Atlantic States Marine Fisheries Commission

Atlantic Coast Diadromous Fish Habitat: A Review of Utilization, Threats, Recommendations for Conservation, and Research Needs

Alewife — *Alosa pseudoharengus*

American eel — *Anguilla rostrata*

American shad — *Alosa sapidissima*

Atlantic sturgeon — *Acipenser oxyrhynchus oxyrinchus*

Hickory shad — *Alosa mediocris*

Striped bass — *Morone saxatilis*

Blueback herring — *Alosa aestivalis*

Habitat Management Series #9
January 2009

Working towards healthy, self-sustaining populations of all Atlantic coast fish species or successful restoration well in progress by the year 2015

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ACKNOWLEDGEMENTS

Selberg, Craig Shirey, members of the ASMFC Habitat Committee, and others that we might have forgotten to mention.

Additionally, we would like to extend our gratitude to Alexa McKerrow, and her crew at North Carolina State University GAP Analysis Project, for compiling the GIS maps for our supplemental DVD.

TABLE OF CONTENTS

LIST OF TABLES

Striped bass tables

Diadromous threats tables

Chapter 1

INTRODUCTION

General Background

The Atlantic States Marine Fisheries Commission (hereafter referred to as ASMFC or the Commission) is the principal agency responsible for the management of many diadromous fish species in state waters. The mission of the Commission's Habitat Program is to work through the Commission, in cooperation with appropriate agencies and organizations, to enhance and cooperatively manage vital fish habitat for conservation, restoration, and protection, and to support the cooperative management of Commission managed species. One of the primary tasks of the Habitat Program is to develop habitat source documents on topics of immediate and broad interest to ASMFC Commissioners that will provide needed information to the states. In this case, Commissioners requested detailed information on the habitat use, threats to habitat, and habitat research needs for all life stages of the ASMFC-managed diadromous species.

ASMFC coordinates interstate fishery management plans for seven diadromous fish species. Of these seven species, striped bass, Atlantic sturgeon, American shad, hickory shad, alewife, and blueback herring are anadromous; the only ASMFC-managed catadromous species is American eel. Throughout their life history, diadromous fishes occupy a broad range of rivers, bays, and estuaries from Florida to Canada, as well as the Atlantic Ocean. All diadromous fish share the common need for fresh, estuarine, and marine waters at various stages in their development. Some of these species, such as the alosines, share similar life history characteristics and range of habitat as well.

Under the 1996 reauthorization of the Magnuson-Stevens Fishery Conservation and Management Act, federal Fishery Management Councils were required to identify *essential fish habitat* (EFH) for all species under federal management; federal agencies proposing projects within EFH areas would then be required to consult with NMFS to determine the impact of those projects on EFH. This mandate was required only for federally managed species, not for species solely under the management authority of interstate Marine Fisheries Commissions. The ASMFC subsequently chose to adopt EFH designations prepared by the federal Fishery Management Councils for any species managed jointly or in association with the Councils. For species solely under Commission management, the Commission has chosen to identify all habitat and Habitat Areas of Particular Concern (HAPCs), but will refrain from identification of EFH. The HAPCs identified by the Commission do not require consultations, or any other regulatory compliance authority.

HAPCs are areas within EFH that may be designated according to the Essential Fish Habitat Final Rule (2002) based on one or more of the following considerations: (i) the importance of the ecological function provided by the habitat, (ii) the extent to which the habitat is sensitive to human-induced environmental degradation, (iii) whether, and to what extent, development activities are, or will be, stressing the habitat type, or (iv) the rarity of the habitat type. Since descriptions of EFH are not currently included in Commission Fishery Management Plans (FMPs), the HAPC definition has been modified to include areas within the species' habitat that satisfy one or more of the aforementioned criteria. A HAPC is a subset of habitats the species is known to occupy, and could include spawning habitat, nursery habitat for larvae, juveniles, and subadults, and/or some amount of foraging habitat for mature adults. HAPCs are geographic locations that are particularly critical to the survival of a species.

All Atlantic coast states are impacted by numerous threats to their natural resources; diadromous fish species are particularly vulnerable because they utilize both coastal and inland habitat during portions of their life history. Poor water quality, altered habitat, blocked access, suboptimal conditions, and invasive species are just a few of the conditions that jeopardize many fish. According to the ASMFC Five-Year Strategic Plan (2009-2013), the loss and degradation of nearshore marine and estuarine fish habitat is a significant factor affecting the long-term sustainability of the nation's fisheries. Diadromous fish species occupy these habitats during a critical period in their life history; it is therefore imperative that fisheries managers provide coordinated management of these areas.

In 2006, the National Fish Habitat Action Plan (NFHAP) was adopted to address the need for improved coordination of fisheries conservation efforts throughout the nation. Currently, the existing NFHAP lacks a Habitat Conservation Plan that directly addresses the needs of diadromous fish species. This document will serve as a basis for the development of the diadromous portion of a conservation strategy for the Atlantic Coastal Fish Habitat Partnership (ACFHP). The ACFHP hopes to conserve habitat for Atlantic coastal, estuarine-dependent, and diadromous fish.

Ecological Significance of Diadromous Fish Species

Diadromous fish have historically played a critical ecological role throughout the range of their habitats. For example, in freshwater, adult shad and river herring returning to spawn are assumed to be food for other fish, reptiles (e.g., snakes and turtles), birds (e.g., ospreys, green herons, eagles, cormorants), and mammals (e.g., mink). Egg, larval, and juvenile shad and river herring may also be consumed by both vertebrate and invertebrate predators in freshwater, estuarine, and nearshore environments. Shad and river herring, which spend several years in the marine environment growing to maturity, bring a significant source of nutrient input to freshwater and estuarine environments. In a second example, American eel are preyed upon by a variety of fish, mammals, and birds, including mink, raccoon, striped bass, and bald eagles. As American eel can contribute up to 25% of the biomass in individual systems, they may be a very important part of the food web. In a third example, documented freshwater predators of Atlantic sturgeon include gar and sea lamprey, and in marine waters, Atlantic sturgeon may be preyed upon by birds, seals, sharks, and other fish.

In addition, semelparous American shad in south Atlantic coastal rivers were a significant food source before their decline. Furthermore, adult blueback herring are commonly preyed upon by striped bass. Although striped bass populations were once depleted, they have now fully recovered; this increased predation may have contributed to a decline in blueback herring abundance in the Connecticut River since 1992. In recent years, predation on alewife and blueback herring by double-crested cormorants staging near the entrance to fishways has increased dramatically in Rhode Island rivers. Predation by otters and herons has also increased in the same area, but to a lesser extent.

Additionally, diadromous fish have historically been a significant food source for human consumption. For example, American eel was once an important food source to Native Americans and early European settlers due to their high nutritional value. They are considered to have the highest nutritional value of fish. In addition, Atlantic sturgeon have been a valuable resource since pre-colonial times. This species was often used by Native Americans, as

evidenced by remains at archeological sites. Atlantic sturgeon were harvested as early as the 1600's by colonists, and were the primary cash crop in Jamestown before tobacco. Their leather was used for clothing and bookbinding, and swim bladders were used for carriage windows and to make gelatin for jellies, wine, beer, and glue. Atlantic sturgeon were also used as fertilizer for plants and fuel for steam-powered vessels. In the 1870's, a major fishery was established for caviar, and within a hundred years, the fishery had completely collapsed.

Most American shad stocks are at historically low levels, and landings have plummeted from a peak of 30,000 metric kg at the turn of the century to a low of 0.6 million kg in 1996. Hickory shad, whose meat is bony and regarded as inferior to American shad, but is prized for its roe, has supported minor commercial fisheries. It is highly sought after by sport fishermen when adults ascend rivers and tributaries during their spawning run, and numbers of fish and landings have increased significantly in recent years. For American eel, landings in the United States have fallen from a high of 1.8 million pounds in 1985 to a low of 641,000 pounds in 2002. For Atlantic sturgeon, in the late 1800's a caviar fishery was established, and by 1890, harvest peaked at approximately 3350 mt (7 million lbs), which lead to a significant reduction in population size. By 1901, landings were 10% of the former peak at 295 mt. Further reductions in populations occurred in the 1970's and 1990's, with landings in the 1990s averaging 84.2 mt. As a result, in 1998, the ASMFC initiated a 40-year commercial fishing moratorium.

Document Content

This document is the most comprehensive compilation of habitat information to date on Commission-managed diadromous species. The primary focus of this document is on inshore and nearshore habitats along the Atlantic coast for all life stages of the included species, but offshore habitat is also discussed. In contrast with the catadromous American eel, the six anadromous species discussed spawn in fresh or brackish waters and spend a portion of their juvenile/sub-adult life stage in freshwater and/or brackish waters. However, American eel spawn in saltwater; following an oceanic larval stage, they migrate to fresh or brackish waters to grow to maturity. Inland and coastal waters provide critical habitat for spawning, growth, feeding, and in some cases, residential habitat for diadromous fish species. Thus, impacts to these areas are likely to have consequences for species that rely on these areas.

In 1998, the Commission published the *ASMFC Guidance for the Development of FMP and Source Document Habitat Sections* (since revised in the *2008 ASMFC Habitat Program Operational Procedures Manual*), which served as the primary guide for preparation of this document. Currently, Commission FMPs and FMP amendments contain varying degrees of habitat information, including habitat-related management objectives and recommendations. Therefore, this document will serve as a tool for fisheries managers to amend existing FMPs to include the most current and comprehensive habitat information.

The Commission's FMP guidance document indicates that the best available information and data should be used in the development of habitat sections, including, but not limited to, peer reviewed literature, gray literature, personal communication with knowledgeable professionals, and unpublished information with adequate citations. In accordance with this directive, this document has utilized many available sources, including state, federal, and private sources to cover the major sections required for FMPs. Furthermore, maps were developed using a GIS

interface that provide a comprehensive source of spawning habitat information for Commission-managed anadromous species (see DVD supplement).

The authors of this document mined existing data sources that identified confirmed or suspected habitats, and those that were deemed important or essential (see text of this document as well as tables included on supplemental DVD). Many new studies have been conducted in recent years, including physical, chemical, and ecological requirements, and are included in this document. Information about the condition of existing habitat has been assessed in some areas, as well as recommendations for reversing impacts or preserving the status quo.

In addition, all Atlantic coastal states submitted a State Wildlife Action Plan (SWAP) to the U.S. Fish and Wildlife Service in 2005. The purpose of the State Wildlife Grants Program is to provide federal dollars to every state and territory to support conservation efforts to prevent wildlife from becoming endangered. The amount of information on diadromous fish species varies within individual SWAPs, but collectively, this represents a significant amount of data that was not previously available before publication of this document. Inclusion of this information provides fish habitat managers with additional resources to identify and protect important habitats.

Unfortunately, we still lack a complete understanding of what habitats are essential to a given species, what the effects of anthropogenic activities are on habitat, and what can be done to mitigate these impacts. This document attempts to address some of these concerns. By identifying all known and suspected habitat, habitat managers can begin to piece together the full range of habitat that each species occupies. Information about physical, chemical, and ecological requirements may help managers to delineate essential habitat for each species at various life history stages. Where information exists on present condition of habitat, managers can predict the fate of resident species. Finally, recommendations for conservation and restoration can be developed to ensure that there will be adequate habitat for all diadromous fish species.

Chapter 2

AMERICAN SHAD

(*Alosa sapidissima*)

Section I. American Shad Description of Habitat

American Shad General Habitat Description and Introduction

American shad (*Alosa sapidissima*) are an anadromous, pelagic, highly migratory, schooling species (Colette and Klein-MacPhee 2002). The historical range of American shad extended from Sand Hill River, Labrador, Newfoundland, to Indian River, Florida, in the western Atlantic Ocean (Lee et al. 1980; Morrow 1980). The present range extends from the St. Lawrence River in Canada to St. Johns River, Florida. In addition, American shad were introduced to the Sacramento River in California, and the Columbia, Snake, and Willamette rivers in Oregon in the late 1800s. Since that time, the species' range in the Pacific Ocean has expanded to Cook Inlet, Alaska, and the Kamchatka Peninsula, Russia, south to Todos Santos Bay, Baja California (Lee et al. 1980; Howe 1981). Attempts to introduce the species in the Gulf of Mexico, Mississippi River drainage, Colorado streams, and the Great Lakes were unsuccessful (Walburg and Nichols 1967; Whitehead 1985). Interestingly, a landlocked population exists in a reservoir of the San Joaquin River on the Pacific coast, but no landlocked populations have been reported along the Atlantic coast (Zydlewski and McCormick 1997a). This document will focus on behaviors of Atlantic populations of anadromous American shad.

American shad spend most of their lives in marine waters, with adults migrating into coastal rivers and tributaries to spawn. On average, American shad spend four to five years at sea, and some individuals from the southernmost range may travel over 20,000 km during this time period (Dadswell et al. 1987). Researchers believe that the historical spawning range of American shad included all accessible rivers and tributaries along the Atlantic coast (MacKenzie et al. 1985). Additionally, rivers, bays, and estuaries associated with spawning reaches are used as nursery areas by American shad (ASMFC 1999).

Over the past 170 years, declines in American shad stocks have been attributed to overfishing, pollution, and habitat loss due to dams, upland development, and other factors (Limburg et al. 2003). Turn of the century catch levels of 30,000 metric tons (Walburg and Nichols 1967) have dropped considerably to a low of 600 metric tons in 1996 (ASMFC 1999). Overfishing contributed to the decline in American shad landings in many East Coast rivers; this decline is seen in harvest records from the 1950s to the 1970s (Talbot 1954; Walburg 1955, 1963; Williams and Bruger 1972; Sholar 1976). Unfortunately, due to habitat loss, American shad stocks have continued to decline in many coastal rivers, including the Hudson River, New York. However, some populations, such as in the Connecticut River, the Pawcatuck River, Rhode Island, and the Santee River, South Carolina, have stabilized or are increasing in numbers (ASMFC 1988; Cooke and Leach 2003).

In 1998, an assessment of American shad confirmed that most stocks were not overfished, however, overall stock abundance was historically low. Researchers concluded that, "the current strategy to restore American shad stocks by improving habitat and fish passage, stocking, and inter-basin transfers will yield much stronger dividends than a strategy of stock restoration based solely on reduction of fishing mortality" (Boreman and Friedland 2003).

Although there is an abundance of literature on adult American shad migration trends, migration physiology, and young-of-the-year ecology, research on American shad habitat

requirements is greatly needed. Much of the information contained in this chapter was derived from fisheries surveys, and research studies on American shad and other fish from the sub-family Alosinae (also referred to as "alosines").

Part A. American Shad Spawning Habitat

Geographical and temporal patterns of migration

The existing Atlantic coast stocks of American shad have a geographic range that currently extends from the St. Johns River, Florida, to the St. Lawrence River, Canada (see above for historic range). Scientists estimate that this species once ascended at least 130 rivers along the Atlantic coast to spawn, but today fewer than 70 systems have runs (Limburg et al. 2003). Most American shad return to their natal rivers and tributaries to spawn (Fredin 1954; Talbot 1954; Hill 1959; Nichols 1966; Carscadden and Leggett 1975), although on average, 3% stray to non-natal river systems (Mansueti and Kolb 1953; Williams and Daborn 1984; Melvin et al. 1985). In fact, Hendricks et al. (2002) demonstrated that hatchery-reared American shad homed to a specific tributary within the Delaware River system several years after stocking, and also preferred the side of the tributary influenced by the plume of their natal river.

The degree of homing by American shad may depend on the nature of the drainage system. If so, mixing of stocks and consequent straying would more likely occur in large and diversified estuarine systems, such as the Chesapeake Bay, while more precise homing could be expected in systems that have a single large river, such as the Hudson River (Richkus and DiNardo 1984).

Timing	Month	Location	Citation
Begin	December	St. Johns River, FL	Williams and Bruger 1972
Peak	January	St. Johns River, FL	Leggett 1976
Begin	mid-January	GA and SC	Walburg and Nichols 1967; Leggett and Whitney 1972
Begin	mid-February	NC and VA	Walburg and Nichols 1967; Leggett and Whitney 1972
Peak	March	NC and VA	Walburg and Nichols 1967; Leggett and Whitney 1972
Peak	April	Potomac River	Walburg and Nichols 1967; Leggett and Whitney 1972
Peak	early May	Delaware River	Walburg and Nichols 1967; Leggett and Whitney 1972
Range	March-June	Hudson & Connecticut rivers	Walburg and Nichols 1967;

Timing	Month	Location	Citation
			Leggett and Whitney 1972
Range	June-August	Androscoggin River, Maine	Brown and Sleeper 2004
End	July-August	Canadian rivers	MacKenzie et al. 1985;
			Scott and Scott 1988

Table 2-1. American shad temporal spawning trends along the Atlantic coast of North America

American shad spring spawning migrations begin in the south and move gradually north as the season progresses and water temperatures increase (Table 2-1; Walburg 1960). Spawning runs typically last 2-3 months, but may vary depending on weather conditions (Limburg et al. 2003). The diel timing of migration may not vary greatly from region to region. In the James River, Virginia, spawning adults ascended mostly between 0900 and 1600 hours (Weaver et al. 2003). Arnold (2000) reported similar results in the Lehigh River, Pennsylvania, where American shad passed primarily between 0900 and 1400 hours.

American shad show varied preferences for migration distance upstream depending on the river system. There does not seem to be a minimum distance from brackish waters at which spawning occurs (Leim 1924; Massmann 1952), but upstream and mid-river segments appear to be favored (Massmann 1952; Bilkovic et al. 2002). It is not unusual for American shad to travel 25 to 100 miles upstream to spawn; some populations historically migrated over 300 miles upstream (Stevenson 1899; Walburg and Nichols 1967). In the 18th and 19th centuries, American shad runs were reported as far inland as 451 miles along the Great Pee Dee and Yadkin rivers in North Carolina (Smith 1907) and over 500 miles in the Susquehanna River (Stevenson 1899).

Male American shad arrive at riverine spawning grounds before females (Leim 1924). Females release their eggs close to the water surface to be fertilized by one or several males. Diel patterns of egg release depend upon water turbidity and light intensity. In clear open water, eggs are released and fertilized after sunset (Leim 1924; Whitney 1961), with peak spawning around midnight (Massmann 1952; Miller et al. 1971; 1975). In turbid waters (or on overcast days; Miller et al. 1982), eggs are released and fertilized during the day (Chittenden 1976a). For example, in the Pamunkey River, Virginia, spawning has been observed throughout the day, which may be due to relatively turbid waters damping light intensity (Massmann 1952). These findings support the hypothesis of Miller et al. (1982) that daily spawning is regulated by light intensity.

Another interesting aspect of American shad migration is the regional difference in spawning periodicity. American shad that spawn north of Cape Hatteras are iteroparous (repeat spawners), while almost all American shad spawning south of Cape Hatteras are semelparous (die after one spawning season). This may be due to the fact that south of North Carolina the physiological limits of American shad are stretched during long oceanic migrations; higher southern water temperatures may also have an effect (Leggett 1969). Moreover, Leggett and Carscadden (1978) suggest that southern stocks produce more eggs per unit of body weight than northern populations to compensate for not spawning repeatedly.

Location	% of repeat spawners	Citations
Neuse River, NC	3	Leggett and Carscadden 1978
York River, VA	24	Leggett and Carscadden 1978
Connecticut River	63	Leggett and Carscadden 1978
Saint John River, Canada	73	Colette and Klein-MacPhee 2002

Table 2-2. Percentage of repeat spawners for American shad along the Atlantic coast of North America

Studies show the percentage of iteroparous adult American shad increases northward along the Atlantic coast (Table 2-2). However, the percentage of repeat spawners may fluctuate over time within the same river due to pollution, fishing pressure, land-use change, or other factors (Limburg et al. 2003). Furthermore, almost 59% of American shad in the St. Lawrence River did not spawn every year following the onset of maturation, skipping one or more seasons (Provost 1987). Additionally, some fish spawn up to five times before they die (Carscadden and Leggett 1975).

Members of this species exhibit asynchronous ovarian development and batch spawning. In addition, American shad spawn repeatedly as they move upriver (Glebe and Leggett 1981a), which some researchers think may be a function of their high fecundity (Colette and Klein-MacPhee 2002). Estimates of egg production for the York River, Virginia, are 20,000 to 70,000 eggs per kg somatic weight spawned every four days (Olney et al. 2001).

However, some researchers believe that fecundity in American shad may be indeterminate, and that previous annual or lifetime fecundity estimates may not be accurate (Olney et al. 2001). Researchers examining batch fecundity of semelparous American shad in the St. Johns River, Florida, and iteroparous individuals in the York and Connecticut rivers in Virginia and Connecticut, respectively, found no statistically significant differences in batch fecundity among the populations. Until spawning frequency, duration, and batch size throughout the spawning season are known, lifetime fecundity for various stocks cannot be determined and previous methods to determine fecundity throughout the coastal range will be inadequate (Olney and McBride 2003). Nevertheless, the habitat productivity potential estimate used in Maine is 2.3 shad per 100 square yards of water surface area (Brown and Sleeper 2004).

It is interesting to note that Olney et al. (2001) found that approximately 70 percent of post-spawning American shad females leaving the York River had only partially spent ovaries, which suggests that the maximum reproduction level of most females in the river system each year is not achieved. Researchers hypothesize that these females utilize partially spent ovaries by reabsorbing unspawned, yoked oocytes to supplement somatic energy sources as they return to the ocean. These fish likely have a greater potential for surviving multiple spawning events than individuals that are fully spent and have no such energy reserves (Olney et al. 2001). Even with energy reserves, spent adults are usually very emaciated and return to sea soon after spawning (Chittenden 1976b), sometimes feeding before reaching saltwater (Atkins 1887).

Layzer (1974) found that American shad selected discrete spawning sites in the Connecticut River and remained there for most of the season despite the large area available for spawning. Sometimes spawners forego areas with highly suitable habitats that are further downstream, suggesting that there are other variables that influence habitat choice (Bilkovic 2000). Ross et al. (1993) suggest that choice of spawning habitat may be unrelated to physical variables, but rather may reflect a selective pressure such as fewer egg predators in selected habitats.

Spawning and the saltwater interface

Adult American shad may spend two to three days in estuarine waters prior to upriver migration (Dodson et al. 1972; Leggett 1976). Leim (1924) observed spawning by American shad in brackish waters, but other researchers believe that spawning occurs only in freshwater (Massman 1952; MacKenzie et al. 1985). Spawning typically occurs in tidal and non-tidal freshwater regions of rivers and tributaries (Chittenden 1976a). While in the Hudson River, American shad ascend beyond the saltwater interface and go as far upstream as they can travel (Schmidt et al. 1988), eggs are typically deposited slightly above the range of tide in the Shubenacadie River, Canada (Leim 1924). In many rivers, adult spawners historically migrated beyond tidal freshwater areas, but they can no longer reach these areas due to dam blockages (Mansueti and Kolb 1953).

Interestingly, American shad tolerate a wide range of salinities during early developmental stages (Chittenden 1969) and adult years (Dodson et al. 1972), even though their eggs are normally deposited in freshwater (Weiss-Glanz 1986). Additionally, Limburg and Ross (1995) concluded that a preference for upriver spawning sites may be genetically fixed, but its advantage or significance was not related to salt intolerance of eggs and larvae.

Leggett and O'Boyle (1976) conducted an experiment to see if American shad require a period of acclimation to freshwater. The researchers determined that fish transferred from seawater to freshwater, with a 6°C temperature increase over a 2.5-hour period, experienced physiologic stress and a 54% mortality rate five hours later. Furthermore, adults did not survive transfers from saltwater (27 ppt) to freshwater with a 14°C temperature increase. Mortality rates varied from 0 to 40% for transfers from waters with salinities ranging from 13 to 25 ppt to freshwater and temperature increases up to 6°C. However, adult American shad may be better adapted to transfers from freshwater to saltwater. They tolerated transfers from freshwater to 24 ppt and temperature increases of up to 9°C (Leggett and O'Boyle 1976).

Spawning substrate associations

Spawning often occurs far upstream or in river channels dominated by flats of sand, silt, muck, gravel, or boulders (Mansueti and Kolb 1953; Walburg 1960; Walburg and Nichols 1967; Leggett 1976; Jones et al. 1978). The importance of substrate type to American shad spawning behavior is still debated. Bilkovic et al. (2002) concluded that substrate type was not predictive of spawning and nursery habitat in two Virginia rivers that were surveyed. Similarly, Krauthamer and Richkus (1987) do not consider substrate type to be an important factor at the spawning site since eggs are released into the water column.

However, eggs are semi-buoyant and may eventually sink to the bottom. Thus, areas predominated by sand and gravel may enhance survival because there is sufficient water velocity to remove particles and prevent suffocation if eggs settle to the bottom (Walburg and Nichols 1967). Furthermore, Layzer (1974) noted that survival rates of shad eggs were highest where gravel and rubble substrates were present. Likewise, Hightower and Sparks (2003) hypothesize that larger substrates are important for American shad reproduction, based on observations of spawning in the Roanoke River, North Carolina. Other researchers have also observed American shad spawning primarily over sandy bottoms free of mud and silt (Williams and Bruger 1972).

Spawning depth associations

Depth is not considered a critical habitat parameter for American shad in spawning habitat (Weiss-Glanz et al. 1986), although Witherell and Kynard (1990) observed adult American shad in the lower half of the water column during the upstream migration. Once they reach preferred spawning areas, adults have been found at river depths ranging from 0.45 to 10 m (Mansueti and Kolb 1953; Walburg and Nichols 1967). However, depths less than 4 m are generally considered ideal (Bilkovic 2000).

Ross et al. (1993) observed that the greatest level of spawning occurred where the water depth was less than 1 m in the Delaware River. Other studies suggest that adults select river areas that are less than 10 ft deep (3.3 m) or have broad flats (Mansueti and Kolb 1953; Leggett 1976; Kuzmeskus 1977). Adults may reside in slow, deep pools during the day, and in the evening move to shallower water where riffle-pools may be present to spawn (Chittenden 1969; Layzer 1974). During the spawning event, females and males can be found close to the surface for the release and fertilization of eggs (Medcof 1957).

Stier and Crance (1985) suggest that for all life history stages, including spawning, egg incubation, larvae, and juveniles, the optimum depth range is between 1.5 and 6.1 m. Depths less than 0.46 m (for spawning adults, larvae, and juveniles) and 0.15 m (for egg incubation), and depths greater than 15.24 (for all life history stages) are considered unsuitable (Stier and Crance 1985). However, recent studies on optimal habitat for spawning events have found that these areas may be defined more narrowly than indicated by studies focused primarily on egg collection. For example, sites deeper than 2 m in the Neuse River, North Carolina, were used less extensively than expected for spawning based on depth availability within the spawning grounds and over the entire river (Beasley and Hightower 2000; Bowman and Hightower 2001).

Spawning water temperature

Activity	Temperature (^oC)	Location	Citation
Migration	5 - 23	Throughout range	Walburg and Nichols 1967
Migration (peak)	8.6 - 19.9 (16 - 19)	North Carolina	Leggett and Whitney 1972
Peak migration	16.5 - 21.5	Southern rivers	Leggett 1976
Spawning	8 - 26	Throughout range	Walburg and Nichols 1967;

Activity	Temperature (^{o}C)	Location	Citation
			Stier and Crance 1985
Optimum spawning	14 - 20	Throughout range	Stier and Crance 1985
Optimum spawning	14 – 24.5	Throughout range	Ross et al. 1993

Table 2-3. American shad migration and spawning temperatures for the Atlantic coast

Spawning for American shad may occur across a broad range of temperatures (Table 2-3). Water temperature is the primary factor that triggers spawning, but photoperiod, water flow and velocity, and turbidity also exert some influence (Leggett and Whitney 1972). Based on the temperature range reported by Leggett and Whitney (1972), Parker (1990) suggests that pre-spawning adults tolerate higher temperatures as they undergo physiological changes and become sexually ripe.

Most spawning occurs in waters with temperatures between 12-21°C (Walburg and Nichols 1967; Leggett and Whitney 1972). Generally, water temperatures below 12°C cause total or partial cessation of spawning (Leim 1924). However, Jones et al. (1978) reported American shad moving into natal rivers when water temperatures were 4° C or lower. Additionally, Marcy (1976) found that peak spawning temperatures varied from year to year. For example, peak spawning temperatures in the Connecticut River were 22°C and 14.8°C in 1968 and 1969, respectively (Marcy 1976).

Other factors, such as the pace of gonadal and egg development may also be related to water temperature. Mansueti and Kolb (1953) found that shad ovaries developed more slowly at 12.8°C than at 20 to 25°C. In theory, eggs may develop slowly at first then mature rapidly with higher temperatures (DBC 1980).

Spawning dissolved oxygen associations

American shad require well-oxygenated waters in all habitats throughout their life history (MacKenzie et al. 1985). Jessop (1975) found that migrating adults require minimum dissolved oxygen (DO) levels between 4 and 5 mg/L in the headwaters of the Saint John River, New Brunswick. Dissolved oxygen levels below 3.5 mg/L have been shown to have sub-lethal effects on American shad (Chittenden 1973a); levels less than 3.0 mg/L completely inhibit upstream migration in the Delaware River (Miller et al. 1982). Additionally, dissolved oxygen levels less than 2.0 mg/L cause a high incidence of mortality (Tagatz 1961; Chittenden 1969), and below 0.6 mg/L cause 100% mortality (Chittenden 1969). Although minimum daily dissolved oxygen concentrations of 2.5 to 3.0 mg/L should be sufficient to allow American shad to migrate through polluted areas, Chittenden (1973a) recommends that suitable spawning areas have a minimum of 4.0 mg/L. Miller et al. (1982) propose even higher minimum concentrations, suggesting that anything below 5.0 mg/L should be considered potentially hazardous to adult and juvenile American shad.

Spawning water velocity/flow

Water velocity (m/sec) is an important parameter for determining American shad spawning habitat (Stier and Crance 1985). Walburg (1960) found that spawning and egg incubation most often occurred where water velocity was 0.3 to 0.9 m/s. In support, Stier and Crance (1985) suggested that this was the optimum range for spawning areas. Ross et al. (1993) observed that American shad spawning activity was highest in areas where water velocity ranged from 0.0 to 0.7 m/s; this suggested that there was no lower suitability limit during this stage and that the upper limit should be modified. However, Bilkovic (2000) determined that the optimum water velocity range for eggs and larvae was 0.3 to 0.7 m/s, and hypothesized that some minimum velocity was required. A minimum velocity is needed in order to prevent siltation and ensure that conditions conducive to spawning and egg incubation occur (Williams and Bruger 1972; Bilkovic 2000).

Appropriate water velocity at the entrance of a fishway is also important for American shad migrating upstream to spawning areas. Researchers found that water velocities of 0.6 to 0.9 m/s at the entrance to a pool-and-weir fishway was needed to attract American shad to the structure (Walburg and Nichols 1967). The Conowingo Dam fish lift on the Susquehanna River uses entrance velocities of 2 to 3 m/s to attract American shad to the lift (R. St. Pierre, U.S. Fish and Wildlife Service, personal communication). At other sites, such as the Holyoke Dam in Massachusetts, American shad have trouble locating fishway entrances among turbulent discharges and avoid the area; thus, too much water velocity and/or turbulence may actually deter this species (Barry and Kynard 1986).

Ross et al. (1993) noted that habitat selection among spawning adult American shad favored relatively shallow (0.5 to 1.5 m) mid-river runs with moderate to high current velocity (0.3-0.7 m/s). To a lesser degree, adults also were located in channels (deeper, greater current velocities, little if any SAV) and SAV shallows (inshore, high densities of SAV, low current velocities). The researchers found adults seemed to avoid pools (wide river segment, deep, low current velocities) and riffle pools (immediately downstream of riffles, deep water, variable current velocity and direction) that contained both deep and slow water. This avoidance of pools and riffle pools may be explained by the fact that the preferred run habitat contained both swift and shallow water characteristics. Channels and SAV shallows may be either swift or shallow; these characteristics may lead to higher survivability of newly spawned eggs compared to deep pool habitat (Ross et al. 1993). Similarly, Bilkovic et al. (2002) found the greatest level of spawning activity in runs.

Water velocity may also contribute in some way to weight loss and mortality during the annual spawning migration, especially for male American shad. Males typically migrate upstream earlier in the season when water velocities are greater, thus expending more energy than females (Glebe and Leggett 1973; DBC 1980).

In addition, areas with high water flows provide a cue for spawning American shad (Orth and White 1993). In 1985, a rediversion canal and hydroelectric dam constructed between the Cooper River and Santee River, South Carolina, increased the average flow of the Santee River from 63 m^3/s to 295 m^3/s. (Cooke and Leach 2003). The increased river flow and access to spawning grounds through the fish passage facility have contributed to increases in American shad populations. Although the importance of instream flow requirements has been previously recognized with regard to spawning habitat requirements or recruitment potential (Crecco and

Savoy 1984; ASMFC 1985; Crecco et al. 1986; Ross et al. 1993; Moser and Ross 1994), Cooke
and Leach (2003) suggested that river flow might be an important consideration for restoring
alosine habitat.

Water flow may have additional importance for American shad populations in the future.
Although Summers and Rose (1987) did not detect direct relationships between stock size and
river flow or water temperature, they found that spawning stock size, river flow rate, and
temperature were important predictors of future American shad population sizes. These
researchers suggested that future studies incorporate a combination of environmental variables,
rather than a single environmental variable, to determine what stimuli affect stock size.

Spawning suspended solid associations

Adults appear to be quite tolerant of turbid water conditions. In the Shuebenacadie River,
Nova Scotia, suspended solid concentrations as high as 1000 mg/L did not deter migrating adults
(Leim 1924). Furthermore, Auld and Schubel (1978) found that suspended solid concentrations
of 1000 mg/L did not significantly affect hatching success of eggs.

Spawning feeding behavior

Early research suggested that adult American shad did not feed in freshwater during
upstream migration or after spawning (Hatton 1940; Moss 1946; Nichols 1959) because the most
available food source in the freshwater community was too small to be retained by adult
gillrakers (Walburg and Nichols 1967). Atkinson (1951) suggested that American shad stopped
feeding due to the physical separation from suitable food sources rather than a behavioral or
physiological reduction in feeding.

More recent studies of feeding habits of American shad in the York River, Virginia,
found that individuals did, in fact, feed as they migrated from the oceanic to coastal waters
(Chittenden 1969, 1976b; Walters and Olney 2003). Walters and Olney (2003) compared
stomach fullness of migrating American shad with individuals in the ocean and estuary, and
found that as American shad moved from oceanic waters to coastal and estuarine waters their
diet composition changed from oceanic copepods, such as *Calanus finmarchicus*, to other
copepods, such as *C. typicus* and *Acartia* spp. (Walters and Olney 2003). The estuarine mysid
shrimp *Neomysis americana* became an important component, replacing euphausids in spent and
partially spent adults. Minor amounts of other crustaceans were also found in spent American
shad stomachs including cumaceans, sevenspine bay shrimp (*Crangon septemspinosa*), and
gammarid amphipods, as well as woody and green plant debris that had little or no nutritional
value (Walters and Olney 2003). This finding suggested that these fish fed if there was suitable
prey available (Atkinson 1951).

The ability to feed during migration and after spawning may be an important factor in
decreasing post-spawning mortality of American shad (Walters and Olney 2003). Migration
requires significant energetic expenditures and causes weight loss (Glebe and Leggett 1981a;
1981b); the resumption of feeding likely represents a return to natural feeding patterns, which
allows the fish to begin regaining lost energy reserves (Walter and Olney 2003). Finally, the
ability to survive spawning has been correlated with the degree of energy lost (Glebe and Leggett
1981b; Bernatchez and Dodson 1987). Therefore, American shad that feed actively before and

after spawning may have a higher likelihood of repeat spawning. Additionally, individuals whose spawning grounds are in closer proximity to estuarine food sources (and do not expend as much energy as those that have to travel farther), and emigrating fish that have partially spent ovaries that can be reabsorbed for energy (Olney et al. 2001), may have a higher frequency of repeat spawning and lower energy expenditures (Walter and Olney 2003).

Spawning competition and predation

Early studies found that seals and humans preyed upon adult American shad (Scott and Crossman 1973), but the species appeared to have few other predators (Scott and Scott 1988). Erkan (2002) found that predation of alosines has increased in Rhode Island rivers, noting that the double-crested cormorant often takes advantage of American shad staging near fishway entrances. Predation by otters and herons has also increased, but to a lesser extent (D. Erkan, Rhode Island Division of Fish and Wildlife, personal communication). A recent study strongly supports the hypothesis that striped bass predation on adult American shad in the Connecticut River has resulted in a dramatic and unexpected decline in American shad abundance since 1992 (Savoy and Crecco 2004). Researchers further suggest that striped bass prey primarily on spawning adults because their predator avoidance capability may be compromised at that time, due to a strong drive to spawn during upstream migration. Rates of predation on ages 0 and 1 alosines was also much lower (Savoy and Crecco 2004).

In south Atlantic coastal rivers where the percentage of repeat spawning is low or non-existent, adult American shad that die after spawning may contribute significant nutrient input from the marine system into freshwater interior rivers (ASMFC 1999). Garman (1992) hypothesized that before recent declines in abundance, the annual input of marine-derived biomass of post-spawning alosines was an important seasonal source of energy and nutrients for the non-tidal James River.

Part B. American Shad Egg and Larval Habitat

Geographical and temporal movement patterns

American shad eggs and larvae have been found at, or downstream of, spawning locations. Upstream areas typically have extensive woody debris where important larval and juvenile American shad prey items reside, and spawning there may ensure that eggs develop within favorable habitats (Bilkovic et al. 2002).

Once American shad eggs are released into the water column, they are initially semi-buoyant or demersal. Survival of eggs is dependent on several factors, including current velocity, dissolved oxygen, water temperature, suspended sediments, pollution, and predation (Krauthamer and Richkus 1987; Bailey and Houde 1989). Whitworth and Bennett (1970) monitored American shad eggs after they were broadcast and found that they traveled a distance of 5 to 35 m downstream before they sank or became lodged on the bottom. Other researchers reported similar observations (Barker 1965; Carlson 1968; Chittenden 1969).

Laboratory experiments suggested that sinking rates for American shad eggs were around 0.5 to 0.7 m/min (1.6 to 2.4 ft/min), with newly spawned eggs sinking at a quicker rate, although hydrodynamic and tidal effects were not accounted for in the experiments (Massmann 1952; Chittenden 1969). Other factors, such as amount of woody debris, influence how far eggs travel and may prevent eggs from settling far from the spawning site (Bilkovic 2000). Once eggs sink to the bottom, they are swept under rocks and boulders and are kept in place by eddy currents. In addition, eggs may become dislodged and swept downstream to nearby pools (DBC 1980).

American shad yolk-sac larvae may not use inshore habitat as extensively as post-yolk-sac larvae (Limburg 1996). One early study (Mitchell 1925, cited by Crecco et al. 1983) found that yolk-sac larvae were near the bottom and swam to shore as the yolk-sac reabsorbed. Metzger et al. (1992) also found yolk-sac larvae mostly in offshore areas along the bottom, while post yolk-sac larvae were more concentrated in quiet areas near shorelines (Cave 1978; Metzger et al. 1992). Yolk-sac larvae are typically found deeper in the water column than post-larvae, due to their semi-buoyant nature and aversion to light. Post-larvae, in contrast, are more abundant in surface waters, especially downstream of spawning sites (Marcy 1976).

Yolk-sac larvae exhaust their food supply within 4 to 7 days of hatching (Walburg and Nichols 1967), usually when they are approximately 10 to 12 mm total length (TL) (Marcy 1972). Survival is affected by water temperature, water flow, food production and density, and predation (State of Maryland 1985; Bailey and Houde 1989; Limburg 1996). Larvae may drift passively into brackish water shortly after hatching occurs, or can remain in freshwater for the remainder of the summer (State of Maine 1982); often they aggregate in eddies and backwaters (Stier and Crance 1985). Ross et al. (1993) reported that American shad larvae frequent riffle pools where water depth is moderate and velocity and direction vary. Alternatively, larvae in the Mattaponi and Pamunkey rivers, Virginia, were dispersed from the upper through the downriver areas. Unlike the presence of eggs, which can be predicted in most cases using physical habitat and shoreline/land use ratings, distinct habitat associations could not be discerned for larval distributions. This may be due to the fact that larvae were carried further downstream than eggs, dispersing them into more variable habitats (Bilkovic et al. 2002).

Eggs, larvae, and the saltwater interface

Although American shad eggs are generally deposited in freshwater, it is unknown whether they hatch in freshwater, brackish water, or in both (Weiss-Glanz 1986). Early attempts to acclimate larval shad to seawater resulted in high mortality rates (Milner 1876). Leim (1924) purported that successful development of embryos and larvae occurs under low salinity conditions. In the Shubenacadie River, Canada, eggs and larvae were most often observed in areas with a salinity of 0 ppt (range 0 to 7.6 ppt). Additionally, while larvae may tolerate salinities as high as 15 ppt, these conditions often result in death. Leim (1924) also found that temperature may influence salinity sensitivities, with lower temperatures (i.e., 12°C) resulting in more abnormalities at 15 and 22.5 ppt than higher temperatures (i.e., 17°C).

In another study, Limburg and Ross (1995) found that salinities of 10 to 20% were favorable for post-yolk sac American shad larvae, and concluded that estuarine salinities neither depressed growth rates nor elevated mortality rates of larval American shad compared with freshwater conditions. These researchers concluded that other ecological factors may play a greater role in influencing spawning site selection by American shad than the physiological effects of salinity.

Egg and larval substrate associations

Areas with sand or gravel substrates may be better for egg and larval survival because they allow sufficient water velocity to remove silt or sand that can suffocate eggs (Walburg and Nichols 1967). Additionally, survival rates of American shad eggs have been found to be highest among gravel and rubble substrates (Layzer 1974). According to Krauthamer and Richkus (1987), bottom composition is not a critical factor in the selection of spawning locations for American shad. After American shad eggs are fertilized, they either sink to the bottom where they become lodged under rocks and boulders, or they are swept by currents to nearby pools (Chittenden 1969). Bilkovic (2000) concluded that substrate type was not a good predictor of spawning and nursery habitat in rivers.

Egg and larval depth associations

Eggs are slightly heavier than water, but may be buoyed by prevailing currents and tides. Most eggs settle at, or near, the bottom of the river during the water-hardening stage (Leim 1924; Jones et al. 1978). In the Connecticut River, American shad eggs are distributed almost uniformly between the surface and the bottom of the river. Larvae are more than twice as abundant in surface waters, and are even more abundant in the water column as they move downstream (Marcy 1976).

Walburg and Nichols (1967) found 49% of American shad eggs in waters shallower than 3.3 m (10 ft), 30% in water 3.7 to 6.7 m (11 to 20 ft), and 21% in water 7 to 10 m (21 to 30 ft). Similarly, Massman (1952) reported that five times more eggs per hour were collected at depths ranging from 1.5 to 6.1 m (4.9 to 20.0 ft), than in deeper waters of the Pamunkey and Mattaponi rivers. In the same river systems, Bilkovic et al. (2002) found eggs at depths of 0.9 to 5.0 m, and larvae at 1 to 10 m.

Egg and larval water temperature

Days	Temperature	Reference
15.5	12° C	Leim 1924
17	12° C	Ryder 1887
7	17° C	Leim 1924
3	24° C	MacKenzie et al. 1985
2	27° C	Rice 1878

Table 2-4. American shad egg development time at various temperatures

Rate of development of shad eggs is correlated with water temperature (Table 2-4; Mansueti and Kolb 1953). According to Limburg (1996), within the temperature range of 11 to 27°C, the time it takes for eggs to develop can be expressed as:

$\log_e(EDT) = 8.9 - 2.484 \times \log_e(T)$, where EDT is egg development time in days and T is temperature in degrees Celsius

Estimates of near-surface water temperatures suitable for development and survival of American shad eggs range from 8 to 30°C (Walburg and Nichols 1967; Bradford et al. 1968; Stier and Crance 1985; Ross et al. 1993). Leim (1924) suggests that optimal conditions for American shad egg development occur in the dark at 17°C and 7.5 ppt salinity.

Characterization	Temperature (^{o}C)	Citation
Suitable	10 - 27	Bradford et al. 1968
Suitable	13.0 - 26.2	Ross et al. 1993
Suitable	10 - 30	Stier and Crance 1985
Optimal	15.5 - 26.5	Leim 1924
Optimal	15 - 25	Stier and Crance 1985

Table 2-5. American shad larval temperature tolerance ranges

Water temperatures above 27°C can cause abnormalities or a total cessation of larval American shad development (Bradford et al. 1968). Few larvae have been found living in temperatures above 28°C (Table 2-5; Marcy 1971; 1973), and no viable larvae develop from eggs incubated above 29°C (Bradford et al. 1968). Ross et al. (1993) recommend that further sampling be conducted for post-larval stages at temperatures greater than or equal to 27°C to confirm upper optimal temperature preferences. In this study, the researchers found no reduction in density of larvae at the upper thermal limit (26 to 27°C) in areas sampled along the Delaware River (Ross et al. 1993).

Laboratory experiments have shown that American shad eggs can tolerate extreme temperature changes as long as the exposure is of relatively short duration (Klauda et al. 1991). Temperature increases after acclimation at various temperatures produced variable results; however, some eggs were found to withstand temperatures of 30.5°C for 30 minutes and 35.2°C for 5 minutes (Schubel and Koo 1976). Furthermore, sensitivity to temperature change decreases as eggs mature (Koo et al. 1976).

Shoubridge (1977) analyzed temperature regimes in several coastal rivers throughout the range of American shad, and found that as latitude increases: 1) the duration of the temperature optima for egg and larval development decreases, and 2) the variability of the temperature regime increases. Based on Shoubridge's work, Leggett and Carscadden (1978) suggest that variation in American shad egg and larval survival, year-class strength, and recruitment also increases with latitude.

Crecco and Savoy (1984) found that low water temperatures (with high rainfall and river flow) were significantly correlated with low American shad juvenile abundance during the month of June in the Connecticut River, while high water temperatures (with low river flow and rainfall) were significantly correlated with high juvenile abundance. In addition, depressed water temperatures can retard the onset and duration of American shad spawning (Leggett and Whitney 1972), larval growth rate (Murai et al. 1979), and the production of riverine zooplankton (Chandler 1937; Beach 1960).

Egg and larval dissolved oxygen associations

Miller et al. (1982) concluded that the minimum dissolved oxygen level for both eggs and larvae of American shad is approximately 5 mg/L. This is the value that Bilkovic (2000) assigned for optimum conditions for survival, growth, and development of American shad.

Although specific tolerance or optima data for eggs and larvae is limited, there are studies that note the presence or absence of eggs and larvae under certain dissolved oxygen conditions (Bilkovic et al. 2002). In the Neuse River, North Carolina, American shad eggs were collected in waters with dissolved oxygen levels ranging from 6 to 10 mg/L (Hawkins 1979). Marcy (1976) did not find any American shad eggs in waters of the Connecticut River where dissolved oxygen concentrations were less than 5 mg/L. Bilkovic (2000) found variations in dissolved oxygen concentrations for eggs (10.5 mg/L), yolk-sac larvae (9.0 mg/L), and post-larvae (8.1 mg/L) in the Mattaponi and Pamunkey rivers.

Marcy (1976) determined that the dissolved oxygen LC_{50} values (i.e., concentration that causes 50% mortality) for American shad eggs in the Connecticut River were between 2.0 and 2.5 mg/L. In the Columbia River, the LC_{50} was close to 3.5 mg/L for eggs and at least 4.0 mg/L for a high percentage of hatched eggs and healthy larvae; less than 1.0 mg/L dissolved oxygen resulted in total mortality (Bradford et al. 1968). Klauda et al. (1991) concluded that a good hatch with a high percentage of normal larvae required dissolved oxygen levels during egg incubation of at least 4.0 mg/L, based on observations by both Maurice et al. (1987) and Chittenden (1973a). Finally, it is worth noting that cleanup of the Delaware River has had a measurably positive effect on increasing dissolved oxygen concentrations in that system (Maurice et al. 1987).

Egg and larval pH and aluminum associations

Level	pH	Citation
Tolerance- egg	5.5 - 9.5	Bradford et al. 1968
Tolerance- egg	6.0 – 7.5	Klauda 1994
Tolerance- egg	6.5 - 8.5	Bilkovic et al. 2002
LD_{50}- egg	5.5	Klauda 1994
Mortality- egg	<5.2	Bradford et al. 1968
Tolerance- larvae	6.7 – 9.9	Klauda 1994
Tolerance- larvae	6.5 - 9.3	Bilkovic et al. 2002
Optimal- larvae	>7.0	Leach and Houde 1999
Tolerance- both	6.0 - 9.0	Leim 1924

Table 2-6. American shad egg and larval environmental pH tolerance ranges

A number of researchers have examined the effects of pH on American shad eggs and larvae (Table 2-6). Klauda (1994) hypothesized that even infrequent and temporary episodes of critical or lethal pH and aluminum exposures in spawning and nursery areas could contribute to significant reductions in egg or larval survival and slow stock recovery. Similarly, Leach and Houde (1999) noted that sudden drops in pH levels, such as those associated with rainfall, could cause sudden mortalities for American shad larvae.

In a laboratory study, Klauda (1994) subjected eggs, yolk-sac larvae, and post-larvae to an array of acid and aluminum conditions; larvae appeared to be more sensitive to acid and aluminum pulses than eggs. When eggs were subjected to aluminum pulses, critical conditions were met at pH 5.7 (with 50 or 200 µg/L Al) and pH 6.5 (with 100 µg/L Al) for 96-hour treatments. The least severe treatment that resulted in critical conditions for 1 to 3 day old yolk-sac larvae was a 24 h exposure to pH 6.1 with 92 µg/L Al. The least severe treatment that resulted in a lethal condition for yolk-sac larvae was a 24 h exposure to pH 5.5 with 214 µg/L Al. Furthermore, post-larvae (6 to 16 days old) were found to be more sensitive to acid and aluminum pulses than both eggs and yolk-sac larvae. Critical conditions occurred at pH 5.2 (with 46 µg/L Al) and pH 6.2 (with 54 or 79 µg/L Al) for 8 hours, and lethal conditions occurred at pH 5.2 (with 63 µg/L Al) for 16 hours (Klauda 1994).

Egg and larval water velocity/flow

Several studies report water velocity preferences for larval American shad, with 0 to 1.0 m/s the most commonly reported range (Walburg 1960; Walburg and Nichols 1967; Stier and Crance 1985; Bilkovic et al. 2002). Kuzmeskus (1977) found freshly spawned eggs in areas with water velocity rates between 0.095 and 1.32 m/s. Williams and Bruger (1972) noted that increased siltation may result if water velocities are less than 0.3 m/s, causing increased egg mortality from suffocation and bacterial infection.

Freshwater discharge can influence both eggs and larvae of American shad. Increased river flow can carry eggs from favorable nursery habitat to unfavorable areas that reduce their chance for survival. Lower flows may result in favorable hydrodynamic, thermal, and feeding conditions (Crecco and Savoy 1987a; Limburg 1996). Larval and juvenile American shad may select eddies and backwater areas where water flow is reduced (Crecco and Savoy 1987b). Limburg (1996) found that high spring river discharges coupled with low temperatures and low food availability contributed to high larval mortality in the Hudson River. Larvae that hatched after May, when the highest discharges occurred, had a higher survival rate (Limburg 1996). Furthermore, year-class strength and river flow showed a significant negative correlation in studies conducted on the Connecticut River (Marcy 1976). Larval survival rates have also been negatively correlated with increased river flow in June, but positively correlated with June river temperatures (Savoy and Crecco 1988).

Although hydrographic turbulence may affect larval American shad survival rates, the precise mechanisms of this influence are uncertain because daily river flow and rainfall levels are nonlinear, time-dependent processes that may act singularly or in combination with other factors, such as temperature and turbidity (Sharp 1980). Decreased temperatures can affect larval growth rates (Murai et al. 1979) and riverine zooplankton production that American shad may require for nourishment (Chandler 1937; Beach 1960). Turbulence can also cause turbidity, which may compromise the ability of larval fish to see their prey (Theilacker and Dorsey 1980). Increased turbidity may also affect the food web. Turbidity can cause reduced photosynthesis by phytoplankton, which in turn may lead to elimination of the cladocerans and copepods that American shad feed upon (Chandler 1937; Hynes 1970; Crecco and Blake 1983; Johnson and Dropkin 1995).

Egg and larval suspended solid associations

American shad eggs are less vulnerable to the effects of suspended solids than larvae. For example, Auld and Schubel (1978) found that suspended solid concentrations of up to 1000 mg/L did not significantly reduce hatching success, while larvae exposed to concentrations of 100 mg/L, or greater, had significantly reduced survival rates.

Egg and larval feeding behavior

Predation and starvation are considered the primary causes of mortality among larval fish of many marine species (May 1974; Hunter 1981). Newly hatched American shad larvae must begin feeding within 5 days, or they will die from malnutrition (Wiggins et al. 1984). Furthermore, older larvae have significantly reduced survival rates if they are deprived of food for as little as 2 days (Johnson and Dropkin 1995). Researchers have also found that larvae fed at intermediate prey densities of 500 L^{-1} survived as well as those fed at high prey densities, and significantly higher than starved larvae, which indicates that some minimal level of feeding in riverine reaches can increase survival (Johnson and Dropkin 1995).

Crecco et al. (1983) suggest that larval American shad survival rates are related to spring and summer zooplankton densities. Additionally, despite larval American shad abundance being highest during May, Limburg (1996) determined that year-class was established by cohorts

hatched after June 1 due to more favorable conditions, including warmer temperatures, lower flow rates, and higher zooplankton densities.

Once the yolk-sac is absorbed, American shad larvae consume zooplankton, copepods, immature insects, and adult aquatic and terrestrial insects (Leim 1924; Mitchell 1925; Maxfield 1953; Crecco and Blake 1983; Facey and Van Den Avyle 1986). Several researchers have noted varying levels of selectivity for copepods and cladocerans (Crecco and Blake 1983; Johnson and Dropkin 1995), but zooplankton and chironomids generally comprise the bulk of larval diets (Maxfield 1953; Levesque and Reed 1972). Larval American shad feeding occurs most actively in late afternoon or early evening, usually peaking between 1200 h and 2000 h (Johnson and Dropkin 1995); feeding is least intensive near dawn (Massman 1963; Grabe 1996). Larval American shad are opportunistic feeders, shifting their diet depending on availability, river location, and their size (Leim 1924; Maxfield 1953; Walburg 1956; Levesque and Reed 1972; Marcy 1976).

Researchers have also attempted to determine if the patchiness of planktonic prey has any effect on cohort survival. Letcher and Rice (1997) found that increasing levels of patchiness enhances survival when productivity or average prey density is low, but will reduce cohort survival when productivity is high. Thus, except when average prey densities of plankton are particularly high, prey patchiness may be a requirement for survival of fish larvae (Letcher and Rice 1997).

Egg and larval competition and predation

American shad eggs and larvae are preyed upon primarily by American eels (*Anguilla rostrata*) and striped bass (*Morone saxatilis*) (Mansueti and Kolb 1953; Walburg and Nichols 1967; Facey et al. 1986), although they may be preyed upon by any fish that is large enough to consume them (McPhee 2002). According to Johnson and Ringler (1998), American shad larvae that were stocked in the Susquehanna River, Pennsylvania, experienced the lowest percentage mortality at releases of 400,000 to 700,00 larvae. A high rate of larval mortality at releases up to 400,000 may have been due to depensatory mechanisms, and releases above 700,000 may have resulted in increased predator aggregation at the site. Although some individual predators consumed up to 900 American shad larvae, mortality of larvae at the stocking site was usually less than 2% (an insignificant source of mortality) (Johnson and Ringler 1998).

Eggs, larvae, and contaminants

Bradford et al. (1968) found that the lethal dose (LD_{50}) of sulfates for American shad eggs is >1000 mg/L at 15.5° C. The LD_{50} of iron for eggs is greater than 40 mg/L between pH 5.5 and 7.2 (Bradford et al. 1968). American shad eggs that are exposed to zinc and lead concentrations of 0.03 and 0.01 mg/L experience high mortality rates within 36 hours (Meade 1976). In addition, when water hardness is low (i.e., 12 mg/L), the toxicity of the zinc and lead are intensified (Klauda et al. 1991).

Part C. American Shad Juvenile Riverine/Estuarine Habitat

Geographical and temporal movement patterns

American shad larvae are transformed into juveniles 3 to 5 weeks after hatching at around 28 mm total length (TL) (Jones et al. 1978; Crecco and Blake 1983; Klauda et al. 1991; McCormick et al. 1996); they disperse at, or downstream of, the spawning grounds, where they spend their first summer in the lower portion of the same river. While most young American shad use freshwater nursery reaches (McCormick et al. 1996), it is thought that their early ability to hypo-osmoregulate allows them to utilize brackish nursery areas during years of high juvenile abundance (Crecco et al. 1983). Juveniles are typically 7 to 15 cm in length before they leave the river and enter the ocean (Talbot and Sykes 1958). For example, in the Hudson River, juvenile American shad and blueback herring were found inshore during the day, while alewives predominated inshore at night (McFadden et al. 1978; Dey and Baumann 1978). Additionally, American shad juveniles use the headpond of the Annapolis River, Nova Scotia, as a nursery area, which has surface water salinities of 25 to 30‰; they were observed remaining in the offshore region of the estuary for almost a month before the correct cues triggered emigration (Stokesbury and Dadswell 1989). Farther south, O'Donnell (2000) found that juvenile American shad in the Connecticut River began their seaward emigration at approximately 80 days post-hatch.

In addition, juvenile American shad may demonstrate temporal and latitudinal migration trends. It seems that juveniles in northern rivers emigrate seaward first, and those from southern rivers emigrate progressively later in the year (Leggett 1977a). For example, downstream emigration peaks at night (i.e., at 1800-2300 hours) (O'Leary and Kynard 1986; Stokesbury and Dadswell 1989) in September and October in the Connecticut River, late October in the Hudson River (Schmidt et al. 1988), and late October through late November in the Upper Delaware River and Chesapeake Bay (Krauthamer and Richkus 1987) and the Cape Fear River, North Carolina (Fischer 1980). Interestingly, some researchers (Chittenden 1969; Limburg 1996; O'Donnell 2000) found evidence that juvenile emigration was already underway by mid-summer, indicating that movement may be triggered by cues other than declining fall temperatures.

The combination of factors that trigger juvenile American shad emigration is uncertain, but some researchers suggest that decreased water temperatures, reduced water flow, or a combination of both during autumn appear to be key factors (Sykes and Lehman 1957; Walburg and Nichols 1967; Moss 1970). In the Susquehanna River, an increase in river flow from October through November may actually help push juveniles downstream (R. St. Pierre, U.S. Fish and Wildlife Service, personal communication). Miller et al. (1973) suggest that water temperature is more important than all other factors, because it directly affects the juvenile American shad. The lower lethal temperature limit that triggers the final movement of juveniles from fresh water is approximately 4 to 6°C (Chittenden 1969; Marcy 1976). In addition, Zydlewski and McCormick (1997a) observed changes in osmoregulatory physiology in migrating juvenile American shad, and concluded that these changes were part of a suite of physiological alterations that occur at the time of migration. While these changes are strongly affected by temperature, researchers suggest that other environmental and/or ontogenetic factors may have an influence on timing of migration (Zydlewski and McCormick 1997a).

Another migration theory deals with the age and growth of juvenile American shad. Limburg (1996) suggested that at the population level, temperature may provide the stimulus for fish to emigrate, or it may be a gradual process that is cued by size of fish, with early cohorts leaving first. Several researchers (Chittenden 1969; Miller et al. 1973; Limburg 1996; O'Donnell 2000) have observed younger, smaller young-of-the-year American shad in upstream reaches, while older and larger individuals within the same age cohorts are found downstream earlier in the season. This apparent behavior has lead researchers to hypothesize that as American shad grow and age, they move downstream (Chittenden 1969; Miller et al. 1973; Limburg 1996; O'Donnell 2000). Similarly, both Chittenden (1969) and Marcy (1976) suggest that factors associated with size appear to initiate the earlier stages of seaward emigration.

In contrast, Stokesbury and Dadswell (1989) suggest that size at emigration may not be the important factor that triggers migration, but that environmental stress may reach a point where seaward movement is necessary regardless of a critical size. O'Leary and Kynard (1986) and Stokesbury and Dadswell (1989) found that American shad movement typically occurred during quarter to new moon periods when water temperatures dropped below 19°C and 12°C, respectively. In these cases, decreasing water temperatures and the new moon phase, which provided dark nights, were considered to be more important in providing cues for emigration than increased river flow.

Habitat Type	Location	Citation
sound	Long Island	Savoy 1993
offshore estuary	New Jersey	Milstein 1981; Cameron and Pritchard 1963
brackish/ freshwater	Potomac River	Hammer 1942
estuary	Neuse River, NC	Holland and Yelverton 1973

Table 2-7. Overwintering habitats for juvenile American shad along the Atlantic coast

Following downstream migration in late fall, juvenile American shad may spend their first year near the mouths of streams, in estuaries, or in other nearshore waters (Hildebrand 1963; Colette and Klein-MacPhee 2002), or they may move to deeper, higher salinity areas, such as in portions of the lower Chesapeake Bay (Table 2-7; Hildebrand and Schroeder 1928). In their southern range, some juveniles may stay in the river for up to one full year (Williams and Bruger 1972). In South Carolina, juvenile American shad were found predominantly in deeper, channel habitats of estuarine systems, during fall and winter. Small crustaceans preyed upon by American shad are generally abundant near the bottom in these areas (McCord 2003).

Juveniles and the saltwater interface

Early studies of juvenile American shad describe a variety of responses to changes in salinity. When accompanied by temperature changes, juveniles generally adapt to abrupt transfers from freshwater to saltwater, but high mortality results when transferred from saltwater to freshwater (Tagatz 1961). For example, Tagatz (1961) observed 60% mortality for juveniles in isothermal transfers (21°C) from freshwater to 30 ppt saltwater; however, no individuals

survived transfers from freshwater (21.1°C) to 33 ppt saltwater (7.2 to 12.8°C). Freshwater transfers to 15 ppt in association with a temperature decrease less than 4°C also resulted in high mortalities (30 to 50%). Conversely, at temperature increases greater than 14°C, all juvenile American shad survived abrupt transfers from saltwater (15 ppt and 33 ppt) to freshwater (Tagatz 1961).

In another study, Chittenden (1973b) observed 0% mortality in isothermal transfers (17°C) from freshwater or 5 ppt to 32 ppt seawater. Additionally, juveniles transferred from 30 ppt seawater to freshwater suffered 100% mortality, but no mortalities resulted when they were transferred from 5 ppt to freshwater. In general, American shad are considered to be capable of surviving a wide range of salinities at early life stages, especially if salinity changes are gradual (Chittenden 1969).

Experiments conducted on American shad and other anadromous fish (Rounsefell and Everhart 1953; Houston 1957; Tagatz 1961; Zydlewski and McCormick 1997a, 1997b) have demonstrated that most fish undergo physiological changes before emigrating to saltwater. This ability to adapt to changes in salinity occurs at the onset of metamorphosis for American shad, between 26 and 45 days post-hatch. Zydlewski and McCormick (1997b) noted that the ability to osmoregulate in full-strength seawater is an important factor that limits American shad early life history stages to freshwater and low-salinity estuaries. The researchers suggested that a decrease and subsequent loss of hyper-osmoregulatory ability may serve as a proximate cue for juveniles to begin their downstream migration (Zydlewski and McCormick 1997b).

Juvenile substrate associations

Although juvenile American shad are often most abundant where boulder, cobble, gravel, and sand are present (Walburg and Nichols 1967; Odom 1997), substrate type is not considered to be a critical factor in nursery areas (Krauthamer and Richkus 1987). Ross et al. (1997) found no overall effect of habitat type on juvenile American shad relative abundance in the upper Delaware River, indicating that juveniles use a wide variety of habitat types to their advantage in many nursery areas. These researchers suggest that in contrast to earlier life stages and spawning adults, pre-migratory juveniles may be habitat generalists; however, a positive relationship was found between abundance of juvenile American shad and percent of SAV cover in SAV habitats only. In addition, Odom (1997) found that juvenile American shad favored riffle/run habitat in the James River, especially areas with extensive beds of water stargrass (*Heteranthera dubia*). These areas provided flow-boundary feeding stations where juveniles could feed on drifting macroinvertebrates while reducing their energy costs (Odom 1997).

Estuarine productivity is linked to freshwater detrital nutrient input to the estuary (Biggs and Flemer 1972; Hobbie et al. 1973; Saila 1973; Day et al.1975) and detritus production in the salt marsh (Teal 1962; Odum and Heald 1973; Reimhold et al. 1973; Stevenson et al. 1975). Based on the assumption that the amount of submerged and emergent vegetation will be a qualitative estimate of the estuary's secondary productivity, and therefore, food availability (zooplankton) to juvenile American shad, Stier and Crance (1985) suggest that estuarine habitat with 50% or more vegetation coverage is optimal.

It is important to note that, although no link has been made between the presence of SAV and abundance of alosines, there seems to be a general agreement that there is a correlation

between water quality and alosine abundance (B. Sadzinski, Maryland Department of Natural Resources, personal communication). Abundance of SAV is often used as an indirect measure of water quality, with factors such as available light (Livingston et al. 1998), salinity, temperature, water depth, tidal range, grazers, suitable sediment quality, sediment nutrients, wave action, current velocity, and chemical contaminants controlling the distribution of underwater grasses (Koch 2001). Maryland has made it a priority to increase the amount of SAV within the Chesapeake Bay watershed in order to improve water quality. According to B. Sadzinski (Maryland Department of Natural Resources, personal communication), if SAV in a given area increases, this can be used as an indicator of improved water quality, which in turn, will likely benefit alosine species.

Juvenile depth associations

Juveniles have been observed at depths ranging from 0.9 to 4.9 m in the Connecticut River (Marcy 1976); however, abundance is related to the distance upstream and not to depth (MacKenzie et al. 1985). In the Connecticut River, juveniles were caught primarily at the bottom during the day (87%) and all were caught at the surface at night (Marcy 1976). Chittenden (1969) observed juveniles in the Delaware River most often in deeper, non-tidal pools away from the shoreline during daylight hours; after sunset juveniles scattered and were found at all depths (Miller et al. 1973).

Although data was sparse for depth optima for juveniles, Stier and Crance (1985) developed a suitability index based on input provided by research scientists. They suggest that for all life history stages, including juveniles, the optimum range for river depth is between 1.5 and 6.1 m. Depths less than 0.46 m and greater than 15.24 m are unsuitable habitat according to the model.

Juvenile water temperature

Characterization	Temperature (^{o}C)	Location	Citation
Optimal range	15.5 - 23.9	N/A	Crance 1985
Optimal range	10 - 25	N/A	Stier and Crance 1985
Range	10 - 30	Connecticut River	Marcy et al. 1972
Critical maximum	34 - 35	Neuse River, NC	Horton and Bridges 1973
Maximum tolerance	35	N/A	Stier and Crance 1985
Minimum preference	8	N/A	MacKenzie et al. 1985
Minimum tolerance	3	N/A	Stier and Crance 1985
Minimum tolerance	31.6	N/A	Ecological Analysts Inc. 1978
Begin migration	19	Connecticut River	Leggett 1976; O'Leary and Kynard 1986

Characterization	Temperature (^{o}C)	Location	Citation
Begin migration	23 - 26	Connecticut River	Marcy 1976
Begin migration	18.3	Connecticut River	Watson 1970
Peak migration	16	Connecticut River	Leggett and Whitney 1972; O'Leary and Kynard 1986
Peak migration	15.1	North Carolina	Neves and Depres 1979; Boreman 1981
End migration	8.3	Delaware River	Chittenden and Westman 1967
End migration	8.3	Chesapeake Bay	Chesapeake Bay Program 1988

Table 2-8. Temperature tolerances, preferences, and cues for juvenile American shad

Juvenile American shad demonstrate some variability in temperature tolerances and preferences among river systems (Table 2-8). Leim (1924) found that juveniles captured in the Shubenacadie River, Canada, were usually found where temperatures tended to be the highest compared to other regions of the river. Additionally, temperature appears to have a significant impact on growth of juvenile American shad. Limburg (1996) found that juveniles in the laboratory had higher initial growth rates at 28.5°C than individuals reared at lower temperatures. O'Donnell (2000) concluded that it may be advantageous for eggs to hatch later in the year because temperatures are higher and growth rates are faster; however, competition and predation rates are also higher.

Juvenile American shad do not appear to be as tolerant to temperature changes as eggs of the same species. In fact, juveniles are sensitive to water temperature changes, and actively avoid temperature extremes, if possible. Laboratory tests suggest that juveniles can tolerate temperature increases between 1° and 4°C above ambient temperature, but beyond that they will avoid changes if given a choice (Moss 1970). For example, juveniles acclimated to 25° C suffered a 100% mortality rate when the temperature was decreased to 15°C. There was also a 100% mortality rate for juveniles acclimated to 15°C and then subjected to temperatures less than 5°C. Finally, no survival was reported for juveniles acclimated to 5°C and then exposed to 1°C (PSE&G 1982).

Juvenile dissolved oxygen associations

Minimum dissolved oxygen values have a more adverse effect upon fish than average dissolved oxygen values; therefore, minimum dissolved oxygen criteria have been recommended. Dissolved oxygen concentrations less than 5.0 mg/L are considered sub-lethal to juvenile American shad (Miller et al. 1982). As with spawning areas, Bilkovic (2000) assigned a value of greater than 5.0 mg/L dissolved oxygen as optimal for nursery areas.

Seemingly healthy juvenile American shad have been collected in the Hudson River, New York, where dissolved oxygen concentrations were 4 to 5 mg/L (Burdick 1954). Similarly, in headponds above hydroelectric dams on the St. John River, New Brunswick, dissolved oxygen must be at least 4 to 5 mg/L for migrating juveniles to pass through (Jessop 1975). In the

Delaware River, dissolved oxygen concentrations less than 3.0 mg/L blocked juvenile migration, and concentrations below 2.0 mg/L were lethal. Emigrating juveniles have historically arrived at the upper tidal section of the Delaware River by mid-October, but do not continue further seaward movement until November or December, when the pollution/low oxygen conditions dissipate (Miller et al. 1982).

Under laboratory conditions, juvenile American shad did not lose equilibrium until dissolved oxygen decreased to 2.5 to 3.5 mg/L (Chittenden 1969, 1973a). Juveniles have been reported to survive brief exposure to dissolved oxygen concentrations of as little as 0.5 mg/L, but survived only if greater than 3 mg/L was available immediately thereafter (Dorfman and Westman 1970).

Juvenile pH associations

Areas that are poorly buffered (low alkalinity) and subject to episodic or chronic acidification may provide less suitable nursery habitat than areas that have higher alkalinities and are less subject to episodic or chronic acidification (Klauda et al. 1991). Once juvenile American shad move downstream to brackish areas with a higher buffering capacity, they may be less impacted by changes in pH (Klauda 1989).

Juvenile water velocity/flow

Ideal water velocity rates are thought to range between 0.06 to 0.75 m/s for the juvenile non-migratory stage of American shad (Klauda et al. 1991). The rate of water velocity is also critical for fish migrating downstream that pass over spillways (MacKenzie et al. 1985). Furthermore, it has been suggested that water flow may serve to orient emigrating juveniles in the downstream direction. Studies conducted on American shad in the St. Johns River, Florida, led researchers to speculate that the lack of water flow as a result of low water levels could result in the inability of juveniles to find their way downstream (Williams and Bruger 1972).

Juvenile suspended solid associations

Ross et al. (1997) suggest that optimal turbidity values for premigratory American shad juveniles in tributaries is between 0.75 and 2.2 NTU. While preliminary, these results could be cautiously applied to other river systems, but consideration should be given to the range and diversity of habitat types in the river system under study before applying the models.

Juvenile feeding behavior

Juvenile American shad begin feeding in freshwater and continue into the estuarine environment. They favor zooplankton over phytoplankton (Maxfield 1953; Walburg 1956), and in general, have a wider selection of prey taxa than larvae due to their increased size and the estuaries' higher diversity. Long, closely-spaced gill rakers enable juveniles to effectively filter plankton from the water column during respiratory movements (Leim 1924). Juvenile American shad are opportunistic feeders, whose freshwater diet includes copepods, crustacean zooplankton, cladocerans, aquatic insect larvae, and adult aquatic and terrestrial insects (Leim

1924; Maxfield 1953; Massmann 1963; Levesque and Reed 1972; Marcy 1976). After juveniles leave coastal rivers and estuaries for nearshore waters, they may prey on some fish, such as smelt, sand lance, silver hake, bay anchovy, striped anchovy, and mosquitofish (Leidy 1868; Bowman et al. 2000).

Although juveniles obtain most of their food from the water column (ASMFC 1999), many of the crustaceans that juveniles prey upon are benthic (Krauthamer and Richkus 1987). Leim (1924) speculated that although American shad obtain a minor amount of food near the bottom of the water column, they do not pick it off the bottom, but rather capture items as they are carried up into the water column a short distance by tidal currents (including mollusks).

Walburg (1956) found that juvenile American shad fed primarily on suitable organisms that were readily available. In contrast, Ross et al. (1997) found that juveniles in SAV habitat fed principally on chironomids, while those feeding in tributaries consumed terrestrial insects almost exclusively, despite the fact that insects were less available than other food sources. Researchers did not attribute the differences to developmental limitations, but concluded that there were true feeding differences between habitats. Other studies have noted different selection of organisms along the same river, but at different locations, such as above a dam (Levesque and Reed 1972) or downstream of a dam (Domermuth and Reed 1980).

Feeding of juvenile American shad may also differ along a stream gradient. In waters of Virginia, Massman (1963) found that juvenile American shad upstream consume more food than juveniles that remain downstream near their spawning grounds. The upstream sections of the river have a higher shoreline to open water ratio that may provide a more abundant source of terrestrial insects, a favored prey item (Massman 1963; Levesque and Reed 1972), while the downstream sections contain more autochthonously-derived prey. In contrast, the lower reach of the Hudson River appears to be more productive (as a function of primary productivity and respiration rates) than upper and middle reaches (Sirois and Fredrick 1978; Howarth et al. 1992). This greater productivity may lead to higher fish production in the lower estuary, as well as a higher relative condition of downriver juvenile American shad earlier in the season, compared to upriver and midriver fish (Limburg 1994).

Juvenile American shad also demonstrate diel feeding patterns. Johnson and Dropkin (1995) found that juveniles increase feeding intensity as the day progresses, achieving a maximum feeding rate at 2000 h. Similarly, juveniles in the Mattaponi and Pamunkey rivers in Virginia, feed during the day with stomachs reaching maximum fullness by early evening (Massman 1963).

In addition, at least one non-native species has proven to have an impact on young-of-the-year American shad. In the Hudson River, there is strong evidence that zebra mussel colonization has reduced the planktonic forage base of the species (Waldman and Limburg 2003).

Juvenile competition and predation

Juveniles in freshwater may be preyed upon by American eel, bluefish, weakfish, striped bass, birds, and aquatic mammals (Mansueti and Kolb 1953; Walburg and Nichols 1967; Facey et al. 1986).

With regard to inter-species competition, differences among alosine species in terms of distribution, diel activity patterns, and feeding habits are evident in many systems, and are likely mechanisms that may reduce competition between juveniles of the different species (Schmidt et al. 1988). For example, several researchers have noted that larger American shad (Chittenden 1969; Marcy 1976; Schmidt et al. 1988) and alewife (Loesch et al. 1982; Schmidt et al. 1988) move downstream first, which helps to segregate size classes of the two species.

Secondly, there is the idea of diel, inshore-offshore segregation. Both American shad and blueback herring juveniles occur in shallow nearshore waters during the day. However, competition for prey between American shad and blueback herring is often reduced by: 1) more opportunistic feeding by American shad, 2) differential selection for cladoceran prey, and 3) higher utilization of copepods by blueback herring (Domermuth and Reed 1980). American shad feed most often in the upper water column, the air-water interface (Loesch et al. 1982), and even leap from the water (Massman 1963), feeding on *Chironomidae* larvae, *Formicidae*, and *Cladocera*; they are highly selective for terrestrial insects (Davis and Cheek 1966; Levesque and Reed 1972). Juvenile bluebacks are more planktivorous, feeding on copepods, larval dipterans, and *Cladocera* (Hirschfield et al. 1966), but not the same cladoceran families that alewife feed upon (Domermuth and Reed 1980).

Juveniles and contaminants

Tagatz (1961) found that the 48 h lethal concentrations (LC_{50}) for juvenile American shad range from 2,417 to 91,167 mg/L for gasoline, No. 2 diesel fuel, and bunker oil. The effects of gasoline and diesel fuel are exacerbated when the dissolved oxygen concentration is simultaneously reduced. Gasoline concentrations of 68 mg/L at 21 to 23°C resulted in a lethal time (LT_{50}) of 50 minutes for juveniles when dissolved oxygen was reduced to 2.6 to 3.2 mg/L. Additionally, juveniles that were exposed to 84 mg/L of diesel fuel at 21 to 23°C with dissolved oxygen between 1.9 and 3.1 mg/L experienced an LT_{50} of 270 minutes (Tagatz 1961).

Part D. American Shad Late Stage Juvenile and Adult Marine Habitat

Geographical and temporal patterns at sea

American shad typically live 5 to 7 years (Leggett 1969) and remain in the ocean for 2 to 6 years before becoming sexually mature, at which point they return to their natal rivers to spawn (Talbot and Sykes 1958; Walburg and Nichols 1967). Both sexes begin to mature at 2 years, with males maturing on average in 4.3 years and females maturing on average in 4.6 years. Fish north of Cape Hatteras are iteroparous and will return to rivers to spawn when temperatures are suitable (Leggett 1969).

Results from 50 years of tagging indicate that discrete, widely separated aggregations of juvenile and adult American shad occur at sea (Talbot and Sykes 1958; Leggett 1977a, 1977b; Dadswell et al. 1987; Melvin et al. 1992). These aggregations are a heterogeneous mixture of individuals from many river systems (Dadswell et al. 1987); it is unknown if American shad from all river systems along the east coast intermingle throughout the entire year (Neves and Depres 1979). Populations that return to rivers to spawn are a relatively homogeneous group (Dadswell et al. 1987), and fish from all river systems can be found entering coastal waters as far south as North Carolina in the winter and spring (Neves and Depres 1979).

Dadswell et al. (1987) presented the following seasonal movement timeline for American shad:

1) *January & February* –found offshore from Florida to Nova Scotia; spawning inshore from Florida to South Carolina;

2) *March & April* –moving onshore and northward from the Mid-Atlantic Bight to Nova Scotia; spawning from North Carolina to the Bay of Fundy;

3) *Late June* – concentrated in the inner Bay of Fundy, inner Gulf of St. Lawrence, Gulf of Maine, and off Newfoundland and Labrador; spawning fish are still upstream from Delaware River to St. Lawrence River;

4) *Autumn* –American shad leaving the St. Lawrence estuary are captured across the southern Gulf of St. Lawrence, while fish leaving the Bay of Fundy are found from Maine to Long Island; some individuals already migrated as far south as Georgia and Florida.

Through an analysis of tag returns, occurrence records, and trawl survey data, Dadswell et al. (1987) found that there are three primary offshore areas where aggregations of American shad overwinter: 1) off the Scotian Shelf/Bay of Fundy, 2) in the Mid-Atlantic Bight, and 3) off the Florida coast. It appears that the majority of American shad that overwinter along the Scotian Shelf spawn in rivers in Canada and New England (Vladykov 1936; Melvin et al. 1985). Fish aggregations that overwinter off the mid-Atlantic coast (from Maryland to North Carolina) are comprised of populations that spawn in rivers from Georgia to Quebec (Talbot and Sykes 1958; Miller et al. 1982; Dadswell et al. 1987).

The regional composition of American shad aggregations overwintering off the Florida coast is unknown. Leggett (1977a) proposed the following estimates for timing and origin of southern migrations for overwintering off Florida based on migration rates and an average departure date of October 1 from the Gulf of Maine/Bay of Fundy region: Rhode Island/Long

Island coast in mid-to-late October, off Delaware Bay in early November, and off the coast of North Carolina, Georgia, and Florida in early December. Additionally, early migration studies of American shad found that during mild winters, small aggregations sometimes enter the sounds of North Carolina during November and December, but disappear if the weather becomes cold (Talbot and Sykes 1958).

Most American shad populations that overwinter off the mid-Atlantic coast (between 36° to 40°N) migrate shoreward in the winter and early spring. Pre-spawning adults homing to rivers in the south Atlantic migrate shoreward north of Cape Hatteras, North Carolina, then head south along the coast to their natal rivers. The proximity of the Gulf Stream to North Carolina provides a narrow migration corridor at Cape Hatteras through which individuals may maintain travel in the preferred temperature range of 3 to 15°C. Although pre-spawning adults are not required to follow a coastal route to North Atlantic rivers because temperatures in the Mid-Atlantic Bight are generally well within a tolerable range in the spring, tag returns indicate that most individuals likely enter coastal waters in the lower mid-Atlantic region, and then migrate north along the coast (Dadswell et al. 1987).

South of Cape Cod, pre-spawning American shad migrate close to shore (Leggett and Whitney 1972), but north of that point the migration corridor is less clear (Dadswell et al. 1987). Pre-spawning adults may detour into estuaries during their coastal migration; however, the timing and duration of the stay is unknown (Neves and Depres 1979). Although poorly documented, immature American shad (age 1+) may also enter estuaries and accompany adults to the spawning grounds, more than 150 km upstream (Limburg 1995, 1998). Additionally, non-spawning adults have been recorded in brackish estuaries (Hildebrand 1963; Gabriel et al. 1976).

Dadswell et al. (1987) found three primary offshore summer aggregations of American shad: 1) Bay of Fundy/Gulf of Maine, 2) St. Lawrence estuary, and 3) off the coast of Newfound and Labrador. Neves and Depres (1979) also found distinct summer aggregations on Georges Bank and south of Nantucket Shoals. Furthermore, American shad from all river systems, including those from south Atlantic rivers, have been collected at the Gulf of Maine feeding grounds during the summer (Neves and Depres 1979). While individuals from north Atlantic rivers are most abundant in the Bay of Fundy in the early summer, the appearance of American shad from the southern range does not peak until mid-summer (Melvin 1984; Dadswell et al. 1987). These migrating groups are a mixture of juveniles, immature sub-adults, and spent and resting adults that originate from rivers along the entire East Coast (Dadswell et al. 1983). Since there are very few repeat spawners in the southern range, the majority (76%) of American shad that migrate to the Bay of Fundy from areas south of Cape Lookout, North Carolina, are juveniles (Melvin et al. 1992).

American shad enter the Bay of Fundy in early summer and move throughout the inner Bay of Fundy for four months in a counterclockwise direction with the residual current (Dadswell et al. 1987). As water temperatures decline in the fall, American shad begin moving through the Gulf of Maine, and continue to their offshore wintering grounds. This species has been captured in late fall and winter 80 to 95 km offshore of eastern Nova Scotia (Vladykov 1936), 65 to 80 km off the coast of Maine, 40 to 145 km off southern New England, and 175 km from the nearest land of southern Georges Bank (Colette and Klein-MacPhee 2002; Dadswell et al. 1987).

Salinity associations at sea

During their residence in the open ocean, American shad sub-adults and adults will live in seawater that is approximately 33 ppt. During coastal migration periods, pre-spawning adults may detour into estuaries where water is more brackish, but the timing and duration of the stay is unknown (Neves and Depres 1979).

Depth associations at sea

While it is known that adult American shad move offshore to deeper waters during the fall and early winter, information regarding preferred depths is lacking. American shad have been found throughout a broad depth range in the ocean, from surface waters to depths of 340 m (Walburg and Nichols 1967; Facey and Van Den Avyle 1986). Alternatively, catch data analyses showed that this species has been caught at depths ranging from surface waters to 220 m (Walburg and Nichols 1967), but are most commonly found at intermediate depths of 50 to 100 m (Neves and Depres 1979). Seasonal migrations are thought to occur mainly in surface waters (Neves and Depres 1979).

The summer and autumn months are a time of active feeding for American shad, and analyzing stomach contents has served as a means to infer distribution in the water column. Studies by Neves and Depres (1979) suggested that American shad follow diel movements of zooplankton, staying near the bottom during the day and dispersing in the water column at night. Other researchers (Dadswell et al. 1983) have suggested that light intensity may control depth selection by American shad. For example, American shad swim much higher in the water column in the turbid waters of Cumberland Basin, Bay of Fundy, than they do in clear coastal waters, where they are found in deeper water. Both areas are within the same surface light intensity range (Dadswell et al. 1983).

Temperature associations at sea

Early studies by Leggett and Whitney (1972) found that American shad move along the coast via a "migrational corridor" where water temperatures are between 13 and 18°C. Neves and Depres (1979) later modified the near-bottom temperature range from 3 to 15°C, with a preferred range of 7 to 13°C. These researchers also hypothesized that seasonal movements are broadly controlled by climate, and that American shad follow paths along migration corridors or oceanic paths of "preferred" isotherms. Melvin et al. (1985) and Dadswell et al. (1987) revised this theory with data indicating movement of American shad across thermal barriers. It was determined that American shad remain for extended periods in temperatures outside their "preferred" range; this species migrates rapidly between regions regardless of currents and temperatures (Melvin et al. 1985; Dadswell et al. 1987). For example, Dadswell et al. (1987) documented non-reproductive American shad migrating from wintering grounds in the Mid-Atlantic Bight through the Gulf of Maine in May-June, where a constant sub-surface temperature of 6°C prevails, to reach the Bay of Fundy by mid-summer.

Temperature change and some aspect of seasonality (i.e., day length) may initiate migratory behavior, but timing of the behavior by different individuals may be influenced by intrinsic (genetic) factors and life history stage of the individual. Chance may also play a small role in determining which direction a fish will travel, at least within a confined coastal region.

Dadswell et al. (1987) concluded that extrinsic factors related to ocean climate, seasonality, and currents may provide cues for portions of non-goal-oriented migration, while intrinsic cues and bi-coordinate navigation appear to be important during goal-oriented migration.

Suspended solid associations at sea

Due to extreme turbidity, the American shad preference zone for light intensity in summer and fall in the Bay of Fundy is limited to surface waters (2 to 10 m). Although this makes the fish more susceptible to fishing gear that operates near surface waters, these waters are highly productive sources of zooplankton. Sight-oriented planktivores may be at a disadvantage in these turbid waters, but American shad, which can use a filter-feeding mechanism, may have a competitive advantage (Dadswell et al. 1983).

Feeding behavior at sea

While offshore, American shad are primarily planktivorous, feeding on the most readily available organisms, such as copepods, mysid shrimps, ostracods, amphipods, isopods, euphausids, larval barnacles, jellyfish, small fish, and fish eggs (Willey 1923; Leim 1924; Maxfield 1953; Massmann 1963; Levesque and Reed 1972; Marcy 1976). Themelis (1986) found that in the Bay of Fundy, American shad mostly consume planktonic and epibenthic crustaceans. Differences in dominant prey items may be attributed to changing availability of zooplankton assemblages and the size of the American shad. Juveniles feed more extensively on copepods than adults and a smaller proportion of their diet is composed of large prey items such as euphausids and mysids (Themelis 1986). In earlier studies, Leim (1924) reported similar observations, with copepods decreasing in importance in the diets of American shad over 400 mm in length. Detritus has also been found in the stomachs of American shad, but it probably provides little nutritional value and is simply ingested during the course of feeding (Themelis 1986).

The Bay of Fundy is regarded as the primary summer feeding grounds for American shad, however, the entire bay does not provide optimal feeding conditions for adults. For example, although both adult and juvenile American shad feed readily in the oceanic lower Bay of Fundy, only juveniles feed to a large extent within the turbid and estuarine waters of the upper bay. This is attributed to the juvenile's ability to successfully filter smaller prey items that dominate the upper bay (Themelis 1982).

Competition and predation at sea

Once in the ocean, American shad are undoubtedly preyed upon by many species including sharks, tunas, king mackerel, bluefish, striped bass, Atlantic salmon, seals, porpoises, other marine mammals, and seabirds, given their schooling nature and lack of dorsal or opercular spines (Melvin et al. 1985; Weiss-Glanz et al. 1986).

Current laboratory research by Plachta and Popper (2003) has found that American shad can detect ultrasonic signals to at least 180 kHz, which is within the range that echolocating harbour porpoises and bottlenose dolphins use to track alosines. In this laboratory environment, American shad have been observed modifying their behavior in response to echolocation beams,

such as turning slowly away from the sound source, forming very compact groups, and displaying a quick "panic" response. Although behavior in a natural environment may be different from that observed in experimental tanks, this study suggests that American shad may have evolved a mechanism to make themselves less "conspicuous" or less easily preyed upon by echolocating odontocetes (Plachta and Popper 2003).

Section II. Significant Environmental, Temporal, and Spatial Factors Affecting Distribution of American Shad

Table 2-9. Significant environmental, temporal, and spatial factors affecting distribution of American shad. Please note that, although there may be subtle variations between systems, the following data include a broad range of values that encompass the different systems that occur along the East Coast. Where a specific range is known to exist, it will be noted. For the subadult–estuarine/oceanic environment and non-spawning adult–oceanic environment life history phases, the information is provided as a general reference, not as habitat preferences or optima. NIF = No Information Found.

Life Stage	Time of Year and Location	Depth (m)	Temperature (°C)	Salinity (ppt)	Substrate	Current Velocity (m/sec)	Dissolved Oxygen (mg/L)
Spawning Adult	Mid-November-August (south to north progression) in natal rivers and tributaries from St. Johns River, Florida to St. Lawrence River, Canada	**Tolerable:** 0.46-15.24 **Optimal:** 1.5-6.1 **Reported:** Variable	**Tolerable:** 8-26 **Optimal:** 14-24.5 **Reported:** Varies across range and may vary between years	**Tolerable:** NIF **Optimal:** NIF **Reported:** Mostly freshwater	**Tolerable:** NIF **Optimal:** NIF **Reported:** Sand, silt, gravel, boulder	**Tolerable:** NIF **Optimal:** 0.3-0.9 **Reported:** Avoid pools but prefer slow flow; velocity is an important factor	**Tolerable:** NIF **Optimal:** NIF **Reported:** Minimum 4
Egg	Mid-November-August (south to north progression) at spawning areas or slightly downstream	**Tolerable:** NIF **Optimal:** NIF **Reported:** Settle at bottom in shallow water	**Tolerable:** 8-30 **Optimal:** NIF **Reported:** Variable	**Tolerable:** NIF **Optimal:** NIF **Reported:** Variable	**Tolerable:** NIF **Optimal:** NIF **Reported:** Gravel, rubble, and sand have highest survival	**Tolerable:** NIF **Optimal:** 0.3-0.9 **Reported:** Low flow	**Tolerable:** NIF **Optimal:** NIF **Reported:** Minimum 5
Larvae	2-17 days after fertilization depending on temperature, downstream of spawning areas	**Tolerable:** 0.46-15.24 **Optimal:** 1.5-6.1 **Reported:** Surface and water column	**Tolerable:** 10-30 **Optimal:** 15-25 **Reported:** Variable	**Tolerable:** NIF **Optimal:** NIF **Reported:** Variable	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** 0.3-0.9 **Reported:** Low flow	**Tolerable:** NIF **Optimal:** NIF **Reported:** Minimum 5

Life Stage	Time of Year and Location	Depth (m)	Temperature (°C)	Salinity (ppt)	Substrate	Current Velocity (m/sec)	Dissolved Oxygen (mg/L)
Early Juvenile – Riverine Environment	3-5 weeks after hatching Downstream of spawning areas as far as brackish waters	**Tolerable:** 0.46-15.24 **Optimal:** 1.5-6.1 **Reported:** Variable	**Tolerable:** 3-35 **Optimal:** 10-25 **Reported:** Variable; growth higher at higher temps	**Tolerable:** 0-30 **Optimal:** NIF **Reported:** Gradual change well tolerated	**Tolerable:** NIF **Optimal:** Possibly areas w/50%+ SAV **Reported:** Boulder, cobble, gravel, sand, SAV	**Tolerable:** NIF **Optimal:** 0.1-0.8 **Reported:** Moderate needed for migration	**Tolerable:** NIF **Optimal:** NIF **Reported:** Minimum 5
Subadult & Non-spawning Adult – Estuarine / Oceanic Environment	2-6 years after hatching; 1) Overwinter offshore of Florida, the Mid-Atlantic Bight, and Nova Scotia 2) Spring – migration route is unknown 3) Late June – inner Bay of Fundy, inner Gulf of St. Lawrence, Gulf of Maine, and Newfoundland and Labrador 4) Autumn – moving offshore	**Tolerable:** Surface waters to 340 m **Optimal:** 50-100 m **Reported:** Variable; possible diel migrations with zooplankton	**Tolerable:** Variable **Optimal:** 7-13 **Reported:** Generally travel in preferred isotherm	**Tolerable:** NIF **Optimal:** NIF **Reported:** Brackish to saltwater	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF

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Chapter 3

HICKORY SHAD

(*Alosa mediocris*)

Section I. Hickory Shad Description of Habitat

Hickory Shad General Habitat Description and Introduction

Hickory shad (*Alosa mediocris*) are anadromous fish that spend most of their adult lives at sea, entering brackish and freshwater only to spawn (Colette and Klein-MacPhee 2002). Little is known about the life history and specific habitat requirements of this species. However, coastal migrations and habitat requirements are thought to be similar to that of other alosine species, particularly American shad (Klauda et al. 1991). Very few spawning studies have been conducted in part due to a general lack of interest in this species relative to other alosines (Klauda et al. 1991).

Historically, hickory shad abundance has been lower than other alosine species in many areas (Atran et al. 1983; Speir 1987). The historical range of hickory shad is thought to have extended as far north as the Gulf of Maine and possibly to Campobello Island, New Brunswick (Hildebrand 1963). The current northern boundary of hickory shad is Cape Cod, Massachusetts (Batsavage and Rulifson 1998), with the highest abundances occurring from New York southward. According to Klauda et al. (1991), spawning does not frequently occur north of Maryland. Hickory shad are reported to occur as far south as central Florida (Hildebrand 1963; Williams et al. 1975; McBride 2000). Waters south of Cape Canaveral, Florida, are unsuitable for hickory shad due to semi-tropical water temperatures (Williams et al. 1975).

Hickory shad have only supported minor commercial fisheries because the bony meat is considered to be inferior to American shad (Whitehead 1985). However, some consider hickory shad roe to be more delectable than the roe of any of the other river herrings (Nichols 1959). Furthermore, adult hickory shad are highly sought after by sport fishermen when they ascend rivers and tributaries during their spawning run (Mansueti 1962; Pate 1972). Although hickory shad populations have not been adequately monitored, there is information indicating that some stocks are healthy (Street 1970; Batsavage and Rulifson 1998; ASMFC 1999). Since 1989, the Albemarle Sound, North Carolina, population of hickory shad has experienced a surge in numbers, which supports a growing sport fishery on the Roanoke River and increased commercial fishing in Albemarle Sound. A short life span and low fecundity, however, makes this North Carolina population vulnerable to overharvest (Batsavage and Rulifson 1998). In contrast, hickory shad have been found to be highly fecund in other areas. For example, egg production was estimated to be as high as 509,749 eggs per female in the Altamaha River, Georgia (Street 1970).

Since the mid-1990s, hickory shad numbers have increased in the upper Chesapeake Bay and its tributaries (ASMFC 1999), including the lower Susquehanna, Potomac near Washington, D.C., upper Rappahannock, and James rivers (R. St. Pierre, U.S. Fish and Wildlife Service, personal communication). Some landings data also support the idea that hickory shad populations are thriving. The National Marine Fisheries Service (NMFS) estimated that 5.6 metric tons of hickory shad were landed in 1990, and by 1999, estimated landings dramatically increased to 61.9 metric tons (Waldman and Limburg 2003).

Part A. Hickory Shad Spawning Habitat

Geographical and temporal patterns of migration

Little is known about hickory shad behavior or utilization of riverine or marine habitats (Colette and Klein-MacPhee 2002). It is assumed that female hickory shad broadcast their eggs into the water between dusk and midnight where one or more males fertilize them; this behavior is similar to the spawning behavior of American shad (Mansueti 1962; Jones et al. 1978). Hickory shad are known to be repeat spawners, with individuals spawning an average of three to five times before dying (Schaeffer 1976). Unlike American shad, there is no progressive increase in spawning frequency from south to north. Most river systems have 70 to 80% repeat spawners, although there are exceptions (Street and Adams 1969; Loesch et al. 1979; Rulifson et al. 1982; Richkus and DiNardo 1984). Data collected from Maryland rivers indicated that 72% of females and 62% of males had previously spawned (B. M. Richardson, Maryland Department of Natural Resources, personal communication). In sharp contrast, Sholar (1977) found that in the Cape Fear River, North Carolina, only 19% of males and 9% of females were repeat spawners.

The age distribution of adult hickory shad in coastal rivers from Florida to North Carolina ranges from two to eight years (Rulifson et al. 1982). Eighty percent of males in the Octoraro Creek, Maryland, were sexually mature at age 2 (Schaeffer 1976). Data collected from a group of Maryland rivers found that 50% of males and 36% of females were sexually mature at age 2; by age 3, 89% of males and 90% of females had spawned (B. M. Richardson, Maryland Department of Natural Resources, personal communication). Further south, in the Altamaha River, Georgia, 75% of females and 49% of males were sexually mature by age 2 (Street and Adams 1969). In general, the majority of females are likely to become sexually mature at least one year later than males (Klauda et al. 1991; Batsavage and Rulifson 1998).

Hickory shad ascend coastal rivers during spring migration. Although it is assumed that these fish return to their natal rivers to spawn, there is no documented evidence of this behavior (Batsavage and Rulifson 1998). Hickory shad distribution in the riverine environment is similar to that of American shad (Rulifson et al. 1982). In North Carolina, the freshwater reaches of coastal rivers are the major spawning sites for hickory shad. In the Roanoke River, eggs have been collected during April and early May from the main channel near Weldon, North Carolina (Sparks 1998; Harris and Hightower 2007), and larvae have been collected farther downstream (Walsh et al. 2005). In the Neuse River, North Carolina, Pate (1972) detected spawning in flooded swamps and sloughs off channels of tributary creeks, but not in the mainstem river. However, Burdick and Hightower (2006) detected spawning in both mainstem Neuse River and tributary sites. In Georgia, hickory shad apparently spawn in flooded areas off the Altamaha River, and not in the mainstem of the upper reaches (Adams 1970). Major spawning sites in Virginia have been discovered in mainstem rivers at the fall line, further downstream, and in tributaries (Davis et al. 1970). Mansueti (1962) found that hickory shad spawned approximately 6 to 10 km (3.7 to 6.2 miles) upriver of major spawning sites of American shad in the mainstem of the Patuxent River, Maryland. In contrast, hickory shad in the St. Johns River, Florida, did not migrate as far upstream as American shad (Moody 1961). Compared to American shad and striped bass, hickory shad in the Neuse River basin tended to spawn further downstream and made greater use of tributaries (Burdick and Hightower 2006).

Adult hickory shad can be found in the St. Johns River, Florida, as early as December or possibly even November (McBride 2000), but may be absent by late January to mid-February (Williams et al. 1975) or early March (McBride 2005). Spawning in the Santee and Cooper rivers, South Carolina, may occur between early March through mid-May (Bulak and Curtis 1979). In the Chesapeake Bay, spawning may begin in early April (Mansueti and Hardy 1967), and typically peaks in early May (Mansueti 1962). However, spawning may occur as late as June in freshwaters of Virginia (Davis et al. 1970). Furthermore, a weaker second run of spawners may also migrate later through the Chesapeake Bay (Hildebrand and Schroeder 1928). It is unknown if the hickory shad that spawn during the fall run also participate in the spring run (Schaeffer 1976).

Large variations in the size of young hickory shad have been reported at spawning sites. This has lead researchers to hypothesize that this species has a protracted spawning period, where small amounts of eggs are released over a long period of time (Mansueti 1962; DesFosse et al. 1994). Mansueti (1962) found very few ripe-running hickory shad on the spawning grounds in the Chesapeake Bay area, and suggested that gonads mature rapidly and spawning occurs at night.

In Albemarle Sound, North Carolina, hickory shad appear to have a prolonged spawning period when compared to other alosines, but that period occurs earlier in the season (Batsavage and Rulifson 1998). It is unknown how long adult hickory shad remain in freshwater after they have spawned.

Spawning substrate associations

B. M. Richardson (Maryland Department of Natural Resources, personal communication) has reported catching adult hickory shad in waters of Maryland rivers, where complex structures, such as ledges and fallen trees are present. Bottom composition in these waters tended to be mud, sand, and/or gravel. Harris and Hightower (2007) reported that hickory shad spawning in the Roanoke River were concentrated in areas of moderate to high water velocity and sediments dominated by cobble, gravel, and sand, but not silt.

Spawning depth associations

Little information is available on spawning depth preferences for hickory shad. Hawkins (1980) noted that hickory shad prefer to spawn in the deep, dark tributaries of the Neuse River, North Carolina. Similarly, Moody (1961) found that hickory shad were more abundant (by frequency of occurrence and by weight) in deeper water than American shad in the St. Johns River, Florida.

Spawning water temperature

Temperature (^oC)	Location	Citation
13 - 21	Albemarle, NC	Street et al. 1975
14 - 19	Tar River, NC	Marshall 1976
15 - 22	Altamaha River, GA	Street 1970
7.8 - 20.5	Maryland rivers	B. M. Richardson, MD DNR, personal communication

Table 3-1. Hickory shad spawning temperatures for locations along the Atlantic coast of North America

Some studies have examined spawning temperature preferences for hickory shad (Table 3-1). Spawning activity occurs in water temperatures that range from 8 to 22°C (Rulifson et al. 1982; Batsavage and Rulifson 1998), but typically peaks in waters temperatures between 15 and 19°C (Mansueti 1962; Street 1970; Pate 1972; Schaeffer 1976; Rulifson et al. 1982). In the Neuse River, North Carolina, spawning occurred at water temperatures of 10 to 23°C , with peak numbers of eggs collected at 12 to 16°C (Burdick and Hightower 2006). Eggs were collected in the Roanoke River at temperatures ranging from 10.2 to 17.0°C (Harris and Hightower 2007).

Spawning dissolved oxygen associations

Adults have been found spawning in Maryland waters where the dissolved oxygen level was between 5.7 and 11.8 mg/L (B. M. Richardson, Maryland Department of Natural Resources, personal communication). Eggs were collected in the Roanoke River at dissolved oxygen levels ranging from 6.76 to 11.27 mg/L (Harris and Hightower 2007).

Spawning water velocity/flow

Hawkins (1980) reported that hickory shad might prefer slow-flowing areas of the Neuse River, North Carolina, for spawning. Conversely, hickory shad in Maryland have been reported to favor habitat with faster moving water than that of American shad (B. M. Richardson, Maryland Department of Natural Resources, personal communication). Roanoke River sites where hickory shad spawning occurred had significantly higher water velocities than nearby sites with no spawning (Harris and Hightower 2007). Main channel sites where spawning occurred had median current velocities of 0.20 to 0.39 m/s (Harris and Hightower 2007).

Spawning feeding behavior

Pate (1972) did not find any stomach contents in over 400 adult migrating hickory shad that he examined from the Neuse River, North Carolina. However, adult hickory shad in the St.

Johns River, Florida, were found actively feeding, with 62.4% of the food items consisting of fish, and to a lesser extent, crustaceans (Williams et al. 1975).

Spawning competition and predation

Although no information on predation was found in the literature, striped bass have been reported preying heavily on hickory shad beginning in early April at Deer Creek, Maryland (B. M. Richardson, Maryland Department of Natural Resources, personal communication).

Part B. Hickory Shad Egg and Larval Habitat

Geographical and temporal movement patterns

        In general, little is known about the movement of hickory shad eggs and larvae. Eggs are generally adhesive and typically sink to the bottom in undisturbed or moderately agitated water, but are semi-demersal in slow moving currents and buoyant under turbulent conditions (Mansueti 1962).

Egg and larval depth associations

        As with adult hickory shad, little habitat information is known about larval individuals. Mansueti (1962) found hickory shad (9 to 20 mm) at depths of 20 feet at approximately 35 to 40 miles upstream from the mouth of the Patuxent River, Maryland.

Egg and larval water temperature

        In the wild, hickory shad eggs have been collected in water temperatures between 9.5 and 22°C in rivers of North Carolina (Street 1970; Pate 1972; Marshall 1976; Hawkins 1980). In the laboratory, early efforts to propagate hickory shad failed. Eventually, Mansueti (1962) successfully hatched eggs in the laboratory at 18.3°C and 21.1°C, with hatching occurring 5 to 10 hours sooner under the warmer conditions. Prolarvae hatching occurred 2 to 3 days after fertilization, with an average hatch time of 55 to 60 hours. Prolarvae fully absorb the yolk sac after 4 to 5 days, and postlarvae begin feeding exogenously at this point. The size range of postlarvae is from 5.5 to 7.0 mm (Mansueti 1962). The state of Maryland reported successful incubation of eggs at 17.8°C (64°F), with hatching occurring in 5 to 6 days (B. M. Richardson, Maryland Department of Natural Resources, personal communication). Newer aquaculture spawning methods have been highly successful, and larvae and fingerlings have been transplanted in large quantities to Chesapeake Bay tributaries (Hendricks 2003).

Egg and larval dissolved oxygen associations

        Viable hickory shad eggs have been collected in the Neuse River, North Carolina, where dissolved oxygen concentrations were between 5 and 10 mg/L (Hawkins 1980).

Egg and larval pH associations

        Hickory shad eggs were found in water with a pH range of 6.4 to 6.6 in the Neuse River, North Carolina (Hawkins 1980).

Part C. Hickory Shad Juvenile (Riverine/Estuarine) Habitat

Geographical and temporal movement patterns

Postlarval hickory shad begin transforming into juveniles when they are 10 to 35 mm long (Ulrich et al. 1979; Krauthamer and Richkus 1987); the minimum size at which they are considered fully developed juveniles is 35 mm (Mansueti and Hardy 1962). Capture of juvenile hickory shad in Maryland rivers often occurs at sharp drop-offs, in schools of several dozen, which suggests a strong schooling behavior (B. M. Richardson, Maryland Department of Natural Resources, personal communication).

Several studies suggest that most young hickory shad leave freshwater and brackish habitats in early summer and migrate to estuarine nursery areas at an earlier age than other anadromous alosines (Mansueti 1962; Adams 1970; Pate 1972; Sholar 1977). Juveniles have also been caught in the surf zone off Long Island, New York, from April to November, which supports this hypothesis (Schaefer 1967). In the Altamaha River, Georgia, juveniles drift downstream and reach the estuary by late spring (Street 1970). Juveniles also drift down the Pee Dee and Waccamaw rivers, in South Carolina, earlier than young American shad, and enter Winyah Bay by July, remaining there throughout the first summer. By early fall, juveniles have moved into oceanic waters (Crochet et al. 1976). Trippell et al. (2007) found a few juvenile hickory shad in the St. Johns River, Florida, near Palatka (rkm 127), from May to October, with the highest catch rates occurring in October.

Some juvenile hickory shad may forego estuarine waters altogether and move directly into saltwater, unlike other alosine species that use freshwater nurseries before moving into marine waters (Pate 1972; Sholar 1977; Batsavage and Rulifson 1998). This ability to move directly into saltwater is believed to occur in hickory shad at an earlier age than for other anadromous alosines (Mansueti 1962; Schaefer 1967; Adams 1970; Pate 1972; Sholar 1977; Batsavage and Rulifson 1998). Additionally, some researchers suggest that juvenile hickory shad initially move to shallow offshore areas in Georgia near the mouth of the Altamaha River, and then disperse farther by August and September (Godwin and Adams 1969; Street 1970). Juvenile hickory shad are thought to be larger in size than other alosines at similar ages due to an earlier spawning period and a faster growth rate (Godwin and Adams 1969). Juvenile hickory shad that are larger than average compared to other alosines have been captured in Maryland (Mansueti 1962; Virginia (Atran et al. 1983) and Georgia rivers (Adams 1970).

Juveniles and the saltwater interface

In Maryland, juvenile hickory shad were captured in waters with salinities that ranged from 0 to 7.2 ppt (B. M. Richardson, Maryland Department of Natural Resources, personal communication). In addition, juveniles were found during the summer in estuarine waters of the Altamaha River, Georgia, when salinities reached 10 ppt, and during the winter, when salinities ranged from 10 to 20 ppt (Street 1970). As noted above, juveniles may forego the oligohaline portion of the estuary in favor of a more saline nursery environment (Pate 1972).

Juvenile depth associations

In South Carolina, juvenile hickory shad are more predominant in shallow expanses of sounds and bays, compared to deeper, channel habitats occupied by juvenile American shad and blueback herring. The variation in distribution is likely the result of differences in food preferences. Small fishes preferred by hickory shad are likely more numerous in shallower habitats adjacent to marshlands (McCord 2003).

Juvenile water temperature

B. M. Richardson (Maryland Department of Natural Resources, personal communication) has caught juveniles in Maryland rivers with water temperatures between 16 and 31°C, usually corresponding to early July through early October. Davis (1973) reported that hickory shad remain in freshwater until temperatures drop in October and November, then move downstream as temperatures continue to decrease.

Juvenile dissolved oxygen associations

Juveniles in Maryland waters were captured where dissolved oxygen ranged from 4.1 to 10.9 mg/L (B. M. Richardson, Maryland Department of Natural Resources, personal communication).

Part D. Hickory Shad Late Stage Juvenile and Adult Marine Habitat

Geographical and temporal patterns at sea

　　As with many aspects of hickory shad life history, very little is known about the distribution and movements of hickory shad in the ocean (Street 1970; Richkus and DiNardo 1984). Adults have been caught along the southern New England coast in the summer and fall (Bigelow and Schroeder 1953) and off Long Island, New York (Schaefer 1967). Anglers report catching them in nearshore waters at Cape May, New Jersey, from May to November, and then capturing them in inlets from November through December (W. Gordon, recreational angler, personal communication). Unlike American shad, hickory shad rarely migrate to the Gulf of Maine or upper Bay of Fundy during the summer (M. J. Dadswell, Canada Department of Fisheries and Oceans, personal communication). Furthermore, some researchers believe that adults do not move far from land while at sea (Mansueti and Hardy 1967).

Temperature associations at sea

　　Little information is available on hickory shad habitat associations offshore. Anglers fishing for hickory shad have reported that they will move further offshore from the nearshore waters of New Jersey, when water temperatures reach above 21°C (W. Gordon, recreational angler, personal communication).

Feeding behavior at sea

　　Adult hickory shad are piscivorous; they generally feed on sand lance, anchovies, cunner, herring, scup, and silversides. This species may also feed on squid, fish eggs, small crabs, and pelagic crustaceans (Hildebrand and Schroeder 1928; Williams et al. 1975; Bigelow and Schroeder 2002).

Section II. Significant Environmental, Temporal, and Spatial Factors Affecting Distribution of Hickory Shad

Table 3-2. Significant environmental, temporal, and spatial factors affecting distribution of hickory shad. Given that there is very little information on hickory shad, this table should be used only as a general reference. The term "reported" is used to denote ranges that were found in the literature, but should not be regarded as the full range tolerated by this species. NIF = No Information Found.

Life Stage	Time of Year and Location	Depth (m)	Temperature (°C)	Salinity (ppt)	Substrate	Current Velocity (m/sec)	Dissolved Oxygen (mg/L)
Spawning Adult	Early December (FL) through late June (VA) in natal rivers and tribuaries from Connecticut River southward to Halifax River, Florida (mostly found from Maryland rivers southward)	**Tolerable:** NIF **Optimal:** NIF **Reported:** May prefer deeper waters than American shad	**Tolerable:** 8-23 **Optimal:** 12-19 **Reported:** Variable	**Tolerable:** NIF **Optimal:** NIF **Reported:** Usually freshwater	**Tolerable:** NIF **Optimal:** NIF **Reported:** Cobble, gravel, sand	**Tolerable:** NIF **Optimal:** NIF **Reported:** Variable	**Tolerable:** NIF **Optimal:** NIF **Reported:** Found 5.7-11.8
Egg & Larvae	Early December (FL) through late June (VA) (eggs may be released in batches) at spawning areas or slightly downstream	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** 9.5-22 (egg) **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** Minimum 5
Early Juvenile – Riverine/ Estuarine Environment	When they reach 35 mm TL (begin moving downstream at an earlier age than other alosines); reach estuaries by late spring/ early summer and ocean by early fall; or may forego estuarine waters and move directly to saltwater	**Tolerable:** NIF **Optimal:** NIF **Reported:** Generally shallow waters	**Tolerable:** 16-31 **Optimal:** NIF **Reported:** Variable	**Tolerable:** NIF **Optimal:** NIF **Reported:** Freshwater, brackish, and saltwater	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** Minimum 4

Life Stage	Time of Year and Location	Depth (m)	Temperature (°C)	Salinity (ppt)	Substrate	Current Velocity (m/sec)	Dissolved Oxygen (mg/L)
Subadult & Non-spawning Adult– Estuarine / Oceanic Environment	Up to 8 years in Cape Cod, Massachusetts to Cape Canaveral, Florida (concentrated from NY southward)	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** Move offshore when temperatures reach 21°C	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF

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Chapter 4

ALEWIFE

(*Alosa pseudoharengus*)

Section I. Alewife Description of Habitat

Alewife General Habitat Description and Introduction

The alewife (*Alosa pseudoharengus*) is an anadromous, highly migratory, euryhaline, pelagic, schooling species. The species spends the majority of its life at sea, returning to freshwater river systems along the Atlantic coast of the United States to spawn (ASMFC 1985). While most alewife are native-anadromous fish, some have been introduced to landlocked systems. Researchers examined two distant anadromous alewife stocks to test whether landlocked stocks were more closely related to St. Croix anadromous stocks or to more geographically distant anadromous stocks. Landlocked alewife were found to be distantly related to all the anadromous stocks tested. A variety of statistical tests confirmed that anadromous and landlocked populations of alewife in the St. Croix are genetically divergent (FST = 0.244). These results implied that very little, if any, interbreeding occurs between the two life history types (Bentzen and Paterson 2006; Willis 2006). Furthermore, significant genetic differences were observed between anadromous alewife populations in the St. Croix and anadromous populations in the LaHave and Gaspereau Rivers, as well as between the two anadromous St. Croix samples (Dennis Stream and Milltown). These results imply homing of alewives to their natal streams and, consequently, at least partial reproductive isolation between spawning runs, even at the level of tributaries within the St. Croix River (Willis 2006).

The historical coastal range of the anadromous alewife was from South Carolina to Labrador, Nova Scotia, and northeastern Newfoundland (Berry 1964; Winters et al. 1973; Burgess 1978). However, more recent surveys indicate that they do not currently occur in the southern range beyond North Carolina (Rulifson 1982; Rulifson et al. 1994). Alewife from the southernmost portion of the species' range migrate long distances (over 2000 km) in ocean waters of the Atlantic seaboard. Patterns of migration may be similar to those of American shad (*Alosa sapidissima*) (Neves 1981). Although alewife and blueback herring co-occur throughout much of their respective ranges, alewife are typically more abundant than blueback herring in the northern portion of their range (Schmidt et al. 2003).

Recent analyses to determine the current status of alewife in the Connecticut, Hudson, and Delaware River systems, suggest that alewife are showing signs of overexploitation (for example, lower mean age, fewer returning spawners, and lower overall abundance) in all of these rivers. However, researchers noted that recently some runs in the northeastern U.S. and Canada have shown increased alewife abundance (Schmidt et al. 2003). Furthermore, alewife appeared to be thriving in inland waters, colonizing many freshwater bodies, including all five Great Lakes (Waldman and Limburg 2003).

While this document will focus primarily on the anadromous alewife populations, much of the research on specific environmental requirements of alewife, such as temperature, dissolved oxygen, salinity, and pH, has been conducted on landlocked populations, not anadromous stocks; therefore data should be interpreted with discretion (Klauda et al. 1991).

Part A. Alewife Spawning Habitat

Geographical and temporal patterns of migration

The spring adult alewife migration to spawning grounds in freshwater and brackish water progresses seasonally from south to north, with populations further north returning later in the season as water temperatures rise. Neves (1981) suggested that alewife migrate from offshore waters north of Cape Hatteras, encountering the same thermal barrier as American shad. Alewife then move south along the Atlantic coast for fish homing to southern rivers, while northbound pre-spawning adults continue traveling up the coast (Stone and Jessop 1992). The species spawns in rivers, ponds, and lakes (lacustrine habitat), as far south as North Carolina and as far north as the St. Lawrence River, Canada (Neves 1981; S. Lary, U.S. Fish and Wildlife Service, personal communication).

State or region	Spawning season	Citations
Bay of Fundy tributaries	late April or early May	Leim and Scott 1996; Dominy 1971, 1973
Gulf of St. Lawrence tributaries	late May or early June	Leim and Scott 1996; Dominy 1971, 1973
Maine	late April to mid-May	Rounsefell and Stringer 1943; Bigelow and Schroeder 1953; Havey 1961; Libby 1981
	mid-May to mid-June	S. Lary, U.S. Fish and Wildlife Service, personal communication
Massachusetts	early to mid-April	Belding 1921; Bigelow and Schroeder 1953
Mid-Atlantic and southern New England	late March or early April	Cooper 1961; Kissil 1969; Marcy 1969; Smith 1971; Saila et al. 1972; Richkus 1974; Zich 1978; Wang and Kernehan 1979
Chesapeake Bay region	mid-March	Jones et al. 1978; Loesch 1987
North Carolina	late February	Holland and Yelverton 1973; Frankensteen 1976

Table 4-1. Reported spawning seasons for alewife along the Atlantic coast of North America

Alewife typically spawn from late February to June in the south, and from June through August in the north (Table 4-1) (Marcy 1976a; Neves 1981; Loesch 1987). Spawning is

triggered most predictably by a change in the water temperature. Movement upstream may be controlled by water flow, with increased movement occurring during higher flow periods (Collins 1952; Richkus 1974). However, extreme high flows can act as a velocity barrier delaying or preventing upstream migration and access to spawning habitat (S. Lary, U.S. Fish and Wildlife Service, personal communication).

Although adult alewife will move upstream at various times of the day, peak migration typically occurs between dawn and noon, and from dusk to midnight (Richkus 1974; Rideout 1974; Richkus and Winn 1979). Researchers have found that high midday movement is restricted to overcast days, and nocturnal movement occurs when water temperatures are abnormally high (Jones et al. 1978). Typically, males arrive before females at the mouths of spawning rivers (Cooper 1961; Tyus 1971; Richkus 1974).

There is strong evidence suggesting that alewife home to their natal rivers to reproduce; however, some individuals have been found to colonize new areas. Alternatively, alewife may reoccupy systems from which they have been extirpated (Havey 1961; Thunberg 1971; Messieh 1977; Loesch 1987). Messieh (1977) found that alewife strayed considerably to adjacent streams in the St. Johns River, Florida, particularly during the pre-spawning period (late winter, early spring), but not during the spawning run. It appears that olfaction is the primary means for homing behavior (Ross and Biagi 1990).

Spawning location (ecological)

Alewife select slow-moving sections of rivers or streams to spawn, where the water may be as shallow as 30 cm (Jones et al. 1978). The species may also spawn in lakes or ponds, including freshwater coves behind barrier beaches (Smith 1907; Belding 1921; Leim and Scott 1966; Richkus 1974; Colette and Klein-MacPhee 2002). In watersheds where dams are an impediment, spawning may occur in shore-bank eddies or deep pools below the dams (Loesch and Lund 1977). Additionally, in New England and Nova Scotia, alewife spawn in lakes and ponds located within coastal watersheds (Loesch 1987). For this reason, they are typically more abundant than blueback herring in rivers with abundant headwater ponds. In rivers where headwater ponds are absent or scarce, alewife are less abundant in headwater reaches; however, blueback herring utilize the mainstream proper for spawning in those systems (Ross and Biagi 1990). In tributaries of the Rappahannock River, Virginia, upstream areas were found to be more important than downstream areas for spawning alewife (O'Connell and Angermeier 1997). Although earlier studies suggested that alewife ascend further upstream than blueback herring (Hildebrand 1963; Scott and Crossman 1973), Loesch (1987) noted that both species have the ability to ascend rivers far upstream.

Boger (2002) found that river herring within the Rappahannock River watershed spawned in larger, elongated watersheds with greater mean elevation and greater habitat complexity. This researcher suggested that such areas are likely to have more stable base flows that can maintain suitable spawning habitat even during dry years. Additionally, spawning areas had a greater percentage of deciduous forest and developed areas and less grassland areas (Boger 2002).

Temporal spawning patterns

Alewife usually spawn 3 to 4 weeks before blueback herring in areas where they co-occur; however, there may be considerable overlap (Loesch 1987) and peak spawning periods may differ by only 2 to 3 weeks (Jones et al. 1978). In a tributary of the Rappahannock River, Virginia, O'Connell and Angermeier (1997) found that blueback herring eggs and larvae were more abundant than those of alewife, but alewife used the stream over a longer period of time. The researchers also reported a minor three-day overlap of spawning by these two alosine species. It has been hypothesized that alewife and blueback herring select separate spawning sites in sympatric areas to reduce competition (Loesch 1987). O'Connell and Angermeier (1997) reported that the two species used different spawning habitat due to a temporal, rather than spatial, segregation that minimizes the competition between the two species.

Alewife may spawn throughout the day, however, most spawning occurs at night (Graham 1956). One female fish and up to 25 male fish broadcast eggs and sperm simultaneously just below the surface of the water or over the substrate (Belding 1921; McKenzie 1959; Cooper 1961). Spawning lasts two to three days for each group or "wave" of fish that arrives (Cooper 1961; Kissil 1969; Kissil 1974), with older and larger fish usually spawning first (Belding 1921; Cooper 1961; Libby 1981, 1982). Following spawning, the adult spent fish quickly return downstream (Colette and Klein-MacPhee 2002).

Maturation and spawning periodicity

State	% of spawners	Citations
Nova Scotia	60%	O'Neill 1980
Maryland	30-72%	Weinrich et al. 1987; Howell et al. 1990
Virginia	61%	Joseph and Davis 1965
North Carolina	13.7% (1993); 61% (1995)	Winslow 1995

Table 4-2. Percentage of repeat spawners for alewife along the Atlantic coast of North America

Many alewife are repeat spawners, with some individuals completing seven or eight spawning events in a lifetime (Table 4-2) (Jessop et al. 1983). It is not clear whether there is a clinal trend from south to north for repeat spawning (i.e., more in the north than south) (Klauda et al. 1991), or if there is a typical percent of the annual return population that repeat spawns (i.e., 30 to 40% repeat spawners throughout their range) (Richkus and DiNardo 1984). Furthermore, Kissil (1974) suggested that alewife might spawn more than once in a season.

Adults will typically spend two to four years at sea before returning to their natal rivers to spawn (Neves 1981). The majority of adults reach sexual maturity at 3, 4, or 5 years of age, although some adults from North Carolina (Richkus and DiNardo 1984) have returned to spawn at age-2 (Jessop et al. 1983). The oldest alewife recorded in North Carolina were age-9 (Street et

al. 1975; Johnson et al. 1979); age-10 fish have been caught in New Brunswick (Jessop et al. 1983) and Nova Scotia (O'Neill 1980). Additionally, Kissil (1974) found that alewife spawning in Bride Lake, Connecticut, spent three to 82 days on the spawning grounds, while Cooper (1961) reported that most fish left within five days of spawning in Rhode Island.

Spawning and the saltwater interface

While it is known that alewife can adjust to a wide range of salinities, published data on alewife tolerance ranges are lacking (Klauda et al. 1991). Richkus (1974) found that adults that were transferred from freshwater to saline water (32 ppt), and vice versa, experienced zero mortality. In the north, Leim (1924) studied the life history of American shad and noted that they do not ascend far beyond the tidal influence of the river, yet alewife migrate as far upstream as they can travel. He concluded that alewife may be less dependent on saltwater for development (Leim 1924). Also, unlike American shad, some populations of alewife have become landlocked and are not at all dependent on saltwater (Scott and Crossman 1973).

Spawning substrate associations

The spawning habitat of alewife can range from sand, gravel, or coarse stone substrates, to submerged vegetation or organic detritus (Edsall 1964; Mansueti and Hardy 1967; Jones et al. 1978). Boger (2002) found that river herring spawning areas along the Rappahannock River, Virginia, had substrates that consisted primarily of sand, pebbles, and cobbles (usually associated with higher-gradient streams). In contrast, areas with little or no spawning activity were dominated by organic matter and finer sediments (usually associated with lower-gradient streams and comparatively more agricultural land use) (Boger 2002).

Pardue (1983) evaluated studies of cover component in alewife spawning areas, suggesting that substrate characteristics and associated vegetation were a measure of the ability of a habitat to provide cover to spawning adults, their eggs, and developing larvae. In high flow areas, there is little accumulation of vegetation and detritus, while in low flow areas, detritus and silt accumulate and vegetation has the opportunity to grow (Pardue 1983). Pardue (1983) suggested that substrates with 75% silt (or other soft material containing detritus and vegetation) and sluggish waters are optimal for alewife**.**

Spawning depth associations

Water depth in spawning habitat may be a mere 15 cm deep (Bigelow and Schroeder 1953; Rothschild 1962), or as deep as 3 m (Edsall 1964); however, spawning typically occurs at less than 1 m (Murdy et al. 1997). Adults may utilize deeper water depths when not spawning in order to avoid high light intensities (Richkus 1974).

Spawning water temperature

Temperature (^oC)	Location	Citation
14.0 – 15.5 (peak)	Rhode Island	Jones et al. 1978
7.0 – 10.9	Lower Connecticut River	Marcy et al. 1976a
10.5 – 21.6	Chesapeake Bay	Jones et al. 1978
11 - 19	Patuxent River, MD	J. Mowrer, Morgan State University, unpublished data
13 (peak)	Lake Mattamuskeet, NC	Tyrus 1974

Table 4-3. Alewife spawning temperatures for locations along the Atlantic coast of North America

Adult alewife have been collected in temperatures ranging from 5.7°C to 32°C (Marcy 1976b; Jones et al. 1978). Spawning temperatures along the Atlantic coast fall within this broader range (Table 4-3). There is some discrepancy regarding the minimum spawning temperature for alewife. Although running ripe fish of both sexes have been reported at temperatures as low as 4.2°C in the Chesapeake Bay area (Mansueti and Hardy 1967), some researchers suggest that the minimum spawning temperature for adult alewife is 10.5°C (Cianci 1965; Loesch and Lund 1977). Additionally, lower temperatures may be dangerous for spawning alewife. Otto et al. (1976) found that the lower incipient lethal temperature range for adults acclimated at 15.0°C and 21.0°C was between 6°C and 8°C. In this study, no fish survived below 3°C, regardless of acclimation temperature (Otto et al. 1976). Furthermore, at temperatures below 4.5°C, normal schooling behavior was significantly reduced for adult alewife from Lake Michigan (Colby 1973).

As water temperatures rise, alewife migration eventually slows. Cooper (1961) noted that upstream migration ceased in a Rhode Island stream when temperatures reached 21°C, while Edsall (1970) reported that spawning ceases altogether at 27.8°C. Ultimately, higher temperatures may cause problems for alewife. In fact, Otto et al. (1976) found that upper incipient lethal temperatures (temperature at which 50% of the population survives) ranged from 23.5°C to 24.0°C for adults that were acclimated at temperatures of 10°C, 15°C, and 20°C. Another study reported upper incipient lethal temperatures of 29.8°C and 32.8°C at acclimation temperatures of 16.9°C and 24.5°C, respectively (Stanley and Holzer 1971). In addition, McCauley and Binkowski (1982) reported upper incipient lethal temperatures of 31°C to 34°C after acclimation at 27°C for a northern population of adults.

In general, alewife may prefer cooler water, and northern populations may be more cold tolerant than other migratory anadromous fish (Stone and Jessop 1992). Richkus (1974) showed that the response of migrating adults to a particular hourly temperature was determined by their relationship to a changing baseline temperature, and not on the basis of the absolute value of temperature. Stanley and Colby (1971) found that decreasing temperatures (from 16°C to 3°C at a rate of 2.5°C per day) reduced adult alewife ability to osmoregulate. Adults were also shown

to survive temperature decreases of 10°C, regardless of acclimation temperature, if the temperature did not drop below 3°C (Otto et al. 1976).

Spawning dissolved oxygen associations

There is little information regarding sensitivities of various life history stages of alewife to dissolved oxygen (Klauda et al. 1991). In one study, adults exposed to dissolved oxygen concentrations ranging from 2.0 to 3.0 mg/L for 16 hours in the laboratory experienced a 33% mortality rate. Alewife were able to withstand dissolved oxygen concentrations as low as 0.5 mg/L for up to 5 minutes, as long as a minimum of 3.0 mg/L was available, thereafter (Dorfman and Westman 1970). Additionally, Jones et al. (1988) suggested that the minimum dissolved oxygen concentration for adult alewife is 5.0 mg/L.

Spawning water velocity/flow

Increased movement upstream occurs during higher water flows (Collins 1952; Richkus 1974), while spawning typically takes place in quiet, slow-moving waters for alewife (Smith 1907; Belding 1921; Marcy 1976a). Some researchers have noted differential selection of spawning areas in alewife. For example, in Connecticut, alewife choose slower moving waters in Bride Lake (Kissil 1974) and Higganum and Mill creeks, while blueback herring select fast-moving waters in the upper Salmon River and Roaring Brook (Loesch and Lund 1977). In other areas where alewife and blueback herring are forced to spawn in the same vicinity due to blocked passage (Loesch 1987), alewife generally spawn along shorebank eddies or deep pools, whereas, blueback herring will typically select the main stream flow for spawning (Loesch and Lund 1977). In North Carolina, alewife utilize slow moving streams and oxbows (Street et al. 2005).

Spawning pH associations

Few researchers have reported on pH sensitivity in alewife (Klauda et al. 1991). Byrne (1988) found that the average pH level was 5.0 in several streams in New Jersey where alewife spawning was known to occur. Laboratory tests found that fish from those streams could successfully spawn at a pH as low as 4.5 (Byrne 1988). In another study, adult alewife tolerated a pH range of 6.5 to 7.3 (Collins 1952). When aluminum pulses were administered in the laboratory, critical conditions for spawning could occur during an acidic pulse between pH 5.5 and 6.2, with concomitant concentrations of total monomeric aluminum ranging from 15 to 137 µg/L for a pulse duration of 8 to 96 hours (Klauda 1989). Klauda et al. (1991) suggested a pH range of 5 to 8.5 as suitable for alewife eggs, but no range was provided for spawning.

Spawning feeding behavior

Adult alewife typically do not feed during their upstream spawning run (Bigelow and Schroeder 1953; Colby 1973). Spent fish that have reached brackish waters on their downstream migration will feed voraciously, mostly on mysids (Colette and Klein-MacPhee 2002). While adults may consume their own eggs during the spawning run (Edsall 1964; Carlander 1969), juveniles reportedly feed more actively on them (Colette and Klein-MacPhee 2002).

Spawning competition and predation

Adult alewife and blueback herring play an important role in the food web and in maintaining the health of the ecosystem. In the inland freshwater and coastal marine environments they provide forage for bass, trout, salmonids, other fish, ospreys, herons, eagles, kingfishers, cormorants, and aquatic fur-bearing mammals (Colby 1973; Royce 1943; Scott and Scott 1988; Loesch 1987; S. Lary, U.S. Fish and Wildlife Service, personal communication). In the marine environment, they are eaten by a variety of predators, such as bluefish, weakfish, striped bass, cod, pollock, and silver hake, as well as marine mammals and sea birds. Additionally, alewife are a host to native freshwater mussels, which they carry up and down rivers in their gills. Furthermore, spawning alewife heading upriver give cover to out-migrating Atlantic salmon smolts in the spring (S. Lary, U.S. Fish and Wildlife Service, personal communication).

Erkan (2002) notes that predation of alosines has increased dramatically in Rhode Island rivers in recent years, especially by the double-crested cormorant, which often takes advantage of fish staging near the entrance to fishways. Populations of nesting cormorant colonies have increased in size and expanded into new areas. Predation by otters and herons has also increased, but to a lesser extent (D. Erkan, Rhode Island Department of Environmental Management, personal communication).

In many coastal communities, the annual alewife run is an integral part of the local culture, and local residents have initiated efforts to protect and restore their cultural link to this fishery, to develop effective management strategies for restoration, to establish self-sustaining harvest levels, and to enhance community education (S. Lary, U.S. Fish and Wildlife Service, personal communication).

Factors affecting stock size

At low stock levels, Havey (1973) and Walton (1987) demonstrated a weak relationship between spawning stock and abundance of juvenile migrant alewife. Jessop (1990) found a stock recruitment relationship for the spawning stock of river herring and year-class abundance at age 3. Despite these results, most studies have been unable to detect a strong relationship between adult and juvenile abundance of clupeids (Crecco and Savoy 1984; Henderson and Brown 1985; Gibson 1994; Jessop et al. 1994). Researchers have suggested that although year-class is driven mostly by environmental factors (see subsequent sections), if the parent stock size falls below a critical level due to natural and manmade environmental impacts, the size of the spawning stock will likely become a factor in determining juvenile abundance (Kosa and Mather 2001).

Part B. Alewife Egg and Larval Habitat

Geographical and temporal movement patterns

Fertilized eggs remain demersal and adhesive for several hours (Mansueti 1956; Jones et al. 1978), after which they become pelagic and are transported downstream (Wang and Kernehan 1979). Marcy (1976a) observed eggs more often near the bottom than at the surface in the Connecticut River. Eggs may hatch anywhere from 50 to 360 hours (2 to 15 days) after spawning, depending on water temperature (Fay et al. 1983); however, eggs most often hatch within 80 to 95 hours (3 to 5 days) (Edsall 1970).

Within two to five days of hatching, the yolk-sac is absorbed and larvae begin feeding exogenously (Cianci 1965; Jones et al. 1978). Post-yolk-sac larvae are positively phototropic (Odell 1934; Cianci 1965). Dovel (1971) observed larvae near or slightly downstream of presumed spawning areas in the Chesapeake Bay, where the water was less than 12 ppt salinity (Dovel 1971). Larvae were also found in or close to observed spawning areas in Nova Scotia rivers in relatively shallow water (2 m) over sandy substrate (O'Neill 1980).

Eggs, larvae, and the saltwater interface

Dovel (1971) found that 99% of alewife eggs in the upper Chesapeake Bay were in freshwater (0 ppt). Larvae were collected where salinities ranged from 0 to 8 ppt, but again, most (82%) were collected in freshwater (Dovel 1971). Klauda et al. (1991) suggested that the optimal range for alewife egg development is 0 to 2 ppt. Additionally, growth rates of larval alewife are considerably faster in saltwater compared to freshwater at temperatures of 26.4°C (Klauda et al. 1991).

Egg and larval substrate associations

As with spawning habitat, Pardue (1983) suggested that optimal egg and larval habitat is found in substrates of 75% silt or other soft material containing detritus and vegetation**.**

Egg and larval water temperature

For alewife in general, average time to median hatch varies inversely with temperature. Edsall (1970) reported the following hatch times for alewife eggs taken from Lake Michigan: 2.1 days at 28.9°C, 3.9 days at 20.6°C, and 15 days at 7.2°C. Reported hatch times in saltwater are comparable: 2 to 4 days at 22°C (Belding 1921); 3 days at 23.8°C to 26.8°C, and 3 to 5 days at 20°C (Mansueti and Hardy 1967); 6 days at 15.5°C (Bigelow and Welsh 1925).

Kellogg (1982) found that eggs from the Hudson River, New York, achieved maximum hatching success at 20.8°C. Edsall (1970) reported some hatching at temperatures ranging from 6.9°C to 29.4°C for eggs from Lake Michigan; however, temperatures below 11°C caused a high percentage of deformed larvae. The optimum hatching performance occurred between 17.2°C and 21.1°C. Although this was the suggested optimal range, it was determined that considerable hatch rates and proper development could occur over a broader range from 10.6°C to 26.7°C

(Edsall 1970). Furthermore, in the upper Chesapeake Bay, alewife eggs were collected where temperatures ranged from 7°C to 14°C, with 70% of eggs found between 12°C and 14°C (Dovel 1971).

Edsall (1970) correlated egg mortality with incubation temperature. His equation follows for predicting incubation time of alewife eggs using a relationship with temperature:

$$t = 6.335 \times 10^6 \, (T)^{-3.1222}$$

where t = time in days

T = incubation temperature in degrees Fahrenheit

Several researchers have attempted to determine the effects of temperature on alewife eggs. One study on the effects of power plants on alewife eggs found that they suffered no significant mortality or abnormal egg development after acclimation at 17° C, and subsequent exposure to 24.5° C for 6 to 60 minutes (Schubel and Auld 1972). Koo et al. (1976) determined that the critical thermal maximum (CTM) for alewife eggs was 35.6° C, acclimated at 20.6° C, with a critical exposure period of 5 to 10 minutes.

Larval alewife were collected at water temperatures between 4°C and 27°C in the upper Chesapeake Bay, although 98% were collected at water temperatures of 25°C (Dovel 1971). In laboratory experiments, larvae acclimated at 18.6°C withstood temperatures as high as 33.6°C for one hour (Koo et al. 1976). The upper temperature tolerance limit for yolk-sac larvae from the Hudson River, New York, acclimated at around 15°C was 31°C (Kellogg 1982); their preferred range when acclimated at 20° C appears to be 23°C to 29°C (EA 1978; Kellogg 1982). Although alewife eggs taken from Lake Michigan were able to hatch at temperatures as low as 6.9°C, larvae held at incubation temperatures below 10.6°C had a 69% rate of deformities (Edsall 1970).

Dovel (1971) found that growth rates of alewife larvae were much lower in freshwater compared to slightly saline water (1.0 to 1.3 ppt) at 26.4°C. He also observed substantial growth increases with small temperature increases above 20.8°C. Average daily weight gain for alewife larvae has been directly correlated with water temperature. The maximum larval growth rate was 0.084 g/day at 29.1°C; net gain in biomass (a function of survival and growth) was highest at 26.4°C (Kellogg 1982).

Based on Kellogg's (1982) observations that the optimum growth temperature (26°C) exceeds peak spawning temperatures by about 10°C to 13°C, it was suggested that the survival and early development of young alewife would not likely be threatened by rapid warming trends following spawning or by moderate thermal discharges. Furthermore, it was indicated that above normal temperature elevations following spawning and hatching would probably be beneficial to alewife populations (Kellogg 1982).

Egg and larval dissolved oxygen associations

Jones et al. (1988) determined that the minimum dissolved oxygen concentration requirement for eggs and larvae is 5.0 mg/L. Furthermore, O'Connell and Angermeier (1997) found that dissolved oxygen and current velocity were the strongest predictors of alewife early egg presence in a Virginia stream.

Egg and larval pH and aluminum associations

Klauda et al. (1991) suggest that a range of pH 5.0 to 8.5 for both the alewife egg and prolarva life stage is optimal. Klauda et al. (1987) suggested that during an acidic pulse between pH 5.5 and 6.2, critical conditions associated with more than 50% direct mortality could occur. Klauda et al. (1991) found that larvae subjected to a single 24-hour, acid-only pulse of pH 4.5 experienced no mortality, while those subjected to a 24-hour single acid pulse and 446 µg/L inorganic monomeric aluminum pulse suffered a 96% mortality rate. A single 12-hour acid-only pulse of 4.0 resulted in 38% mortality (Klauda et al. 1991).

Egg and larval water velocity/flow

Sismour (1994) observed a rapid decline in abundance of early preflexion river herring larvae in the Pamunkey River, Virginia, following high river flow in 1989. This observation lead to speculation that high flow leads to increased turbidity, which reduces prey visibility, leading to starvation of larvae (Sismour 1994). Additionally, O'Connell and Angermeier (1997) found that current velocity and dissolved oxygen were the strongest predictors of alewife early egg presence in a Virginia stream. Further north, drought conditions in Rhode Island in the summer of 1981 were strongly suspected of impacting the 1984-year class, which was only half of its expected size (ASMFC 1985). In tributaries of the Chowan system, North Carolina, water flow was related to recruitment of larval river herring (O'Rear 1983).

Egg and larval suspended solid associations

Alewife eggs subjected to suspended solids concentrations up to 1000 mg/L did not exhibit a reduction in hatching success (Auld and Schubel 1978). Despite these results, high levels of suspended sediment may significantly increase rates of egg infection from naturally occurring fungi, as was witnessed in earlier experiments (Schubel and Wang 1973); this can lead to delayed mortality (Klauda et al. 1991).

Egg and larval feeding behavior

Once alewife larvae begin feeding exogenously, they select relatively small cladocerans and copepods, adding larger species as they grow (Norden 1968; Nigro and Ney 1982). Alewife larvae are highly selective feeders (Norden 1967), usually favoring cladocerans (mainly *Cyclops* sp. and *Limnocalanus* sp.) and copepods over other food types (Norden 1968; Johnson 1983).

Egg and larval competition and predation

Alewife eggs may be consumed by yellow perch, white perch, spottail shiner, and other alewife (Edsall 1964; Kissil 1969). Alewife larvae are preyed upon by both vertebrate and invertebrate predators (Colby 1973).

Part C. Alewife Juvenile Riverine/Estuarine Habitat

Geographical and temporal movement patterns

In North Carolina, juveniles may spend the summer in the lower ends of rivers where they were spawned (Street et al. 1975). In the Chesapeake Bay, juveniles can be found in freshwater tributaries in spring and early summer, but may head upstream in mid-summer when saline waters encroach on their nursery grounds (Warriner et al. 1970). Some juveniles in the Chesapeake Bay remain in brackish water through the summer (Murdy et al. 1997).

Further north, juveniles in the Hudson River usually remain in freshwater tributaries until June (Schmidt et al. 1988). In contrast to the inshore abundance of American shad and blueback herring during the day, juvenile alewife were found to be most abundant in inshore areas at night in the Hudson River (McFadden et al. 1978; Dey and Baumann 1978). Hudson River juveniles were observed in shallow portions of the upper and middle estuary in late June and early July, where they remained for several weeks before moving offshore (Schmidt et al. 1988). Alewife typically spend three to nine months in their natal rivers before returning to the ocean (Kosa and Mather 2001).

In the summer in the Potomac River, juveniles are abundant near surface waters during the day; however, they shift to mid-water and bottom depths in September, where they remain until they emigrate in November (Warriner et al. 1970). Juvenile alewife respond negatively to light and follow diel movement patterns similar to blueback herring. Nevertheless, there appears to be some separation between the alewife and blueback herring as they emigrate from nursery grounds in the fall. The difference occurs most notably at night when alewife can be found more frequently at mid-water depths, while blueback herring are found mostly at the surface (Loesch and Kriete 1980). This behavior may reduce interspecific competition for food, given that the species' diets are similar (Davis and Cheek 1966; Burbidge 1974; Weaver 1975).

Once water temperatures begin to drop in the late summer through early winter (depending on geographic area), juveniles start heading downstream, initiating their first phase of seaward migration (Pardue 1983; Loesch 1987). Some researchers have found that movement of alewife peaks in the afternoon (Richkus 1975a; Kosa and Mather 2001), while others have found that it peaks at night (Stokesbury and Dadswell 1989). Migration downstream is also prompted by changes in water flow, water levels, precipitation, and light intensity (Cooper 1961; Kissil 1974; Richkus 1975a, 1975b; Pardue 1983). Other researchers have suggested that water flow plays only a minor role in providing migration cues under riverine conditions. Rather, these researchers think that migration timing is triggered by water temperature and moon phases that provide dark nights (i.e., new and quarter moons) (O'Leary and Kynard 1986; Stokesbury and Dadswell 1989). Additionally, Stokesbury and Dadswell (1989) found that alewife remained in the offshore region of the Annapolis estuary, Nova Scotia, for nearly one month before the correct migration cues triggered emigration. Furthermore, large juveniles begin moving downstream before smaller juveniles (Schmidt et al. 1988), inhabiting saline waters before they begin their seaward migration (Loesch 1969; Marcy 1976a; Loesch and Kriete 1980).

The influence and magnitude of migration cues on emigrating alewife may vary considerably. Richkus (1975a) observed waves of juvenile alewife leaving systems following environmental changes (e.g., changes in water flow, water levels, precipitation, and light

intensity), but the number of fish leaving was unrelated to the level of magnitude of the change. Most fish (60% to 80%) emigrated during a small percentage (approximately 8%) of available days. These waves also lasted two to three days, regardless of the degree of environmental change (Richkus 1975a). Similarly, other researchers have observed that the majority (>80%) of river herring emigrate in waves (Cooper 1961; Huber 1978; Kosa and Mather 2001). Richkus (1975a) also noted that in some instances, high abundances of juvenile alewife may trigger very early (i.e., summer) emigration of large numbers of small juveniles from the nursery area, which is likely a response to a lack of forage. Additionally, juvenile migration of alewife occurs about one month earlier than that of blueback herring (Loesch 1969; Kissil 1974).

Although most juveniles emigrate offshore during their first year, some overwinter in the Chesapeake (Hildebrand 1963) and Delaware bays (Smith 1971). Marcy (1969) suggested that many juveniles (age-1+) spend their first winter close to the mouth of their natal river due to their presence in the lower portion of the Connecticut River in early spring. Other researchers concur that some juvenile alewife may remain in deep estuarine waters through the winter (Hildebrand and Schroeder 1928). There is some indication that alewife in northern states may remain in inshore waters for one to two years (Walton 1981). Conversely, since juvenile river herring cannot survive water temperatures of 3°C or below (Otto et al. 1976), they likely do not overwinter in coastal systems where temperatures are below 3°C (Kosa and Mather 2001).

Juveniles and the saltwater interface

Richkus (1974) reported that juvenile alewife that were transferred from freshwater to saline water (32 ppt), and vice versa, experienced zero mortality. Juvenile alewife in the upper Chesapeake Bay were found in salinities ranging from 0 to 8 ppt, but most (82%) were collected from freshwater (Dovel 1971). Furthermore, Pardue (1983) suggested that salinities less than or equal to 5 ppt are optimal for juveniles of this species.

Juvenile substrate associations

Olney and Boehlert (1988) found juvenile alewife among submerged aquatic vegetation (SAV) beds of the lower Chesapeake Bay and suggested that SAV likely confers some level of protection from predation. No other information was available regarding substrate preferences for juvenile alewife.

Juvenile depth associations

Jessop (1990) reported that juvenile alewife were completely absent from near-surface water during daylight hours. No other information was available regarding depth preferences or optima for juvenile alewife.

Juvenile water temperature

Characterization	Acclimation Temp (°C)	Temp Range (°C)	Location	Citation
Optimal	N/A	15 - 20	Many	Pardue 1983
Suitable	N/A	10 - 28	Many	Klauda et al. 1991
Present	N/A	4 - 27	Upper Chesapeake Bay	Dovel 1971
Present	N/A	13.5 – 29.0	Cape Fear River, NC	Davis and Cheek 1966
Avoidance	26	>34	Delaware River	PSECG 1984
Preferred	15 - 21	17 – 23 (at 4 – 7 ppt)	Delaware River	Meldrim and Gift 1971; PSE&G 1982
Preferred	15 - 18	25.0	Lake Michigan	Otto et al. 1976

Table 4-4. Juvenile alewife temperature tolerances/preferences along the Atlantic coast

Temperature tolerance range estimates for juvenile alewife vary somewhat between researchers (Table 4-4). Dovel (1971) found that ninety-eight percent of juvenile alewife in the upper Chesapeake Bay were collected at 25°C.

According to McCauley and Binkowski (1982), the upper lethal temperature for juvenile alewife is approximately 30°C. Concurrently, in Lake Michigan, upper incipient lethal limits (i.e., temperature at which 50% of the population survives) for young-of-the-year alewife acclimated to 10°C, 20°C, and 25°C, was estimated to be slightly less than 26.5°C, 30.3°C, and 32.1°C, respectively (Otto et al. 1976). Another study found that juveniles exposed to water at 35°C for 24 hours, after acclimation to water at 18.9 to 20.6°C, had a 20% survival rate (Dorfman and Westman 1970). Moreover, young-of-the-year alewife seem to have critical thermal maxima (CTM) that are 3 to 6°C higher than adults (Otto et al. 1976).

Alternatively, when juvenile alewife were subjected to decreasing temperatures (15.6°C down to 2.8°C) over the course of 15 days, they suffered greater than 90% mortality (Colby 1973). In another study, juvenile alewife exposed to 9°C, following acclimation at 20°C in 5.5 ppt salinity, suffered no mortality. However, when the temperature was decreased to 7°C for 96 h, they suffered 27 to 60% mortality (PSE&G 1984). Comparatively, the lower limit at which juvenile river herring are unable to survive is 3°C or less (Otto et al. 1976).

Juvenile dissolved oxygen associations

Jones et al. (1988) determined that the minimum dissolved oxygen concentration for juveniles is 3.6 mg/L. Dorfman and Westman (1970) reported that at dissolved oxygen

concentrations below 2.0 mg/L, juvenile alewife became physically stressed. At concentrations as low as 0.5 mg/L, juveniles survived for approximately five minutes in oxygen (Dorfman and Westman 1970). In the Cape Fear River system, juveniles preferred waters where dissolved oxygen levels ranged from 2.4 to 10.0 mg/L (Davis and Cheek 1966).

Juvenile pH and aluminum associations

Kosa and Mather (2001) reported that juvenile river herring abundance peaks at a pH of 8.2 in coastal systems in Massachusetts, and suggest that that pH appears to contribute to variations in juvenile abundance.

Juvenile water velocity/flow

Water discharge is an important variable influencing relative abundance and emigration of juvenile alewife. Extremely high discharges may adversely affect juvenile emigration, and high or fluctuating discharges may lead to a decrease in the relative abundance of adults and juveniles (Kosa and Mather 2001). Laboratory experiments suggest that juvenile alewife avoid water velocities greater than 10 cm/s, especially in narrow channels (Gordon et al. 1992). In large rivers where greater volumes of water can be transported per unit of time without substantial increases in velocity, the effects of discharge may differ (Kosa and Mather 2001).

Kissil (1974) observed juvenile alewife leaving Lake Bride, Connecticut, between June and October; they noted especially high migration occurring during times of heavy water flow. These results are consistent with Cooper's (1961) observations that 98% of juveniles left after periods of heavy rainfall. Huber (1978) also noted that juvenile emigration in the Parker River, Massachusetts, was triggered by an increase in water flow. Furthermore, Jessop (1994) found that the juvenile abundance index (JAI) of alewife decreased with mean river discharge during the summer. Daily instantaneous mortality also increased with mean river discharge from July to August at the Mactaquac Dam headpond on the Saint John River, New Brunswick, Canada (Jessop 1994).

Juvenile feeding behavior

Juvenile alewife are opportunistic feeders that usually favor seasonally available items (Gregory et al. 1983). For example, in the Hamilton Reservoir, Rhode Island, juveniles feed primarily on dipteran midges in July, and cladocerans in August and September (Vigerstad and Colb 1978). Juveniles either select their prey individually or switch to a non-selective filter-feeding mode, which is a behavior utilized more at night (Janssen 1976). Grabe (1996) found that juvenile alewife fed on chironomids, odonates, and other amphipods during the day and early evening hours in the Hudson River. Juveniles have also been observed consuming epiphytic fauna especially at night (Weaver 1975; Grabe 1996). Juveniles may also feed extensively on benthic organisms, including ostracods, chironomid larvae, and oligochaete worms (Watt and Duerden 1974).

The number of zooplankton per liter consumed is assumed to be critical for the survival and growth of juvenile alewife. Pardue (1983) suggests that habitats containing 100 or more zooplankton per liter are optimal. Walton (1987) found that juvenile alewife abundance in

Damariscotta Lake, Maine, was controlled by competition for zooplankton, rather than parental stock abundance and recruitment. It has been suggested that clupeids evolved to synchronize the larval stage with the optimal phase of annual plankton production cycles (Blaxter and Hunter 1982). In addition, Morsell and Norden (1968) found that juvenile alewife consume zooplankton until they reach 12 cm TL, and may then switch to increasing amounts of the benthic amphipod *Pontoporeia* sp. Several researchers (Vigerstad and Colb 1978; O'Neill 1980; Yako 1998) hypothesize that a change in food availability may provide a cue for juvenile anadromous herring to begin emigrating seaward, but no causal link has been established.

Unfortunately, invasive species may threaten food sources for alewife. There is strong evidence that juveniles in the Hudson River have experienced a reduced forage base as a result of zebra mussel colonization (Waldman and Limburg 2003).

Juvenile competition and predation

It is often noted throughout the literature that alewife and blueback herring co-exist in the same geographic regions, yet interspecific competition is often reduced through several mechanisms. For example, juveniles of both species may consume different sizes of prey (Crecco and Blake 1983). Juvenile alewife in the Minas Basin, Nova Scotia, Canada, favor larger benthic prey (particulate-feeding strategy) compared to juvenile blueback herring (filter-feeding strategy) (Stone 1985; Stone and Daborn 1987). In the Cape Fear River, North Carolina, juvenile alewife consume more ostracods, insect eggs, and insect parts than blueback herring (Davis and Cheek 1966).

Alewife also spawn earlier than blueback herring, thereby giving juvenile alewife a relative size advantage over juvenile bluebacks, allowing them a larger selection of prey (Jessop 1990). Differences in juvenile diel feeding activity further reduce competition. One study noted that diurnal feeding by juvenile alewife was bimodal, with peak consumption about one to three hours before sunset and a minor peak occurring about two hours after sunrise (Weaver 1975). In comparison, juvenile blueback herring begin to feed actively at dawn, increasing throughout the day and maximizing at dusk, then diminishing from dusk until dawn (Burbidge 1974).

With regard to predation, juvenile alewife are consumed by American eel, white perch, yellow perch, grass pickerel, largemouth bass, pumpkinseed, shiners, walleye and other fishes, as well as turtles, snakes, birds, and mink (Kissil 1969; Colby 1973; Loesch 1987). In the estuarine waters of Maine, juvenile bluefish prey heavily on alewife (Creaser and Perkins 1994). In Massachusetts rivers, juvenile alewife are energetically valuable and a key food source for largemouth bass during late summer (Yako et al. 2000).

Juveniles and contaminants

A 24-hour LC_{50} (i.e., concentration at which 50% of the population dies) of 2.25 mg/L for total residual chlorine (TRC) was reported for juvenile alewife exposed for 30 minutes at 10°C (Seegert and Latimer 1977). Thirty-minute LC_{50} values for TRC were 2.27 and 0.30 mg/L for juveniles exposed at 10°C and 30°C, respectively (Brooks and Seegert 1978; Seegert and Brooks 1978). Juvenile alewife held at 15°C in 7 ppt salinity exhibited an avoidance response to 0.06 mg/L TRC (PSE&G 1980). Juveniles held at 19 to 24°C in freshwater exhibited an

avoidance response at <0.03 mg/L TRC; fish subjected to 0.48 mg/L TRC for 2 hours at 22°C suffered 100% mortality (Bogardus et al. 1978).

Part D. Alewife Late Stage Juvenile and Adult Marine Habitat

Geographical and temporal patterns at sea

Some young-of-the-year alewife over-winter in deep, high salinity areas of the Chesapeake Bay (Hildebrand and Schroeder 1928). Dovel (1971) reported juvenile populations in the upper Chesapeake Bay that did not emigrate until early spring of their second year. Milstein (1981) found that juvenile alewife over-wintered in waters approximately 0.6 to 7.4 km from the shore of New Jersey, at depths of 2.4 to 19.2 m, in what is considered an offshore estuary. This area is warmer with higher salinity than the cooler, lower salinity river-bay estuarine nurseries where alewife reside in fall. The majority of alewife are present in March when bottom temperatures range from 4.4 to 6.5°C and salinity is between 29.0 and 32.0 ppt (Cameron and Pritchard 1963).

Young alewife have been found overwintering off the North Carolina coast from January to March, concentrated at depths of 20.1 to 36.6 m (Holland and Yelverton 1973; Street et al. 1973). However, other sources have noted that juvenile alewife tend to remain near the surface during their first year in saltwater (Bigelow and Schroeder 1953). In Lake Michigan, age-1 fish are usually pelagic, except in spring and fall, where they often occur on the bottom; age-2 fish are typically found on the bottom (Wells 1968).

Information on the life history of young-of-the-year and adult alewife after they emigrate to the sea is sparse (Klauda et al. 1991). Sexual maturity of alewife is reached at a minimum of age-2, but timing may vary regionally. In North Carolina, sexual maturity occurs mostly at age-3. In Connecticut, most males achieve maturity at age-4, and most females at age-5 (Jones et al. 1978). It is generally accepted that juveniles join the adult population at sea within the first year of their lives and follow a north-south seasonal migration along the Atlantic coast, similar to that of American shad (Neves 1981). Despite a lack of conclusive evidence, it is thought that alewife are similar to other anadromous clupeids in that they may undergo seasonal migrations within preferred isotherms (Fay et al. 1983). In fact, alewife typically migrate in large schools of similar sized fish, and may even form mixed schools with other herring species (Colette and Klein-MacPhee 2002).

During spring, alewife from the Mid-Atlantic Bight move inshore and north of 40° latitude to Nantucket Shoals, Georges Bank, coastal Gulf of Maine, and the inner Bay of Fundy. Commercial catch data indicates that alewife are most frequently caught on Georges Bank and south of Nantucket Shoals (Neves 1981; Rulifson et al. 1987). Distribution in the fall is similar to the summer, but alewife concentrate along the northwest perimeter of the Gulf of Maine. In the fall, individuals move offshore and southward to the mid-Atlantic coast between latitude 40°N and 43°N, where they remain until early spring (Neves 1981). It is not known to what extent alewife overwinter in deep water off the continental shelf, but they have rarely been found more than 130 km from the coast (Jones et al. 1978).

Alewife also experience diel movement patterns. At sea alewife are more available to bottom trawling gear during the day, suggesting that they follow the diel movement of plankton in the water column and are sensitive to light (Neves 1981). It also seems that feeding and vertical migration are likely controlled by light intensity patterns within thermal preference zones (Richkus and Winn 1979; Neves 1981).

Results from Canadian spring surveys show river herring distributed along the Scotian Gulf, southern Gulf of Maine, and off southwestern Nova Scotia from the Northeast Channel to the central Bay of Fundy; they are found to a lesser degree along the southern edge of Georges Bank and in the canyon between Banquereau and Sable Island Banks (Stone and Jessop 1992). A large component of the overwintering population on the Scotian Shelf (and possibly some of the U.S. Gulf of Maine population) moves inshore during spring to spawn in Canadian waters. Summer aggregations of river herring in the Bay of Fundy/eastern Gulf of Maine may consist of a mixture of stocks from the entire Atlantic coast, as do similar aggregations of American shad (Dadswell et al. 1987). However, based on commercial offshore catches by foreign fleets in the late 1960s, it was believed that coastal river herring stocks did not mingle to the extent that American shad stocks apparently did, at least during the seasons that foreign harvests were made (ASMFC 1985).

Salinity associations at sea

As noted above, young-of-the-year alewife have been found over-wintering offshore of New Jersey, where salinities range from 29.0 to 32.0 ppt (Milstein 1981). For sub-adults and non-spawning adults that remain in the open ocean, they will reside in full strength seawater. Since alewife may follow a north-south seasonal migration along the Atlantic coast similar to that of American shad (Neves 1981), and pre-spawning adult American shad may detour into estuaries (Neves and Depres 1979), alewife may inhabit more brackish waters during migration.

Depth associations at sea

National Marine Fisheries Service catch data found that in offshore areas, alewife were caught most frequently in waters with depths of 56 to 110 m. The vertical position of alewife in the water column may be influenced by zooplankton concentrations (Neves 1981). Zooplankton usually concentrate at depths <100 m in the Gulf of Maine (Bigelow 1926). Stone and Jessop (1992) found that alewife offshore of Nova Scotia, the Bay of Fundy, and the Gulf of Maine, were at depths of 101 to 183 m in the spring; they were in shallower nearshore waters (46 to 82 m) in the summer, and in deeper offshore waters (119 to 192 m) in the fall.

Stone and Jessop (1992) also found differences in depth distribution between smaller fish (sexually immature) and larger fish. Smaller fish occurred in shallow regions (<93 m) during spring and fall, while larger fish were found in deeper areas (≥93 m) throughout the year (Stone and Jessop 1992). Furthermore, Jansen and Brandt (1980) reported that the nocturnal depth distribution of adult landlocked alewife differed by size class, with the smaller fish present at shallower depths.

Interestingly, in coastal waters juvenile alewife are found in deeper water than blueback herring despite their identical diets (Davis and Cheek 1966; Burbidge 1974; Watt and Duerden 1974; Weaver 1975).

Temperature associations at sea

From Cape Hatteras to Nova Scotia, alewife have been caught offshore where surface water temperatures ranged from 2 to 23°C and bottom water temperatures ranged from 3 to

17°C. Catches in this area were most frequent where the average bottom water temperature was between 4 and 7°C (Neves 1981). Stone and Jessop (1992) reported a temperature range of 7 to 11°C for alewife in the northern range off Nova Scotia, the Bay of Fundy, and the Gulf of Maine. The researchers also noted that the presence of a cold (<5°C) intermediate water mass over warmer, deeper waters on the Scotian Shelf, where the largest catches of river herring occurred, may have restricted the extent of vertical migration during the spring. Since few captures were made where bottom temperatures were <5°C, vertical migration may have been confined by a water temperature inversion in this area during the spring (Stone and Jessop 1992).

Alewife may prefer and be better adapted to cooler water than blueback herring (Loesch 1987; Klauda et al. 1991). Northern populations may also exhibit more tolerance to cold temperatures (Stone and Jessop 1992). Additionally, antifreeze activity was found in blood serum from an alewife off Nova Scotia, but not in any captured in Virginia (Duman and DeVries 1974).

Feeding behavior at sea

At sea, alewife feed largely on particulate zooplankton including euphausiids, calanoid copepods, mysids, hyperiid amphipods, chaetognaths, pteropods, decapod larvae, and salps (Edwards and Bowman 1979; Neves 1981; Vinogradov 1984; Stone and Daborn 1987; Bowman et al. 2000). Alewife also consume small fishes, including Atlantic herring, other alewife, eel, sand lance, and cunner (Colette and Klein-MacPhee 2002). They feed either by selectively preying on individuals or non-selectively filter-feeding with gill rakers. Feeding mode depends mostly on prey density, prey size, and water visibility, as well as size of the alewife (Janssen 1976, 1978a, 1978b). In Minas Basin, Bay of Fundy, alewife diets shift from micro-zooplankton in small fish to mysids and amphipods in larger fish. Feeding intensity also decreases with increasing age of fish (Stone 1985).

Alewife generally feed most actively during the day; nighttime predation is usually restricted to larger zooplankton that are easier to detect (Janssen 1978b; Janssen and Brandt 1980; Stone and Jessop 1993). In Nova Scotia, alewife feeding peaks at midday during the summer and mid-afternoon during the winter. Alewife also have a higher daily ration in the summer than in the winter (Stone and Jessop 1993). Although direct evidence is lacking, alewife catch in specific areas along Georges Bank, the perimeter of the Gulf of Maine, and south of Nantucket Shoals, may be related to zooplankton abundance (Neves 1981).

Competition and predation at sea

Schooling fish such as bluefish, weakfish, and striped bass, prey upon alewife (Bigelow and Schroeder 1953; Ross 1991). Other fish such as dusky shark, spiny dogfish, Atlantic salmon, goosefish, cod, pollock, and silver hake, also prey on alewife (Bowman et al. 2000; R. Rountree, University of Massachusetts, unpublished data). Of these species, spiny dogfish appears to have the greatest affinity for alewife (R. Rountree, University of Massachusetts, unpublished data). Also, see Part C of this chapter for additional information.

Section II. Significant Environmental, Temporal, and Spatial Factors Affecting Distribution of Alewife

Table 4-5. Significant environmental, temporal, and spatial factors affecting distribution of alewife. Please note that, although there may be subtle variations between systems, the following data include a broad range of values that encompass the different systems that occur along the East Coast. Where a specific range is known to exist, it will be noted. For the subadult–estuarine/oceanic environment and non-spawning adult–oceanic environment life history phases, the information is provided as a general reference, not as habitat preferences or optima. NIF = No Information Found.

Life Stage	Time of Year and Location	Depth (m)	Temperature (°C)	Salinity (ppt)	Substrate	Current Velocity (m/sec)	Dissolved Oxygen (mg/L)
Spawning Adult	Late February (south) through August (north); slow-moving sections of streams/ponds/lakes, and shorebank eddies or deep pools, from North Carolina to Labrador & Newfoundland	**Tolerable:** 0.2-3 **Optimal:** NIF **Reported:** Typically spawn in shallower (<1) areas	**Tolerable:** 7-27.8 **Optimal:** 13-20 **Reported:** Broad range; disagreement on minimum temperature for spawning	**Tolerable:** NIF **Optimal:** NIF **Reported:** Migrate as far upstream in freshwater as possible	**Tolerable:** NIF **Optimal:** NIF **Reported:** Usually sand, gravel, cobble, and other coarse stone; some report SAV and detritus	**Tolerable:** NIF **Optimal:** NIF **Reported:** Slow-moving waters	**Tolerable:** >5.0 **Optimal:** NIF **Reported:** Only tolerate low DO for short periods
Egg	Late February (south) through August (north); hatch 50-360 hours after fertilization, but usually within 80-95 hours at spawning site or slightly downstream	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** 10.6-26.7 **Optimal:** 17.2-21.1 **Reported:** Average time to median hatch varies inversely w/temperature	**Tolerable:** NIF **Optimal:** 0-2 **Reported:** Mostly found in freshwater	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** Usually found in low flow; w/DO, strongest predictor of egg presence	**Tolerable:** ≥5.0 **Optimal:** NIF **Reported:** With velocity, strongest predictor of egg presence
Prolarvae	Hatch in 50 to 360 hours, but usually within 80-95 hours downstream of spawning site	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** 8-31 **Optimal:** 15-24 **Reported:** Variable	**Tolerable:** NIF **Optimal:** 0-3 **Reported:** Mostly found in freshwater	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** Usually found in low flow	**Tolerable:** ≥5.0 **Optimal:** NIF **Reported:** NIF

Life Stage	Time of Year and Location	Depth (m)	Temperature (°C)	Salinity (ppt)	Substrate	Current Velocity (m/sec)	Dissolved Oxygen (mg/L)
Postlarvae	2 to 5 days downstream of spawning site after prolarvae stage is reached	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** 14-28 **Optimal:** 20-26 **Reported:** Variable	**Tolerable:** NIF **Optimal:** NIF **Reported:** Larvae grow faster in saltwater	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** Usually found in low flow	**Tolerable:** ≥5.0 **Optimal:** NIF **Reported:** NIF
Early Juvenile – Riverine	3-9 months in natal rivers after reaching juvenile stage in brackish waters or upstream in freshwater	**Tolerable:** NIF **Optimal:** NIF **Reported:** Absent from near-surface during daylight	**Tolerable:** 10-28 **Optimal:** 15-20 **Reported:** 4-29	**Tolerable:** NIF **Optimal:** NIF **Reported:** Variable	**Tolerable:** NIF **Optimal:** NIF **Reported:** SAV for protection	**Tolerable:** NIF **Optimal:** NIF **Reported:** Avoid >10cm/s; high migration rates in heavy flow	**Tolerable:** ≥3.6 **Optimal:** NIF **Reported:** NIF
Subadult & Non-spawning Adult – Estuarine/ Oceanic	2-5 years after hatching in nearshore estuarine waters or offshore marine waters	**Tolerable:** NIF **Optimal:** 46-192 **Reported:** Zooplankton may influence depth; smaller fish generally in shallower water	**Tolerable:** 2-23 **Optimal:** 4-7 (bottom temp) **Reported:** Northern populations may be more cold tolerant	**Tolerable:** NIF **Optimal:** NIF **Reported:** Brackish to saltwater	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF

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Chapter 5

BLUEBACK HERRING

(*Alosa aestivalis*)

Section I. Blueback Herring Description of Habitat

Blueback Herring General Habitat Description and Introduction

Blueback herring (*Alosa aestivalis*) are an anadromous, highly migratory, euryhaline, pelagic, schooling species. Both blueback herring and alewife are often referred to as "river herring," which is a collective term for these two often inter-schooling species (Murdy et al. 1997). This term is often used generically in commercial harvests with no distinction between the two species (ASMFC 1985); to further this lumping tendency, landings for both species are reported as alewife (Loesch 1987). Blueback herring spend most of their lives at sea, returning to freshwater only to spawn (Colette and Klein-MacPhee 2002). Their range is commonly cited as spanning from the St. Johns River, Florida (Hildebrand 1963; Williams et al. 1975) to Cape Breton, Nova Scotia (Scott and Crossman 1973) and the Miramichi River, New Brunswick (Bigelow and Schroeder 1953; Leim and Scott 1966). However, Williams et al. (1975) have reported that blueback herring occur as far south as Tomoka River, a small freshwater tributary of the Halifax River in Florida (a brackish coastal lagoon). Additionally, some landlocked populations occur in the Southeast (Klauda et al. 1991), but landlocking occurs less frequently in blueback herring than in alewife (Schmidt et al. 2003).

Blueback herring from the South are capable of migrating extensive distances (over 2000 km) along the Atlantic seaboard, and their patterns of migration may be similar to those of American shad (Neves 1981). This species is most abundant south of the warmer waters of the Chesapeake Bay (Manooch 1988; Scott and Scott 1988), occurring in virtually all tributaries to the Chesapeake Bay, the Delaware River, and in adjacent offshore waters (Jones et al. 1978). Although blueback herring and alewife co-occur throughout much of their range, blueback herring are more abundant by one or perhaps two orders of magnitude along the middle and southern parts of their ranges (Schmidt et al. 2003).

Several long-term data sets were recently analyzed to determine the current status of blueback herring in large river systems along the East Coast, including the Connecticut, Hudson, and Delaware rivers. Blueback herring show signs of overexploitation in all of these rivers, including reductions in mean age, decreases in percentage of returning spawners, and decreases in abundance. Although researchers did not include smaller drainages in the analysis, they did note that some runs in the northeastern U.S. and Atlantic Canada have observed increased population abundance of blueback herring in recent years (Schmidt et al. 2003).

Please note that some of the data presented in this chapter have been derived from studies of landlocked populations and the applicability of environmental requirements is unknown; therefore, they should be interpreted with discretion (Klauda et al. 1991).

Part A. Blueback Herring Spawning Habitat

Geographical and temporal patterns of migration

Adult blueback herring populations in the South return earliest to spawn in freshwater and sometimes brackish waters, with populations further north migrating inland later in the spring when water temperatures have increased. Researchers believe that blueback herring migrate inland from offshore waters north of Cape Hatteras, North Carolina, encountering the same thermal barrier as American shad. Individuals then turn south along the coast if they are homing to South Atlantic rivers (Neves 1981); northbound pre-spawning adults head north along the coast (Stone and Jessop 1992). Adults begin migrations from the offshore region in response to changes in water temperature and light intensity (Pardue 1983). It is assumed that adults return to the rivers in which they were spawned, but some may stray to adjacent streams or colonize new areas; some individuals have even reoccupied systems in which the species was previously extirpated (Messieh 1977; Loesch 1987).

Blueback herring will ascend freshwater far upstream (Massman 1953; Davis and Cheek 1966; Perlmutter et al. 1967; Crecco 1982); their distribution is a function of habitat suitability and hydrological conditions, such as swift flowing water (Loesch and Lund 1977). Earlier hypotheses that blueback herring do not ascend as far upstream as alewife are unfounded (Loesch 1987). In fact, in tributaries of the Rappahannock River, Virginia, upstream areas were found to be more important for blueback herring spawning than downstream areas (O'Connell and Angermeier 1997).

Spawning location (ecological)

Generally, blueback herring and alewife attempt to occupy different freshwater spawning areas. However, if blueback herring and alewife are forced to spawn in the same vicinity (i.e., due to blocked passage) (Loesch 1987), some researchers have suggested that the two species occupy separate spawning sites to reduce competition. For example, Loesch and Lund (1977) note that blueback herring typically select the main stream flow for spawning, while neighboring alewife spawn along shorebank eddies or deep pools. In rivers where headwater ponds are absent or poorly-developed, alewife may be most abundant farther upstream in headwater reaches, while blueback herring utilize the mainstream proper for spawning (Ross and Biagi 1990). However, in some areas blueback herring are abundant in tributaries and flooded low-lying areas adjacent to main streams (Erkan 2002).

In the allopatric range, where there is no co-occurrence with alewife (south of North Carolina), blueback herring select a greater variety of spawning habitat types (Street 1970; Frankensteen 1976; Christie 1978), including small tributaries upstream from the tidal zone (ASMFC 1999), seasonally flooded rice fields, small densely vegetated streams, cypress swamps, and oxbows, where the substrate is soft and detritus is present (Adams and Street 1969; Godwin and Adams 1969; Adams 1970; Street 1970; Curtis et al. 1982; Meador et al. 1984). Furthermore, despite the fact that blueback herring generally do not spawn in ponds in their northern range (possibly to reduce competition), they have the ability to do so (Loesch 1987).

Loesch (1987) has reported that blueback herring can adapt their spawning behavior under certain environmental conditions and disperse to new areas if the conditions are suitable.

This behavior was demonstrated in the Santee-Cooper System, South Carolina, where hydrological alterations resulting from the creation of a rediversion canal led to changes in spawning site selection in both rivers. In the Cooper River, blueback herring lost access to formerly impounded rice fields along the river, which were important spawning areas. Following the construction of the rediversion canal, there was an increase in the number and length of tributaries along the river that were used as spawning habitat. In the adjacent Santee River, adults dispersed into the rediversion canal itself in favor of their former habitat, which was further upstream (Eversole et al. 1994).

Temporal spawning patterns

Spawning of blueback herring typically commences in the given regions at the following times: 1) Florida – as early as December (McLane 1955); 2) South Carolina (Santee River) – present in February (Bulak and Christie 1981), but spawning begins in early March (Christie 1978; Meador 1982); 3) Chesapeake Bay region - lower tributaries in early April and upper reaches in late April (Hildebrand and Schroeder 1928); 4) Mid-Atlantic region – late April (Smith 1971; Zich 1978; Wang and Kernehan 1979); 5) Susquehanna River - abundance peaks in early to mid-May (R. St. Pierre, U.S. Fish and Wildlife Service, personal communication); 6) Connecticut River – present in lower river mid-April, but spawning begins in mid-May (Loesch and Lund 1977); and 7) Saint John River, New Brunswick – present in May (Messieh 1977; Jessop et al. 1983), but spawning doesn't commence until June and may run through August (Leim and Scott 1966; Marcy 1976b).

Blueback herring generally spawn 3 to 4 weeks after alewife in areas where they co-occur; however, there may be considerable overlap (Loesch 1987) and peak spawning periods may differ by only 2 to 3 weeks (Hildebrand and Schroeder 1928). In a tributary of the Rappahannock River, Virginia, researchers found that blueback eggs and larvae were more abundant than those of alewife, but that alewife used the stream over a longer period of time. In addition, there was only a three- day overlap of spawning by alewife and blueback herring (O'Connell and Angermeier 1997). Although it has been suggested that alewife and blueback herring select separate spawning sites in sympatric areas to reduce competition (Loesch 1987), O'Connell and Angermeier (1997) did not find that the two species used different spawning habitat in the areas they examined. The researchers suggested that there was a temporal, rather than spatial, segregation that minimized the competition between the two species (O'Connell and Angermeier 1997).

Spawning may occur during the day, but blueback herring spawning activity is normally most prolific from late afternoon (Loesch and Lund 1977) into the night (Johnston and Cheverie 1988). During spawning, a female and two or more males will swim approximately one meter below the surface of the water; subsequently, they will dive to the bottom (Loesch and Lund 1977), simultaneously releasing eggs and sperm over the substrate (Colette and Klein-MacPhee 2002). Spawning typically occurs over an extended period, with groups or "waves" of migrants staying 4 to 5 days before rapidly returning to sea (Hildebrand and Schroeder 1928; Bigelow and Schroeder 1953; Klauda et al. 1991). In a temporal context, the majority of spent adult blueback herring emigrating from the Connecticut River moved through fish passage facilities between 1700 and 2100 hours (Taylor and Kynard 1984).

Maturation and spawning periodicity

Blueback herring are repeat spawners at an average rate of 30 to 40% (Richkus and DiNardo 1984). In general, there appears to be an increase in repeat spawning from south to north (Rulifson et al. 1982). Researchers have found that approximately 44 to 65% of the blueback herring in Chesapeake Bay tributaries had previously spawned (Joseph and Davis 1965), while 75% of those in Nova Scotia had previously spawned (O'Neill 1980). In the Chowan River, North Carolina, as many as 78% of individuals were first-time spawners (Winslow and Rawls 1992). First spawning occurs when adults are between 3 and 6 years old, but most first-time spawners are age 4 fish (Messieh 1977; Loesch 1987). Joseph and Davis (1965) reported that some blueback herring spawn as many as six times in Virginia.

Jessop (1990) found a stock-recruitment relationship for the spawning stock of river herring and year-class abundance at age 3. Despite these results, most studies have been unable to detect a strong relationship between adult and juvenile abundance of clupeids (Crecco and Savoy 1984; Henderson and Brown 1985; Jessop 1994). Researchers have suggested that although year-class is driven mostly by environmental factors, if the parent stock size falls below a critical level, the size of the spawning stock may become a factor in determining juvenile abundance (Kosa and Mather 2001). To the extent that environmental factors have been linked to year-class abundance, they will be discussed in subsequent sections.

Spawning and the saltwater interface

Blueback herring generally spawn in freshwater above the head of tide; brackish and tidal areas are rarely used for spawning by this species (Nichols and Breder 1927; Hildebrand 1963; Fay et al. 1983; Murdy et al. 1997). Adults, eggs, larvae, and juveniles can tolerate a wide range of salinities, but seem to prefer a more narrow range, depending on life history stage. For example, while spawning may occur in salinities ranging from 0 to 6 ppt, it typically takes place in waters that are less than 1 ppt (Klauda et al. 1991). Boger (2002) presented a modified salinity range for Virginia rivers, suggesting that a suitable salinity range for spawning adults is 0 to 5 ppt. Alternatively, spawning adult blueback herring have been found in brackish ponds at Woods Hole, Massachusetts (Nichols and Breder 1927; Hildebrand 1963).

Spawning substrate associations

In areas where blueback herring and alewife co-occur (sympatric region), blueback herring prefer to spawn over gravel and clean sand substrates where the water flow is relatively swift, and actively avoid areas with slow-moving or standing water (Bigelow and Welsh 1925; Marcy 1976b; Loesch and Lund 1977; Johnston and Cheverie 1988).

In the allopatric range, there seems to be some variation in blueback herring spawning substrate. Where water flow is more sluggish, there is ample opportunity for detritus and silt to accumulate. Pardue (1983) considered substrates with 75% or more silt and other soft materials (e.g., detritus and vegetation) as optimal for blueback herring spawning because it provides cover for eggs and larvae. However, more recently Boger (2002) found that river herring spawning areas along the Rappahannock River, Virginia, had substrates that consisted primarily of sand, pebbles, and cobbles (usually associated with higher-gradient streams), while areas with little or

no spawning were dominated by organic matter and finer sediments (usually associated with lower-gradient streams and comparatively more agricultural land use).

Spawning depth associations

During their freshwater migration, blueback herring swim at mid-water depths (compared to deeper water used by American shad) (Witherell 1987). This species is reported to spawn in both shallow (Jones et al. 1978) and deep streams (Johnston and Cheverie 1988).

Spawning water temperature

O'Connell and Angermeier (1997) found that temperature was the strongest predictor of blueback herring adult and early egg presence in a tributary of the Rappahannock River, Virginia. Blueback herring are reported to spawn at temperatures ranging from a minimum of 13°C (Hawkins 1979; Rulifson et al. 1982) to a maximum of 27°C (Loesch 1968). Loesch and Lund (1977) noted that spawning adults were found in the lower Connecticut River in mid-April when water temperatures were as low as 4.7°C, but spawning did not occur until several weeks later when the water temperature had risen. Meador et al. (1984) noted that rapid changes in water temperature appeared to be an important factor influencing the timing of spawning. Optimal spawning temperature range is suggested to be 21 to 25°C (Cianci 1969; Marcy 1976b; Klauda et al. 1991) and 20 to 24° C (Pardue 1983). Fish in the laboratory acclimated to 15°C and 29 ppt salinity exhibited a final temperature preference of 22.8°C (Terpin et al. 1977).

Spawning dissolved oxygen associations

Adult blueback herring require a minimum of 5.0 mg/L of dissolved oxygen (Jones et al. 1978). For example, adults caught in the Cooper and Santee Rivers, South Carolina, were always captured in areas that had a dissolved oxygen concentration of 6 mg/L or higher (Christie et al. 1981).

Spawning water velocity/flow

In the sympatric range, blueback herring prefer to spawn in large rivers and tributaries where the water flow is relatively swift, actively avoiding areas with slow-moving or standing water (Bigelow and Welsh 1925; Marcy 1976b; Johnston and Cheverie 1988). In such areas, blueback herring will concentrate and spawn in the main-stream flow, while alewife favor shorebank eddies or deep pools for spawning (Loesch and Lund 1977). In Connecticut, blueback herring select the fast-moving waters of the upper Salmon River and Roaring Brook, while alewife are found in the slower-moving waters of Higganum and Mill creeks (Loesch and Lund 1977) and Bride Lake (Kissil 1974). Researchers suggest that there is differential selection of spawning in these areas (Loesch and Lund 1977).

In the allopatric range, blueback herring favor lentic sites, but may also occupy lotic sites (Loesch 1987; Klauda et al. 1991). Additionally, they may select slower-flowing tributaries and flooded low-lying areas adjacent to main streams with soft substrates and detritus (Street et al. 1975; Sholar 1975, 1977; Fischer 1980; Hawkins 1979).

Meador et al. (1984) found that high flows (and accompanying low water temperatures) associated with flood control discharges in the Santee River, South Carolina, immediately prior to the spawning season, resulted in lower numbers of blueback herring larvae that year. In the preceding year without flood control discharges, spawning occurred farther upstream (Meador et al. 1984). Furthermore, ripe adults were found below the sampling site heading downstream the year that high flows occurred, apparently without having spawned (Bulak and Christie 1981). Concurrently, other studies (Bulak and Curtis 1977; West et al. 1988) have found spawning adults moving downstream from spawning areas following a sudden change in water discharge.

In a similar example in the same river system, a rediversion canal and hydroelectric dam with a fish passage facility were constructed between the Cooper River and Santee River, which increased the average flow of the Santee River from 63 m^3/s to 295 m^3/s (Cooke and Leach 2003). Following the rediversion, blueback herring did not concentrate below the dam and few were attracted into the fish lock during periods of zero discharge. Too much water flow also posed a problem, as adults were found concentrating below the dam during periods of discharge, but were unable to locate the entrance to the fish lock due to high turbulence (Chappelear and Cooke 1994). As a result, blueback herring changed migration patterns by abandoning the Santee River, and following the dredged canal to the higher flow of the St. Stephen Dam. Subsequently, access to spawning grounds was increased, which contributed to increases in blueback herring populations (Cooke and Leach 2003). Although the importance of instream flow requirements has been previously recognized (Crecco and Savoy 1984; ASMFC 1985; Crecco et al. 1986; Ross et al. 1993), it has usually been with regard to spawning habitat requirements or recruitment potential (Moser and Ross 1994). Cooke and Leach (2003) concluded that the study of, and possible adjustment of, river flow may be an important consideration for restoring alosine habitat.

Spawning pH and aluminum associations

Adult blueback herring captured in the Santee-Cooper River system, South Carolina, were found within a range of pH 6.0 to 7.5 (Christie and Barwick 1985; Christie et al. 1981). Further north, within tributaries of the Delaware River, New Jersey, spawning runs were found within a broader range of pH 4.7 to 7.1 (mean pH 6.2) (Byrne 1988). Based on suggested ranges for eggs (cited in Klauda et al. 1991), Boger (2002) suggested a suitable range of pH 6 to 8, and an optimal range of pH 6.5 to 8 for spawning habitat.

Spawning feeding behavior

Adult blueback herring feed during upstream spawning migrations (Rulifson et al. 1982; Frankensteen 1976), consuming large and diverse quantities of copepods, cladocerans, ostracods, benthic and terrestrial insects, molluscs, fish eggs, hydrozoans, and stratoblasts (Creed 1985). Sampling of adult blueback herring along the St. Johns River, Florida, found that they also consume vegetation (FWC 1973).

Spawning competition and predation

Information is lacking that identifies which predator species prey on adult blueback herring during their spawning runs, but it is assumed that they are consumed by other fish, reptiles (e.g., snakes and turtles), birds (e.g., ospreys, eagles, and cormorants), and mammals (e.g., mink) (Loesch 1987; Scott and Scott 1988). Erkan (2002) notes that predation of alosines has increased dramatically in Rhode Island rivers in recent years, especially by the double-crested cormorant, which often takes advantage of fish staging near the entrance to fishways. Populations of nesting cormorant colonies have increased in size and have expanded into areas in which they were not previously observed. Predation by otters and herons has also increased, but to a lesser extent (Erkan 2002).

Several researchers have found evidence of striped bass predation on blueback herring (Trent and Hassler 1966; Manooch 1973; Gardinier and Hoff 1982). A recent study by Savoy and Crecco (2004) strongly supports the hypothesis that striped bass predation in the Connecticut River on adult blueback herring has resulted in a dramatic and unexpected decline in blueback herring abundance since 1992. The researchers further suggest that striped bass prey primarily on spawning adults because their predator avoidance capability may be compromised at that time, due to the strong drive to spawn during upstream migration. Rates of predation on age 0 and 1 alosines was much lower than that of adults (Savoy and Crecco 2004).

Part B. Blueback Herring Egg and Larval Habitat

Geographical and temporal movement patterns

On average, blueback herring eggs are hatched within 38 to 60 hours of fertilization (Adams and Street 1969). Yolk-sac larvae drift passively downstream with the current to slower moving water, where they grow and develop into juveniles (Johnston and Cheverie 1988). Yolk-sac absorption occurs in 2 to 3 days after hatching, and soon thereafter larvae begin to feed exogenously (Cianci 1969). Larvae are sensitive to light, so larval abundance at the surface increases as dusk approaches and reaches a maximum by dawn (Meador 1982).

Eggs, larvae, and the saltwater interface

Although spawning often occurs in freshwater, blueback herring eggs and larvae can survive in salinities as high as 18 to 22 ppt (Johnston and Cheverie 1988). Klauda et al. (1991) suggest an optimal range of 0 to 2 ppt for eggs only.

Egg and larval substrate associations

As with spawning habitat, Pardue (1983) suggested that substrates with 75% silt or other soft materials containing detritus and vegetation were optimal for egg and larval habitat. In contrast, Johnston and Cheverie (1988) found eggs adhered to sticks, stones, gravel, and aquatic vegetation along the bottom of a fast-flowing stream in the Gulf of St. Lawrence.

Egg and larval depth associations

Both Wang and Kernehan (1979) and Meador et al. (1984) observed that larval blueback herring achieved the greatest density at the surface during the night. This pattern of diel periodicity has also been described for the juvenile life stage of blueback herring (Loesch and Lund 1977; Loesch et al. 1982; Johnson et al. 1978).

Egg and larval water temperature

Blueback herring eggs were collected in the upper Chesapeake Bay where temperatures ranged from 7 to 14°C; 90% were collected at 14°C (Dovel 1971). Researchers did not report a significant reduction in hatching success for eggs acclimated at 15 to 18.3°C and exposed to temperatures of 22 to 28.3°C for 5 to 30 minutes in the laboratory (Schubel 1974), as well as those acclimated at 17.9 to 21.1°C and then exposed to 31.1°C for 30 minutes (Schubel and Koo 1976). Eggs acclimated at 32.9 to 36.1°C for 5 to 15 minutes experienced significant mortality, with total egg mortality occurring at 37.9°C. In their review of the literature, Klauda et al. (1991) concluded that suitable and optimal temperature ranges for eggs were 14 to 26°C and 20 to 24°C, respectively.

Blueback herring egg incubation is complete after 80 to 94 hours at 20 to 21°C (Kuntz and Radcliffe 1917; Jones et al. 1978) and 55 to 58 hours at 22.2 to 23.7°C (Cianci 1969; Klauda

et al. 1991). Following incubation, blueback herring eggs typically require 38 to 60 hours for hatching (Adams and Street 1969; Cianci 1969; Morgan and Prince 1976).

Larval blueback herring have been collected in the upper Chesapeake Bay where water temperatures ranged from 13 to 28°C; 96% were collected at 23 to 28°C (Dovel 1971). Blueback herring eggs and larvae collected from the Washademoak River, New Brunswick, were acclimated at 19°C, and then exposed to 29 and 34°C for 1 to 3 hours in the laboratory. While egg mortality and hatchability were deemed poor indicators of the effects of temperatures, larval deformity was considered a good indicator. Deformity rates over the three hour period were 0 to 25% at 29°C, and 100% at 34°C; such deformities were permanent and would have been lethal in the natural environment (Koo and Johnston 1978). In their review of the literature, Klauda et al. (1991) concluded that suitable temperature ranges for prolarvae and postlarvae were 14 to 26°C and 14 to 28°C, respectively.

Egg and larval dissolved oxygen associations

Larvae require a minimum of 5.0 mg/L of dissolved oxygen for survival (Jones et al. 1978).

Egg and larval pH and aluminum associations

Klauda (1989) conducted laboratory research on blueback herring fertilized eggs and yolk-sac larvae, and suggested that critical acidity conditions (defined as laboratory and field test exposures associated with greater than 50% direct mortality) for successful blueback herring reproduction in Maryland coastal plain streams occur during a single 8 to 96 hour pulse of acid (pH 5.5 to 6.2), with concomitant total monomeric aluminum concentrations of 15 to 137 µg/L. Eggs that were subjected to four treatments ranging from pH 5.7 to 7.5 and five aluminum treatments of 0 to 400 µg/L at a continuous exposure time between 96 and 120 h revealed the following results: 4-hour old embryos were sensitive to aluminum in the test treatments of pH 5.7 to 6.7; 12-hour old embryos were most sensitive to pH 5.7 with no aluminum present; and 24-hour old embryos suffered no mortality at all pH and aluminum levels (Klauda and Palmer 1987a).

Laboratory tests by Klauda et al. (1987) found a pH-induced mortality threshold for yolk-sac larvae of pH 5.7 to 6.5, and a 96-hour LC_{50} pH of 6.37 (pH that induced 50% mortality); no aluminum was administered. Additional tests by Klauda and Palmer (1987b) found that as the exposure time was doubled (12 to 24 hours), mortality rates increased among yolk-sac larvae (25 to 49%) at a pH value of 5.5. When coupled with a concomitant exposure of total aluminum maxima of 100 to 150 µg/L, mortality increased to 19, 66, 98, and 100% after 4, 8, 12, and 24 hours exposure, respectively. Tests also revealed highly variable mortality rates (3 to 75%) for yolk-sac larvae at a pH of 6.7. In general, the data indicated that blueback herring larvae were more sensitive to lower pH values (5.7 and 6.2) with no aluminum added, and were more tolerant of higher pH values (6.7 and 7.5) (Klauda and Palmer 1987b). Furthermore, yolk-sac larvae were more sensitive than 4-hour old embryos to pH and aluminum treatments (Klauda and Palmer 1987a). Klauda et al. (1991) suggested overall suitable ranges for eggs and prolarvae of 5.7 to 8.5 and 6.2 to 8.5, respectively; optimal ranges were suggested to be 6.0 to 8.0 and 6.5 to 8.0, respectively.

Median pH values (6.27) where blueback herring were spawning in the Rappahannock River, Virginia, reported by O'Connell and Angermeier (1997) were within the lethal range (5.7 to 6.5) and below a 96-h LC_{50} of 6.37 for larvae. Reduced pH levels may represent episodic events, such as acid precipitation, but additional study is required to determine what the effects of occasional pH depressions might be.

Egg and larval water velocity/flow

Initially, blueback herring eggs are demersal, but during the water-hardening stage, they are less adhesive and become pelagic (Johnston and Cheverie 1988). In general, blueback herring eggs are buoyant in flowing water, but settle along the bottom in still water (Ross and Biagi 1990).

Water flow rates may have a notable impact on larval populations of blueback herring. For example, year-class size of blueback herring decreased with increasing discharge during May-June from the headpond at the Mactaquac Dam (Saint John River, New Brunswick) (Jessop 1990). Researchers speculated that this was due to a low abundance of phytoplankton and zooplankton that larvae rely on at first feeding; these reductions can result when high discharges occur (Laberge 1975). This effect was not observed for alewife, which spawn 2 to 3 weeks earlier than blueback herring. Sismour (1994) also observed a rapid decline in abundance of early preflexion river herring larvae (includes both alewife and blueback herring) in the Pamunkey River, Virginia, following high river flow in 1989. Similar to Jessop (1990), Sismour (1994) speculated that high flow led to increased turbidity, which reduced prey visibility, leading to starvation of larvae. Furthermore, in tributaries of the Chowan system, North Carolina, water flow was determined to be related to recruitment of larval river herring (O'Rear 1983).

Dixon (1996) found that seasonally high river flow and low water temperature during one season in several Virginia rivers were associated with delayed larval emergence, reduced relative abundance, depressed growth rate, and increased mortality compared with the previous season. It was suggested that high river flow may be a forcing mechanism on another abiotic factor, perhaps turbidity, which directly affects larval growth and survival (Dixon 1996).

Egg and larval suspended solid associations

As with alewife, blueback herring eggs have proven extremely tolerant to suspended solids, with no significant reduction in hatching success at concentrations up to 1000 mg/L (Auld and Schubel 1972). Schubel and Wang (1973) demonstrated that high levels of suspended solids during and after spawning significantly increase the rate of egg infections from naturally occurring fungi in alewife, which cause delayed mortalities; it may be likely that the same effects would be observed in blueback herring eggs (Klauda et al. 1991). Two *in situ* studies (Klauda and Palmer 1987b; Greening et al. 1989) note that yolk-sac larvae appear to be more sensitive to suspended solids than eggs, but given that observations were made following storm events, which also resulted in changes to pH and current velocity, the effects of turbidity alone were inconclusive. Klauda et al. (1991) later noted a suitable concentration range of less than 500 mg/L for the prolarva life stage.

Egg and larval feeding behavior

First-feeding larvae in the Connecticut River primarily consumed rotifers; they shift to cladocerans as they grow larger (Crecco and Blake 1983). In general, it has been suggested that clupeids have evolved to synchronize the larval stage with the optimal phase of annual plankton production cycles (Blaxter et al. 1982).

Egg and larval competition and predation

All life stages of blueback herring, including the egg and larval stages, are important prey for freshwater fishes, birds, amphibians, reptiles, and mammals (Klauda et al. 1991). The ability of blueback herring to feed extensively on rotifers is offered as an explanation for their dominance over American shad in some rivers along the East Coast (Marcy 1976a; Loesch and Kriete 1980).

Eggs, larvae, and chlorine

Morgan and Prince (1977) reported an 80 h LC_{50} of 0.33 mg/L total residual chlorine (TRC) for blueback herring eggs incubated at 20.9°C in freshwater. The LC_{50} for 1-day old larvae exposed to TRC for 48 and 54 h ranged from 0.24 to 0.32 mg/L; LC_{50} for 2-day old larvae was between 0.25 and 0.32 mg/L (Morgan and Prince 1977). TRC concentrations that were greater than or equal to 0.30 mg/L increased the percentage of abnormally developed larvae (Morgan and Prince 1978).

Part C. Blueback Herring Juvenile Riverine/Estuarine Habitat

Geographical and temporal movement patterns

Recruitment to the juvenile stage for blueback herring begins later in the year than for other alosines because they spawn later and have a shorter growing season (Hildebrand and Schroeder 1928; Schmidt et al. 1988). The juvenile stage is reached when fish are about 20 mm TL (Klauda et al. 1991), with growth occurring very rapidly (Colette and Klein-MacPhee 2002).

Massman (1953), Warriner et al. (1970), and Burbidge (1974) have reported that juvenile blueback herring are most abundant upstream of spawning grounds in waters of Virginia. While Burbidge (1974) noted a greater prey density at these locations, he was unsure if fish were actually moving upstream in large numbers, if survival rates upstream were higher compared to survival rates downstream, or if fish were simply moving out of tributaries and oxbows into these areas. Michael Odom (U.S. Fish and Wildlife Service, personal communication) has noted that juvenile blueback herring select the pelagic main channel portion of tidal waters of the Potomac River, while American shad juveniles select shallower nearshore flats adjacent to and within submerged aquatic vegetation (SAV) beds. Odom speculates that these two species tend to partition the habitat in this river.

In North Carolina waters, Street et al. (1975) found that juveniles typically reside in the lower ends of the rivers in which they were spawned. In Chesapeake Bay tributaries, young-of-the-year blueback herring can be found throughout tidal freshwater nursery areas in spring and early summer; they subsequently head upstream later in the summer when saline waters encroach on their nursery grounds (Warriner et al. 1970). Schmidt et al. (1988) reasoned that juvenile blueback herring in the Hudson River remained in the vicinity of their natal areas throughout the summer because they were relatively absent downriver until late September.

Nursery areas of the Neuse River, North Carolina, have been characterized as relatively deep, slow-flowing, black waters that drain hardwood swamps (Hawkins 1979). In South Carolina, juvenile blueback herring and American shad were found to co-occur predominantly in deeper, channel habitats of estuarine systems, during fall and winter, while hickory shad selected shallow expanses of sounds and bays. Small crustaceans, favored by blueback herring and American shad, are generally abundant near the bottom in estuarine channels (McCord 2005).

Juvenile blueback herring spend three to –nine months in their natal rivers before returning to the ocean (Kosa and Mather 2001). Observations by Stokesbury and Dadswell (1989) found that blueback herring remained in the offshore region (25 to 30% seawater) of the Annapolis estuary (Nova Scotia) for almost a month before the correct migration cues triggered emigration. Once water temperatures begin to drop in the late summer through early winter (depending on geographic area), juveniles start heading downstream, initiating their first phase of seaward migration (Pardue 1983; Loesch 1987). Migration downstream is also thought by some researchers to be prompted by changes in water flow, water levels, precipitation, and light intensity (Kissil 1974; Pardue 1983). In contrast, other researchers have suggested that water flow plays little role in providing the migration cue under riverine conditions; these researchers think that migration timing is more dependent on water temperature and new to quarter moon phases, which provide dark nights (O'Leary and Kynard 1986; Stokesbury and Dadswell 1989).

In the Connecticut River, juvenile blueback herring were found to move out of river systems rapidly, within a 24-hour period, with peak migration occuring in the early evening at 1800 hours (O'Leary and Kynard 1986). Kosa and Mather (2001) studied juvenile river herring movement from 11 small coastal systems in Massachusetts, and found that most individuals emigrated between 1200 and 1600 hours. Farther north, emigration by juvenile blueback herring in the Annapolis River, Nova Scotia, peaked at night between 1800 and 2300 hours (Stokesbury and Dadswell 1989).

Juvenile blueback herring (age 1+) were found in the lower portion of the Connecticut River in early spring by Marcy (1969), which led him to speculate that many juveniles likely spend their first winter close to the mouth of the river. To the South, some young-of-the-year may overwinter in deeper, higher salinity areas of the Chesapeake Bay (Hildebrand and Schroeder 1928). In fact, Dovel (1971) reported juvenile populations in the upper Chesapeake Bay that did not emigrate until the early spring of their second year. Juveniles have also been reported overwintering in the Delaware Bay (Jones et al. 1978). Since juvenile river herring do not survive temperatures of 3°C or less (Otto et al. 1976), they would not be expected to overwinter in coastal systems where such temperatures persist (Kosa and Mather 2001).

Juveniles and the saltwater interface

Juvenile blueback herring are found most often in waters of 0 to 2 ppt prior to fall migration (Jones et al. 1988), but are tolerant of much higher salinities early in life. Pardue (1983) concluded that juveniles prefer low salinities in the spring and summer, with an optimal range between 0 and 5 ppt. Chittenden (1972) captured older juveniles in freshwater and subjected them to 28 ppt salinity at 22°C and all but one fish survived (mortality may have been due to handling stress). Furthermore, Klauda et al. (1991) suggested that 0 to 28 ppt was a suitable range for juveniles. Their ability to tolerate salinities as low as 0 ppt, and as high as 28 ppt, allows them to utilize both freshwater and marine nursery areas. However, both Loesch (1968) and Kissil (1968) found that juvenile blueback herring remained in freshwater up to one month longer than juvenile alewife.

In some cases, changes in one environmental factor may impact other environmental factors causing changes in behavior patterns. For example, in the Chowan River, North Carolina, juvenile blueback herring became scarce in sampling areas following drought conditions during the summer of 1981, which resulted in saline waters encroaching farther upriver into nursery areas. Researchers suggested that blueback herring had possibly moved further upstream to freshwater areas to avoid the saltwater intrusion (Winslow et al. 1983).

Juvenile substrate associations

Juvenile blueback herring have been found among submerged aquatic vegetation (SAV) beds of the lower Chesapeake Bay, and researchers have suggested that juveniles may benefit from reduced predation in such areas (Olney and Boehlert 1988). It is important to note, however, that no link has been made between the presence of SAV and abundance of alosines. Rather, SAV is known to improve the water quality, which may increase the abundance of alosines (B. Sadzinski, Maryland Department of Natural Resources, personal communication). Moreover, juvenile blueback herring are a pelagic schooling fish that likely do not rely on SAV

to the extent of other anadromous fishes, such as striped bass (D. A. Dixon, Electric Power Research Institute, personal communication).

Juvenile depth associations

Unlike alewife, juvenile blueback herring in the Potomac River remained at the surface or at mid-water depths during daylight hours from July through November, with almost no fish appearing at the bottom. However, at night over half of juvenile blueback herring captured were taken in bottom trawls (Warinner et al. 1970). Burbidge (1974) also reported that juvenile blueback herring were more abundant in surface waters of the James River, Virginia, during the day. Contrary to these results, Jessop (1990) found that abundance of juvenile bluebacks was greater in surface waters at night than during the day, but fish did not exhibit a strict negative phototropism. One explanation for these observed differences is the minimal sewage treatment that was required during the 1970's, which led to major phytoplankton and algal blooms in freshwater areas, reducing light penetration. Since that time, water clarity has greatly improved (Dennison et al. 1993).

In an additional study, Dixon (1996) found that juvenile blueback herring were more available to surface sampling gear approximately 30 minutes after sunset and before sunrise, where there was a corresponding light intensity of 10^{-2} to 10^{-3} uE/m^2/s. Because he did not detect a corresponding change in availability of primary zooplankton prey, he concluded that juveniles migrate to the surface water within a specific isolume with changes in incident light intensity, not as a response to prey movement. A light intensity of 10^{-2} to 10^{-3} uE/m^2/s may be a threshold that controls retinomotor responses to support selective feeding and schooling behavior in this species. Dixon (1996) concluded that juveniles find a depth and isolume that optimizes schooling (for predation protection) and selective feeding during the day, balancing predation risks versus preferred food availability. These results further support and refine the observations of Loesch et al. (1982), who first reported the diel changes in movement of juveniles.

Juvenile water temperature

Characterization	Temperature Range (^{o}C)	Acclimation Temperature (^{o}C)	Salinity (ppt)	Location	Citation
Present	11.5 – 32.0	N/A		Cape Fear River, NC	Davis and Cheek 1966
Present	6.7 – 32.5	N/A		Connecticut River	Marcy 1976b
Suitable	10 – 30	N/A		Chesapeake Bay	Klauda et al. 1991
Optimal	20 – 30			Many	Pardue 1983
Selection	20 – 22	15 – 20	4 – 6	Delaware River,	Meldrim and

Characterization	Temperature Range (°C)	Acclimation Temperature (°C)	Salinity (ppt)	Location	Citation
				NJ	Gift 1971
Preference	24 – 28	25 – 26	7 – 8	Laboratory	PSE&G 1978
Avoidance	36	25 – 26	7 – 8	Laboratory	PSE&G 1978
62% Mortality	32 – 33 for 4 to 6 minutes	19		Laboratory	Marcy and Jacobson 1976
100% Mortality	32 – 33 for 4 to 6 minutes	22.7		Laboratory	Marcy and Jacobson 1976
100% Mortality	30.5 for 6 minutes	15		Laboratory	PSE&G 1984
100% Mortality	32 for 6 minutes	15	29	Laboratory	Terpin et al. 1977
100% Mortality	10	25	6.5 – 7	Laboratory	PSE&G 1978
100% Mortality	0.2	5	8.5 – 10	Laboratory	PSE&G 1978

Table 5-1. Juvenile blueback herring water temperature associations

Juvenile blueback herring have a wide range of temperature tolerances (Table 5-1). Additionally, certain temperatures create cues for the juveniles to begin migration. For example, in the Connecticut River, emigration began when the water temperatures dropped to 21°C in September, peaked at 14 to 15°C, and ended when the temperature dropped to 10°C, in late October and early November (O'Leary and Kynard 1986). Milstein (1981) found juveniles overwintering in an estuary off the coast of New Jersey where bottom temperatures ranged from 2.0 to 10.0°C. These waters were warmer and had a higher salinity than the cooler, lower salinity estuarine nurseries where the juveniles reside in the fall.

Juvenile dissolved oxygen associations

Juvenile blueback herring have been collected in waters of the Cape Fear River, North Carolina, where dissolved oxygen concentrations ranged from 2.4 to 10.0 mg/L (Davis and Cheek 1966). In the laboratory, juveniles that were exposed to dissolved oxygen concentrations of 2.0 to 3.0 mg/L for 16 hours experienced a 33% mortality rate. Researchers determined that the juveniles were unable to detect and avoid waters with low dissolved oxygen (Dorfman and

Westman 1970). As a result, mass mortalities of juveniles resulted from low dissolved oxygen in the Connecticut River over several years during June and July, most notably in the early morning hours when dissolved oxygen was below 3.6 mg/L and temperature was 27.6°C (Moss et al. 1976). In addition, Klauda et al. (1991) concluded that juveniles require a minimum of 4.0 mg/L of dissolved oxygen.

Juvenile pH and aluminum associations

In the Cape Fear River, North Carolina, juvenile blueback herring were collected where pH was between 5.2 and 6.8 (Davis and Cheek 1966), but the length of time spent within these areas was unknown. In contrast, Kosa and Mather (2001) found that abundance of juvenile river herring peaked at a pH of 8.2 in coastal systems in Massachusetts. Researchers speculated that between 7.2 and 8.2, increases in river herring abundance may be related to changes in system productivity. Although researchers were unable to determine the exact mechanism for the impact of pH on river herring, they suggested that pH does appear to contribute to variations in juvenile abundance (Kosa and Mather 2001).

Juveniles and water velocity/flow

Discharge is an important factor influencing variability in relative abundance and emigration of juvenile river herring across smaller systems. Extremely high discharge may adversely affect juvenile emigration, and high or fluctuating discharge may decrease relative abundance of adult and juvenile blueback herring (Meador et al. 1984; West et al. 1988; Kosa and Mather 2001). In laboratory experiments, juvenile river herring avoided water velocities greater than 10 cm/s, especially in narrow channels (Gordon et al. 1992). However, in large rivers, where greater volumes of water can be transported per unit of time without substantial increases in velocity, the effects of discharge may differ (Kosa and Mather 2001). Jessop (1994) found that the juvenile abundance index (JAI) of blueback herring decreased, and daily instantaneous mortality increased, with mean July-August river discharge from the Mactaquac Dam headpond on the Saint John River, New Brunswick, Canada. Impacts may have been the result of advection from the headpond, or from mortality as a result of reduced phytoplankton and zooplankton prey (Jessop 1994).

Juvenile feeding behavior

Juvenile blueback herring in nursery areas feed mostly on copepods, cladocerans (Domermuth and Reed 1980), and larval dipterans (Davis and Cheek 1966; Burbidge 1974). In fact, as much as 40% of the juvenile's diet may consist of benthic organisms (Watt and Duerden 1974). Additionally, Burbidge (1974) found that juveniles often selectlarger items in the James River, Virginia, such as adult copepods, rather than smaller prey, such as *Bosminia* sp., except where there is a high relative abundance of smaller prey.   Several researchers (Vigerstad and Colb 1978; O'Neill 1980; Yako 1998) have hypothesized that a change in food availability may provide a cue for juvenile anadromous herring to begin emigrating seaward, but no causal link has been established.

Juvenile blueback herring feed mostly at the surface, below the surface of the water, and to a lesser degree, on benthic prey (Domermuth and Reed 1980; Colette and Klein-MacPhee 2002). Some researchers (Burbidge 1974; Jessop 1990) observed juveniles feeding somewhat at dawn, and increasing feeding throughout the day with a maximum at dusk, then declining overnight. It is suggested that during the day, juveniles will remain within, or near, their zone of preferred light intensity, and feed in a selective mode (Dixon 1996), such as a "particulate" feeding mode (Janssen 1982).

Dixon (1996) noted that the size and age of juvenile blueback herring in the nursery zone increased in the downstream direction. Burbidge (1974) made similar observations that larger juveniles were found in downstream reaches of the James River. Dixon (1996) noted that the relative age distribution and density of juveniles (center of abundance) persisted in the nursery zone throughout the sampling season, which precluded the hypothesis that cohorts move downriver as a function of age and size. Instead, Dixon (1996) referenced Sismour's (1994) theory that as river herring larvae hatch at different times and locations along the river, they will encounter varying concentrations and combinations of potential prey. It is these differences that will affect larval nutrition and survival. In early spring, larvae that are closer to the center of the chlorophyll maxima along the river (which likely support development and expansion of zooplankton assemblages) are more likely to find suitable prey items. Early in the season, sufficient prey in upriver areas may be lacking. As the season progresses and the zooplankton prey field expands to upriver reaches, larvae in these areas may find suitable prey quantities and grow to the juvenile stage (Sismour 1994; Dixon 1996). Pardue (1983) considered habitats that contained 100 or more zooplankton per liter as optimum, which he suggested was critical for survival and growth at this stage. Burbidge (1974) demonstrated a direct relationship between density of zooplankton and distribution and growth of blueback herring. This differential survival rate within the nursery zone over time may account for younger juveniles in upstream reaches (Dixon 1996).

Juvenile competition and predation

Young-of-the-year blueback herring are preyed upon by many freshwater and marine fishes, birds, amphibians, reptiles, and mammals. Eels, yellow perch, white perch, and bluefish are among the fish species that prey on blueback herring (Loesch (1987; Juanes et al. 1993). Researchers have suggested that excessive predation by striped bass may be contributing to the decline of blueback herring stocks in the Connecticut River (Savoy and Crecco 1995). Furthermore, suitably sized juvenile blueback herring were found to be energetically valuable and potentially a key prey item for largemouth bass in two Massachusetts rivers during the late summer. Although largemouth bass do not consistently consume blueback herring, they are energy-rich prey, which provide the highest growth potential (Yako et al. 2000).

It is often noted throughout the literature, that alewife and blueback herring co-exist in the same geographic regions, yet interspecific competition is often reduced through several mechanisms. For example, juveniles of both species in the Connecticut River consume or select different sizes of prey, leading researchers to conclude that intraspecific competition may be greater than interspecific competition (Crecco and Blake 1983). This behavior is also evident in the Minas Basin, Nova Scotia, where juvenile blueback herring favor smaller and more planktonic prey (filter feeding strategy) than do juvenile alewife (particulate-feeding strategy)

(Stone 1985; Stone and Daborn 1987). In addition, alewife spawn earlier than blueback herring, thereby giving juvenile alewife a relative size advantage over juvenile blueback herring, which allows them access to a larger variety of prey (Jessop 1990).

Furthermore, differences in juvenile diel feeding activity serve to reduce competition. One study noted that diurnal feeding by juvenile alewife is bimodal, with peak consumption about one to three hours before sunset and a minor peak occurring about two hours after sunrise (Weaver 1975). Another study found that juvenile blueback herring begin to feed actively at dawn, with feeding increasing throughout the day and maximizing at dusk, then diminishing from dusk until dawn (Burbidge 1974). Blueback herring are also found closer to the surface at night than alewifethat are present at mid-water depths; this behavior may further reduce interspecific competition for food between the two species (Loesch 1987).

Blueback herring and American shad juveniles also co-occur in shallow nearshore waters during the day, but competition for prey is often reduced by: 1) more opportunistic feeding by American shad; 2) differential selection for cladoceran prey; and 3) higher utilization of copepods by blueback herring (Domermuth and Reed 1980). Juvenile blueback herring are more planktivorous, feeding on copepods, larval dipterans, and cladocerans (Hirschfield et al. 1966, Burbidge 1974).

Blueback herring have shown signs of being impacted by invasive species as well. For example, there is strong evidence that juveniles in the Hudson River have experienced a reduced forage base as a result of zebra mussel colonization (Waldman and Limburg 2003).

Juveniles and alkalinity, carbon dioxide, and chlorine

Davis and Cheek (1966) captured juvenile blueback herring in the Cape Fear River, North Carolina, where the alkalinity ranged from 5 to 32 mg/L. This same study also found that juveniles selected areas where free carbon dioxide concentrations were between 4 and 22 ppm (Davis and Cheek 1966). Another study found that juvenile blueback herring held in freshwater avoided 0.1 mg/L total residual chlorine (TRC) at 17.5°C (PSE&G 1978).

Part D. Blueback Herring Late Stage Juvenile and Adult Marine Habitat

Geographical and temporal patterns at sea

Juvenile river herring have been found overwintering in an offshore estuary (Cameron and Pritchard 1963) 0.6 to 7.4 km from the shore of New Jersey, at depths of 2.4 to 19.2 m (Milstein 1981). This estuary is warmer and has a higher salinity than the cooler, lower salinity river-bay estuarine nurseries where river herring reside in the fall. The majority of river herring are present in this offshore estuary during the month of March, when bottom temperatures range from 4.4 to 6.5°C and salinity varies between 29.0 and 32.0 ppt (Cameron and Pritchard 1963). Further south, young blueback herring have been found overwintering off the North Carolina coast from January to March, concentrated at depths of 5.5 to 18.3 m (Holland and Yelverton 1973; Street et al. 1975).

Sexual maturity is reached between ages 3 and 6 for blueback herring. Life history information for young-of-the-year and adult blueback herring after they emigrate to the sea, and before they return to freshwater to spawn, is incomplete (Klauda et al. 1991). Researchers assume that most juveniles join the adult population at sea within the first year of their lives, and follow a north-south seasonal migration along the Atlantic coast, similar to that of American shad; changes in temperature likely drive oceanic migration (Neves 1981).

Neves (1981) reported that 16 years of catch data showed that blueback herring were distributed throughout the continental shelf from Cape Hatteras, North Carolina, to Nova Scotia during the spring. Most were found south of Cape Cod, but, unlike alewife, no blueback herring catches were recorded for Georges Bank. During the summer, blueback herring moved north and inshore, but catch records were too infrequent to determine summer occurrence for the species, although several catches were made near Nantucket Shoals and Georges Bank. This species was never collected south of 40° N in the summer. By early fall, the blueback herring were found along Nantucket Shoals, Georges Bank, and the inner Bay of Fundy, but were concentrated mostly along the northwest perimeter of the Gulf of Maine (Neves 1981). In the autumn, they began moving southward and offshore for overwintering along the mid-Atlantic coast until early spring (Neves 1981; Rulifson et al. 1987). Although winter sampling stations were inadequate to define wintering grounds, the few catches that were reported were primarily between latitude 40° N and 43° N. It is unknown to what extent blueback herring overwinter in deep water off the continental shelf of the United States (Neves 1981). This species has been found offshore as far as 200 km (Bigelow and Schroeder 1953; Netzel and Stanek 1966), but they are rarely collected more than 130 km from shore (Jones et al. 1978).

Canadian spring survey results also reveal river herring distributed along the Scotian Gulf, southern Gulf of Maine, and off southwestern Nova Scotia from the Northeast Channel to the central Bay of Fundy. They are also found to a lesser degree along the southern edge of Georges Bank and in the canyon between Banquereau and Sable Island Banks. A large component of the overwintering population on the Scotian Shelf moves inshore during spring to spawn in Canadian waters, but may also include the U.S. Gulf of Maine region (Stone and Jessop 1992).

Salinity associations at sea

Adult blueback herring have been collected in salinities ranging from 0 to 35 ppt (Klauda et al. 1991). Chittenden (1972) subjected adults to gradual and abrupt changes in salinity, including direct transfers from fresh to saltwater and vice versa, with no mortality. Non-spawning adults that do not ascend freshwater streams will likely be found mostly in seawater, and possibly brackish estuaries as they make their way up the coast to their summer feeding grounds (Chittenden 1972).

Depth associations at sea

The extent to which blueback herring overwinter in deep waters off the continental shelf is unknown. Individuals have been caught most frequently at 27 to 55 m throughout their offshore range. While at sea, blueback herring are more susceptible to bottom trawling gear during the day; this concept led early researchers to conclude that the species is aversive to light and follows the diel movement of plankton in the water column (Neves 1981). In the Gulf of Maine region, zooplankton concentrations are at depths less than 100 m (Bigelow 1926). Since blueback herring are rarely found in waters greater than 100 m in this area, it is speculated that zooplankton influence the depth distribution of blueback herring at sea (Neves 1981). A more recent study of juveniles within the riverine environment (see *Juvenile depth* under Part C of this chapter) found that they migrate to the surface within a specific isolume as light intensity changes (Dixon 1996).

Stone and Jessop (1992) found blueback herring offshore of Nova Scotia, the Bay of Fundy, and the Gulf of Maine, at mid-depths of 101 to 183 m in the spring, in shallower nearshore waters of 46 to 82 m in the summer, and in deeper offshore waters of 119 to 192 m in the fall. The researchers also found differences in depth distribution, with smaller fish (sexually immature) occurring in shallow regions (<93 m) during spring and fall, while larger fish occurred in deeper areas (≥93 m) in all seasons (Stone and Jessop 1992). In addition, the semi-pelagic nature of juveniles may provide them with protection from the effects of overfishing (Dadswell 1985).

Temperature associations at sea

Although data on offshore temperature associations is limited, researchers speculate that blueback herring are similar to other anadromous clupeids, in that they may undergo seasonal migrations within preferred isotherms (Fay et al. 1983). Neves (1981) found that blueback herring were caught in an offshore area where surface water temperatures were between 2 and 20°C and bottom water temperatures ranged from 2 to 16°C; almost all of the fish were caught in water temperatures less than 13°C. Catches were most frequent where bottom temperatures averaged between 4 and 7°C (Neves 1981).

Stone and Jessop (1992) found that the presence of a cold (<5°C) intermediate water mass over warmer, deeper waters on the Scotian Shelf (Hatchey 1942), where the largest catches of river herring occurred, may have restricted the extent of vertical migration during the spring. Since few captures were made where bottom temperatures were less than 5°C during the spring, researchers concluded that vertical migration may be confined by a water temperature inversion in this area (Stone and Jessop 1992).

Feeding behavior at sea

Blueback herring are size-selective zooplankton feeders (Bigelow and Schroeder 1953), whose diet at sea consists mainly of ctenophores, calanoid copepods, amphipods, mysids and other pelagic shrimps, and small fish (Brooks and Dodson 1965; Neves 1981; Stone 1985; Stone and Daborn 1987; Scott and Scott 1988; Bowman et al. 2000). In Minas Basin, Bay of Fundy, smaller blueback herring feed mostly on microzooplankton, while larger fish consume larger prey, including mysids and amphipods; feeding intensity also decreases with increasing age of fish (Stone 1985).

Neves' (1981) analysis of offshore survey results led to the conclusion that blueback herring follow the diel movement of zooplankton while at sea. As discussed above (see ***Juvenile depth*** under Part C of this chapter), Dixon's (1996) study in freshwater concluded that juvenile blueback herring followed diel movements in response to light intensity, not prey movement. Although direct evidence is lacking, catches of blueback herring in specific areas along Georges Bank, the perimeter of the Gulf of Maine, and south of Nantucket Shoals may be related to zooplankton abundance (Neves 1981).

Competition and predation at sea

Complete information on predation at sea is lacking for blueback herring (Scott and Scott 1988). Fish that are known to prey on blueback herring in the marine environment include spiny dogfish, American eel, cod, Atlantic salmon, silver hake, white hake, and Atlantic halibut, as well as larger schooling species, including bluefish, weakfish, and striped bass (Dadswell 1985; Ross 1991; Bowman et al. 2000). Seals, gulls, and terns may also feed on blueback herring in the ocean.

Section II. Significant Environmental, Temporal, and Spatial Factors Affecting Distribution of Blueback Herring

Table 5-2. Significant environmental, temporal, and spatial factors affecting distribution of blueback herring. Please note that, although there may be subtle variations between systems, the following data include a broad range of values that encompass the different systems that occur along the East Coast. Where a specific range is known to exist, it will be noted. For the subadult–estuarine/oceanic environment and non-spawning adult–oceanic environment life history phases, the information is provided as a general reference, not as habitat preferences or optima. NIF = No Information Found.

Life Stage	Time of Year and Location	Depth (m)	Temperature (°C)	Salinity (ppt)	Substrate	Current Velocity (m/sec)	Dissolved Oxygen (mg/L)
Spawning Adult	December (Florida) through late August (Nova Scotia) in Atlantic coast rivers from St. Johns River, FL to Nova Scotia. **Sympatric range:** Freshwater or brackish water above the head of the tide in fast-moving waters, also brackish ponds. **Allopatric range:** Slower-flowing tributaries and flooded low-lying areas adjacent to main streams	**Tolerable:** NIF **Optimal:** NIF **Reported:** Variable	**Tolerable:** 13-27 **Optimal:** 20-25 **Reported:** Variable	**Tolerable:** 0-6 **Optimal:** <1 **Reported:** Generally freshwater	**Tolerable:** NIF **Optimal:** NIF **Reported:** Sympatric: gravel, sand; Allopatric: Variable	**Tolerable:** NIF **Optimal:** NIF **Reported:** Fast flow	**Tolerable:** NIF **Optimal:** NIF **Reported:** Minimum 5
Egg	December to August (south to north progression) at spawning site or slightly downstream of spawning site	**Tolerable:** NIF **Optimal:** NIF **Reported:** Usually found at bottom	**Tolerable:** 7-14 **Optimal:** NIF **Reported:** Variable	**Tolerable:** 0-22 **Optimal:** 0-2 **Reported:** Usually freshwater	**Tolerable:** NIF **Optimal:** NIF **Reported:** Variable	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** Minimum 5
Larvae	38-60 hours after fertilization downstream of spawning site	**Tolerable:** NIF **Optimal:** NIF **Reported:** Diel movement	**Tolerable:** 13-28 **Optimal:** NIF **Reported:** Variable	**Tolerable:** 0-22 **Optimal:** NIF **Reported:** Usually freshwater	**Tolerable:** NIF **Optimal:** NIF **Reported:** Variable	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** ≥ 5 **Optimal:** NIF **Reported:** Minimum 5

Life Stage	Time of Year and Location	Depth (m)	Temperature (°C)	Salinity (ppt)	Substrate	Current Velocity (m/sec)	Dissolved Oxygen (mg/L)
Early Juvenile – Riverine Environment	3-9 months in natal rivers after reaching juvenile stage upstream or downstream of spawning sites, as far as offshore estuaries	**Tolerable:** NIF **Optimal:** NIF **Reported:** Surface or mid-water (daytime); bottom (nighttime)	**Tolerable:** 11-32 **Optimal:** 20-30 **Reported:** Variable; temp gives migration cues	**Tolerable:** 0-28 **Optimal:** 0-5 (summer) **Reported:** Variable	**Tolerable:** NIF **Optimal:** NIF **Reported:** SAV	**Tolerable:** NIF **Optimal:** NIF **Reported:** Variable	**Tolerable:** NIF **Optimal:** NIF **Reported:** Minimum 4
Subadult & Non-spawning Adult– Estuarine / Oceanic Environment	3-6 years after hatching in nearshore estuarine waters or offshore marine waters	**Tolerable:** NIF **Optimal:** NIF **Reported:** Diel migrations with zooplankton; most frequently caught at 27-55	**Tolerable:** NIF **Optimal:** NIF **Reported:** Probably travel in preferred isotherm like other alosines	**Tolerable:** NIF **Optimal:** NIF **Reported:** Brackish to saltwater	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF

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Chapter 6

HABITAT AREAS OF PARTICULAR CONCERN FOR ALOSINES

Section I. Identification and Distribution of Habitat and Habitat Areas of Particular Concern for Alosines

NOTE: **Due to the dearth of information on Habitat Areas of Particular Concern (HAPC) for alosine species, the information in this chapter is applicable to American shad, hickory shad, alewife, and blueback herring. Information about one alosine species may be applicable to other alosine species, and is offered for comparison purposes only. Certainly, more information should be obtained at individual HAPCs for each of the four alosine species.**

All habitats described in the preceding chapters (spawning adult, egg, larval, juvenile, sub-adult, and adult resident and migratory) are deemed essential to the sustainability of anadromous alosine stocks as they presently exist (ASMFC 1999). Klauda et al. (1991) concluded that the critical life history stages for American shad, hickory shad, alewife, and blueback herring, are the egg, prolarva (yolk-sac or pre-feeding larva), post-larva (feeding larva), and early juvenile (through the first month after transformation). Nursery habitat for anadromous alosines consists of areas in which the larvae, post-larvae, and juveniles grow and mature (ASMFC 1999). These areas include spawning grounds and areas through which the larvae and post-larvae drift after hatching, as well as the portions of rivers and estuaries in which they feed, grow, and mature. Juvenile alosines, which leave the coastal bays and estuaries prior to reaching adulthood, also use the nearshore Atlantic Ocean as a nursery area (ASMFC 1999).

Sub-adult and adult habitat for alosines consists of: the nearshore Atlantic Ocean from the Bay of Fundy in Canada to Florida; inlets, which provide access to coastal bays and estuaries; and riverine habitat upstream of the spawning grounds (ASMFC 1999). American shad and river herring have similar seasonal distributions, which may be indicative of similar inshore and offshore migratory patterns (Neves 1981). Although the distribution and movements of hickory shad are essentially unknown after they return to the ocean (Richkus and DiNardo 1984), due to harvest along the southern New England coast in the summer and fall (Bigelow and Schroeder 1953) it is assumed that they also follow a migratory pattern similar to American shad (Dadswell et al. 1987).

Critical habitat in North Carolina is defined as, "The fragile estuarine and marine areas that support juvenile and adult populations of economically important seafood species, as well as forage species important in the food chain." Among these critical habitats are anadromous fish spawning and nursery areas in all coastal fishing waters (NCAC 3I.0101 (20) (NCDEHNR 1997). Although most states have not formally designated essential or critical alosine habitat areas, most states have identified spawning habitat, and some have even identified nursery habitat.

Tables in Section II of each alosine species chapter contain significant environmental, temporal, and spatial factors that affect the distribution of American shad, hickory shad, alewife, and blueback herring. Additional tables found on the included DVD contain confirmed, reported, suspected, or historical state habitat for American shad, hickory shad, alewife, and blueback herring. Alosines spend the majority of their life cycle outside of state waters, and the Commission recognizes that all habitats used by these species are essential to their existence.

Section II. Present Condition of Riverine Habitats and Habitat Areas of Particular Concern for Alosines

Fisheries management measures cannot successfully sustain anadromous alosine stocks if the quantity and quality of habitat required by all species are not available. Harvest of fisheries resources is a major factor impacting population status and dynamics, and is subject to control and manipulation. However, without adequate habitat quantity and quality, the population cannot exist (ASMFC 1999).

Habitat quantity

Thousands of kilometers of historic anadromous alosine habitat have been lost due to development of dams and other obstructions to migration. In the 19th century, organic pollution from factories created zones of hypoxia or anoxia near large cities (Burdick 1954; Talbot 1954; Chittenden 1969). Gradual loss of spawning and nursery habitat quantity and quality, and overharvesting are thought to be the major causative factors for population declines of American shad, hickory shad, alewife, and blueback herring (ASMFC 1999). Although these threats are considered the major causative factors in the decline of shad and river herring, additional threats are discussed in the Threats chapter.

It is likely that American shad spawned in all rivers and tributaries throughout the species' range on the Atlantic coast prior to dam construction in this country (Colette and Klein-MacPhee 2002). While precise estimates are not possible, it is speculated that at least 130 rivers supported historical runs; now there are fewer than 70 systems that support spawning. Individual spawning runs may have numbered in the hundreds of thousands. It is estimated that runs have been reduced to less than 10% of historic sizes. One recent estimate of river kilometers lost to spawning is 4.36×10^3 compared to the original extent of the runs. This is an increase in available habitat over estimates from earlier years, with losses estimated at 5.28×10^3 in 1898 and 4.49×10^3 in 1960. The increase in available habitat has largely been due to restoration efforts and enforcement of pollutant abatement laws (Limburg et al. 2003).

Some states have general characterizations of the degree of habitat loss, but few studies have actually quantified impacts in terms of the area of habitat lost or degraded (ASMFC 1999). It has been noted that dams built during the 1800's and early to mid-1900's on several major tributaries to the Chesapeake Bay have substantially reduced the amount of spawning habitat available to American shad (Atran et al. 1983; CEC 1988), and likely contributed to long-term stock declines (Mansueti and Kolb 1953). North Carolina characterized river herring habitat loss as "considerable" from wetland drainage, stream channelization, stream blockage, and oxygen-consuming stream effluent (NCDENR 2000).

Some attempts have been made to quantify existing or historical areas of anadromous alosine habitat, including spawning reaches. For example, Maine estimated that the American shad habitat area in the Androscoggin River is 10,217,391 yd^2. In the Kennebec River, Maine, from Augusta to the lower dam in Madison, including the Sebasticook and Sandy rivers, and Seven Mile and Wesserunsett streams, there is an estimated 31,510,241 yd^2 of American shad habitat and 24,606 surface acres of river herring habitat. Lary (1999) identified an estimated 90,868 units (at 100 yd^2 each) of suitable habitat for American shad and 296,858 units (at 100

yd^2 each) for alewife between Jetty and the Hiram Dam along the Saco River, Maine. Above the Boshers Dam on the James River, Virginia, habitat availability was estimated in terms of the number of spawning fish that the main-stem area could support annually, which was estimated at 1,000,000 shad and 10,000,000 river herring (Weaver et al. 2003).

Although many stock sizes of alosine species are decreasing or remain at historically low levels, some stock sizes are increasing. It has not been determined if adequate spawning, nursery, and adult habitat presently exist to sustain stocks at recovered levels (ASMFC 1999).

Habitat quality

Concern that the decline in anadromous alosine populations is related to habitat degradation has been alluded to in past evaluations of these stocks (Mansueti and Kolb 1953; Walburg and Nichols 1967). This degradation of alosine habitat is largely the result of human activities. However, it has not been possible to rigorously quantify the magnitude of degradation or its contribution to impacting populations (ASMFC 1999).

Of the habitats used by American shad, spawning habitat has been most affected. Loss due to water quality degradation is evident in the northeast Atlantic coast estuaries. In most alosine spawning and nursery areas, water quality problems have been gradual and poorly defined; it has not been possible to link those declines to changes in alosine stock size. In cases where there have been drastic declines in alosine stocks, such as in the Chesapeake Bay in Maryland, water quality problems have been implicated, but not conclusively demonstrated to have been the single or major causative factor (ASMFC 1999).

Toxic materials, such as heavy metals and various organic chemicals (i.e., insecticides, solvents, herbicides), occur in anadromous alosine spawning and nursery areas and are believed to be potentially harmful to aquatic life, but have been poorly monitored. Similarly, pollution in nearly all of the estuarine waters along the East Coast has certainly increased over the past 30 years, due to industrial, residential, and agricultural development in the watersheds (ASMFC 1999). Specific challenges that currently exist are identified and discussed in greater detail in the Threats Chapter.

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<u>Chapter 7</u>

AMERICAN EEL

(*Anguilla rostrata*)

Section I. American Eel Description of Habitat

American Eel General Habitat Description and Introduction

American eel (*Anguilla rostrata*) are found in fresh, brackish, and coastal waters from the southern tip of Greenland to northeastern South America (Facey and Van den Avyle 1987). Additionally, there may be hybridization, or at least genetic introgression, of American eel into the population of European eel in Iceland. Therefore, the range might possibly be extended to Iceland in the north (Williams et al. 1984; Avise et al. 1990).

American eel are ubiquitous in many habitats (Jacobs et al. 2003), and can contribute up to more than 25% of the total fish biomass in some individual systems (Smith and Sauders 1955; Ogden 1970; J. McCleave, University of Maine, personal communication). In Connecticut rivers and streams, the American eel was found in one case to be four times more abundant than any other species (Jacobs et al. 2003). American eel habitats include the open ocean, estuaries, large coastal tributaries, rivers, small freshwater streams, lakes, and ponds. They utilize habitats from the East Coast of North America and the northern portion of South America, into the inland areas of the Mississippi River and the Great Lake drainages (primarily Lake Ontario), and north into Canadian tributaries. American eel are sometimes found in land locked lakes, particularly in the northeastern United States (Facey and Van den Avyle 1987). The latitudinal range for the American eel has been documented as 5°N to 60°N (Bertin 1956), and their range covers approximately 30,000 km of coastline (Federal Register 2007). American eel are thought to occupy the broadest array of habitats of any fish in the world (Helfman et al. 1987).

American eel are a catadromous species that reproduces in salt water, and after an oceanic larval stage, migrates to brackish or fresh water for growth to maturity. Upon reaching maturity, the American eel migrate back to the ocean to spawn. Spawning occurs in the winter and spring in the Sargasso Sea, and the newly hatched larvae (pre-leptocephalus and leptocephalus stages) passively drift and swim toward the continental shelf where they metamorphose into glass eels (Kleckner and McCleave 1982; Kleckner and McCleave 1985; McCleave et al. 1987).

The transformation from a leptocephalus larvae into a glass eel includes a decrease in body length and weight due to a loss in water concentration, an increase in body thickness, loss of larval teeth, darkening of the eye, changes in the morphology of the head and jaw, and further development of the digestive system (Fahay 1978). Glass eels are miniature transparent American eel that are morphologically similar to elvers (the next life stage), but they are unpigmented. As American eel develop pigment, some begin to migrate into freshwater. These young pigmented American eel are termed elvers. Some elvers remain in coastal rivers and estuaries, while others may continue movements upstream in the winter and the spring (Facey and Van den Avyle 1987). In fact, upstream migration may continue into the yellow- phase for at least three to five years (Haro and Krueger 1991).

The next life stage for American eel is the yellow-phase, which is the primary growth stage where individuals spend most of their lives. The yellow-phase is characterized by a lack of sexual maturity and may last many years. Sexual differentiation begins when eels reach approximately 300 mm TL, primarily during the yellow-phase. Following sexual differentiation,

American eel eventually begin to migrate downstream (Krueger & Olivera 1999). Yellow-phase eels gradually metamorphose into silver-phase adults through a process that involves a number of physiological changes. Physiological changes reviewed by Facey and Van den Avyle (1987) include a change in color to a metallic bronze black sheen, pectoral fin color change from yellow-green to black, fattening of the body, thickening of the skin, increased length of capillaries in the rete of the swim bladder, and degeneration of the digestive tract. Additionally, the eyes become enlarged and the visual pigments in the eye are altered (Vladykov 1973; Beatty 1975). These changes are thought to better suit the American eel for migration at deeper depths (Beatty 1975; Kleckner and Kruger 1981; Facey and Van den Avyle 1987). During maturation, American eel migrate downriver to marine waters and out to the Sargasso Sea, where they are thought to spawn once and die (Facey and Van den Avyle 1987).

All American eel comprise one panmictic population, meaning that they are a single breeding population that exhibits random mating. Thus, for example, an American eel from the northern portion of the range could mate with an American eel from the southern portion of the range, and their offspring could inhabit any portion of the range. As a result, recruits to a particular system are likely not the offspring of the adults that migrated out of that system (ASMFC 2000; Avise 2003).

Life history information for American eel remains incomplete, and for some life stages, habitat-specific information is lacking. There is a high degree of uncertainty regarding the range of variation in life history traits that occurs throughout the entire population. Knowledge is lacking on silver eel migration from freshwater to the sea, as well as the egg, leptocephali, and glass eel life stages while in marine waters. Furthermore, while a potential spawning area of the American eel has been hypothesized in the Sargasso Sea, the specific spawning location remains unknown and no spawning activity has been witnessed (ASMFC 2000).

Many studies have indicated that American eel populations are declining (Castonguay et al. 1994 a, b; Haro et al. 2000b). Recent research by Richkus and Whalen (1999, 2000) has shown a decrease in yellow-phase and silver-phase American eel abundance in Ontario, Quebec, New York, and Virginia. For example, during the 31-day peak migration period in 2004, the mean number of American eel passing through the Moses-Saunders Hydroelectric Dam at Cornwall, Ontario, decreased from previous estimates of over 27,000 individuals per day to 274 individuals per day (Casselman *In press*).

Concerns about the decline in American eel abundance prompted a petition in 2004 to list the American eel as endangered under the Endangered Species Act (16 U.S.C. §§ 1531 – 1544). NOAA Fisheries and the United States Fish and Wildlife Service subsequently completed a 12-month status review to determine whether an endangered finding was justified. The findings of the status review indicated that listing the American eel as a threatened or endangered species was not currently warranted due to the fact that American eel are widely distributed and that their overall population abundance remains in the millions. The review also noted that ample historic habitat is available to American eel, and they have the flexibility to complete their lifecycle using marine and estuarine waters, in addition to freshwater. Furthermore, recruitment trends appear to be stable and the factors affecting American eel do not appear to threaten the species at a population level (Federal Register 2007).

Due to their diverse habitat requirements, American eel are subjected to a number of anthropogenic impacts. Fishing pressures and habitat loss are implicated as contributing factors

in the American eel decline. Some habitat threats include blockage of stream access, pollution, nearshore habitat destruction, and oceanic changes (Castonguay et al. 1994a, b; ASMFC 2000).

Part A. American Eel Spawning Habitat

Geographical and temporal patterns of migration

American eel are believed to spawn in the Sargasso Sea, which constitutes a large portion of the western North Atlantic Ocean east of the Bahamas and south of Bermuda. Spawning occurs during the winter and the spring, from February to April, and possibly later into the year (McCleave et al. 1987). No other information exists on the spawning requirements, behavior, or exact location of spawning in the Sargasso Sea. Some researchers have speculated that the spawning area is located south of Bermuda and north of the Bahamas in a zone centered at about 25°N and 69°W (Tesch 1977). McCleave et al. (1987) reported spawning in the area from 52ºW to 79°W longitude and 19.5ºN to 29ºN latitude.

Kleckner et al. (1983) and Kleckner and McCleave (1988) hypothesize that within this area, spawning occurs in the subtropical front systems of the oligotrophic subtropical gyres. This frontal zone is located within the North Atlantic Subtropical Convergence and occurs yearly during the time span when spawning is thought to take place. This area separates the warm saline water mass of the southern Sargasso Sea from the lower salinity cool water mass of the northern Sargasso Sea. The area occurs in the upper 500 m of the water column, and it is thought that spawning occurs on the warm side of this front (McCleave and Kleckner 1985; McCleave et al. 1987). However, no direct observations of American eel spawning have been reported anywhere in the world, and no adult American eel have been captured in the Sargasso Sea. Thus, the exact location of spawning area has only been inferred from the collection of leptocephali, or larvae, less than 7 mm in size (Kleckner et al. 1983; Kleckner and McCleave 1985).

The northern limit of the spawning area for American eel appears to be the thermal fronts that separate the northern and southern water masses of the Sargasso Sea (Kleckner et al. 1983). Kleckner et al. (1983) found that the smallest leptocephali collected during their study (3.9 to 5.5 mm) were located on the warm side of these fronts and were rare on the cold side of the fronts. Kleckner and McCleave (1985) suggest that the northern limit for spawning occurs between 24°N and 29°N, and the Bahamas/Antilles Arc forms the southern and western borders. Thus far, the eastern limit of American eel spawning has not been hypothesized (Kleckner and McCleave 1985). Kleckner and McCleave (1985) suggest that this eastern limit may be controlled by a directional orientation mechanism used by American eel adults to locate the spawning area.

It remains unknown how American eel locate the spawning area in the Sargasso Sea and what cues cause them to cease migration. McCleave and Kleckner (1985) offer three hypotheses relating to how American eel migrate in the open ocean. Their first hypothesis is that swimming in one general compass direction (south), in addition to oceanic circulation, allows the American eel to reach the spawning area from anywhere within the species geographical range. Their second hypothesis is that only a moderate directional orientation will result in successful migrations. Their final hypothesis is that migration occurs within the upper three hundred meters

of water, which McCleave and Kleckner (1985) speculate is significant with regard to the mechanism of migration. Alternatively, Stasko and Rommel (1977) suggest that American eel orient themselves using geoelectrical fields generated by ocean currents.

Kleckner et al. (1983) suggest that American eel cease migrating when they cross the frontal zone, an area located between 24ºN and 29ºN, which meanders from east to west for hundreds of kilometers. The researchers believe that some feature of the surface water south of the front cues the American eel to cease migration; it may be indicated by a thermal or chemical characteristic of the surface water. In addition, temperature and odor might also serve as cues to halt migration (McCleave and Kleckner 1985). For example, the temperature between the zones may vary as much as 2ºC, and the northern and southern zones exhibit differing species compositions of phytoplankton, zooplankton, and mesopelagic fishes, which could account for a change in odor (McCleave and Kleckner 1985). Furthermore, the upper layers in the pycnocline in the Sargasso Sea may contain dissolved amino acids that are known to be potent to American eel (Liebezeit et al. 1980; Silver 1979). McCleave and Kleckner (1985) suggest it is possible that the leptocephalus larvae imprint to this area in the same way that salmon imprint to a home stream.

American eel are thought to be semelparous, meaning that they die after one spawning event. Evidence for this includes no observations of adult American eel migrating upriver, and no spent adults reported in the literature (Facey and Van den Avyle 1987).

Spawning and the saltwater interface

Salinity might be a key habitat parameter for spawning adult American eel, as spawning is thought to occur on the side of the front in the Sargasso Sea that has warmer temperatures and more saline waters (Kleckner et al. 1983; Kleckner and McCleave 1985). The spawning grounds of the American eel may occur in a high salinity region of the Sargasso Sea where the salinity reaches a maximum of 36.6 ppt (Kleckner and McCleave 1985).

Spawning substrate associations

Bottom composition is not known to be important to spawning adult American eel, as reproduction is thought to occur in the upper 150 to 200 m of the water column (Kleckner et al. 1983; McCleave and Kleckner 1985).

Spawning depth associations

Kleckner et al. (1983) and McCleave and Kleckner (1985) suggest that morphological and physiological evidence indicate that American eel spawning occurs in the upper few hundred meters of the water column. Furthermore, larval American eel (less than 5 mm long) have been located in water 50 to 350 m deep, suggesting that spawning occurs in the upper water column (Kleckner and McCleave 1982).

Spawning water temperature

Temperature may be significant to spawning adult American eel, as they are thought to spawn on the warmer side of the front in the Sargasso Sea (Kleckner et al. 1983; Kleckner and McCleave 1985). Spawning is thought to occur in an area where water temperatures are characterized by 18 to 19°C isotherms between 200 and 300 m (Kleckner et al. 1983). Kleckner and McCleave (1985) describe the hypothesized spawning area as having temperatures greater than 18.2°C. Haro (1991) found that mean preferred water temperature for sexually mature male American eels test in the laboratory ranged between 17.2 and 18.1°C.

Spawning feeding behavior

Once the spawning migration begins, American eel cease feeding and their digestive system atrophies. Gray and Andrews (1971) found no prey and shrunken stomachs in silver eels, suggesting that the subjects ceased feeding before migration.

Spawning competition and predation

Both American eel and European eel (*Anguilla anguilla*) are thought to use the Sargasso Sea for spawning grounds (McCleave et al. 1987). However, McCleave et al. (1987) speculate that American eel spawn from February to April from approximately 19°N to 29°N latitude and 52°W to 79°W longitude, while European eel spawn from March to June from approximately 23°N to 30°N latitude and 48°W and 74°W longitude. Thus, their overlap area may not be significant enough to induce competition.

Part B. American Eel Egg and Larval Habitat

Geographical and temporal movement patterns

Little information exists on the environmental requirements or the incubation period of American eel eggs. It is assumed that the eggs hatch in the same area as they are laid in the Sargasso Sea (see discussion in above section). Hatching is thought to occur from February through April (McCleave et al. 1987), with a possible peak occurring in February (Tesch 1977).

After hatching, American eel undergo a brief pre-larval stage, and then enter the larval leptocephalus life stage. Leptocephali are flattened from side to side and resemble a willow leaf (ASMFC 2000). They grow to between 55 and 65 mm before metamorphosis to the glass eel stage (Kleckner and McCleave 1985). While growing, the leptocephali drift and swim in the upper water column of the open ocean. Their distribution is a result of the oceanic circulation patterns and the swimming behavior of the larvae (ASMFC 2000).

Kleckner and McCleave (1985) reported on the spatial and temporal distribution of leptocephali by collecting specimens and analyzing data collected by Schmidt in the 1920's. They found that leptocephali 7 to 10 mm in length were caught from mid-February to the end of April. In addition, specimens longer than 45 mm were acquired during all months. Kleckner and McCleave (1985) identified two year classes that occurred from February to mid-June: a 0-year class that constituted most samples, and a 1-year class, which represented only a few larvae.

Kleckner and McCleave (1985) collected the majority of leptocephalus larvae between 11°00'N and 42°35'N latitude and 43°50'W and 87°00'W longitude. One 70 mm leptacephalus (a member of the 1-year class) was collected at 49°43'N, 20°45'W. The researchers stated that all leptocephali 10 mm TL or less, and all 0-year leptocephali, were found within a 550 km arc east of the Bahamas and north of the Hispanola Islands. These specimens were found from February to March. Sampling father north and east yielded no leptacephali (Kleckner and McCleave 1985).

From April to May, only one young-of-the-year leptocephalus was collected in the eastern Sargasso Sea from 23°N to 28°N and 51°W to 63°W (Kleckner and McCleave 1985). Kleckner and McCleave (1985) also found young-of-the-year American eel in the Caribbean Current along the western shore of the Yucatan Channel in the Straits of Florida, and in the Gulf Stream to the east of Cape Hatteras, in April and May. Despite the use of nets capable of capturing small leptocephali, no larvae were collected from 38°N to 44°N and 41°W to 55°W in the North Atlantic current (Kleckner and McCleave 1985).

Throughout June and July, young-of-the-year American eel were taken in the Caribbean, Gulf Loop, Florida, and Gulf Stream currents. The samples were taken east to 54°15'W in the southern Sargasso Sea and northeast of Bermuda east to 56°46'W. No larvae were found in the eastern North Atlantic Current at that time. The authors were also unable to define an eastern limit of young-of-the-year larvae during these months in the Gulf Stream due to a lack of collections south of Newfoundland (Kleckner and McCleave 1985).

By August, American eel larvae 40 to 67 mm occupied the entire Gulf Stream area up to the Gulf of Maine. From August through October, only a few large leptocephali, or newly metamorphosed glass eels, remained far out in the Western Atlantic coast (Kleckner and

McCleave 1985). Kleckner and McCleave (1985) reported that during August and September, they collected leptocephali in the southern Caribbean Sea, Gulf Loop Current, Florida Current, Gulf Stream, and North Atlantic Current. Throughout the fall, American eel approached the North American continent and Greenland in the glass eel phase (Kleckner and McCleave 1980; Kract and Tesch 1981). Kleckner and McCleave (1985) found American eel leptocephali in collections in the Caribbean Sea from south of Puerto Rico to the Yucatan Channel in October and November. Likewise, leptocephali were found south of the northeastern United States in October and November, inshore and offshore of the Gulf Stream, and in the Canadian Maritime provinces. However, leptocephali in the south and east in the Sargasso Sea were scarce (Kleckner and McCleave 1985).

Kleckner and McCleave (1985) found that age 1 American eel were scattered widely in collections taken in the Caribbean Sea and western North Atlantic Ocean from February through May. Many specimens were taken near the Bahaman Islands and the Florida Current off the Southeastern United States. They also found metamorphosing American eel leptocephali located north of the Gulf Stream between 65°42'W and 73°30'W, 55 km southwest of Bermuda, and approximately 45 km southeast of Cape Hatteras. One specimen was taken 110 km north of Campeche Bank in the Gulf of Mexico (Kleckner and McCleave 1985).

Larvae are transported northwest from the spawning grounds to the eastern seaboard by the Antilles Current, Florida Current, and the Gulf Stream (Facey and Van den Avyle 1987). The proposed route of American eel larval transport is a westward drift from the spawning grounds in the Sargasso Sea via the Antilles Current, and then moving north with the Florida Current to join the Gulf Stream north of Bermuda (Kleckner and McCleave 1985; McCleave 1993; McCleave et al. 1998).

A small portion of leptocephali reach the Caribbean Sea, Gulf of Mexico, and the Straits of Florida. The proposed route of these larvae occurs to the west and southwest of the spawning grounds via the Windward and Mona Passages, which transport the larvae to the Caribbean Sea. From here, eddies could carry them along the Caribbean coast, or the Caribbean current could convey them through the Yucatan Channel into the Gulf of Mexico and the Gulf loop current (Kleckner and McCleave 1985; McCleave and Kleckner 1987). Leptocephali entering the Straits of Florida are likely carried by the Gulf Loop Current, which flows out of the Gulf of Mexico as the Florida Current. Additionally, they may be conveyed into the Straits of Florida from the Bahamas/Antilles archipelago by currents through the Old Bahama Channel, then the Nicholas and Santaren Channels north of Cuba, or through the Northwest Providence Channel south of Grand Bahaman Island (Kleckner and McCleave 1985).

It is possible that some eel larvae become trapped in the Sargasso Sea for over a year by recirculating currents (Knights 2003). This occurs when the larvae become trapped in the sub-gyre where the Florida and Antilles Currents interact, thus causing the larvae to drift north, or recirculate back into the oligotrophic Sargasso Sea from the Gulf Stream (Boëtius and Harding 1985).

 As the larvae approach the edge of the continental shelf, they metamorphose into miniature transparent eel, called glass eels (Kleckner and McCleave 1985). This occurs by early October when the American eel are between 55 mm and 65 mm (Kleckner and McCleave 1985).

Eggs, larvae, and the saltwater interface

The salinity requirements of eggs and larvae have not been documented in literature. Facey and Van den Avyle (1987) state that post-larval American eel are tolerant of a broad range of salinities because they occur both in freshwater and marine habitats. Additionally, leptocephali are in near-ionic equilibrium with seawater (Hulet et al. 1972).

Egg and larval substrate associations

Bottom substrate is not important to this lifestage, as American eel larvae are planktonic and float and drift in the water column. Thus, no bottom substrate is used during this life stage (Kleckner and McCleave 1985).

Egg and larval depth associations

The importance of depth to the American eel egg stage is not stated in the literature. No information exists on the depth that eggs are found, as they have never been collected in the Sargasso Sea (ASMFC 2000).

Once American eel enter the leptocephalus stage, they are found in the upper 250 m of the water column (Castonguay and McCleave 1987). Larvae less than 5 mm long have been captured at depths between 50 m and 350 m. Furthermore, larvae between 5 and 10 mm appear to vertically migrate, as they are found between 100 m and 150 m during the day and between 50 m and 100 m at night. (Castonguay and McCleave 1987; McCleave et al. 1987).

Egg and larval water temperature

No studies have concluded the egg and larval temperature requirements of American eel in the wild. However, Japanese eel (*Anguilla japonica*) eggs hatch in 38 to 45 hours at 23°C (Yamamoto and Yamauchi 1974). Spawning and hatching is likely to occur on the warm side of the front in the Sargasso Sea where temperatures are greater than 18.2°C (Kleckner and McCleave 1985).

Egg and larval competition and predation

Both American and European eel use the Sargasso Sea as a spawning ground. As a result, the youngest stages of both eel species may share a small portion of the same habitat. However, Kleckner and McCleave (1985) state that while there is an overlap in range, competition does not occur. American eel larvae are predominately found west of 62°W and south of 25°N, while European eel are located in a different area (Kleckner and McCleave 1985; McCleave and Kleckner 1987).

One study by Appelbaum (1982) suggests that predation on American eel larvae in the Sargasso Sea may be minimal. Researchers found that of 1,000 pelagic fish representing 25 species, only the myctophid, *Ceratoscopelus warmingii*, had American eel leptocephali in its stomach. This suggests that American eel may spawn in a nutritionally poor area, thus

increasing the chance of survival due to a lack of predation. However, more research is needed to fully explore the issue (Appelbaum 1982).

Part C. American Eel Elver (including Glass Eel) Habitat

Geographical and temporal movement patterns

American eel metamorphose from leptocephalus larvae to glass eels over the Continental Shelf. Shortly after metamorphosis, the unpigmented glass eels enter estuaries, eventually migrate to freshwater, and ascend rivers during the late winter and early spring. It is thought that glass eels and elvers use olfaction to locate freshwater (Sheldon 1974; Sorensen 1986; Sorensen and Bianchini 1986); however, the specifics of this theory are mostly unknown. For example, Sorensen (1986) reported that American eel were attracted to the smell of brook water, as well as the smell of leaf detritus. Furthermore, Creutzberg (1959, 1961) demonstrated that European eel were able to detect the odor of freshwater, and alter their behavior accordingly.

Vladykov (1966) stated that the American eel migration upriver occurred earlier in the southern portion of the range than in the north. However, other studies showed variations and overlaps in migration timing (Facey and Van den Avyle 1987). Migrating American eel in the Southeastern states and the Mid-Atlantic have been collected from January through May (Jeffries 1960; Smith 1968, Fahay 1978; Hornberger 1978; Sykes 1981; Helfman et al. 1984). In the Northern states, migrating glass eels reach estuaries as early as late winter (Jeffries 1960), although the main migration occurs in the spring. In the East River, Chester, Nova Scotia, Jessop (2000) reported eel recruitment in the river mouth from May through June, and upstream migrations from July through September. Dutil et al. (1989) reported that the glass eel and elver migration to the St. Lawrence estuary occurred in the second half of June and was finished by the end of July.

Slightly south, American eel in Maine were documented arriving upstream from the end of March to the beginning of May (Facey and Van den Avyle 1987). Ricker and Squires (1974) and Sheldon (1974) reported that American eel ran in Maine from late April to June. In Rhode Island, migrations peaked during April and May (Facey and Van den Avyle 1987). Further south, in North Carolina, Rulifson et al. (2004) found that recruitment of elvers occurred from January through April, with the highest density of American eel present from March to April.

Glass eels enter estuaries by drifting on flood tides and holding position near the bottom of ebb tides (McCleave and Wippelhauser 1987), and by actively swimming along shore in estuaries above tidal influence (Barbin and Krueger 1994). Movements of glass eels are primarily nocturnal (Dutil et al. 1989). Eventually, glass eels in estuaries change into pigmented elvers (Haro 1991).

Throughout the elver life stage, American eel are mostly active at night. During the day elvers either burrow or remain in deep waters (Deelder 1958). Elvers move back up into the water column on flood tides and return to the bottom during ebb tides (Pacheco and Grant 1973; McCleave and Kleckner 1985; McCleave and Wippelhauser 1987).

Documentation shows that American eel stall their inward migration before they enter freshwater (McCleave and Kleckner 1985). The cues that trigger this behavior are unknown. Some researchers hypothesize that American eel may be able to detect the odor of freshwater (Creutzberg 1959, 1961; Sorensen 1986). Stalling at the freshwater interface may allow individuals to adjust physiologically and behaviorally before entering the new environment (Sorensen and Bianchini 1986). This upstream migration is possibly triggered by water

chemistry changes associated with the intrusion of estuarine water during the high spring tides (Sorensen and Bianchini 1986).

Elvers eventually begin their upstream migration and become more active during the day (Sorensen and Bianchini 1986). Tesch (1977) reported that European elvers oriented themselves with river currents for upstream movement. If the current was too weak or strong, the European eel moved into backwater areas and delayed migration. Since American eel and European eel have similar behavior patterns, it is possible that fast or slow currents also affect American eel (Tesch 1977).

Factors that are thought to influence the daily abundance of migrating elvers include nightly tidal height, river water temperature and discharge, and the difference between bay and river temperatures (McCleave and Kleckner 1985; Sorensen and Bianchini 1986; Ciccotti et al. 1995; McCleave and Wipplehauser 1987; Wipplehauser and McCleave 1987; Martin 1995; Jessop 2003). Migration occurs in waves and is initially triggered by an increase in temperature to between 12 and 14°C. After initiating migration, temperature does not appear to have a functional influence on migrating elvers (Jellyman and Ryan 1983; Martin 1995; Jessop 2003). River discharge appears to control the daily abundance of upstream migrants, with decreases in abundance coinciding with increases in river discharge. Jessop (2003) stated that increased tidal height delivered an increased abundance of elvers to the river mouth. Temperature then triggered upstream migration, while discharge controlled the rate of movement upstream (Jessop 2003).

While most American eel elvers migrate into freshwater, some may cease migration in coastal waters and estuaries and remain there from the time they arrive until they reach the mature silver eel stage and begin the spawning migration (Morrison et al. 2003, Lamson et al. 2006). In addition to the upriver migration, fall and spring migrations have been documented (Smith and Saunders 1955; Medcof 1969).

Elvers and the saltwater interface

Little is known about the salinity requirements of juvenile American eel. Sheldon and McCleave (1985) documented glass eels in Penobscot, Maine, in salinities ranging from 0 to 25.2 ppt.

Elver substrate associations

Substrate may be an important habitat parameter for juvenile American eel, as elvers have been seen burrowing during the day and in between movements upstream. American eel appear to use many different types of substrates. Facey and Van den Avyle (1987) stated that migrating elvers make use of soft undisturbed bottom sediments as shelter. Furthermore, a study by Edel (1979) demonstrated that American eel are less active when there is shelter present. Fahay (1978) stated that post-larval American eel are benthic and utilize burrows, tubes, snags, plant masses, other types of shelter, and the substrate itself. Additionally, American eel have been documented burrowing in both mud and sand (P. Geer, Georgia Department of Natural Resources, personal communication). Elvers may also use the hydraulic boundary layer of rough substrates to facilitate migration upstream, or migrate through interstitial spaces within a substrate to avoid high water velocities during upstream migration (Barbin and Krueger 1994).

Elver depth associations

Creutzberg (1961) reported that at night, unpigmented European eel in coastal waters were found in a variety of depths throughout the water column during incoming tides. During the day, elvers move to the bottom and bury themselves in the substrate (Deedler 1958).

Elver water temperature

Temperature is important to elvers because it is thought to trigger upstream migration. Migrations of American eel begin when the temperature rises above 10ºC, with the majority of movement occurring at temperatures greater than 20ºC (Moriarty 1986; Haro and Krueger 1991; Richkus and Whalen 1999; Jessop 2003). Jessop (2003) found that elvers in the East River, Chester, Nova Scotia, actively moved upstream when river temperatures reached 10 to 12°C, and the first wave of migrants peaked at 11 to 16°C. Water temperatures of less than 10°C had a gating effect on the elvers (Jessop 2003).

Other researchers have found similar results. Helfman et al. (1984) noted migrations in Georgia at 11°C, Soreson and Bianchini (1986) found a range of 10 to 15°C in Rhode Island, with a peak at 14°C, and Smith (1955) and Groom (1975) found a temperature range of 10 to 12°C for migrating American eel in New Brunswick. While temperature is thought to play an active role in stimulating migration, other factors also play a role in the abundance of American eel migrating upstream (Jessop 2003).

Beyond stimulating migration, temperature does not appear to play a key role in the elver life cycle. Juvenile American eel utilize a broad range of habitats and are likely to have flexible temperature tolerance ranges. Glass eels were documented in Penobscot, Maine, in temperatures ranging from 3.9 to 13.8ºC (Sheldon and McCleave 1985). Elvers have been documented in a wide variety of temperatures, including cold freshwater streams and lakes, and warm brackish coastal bays and lakes. In fact, elvers have been found at temperatures as low as -0.8°C in the Narragansett Bay, Rhode Island (Jeffries 1960).

Elver water velocity/flow

Sheldon and McCleave (1985) noted that in Penobscot, Maine, glass eels accumulated on the surface when surface currents on the ebb tide decreased below 15 cm·s^{-1}. In another study, river discharge and its effects on water velocity were found to be the primary factor influencing the rate of elver upstream migrations (Jessop 2000). In velocities exceeding 35 to 40 cm·s^{-1}, elvers had difficulty swimming and maintaining their position (McCleave 1980; Barbin and Krueger 1994). Jessop (2000) found that most elvers would not swim at water velocities exceeding 25 cm·s^{-1}, and instead would remain resting in the substrate. Some researchers have found that delays or prevention of upstream elver migration can be caused by high flows (Lowe 1951; Jessop and Harvie 2003). Similarly, Lowe (1951) noted that high flows on the Bann River, Ireland, delayed European eel (*A. Anguilla*) elver migrations for many weeks.

Elver feeding behavior

Dutil et al. (1989) found that the stomachs of elvers contained 90% *Chironomidae* and 8% *Simuliidae*. No food remains were found in the stomachs or intestines of glass eels (Dutil et al. 1989).

Glass eel competition and predation

Glass eels are preyed upon by many fish species including striped bass. American eel were found in 20% of striped bass stomachs in the Merrimack River, New Hampshire. Additionally, migrations of striped bass coincide with upstream elver migrations (reviewed in Richkus and Whalen 1999). Jessop (2000) found that a major source of predation on American eel elvers in the East River, Chester, Nova Scotia, was cannibalism by larger individuals of the same species. Other authors have also reported cannibalism on younger American eel (Tesch 2003).

Part D. Yellow-phase American Eel Habitat

Geographic and temporal movement patterns

Some yellow-phase American eel continue migrating upstream until they reach maturity, while others remain in the lower portions of coastal estuaries and rivers (Morrison et al. 2003; Cairns et al. 2004; Lamson et al. 2006). Morrison et al. (2003) studied the migration histories of yellow eels using otolith microchemistry. Yellow eels in the Hudson River, New York, showed three modes of habitat use: 1) the freshwater mode, in which yellow eels and elvers utilized only freshwater habitats; 2) the mixed mode, where American eel resided in freshwater for at least 2 years before migrating back to brackish water; and 3) the brackish mode, where American eel remained entirely in brackish habitats, without ever utilizing freshwater environments (Morrison et al. 2003). Individuals that exhibited the brackish mode had increased growth rates, earlier maturation, and began their downstream migrations sooner than those that utilized freshwater habitats (Morrison et al. 2003; Cairns et al. 2004; Lamothe et al. 2000). These findings support the Helfman et al. (1987) hypothesis that brackish water habitats are more productive than freshwater for American eel.

Lamson et al. (2006) also used microchemistry to trace movements of American eel in Prince Edwards Island, Canada. Findings of this study showed that 69% of individuals moved between salt and freshwater. Half of the freshwater American eel sampled showed freshwater residency only. The authors state that this may have been due to distances to other salinity zones or dams that impede movements. American eel were also found to be able to complete their lifecycle entirely in brackish water habitats (Lamson et al. 2006). Other research (Thibault et al. 2007) indicates that movements between freshwater and estuarine zones may be regular and seasonal in nature, as a response to low winter temperatures in the estuary.

Movement of yellow eels and upstream migrations occur primarily at night from dusk to dawn. However, movement does sometimes occur during the day (Dutil et al. 1988; McGrath et al. 2003c; Verdon et al. 2003). Some studies have indicated that American eel migrate in response to the lunar cycle, with individuals being less active during moonlit periods (Sorensen and Bianchini 1986; Cairns and Hooley 2003; Hildebrand 2005). Other studies indicate that high tides and increased river flow may increase movements (Dutil et al. 1988; Hildebrand 2005). Dutil et al. (1988) found that American eel moved upstream during high tides and were more than two times as active during high tides compared with low tides.

Yellow eels remain in freshwater and brackish systems for up to 30 years before maturing into silver eels and migrating to the sea to spawn (Tesch 1977; Helfman et al. 1987; Able and Fahay 1998). Few young American eel are found in inland lakes (Hurley 1972; Facey and LaBar 1981); migrants to farther reaches upstream tend to be older, larger, more mature females (Helfman et al. 1987; Haro and Krueger 1991; Oliveira 1999; Morrison et al. 2003).

American eel migrations upstream occur from March through October, and peak in May and July depending on location (Richkus and Whalen 1999). McGrath et al. (2003c) found that the numbers of American eel in the St. Lawrence River, New York, approaching the Moses-Saunders Power Dam peaked in early July and early October. Verdon et al. (2003) found that American eel in the Richelieu River, Quebec, began upstream migrations as early as June 11th and ended in late September. Hildebrand (2005) found that in the Shenandoah River, West

Virginia, American eel utilized the eel ladder at Millville Dam from March through October (the duration of time that the ladder was installed).

There is substantial evidence that some American eel establish a home range (Table 7-1). A home range is defined as the spatial extent or outside boundary of an animal's movement during the course of its everyday activities (Burt 1943). The size of the home range can be influenced by food availability, competition, and predator density (Bozeman et al. 1985). Ford and Mercer (1986) found some evidence of a home range and territoriality, and found that larger American eel were located primarily in large creeks, while smaller American eel were found in narrow creeks at the back of the marsh, in the Great Sippewisset Marsh, Massachusetts. They found that 93% of the American eel in their study traveled less than 100 m (Ford and Mercer 1986).

Citation	Home Range	Method	Waterbody Type
Ford and Mercer 1986	0.0209 ha	Areal analysis	Tidal creek
Dutil et al. 1988	0.5 – 2.0 ha	Linear distance	Tidal river
Parker 1995	~325 ha	Areal analysis	Tidal estuary
Gunning and Shoop 1962	<137 m	Mark-recapture	Estuary
Helfman et al. 1983	~1 ha	Polygons	Estuarine stream
Oliviera 1997	Max 4.7 km	Mark-recapture	River
Morrison and Secor 2003	Max 4.2 km	Mark-recapture	River
La Bar and Facey 1983	2.4 – 65.4 ha	Displacement polygons	Bay/lake
Thomas 2006	3 ha (18 ha)	50% (95%) kernels	Impounded lake

Table 7-1. Yellow-phase American eel home range estimates (adapted from Thomas 2006)

Parker (1995) found that homing in yellow-phase American eel in the Penobscot Estuary, Maine, was precise. More than half of the displaced American eel returned to within 300 m of their capture site, and three American eel moved towards their capture sites, but did not arrive there while under observation. Some of the American eel returned to within 50 m of the capture site and remained there for several days, indicating that the American eel returned to a specific area and not just a general location. In another study, Lamothe et al. (2000) found that American eel returned to home ponds after being moved to an adjacent pond.

Morrison and Secor (2003) also found that American eel in the Hudson River established a home range. They found that more than 70% of their PIT tagged American eel moved less than 1 km from the original tagging area in a 2 to 12 month time period. The longest dispersal was 4.2 km from the tagging site. However, the authors did suggest that based on otolith microchemistry, some American eel may have dispersed in the estuary over longer time periods (Morrison and Secor 2003).

Habitat influence on sexual differentiation

Many studies indicate that sex ratios of American eel are highly variable. This occurs both regionally and within individual systems (Hansen and Eversole 1984; Helfman et al. 1984). Hansen and Eversole (1984) found that females dominated males 23 to 1 in the Cooper River, South Carolina. Helfman et al. (1984) found that 36% of the American eel in a Georgia estuary were male, while only 6% of American eel in freshwater were male. Goodwin and Angermeier (2003) determined that while presence of male American eel in the Potomac River tributaries ranged from 0 to 100%, in the mainstem river 29% of individuals were male. Furthermore, 100% of the American eel in the Shenandoah River were female (Goodwin and Angermeier 2003). Further north, Oliveira et al. (2001) found that 49% to 98% of American eel in the Chandler, East Machias, and Sheepscot Rivers, Maine, were male. Additionally, females have been reported as dominant in most Canadian habitats (Gray and Andrews 1971; Dolan and Power 1977; Jessop 1987).

Various hypotheses have been developed to explain the skewed sex ratios. Vladykov (1966) hypothesized that females were found predominately in higher latitudes, while males were found in lower latitudes. However, Krueger and Oliveira (1997) and Oliveira et al. (2001) found the opposite. Krueger and Oliveira (1997) found that males outnumbered females 3 to 1 in the Annaquatucket River, Rhode Island. In addition, Oliveira et al. (2001) found that males made up 0% to 98% of the population within a single degree of latitude between Maine and Nova Scotia rivers.

Another theory is that female American eel are found in freshwater, while males are found in estuaries (Vladykov 1966; Tesch 1977). However, Winn et al. (1975) contradicted this hypothesis when they found more males in freshwater habitats and more females in estuaries in Rhode Island. Alternatively, Helfman et al. (1987) suggested that males were found in estuaries because these productive habitats led to fast growth. Females, on the other hand, preferred freshwater habitats that led to slower growth and increased fecundity (Helfman et al. 1987).

Helfman et al. (1987) also proposed that females delayed metamorphosis until they reached areas of higher latitude. However, Oliveira et al. (2001) found high variation in the proportion of males along a 30 km stretch of the Chandler, East Machias, and Pleasant Rivers in Maine, suggesting that delayed metamorphosis by females was unlikely.

Oliveira et al. (2001) also found that the proportion of males was inversely related to the amount of available lacustrine habitat; this finding was independent of distance inland. American eel from lacustrine habitats were found to be female, while samples from fluvial habitats were mostly male. The researchers concluded that river habitat may affect the distribution of sexes and play a role in sexual determination (Oliveira et al. 2001).

Other evidence suggests that density of American eel plays the key role in determining the sex of an individual; males are produced in high density areas, and females in low density areas. Thus, females are more common in upper reaches of rivers where density is lowest (Krueger and Oliveira 1999). Oliveira (1999) and Oliveira et al. (2001) hypothesize that males are produced in areas where crowding is occurring. Furthermore, males favor areas closer to the sea and spawning ground in more productive habitats, where they can grow and mature faster (Helfman et al. 1987). On the other hand, females tend to disperse widely within their range and utilize all suitable habitats. They favor slower growth and greater size, thus increasing fecundity and swimming ability (Krueger and Oliveira 1999; Goodwin and Angermeier 2003). In fact, in

upper reaches of rivers, American eel tend to mature at older ages and larger sizes (Helfman et al. 1987).

Yellow eels and the saltwater interface

Salinity is not likely a key habitat parameter for American eel, as they are found in a wide range of salinities (Morrison et al. 2003). Geer (2003) reported that in the Chesapeake Bay, Virginia, more American eel were present in the upper tributaries near or above the saltwater interface. Eighty-nine percent were caught in salinities below 12 ppt, and 27% of the catch occurred in waters less than 2 ppt (Geer 2003). Additionally, Dutil et al. (1988) found that American eel selected salinities less than 12 ppt in areas where mid-channel salinity levels reached 24 ppt.

While American eel do not, in general, seem to select habitats based on salinity, it may influence growth rates. Morrison et al. (2003) found that yellow eels that showed evidence of freshwater residency had slower growth rates than those that spent their entire lives in brackish water. Brackish water habitats are thought to have higher food abundances, better quality food, lower predation pressure, and less thermal and osmotic stress. Helfman et al. (1987) suggested that productivity was higher downriver in brackish habitats as compared to upriver habitat. Yellow eels in brackish water are thought to grow faster, mature earlier, and migrate downstream as silver eels sooner. Freshwater habitats are thought to lead to later maturation and overall larger individuals (Helfman et al. 1987).

Yellow eel substrate associations

Yellow-phase American eel are bottom/substrate oriented and may show little movement, particularly during the day (Eales 1968; Ogden 1970; Tesch 1977; LaBar and Facey 1983; Helfman 1986). However, the substrate preference of American eel is not well documented in the literature. LaBar and Facey (1983) reported that American eel in Lake Champlain were found over weedy bottoms. Ford and Mercer (1986) documented small American eel in soft-bottomed creeks of landward marshes, and larger American eel in soft mud to sandy-bottomed creeks of seaward marshes. Geer (2003) found that in the Chesapeake Bay, Virginia, American eel were mostly found over detritus, hydroid, or shell bottoms. Chaput et al. (1997) state that American eel in the St. Lawrence River use soft sediments to burrow during the winter.

Thomas (2006) suggested that riparian vegetation and complex substrate were important to yellow-phase American eel in impounded systems. Additionally, American eel were more likely to be found in areas with coarser substrates (i.e., sand, gravel, or rock) in the morning-afternoon, and winter-spring because individuals were less active and seeking shelter during those times. However, during comparatively more active times (i.e., evening-night and summer-fall) in an impounded system, American eel were more likely to be in areas with finer substrates (i.e., silt or clay) (Thomas 2006).

Yellow eel depth associations

Little information exists regarding the depths at which American eel are found. Due to the diverse range of habitats that American eel utilize, depth range probably varies greatly. Facey and LaBar (1981) found American eel in water 1 to 2 m deep. Geer (2003) found that the majority of yellow eels were caught in the upper tributaries of the Chesapeake Bay in depths of 4 to 10 m.

Thomas (2006) found that yellow-phase American eel in an impounded system typically occupied depths of 0.4 to 1.5 m (available depths of 0 to 2.93 m). In addition, while mean morning (1.1 m) and afternoon (1.1 m) depths were relatively shallow, mean evening (1.3 m) and night (1.4 m) depths were slightly deeper. Given the relatively shallow nature of the impounded system, these changes in depth usually represented areas with different substrate and variable distances from shore. Furthermore, mean winter (0.8 m) and spring (0.9 m) depths showed use of shallow habitat, while mean summer (1.2 m) and fall (1.3 m) depths showed use of deeper areas. Therefore, American eel utilization of different depth areas may be dependent upon time of day and season (Thomas 2006).

Yellow eel water temperature

Researchers hypothesize that the onset of upstream migration in yellow eels is linked to water temperature (Moriarty 1986; Haro and Krueger 1991; EPRI 1999). Knights and White (1998) found that European eel were stimulated to migrate by temperatures greater than 14 to 16°C, and increases in migrations occurred at temperatures greater than 20°C. Similarly, Verdon et al. (2003) determined that migration occurred earlier in the Richelieu River, Quebec, than in the upper St. Lawrence River. The St. Lawrence is a larger lake-fed system that has more gradual and less variable temperature increases than the Richelieu system; the researchers hypothesized that this pattern might cause a delayed upstream migration (Verdon et al. 2003). In the upper St. Lawrence River, upstream migration begins in late June and peaks at the end of July (Verdon and Desrochers 2003).

Verdon and Desrochers (2003) found that captures of American eel in the St. Lawrence River peaked when temperatures reached 22 to 23°C, and decreased as water temperatures dropped from 24°C to 21°C. Once the temperatures fell below 21°C, captures of American eel became scarce (Verdon and Desrochers 2003). McGrath et al. (2003c) noted a decrease in migrant yellow eels at the Moses-Saunders Power Dam in the St. Lawrence River, when temperatures declined to 10°C in the fall. Additionally, Geer (2003) reported that American eel in the Chesapeake Bay, Virginia, were found between 13°C and 27°C. They were most abundant in waters where the temperature was 26 to 28°C and least abundant in waters less than 8°C. Low catch rates at these temperatures suggested inactivity. However, researchers found no direct correlation between temperature and catch, although peaks seemed to coincide with increased temperature (Geer 2003). Haro (1991) determined the range of preferred temperatures for yellow eels in a freshwater laboratory was between 17.8 and 19.8°C.

Yellow eels live in a variety of habitats, including cold, high-elevation or high-latitude freshwater streams and lakes, to warm, brackish coastal bays and estuaries in the Gulf of Mexico (Facey and Van den Avyle 1987). American eel have been reported to survive passage through a nuclear power plant, where they were exposed to elevated temperatures for 1 to 1.5 hours (Marcy

1973). Furthermore, American eel are thought to become torpid at temperatures less than 10°C. Walsh et al. (1983) held yellow eel at 5°C for over five weeks, and found that at temperatures less than 8°C they stopped feeding and remained inactive for months.

Yellow eel dissolved oxygen associations

Rulifson et al. (2004) found that catch of American eel was affected by dissolved oxygen rates, and determined that dissolved oxygen was a strong predictor of the distribution of American eel in North Carolina. High catches of American eel were almost always in waters with dissolved oxygen levels above 4 mg/L (Rulifson et al. 2004). Similarly, Geer (2003) found that 82% of the American eel caught in the Chesapeake Bay, Virginia, were found in waters with dissolved oxygen levels between 5 and 9 mg/L. However, no association was found between dissolved oxygen and catch (Geer 2003). This could be due to the fact that sampling was conducted only in the areas with dissolved oxygen levels above 5 mg/L (Rulifson et al. 2004).

Yellow eel water velocity/flow

Yellow eels are likely not water velocity dependent, as high densities of American eel have been found in lakes and ponds where velocity is low or nonexistent (K. McGrath, New York Power Authority, personnel communication). However, Wiley et al. (2004) found that in Maryland, velocity-depth diversity was the only stream habitat variable related to American eel density. The highest densities of eel occurred in sites that had four velocity-depth regimes: slow (<0.3 m/s)-deep (>0.5 m/s), slow-shallow (<0.5 m/s), fast (>0.3 m/s)-deep, and fast-shallow. Sites with only one of two velocity-depth regimes had significantly lower American eel densities (Wiley et al. 2004).

Yellow eel feeding behavior

The yellow eel phase is the feeding and growth stage for the American eel. American eel are thought to be opportunistic feeders, preying upon whatever is available in their habitat (Colette and Klein-MacPhee 2002). American eel can feed heavily on demersal fish eggs, larvae, and juveniles (Knotek and Orth 1998). Mature American eel have been documented feeding on invertebrates including insects, crayfish, snails, worms, and small fish (Ogden 1970; Scott and Crossman 1973; Facey and LaBar 1981). They have also been documented consuming plant material (Moriarity 1978) and carrion (Ogden 1970). Additionally, cannibalism on smaller conspecifics has been documented in the literature (Domingos et al. 2006).

Godfrey (1957) found that 90% of the American eel's diet consisted of insects, while 10% consumed whole fish. Facey and LaBar (1981) reported that American eel feed heavily upon benthic organisms. They found that 43% of stomachs contained insects, 26% contained fish and crayfish, and 20% contained gastropods. The rest of the stomachs were empty. The authors noted that American eel in this study consumed fish more than in other studies, and suggested that yellow eels in Lake Champlain, Vermont, relied more on fish due to their large sizes (Facey and LaBar 1981). In another study, Wenner and Musick (1975) documented American eel preying heavily on blue crabs (*Callinectes sapidus*) and bivalves (*Mya arenaria*,

Mulinia lateralis, and *Macoma* spp.) in the James, York, and Rappahannock Rivers, Virginia. They also found that American eel preyed upon alewife (Wenner and Musick 1975).

Denoncourt and Stauffer (1993) found that American eel in the Delaware River fed on 56 taxa, including 4 fish species and 52 macroinvertebrates. Macroinvertebrates were found in 98.8% of the feeding American eel. Mayflies (Ephemeroptera) and stoneflies (Plecoptera) made up 69% of the prey items, followed by caddisflies (Trichoptera, 33.9%), beetles (Coleoptera, 23.4%), flies (Diptera, 16.4%), fishflies and hellgrammites (Megaloptera, 12.8%), and dragonflies and damselflies (Odonata, 11.1%). Fish species were found in 7% of the feeding American eel and included lamprey ammocetes (*Petromyzon marinus*), madtoms (*Noturus insignis*), and minnows (*Notropis* sp.). Other items in the stomachs included detritus and vegetation, bones and flesh, and sand and gravel (Denoncourt and Stauffer 1993).

Lookabaugh and Angermeier (1992) also found that prey size increased with the size of the American eel. In the piedmont regions of the James River drainage (Virginia), small American eel fed primarily on aquatic insects, whereas larger American eel consumed fish and crayfish (Decapoda). In the coastal plain, small and medium sized American eel preyed upon microcrustaceans and aquatic insects, while large American eel fed on crayfish. Similarly, Ogden (1970) determined that smaller American eel (less than 40 cm) in New Jersey streams mostly fed on aquatic insect larvae, including Ephemeroptera, Megaloptera, and Trichoptera, while the larger American eel consumed fish and crustaceans. Smith (1985) also reported smaller American eel feeding on mayflies, magalopterans, and caddisflies. In addition, Rulifson et al. (2004) found that in North Carolina, large American eel consumed crayfish and fish (mullet and centrarchids). Smaller American eel fed on arthropods, small mullet and minnows, polychaetes, unidentifiable matter, and plant material. Fish, crustaceans, and arthropods were the most important prey items (Rulifson et al. 2004).

In addition, Sorensen et al. (1986) reported that in Rhode Island American eel fed primarily at night, with activity peaking at nightfall.

Part E. Silver American Eel Habitat

Geographic and temporal patterns at sea

Once American eel enter their final life stage, termed silver-phase, the maturation process accelerates and they migrate out to the Sargasso Sea to spawn. In New England tributaries, spawning migrations begin in the late summer and continue through fall. American eel migrate later in the Southeastern states and in the Mid-Atlantic than in the Northern states. It is hypothesized that this delay helps to synchronize the arrival of the American eel at the spawning grounds in the Sargasso Sea (Wenner 1973; Facey and Helfman 1985; Helfman et al. 1987).

Yellow eels transform into silver eels before migrating out to sea. Little is known about this final phase of their life history (ASMFC 2000). Downstream migrations occur in sudden bursts with long periods of no movement and peaks of intensive movements (Barbin et al. 1998). The rate of migration varies, with pauses occurring while the silver eels wait for specific environmental cues (Richkus and Whalen 1999).

Silver eel migration begins at different times of year depending on location, but occurs primarily in the fall, although winter migrations have been documented (Facey and Helfman 1985; Euston et al. 1997, 1998). In Newfoundland, the largest American eel migrations occur in late September and early October (Bouillon and Haedrich 1985). McGrath et al. (2003a) found that American eel in the upper portion of the St. Lawrence River migrated downstream from the end of June to the beginning of October, and that the primary migration in the lower estuarine portion of the river occurred in October. Slightly south, Winn et al. (1975) documented American eel migrating in Rhode Island from September through November.

Migration of mature American eel is thought to occur mostly at night (Winn et al. 1975; Haro et al. 2000a; McGrath et al. 2003b). Haro et al. (2000a) stated that silver eels in the Connecticut River, Massachusetts, migrated primarily at night within several hours after sunset, and became inactive during the day. The variables thought to influence downstream migration of silver eels include water temperature, river and stream discharge, odor, and light-intensity, including moon phase (Hain 1975; Westin 1990; Haro 1991; Richkus and Whalen 1999; Richkus and Dixon 2003). In fact, research has indicated that catch rates of American eel are higher during the dark phases of the moon and when cloud cover is highest (Winn et al. 1975; Cairns and Hooley 2003; McGrath et al. 2003b). Cairns and Hooley (2003) found that in tidal bays and estuaries in Prince Edward Island, Canada, catch per unit effort (CPUE) for silver and yellow eels decreased at full moon. CPUE was negatively correlated with the proportion of moon fullness and was negatively correlated with the illuminance index (Cairns and Hooley 2003). Cairns and Hooley (2003) suggest that this is a mechanism to avoid predation. Furthermore, some studies indicate that American eel exhibit an endogenous lunar cycle of activity (Boëtius 1976; Hain 1975; Edel 1976).

Rainfall, which leads to increased river discharge, may also have an impact on silver eel migrations (Lowe 1951; Winn et al. 1975; Charles Mitchell & Associates 1995; Euston et al. 1997, 1998). Winn et al. (1975) noted increased migrations after rains, as well as during the third and fourth lunar quarter. Haro et al. (2003) found in Maine that more American eel were captured on, or soon after, days with rain than on dry days.

The age and size at which migration begins varies geographically. American eel in the northern part of the range exhibit slower growth and remain longer in freshwater and estuarine systems before beginning migration back to sea (Facey and LaBar 1981). Various studies in Newfoundland, Lake Ontario, and Lake Champlain have shown that American eel migrate back to sea after about 12 to 13 years, and at a mean size of 69 cm (Gray and Andrews 1971; Hurley 1972; Facey and LaBar 1981; McGrath et al. 2003a). In the southern part of their range, American eel begin migrating earlier than in the north (Hansen and Eversole 1984; Helfman et al. 1984; Owens and Geer 2003). Hansen and Eversole (1984) found that in the Cooper River, South Carolina, American eel older than 7 years old and greater than 65 cm in length were sparse, suggesting that adults migrate at a younger age and smaller size. Helfman et al. (1984) found similar results in the Altamaha River, Georgia. More recently, Owens and Geer (2003) found that populations in Virginia tidal rivers were comprised mostly of American eel less than 7 years old, indicating that migrations had occurred by this age.

Silver eel salinity associations

The importance of salinity to silver-phase American eel has not been documented in the literature. As a habitat generalist, American eel utilize a wide variety of salinities from freshwater to saltwater, thus migrations occur through a broad range of salinities. Barbin et al. (1998) suggested that changes in salinity could be used as a mechanism to help orient American eel out of estuaries. These researchers documented American eel in the Souadabscook stream (tributary to the mouth of the estuary) and the Penobscot Estuary, Maine, in salinities ranging from 0 to 30 ppt (Barbin et al. 1998).

Silver eel substrate associations

There is little information documented in the literature on the substrate requirements of silver-phase American eel. One study by Valdykov (1955) reported that silver eels in the northern habitats utilized muddy substrates during the winter months. Goodwin and Angermeier (2003) found that the highest catch of American eel in Shenandoah River drainages appeared to be associated with site characteristics including leaf packs, rootwads, woody debris, and flowing water.

Silver eel depth associations

Depth does not appear to be an important habitat characteristic for silver-phase American eel, as authors have documented use of a wide range of depths during outmigrations. Haro et al. (2000a) found that silver eels in a hydroelectric forebay on the Connecticut River, Massachusetts, used many depths, but occupied depths most frequently between 6.6 and 10 m. However, American eel were also observed swimming at night near the surface of the water (Haro et al. 2000a). Similarly, McGrath et al. (2003b) found, during their surface and midwater trawling study, that American eel were caught at the highest rates between 6 and 10 m. However, the researchers stated that they were unsure if these findings were significant since sampling was limited near the bottom (between 18 and 24 m) (McGrath et al. 2003b).

Barbin et al. (1998) documented eels occupying a variety of depths in the Penobscot Estuary, Maine. The researchers found that American eel moved freely between surface waters and the bottom, and that when movement occurred, it was near the surface on ebbing tides (Barbin et al. 1998).

Upon entering the ocean, American eel appear to migrate in the upper water column. Evidence for this includes physiological changes, including the color change, changes to the visual system, and morphological changes to the swim bladder (McCleave and Kleckner 1985). The color change from yellow to silver provides the American eel with a more countershaded appearance. This form of camouflage is thought to only be effective in the photic zone of the ocean, possibly only in the upper 600 m (McCleave and Kleckner 1985). Other fishes found below 600 m are often dark and not countershaded (Marshall 1971, 1972).

American eel also undergo changes in vision, including an increased eye diameter, an increase in retinal surface area, the addition of new rod cells, an increase of convergence of rods on each neural pathway, decreases in cone density, and changes in vision pigments (Winn et al. 1975; Beatty 1975; Pankhurst 1982; Pankhurst and Lythgoe 1982, 1983). These changes allow the American eel to adapt to the low light conditions they would likely be migrating through (Jerlov 1976; McCleave and Kleckner 1985). Lastly, the swim bladder changes during metamorphosis, allowing American eel to maintain an inflated swim bladder at greater depths (Kleckner 1980).

Tesch (1978a, 1978b) tracked European silver eels (*Anguilla anguilla*) over the European continental slope and found that they swam at depths between 50 and 400 m; the maximum depth in this area was 2000 m. However, the tracking was terminated prematurely due to pressure-transmitter failure. Additionally, Wenner (1973) documented American eel at depths ranging from 15 to 68 m in the Chesapeake Bay, Maryland, and Cape Cod, Massachusetts. The deepest known record for *Anguilla* was reported by Robins et al. (1979) as approximately 2000 m.

Silver eel temperature associations

Temperature may be an important trigger for migrating silver eels, which travel during the fall and winter months. Vøllestad et al. (1986) documented that migrating European eel in Norwegian streams showed the most activity in a temperature range of 9ºC to 18ºC. Similarly, Barbin et al. (1998) documented American eel migrating in September and October in the Penobscot Estuary, Maine, in water temperatures ranging from 9.6ºC to 17.6ºC. Moreover, commercial fishermen in the Elbe estuary have noted that lingering summer temperatures into the fall cause a delay in migration (Tesch 2003).

Like juveniles, mature silver eels utilize a broad range of habitats, and thus are likely to tolerate a wide range of temperatures (Facey and Van den Avyle 1987). A few studies have been done to determine the preferred temperatures of American eel. Barila and Stauffer (1980) reported a temperature preference of 16.7°C, while Karlsson et al. (1984) found that American eel preferred a temperature of 17.4 ± 2.0°C. Haro (1991) reported preferred temperatures of 19.6°C for unmatured silver eels in freshwater, and 15.8 to 18.9 °C for unmatured silver eels acclimated to saltwater.

Silver eel feeding behavior

Silver phase American eel presumably do not feed during their migration to the Sargasso Sea (Gray and Andrews 1971).

Silver eel competition and predation

American eel are preyed upon by many different species, including fish, aquatic mammals, birds, and mammals (mink) (Sinha and Jones 1967; Seymour 1974). However, the importance of American eel as a food source for other animals has not been well recorded in the literature (ASMFC 2000). Thompson et al. (2005) documented the American bald eagle using American eel as a food source. In the Hudson River, New York, 50% of the bald eagle's diet was comprised of 3 fish species, one of which was the American eel (Thompson et al. 2005).

Section II. Identification and Distribution of Habitat Areas of Particular Concern for American Eel

Habitat types that qualify as Habitat Areas of Particular Concern (HAPCs) for American eel include the spawning and hatching grounds, nursery and juvenile habitat, and adult habitat.

Oceanic waters of the Sargasso Sea comprise the spawning and hatching grounds for American eel. This is the only suspected location of reproduction for American eel, and therefore, is essential to the survival of the species. Little is known about American eel habitat in the Sargasso Sea, and the exact location of spawning and hatching has not been identified.

Continental Shelf waters usher the final stage of the larval American eel migration into coastal waters, and are important to larval feeding and growth. This is also where American eel metamorphose into the glass eel stage. Silver-phase eels also cross the shelf during their migration to the Sargasso Sea.

Estuaries and freshwater habitat, including rivers, streams, and lakes, serve as juvenile, sub-adult, and adult migration corridors, as well as feeding and growth areas for juveniles and sub-adults (ASMFC 2000). After American eel larvae transform into glass eels over the continental shelf, they enter estuaries, and ascend the tidal portions of rivers. Glass eels metamorphose into the elver life stage and either continue upstream movements, or cease migrating in the lower saline portions of estuaries and rivers. These estuaries and freshwater habitats serve as foraging grounds for American eel and are important for growth and maturation. American eel can remain in these systems for up to thirty years before maturing and returning to sea.

While estuarine and riverine habitats have been identified as important for the rearing and growth of American eel, many studies failed to find specific American eel habitat associations within them (Huish and Pardue 1978; Meffe and Sheldon 1988; Smogor et al. 1995; Bain et al. 1988; Wiley et al. 2004). Huish and Pardue (1978) found no difference in American eel abundance in relation to width, substrate, flow, and depth in North Carolina streams. Likewise, Bain et al. (1988) found that American eel habitat use was not related to specific habitat features including depth, water velocity, and substrate in two Connecticut River tributaries. Wiley et al. (2004) also did not find any eel-stream habitat relations. The researchers found that eel density was correlated with distance from the ocean (Wiley et al. 2004). While anguillid eels have the ability to survive in a wide variety of habitats, water quality is still an important factor to their health and survival.

Given the great variation in demographics that occurs across latitudinal and distance-inland gradients, all areas may not contribute equally to American eel production and recruitment. Despite this, geographic patterns of differential recruitment are unexplored. This issue must be addressed before identifying specific Habitat Areas of Particular Concern.

Section III. Present Conditions of Habitat and Habitat Areas of Particular Concern for American Eel

Habitat quantity

Much of American eel habitat has not been quantified. American eel utilize a wide range of habitat types throughout their life history, including the Sargasso Sea, oceanic waters off the continental shelf, estuaries, and rivers. Some researchers think that habitat availability for American eel growth areas is rapidly declining. An extreme example by Busch et al. (1998) showed that stream habitat for American eel was reduced from 556,801 km to 90,755 km by dams (assuming that all dams completely block all migration). According to Busch et al. (1998), 15,115 dams block upstream and downstream migrations. Fortunately, American eel are habitat generalists, and therefore may be somewhat resilient to impacts on habitat availability. The increased human impact on aquatic habitat in recent years may not have had as high an impact on American eel as on other diadromous species because they are able to survive and thrive under a variety of conditions.

In general, the use of the estuarine and marine habitat by American eel is less well known than freshwater habitat utilization. Consequently, little information is known on requirements for mature, egg, and larval stages of this species. This is important to note because the marine and estuarine portions of the total population could be quite significant.

Habitat quality

The quality of American eel habitat has been impacted by human actions. Since European settlement, habitat loss has potentially contributed to a possible decline in stocks. However, anthropogenic impacts on American eel at the population level are poorly understood and the magnitude of these threats remains unknown.

Section IV. Significant Environmental, Temporal, and Spatial Factors Affecting Distribution of American Eel

Table 7-2. Significant environmental, temporal, and spatial factors affecting distribution of American eel. This table summarizes the current literature on American eel habitat associations. For most categories, optimal and tolerable ranges have not been identified, and the summarized habitat parameters are listed under the category reported. In some cases, unsuitable habitat parameters are defined. NIF = No Information Found. N/A = Not Applicable. Reported = Ranges or information recorded in the literature.

Life Stage	Time of Year and Location	Depth (m)	Temperature (°C)	Salinity (ppt)	Substrate	Current Velocity (m/sec)	Dissolved Oxygen (mg/L)
Silver eel (spawning)	From February to April in Sargasso Sea; spawn in thermal fronts that separate southern and northern water masses; 19.2°N to 29°N and 52°W to 79°W	**Tolerable:** NIF **Optimal:** NIF **Reported:** Upper few hundred meters of the water column	**Tolerable:** NIF **Optimal:** NIF **Reported:** 18-19	**Tolerable:** NIF **Optimal:** NIF **Reported:** Maximum 36.6	**Tolerable:** N/A **Optimal:** N/A **Reported:** N/A	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF
Egg	Same as spawning	**Tolerable:** NIF **Optimal:** NIF **Reported:** Assumed that hatching occurs in the water column	**Tolerable:** NIF **Optimal:** NIF **Reported:** Hatching occurs at 23 in Japanese eel	**Tolerable:** NIF **Optimal:** NIF **Reported:** Sea water	**Tolerable:** N/A **Optimal:** N/A **Reported:** N/A	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF
Leptocephali (larval)	Larvae drift and swim in upper water column for a couple of months; distributed by water currents (e.g., Antilles, Florida, and Gulf Stream); transform into glass eels over continental shelf	**Tolerable:** NIF **Optimal:** NIF **Reported:** Found in upper 250; vertical migrations occur 50-300	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** Sea water	**Tolerable:** N/A **Optimal:** N/A **Reported:** N/A	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF

Life Stage	Time of Year and Location	Depth (m)	Temperature (ºC)	Salinity (ppt)	Substrate	Current Velocity (m/sec)	Dissolved Oxygen (mg/L)
Glass eel/elver	Migrations from late winter to early spring (earlier in South than North) into estuaries, and ascend rivers along Atlantic coast	**Tolerable:** NIF **Optimal:** NIF **Reported:** Variety	**Tolerable:** NIF **Optimal:** NIF **Reported:** 10-12 triggers migrations; utilize wide range of temperatures	**Tolerable:** NIF **Optimal:** NIF **Reported:** 0 – 25	**Tolerable:** NIF **Optimal:** NIF **Reported:** Burrow during the day and in between movements upstream in sand, mud, tubes, snags, plant masses, and other materials	**Tolerable:** >25 cm·s⁻¹ **Optimal:** NIF **Reported:** Upper limit is 35 cm·s⁻¹. Most will not swim in waters > 25 cm·s⁻¹	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF
Yellow eel	May continue migrating upstream (March through October; peak May/June) and eventually settle into any habitat available (e.g., estuaries, lakes, ponds, rivers, and streams) for up to 30 years	**Tolerable:** NIF **Optimal:** NIF **Reported:** Variable depending on system	**Tolerable:** Variable **Optimal:** NIF **Reported:** Migration thought to be linked to temperature; increases in migration occur >20	**Tolerable:** NIF **Optimal:** NIF **Reported:** Variety; high tolerance for changes in salinity	**Tolerable:** NIF **Optimal:** NIF **Reported:** Bottom/substrate oriented; most prefer coarse substrates	**Tolerable:** NIF **Optimal:** NIF **Reported:** Highest density in areas with a large variety of velocities and depths	**Tolerable:** NIF **Optimal:** NIF **Reported:** >4
Silver eel (migratory)	Migrate out to sea after maturation; migrations begin at different times depending on location, but occur mostly during the fall, although winter and summer migrations have been documented	**Tolerable:** NIF **Optimal:** NIF **Reported:** Depths vary and may be related to light intensity and turbidity	**Tolerable:** NIF **Optimal:** 17.4±20; activity decreases around 12 **Reported:** Tolerate wide range of temperatures	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** N/A **Optimal:** N/A **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** N/A	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF

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Chapter 8

ATLANTIC STURGEON

(*Acipenser oxyrinchus oxyrinchus*)

Section I. Description of Atlantic Sturgeon Habitat

Atlantic Sturgeon General Habitat Description and Introduction

The Atlantic sturgeon (*Acipenser oxyrinchus oxyrinchus*) is an anadromous species found in Atlantic Coastal waters of the United States, and major river basins from Labrador (Churchill River, George River, and Ungava Bay), to Port Canaveral and Hutchinson Island, Florida (Van den Avyle 1984). Historically, Atlantic sturgeon once inhabited northern Europe as well, but since have become extinct (ASSRT 2007). According to historical records, important sturgeon fisheries existed in nearly all Piedmont river basins on the Atlantic Coast at some point in time (Goode 1887). Early accounts of sturgeon fishery landings did not distinguish between Atlantic sturgeon and the smaller shortnose sturgeon (*Acipenser brevirostrum*). However, it is likely that the accounts referred to the larger and more valuable Atlantic sturgeon. Following intense exploitation for food, and construction of mainstem river dams during the 19th and early 20th centuries, sturgeon populations were drastically reduced throughout their range and extirpated in some rivers (ASMFC 1998; USFWS-NMFS 1998; ASSRT 2007). Scientists believe that spawning populations of Atlantic sturgeon were extirpated from the St. Marys River in Georgia, the Housatonic River in Connecticut, the Connecticut River, the Taunton River in Massachusetts and Rhode Island, and all Maryland and Pennsylvania tributaries of the Chesapeake Bay (Burkett and Kynard 1993; Rogers and Weber 1995; ASMFC 1998; USFWS-NMFS 1998; ASSRT 2007).

Atlantic sturgeon are motile, long lived, and utilize a wide variety of habitats. Atlantic sturgeon require freshwater habitats for reproduction and early life stages, in addition to hard bottom substrate for spawning (Vladykov and Greeley 1963; Huff 1975; Smith 1985b). Coastal migrations and frequent movements between the estuarine and upstream riverine habitats are characteristic of this species (ASMFC 1998). Historical accounts describe captures of large sturgeon, most probably *A. oxyrinchus oxyrinchus*, during the summer and fall in fall-line habitats on the Savannah River (Lawson 1709). In some systems, Atlantic sturgeon may prefer extensive reaches of silt-free higher gradient boulder, bedrock, cobble-gravel, and coarse sand substrates for spawning habitat (Brownell et al. 2001). Juvenile and adult Atlantic sturgeon frequently congregate in upper estuary habitats around the saltwater interface, and may travel upstream and downstream throughout the summer and fall, and during late winter and spring spawning periods. Adult Atlantic sturgeon may spend many years between spawning periods in marine waters (Brundage and Meadows 1982; Bain 1997; ASMFC 1998; USFWS-NMFS 1998; Savoy and Pacileo 2003; ASSRT 2007).

Due to a variety of anthropogenic impacts, including river impoundments, water quality deterioration, and overfishing, only 20 of the 35 existing stocks of Atlantic sturgeon are reproducing, with many stocks likely at historically low levels (ASSRT 2007). In 1991, Atlantic sturgeon was listed as a candidate species (56 FR 26797) under the Endangered Species Act (ESA) and remained on the revised list in 1997 (62 FR 37560). In 1998, a status review of Atlantic sturgeon found that the continued existence of Atlantic sturgeon was not threatened by any of the five ESA listing factors. Therefore, Atlantic sturgeon was not listed as a threatened or endangered species (USFWS-NMFS 1998). In 2003, the National Marine Fisheries Service (NMFS) and U.S. Fish and Wildlife Service (USFWS) held a workshop on the "Status and Management of Atlantic Sturgeon" to discuss the current status along the Atlantic coast to

determine what obstacles, if any, were impeding the recovery of Atlantic sturgeon. The results of the conference reported "mixed" reviews where some populations seemed to be recovering while others were declining. Bycatch and habitat degradation were noted as possible causes for some population declines (Kahnle et al. 2005). Based on the information gathered from the 2003 workshop on Atlantic sturgeon, NMFS initiated a second status review in 2006, and the results are currently under consideration by the Secretary of Commerce, as to whether the species warrants listing as threatened or endangered (W. Patrick, NOAA Fisheries Service, personal communication).

In 1990, the Atlantic States Marine Fisheries Commission wrote a Fisheries Management Plan (FMP) for Atlantic sturgeon, which was amended in 1998. In 1998, the ASMFC closed all Atlantic sturgeon fisheries coastwide in the United States, and recommended a 20 to 40 year moratorium so that the spawning stock of the slow-reproducing fish could be restored to a level where 20 year classes of adult females are present (ASMFC 1998). This action was followed by NMFS with a similar moratorium in Federal waters (K. Damon-Randall, NOAA Fisheries Service, personal communication).

Much of the habitat information on Atlantic sturgeon remains incomplete. Due to the relatively low numbers of fish in many river basins, habitat utilization patterns have been difficult to establish with certainty (Collins et al. 2000a). Life history, behavior, and movements have been most thoroughly documented in the Hudson River, New York, while many other river systems are lacking in vital life history information (Bain 1997; Bain et al. 2000; Gross et al. 2002). Below is a discussion of some of the general habitat requirements for the Atlantic sturgeon.

Part A. Atlantic Sturgeon Spawning Habitat

Since adult Atlantic sturgeon migrate through rivers and estuaries during their spawning migration, the discussion of adult Atlantic sturgeon estuarine and spawning habitat utilization patterns will be combined in this section. For the purposes of this report, female spawning adults are considered to be at least 15 years of age, and are a minimum of 1800 mm fork length (FL) or 2000 mm total length (TL). Male adult Atlantic sturgeon are 12 to 20 years of age, and between 1350 and 1900 mm FL or 1500 and 2100 mm TL (Bain 1997). See Table 8-1 for information on length-at-age.

Life Interval	Age Range (years)	Fork Length (mm)	Total Length (mm)
Larvae	<0.08		≤ 30
Juvenile	0.08-11	~20-1340	~30-1490
Non-spawning adults	≥ 12	≥ 1350	≥ 1500
Female spawners	≥ 15	≥ 1800	≥ 2000
Male spawners	12-20	$\geq 1350-1900$	$\geq 1500-2100$

Table 8-1. Age and size range of Atlantic sturgeon throughout their life cycle

Geographical and temporal patterns of migration

Atlantic sturgeon most often spawn in tidal freshwater regions of large estuaries (Hildebrand and Schroeder 1928; Bain 1997; Colette and Klein-MacPhee 2002; Moser and Ross 1995). This pattern is prevalent in New England and U.S. mid-Atlantic estuaries, where obstructions to migration at the fall line preclude upriver migration. In the South where many rivers remain unblocked, documentation shows that Atlantic sturgeon ascend hundreds of miles upstream into non-tidal rivers to spawn (M. Collins, South Carolina Department of Natural Resources, personal communication).

Spawning migrations are cued by temperature, which causes fish in U.S. South Atlantic estuaries to migrate earlier than those in mid-Atlantic and New England portions of their range (Smith 1985b). In Florida, Georgia, and South Carolina, spawning migrations begin in February. Collins et al. (2000b) found that in the Edisto River, South Carolina, ripe males were captured as early as March 2nd, and a single ripe female was captured on March 7th. Additionally, the researchers captured spent males as early as late March, and spent females as late as mid-May (Collins et al. 2000b). In contrast, researchers in the mid-Atlantic region report that spawning migrations for Atlantic sturgeon begin between April and May (Hildebrand and Schroeder 1928; Secor and Waldman 1999; Dovel and Berggren 1983; Bain et al. 2000). In New England and Canada, spawning migrations occur from May through July (Collette and Klein-MacPhee 2002). Furthermore, Hatin et al. (2002) reported that spawning occurred from early June to approximately July 20th, in the St. Lawrence River, Québec.

In addition to a spring migration, many studies document the occurrence of a fall migration (Smith et al. 1984; Smith 1985b; Collins et al. 2000b; Laney et al. 2007). Most fall migrations are movements out of the estuaries into marine habitats. Fall migrations occur from September through December, again, depending on the latitude (Smith 1985b). In addition, some researchers have proposed that an alternate fall migration into estuaries may be related to spawning (Smith et al. 1984; Rogers and Weber 1995; Weber and Jennings 1996, Moser et al. 1998; Collins et al. 2000b; Laney et al. 2007). Whether this fall migration results in fall spawning remains unknown. Smith et al. (1984) reported an upriver migration of fish in late August and September in South Carolina. Similarly, Collins et al. (2000b) noted the appearance of ripe males in South Carolina at the end of August and September; by October, 86% of the males were ripe. Furthermore, Collins et al. (2000b) tracked two sturgeon via radio and acoustic transmitters in the Edisto River, South Carolina. After spending the summer in the lower river, these fish migrated upriver to RKM 190 in October, which led the researchers to hypothesize that a fall spawning migration was occurring (Collins et al. 2000b). An alternative explanation is that the fall migration was comprised of fish that would reside in the upper river through the winter and spawn the following spring, as is reported to occur in Caspian Sea sturgeons (D. Secor, Chesapeake Biological Laboratory, University of Maryland Center for Environmental Science, personal communication). The general phenomenon of fall spawning remains uncertain and merits further study. Spring spawning, however, has been well documented in the literature (ASMFC 1998; USFWS-NMFS 1998; ASSRT 2007), and is most likely the dominant behavior of all North American sturgeon species.

Despite extensive mixing in coastal waters, Atlantic sturgeon return to their natal river to spawn as indicated from tagging records (Collins et al. 2000b; K. Hattala, New York State Department of Environmental Conservation, personal communication), and the relatively low

rates of gene flow reported in population genetic studies (King et al. 2001; Waldman et al. 2002; from ASSRT 2007).

Spawning location (geographical)

The following information on Atlantic sturgeon spawning location (geographical) along the Atlantic coast was excerpted from ASSRT (2007):

Maine Rivers

The geomorphology of most small coastal rivers in Maine is not sufficient to support Atlantic sturgeon spawning populations, except for the Penobscot and the estuarial complex of the Kennebec, Androscoggin, and Sheepscot rivers. During the summer months, the salt wedge intrudes almost to the site of impassable falls in these systems: St. Croix River (rkm 16), Machias River (rkm 10), and the Saco River (rkm 10). Although surveys have not been conducted to document Atlantic sturgeon presence, subadults may use the estuaries of these smaller coastal drainages during the summer months (ASSRT 2007).

St. Croix River – Maine/Nova Scotia

The historic and current status of a St. Croix Atlantic sturgeon population is largely unknown. Mike Dadswell (Arcadia University, Canada) notes from personal communications with Nova Scotia Power (in 1993) that a small population of large sturgeon may be spawning annually below the hydropower dam on the St. Croix River (Dadswell 2006). Other than this personal communication, there is no additional information that an Atlantic sturgeon population exists on the St. Croix or regarding their status (ASSRT 2007).

Penobscot River – Maine

There have been two surveys conducted in the last 15 years to document the presence of shortnose and Atlantic sturgeon in the Penobscot River. The Maine Department of Marine Resources (MEDMR) conducted a limited sampling effort in 1994 and 1995 to assess whether shortnose sturgeon were present in the Penobscot River. The MEDMR made 55 sets of 90 meter experimental gill nets for a total fishing effort of 409 net hours (1 net hour = 100 yards fished for 1 hour). The majority of the fishing effort in the Penobscot River was in the upper estuary near head-of-tide. No shortnose or Atlantic sturgeon were captured. The sampling was determined to be inadequate to assess the presence of adult Atlantic sturgeon because the mesh sizes would have been selective only for subadult Atlantic sturgeon that are commonly found in the lower estuary of larger river systems. In 2006, a similar gill net survey was implemented in the lower river using both 15 cm and 30 cm stretched mesh sinking gill nets. As of January 2007, sixty-two shortnose and seven Atlantic sturgeon have been captured in 1004.39 net hours, 506.18 net hours using the smaller mesh and 498.21 net hours using the larger mesh (M.

Kinnison, University of Maine, personal communication). One of these Atlantic sturgeon, captured in July, may have been an adult based on its size (145 cm TL) and time of capture. Thus, it is probable that a small population of Atlantic sturgeon persists in the Penobscot River. This speculation is supported by archeological evidence that sturgeon were present, occasional observations by fishers, and at least one capture of an adult Atlantic sturgeon by a recreational fisherman (Bangor Daily News 2005; ASSRT 2007).

Estuarial Complex of the Kennebec, Androscoggin, and Sheepscot Rivers – Maine

Atlantic sturgeon were historically abundant in the Kennebec River and its tributaries, including the Androscoggin and Sheepscot rivers (Bigelow and Schroeder 1953; Vladykov and Greeley 1963; Kennebec River Resource Management Plan 1993). In 1849, a directed fishery for Atlantic sturgeon landed 160 mt. Population estimates based on the landings indicated that approximately 10,240 adult sturgeon were present prior to 1843 (Kennebec River Resource Management Plan 1993). Three hundred and thirty-six Atlantic sturgeon (nine adults and 327 subadults) have been captured in the Kennebec River in a multi-filament gill net survey conducted intermittently from 1977 through 2000 (Squiers 2004). During this period, the CPUE of Atlantic sturgeon has increased by a factor of 10 to 25% (CPUE from 1977 to 1981 was 0.30 versus CPUE from 1998 to 2000 at 7.43). The mean length of the 327 subadults was 86.7 cm TL with a range from 48 to 114.5 cm TL (a subadult was classified as being 40 to 130 cm TL). The majority of the adult captures were in July between Merrymeeting Bay and Gardiner. Additional insight concerning the timing of Atlantic sturgeon spawning season emerged from a small commercial fishery on the Kennebec River in South Gardiner near Rolling Dam from June 15 – July 26, 1980. Thirty-one adult Atlantic sturgeon (27 males, four of which were ripe, and four females, one of which was ripe) were captured. Two adults tagged in 1978 by the MEDMR in South Gardiner were recaptured in this fishery (ASSRT 2007).

On July 13, 1994, while sampling for sturgeon, the MEDMR captured seven adult Atlantic sturgeon just below the spillway of the Edwards Dam in Augusta. Five of the seven Atlantic sturgeon (56 to 195 cm TL) were males expressing milt. In 1997, a biweekly trawl survey conducted from April through November by Normandeau Associates in the lower Kennebec River, captured 31 subadults and one adult Atlantic sturgeon. Subadults were also captured by the MEDMR in September of 1997 in the Eastern River (n = 18) and the Cathance River (n = 5), which are freshwater tributaries to the Kennebec, in overnight sets of gill nets (T. Squiers, MEDMR, personal communication). Additional sampling from 2000 through 2003 of the MEDMR inshore groundfish trawl survey collected 13 subadults at the mouth of the Kennebec River, which had the greatest occurrences of Atlantic sturgeon among five regions sampled along the New Hampshire and Maine coasts (Squiers 2003). The most recent capture of an adult Atlantic sturgeon occurred in June of 2005, where a 178 cm TL sturgeon was captured in an American shad gill net (12.7 cm stretched mesh) in Ticonic Bay, just upstream of the confluence between Sebasticook and the Kennebec rivers (Squiers 2005; ASSRT 2007).

The presence of adult male Atlantic sturgeon in ripe condition near the head-of-tide during June and July of 1994, 1997, and possibly in 2005 presents strong evidence that a spawning population still exists in the Kennebec River. While no eggs, larvae, or YOY have been captured in the last 15 years, the presence of subadults (48 cm to over 100 cm TL) in tidal freshwater tributaries and the mid-estuary and mouth of the Kennebec River from at least April – November provides additional evidence that a spawning population of Atlantic sturgeon persists in the Kennebec River estuary (ASSRT 2007).

The only documented occurrence of Atlantic sturgeon in the Androscoggin River was an adult captured and released approximately one km downstream of the Brunswick Dam in 1975. No studies have been conducted to assess whether Atlantic sturgeon are presently utilizing the Androscoggin River for spawning. Subadults have been captured in the Sheepscot River, which may function as a nursery area for Kennebec River Atlantic sturgeon (ASSRT 2007).

Piscataqua River/Great Bay Estuary System – New Hampshire

Few Atlantic sturgeon have been captured in the Piscataqua River (Hoff 1980). A subadult Atlantic sturgeon (57 cm; likely age-1) was captured by New Hampshire Fish and Game (NHFG) in June 1981 at the mouth of the Oyster River in Great Bay (NHFG 1981). Between July 1, 1987, and June 30, 1989, NHFG surveyed the deeper tributaries of the Great Bay Estuary, including the Piscataqua, Oyster, Little and Lamprey rivers, as well as the Great Bay for shortnose sturgeon, using 30.5 m nets (3 m deep, with 14 and 19 cm stretch mesh) that were fished for 146 net days. In 1988, sampling occurred in suspected spawning areas (salinities 0 to 10 ppt) in the spring and in suspected feeding areas (salinities around 24 ppt) in the summer. In 1989, nets were fished in May and June only (salinities 6 to 15 ppt). No Atlantic sturgeon were captured. However, a large gravid female Atlantic sturgeon (228 cm TL) weighing 98 kg (of which 15.9 kg were eggs) was captured by a commercial fisherman in a small mesh gill net at the head-of-tide in the Salmon Falls River in South Berwick, Maine, on June 18, 1990 (D. Grout, NHFG, personal communication). The Salmon Falls River is a shallow tributary of the Piscataqua and is the delineation between New Hampshire and Maine state lines. Since 1990, the NHFG has not observed or received reports of Atlantic sturgeon of any age-class being captured in the Great Bay Estuary and its tributaries (B. Smith, NHFG, personal communication). It is the conclusion of the Atlantic Sturgeon Status Review Team and NHFG biologists that the Great Bay Atlantic sturgeon population is likely extirpated (ASSRT 2007).

Merrimack River – New Hampshire and Massachusetts

Historical reports of Atlantic sturgeon in the Merrimack River include a 104 kg sturgeon taken at Newburyport on September 14, 1938, while netting for blueback herring (Hoover 1938). An intensive gill net survey was conducted in the Merrimack River from 1987 through 1990 to determine annual movements, spawning, summering, and wintering areas of shortnose and Atlantic sturgeon (Kieffer and Kynard 1993).

Thirty-six Atlantic sturgeon were captured (70 to 156 cm TL); most being under 100 cm TL. One dead Atlantic sturgeon was found on June 30, 1990 at the shortnose spawning area in Haverhill, Massachusetts (between rkm 31 and 32). Of 23 subadult Atlantic sturgeon sonically tracked in the river, 11 left the river within seven days, and the rest left by September or October of each year (Kieffer and Kynard 1993). Fish captured in one year were not observed in the river during subsequent years. On June 9, 1998, a 24 inch (estimated length) Atlantic sturgeon was captured and released in the Merrimack River by the United States Fish and Wildlife Service (USFWS) personnel who were conducting a contaminant study on the river (D. Major, USFWS, personal communication). This information provides no evidence of a spawning population of Atlantic sturgeon in the Merrimack River, although it seems that the estuary is used as a nursery area (B. Kynard, USGS Conte Anadromous Fish Research Center, personal communication; ASSRT 2007).

Taunton River – Massachusetts and Rhode Island

Historical records indicate that Atlantic sturgeon spawned in the Taunton River at least until the turn of the century (Tracy 1905). A gill net survey was conducted in the Taunton River during 1991 and 1992 to document the use of this system by sturgeon. Three subadult Atlantic sturgeon were captured but were determined to be non-natal fish (Burkett and Kynard 1993). In June 2004, a fisherman fishing in state waters noted that the first three fathoms of towed up gear held three juvenile Atlantic or shortnose sturgeon (Anoushian 2004). Trawlers fishing in state waters (less than three miles offshore) also occasionally report Atlantic sturgeon captures. Since 1997, only two sturgeon have been captured by the Rhode Island Department of Environmental Management Trawl Survey (RIDEM), one measuring 85 cm TL was captured in 1997 in Narragansett Bay, and another (130 cm TL) was captured in October 2005 in Rhode Island Sound (A. Libby, RIDEM, personal communication). The NMFS observer program has also documented Atlantic sturgeon bycatch off the coast of Rhode Island in Federal waters. Since spawning adults were not found during the expected spawning period of May and June, it is likely that a spawning population of Atlantic sturgeon does not occur in the Taunton River, though the system is used as a nursery area for Atlantic sturgeon (Burkett and Kynard 1993; ASSRT 2007).

Thames River – Connecticut

The Thames River is formed by the joining of the Yantic and Shetucket rivers in Norwich Harbor, Connecticut. Information on abundance of Atlantic sturgeon in the Thames River is scarce. Sturgeon scutes have been documented at an archeological site along the river, and historical reports note sturgeon use by Native Americans. Atlantic sturgeon were reportedly abundant in the system until the 1830s (reviewed in Minta 1992). Whitworth (1996) speculated that populations of both shortnose and Atlantic sturgeon in the Thames were always low because the fall line is located near the limit of saltwater intrusion, leaving little to no freshwater habitat for spawning. The construction of the Greenville Dam in 1825 further restricted available habitat and probably prevented sturgeon from spawning in the river. There have been some reports of low dissolved

oxygen (DO) levels during the summer months. The mouth of the river is dredged to accommodate the shipyard, and the channel was recently improved to provide deeper depths to accommodate the Sea Wolf submarine. Subadult Atlantic sturgeon have been captured in the estuary (Whitworth 1996), but it is unlikely that a spawning population is present (ASSRT 2007).

Connecticut River – Massachusetts and Connecticut

Judd (1905) reports that sturgeon were speared at South Hadley Falls in the mid 1700s. There are historical reports of sturgeon migration as far as Hadley, Massachusetts, but regular migration of Atlantic sturgeon beyond Enfield, Connecticut, is doubtful due to presence of significant rapids (Judd 1905). A dam constructed at Enfield in 1827 effectively blocked any migration beyond this point, until 1977 when the dam was breached. Until recently, there has been no evidence that Atlantic sturgeon currently use the Massachusetts portion of the Connecticut River. On August 31, 2006, a 152.4 cm TL Atlantic sturgeon was observed in the Holyoke Dam spillway lift (around rkm 143). The Atlantic sturgeon was not sexed and was described as a subadult (R. Murray, Holyoke Gas and Electric, personal communication). However, based on the size of the Atlantic sturgeon it is possible that the fish was a mature adult. This is the first time an Atlantic sturgeon has been reported at the Holyoke Dam fish lift (ASSRT 2007).

Six juvenile fish (9 to 11 kg) were reportedly taken opposite Haddam Meadows in 1959, but it is unclear if these were Atlantic or shortnose sturgeon. As late as the 1980s, the Connecticut Department of Environmental Protection (CTDEP) fisheries staff reported occasional visual observations of Atlantic sturgeon below the Enfield Dam during May and June. From 1984 to 2000, the CTDEP studied the abundance, locations, and seasonal movement patterns of shortnose sturgeon in the lower Connecticut River and Long Island Sound (Savoy and Pacileo 2003). Sampling was conducted using gill nets ranging from 10 to 18 cm stretched mesh in the lower Connecticut River (1988 to 2005) and a stratified random-block designed trawl survey (12.8 m from 1984 to 1990, and 15.2 m from 1990 to 2005) in the Long Island Sound (also referred to as the LIS Trawl Survey). One hundred and thirty-one Atlantic sturgeon were collected from the lower Connecticut River gill net survey, and average lengths of fish reported from 1988 to 2000 were 77 cm FL (51 to 107 cm FL). The majority of these subadult Atlantic sturgeon were captured in the lower river (between rkm 10 and 26) within the summer range of the salt wedge (Savoy and Shake 1993). A total of 347 fish were collected in the LIS Trawl Survey from 1984 through 2004, of these with reported lengths (1984 to 2000) the mean length was 105 cm FL (ranging from 63 to 191 cm FL). Data from 1984 through 2000, indicated that 68% of the Atlantic sturgeon captured in the trawl survey came from the Central Basin (off Faulkner Island), while 6% of catches occurred in northern portions of the LIS survey near the mouth of the Connecticut River (ASSRT 2007).

While research efforts have not specifically investigated the occurrence of Atlantic sturgeon in the upper Connecticut River, the species has never been collected incidentally in this region during extensive sampling for shortnose sturgeon. Occasional reports, sightings, and capture of large Atlantic sturgeon (150 to 300 cm) are made, but

most Atlantic sturgeon captured within tidal waters or freshwater in Connecticut are consistent with the size and seasonal locations of immature Atlantic sturgeon from the Hudson River (Savoy 1996). Based on the lack of evidence of spawning adults, stocks of Atlantic sturgeon native to Connecticut waters are believed to be extirpated (Savoy 1996; ASSRT 2007).

<u>Housatonic River – Connecticut</u>

Coffin (1947) reports that Atlantic sturgeon were abundant in the Housatonic River and were captured by Native Americans. According to Whitworth (1996), there was a large fishing industry for sturgeon in this basin, and subadults have been captured in the estuary. Atlantic sturgeon likely spawned at a natural fall (Great Falls) at rkm 123 until 1870 when the Derby Dam was constructed at rkm 23.5. The Derby Dam restricted access to approximately 100 km, or 81%, of historical habitat. The Housatonic has not been systematically sampled for sturgeon in recent years (last 15 years), but it is unlikely that a spawning population is present (USFWS-NMFS 1998; ASSRT 2007).

<u>Hudson River – New York</u>

Atlantic sturgeon in the Hudson River have supported subsistence and commercial fishing since colonial times (Kahnle et al. 1998). No data on abundance of juveniles are available prior to the 1970s; however, catch depletion analysis estimated conservatively that 6,000 to 6,800 females contributed to the spawning stock during the late 1800s (Secor 2002, Kahnle et al. 2005). Two estimates of immature Atlantic sturgeon have been calculated for the Hudson River stock, one for the 1976 year class and one for the 1994 year class. Dovel and Berggren (1983) marked immature fish from 1976 to 1978. Estimates for the 1976 year class at age one ranged from 14,500 to 36,000 individuals (mean of 25,000). In October of 1994, the New York State Department of Environmental Conservation (NYSDEC) stocked 4,929 marked age-0 Atlantic sturgeon, provided by a USFWS hatchery, into the Hudson Estuary at Newburgh Bay. These fish were reared from Hudson River brood stock. In 1995, Cornell University sampling crews collected 15 stocked and 14 wild age-1 Atlantic sturgeon (Peterson et al. 2000). A Petersen mark-recapture population estimate from these data suggests that there were 9,529 (95% CI = 1,916 to 10,473) age-0 Atlantic sturgeon in the estuary in 1994. Since 4,929 were stocked, 4,600 fish were of wild origin, assuming equal survival for both hatchery and wild fish and that stocking mortality for hatchery fish was zero. Estimates of spawning adults were also calculated by dividing the mean annual harvest from 1985 to 1995 by the exploitation rate (u). The mean annual spawning stock size (spawning adults) was 870 (600 males and 270 females) (Kahnle et al. *In press*; ASSRT 2007).

Current abundance trends for Atlantic sturgeon in the Hudson River are available from a number of surveys. From July to November during 1982 to 1990 and 1993, the NYSDEC sampled the abundance of juvenile fish in Haverstraw Bay and the Tappan Zee Bay. The CPUE of immature Atlantic sturgeon was 0.269 in 1982 and declined to zero by 1990. The American shad gill net fishery in the Hudson River estuary, conducted from early April to late May, incidentally captures young Atlantic sturgeon (< 100 cm)

and therefore, has been monitored by onboard observers since 1980. Annual CPUE data from the observer program were summarized as total observed catch/total observed effort. Catch-per-unit-of-effort of Atlantic sturgeon as bycatch was greatest in the early 1980s and decreased until the mid 1990s. It has gradually begun to increase since that time (ASSRT 2007).

Hudson River Valley utilities (Central Hudson Electric and Gas Corp., Consolidated Edison Company of New York, Inc., New York Power Authority, Niagara Mohawk Power Corporation, Orange and Rockland Utilities, Inc.) conduct extensive river-wide fishery surveys to obtain data for estimating impacts of power plant operations. Detailed survey descriptions are provided in the utilities' annual reports (CONED 1997). Two surveys regularly catch sturgeon, despite the fact that these surveys were not specifically designed to capture sturgeon. The Long River Survey (LRS) samples ichthyoplankton river-wide from the George Washington Bridge (rkm 19) to Troy (rkm 246) using a stratified random design (CONED 1997). These data, which are collected from May through July, provide an annual index of juvenile Atlantic sturgeon in the Hudson River estuary since 1974. The Fall Shoals Survey (FSS), conducted from July through October by the utilities, calculates an annual index of the number of fish captured per haul. Between 1974 and 1984, the shoals in the entire river (rkm 19 to 246) were sampled by epibenthic sled; in 1985 the gear was changed to a three-meter beam trawl. Length data are only available for the beam trawl survey from 1989 to the present; fish length ranged from 10 to 100 cm TL, with most fish less than 70 cm TL. Based on these length data, it seems that ages-0 (YOY), 1, and 2 sturgeon are present in the river. Indices from utility surveys conducted from 1974 to the present (LRS and FSS) indicate a trend consistent with NYSDEC American shad monitoring data. Abundance of young juvenile Atlantic sturgeon has been declining, with CPUE peaking at 12.29 in 1986 (peak in this survey) and declining to 0.47 in 1990. Since 1990, the CPUE has ranged from 0.47 to 3.17, increasing in recent years to 3.85 (2003). In 2000, the NYSDEC created a sturgeon juvenile survey program to supplement the utilities' survey; however, funds were cut in 2000, and the USFWS was contracted in 2003 to continue the program. In 2003 to 2005, 579 juveniles were collected (N = 122, 208, and 289, respectively) (Sweka et al. 2006). Pectoral spine analysis showed they ranged from one to eight years of age, with the majority being ages two and six. None of the captures were found to be young-of-the-year (YOY; smaller than 41 cm TL) (ASSRT 2007).

Indices for post-migrant Atlantic sturgeon are provided by the New Jersey Bureau of Marine Fisheries from surveys of the coastal waters along the entire state (Sandy Hook to Delaware Bay). Since 1988 when the survey was initiated, a total of 96 Atlantic sturgeon have been captured. Abundances of post-migrants seem to be declining as CPUE has decreased from a high of 8.75 in 1989 to 1.5 in 2003. This trend differs from Hudson River Fall Shoals Utility Survey, which indicated an increasing or stable trend over the last several years (ASSRT 2007).

All available data on abundance of juvenile Atlantic sturgeon in the Hudson River estuary (i.e., mark/recapture studies, bycatch data from commercial gill net fishery, and utilities sampling) indicate a substantial drop in production of young since the mid-1970s. The greatest decline seemed to occur in the middle to late 1970s, followed by a secondary

drop in the late 1980s. Sturgeon are still present, and juveniles (age-0 (YOY), 1, and 2 years) were captured in recent years and a slight increasing trend in CPUE has been observed. The capture of YOY sturgeon in 1991, 1993 to 1996, and 2003, provides evidence of successful spawning (ASSRT 2007).

Delaware River – New Jersey, Delaware, and Pennsylvania

The Delaware River, flowing through New Jersey, Delaware, Pennsylvania and into Delaware Bay, historically may have supported the largest stock of Atlantic sturgeon of any Atlantic coastal river system (Kahnle et al. 1998; Secor and Waldman 1999; Secor 2002). Prior to 1890, it is expected that more than 180,000 adult females were spawning in the Delaware River (Secor and Waldman 1999, Secor 2002). Juveniles were once abundant enough to be considered a nuisance bycatch of the American shad fishery. Very little is known about adult stock size and spawning of Atlantic sturgeon in the Delaware river; however, based on reported catches in gill nets and by harpoons during the 1830s, they may have spawned as far north as Bordentown, south of Trenton, New Jersey (Pennsylvania Commission of Fisheries 1897). A recent sonic tracking project, on-going in 2006, has reported at least one adult Atlantic sturgeon migrating to Bordentown during the spawning season (D. Fox, Delaware State University, personal communication). Borodin (1925) reported that running-ripe sturgeon were captured near Delaware City, Delaware adjacent to Pea Patch Island. Spawning grounds with appropriate substrate occurred near Chester, Pennsylvania. Ryder (1888) suggested that juvenile Atlantic sturgeon used the tidal freshwater reach of the estuary as a nursery area. Lazzari et al. (1986) reported that the Roebling-Trenton stretch of the river may be an important nursery area for the species (ASSRT 2007).

The current abundance of all Atlantic sturgeon life stages in the Delaware River has been greatly reduced from the historical level. Brundage and Meadows (1982) recorded 130 Atlantic sturgeon captures between the years of 1958 through 1980. The Delaware Division of Fish and Wildlife (DNREC) began sampling Delaware Bay in 1966 by bottom trawl and have rarely captured Atlantic sturgeon. During the period from 1990 to 2004, the trawl survey captured 17 Atlantic sturgeon (Murphy 2005). However, there are several areas within the estuary where juvenile sturgeon regularly occur. Lazzari et al. (1986) frequently captured juvenile Atlantic sturgeon from May to December in the upper tidal portion of the river below Trenton, New Jersey (N = 89, 1981 to 1984). In addition, directed gill net surveys by DNREC from 1991 through 1998 consistently took juvenile (N > 1,700 overall) Atlantic sturgeon in the lower Delaware River near Artificial Island and Cherry Island Flats from late spring to early fall (Shirey et al. 1999). The number of fish captured in the lower river annually has declined dramatically throughout this time period from 565 individuals in 1991 to 14 in 1998. Population estimates based on mark and recapture of juvenile Atlantic sturgeon declined from a high of 5,600 in 1991 to less than 1,000 in 1995; however, it is important to note that population estimates violated most tagging study assumptions and should not be used as unequivocal evidence that the population has declined dramatically. No population estimates are available from 1996 and 1997, given the low number of recaptures. Voluntary logbook reporting of Atlantic sturgeon bycatch in the spring gill net fishery indicate that abundance varies year to year with no indication of decline or increase mainly because the number of bycatch

reports varies considerably by commercial fishers reporting. Bycatch data are represented as the average bycatch per fisher per year (total bycatch/number of fishers). An annual small mesh gill net survey began in 1991 until 1998 when sampling was restricted to every three years in the lower Delaware River. The results of this study indicated that CPUE (fish per gill net hour) estimates have declined from 32 fish per effort hour in 1991 to only 2 fish per effort hour in 2004 (ASSRT 2007).

Carcasses of large adult fish (> 150 cm TL) are commonly reported along the lower Delaware River and upper Delaware Bay during the historic spawning season (G. Murphy, Delaware Division of Fish and Wildlife, personal communication). Fifteen adult size fish have been documented since 1994, including several gravid females and males. A 2.4 m female Atlantic sturgeon was found dead on June 14, 1994, adjacent to Port Penn; ageing of a pectoral spine indicated it was approximately 25 years old (D. Secor, University of Maryland, personal communication). Three years later, a second female sturgeon was found in late spring/early summer of 1997 adjacent to Port Penn, just south of the eastern end of the Chesapeake/Delaware Canal. A male sturgeon carcass was found on May 19, 1997, just north of the mouth of the Cohansey River, on Beechwood Beach; it seemed that the fish was cut in half by the propeller of a large vessel. Gonadal tissue and a pectoral spine were collected and sent to USFWS-Northeast Fisheries Center (NEFC), Fish Technology Section, Lamar, Pennsylvania, for analysis, where it was confirmed to be a male (W. Andrews, New Jersey Division of Fish, Game, and Wildlife, personal communication). In 2005, DNREC began tracking reported sturgeon mortalities during the spawning season. During the first year, six adults were found dead washed ashore in May 2005, including two from Woodland Beach (approximately 250 cm and 170 cm TL), one from Artificial Island (larger than 180 cm TL), one from South Bowers Beach (205 cm TL), one from Conch Bar (160 cm TL), and one from Slaughter Beach (160 cm TL). Six additional carcasses, presumed adults, were found during April through May 2006, including a gravid female at Augustine Beach (144 cm), a gravid male at Sleusch Ditch (180 cm), one at South Bowers Beach (119 cm), one at Brockonbridge Gut (112 cm), one at Kitts Hummock (208 cm), and one at Little Tinicum Island, Pennsylvania (106 cm). The majority of adults documented had substantial external injuries and were severed (ASSRT 2007).

In addition to the carcasses reported annually during the spawning season, several males were captured by directed gill net efforts and a reward program conducted by Delaware State University during April and May 2006. These males were collected in the lower Delaware River and upper Delaware Bay and were implanted with sonic transmitters to assist in determining spawning locations in the Delaware River (D. Fox, Delaware State University, personal communication). Although catch rates declined throughout the mid 1990s, the mature adults documented within the Delaware System provide evidence that a reproducing population exists. It is speculated, however, that the abundance of subadults within the Delaware River during the 1980s and early 1990s was the result of a mixture of stocks including the Hudson River stock. However, genetic data indicate that the Delaware River has a distinct genetic signature of a remnant population (Waldman et al. 1996a; Wirgin 2006; King supplemental data 2006; ASSRT 2007).

<u>Chesapeake Bay and Tributaries (Potomac, Rappahannock, York, James, Susquehanna,
Nanticoke) – Pennsylvania, Maryland, and Virginia</u>

Historically, Atlantic sturgeon were common throughout the Chesapeake Bay and
its tributaries (Kahnle et al. 1998; Wharton 1957; Bushnoe et al. 2005). There are several
newspaper accounts of large sturgeon in the lower reaches of the Susquehanna River
from 1765 to 1895, indicating that at one time, Atlantic sturgeon may have spawned
there. Commercial landings data during the 1880s are available for the Rappahannock (8
mt), York (23 mt), and James (49 mt) providing evidence that Atlantic sturgeon were
historically present in these rivers as well (Bushnoe et al. 2005). Historical harvests were
also reported in the Patuxent, Potomac, Choptank, Nanticoke, and Wicomico/Pocomoke
rivers (S. Minkkinen, USFWS, personal communication). Prior to 1890, when a sturgeon
fishery began, Secor (2002), using U.S. Fish Commission landings, estimated
approximately 20,000 adult females inhabited the Chesapeake Bay and its tributaries
(ASSRT 2007).

For the past several decades, state fishery agencies and research facilities
operating in the Chesapeake Bay have conducted extensive finfish sampling surveys in
the mainstem Bay and all major tributaries. These surveys occurred in all seasons and
were conducted using many gear types, including trawls, seines, and gill nets. While no
surveys were directed at sturgeon, incidental captures were recorded. These data
supplement reports of sturgeon captures from commercial fishers using gill nets, pound
nets, and fyke nets with occasional visual observations of large sturgeon, including
carcasses found on beaches during the summer (ASSRT 2007).

A mixed stock analysis, performed from nDNA microsatellite markers, indicated
that the Chesapeake Bay population was comprised of three main stocks: 1) Hudson
River (23 to 30%), 2) Chesapeake Bay (0 to 35%), and 3) Delaware River (17 to 27%)
(King et al. 2001). The contribution of fish with Chesapeake Bay origin fish, which had
not been identified in previous genetic studies, indicates the likely existence of a
reproducing population within the Bay. This is further supported and substantiated by the
capture of young juveniles at the mouth of the James River and two YOY Atlantic
sturgeon captured in the river in 2002 and 2004 (Florida Museum of Natural History
2004; A. Spells, USFWS, personal communication; ASSRT 2007).

Several sturgeon sightings were made by commercial fishers and researchers
between 1978 and 1987 near the Susquehanna River mouth. A deep hole (19 m) on the
Susquehanna River near Perryville, Maryland also supported a limited sturgeon fishery
(R. St. Pierre, USFWS retired, personal communication). Maryland Department of
Natural Resources (DNR) personnel reported a large mature female Atlantic sturgeon in
the Potomac in 1970 and another in the Nanticoke River in 1972 (H. Speir, Maryland
DNR, personal communication; ASSRT 2007).

A Virginia Institute of Marine Science (VIMS) trawl survey was initiated in 1955
to investigate finfish dynamics within the Chesapeake Bay; the survey was standardized
in 1979. Since 1955, 40 Atlantic sturgeon have been captured, 16 of which were
captured since 1990, and two of these collections may have been YOY based on size. No
fish were captured between 1990 and 1996; however, seven were captured in 1998. In
subsequent years, catch declined ranging between zero and three fish per year. Similarly,

American shad monitoring programs (independent stake gill net survey) also recorded a spike in Atlantic sturgeon bycatch that peaked in 1998 (N = 34; 27 from James River) and declined dramatically in later years to only one to three sturgeon being captured in each year from 2002 to 2004. These observations could be biased by stocking 3,200 juveniles in the Nanticoke River in 1996; however, the capture of wild fish in the Maryland Reward Tagging program conducted from 1996 to present shows identical rates of capture for wild fish (ASSRT 2007).

The Maryland reward tagging has resulted in the capture of 1,700 Atlantic sturgeon. Five hundred and sixty seven of these fish were hatchery fish, of which 462 were first time captures (14% recapture rate), the remaining captures (1,133) were wild. However, none of these 1,700 Atlantic sturgeon were considered YOY based on length data (S. Minkkinen, USFWS, personal communication). Similarly, Virginia initiated a reward tagging program in 1996 through 1998. The majority of their recaptures were wild Atlantic sturgeon taken from the lower James and York rivers in the 20 to 40 cm size range and are believed to be YOY (A. Spells, USFWS, personal communication). Captures of YOY and age-1 sturgeon in the James River during 1996 and 1997 suggest spawning has occurred in that system. Since then, captures from the reward program have varied, declining from 1999 to 2002 and then increasing in 2005 to levels similar to that of 1998 and with record levels during 2006. Further evidence that spawning may have occurred recently is provided by three carcasses of large adults found in the James River in 2000 to 2003, the discovery of a 213 cm carcass of an adult found in the Appomattox River in 2005, as well as the release of a 2.4 m Atlantic sturgeon near Hoopers Island (Chesapeake Bay) in April, 1998 (S. Minkkinen, USFWS, personal communication; ASSRT 2007).

These data indicate that some of the Chesapeake Bay tributaries may continue to support spawning populations as evidenced by YOY captures (James River) and carcasses of mature adults being found occasionally within the Bay during the spawning season. Commercial fishers have regularly reported observations of YOY or age-1 juveniles in the York River over the past few years (K. Place, commercial fisherman, personal communication). In 2006, tissue samples from 38 juvenile Atlantic sturgeon measuring between 500 to 600 mm TL (around age-1) were haplotyped and genotyped by researchers (I. Wirgin – NIEM, and T. King – USGS, supplemental data 2006). These 38 juveniles from the York River were significantly different (P < 0.01) from neighboring subpopulations including the James River subpopulation, based on frequency differences in mtDNA and nDNA markers. However, the York River does not contain unique mtDNA haplotypes differentiating it directly from other sturgeon populations, and the population could not be differentiated from the James River population using classification techniques. Additionally, a review of spawning habitat availability in the Chesapeake Bay and its tributaries indicated that spawning habitat is available in the James, York, and Appomattox rivers (Bushnoe et al. 2005). Therefore, the above information provides some evidence that a spawning population may exist in the York River, as the population exhibits significantly different haplotype frequencies from its neighboring subpopulations, and spawning habitat appears to be available. However, there is a possibility that samples taken from the York River were of a mixed stock since they measured 500 to 600 mm in total length (the size range of migratory subadults) and many of the collections were taken from the mouth of the river (ASSRT 2007).

North Carolina Rivers

Historically, Atlantic sturgeon were abundant in most North Carolina coastal rivers and estuaries; the largest fishery occurring in the Roanoke River/Albemarle Sound system and in the Cape Fear River (Kahnle et al. 1998). Historic landing records from the late 1800s indicated that Atlantic sturgeon were very abundant within the Albemarle Sound (around 61.5 mt/yr); however, these landings are relatively small compared to the Delaware fishery (around 2,700 mt/yr) (Secor 2002). Abundance estimates derived from these historical landings records indicated that between 7,200 and 10,500 adult females were present within North Carolina prior to 1890 (Armstrong and Hightower 2002; Secor 2002).

Albemarle Sound (Roanoke and Chowan/Nottoway Rivers) – North Carolina

Historic and current survey data indicate that spawning occurs in the Roanoke River/Albemarle Sound system, where both adults and small juveniles have been captured. Since 1990, the NC Division of Marine Fisheries (NCDMF) has conducted the Albemarle Sound Independent Gill Net Survey (IGNS), initially designed to target striped bass. The survey is conducted from November to May, using a randomized block sampling design and employing 439 m of gill net, both sinking and floating, with stretched mesh sizes ranges from 63.5 mm (2.5 in) to 254 mm (10 in). Since 1990, 842 sturgeon have been captured ranging from 15.3 to 100 cm FL, averaging 47.2 cm FL. One hundred and thirty-three (16%) of the 842 sturgeon captured could be classified as YOY (≤ 41 cm TL, ≤ 35 cm FL); the others were subadults. Incidental take of Atlantic sturgeon in the IGNS indicate that the subpopulation has been increasing in recent years (1990 to 2000), but since then recruitment has dramatically declined. Similarly, the

North Carolina Division of Marine Fisheries (NCDMF) Observer Program documented the capture of 30 Atlantic sturgeon in large and small mesh gill nets; two of these individuals being YOY (less than 410 mm TL) (Blake Price, NCDMF, personal communication; ASSRT 2007).

In 1997 and 1998, North Carolina State University (NCSU) researchers characterized the habitat use, growth, and movement of juvenile Atlantic sturgeon (Armstrong and Hightower 2002). Their survey collected 107 Atlantic sturgeon, of which 15 (14%) could be considered YOY (≤ 41 cm TL or 35 cm FL). Young juveniles were observed more often over organic rich mud bottoms and at depths of 3.6 to 5.4 meters. Adult running ripe sturgeon have not been collected in the Roanoke River even though the North Carolina Wildlife Resources Commission (NCWRC) has sampled the spawning grounds since the 1990s during their annual striped bass electrofishing survey. However, in 2005, an angler captured a YOY (39 cm TL) Atlantic sturgeon in the Roanoke River, near the city of Jamesville, North Carolina. These multiple observations of YOY from the Albemarle Sound and Roanoke River provide evidence that spawning continues, and catch records indicate that this population seemed to be increasing until 2000, when recruitment began to decline (ASSRT 2007).

Pamlico Sound (Tar and Neuse Rivers) – North Carolina

Evidence of spawning was reported by Hoff (1980), who noted captures of very young juveniles in the Tar and Neuse rivers. More recently, two juveniles (approximately 45 and 60 cm TL) were observed dead on the bank of Banjo Creek, a tributary to the Pamlico system (B. Brun, USFWS and U.S. Army Corps of Engineers (retired), personal communication). An independent gill net survey, following the Albemarle Sound IGNS methodology, was initiated in 2001. Collections were low during the periods of 2001 to 2003, ranging from zero to one fish per year. However, in 2004, this survey collected 14 Atlantic sturgeon ranging from 460 to 802 mm FL, and averaging 575 mm FL. During the same time period (2002 to 2003), four Atlantic sturgeon (561 – 992 mm FL) were captured by NCSU personnel sampling in the Neuse River (Oakley 2003). Similarly, the NCDMF Observer Program documented the capture of 12 Atlantic sturgeon in the Pamlico Sound from April 2004 to December 2005; none of these were YOY or spawning adults, averaging approximately 600 mm TL (Blake Price, NCDMF, personal communication).

The incidental capture of two juvenile Atlantic sturgeon in the Tar (1) and Neuse rivers (1) in 2005 also provides evidence that spawning may be occurring within those rivers. The Tar River juvenile was captured near Greenville, North Carolina, by an angler and reported to be less than 40 cm TL (P. Kornegay, NCWRC, personal communication). The other juvenile was captured in an illegal gill net set upstream of New Bern, North Carolina, and measured 46 cm TL. Although not confirmed as YOY, these two captures are important in that they represent the only evidence of possible spawning activities within the Pamlico Sound Drainage for at least the last 15 years (ASSRT 2007).

Cape Fear River – North Carolina

A gill net survey for adult shortnose and juvenile Atlantic sturgeon was conducted in the Cape Fear River drainage from 1990 to 1992, and replicated 1997 to 2005. Each sampling period included two overnight sets (checked every 24 hours). The 1990 to 1992 survey captured 100 Atlantic sturgeon below Lock and Dam #1 (rkm 95) for a CPUE of 0.11 fish/net-day. No sturgeon were collected during intensive sampling above Lock and Dam #1. In 1997, 16 Atlantic sturgeon were captured below Lock and Dam #1, an additional 60 Atlantic sturgeon were caught in the Brunswick (a tributary of the Cape Fear River), and 12 were caught in the Northeast Cape River (Moser et al. 1998). Relative abundance of Atlantic sturgeon below Lock and Dam #1 seemed to have increased dramatically since the survey was conducted in 1990 to 1992 (Moser et al. 1998) as the CPUE of Atlantic sturgeon was two to eight times greater during 1997 than in the earlier survey. Since 1997, Atlantic sturgeon CPUE has been gradually increasing; a regression analysis revealed that CPUE doubled between the years of 1997 (around 0.25 CPUE) and 2003 (0.50 CPUE) (Williams and Lankford 2003). This increase may reflect the effects of North Carolina's ban on Atlantic sturgeon fishing that began in 1991; however, the increase in CPUE may also be artificial as these estimates are similar among years except in 2002 (large increase) that likely skewed the regression analysis.

In 2003, the NCDMF continued the sampling program (Cape Fear River Survey) and have collected 91 Atlantic sturgeon (427 to 1473 mm FL) (ASSRT 2007).

Adult Atlantic sturgeon have been observed migrating upstream in the fall within the Cape Fear River, indicating that there may be two spawning seasons or some upstream overwintering may be occurring (M. Williams, formerly of University of North Carolina at Wilmongton, personal communcation). One large Atlantic sturgeon was tracked moving upstream in the Black River, which is a tributary of the Cape Fear River, in early October. Moreover, all of the largest sturgeon collected by University of North Carolina at Wilmington personnel were later captured only during September and October in both the Cape Fear and Northeast Cape Fear rivers. Finally, a carcass of an adult female Atlantic sturgeon with fully developed ovaries was discovered in an area well upstream of the saltwater-freshwater interface in mid-September. Studies in other river systems have also demonstrated that some sturgeon will participate in upstream spawning migrations in the fall (Rogers and Weber 1995; Weber and Jennings 1996; Moser et al. 1998; ASSRT 2007).

South Carolina Rivers

Historically, Atlantic sturgeon were likely present in many South Carolina river/estuary systems, but it is not known where spawning occurred. Secor (2002) estimated that 8,000 spawning females were likely present prior to 1890, based on US Fish Commission landing records. Since the 1800s, however, populations have declined dramatically (Collins and Smith 1997). During the last two decades, Atlantic sturgeon have been observed in most South Carolina coastal rivers, although it is not known if all rivers support a spawning subpopulation (Collins and Smith 1997; ASSRT 2007).

Winyah Bay (Waccamaw, Great Pee Dee, and Sampit Rivers) – South Carolina

Recent shortnose sturgeon sampling (using 5, 5.5, 7, and 9 inch stretched mesh experimental gill nets; 16' otter trawl) conducted in Winyah Bay captured two sub-adult Atlantic sturgeon during 4.2 hrs of effort in 2004. Captures of age-1 juveniles from the Waccamaw River during the early 1980s suggest that a reproducing population of Atlantic sturgeon may persist in that river, although the fish could have been from the nearby Great Pee Dee River (Collins and Smith 1997). In 2003 and 2004, nine Atlantic sturgeon (48.4 to 112.2 cm FL) were captured in the Waccamaw River during the South Carolina Department of Natural Resources (SCDNR) annual American shad gill net survey, although none were considered spawning adults or YOY. However, Collins et al. (1996) note that unlike northern populations, in South Carolina, YOY are considered to be less than 50 cm TL or 42.5 cm FL, as growth rates are greater in the warmer southern waters compared to cooler northern waters. Therefore, the capture of a 48.4 cm FL sturgeon provides some evidence that YOY may be present in the Waccamaw River and some evidence of a spawning subpopulation. Lastly, watermen on the lower Waccamaw and Pee Dee rivers have observed jumping sturgeon, which suggest that rivers either serve as a nursery/feeding habitat or support an extant subpopulation(s) (W. Laney, USFWS, personal communication; ASSRT 2007).

Until recently, there was no evidence that Atlantic sturgeon spawned in the Great Pee Dee River, although subadults were frequently captured and large adults were often observed by fishers. However, a fishery survey conducted by Progress Energy Carolinas Incorporated captured a running ripe male in October of 2003 and observed other large sturgeon, perhaps revealing a fall spawning run (ASSRT 2007).

There are no data available regarding the presence of YOY or spawning adult Atlantic sturgeon in the Sampit River, although it did historically support a subpopulation and is thought to serve as a nursery ground for local stocks (ASSRT 2007).

Santee and Cooper Rivers – South Carolina

The capture of 151 subadults, including age-1 juveniles, in the Santee River in 1997 suggests that an Atlantic sturgeon population exists in this river (Collins and Smith 1997). This is supported by three adult Atlantic sturgeon carcasses found above the Wilson and Pinopolis dams in Lakes Moultrie (Santee-Cooper reservoirs) during the 1990s (M. Collins, SCDNR, personal communication). Although shortnose sturgeon spawning above the dam has been documented, there is scant information to support existence of a land-locked subpopulation of Atlantic sturgeon. In 2004, 15 subadult Atlantic sturgeon were captured in shortnose sturgeon surveys during 156.6 hours of effort conducted in the Santee estuary. The previous winter, four juvenile (YOY and subadults) Atlantic sturgeon were captured (360 to 657 mm FL) from the Santee (N = 221) and Cooper (N = 3) rivers. These data support previous hypotheses that a fall spawning run occurs within this system, similar to that observed in other southern river systems. However, SCDNR biologists are skeptical as to whether these smaller sturgeon (360 and 378 mm FL) from the Santee-Cooper are resident YOY as flood waters from the Pee Dee or Waccamaw River could have transported these YOY to the Santee-Cooper system via Winyah Bay and the Intercoastal Waterway (ICW) (McCord 2004; ASSRT 2007).

Ashley River – South Carolina

The Ashley River, along with the Cooper River, drains into Charleston Bay; only shortnose sturgeon have been sampled in these rivers. While the Ashley River historically supported an Atlantic sturgeon spawning subpopulation, it is unknown whether the subpopulation still exists (ASSRT 2007).

ACE Basin (Ashepoo, Combahee, and Edisto Rivers) – South Carolina

From 1994 through 2001, over 3,000 juveniles have been collected in the ACE Basin including 1,331 YOY sturgeon (Collins and Smith 1997; M. Collins, SCDNR, personal communication). Sampling for adults began in 1997, with two adult sturgeon captured in the first year of the survey, including one gravid female (234 cm TL) captured in the Edisto River and one running ripe male (193 cm TL) captured in the Combahee River. The running ripe male in the Combahee River was recaptured one week later in the Edisto River, which suggests that the three rivers that make up the ACE

basin may support a single subpopulation that spawns in at least two of the rivers. In 1998, an additional 39 spawning adults were captured (M. Collins, SCDNR, personal communication). These captures show that a current spawning subpopulation exists in the ACE Basin as both YOY and spawning adults are regularly captured (ASSRT 2007).

Broad/Coosawatchie River – South Carolina

There has been little or no scientific sampling for Atlantic sturgeon in the Broad/Coosawatchie River. One fish of unknown size was reported from a small directed fishery during 1981 to 1982 (Smith and Dingley 1984; ASSRT 2007).

Savannah River – South Carolina and Georgia

The Savannah River supports a reproducing subpopulation of Atlantic sturgeon (Collins and Smith 1997). According to the NOAA-National Ocean Service, 70 Atlantic sturgeon have been captured since 1999 (J. Carter, National Ocean Service, supplemental data 2006). Twenty-two of these fish have been YOY (less than 410 mm TL). A running ripe male was captured at the base of the dam at Augusta during the late summer of 1997, which supports the hypothesis that spawning occurs there in the fall (ASSRT 2007).

Georgia Rivers

Prior to the collapse of the fishery in the late 1800s, the sturgeon fishery was the third largest fishery in Georgia. Secor (2002) estimated from U.S. Fish Commission landing reports that approximately 11,000 spawning females were likely present prior to 1890. The sturgeon fishery was mainly centered on the Altamaha River, and in more recent years, peak landings were recorded in 1982 (13,000 lbs). Based on juvenile presence and abundance, the Altamaha seems to currently support one of the healthiest Atlantic sturgeon subpopulations in the southeast (D. Petersen, University of Georgia, personal communication). Atlantic sturgeon are also present in the Ogeechee River; however, the absence of age-1 fish during some years and the unbalanced age structure suggests that the subpopulation is highly stressed (Rogers and Weber 1995). Spawning adults have been collected in recent years from the Satilla River (Waldman et al. 1996b). Recent sampling of the St. Mary's River failed to locate any sturgeon, which suggests that the subpopulation may be extirpated (Rogers et al. 1994). In Georgia, Atlantic sturgeon are believed to spawn in the Savannah, Ogeechee, Altamaha, and Satilla rivers (ASSRT 2007).

Ogeechee River – Georgia

Previous studies have shown the continued persistence of Atlantic sturgeon in this river, as indicated by the capture of age +1 fish. Sampling efforts (including 1991 to 1994, 1997, and 1998) to collect age-1 sturgeon as part of the Savannah River genetics study suggest that juvenile abundance is rare with high inter-annual variability, indicating spawning or recruitment failure. However, the Army's Environmental and Natural

Resources Division (AENRD) at Fort Stewart, Georgia, collected 17 sturgeon in 2003 considered to be YOY (less than 30 cm TL) and an additional 137 fish in 2004, using a 30 m x 2 m experimental gill net (3.8, 7.7, 12.7, 15.2, 17.8 cm stretched mesh). Most of these fish were juveniles; however, nine of these fish measured less than 41 cm TL and were considered YOY. In 2003, 17 sturgeon captured in this survey were also considered YOY (reported as less than 30 cm TL). The AENRD survey provides the most recent captures of YOY in the Ogeechee (ASSRT 2007).

Altamaha River – Georgia

The Altamaha River supports one of the healthiest Atlantic sturgeon subpopulations in the Southeast, with over 2,000 subadults captured in trammel nets, 800 of which were nominally age-1 as indicated by size. Independent monitoring of the American shad fishery also documents the incidental take of Atlantic sturgeon within the river. Using these data, the subpopulation does not seem to be increasing or decreasing, as catch trends are variable (ASSRT 2007).

A survey targeting Atlantic sturgeon was initiated in 2003 by the University of Georgia. Trammel nets (91 m x 3 m) and gill nets were set in the lower 27 rkm of the Altamaha River, and were fished for 20 to 40 minutes during slack tides only. Sampling for adults was conducted using large mesh-gill nets set by local commercial fishermen during the months of April through May 2003. During 2005, similar gill nets were drift set during slack tides to supplement catches. As of October 2005, 1,022 Atlantic sturgeon have been captured using these gear types (trammel and large gill nets). Two hundred and sixty seven of these fish were collected during the spring spawning run in 2004 (N = 74 adults) and 2005 (N = 139 adults). From these captures, 308

(2004) and 378 (2005) adults were estimated to have participated in the spring spawning run, which is 1.5% of Georgia's historical spawning stock (females) that were estimated from U.S. Fish Commission landing records (Schuller and Peterson 2006; Secor 2002; ASSRT 2007).

Satilla River – Georgia

Sampling results indicate that the Atlantic sturgeon subpopulation in the Satilla River is highly stressed (Rogers and Weber 1995). Only four spawning adults or YOY, which were used for genetic analysis (Ong et al. 1996), have been collected from this river since 1995 (ASSRT 2007).

St. Mary's River – Georgia and Florida

The lack of Atlantic sturgeon captures (in either scientific sampling and/or as bycatch in other fisheries) in the St. Mary's River indicates that the river neither supports a spawning subpopulation nor serves as a nursery ground for Atlantic sturgeon (Rogers and Weber 1995; Kahnle et al. 1998). However, no directed sampling surveys have been conducted in recent Years (ASSRT 2007).

St. Johns River – Florida

In the 1970s and 1980s, there were several reports of Atlantic sturgeon being captured by commercial fishermen, although these fish were considered juveniles measuring 69 to 84 cm in length (J. Holder, Florida Fish and Wildlife Commission, personal communication). There have been reports of Atlantic sturgeon tagged in the Edisto River (South Carolina) having been recaptured in the St. Johns River, indicating this river may serve as a nursery ground; however, there are no data to support the existence of a spawning subpopulation (i.e., YOY or running ripe adults) (Rogers and Weber 1995; Kahnle et al. 1998; ASSRT 2007).

Spawning location (ecological)

A study by Collins et al. (2000b) indicated that adult Atlantic sturgeon in South Carolina utilize a wide variety of habitats during the summer. They found sturgeon in the upper fresh/brackish interface zone, the lower interface zone, and in the high salinity portions of the estuary in the Edisto River, South Carolina. Atlantic sturgeon were present in this river from March to October. During the winter, southern Atlantic sturgeon resided in the ocean (Collins et al. 2000b). Adult Atlantic sturgeon in southern rivers exhibit behavior much like gulf sturgeon (*Acipenser oxyrinxhus desotoi*) in that they spend 9 months within the river system and 3 winter months in marine waters (M. Collins, South Carolina Department of Natural Resources, personal communication).

Most studies indicate that after spawning, Atlantic sturgeon migrate to salt water (Vladykov and Greeley 1963); these down-estuary migrations may occur over several months (Bain 1997). In the St. Lawrence River, migrations downstream have been reported from September through November (Scott and Crossman 1973). Hatin et al. (2002) found that the majority of Atlantic sturgeon were gone from the upper St. Lawrence River by late September in some years, while in other years, the sturgeon remained in the upper river through early December. In the Hudson River, females migrate back to salt water immediately following spawning, while males remain until the onset of cold temperatures in the fall (Smith 1985a). Additionally, Bain et al. (2000) reported post-spawn adult sturgeon and older juveniles congregating in deep water habitat during the summer in the Hudson River, New York.

Maturation and spawning periodicity

Atlantic sturgeon mature at different times along the Atlantic coast, with maturity occurring earlier in the Southern regions (Vladykov and Greeley 1963). Females in South Carolina first spawn between the ages of 7 and 19, and males first spawn between 5 and 13 years. In the Hudson River, New York, females first spawn between the ages of 15 and 30 years, and males between 8 and 20 years (Dovel 1979; Smith et al. 1982; Smith 1985a; Smith 1985b; Young et al. 1988; Stevenson and Secor 1999). Scott and Crossman (1973) report that in the St. Lawrence River, Canada, female Atlantic sturgeon mature at approximately 27 years, and males mature between 22 and 34 years. Although most researchers have not verified age determination methods, Stevenson and Secor (2000) used marginal increment analysis and rearing studies to confirm the seasonality of annulus formation; they reported an aging precision of $\pm$ 5 years for Hudson River Atlantic sturgeon.

Sexually mature Atlantic sturgeon do not usually spawn every year (Van Eenennaam et al. 1996; Caron et al. 2002). However, some fish participate in spawning migrations even when they do not spawn (Smith 1985b). There remains a high degree of uncertainty on the frequency of spawning due in part to imprecision in methods, but also due to large natural variability expected in this parameter. In South Carolina, females are thought to spawn every 3 to 5 years, while males spawn at 1 to 5 year intervals (Smith 1985b). Additionally, Smith et al. (1982) found that an average interval of 5.4 years occurred between first and second spawnings, and 3.5 years between second and third spawnings. Vladykov and Greeley (1963) concluded that females spawn once every 2 to 3 years. Results from recent research on gonad histology and hard part analysis of Hudson River Atlantic sturgeon suggest a spawning frequency of 3-5 years (Van Eenennaam et al. 1996; Stevenson and Secor 2000). Interestingly, Collins et al. (2000b) caught and then recaptured a male sturgeon (in 1998 and 1999, respectively) that was in spawning condition for two successive spawning seasons. Similarly, Scott and Crossman (1973) indicated that spawning might occur every year in some females.

Spawning and the saltwater interface

Atlantic sturgeon generally spawn in tidal freshwater regions of estuaries, but may spawn in nontidal freshwater rivers in the southeastern part of their range. Most studies report that Atlantic sturgeon spawn in freshwater above the salt wedge in estuaries (Dovel 1978, 1979; Smith 1985b; Van Eenennaam et al. 1996; Bain et al. 2000). For instance, Dovel (1978, 1979) reported that Atlantic sturgeon in the Hudson River, New York, spawn in freshwater above the salt wedge. Smith (1985a) suggested that spawning fish may migrate seasonally, following the salt front upriver as the season progresses. Dovel and Berggren (1983) reported that the majority of spawning occurred between RKM 56 and 132 in the Hudson River. However, Van Eenennaam et al. (1996) suggest that these results might be questionable because the salt wedge extends to RKM 98. Atlantic sturgeon eggs cannot tolerate high salinity, thus it is more likely that sturgeon spawn above the salt wedge, and not in brackish waters (Van Eenennaam et al. 1996). In addition, Van Eenennaam et al. (1996) found ovulating sturgeon around RKM 136 in the Hudson River system.

Spawning substrate associations

Substrate is a key habitat parameter for Atlantic sturgeon, because a hard bottom substrate is required for successful egg attachment and incubation (Vladykov and Greeley 1963; Huff 1975; Smith 1985b; Gilbert 1989; Smith and Clugston 1997; Secor et al. 2002; Bushnoe et al. 2005). Within rivers, the areas of cobble-gravel, coarse sand, and bedrock outcrops, which occur in the rapids complex, may be considered prime habitat (Table 8-2). In northern rivers, these areas are nearer to the salt-wedge than in southern rivers. South of the Chesapeake Bay, nearly all rivers have extensive rapid-complex habitats in and/or near the fall line zone; these areas are generally at least 100 km upstream from the saltwater interface (P. Brownell, NOAA Fisheries, Southeast Regional Office, personal communication). This habitat provides Atlantic sturgeon with well-oxygenated water, clean substrates for egg adhesion, crevices that serve as shelter for post-hatch larvae, and macroinvertebrates for food (P. Brownell, NOAA Fisheries, Southeast Regional Office, personal communication).

Substrate	Activity	Location	Citation
Rock and bedrock	spawning	St. Lawrence River, Québec	Hatin et al. 2002
Rock, clay, & sand	spawning	St. Lawrence River, Québec	Caron et al. 2002
Irregular bedrock, silt, & clay	spawning	Hudson River, NY	Bain et al. 2000
Clay/silt with rocky shoreline	post-spawning	Hudson River, NY	Bain et al. 2000
Hard clay	spawning	Delaware River	Borodin 1925
Small rubble & gravel	spawning	Delaware River	Dees 1961
Clay	spawning	Delaware River	Scott & Crossman 1973
Limestone	spawning	Edisto River, SC	Collins et al. 2000b
Fine mud, sand, pebbles, & shell	post-spawning	Edisto River, SC	Collins et al. 2000b
Cobble/gravel	spawning	HSI Model	Brownell et al. 2001

Table 8-2.   Spawning (and post-spawn) substrate type for Atlantic sturgeon along the Atlantic coast

Some researchers have attempted to identify likely spawning areas for Atlantic sturgeon using modeling techniques. Brownell et al. (2001) developed a Habitat Suitability Index (HSI) model for spawning Atlantic sturgeon and early egg development, and found that cobble/gravel (64 mm to 250 mm) was the optimal spawning substrate for Atlantic sturgeon. Boulder (250 mm to 4000 mm) scored the second highest in the model, and silt/sand (<2.0mm) and mud/soft clay/fines scored the lowest. The curve and the data values were based on the shortnose sturgeon model, and factors such as oxygenation, substrate embeddedness, available egg attachment sites, protection of eggs from predators, light intensity, and solar warming were all hypothesized to be available in cobble/gravel and boulder substrates (Brownell et al. 2001).

Bushnoe et al. (2005) identified potential spawning areas for Atlantic sturgeon in Virginia based on the location of suitable hard substrate and a variety of other water quality parameters, including temperature, dissolved oxygen, pH, salinity, hardness, and conductivity. They concluded that Turkey Island oxbow and the James Neck oxbow in the James River, the Appomattox River, the Mattaponi, and Pamunkey River in the York River system, and the Rappahannock River, all represented potential spawning habitat (Bushnoe et al. 2005).

Spawning depth associations

Atlantic sturgeon have been documented spawning in water from 3 m to 27 m in depth (Table 8-3) (Borodin 1925; Dees 1961; Scott and Crossman 1973; Shirey et al. 1999; Bain et al. 2000; Collins et al. 2000b; Caron et al. 2002; Hatin et al. 2002). Spawning depth seems to vary greatly depending upon the available depth range.

Depth Range (m)	Status	Location	Citation
10 - 22	Spawning	St. Lawrence River, Québec	Caron et al. 2002
17 - 21	Non-spawning	St. Lawrence River, Québec	Caron et al. 2002
15 - 27 (mean)	All	St. Lawrence River, Québec	Hatin et al. 2002
6 - 60	Spawning	St. Lawrence River, Québec	Hatin et al. 2002
>7.6	Migrating	Hudson River, NY	Dovel and Berggren 1983
12 - 27	Spawning	Hudson River, NY	Bain et al. 2000
11 - 13	Spawning	Delaware River	Borodin 1925; Scott & Crossman 1973
1.5 - 13	All	Edisto River, SC	Collins et al. 2000b
2.4 - 8	Spawning	HSI Model	Brownell et al. 2001

Table 8-3. Spawning (and non-spawn) depth ranges for Atlantic sturgeon along the Atlantic coast

A recent HSI model developed by Brownell et al. (2001) showed that the optimal depth range in the South for spawning Atlantic sturgeon and egg incubation ranged from 2.4 m to 8 m. It should be noted that depth in this model had a maximum range of 8 m because areas where spawning is likely to occur (areas above the fall zone) in the South are not much deeper than 8 m (P. Brownell, NOAA Fisheries, Southeast Regional Office, personal communication).

Spawning water temperature

Atlantic sturgeon reportedly spawn in waters where temperatures range from 13°C to 26°C (Table 8-4) (Borodin 1925; Huff 1975; Smith 1985b; Bain et al. 2000; Caron et al. 2002; Hatin et al. 2002). Temperature appears to be a universal determining factor in spawning migration times. Migration temperatures seem to be fairly uniform across the Atlantic Coast, with southern fish migrating earlier in the spring, and northern fish following a few weeks later once the waters reach the appropriate temperature. Generally, male Atlantic sturgeon commence upstream migration when waters reach around 6°C (Smith et al. 1982; Dovel and Berggren 1983; Smith 1985a). Females usually follow a few weeks later when temperatures are closer to 12°C or 13°C (Dovel and Berggren 1983; Smith 1985a; Smith 1985b; Collins et al. 2000b). Spawning has been found to occur most often in waters 13°C to 21°C (Ryder 1888; Scott and Crossman 1973; Bain et al. 2000; Caron et al. 2002). In addition, Mohler (2003) stated in the "Culture Manual for Atlantic Sturgeon" that the preferred temperature for induced spawning in cultured sturgeons is between 20°C and 21°C.

Sex	Activity	Month	Temperature Range (°C)	Location	Citation
M/F	Spawning	N/A	14.5 - 23.4	St. Lawrence River, Québec	Caron et al. 2002; Hatin et al. 2002
M	Migration Up	N/A	5.6 - 6.1	Hudson River, NY	Smith 1985a
F	Migration Up	♂ + few weeks	12.2 - 12.8	Hudson River, NY	Smith 1985a
M	Migration Up	April	6	Hudson River, NY	Dovel and Berggren 1983
F	Migration Up	♂ + few weeks	13	Hudson River, NY	Dovel and Berggren 1983
M/F	Spawning	N/A	14 - 26	Hudson River, NY	Bain et al. 2000
M/F	Spawning	April - June	12.8 - 18.3	Delaware River	Ryder 1888
M/F	Spawning	N/A	13.3 - 17.8	Delaware River	Scott and Crossman 1973
M	Migration Up	N/A	13 - 19	South Carolina	Smith 1985b
F	Spent	Sept. - Oct.	17 - 18	Edisto River, SC	Collins et al. 2000b
M/F	Migration Up	March	13.6	Edisto River, SC	Collins et al. 2000b
M/F	Present	Summer	up to 33.1	Edisto & Combahee Rivers, SC	Collins et al. 2000b
M/F	Spawning	N/A	20 - 21	Aquaculture facility	Mohler 2003
M/F	Spawning	N/A	16 - 21	HSI Model	Brownell et al. 2001

Table 8-4. Spawning and migration temperatures for Atlantic sturgeon along the Atlantic coast

Spawning water velocity/flow

Atlantic sturgeon lay their eggs in flowing water (Vladykov and Greeley 1963; Van den Avyle 1983). Modeling studies suggest that the optimal water velocities for Atlantic sturgeon spawning range from 0.46 m/s to 0.76 m/s. Furthermore, velocities lower than 0.06 m/s and higher than 1.07 cm/s are unsuitable for spawning (Crance 1987). A recent HSI developed for spawning Atlantic sturgeon showed that optimal water velocity for spawning and egg incubation ranged from 0.2 m/s to 0.76 m/s (Brownell et al. 2001).

Spawning and other water parameters

Reports of gulf sturgeon (*Acipenser oxyrinchus desotoi*) indicate that other important habitat factors include hardness and conductivity. Sulak and Clugston (1999) and Fox et al. (2000) describe the spawning sites of gulf sturgeon on the Suwannee River, Florida, as having a moderate Ca++ ion concentration and conductivity ranging from 10 µS to 110µS. Bushnoe et al. (2005) used these criteria to identify Atlantic sturgeon spawning habitat in rivers in Virginia. More research will be needed to clarify the importance of these parameters.

Spawning feeding behavior

It has been hypothesized that Atlantic sturgeon do not feed during spawning migrations. Research is currently being conducted in South Carolina to test this hypothesis (M. Collins, South Carolina Department of Natural Resources, personal communication). Post-spawning adults that remain in freshwater systems have been documented feeding on gastropods and other benthic organisms (Scott and Crossman 1973). In general, adult Atlantic sturgeon feed indiscriminately throughout their lives and are considered to be opportunistic feeders (Vladykov and Greeley 1963; Murawski and Pacheco 1977; Van den Avyle 1983; Haley and Bain 1997; Colette and Klein-MacPhee 2002). They feed on mollusks, polychaetes, gastropods, shrimps, isopods, and benthic fish in estuarine areas (Dadswell et al. 1984; Secor et al. 2000b; Colette and Klein-MacPhee 2002). In freshwater, their prey includes aquatic insects, nematodes, amphipods, and oligochaetes (Colette and Klein-MacPhee 2002; Hain et al. 2002).

Spawning competition and predation

Adult Atlantic sturgeon appear to have few ecological competitors. They spawn later in the season and in different areas than shortnose sturgeon, thus avoiding competition for egg deposition space in areas where their habitat overlaps (Bath et al. 1981; Gilbert 1989; Kynard and Horgan 2002). Other species that might utilize the same spawning habitat include anadromous species, such as white perch, striped bass, and American shad (D. Secor, Chesapeake Biological Laboratory, University of Maryland Center for Environmental Science, personal communication).

The ASSRT (2007) notes the following information on competition and predation in Atlantic and shortnose sturgeon:

Atlantic sturgeon are benthic predators and may compete for food with other bottom-feeding fishes and invertebrates including suckers (*Moxotoma* sp.), winter flounder (*Pleuronectes americanus*), tautog (*Tautoga onitis*), cunner (*Tautagolabrus adspersus*), porgies (Sparidae), croakers (Sciaenidae), and stingrays (*Dasyatis* sp.) (Gilbert 1989). Specific information concerning competition between Atlantic sturgeon and other species over habitat and food resources is scarce. There are no known exotic or non-native species that compete directly with Atlantic sturgeon. There is a chance that species such as suckers or other bottom forage fish would compete with Atlantic sturgeon, but these interactions have not been elucidated (from ASSRT 2007).

The relationship between the Federally endangered shortnose sturgeon and the Atlantic sturgeon has recently been explored. Shortnose sturgeon are sympatric with Atlantic sturgeon throughout most of their range. Larger, adult shortnose are suspected to compete for food and space with juvenile Atlantic sturgeon in rivers of co-occurrence (Pottle and Dadswell 1979; Bain 1997). Haley and Bain (1997) found that while shortnose and Atlantic sturgeon overlap in their use of the lower estuary, the overall distribution of the two species differed by river kilometers, providing evidence that Atlantic and shortnose sturgeon partition space within the Hudson River despite co-occurrence in channel habitats. This finding is consistent with Kieffer and Kynard (1993) who found that subadult Atlantic and adult shortnose sturgeon in the Merrimack River, MA were spatially separate except for brief use of the same saline reach in the spring.

Kahnle and Hattala (1988) conducted late summer-fall bottom trawl collections in the lower Hudson River Estuary from 1981-1986 and found that most shortnose sturgeon occupied rkm 55-60 in water depths of greater than six meters. Even though there was overlap in river miles, there was separation by water depth. In Georgia, the distributions of adult shortnose and juvenile Atlantic sturgeons overlap somewhat, but Atlantic sturgeon tend to use more saline habitats than shortnose sturgeon (G. Roger, formerly Georgia Department of Natural Resources, personal communication; from ASSRT 2007).

Juvenile shortnose sturgeon apparently avoid competition for food with Atlantic sturgeon in the Saint John River, Canada by spatial separation, but adult shortnose may compete for space with similar-sized juvenile Atlantic sturgeon (Dadswell et al. 1984). Haley and Bain (1997) analyzed stomach contents of Atlantic and shortnose sturgeon in the Hudson River using gastric lavage and found clear differences in their diets. Polychaetes and isopods were primary foods retrieved from Atlantic sturgeon while amphipods were the dominant prey obtained from shortnose sturgeon (Haley and Bain 1997; from ASSRT 2007).

Very little is known about natural predators of Atlantic sturgeon. The presence of bony scutes are likely effective adaptations for minimizing predation of sturgeon greater than 25 mm TL (Gadomski and Parsley 2005). Documented predators of sturgeon (*Acipenser* sp.) include sea lampreys (*Petromyzon marinus*), gar (*Lepisosteus* sp.), striped bass, common carp (*Cyprinus carpio*), northern pikeminnow (*Ptychocheilus oregonensis*), channel catfish (*Ictalurus punctatus*), smallmouth bass (*Micropterus dolomieu*), walleye (*Sander vitreus*), grey seal (*Halichoerus grypus*), fallfish (*Semotilus corporalis*) and sea lion (*Zalophus californianus*) (Scott and Crossman 1973; Dadswell et al. 1984; Miller and Beckman 1996; Kynard and Horgan 2002; Gadomski and Parsley 2005; Fernandes 2006; Wurfel and Norman 2006). In contrast to these findings, Moser et al. 2000 tested whether flathead catfish (*Pylodictus olivaris*) preyed on shortnose sturgeon (30 cm) in a controlled system, and despite sturgeon being the only prey available none were consumed. However, Gadomksi and Parsely (2005) tested at what size white sturgeon were preyed upon by channel catfish, northern pikeminnow, walleyes, and prickly sculpins (*Cottus asper*). Their results found that channel catfish (mean TL = 472 mm), northern pikeminnow (mean TL = 464 mm), and prickly sculpin (mean TL = 126 mm) fed on juvenile sturgeon of an average size of 121 mm TL, 134 mm TL, and 50 mm TL, respectively. Oddly, similar size walleye (~470 mm TL) rarely fed on white sturgeon, but juvenile walleye (mean TL = 184 mm) consumed sturgeon with a mean size of 59 mm TL. Gadomski and Parsley (2005) suggest that these findings indicate that predation could play an important role in sturgeon recovery (from ASSRT 2007).

Similarly, Brown et al. (2005) concluded that the "…introduction of [flathead catfish] has the potential to adversely affect ongoing anadromous fish restoration programs and native fish conservation efforts in the Delaware and Susquehanna basins." The same concern has been stated by fishery management agencies for south Atlantic river basins where flathead catfish are firmly established and have reached significant biomass, significantly altering native fish assemblages and biomass in the process. There is, however, no current evidence that predation rates on Atlantic sturgeon are elevated above "natural" levels (from ASSRT 2007).

Part B. Atlantic Sturgeon Egg and Larval Habitat

Geographical and temporal movement patterns

Due to a low tolerance for saline environments, Atlantic sturgeon eggs must be spawned upstream of the salt front (Van Eenennaam et al. 1996). On the other hand, research on the conspecific *A. o. desotoi* (Gulf sturgeon) indicates that Atlantic sturgeon probably select regions with high conductivity, above the salt wedge, but below fall line regions containing freshwater with low conductivity (Sulak and Clugston 1999; Fox et al. 2000).

Eggs are deposited into flowing water and disperse following fertilization. After approximately twenty minutes, the demersal eggs become strongly adhesive and attach to hard substrates (Murawski and Pacheco 1977; Van den Avyle 1983). The eggs hatch after 94 to 140 hours; subsequent to a pelagic yolksac larval period of about 10 days, late-stage larvae settle in the demersal habitat. This will be the principal type of habitat for the remainder of the sturgeon's life (USFWS-NMFS 1998).

Little is known about the habitat of larval Atlantic sturgeon. Larval Atlantic sturgeon are less than 4 weeks old, with lengths less than 30 mm (TL) (Van Eenennaam et al. 1996); they are assumed to inhabit the same riverine or estuarine areas where they were spawned (Bain et al. 2000; Kynard and Horgan 2002). Newly hatched larvae are active swimmers and leave the bottom to swim in the water column. Once the yolk sac is absorbed, the larvae exhibit benthic behavior (Smith et al. 1980, 1981). Bath et al. (1981) caught free embryos by actively netting the bottom near the spawning area, demonstrating that early life stages are benthic.

For a more controlled experiment, Kynard and Horgan (2002) raised captive Atlantic sturgeon in chambers. They found that upon hatching, the embryos sought cover where they remained for a few days. The fish left cover and began to migrate around day 8. Following the passage of a few more days, the larvae stopped migrating and exhibited foraging behavior. Downstream migration resumed again during the juvenile period when the temperature dropped. Atlantic sturgeon larvae are capable of dispersing long distances. Movement occurs at night during the first half of the larval migration; eventually, the fish become active during both the day and night (Kynard and Horgan 2002). Kynard and Horgan (2002) hypothesize that this foraging behavior is a way to reduce daytime predation while the larvae are still developing, yet still enable them to forage when there is daylight to aid in the visual detection of prey.

Mohler (2003) found similar results. Cultured Atlantic sturgeon were mostly pelagic after hatching and exhibited a "swim up and drift down" behavior. After three to four days, fry began to exhibit benthic clumping behavior and swam against the direction of water flow in the tank. Fry remained benthic for approximately four days, before moving around the tank in search of food. At this stage, the larval Atlantic sturgeon were noted to be pelagic, until live brine shrimp were thrown into the tank and the fry moved to the bottom of the tank to feed. Atlantic sturgeon fry did not actively seek out a food source, but rather waited until the currents brought food to them (Mohler 2003).

The ASSRT (2007) notes that downstream dispersal patterns may be different among watersheds:

> Differences in the innate dispersal patterns of sturgeon species in early life stages also suggest that there are markedly separated differences in behavior between

subpopulations of sturgeon (B. Kynard, USGS Conte Anadromous Fish Research Center, personal communication). Boyd Kynard, a researcher at the USGS Conte Anadromous Fish Research Center (Turner Falls, Massachusetts), has noted major differences in innate dispersal patterns of early life stage sturgeon species including *Acipenser fulvescens* (Wolf and Menominee rivers), *A. brevirostrum* (Connecticut and Savannah rivers), *A. transmontanus* (Sacremento and Kootenai rivers), and Atlantic/Gulf sturgeon subpopulations (Hudson and Suwannee rivers). This research suggests that Atlantic sturgeon are likely adapted to unique features of their watershed, considering their genetic discreteness and differing migration behaviors. These findings are similar to research conducted on striped bass (*Morone saxatilis*), an anadromous fish like Atlantic sturgeon, which correlated egg characteristics (e.g., egg diameter, egg density, etc.) with watershed type (e.g., low, medium, high energy) (Bergey et al. 2003). Differences in egg characteristics likely are the result of subpopulation adaptations to the watershed, but the manner in which these adaptations were produced were not determined. The ASSRT concluded that unique behavioral and physiological traits likely exist for each extant subpopulation of Atlantic sturgeon – except those that share a drainage basin (similar adaptations) (from ASSRT 2007).

Eggs, larvae, and the saltwater interface

Salinity is very important to the survival of sturgeon eggs (McEnroe and Chech 1985; Jenkins et al. 1993; Van Eenennaam et al. 1996). Eggs are spawned in regions between the salt front and the fall-line of large rivers or estuarine tributaries (Borodin 1925; Leland 1968; Scott and Crossman 1973; Crance 1987; Bain et al. 2000). Bath et al. (1981) collected larval sturgeon in salinities of 0 ppt to 22 ppt in the Hudson River, New York. Dovel and Berggren (1983) recorded sturgeon embryos from RKM 60 to RKM 148, which includes some brackish water. However, Van Eenennaam et al. (1996) report that Atlantic sturgeon embryo habitat must be well above the salt wedge, due to their low tolerance to salinity. Other species of sturgeon show this same salt intolerance. For example, free embryos, larvae, and age-0 juveniles of white sturgeon and shortnose sturgeon also exhibit low salt tolerance. Mortality has been documented at salinities as low as 5 ppt to 10 ppt (McEnroe and Chech 1985; Jenkins et al. 1993).

Egg and larval substrate associations

Atlantic sturgeon deposit their eggs on benthic hard substrate (Gilbert 1989; Smith and Clugston 1997). The eggs contain adhesive strings that attach to stones, shells, sticks, and weeds (Vladykov and Greeley 1963; Colette and MacPhee 2002). Hard substrate is also important to larval Atlantic sturgeon, as it provides refuge from predators (Kieffer and Kynard 1996; Fox et al. 2000). A study by Kynard and Horgan (2002) showed that after hatching, embryos immediately sought cover. Some scientists hypothesize that rapid-complex habitats might serve as hatcheries for Atlantic sturgeon because they provide cover, well-oxygenated hiding places, and a food source of microinvertebrates (P. Brownell, NOAA Fisheries, Southeast Regional Office, personal communication).

Egg and larval depth associations

The importance of depth to embryonic and larval Atlantic sturgeon has not been thoroughly discussed in the literature, but it is likely not as important to this species as benthic substrate characteristics (P. Brownell, NOAA Fisheries, Southeast Regional Office, personal communication). However, depth of migrating larvae would be an important issue to address for a project inserting intake structures into a river near nursery grounds (W. Patrick, NOAA Fisheries Service, personal communication). Additionally, Bain (1997) found that embryos remain on the bottom of deep channel habitats, and Bath et al. (1981) collected larval samples from 9.1 m to 19.8 m.

Egg and larval water temperature

Smith et al. (1980) found that Atlantic sturgeon eggs optimally hatch at temperatures ranging from 18°C to 20°C. Hatching occurs approximately 94 to 140 hours after egg deposition at temperatures of 20°C and 18°C, respectively, and larvae assume a demersal existence (Smith et al. 1980). Similarly, Mohler (2003) states that in a culture setting, a temperature range of 20°C to 21°C is favorable for the incubation of Atlantic sturgeon eggs. Temperatures below 18°C prolong hatching and increase the risk of fungal infestation to dead eggs, which in turn can kill the viable individuals. Hatching occurs in 60 hours at this temperature range (Mohler 2003).

Bath et al. (1981) collected larval sturgeon in the Hudson Bay, New York, in temperatures of 15.0°C to 24.5°C. Researchers recommend that first-feeding cultured Atlantic sturgeon fry be kept in water temperatures of 15°C to 19°C, and that a temperature of 19°C yields higher growth rates (Kelly and Arnold 1999; Mohler 2003).

Egg and larval feeding behavior

There are no studies to indicate what larval Atlantic sturgeon prey upon in the wild. However, it is assumed that after they absorb the yolk sac, they feed on small bottom dwelling organisms (Gilbert 1989). Studies of other sturgeon species indicate that larvae in rivers feed on small mobile invertebrates, including cladocerans and copepods (Baranova and Miroshnichenko 1969; Miller et al. 1991). Miller et al. (1991) found that white sturgeon larvae primarily fed on amphipods.

During their lab test, Kynard and Horgan (2002) found that Atlantic sturgeon larvae (30 to 50 days old) preferred illumination and a white substrate. They hypothesize that an illuminated bright substrate may make it easier for young sturgeon to locate moving prey. Laboratory rearing of larvae depends principally on *Artemia* sp. as prey, which the Atlantic sturgeon can readily consume (Kynard and Horgan 2002).

Egg and larval competition and predation

Kynard and Horgan (2002) hypothesize that larval and juvenile Atlantic sturgeon have a low predation risk. This hypothesis is based on the theory that migration upon hatching is stimulated by predation risk to embryos. Species that undergo high predation tend to migrate from the area immediately after hatching (Kynard and Horgan 2002). While this hypothesis has

not been fully tested, Kynard and Horgan (2002) have determined that shortnose sturgeon embryos have few predators. After sampling predators in a spawning area, they found that only one fish, the fallfish (*Semotilus corporalis*), had sturgeon eggs in its stomach (Kynard and Horgan 2002).

Part C. Atlantic Sturgeon Juvenile Estuarine Habitat

Geographical and temporal movement patterns

For the purposes of this report, a sturgeon will be considered juvenile according to the guidelines found in the ASSRT (2007), which broke juveniles down as such:

1) YOY (AGE-0): Thought to be natal to the river they were captured in and used as evidence in identifying extant populations

2) Juveniles or subadults (AGE-1 to AGE-15): Considered possible migrants from other systems though the older individuals could be reproducing (maybe in more northern waters)

3) Mature adults (AGE-15) or 150 cm TL: Generally considered mature, and if they were captured in a river during the spawning season it was assumed that they were going to spawn in that river (used to identify extant populations) (ASSRT 2007)

Most researchers have found that growth rates and sizes of Atlantic sturgeon vary by latitude, with rapid growth occurring in the southern latitudes and larger maximum sizes occurring in the north (Vladykov and Greeley 1963; Smith 1985a; Smith 1985b; Dovel and Berggren 1983; Collins et al. 1996; Stevenson and Secor 1999). However, Johnson et al. (2005), working off the New Jersey coast, found that their data did not fit this pattern. They suggested that this might have been due to a mixed sample composed of Atlantic sturgeon from different populations that had different growth rates (Johnson et al. 2005). These findings are partially supported by genetic studies performed by Waldman et al. (1996a) who showed that approximately 90% of the Atlantic sturgeon catch in the New York Bight was of Hudson River origin.

The ASSRT (2007) notes the following information on juvenile Atlantic sturgeon migrations:

Upon reaching a size of approximately 76 to 92 cm, the sub-adults may move to coastal waters (Murawski and Pacheco 1977; Smith 1985b), where populations may undertake long-range migrations (Dovel and Berggren 1983; Bain 1997; T. King, USGS Leetown Science Center, Aquatic Ecology Laboratory, Kearneysville, West Virginia, supplemental data). Tagging and genetic data indicate that sub-adult and adult Atlantic sturgeon may travel widely once they emigrate from rivers. Sub-adult Atlantic sturgeon wander among coastal and estuarine habitats, undergoing rapid growth (Dovel and Berggren 1983; Stevenson 1997). These migratory sub-adults, as well as adult sturgeon, are normally captured in shallow (10 to 50m) near shore areas dominated by gravel and sand substrate (Stein et al. 2004; from ASSRT 2007).

Juvenile Atlantic sturgeon are thought to remain close to their natal habitats within the freshwater portion of the estuary for at least one year before commencing migration out to sea (Secor et al. 2000b). Migrations out to coastal areas occur between two and six years of age (Smith 1985b), and are seasonal, with movement occurring north in the late winter, and south in fall and early winter (Dovel 1978; Smith 1985b; USFWS-NMFS 1998). Seasonal migrations of juveniles are regulated by changes in temperature gradients between fresh and brackish waters (Van Den Avyle 1984). For example, hatchery-reared juveniles released in the Chesapeake Bay

used brackish waters close to the estuary mouth during colder months, and moved upriver during warmer months (Secor et al. 2000b).

Similar behavior has been seen in a number of river systems, including the Delaware River, Hudson River, and the Winyah Bay system (South Carolina) (Brundage and Meadows 1982; Smith et al. 1982; Dovel and Berggren 1983; Gilbert 1989). Dovel and Berggren (1983) reported a mass down-estuary migration of juvenile Atlantic sturgeon in the Hudson Estuary, New York, when the temperature dropped below 20°C. Down-river/down-estuary migrations peak at the end of October in the Hudson system. At this time, many juveniles overwinter in deep holes, while others leave the Hudson River and move south along the Atlantic coast (Dovel and Berggren 1983). In contrast, Moser and Ross (1995) found that juvenile sturgeon in the Cape Fear River, North Carolina, kept the same center of distribution near the saltwater-freshwater interface year round. However, these fish were unable to move upriver because of the location of the Cape Fear Lock and Dam No. 1, just above the estuary (0.5 ppt interface) (P. Brownell, NOAA Fisheries, Southeast Regional Office, personal communication).

Coastal features or shorelines where migratory Atlantic sturgeon commonly aggregate include the Bay of Fundy, Massachusetts Bay, Rhode Island, New Jersey, Delaware, Delaware Bay, Chesapeake Bay, and North Carolina, which presumably provide better foraging opportunities (Dovel and Berggren 1983; Johnson et al. 1997; Rochard et al. 1997; Kynard et al. 2000; Eyler et al. 2004; Stein et al. 2004; Dadswell 2006). Smith (1985b) stated that fish tagged off South Carolina migrated as far north as Pamlico Sound and Chesapeake Bay. Most data indicate that Atlantic sturgeon in the northern rivers travel more extensively than those in the southern rivers (ASMFC 1998). However, research in the southern region has not adequately addressed inter-basin movements in the south (P. Brownell, NOAA Fisheries, Southeast Regional Office, personal communication).

Later-stage juveniles often enter and reside in non-natal rivers that lack active spawning sites (Bain 1997). Inter-estuarine migrations have been documented extensively in the literature (Dovel and Berggren 1983; Smith 1985b; Welsh et al. 2002; Savoy and Pacileo 2003). These non-natal estuarine habitats serve as nursery areas, providing abundant foraging opportunities and thermal and salinity refuges. Therefore, these areas are very important to the Atlantic sturgeon's survival (Moser and Ross 1995).

Juveniles and the saltwater interface

There is a large amount of variation in the salinity tolerance of juvenile Atlantic sturgeon (Table 8-5). Some Atlantic sturgeon may occupy freshwater habitats for two or more years, while others move downstream to brackish waters when the water temperature drops (Scott and Crossman 1973; Dovel 1978; Hoff 1980; Lazzari et al. 1986). Additionally, bioenergetic studies on YOY juveniles indicate poor survival at salinities greater than 8 ppt, but euryhaline behaviors are exhibited by juveniles age-1 and 2 (Niklitschek 2001).

Salinity Range (ppt)	Location	Citation
>3	Hudson River, New York	Appy and Dadswell 1978
3 - 16	Hudson River, New York	Dadswell 1979
3 - 16	Hudson River, New York	Brundage and Meadows 1982
0 - 6	Hudson River, New York	Dovel and Berggren 1983
3 - 16	Hudson River, New York	Smith 1985b
3 - 16	Hudson River, New York	Haley et al. 1996
>3	Hudson River, New York	Bain et al. 2000
0 - 12	Delaware River	Shirey et al. 1999
<10	Brunswick River, North Carolina	Moser and Ross 1995

Table 8-5. Salinity tolerance ranges for young juvenile Atlantic sturgeon along the Atlantic coast

Juvenile substrate associations

Kynard et al. (2000) reported that juvenile Atlantic sturgeon in Massachusetts were found mostly over sand substrates, but other associated substrates included rock, cobble, and mud (Kynard et al. 2000). Savoy and Pacileo (2003) found that 85% of the juvenile Atlantic sturgeon caught in Long Island Sound were in mud or transitional bottom habitats. Correspondingly, Bain et al. (2000) found juveniles off Long Island Sound over mud substrates. In the Hudson River, Haley et al. (1996) collected juvenile Atlantic sturgeon at sites that had silt substrates. However, the researchers state that it is unclear whether this represents habitat preference or habitat use, as the majority of sites sampled was composed of this substrate (Haley et al. 1996). In the same system, Bain et al. (2000) documented juveniles over clay, silt, and sand substrates. Stein et al. (2004) found migratory sub-adults, as well as adult Atlantic sturgeon, generally in areas dominated by gravel and sand substrate.

Juvenile depth associations

Many researchers have found that juvenile Atlantic sturgeon tend to congregate in deep waters (Table 8-6) (Moser and Ross 1995; Bain et al. 2000; Savoy and Pacileo 2003). Moser and Ross (1995) report that juvenile Atlantic sturgeon in North Carolina use deep and cool areas as thermal refuges, particularly in the summertime.

Depth Range (m)	Location	Citation
2 - 12	Massachusetts	Kynard et al. 2000
30 - 40	Long Island Sound, Connecticut	Bain et al. 2000
27 - 37	Long Island Sound, Connecticut	Savoy and Pacileo 2003
Mean = 22.7	Hudson River, New York	Haley et al. 1996
10 – 25 (<700 mm TL)	Hudson River, New York	Bain et al. 2000
16 – 26 (>700 mm TL)	Hudson River, New York	Bain et al. 2000
7 - 16	Delaware River, Pennsylvania	Lazzari et al. 1986
5.5 - 11	Delaware River, Delaware	Shirey et al. 1999
<20	Chesapeake Bay, Virginia	Musick et al. 1994
<7	Brunswick River, North Carolina	Moser and Ross 1995
>10	Cape Fear River, North Carolina	Moser and Ross 1995
1.8 - 5.4	Albemarle Sound, North Carolina	Armstrong and Hightower 2002

Table 8-6. Depth ranges for young juvenile Atlantic sturgeon along the Atlantic coast

Juvenile Atlantic sturgeon farther north also seem to prefer deeper areas. Bain et al. (2000) stated that those juveniles that did not migrate out to sea during the winter occupied deep-water habitat in the Hudson River, New York. Further north, Savoy and Pacileo (2003) found that juvenile Atlantic sturgeon in Long Island Sound preferred the deep-water areas within the central basin of the Sound. They reported that 71% of the Atlantic sturgeon were caught in areas of the deepest stratum (deeper than 27 m). This area comprised only 26% of the available habitat (Savoy and Pacileo 2003). Savoy and Pacileo (2003) also reported that Atlantic sturgeon were rarely caught in the shallow areas (5 m to 9 m), and that the 20 fish caught in the shallow stratum were fish migrating in and out of Long Island Sound.

While the majority of juvenile Atlantic sturgeon have been collected at the deepest depths available, some have also been collected in shallower waters (Table 8-6). A telemetry study on hatchery-released age-1 juveniles showed that most Atlantic sturgeon utilized depths less than 6 m (Secor et al. 2000b).

Juvenile water temperature

Temperature Range (°C)	Location	Citation
13.2 – 26.7	Merrimack River, Massachusetts	Kieffer and Kynard 1993
24.2 – 24.7	Hudson River, New York	Dovel and Berggren 1983
27	Hudson River, New York	Haley et al. 1996
24 – 28	Hudson River, New York	Bain et al. 2000

Table 8-7.   Summer temperature ranges for juvenile Atlantic sturgeon along the Atlantic coast

Temperature is a key habitat parameter for the structuring of juvenile Atlantic sturgeon summer habitat (Table 8-7) (Niklitschek and Secor 2005). Temperatures in excess of 28°C are judged to have sublethal effects on Atlantic sturgeon. An increase in temperature coupled with low dissolved oxygen and high salinity can cause loss of juvenile Atlantic sturgeon nursery habitat. Their low tolerance to temperature and low oxygen is of particular concern during the first two summers of life when juveniles are restricted to lower saline waters, and are unable to seek out thermal refuge in deeper waters (Secor and Gunderson 1998; Niklitschek 2001; Niklitschek and Secor 2005).

Temperature may also be an important habitat parameter with regard to migration patterns, since juvenile Atlantic sturgeon appear to migrate in response to certain temperature thresholds. Dovel and Berggren (1983) stated that downstream migrations in the Hudson River began when temperatures reached 20°C, and peaked between 12°C and 18°C. By the time the temperature was 9°C, juvenile Atlantic sturgeon had congregated for the winter in deep holes (Dovel and Berggren 1983) where water temperatures can approach 0°C (Bain et al. 2000). Similar migration patterns were noted by Dovel (1979) in the Hudson River and by Brundage and Meadows (1982) in the Delaware River. However, Lazzari et al. (1986) reported that juvenile Atlantic sturgeon in the Delaware River used the tidal portion of the bay for a longer period of time and at lower temperatures than reported by other researchers. T hey found Atlantic sturgeon in these areas through December when temperatures approached 0.5°C.

Kieffer and Kynard (1993) found during their biotelemetry studies that juvenile Atlantic sturgeon in the Connecticut and Merrimack Rivers, Massachusetts, did not enter the river until mid-May when the temperatures were 14.8°C to 19.0°C. The fish left the river by September or October when river temperatures were 13°C to 18.4°C (Kieffer and Kynard 1993).

Temperature may also affect juvenile Atlantic sturgeon feeding behavior. Mohler (2003) found that in cultured juvenile Atlantic sturgeons, a noticeable decrease in feeding occurred when temperatures dropped to 10°C. However, minimum weight gains were noticed at temperatures as low as 5.4°C, with weight loss occurring at lower water temperatures (Mohler 2003).

Juvenile dissolved oxygen associations

Dissolved oxygen is a very important habitat parameter for juvenile Atlantic sturgeon. A large proportion of Atlantic sturgeon nursery habitat has been degraded as a result of persistent low levels of dissolved oxygen. Secor and Niklitschek (2001) report that in habitats with less than 60% oxygen saturation (4.3 mg/L to 4.7 mg/L at 22°C to 27°C), YOY fish aged 30 to 200 days, will experience a loss in growth. Mortality of juvenile Atlantic sturgeon has been observed for summer temperatures at levels of less than or equal to 3.3 mg/L (Secor and Niklitschek 2001). Recently, the Chesapeake Bay Program adopted dissolved oxygen guidelines based upon levels that would protect Atlantic and shortnose sturgeon, which show unusually high sensitivity to low oxygen concentrations among estuarine living resources (Secor and Niklitschek 2002; EPA 2003).

Juvenile feeding behavior

Pottle and Dadswell (1982) examined the gut contents of juvenile Atlantic sturgeon in the St. Johns River, Florida. They found that juvenile Atlantic sturgeon fed on diptera and trichoptera, in addition to amphipods. Secor et al. (2000b) found that juvenile Atlantic sturgeon in the Chesapeake Bay preyed upon annelid worms, isopods, amphipods, chironomid larvae, and mysids. Moser and Ross (1995) found polycheate worms, isopods, and mollusk shell fragments in the stomachs of juvenile sturgeon in North Carolina. An examination of 12 juvenile Atlantic sturgeon in the Connecticut and Merrimack Rivers showed a mix of amphipods and polychaetes (Kynard et al. 2000). In freshwater, juvenile Atlantic sturgeon ate plant and animal matter, sludgeworms, chironomid larvae, mayfly larvae, isopods, amphipods, and small bivalve mollusks (Scott and Crossman 1973). Scott and Crossman (1973) also noted that sturgeon consumed mud while foraging on the bottom.

Secor et al. (2000b) analyzed the gut content of 12 juvenile Atlantic sturgeon in the Chesapeake Bay and found that sand, silt, and detritus accounted for 34% of the gut contents. Annelid worms made up 61% of the prey items, followed by isopods (*Cyathura polita* and *Cyathura* sp.; 23%), amphipods (*Leptocheirus plumulosus* and *Gammarus* sp.; 10%), chironomid larvae (1.6%), and mysids (*Neomysis americana*; 1.5%). One-third of the Atlantic sturgeon had empty guts (Secor et al. 2000b). In this small study, Secor et al. (2000b) did not find that juvenile Atlantic sturgeon preyed upon mollusks, despite their high biomass in the Chesapeake Bay.

Juvenile competition and predation

Both juvenile Atlantic sturgeon and shortnose sturgeon occupy the same freshwater/saltwater interface nursery habitat, although shortnose sturgeon tend to be located in freshwater, while Atlantic sturgeon utilize more saline areas (Dadswell 1979; Dovel and Berggren 1983; Dovel et al. 1992; Kieffer and Kynard 1993; Haley et al. 1996; Bain 1997). Haley et al. (1996) collected the majority of juvenile Atlantic sturgeon in the Hudson River in deeper, mesohaline (3.0 ppt to 16.0 ppt) regions, while juvenile shortnose sturgeon were found most often in the shallower, freshwater (<0.5 ppt) zones of the estuary. Furthermore, bioenergetic comparisons showed that age-1 Atlantic sturgeon demonstrated better growth in brackish water (1 ppt to 10 ppt), than sympatric shortnose sturgeon juveniles (Niklitschek 2001).

In contrast, Bain (1997) found that early juvenile Atlantic sturgeon had the same distribution as juvenile shortnose sturgeon in the Hudson River estuary during all seasons. Both species were similar in size, grew at about the same rate, had similar diets, and shared deep channel habitats early in life (Bain 1997). Additionally, Bain (1997) found that the distribution of adult shortnose sturgeon overlapped with the distribution of juvenile Atlantic sturgeon.

Haley et al. (1996) hypothesized that the freshwater/saltwater interface where both sturgeon species concentrate, may serve as a foraging ground, and that Atlantic and shortnose sturgeon may compete for food in this area. However, Pottle and Dadswell (1982) found that juvenile Atlantic and shortnose sturgeon in the St. Johns River preyed on different species. They found that Atlantic sturgeon preyed upon diptera, trichoptera, and some amphipods, while shortnose sturgeon preyed mostly upon cladocerans, amphipods, mollusks, and insect larvae (Pottle and Dadswell 1982). When reared in large outdoor tanks and fed an artificial diet, shortnose sturgeon juveniles fed at higher rates and grew more rapidly than similar sized Atlantic sturgeon (Niklitschek 2001).

In more southern rivers, juvenile Atlantic sturgeon and adult shortnose sturgeon may share parts of the river with similar salinity levels. This has been documented in the Savannah River during the fall and winter, and in the Altamaha River during warm summers (Kieffer and Kynard 1993).

Atlantic sturgeon juveniles would be expected to compete with other demersal feeding fishes in estuaries. In mid-Atlantic estuaries these demersal feeders include catfishes, white perch, carp, spot, croaker, and hogchoker (Murdy et al. 1997).

Part D. Atlantic Sturgeon Late Stage Juvenile and Adult Marine Habitat

All estuarine habitats for adult and juvenile Atlantic sturgeon are discussed under previous sections. This section focuses entirely on juvenile and adult Atlantic sturgeon habitat in marine waters.

Geographical and temporal patterns at sea

Juvenile Atlantic sturgeon are known to emigrate out of their natal estuarine habitats and migrate long distances in the marine environment (Murawski and Pacheco 1977); the longest oceanic journey recorded was 1,450 km (Magnin and Beaulieu 1963). Tag returns (n = 120) of juvenile Atlantic sturgeon that were originally tagged in the Delaware River provide insight into the coastal migration of this life stage that encompasses a broad size range (C. Shirey, Delaware Department of Fish and Wildlife, unpublished data). After leaving the Delaware River estuary during the fall, juvenile Atlantic sturgeon were recaptured by commercial fishermen in nearshore waters along the Atlantic coast as far south as Cape Hatteras, North Carolina, where they were recaptured from November through early March. Juvenile Atlantic sturgeon repeatedly crossed the mouth of the Chesapeake Bay and traveled around the Delmarva Peninsula in March and April, with a portion of the tagged fish re-entering the Delaware River estuary. However, many fish continued this northerly coastal migration through the mid-Atlantic and into southern New England waters where they were recovered throughout the summer months, primarily in the waters of Massachusetts, Rhode Island, and Long Island, New York. Movements as far north as Maine were documented. A southerly coastal migration was apparent from tag returns reported in the fall. The majority of these tag returns were reported from relatively shallow nearshore fisheries with few fish reported from waters in excess of 25 m (C. Shirey, Delaware Department of Fish and Wildlife, unpublished data).

Little is known about the habitat use of adult Atlantic sturgeon during the non-spawning season, particularly when the sturgeon return to marine waters (Bain 1997; Collins et al. 2000b). While at sea, adult Atlantic sturgeon have been documented using relatively shallow nearshore habitats (10 m to 50 m) (Laney et al. 2007; Stein et al. 2004). It is possible that individual fish select habitats in the same areas, or even possibly school to some extent (Bain et al. 2000; Stein et al. 2004; Laney et al. 2007).

Substrate associations at sea

Stein et al. (2004) reported that Atlantic sturgeon were found mostly over sand and gravel substrate, and that they were associated with specific coastal features, such as the mouths of the Chesapeake Bay and Narragansett Bay, and inlets in the North Carolina Outer Banks. Laney et al. (2007) found similar results off the coasts of Virginia and North Carolina. The researchers used GIS layers to analyze data from the Cooperative Winter Tagging Cruise, and found that Atlantic sturgeon were located primarily in sandy substrates. However, the authors state that GIS does not depict small-scale sediment distribution, thus only a broad overview of sediment types was used. In addition, sediment sampling done along the North Carolina coast shows that gravel substrates are found a little farther offshore from where the sturgeon were found (Laney et al. 2007).

Depth associations at sea

The greatest depth in the ocean at which Atlantic sturgeon have been reported caught was 75 m (Colette and Klein-MacPhee 2002). Collins and Smith (1997) report that Atlantic sturgeon were captured at depths of 40 m in marine waters off South Carolina. Stein et al. (2004) investigated data collected by on-board fishery observers from 1989-2000 to determine habitat preferences of Atlantic sturgeon. They found that Atlantic sturgeon were caught in shallow (<60 m) inshore areas of the Continental Shelf. Sturgeon were captured in depths less than 25 m along the Mid-Atlantic Bight, and in deeper waters in the Gulf of Maine (Stein et al. 2004).

The Northeast Fisheries Science Center bottom trawl survey caught 139 Atlantic sturgeon from 1972-1996 in waters from Canada to South Carolina. They found the fish in depths of 7 m to 75 m, with a mean depth of 17.3 m. Of the fish caught, 40% were collected at 15 m, 13% at 13 m, and less than 5% at all the depth strata (NEFC, unpublished data, reviewed in Savoy and Pacileo 2003).

Upon entering the marine habitat, Atlantic sturgeon have been documented near the shore in shallow waters where the depths measure less than 20 m (Gilbert 1989; Johnson et al. 1997). During their tagging cruise off the coasts of Virginia and North Carolina, Laney et al. (2007) captured Atlantic sturgeon at depths up to approximately 6 m. Vladykov and Greeley (1963) record a maximum depth of at least 18 m. Additionally, Johnson et al. (2005) reported that Atlantic sturgeon were caught within 5 km of the coast of New Jersey in waters approximately 15 m deep.

Feeding behavior at sea

There is little information regarding the marine diet of Atlantic sturgeon. Johnson et al. (1997) suggest that this is because of the low population density of Atlantic sturgeon offshore, and the fact that most studies have focused on rivers and estuaries. A stomach content study by Johnson et al. (1997) found that Atlantic sturgeon off the coast of New Jersey preyed upon polychaetes, isopods, decapods, and amphipods. They also found that mollusks and fish contributed little to the diet, and that sand and organic debris were major components (Johnson et al. 1997). Scott and Crossman (1973) stated that in marine waters, Atlantic sturgeon fed on mollusks, polychaete worms, gastropods, shrimps, amphipods, isopods, and small fish (particularly sand lances).

Competition and predation at sea

Atlantic sturgeon compete with other bottom feeding fish and invertebrates. Gilbert (1989) lists winter flounder (*Pleuronectes americanus*), tautog (*Tautoa onitis*), cunner (*Tautogolabrus adspersus*), porgies (Sparidae), croakers (Sciaenidae), and stingrays (*Dasyatis* sp.) as possible competitors. Scott and Crossman (1973) report that Atlantic sturgeon are killed by sea lampreys, *Petromyzon marinus*; in South Carolina, long nose gar have been reported attacking sturgeon (Smith 1985b). Other predators can be found in Part A of this chapter.

Section II. Identification and Distribution of Habitat Areas of Particular Concern for Atlantic Sturgeon

Habitat types that qualify as Habitat Areas of Particular Concern for Atlantic sturgeon include spawning sites/hatching grounds, nursery areas, inlets, and wintering grounds.

Spawning sites/hatching grounds occur in freshwater portions of estuaries and large river tributaries along the Atlantic coast. These areas provide the habitat parameters essential for reproduction, including well oxygenated water, clean substrates for egg adhesion, and crevices that provide cover for post-hatch larvae and abundant macroinvertebrate prey items. This habitat type is very sensitive to anthropogenic impacts, including dams and other river impoundments, nutrient and sediment loading, pollution, navigational dredging, and other coastal developments (especially those with intake structures). Spawning sites are very limited and have been rendered inaccessible and/or degraded since coastal areas have become industrialized and developed.

Nursery areas are limited to freshwater/estuarine tributaries for Atlantic sturgeon age 0-2; nursery areas include bays, estuaries, and nearshore ocean environments for older juveniles (age >2). Freshwater and low salinity areas are important to larvae and age-0 juveniles, because they cannot tolerate high salinity (Secor and Niklitschek 2002). Nursery habitats for juvenile Atlantic sturgeon are essential for growth of this species. This habitat provides foraging grounds for juvenile Atlantic sturgeon, and in some cases, thermal refuge during the summer and winter months (Moser and Ross 1995). Nursery habitats are severely impacted by hypoxic conditions, particularly during summer months when high temperatures can combine with low oxygen levels to degrade and eliminate valuable habitat for juveniles (Secor and Niklitschek 2002; McBride 2004). Other anthropogenic impacts include navigational dredging and port development, sedimentation, nutrient loading (which leads to hypoxic conditions), and recreational and commercial vessel traffic. While nursery areas are less limited in extent than spawning areas, they are still scarce.

Estuarine inlets provide adult and intermediate/late juvenile Atlantic sturgeon with migration corridors to and from freshwater spawning habitat and estuarine nursery grounds. The importance of these areas to Atlantic sturgeon has not been researched; inlets are potentially more rare than spawning habitats. Inlets are impacted by channel alterations (deepening and stabilization) and commercial and recreational coastal development activities. Examples of inlets used by juvenile and adult Atlantic sturgeon include New York Harbor, Delaware Bay, Oregon Inlet, Hatteras Inlet, and Okracoke Inlet for Atlantic sturgeon entering/leaving the Cape Fear River, North Carolina. For movement into or out of the James River, Virginia, fish must migrate through the mouth of the Chesapeake Bay (W. Laney, U. S. Fish and Wildlife Service, personal communication).

Wintering Grounds for adult and late juvenile Atlantic sturgeon include the nearshore areas off the Atlantic coast from the Gulf of Maine south to at least Cape Lookout, North Carolina (Stein et al. 2004; Laney et al. 2007). These areas provide Atlantic sturgeon with foraging grounds and habitat for most of the year (Johnson et al. 1997). Anthropogenic impacts include habitat degradation due to fishing activities, commercial navigation, oil and gas exploration, and construction of offshore liquefied natural gas (LNG) facilities. Ghost fishing may result in sturgeon losses due to entanglement in lost gear. Winter habitat occurs in coastal nearshore waters, which is expected to not be as limited as spawning habitats and inlets.

Section III.   Present Conditions of Habitat and Habitat Areas of Particular Concern for Atlantic Sturgeon

Habitat quantity

Although the amount has not been quantified, Atlantic sturgeon habitat has decreased or been degraded by clear-cutting, agricultural practices, dams, and other channel and watershed modifications since the eighteenth and nineteenth centuries (Hill 1996; Secor et al. 2002; Bushnoe et al. 2005). Historically, Atlantic sturgeon were documented in 38 rivers ranging from the Hamilton Inlet on the coast of Labrador to the St. Johns River in Florida. The ASSRT (2007) most recently reported that 35 of those historical rivers have Atlantic sturgeon present, and 20 are believed to be extant reproducing populations. Once abundant in every river and associated estuary within their range, Atlantic sturgeon have now either been extirpated, or are at historically low levels. Consequently, although Atlantic sturgeon still remain throughout much of their former range, their numbers have been severely reduced (ASSRT 2007).

Habitat quality

The quality of Atlantic sturgeon habitat has been seriously impacted by human actions. Since European settlement, overfishing, habitat loss, and poor water quality have all contributed to the decline of Atlantic sturgeon stocks. Most of these impacts have been gradual and are poorly understood (Smith 1985b; ASFMC 1998; USFWS-NMFS 1998; Secor and Gunderson 1998; Secor et al. 2000a; Secor and Niklitschek 2001; ASSRT 2007).

Section IV. Significant Environmental, Temporal, and Spatial Factors Affecting Distribution of Atlantic Sturgeon

Table 8-8. Significant environmental, temporal, and spatial factors affecting distribution of Atlantic sturgeon. This table summarizes the current literature on Atlantic sturgeon habitat associations. For most categories, optimal and tolerable ranges have not been identified, and the summarized habitat parameters are listed under the category reported. In some cases, unsuitable habitat parameters are defined. NIF = No Information Found. N/A = Not Applicable.

Life Stage	Time of Year and Location	Depth (m)	Temperature (°C)	Salinity (ppt)	Substrate	Current Velocity (m/sec)	Dissolved Oxygen (mg/L)
Adult (Spawning)	Freshwater rivers and possibly tidal freshwater regions of large estuaries (in the north) Feb – Southern states April-May – Mid-Atlantic May-July – Northern States and Canada Sept-Dec – Second spawning documented in Southern regions	**Tolerable:** NIF **Optimal:** 2.4-8+ (HSI model for Southern Regions) **Reported:** 3-27	**Tolerable:** NIF **Optimal:** 16-21 (HSI model for Southern Regions); 20-21 for cultured sturgeon **Reported:** Male migrations 5.6-6.1; Female migrations 12.2-13; Spawning 13-23.4	**Tolerable:** NIF **Optimal:** NIF **Reported:** Above the salt wedge in fresh water.	**Tolerable:** NIF **Optimal:** Cobble/gravel >64mm-250mm (HSI model for Southern Regions) **Reported:** Hard substrate, including rubble, gravel, clay, rock, bedrock, slag from old steel mills and limestone	**Tolerable:** NIF **Optimal:** 0.2 - 0.76 **Reported:** 0.46 – 0.76 okay; unsuitable if $\leq$0.06, or $\geq$1.07	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF
Adult (Estuarine)	Sturgeon do not spawn every year, yet may participate in an upstream migration. After spawning, some sturgeon remain in the rivers through the summer, while others migrate to sea. Downstream migrations occur Sept – Nov in Canada. Present in South March – Oct. Overwinter in the ocean.	**Tolerable:** NIF **Optimal:** NIF **Reported:** 1.5-60	**Tolerable:** NIF **Optimal:** NIF **Reported:** Adult sturgeon documented in waters with temperatures as high as 33.1 in SC.	**Tolerable:** NIF **Optimal:** NIF **Reported:** Documented summer habitat in upper/fresh/ brackish interface, lower interface, and high salinity portions of estuaries in SC. Salinity ranged from 0-28.6	**Tolerable:** NIF **Optimal:** NIF **Reported:** Found over fine mud, sand, pebbles, and shell substrate	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF

Life Stage	Time of Year and Location	Depth (m)	Temperature (°C)	Salinity (ppt)	Substrate	Current Velocity (m/sec)	Dissolved Oxygen (mg/L)
Egg and Larval	Eggs are laid in flowing water in rivers along the Atlantic coast. Larval sturgeon are found in same habitat where spawned and are benthic.	**Tolerable:** NIF **Optimal:** 2.4-8+ for egg incubation (HSI model for Southern Regions) **Reported:** Embryos remain in deep channels. Larval collected at 9.1-19.8	**Tolerable:** NIF **Optimal:** 20-21 Cultured sturgeon **Reported:** Eggs hatch in 94-140 hours ranging from 15.0 – 24.5	**Tolerable:** NIF **Optimal:** NIF **Reported:** Found upstream of salt front; have a low tolerance to salinity; mortality reported in 5-10 for some sturgeon species	**Tolerable:** NIF **Optimal:** Cobble/gravel >64mm-250mm (HSI model for Southern Regions) **Reported:** After 20 minutes, eggs become adhesive and attach to hard substrate. Larvae also use hard substrate as refuge	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF
Juvenile (Estuarine)	Remain in natal habitats within estuary for up to a year before migrating out to sea. Migrations to other estuaries are common. Use brackish water near month of estuary during winter and move up-estuary during warmer months	**Tolerable:** NIF **Optimal:** Deep water and holes serve as thermal refuge **Reported:** 2-37	**Tolerable:** 3-28 **Optimal:** ~20 **Unsuitable:** Temperatures >28 are sub-lethal **Reported:** Downstream migration begins when water reaches 20°C and peaks between 12-18°C. Documented range of 0.5-27	**Tolerable:** NIF **Optimal:** ~10 **Reported:** Large juveniles found mostly where salinity is >3; found in 0-27.5	**Tolerable:** NIF **Optimal:** NIF **Reported:** Found mostly over sand substrate and mud or transitional habitats. Also found over rocks and cobble	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** >5 mg/L **Reported:** Mortality at summer temperatures (26°C) observed at levels <3.3mg/L

Life Stage	Time of Year and Location	Depth (m)	Temperature (°C)	Salinity (ppt)	Substrate	Current Velocity (m/sec)	Dissolved Oxygen (mg/L)
Juvenile and adult (At-sea)	Utilize marine waters during non-spawning seasons. Nearshore areas off the Atlantic coast from the Gulf of Maine to at least Cape Lookout, NC. Little is known about this part of their lives	**Tolerable:** NIF **Optimal:** NIF **Reported:** Most found in shallow waters; greatest depth recorded = 75; depth range 7-43	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** Marine waters off the continental shelf	**Tolerable:** NIF **Optimal:** NIF **Reported:** Sand, gravel, silt and clay. Suggested that they will use any substrate that supports their food resource	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF

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Chapter 9

STRIPED BASS

(*Morone saxatilis*)

In Memoriam

James Benton: April 19, 1958 – November 15, 1995
David G. Deuel: July 31, 1939 – February 17, 1995
Dr. Eileen Setzler-Hamilton: April 28, 1943 – March 12, 2003
Dr. William W. Hassler: June 16, 1917 – February 16, 2008

This chapter is dedicated to the memory of Jim Benton, David G. Deuel, Dr. Eileen Setzler-Hamilton, and Dr. William W. Hassler, four friends and valued colleagues with whom those of us in the striped bass management community were privileged to work for not nearly enough years. Jim, Dave, Bill, and Eileen all worked tirelessly during their careers for the conservation of the striped bass and its supporting ecosystems in the Chesapeake Bay, the Roanoke and Neuse river basins, and off the Outer Banks of North Carolina and Virginia, and their efforts bore much fruit. They are very much missed, and remembered.

Section I. Striped Bass Description of Habitat

Striped Bass General Habitat Description and Introduction

The striped bass (*Morone saxatilis*) was one of the first fish species in North America to be actively used and managed by society (Mann 2005, 2007; Smith and Olsenius 2007). Historically, striped bass was a highly important subsistence, commercial, and recreational species to Native Americans for millennia, and to European travelers and invaders beginning with the Vikings for centuries. Their importance as a harvested species continues into the present. Aside from their importance to humans, it is likely that striped bass provide several highly important ecosystem functions, including structuring fish and invertebrate communities through predation, and providing trophic linkages between productive rivers and estuaries and the coastal Atlantic Ocean (Able 2004). From this perspective, the striped bass may be seen as an indicator of estuarine and coastal health and habitat quality. The importance of this fish species remains undiminished today, and if anything, the relatively recent collapse (early 1980's) and restoration (1995) of the migratory striped bass population and fishery has only heightened public interest in management efforts (W. Laney, personal observation).

The Chesapeake Bay is the epicenter of migratory striped bass abundance and production on the East Coast. However, other estuaries from the Cape Fear River, North Carolina, to the St. Lawrence River, Canada, as well as the nearshore Atlantic Ocean, contribute to production and are essential for the long-term survival and sustainability of the species (W. Laney, personal observation). The purpose of this chapter is to describe the habitats used by all life stages of migratory striped bass, and establish a basis for formal habitat designation.

The striped bass is an anadromous, schooling species with a historic native range extending discontinuously from the Canadian Maritime Provinces to the Gulf of Mexico (Lee et al. 1980; Fay et al. 1983; Hill et al. 1989; Rago 1992; Rulifson and Dadswell 1995; Richards and

Rago 1999). On the Atlantic coast, the range of striped bass is continuous from the St. Lawrence River and southern Gulf of St. Lawrence, Quebec, to the St. Johns River, Florida (McLane 1955; Leim and Scott 1966). The species is absent from southeast and southwest Florida rivers below roughly 29°N latitude; it appears again in the Gulf of Mexico from the Suwannee River, Florida, to Lake Pontchartrain, Louisiana (Jordan 1884; Lee et al. 1980).

Many striped bass in Atlantic Coast rivers from Albemarle Sound, North Carolina, to the St. Lawrence River are migratory as adults. They travel annually from oceanic waters to riverine spawning grounds and back to the ocean, where they undertake a northern summer migration and southward winter migration (Boreman and Lewis 1987). However, recent studies of otolith microchemistry (Morris et al. 2003; Zlokovitz et al. 2003) indicate that striped bass residing in some longer river systems (Roanoke River, North Carolina, and Hudson River, New York, respectively) may exhibit multiple life history strategies, with some individuals remaining year-round in the upper freshwater portion of the system. Additionally, one group of individuals resides in the lower river and upper estuary, another group migrates to the coastal ocean, and a final group exhibits a mid-life habitat shift between freshwater and saltwater environments (Morris et al. 2003; Zlokovitz et al. 2003).

Striped bass populations south of Albemarle Sound, North Carolina, and in the Gulf of Mexico are thought to be endemic to each river system, and are considered essentially non-migratory by most researchers (Vladykov 1947; Scruggs and Fuller 1955; Scruggs 1957; Raney 1957; Murawski 1958; Barkuloo 1970; Dudley et al. 1977; McIlwain 1980; Richkus 1990). However, it might be that past and present management measures used for these stocks have largely precluded most fish from reaching a minimum size for migration (i.e., small size limits and liberal bag limits that, in combination, effectively maintain an artificially young age structure for a species well-documented to live to at least age 30) (W. Laney, personal observation).

Historic and recent recaptures of tagged striped bass suggest that migratory behavior in southeastern stocks is displayed by at least some small percentage of larger individuals. Hess et al. (1999) reported that movement between the adjacent Savannah and Ogeechee rivers in Georgia has occasionally occurred via coastal waters. For example, a striped bass tagged in Alligator Creek (a tributary to the Cape Fear River, North Carolina) on February 18, 2004, was captured by an angler on May 13, 2005, at the mouth of the Cape Cod Canal in Buzzard's Bay, Massachusetts (Mark Westendorf, Coastal Zone Resources, Wilmington, North Carolina, personal communication).

Additionally, two striped bass populations on the Atlantic coast, one in the John H. Kerr Reservoir on the North Carolina/Virginia border and another in the Santee-Cooper Reservoirs in South Carolina, developed upstream spawning migrations to reservoir tributaries after downstream migration was precluded by dam construction (Scruggs and Fuller 1955; Scruggs 1957; Stevens 1958; Jenkins and Burkhead 1993). Additional reproducing freshwater populations have been established in U.S. reservoirs outside the historic range of the species (in California, Oklahoma, Oklahoma/Texas and Utah; see list in Crance 1984).

On the Pacific coast, striped bass were introduced in the San Francisco Bay estuary in 1879 and 1882, and have since spread north to Vancouver Island, British Columbia, and south to Baja California, Mexico (Lee et al. 1980). The species has also been widely stocked in inland

reservoirs and coastal rivers in the United States (Fuller et al. 1999) and abroad (France, Portugal and Russia; see Hill et al. 1989).

The detailed descriptions of striped bass habitats and environmental requirements in this chapter focus on those areas used by the Atlantic coastal migratory stocks under the jurisdiction of the Atlantic States Marine Fisheries Commission (ASMFC) and its member states. The stock is defined as, "...all coastal migratory striped bass stocks on the East Coast of the United States, excluding the Exclusive Economic Zone [EEZ] (3-200 nautical miles offshore), which is managed separately by NOAA Fisheries." Migratory striped bass stocks occur in the riverine, estuarine, and coastal areas of all states and jurisdictions from Maine through North Carolina, as congressionally mandated in the Atlantic Striped Bass Conservation Act (PL 98-613) (Atlantic Striped Bass Plan Development Team 2003). All habitats used by the stock are addressed, including habitats outside ASMFC jurisdiction in the EEZ. Significant environmental, temporal, and spatial factors affecting distribution of striped bass are summarized in Table 9-5. Since tagging studies have documented exchange between migratory striped bass in U.S. and Canadian rivers, cursory descriptions of Canadian striped bass habitats are also included.

Striped bass habitat use information in this document is largely based on material in Bigelow and Schroeder (1953), Hardy (1978), Bain and Bain (1982), Rulifson et al. (1982), Fay et al. (1983), Crance (1984), Richkus (1990), Funderburk et al. (1991), Rago (1992), and Rulifson and Dadswell (1995), but has been supplemented by many other sources, including references from the following anadromous fish and striped bass bibliographies: Street and Hall (1973); Pfuderer et al. (1975); Rogers and Westin (1975); Horseman and Kernehan (1976); Smith and Wells (1977); Westin and Rogers (1978); Setzler et al. (1980); and Bettross (1991).

Striped bass life stages for the purposes of this document are defined as follows (after Hardy 1978). **Eggs** are extruded, fertilized, or unfertilized ova. **Yolk-sac larvae** are newly-hatched individuals that range in length from 2.0 to 3.7 mm (Mansueti 1958a; Fay et al. 1983), with a mean of 3.1 mm (Mansueti 1964) and maximum length of 6.0 to 7.0 mm, prior to yolk absorption. **Larvae** are individuals that have absorbed the yolk sac, but have not yet acquired the minimum adult fin ray complement and assumption of adult body form. Larvae range in size from 5.0 to 36 mm (Pearson 1938; Mansueti 1958a, 1964), and include the finfold and post-finfold larval stages as described by some authors (see Setzler et al. 1980). **Juveniles** range from 36 to approximately 174 mm minimum size for males (Raney 1952) and 432 mm for females (Clark 1968), and have acquired the minimum adult fin ray complement, but have yet to reach sexual maturity. **Adults** are any fish these lengths or larger that have reached sexual maturity.

Habitat Suitability Index Models

The U.S. Fish and Wildlife Service has developed several Habitat Suitability Index (HSI) models for various applications to striped bass stocks (Bain and Bain 1982; Crance 1984). In one instance, Bain and Bain (1982) developed a model for estuarine-associated coastal stocks of striped bass that contains individual components corresponding to the spawning, egg, larval, juvenile, and adult life history stages. The model is intended for use year-round on estuarine-associated striped bass stocks located on the Atlantic, Gulf, and Pacific coasts of the United States. This model can yield one HSI value for the entire life cycle of the striped bass, if all

components are used, or individual HSI values can be generated for each life stage. The model was not designed for evaluation of marine habitat. It is also not applicable to areas where partial or extensive reduction in habitat availability has occurred due to contamination by toxic substances. Habitat parameters required for running the model include: 1) For riverine habitats- percent of natural river discharge, maximum total dissolved solids, average water temperature, minimum dissolved oxygen, and average current velocity; and 2) For estuarine habitats- percent of original salt marsh, percent of original freshwater input to estuary, average water temperature, average salinity, and minimum and average dissolved oxygen. This model assumes that striped bass habitat suitability is primarily associated with water quality (physicochemical conditions) during most life stages (Bain and Bain 1982). However, food availability and water quantity are considered particularly important life requisites in some life stages (W. Laney, personal observation).

The U.S. Fish and Wildlife Service's Charleston Ecological Services Field Office modified the Bain and Bain (1982) striped bass model for use on the Savannah River, Georgia/South Carolina (EuDaly 2002). The HSI was developed specifically to assess the modeled impacts of Savannah Harbor deepening on striped bass spawning, egg, and larval habitats through changes in flow velocity, dissolved oxygen, and salinity concentrations caused by channel modifications (EuDaly 2002; Van Den Avyle et al. 1990; Winger and Lasier 1990; Reinert and Jennings 1998; Will et al. 2000). This modified model may have application potential in tributaries where migratory striped bass spawn closer to the estuary (W. Laney, personal observation). Another model, developed by Crance (1984), applies to riverine or lacustrine habitat of striped bass throughout the 48 conterminous states.   The lacustrine component of the model is generally inapplicable for migratory striped bass. The riverine model applies during the spawning season, and can be used to assess spawning habitat for those populations that spawn in the inland portions of East Coast rivers. The minimum length of river required for riverine reproductive habitat in this model is about 52.6 km. This estimate may not represent the actual minimum river length required if: 1) eggs are not moving at water velocity; 2) water temperature varies from optimal; or 3) the distance required is increased by suspension of the newly-hatched embryo (suspension may be required for about 15 hours post-hatch).

Variables required to run the riverine spawning habitat model include: water temperature; dissolved oxygen concentration; and current velocity (Crance 1984). At the time of its publication, the model had not been field-tested.

Both of the striped bass models developed for the U.S. Fish and Wildlife Service (Bain and Bain (1982) and Crance (1984)) are available on the internet at the U.S. Geological Survey National Wetlands Research Center website: http://www.nwrc.usgs.gov/wdb/pub/ hsi/hsiintro.htm.

An additional document prepared by the Gulf States Marine Fisheries Commission (Lukens 1988) provides a useful example of how striped bass habitat criteria may be used in assessing and prioritizing habitats for striped bass restoration efforts. HSI models should not be considered proven statements, but instead are hypothetical species-habitat relationships (Stier and Crance 1985). Values provided may not be precisely applicable to all areas along the Atlantic Coast. Information pertaining to a particular habitat should be evaluated with regard to model criteria. Despite their limitations, HSI models are useful for evaluating species-habitat relationships (Bilkovic 2000).

Part A. Striped Bass Spawning Habitat

Geographical and temporal patterns of migration

Merriman (1937b) indicated that spawning probably occurred historically in every river of any size in the northeastern United States where proper conditions were present. The present range of migratory striped bass documented as returning regularly from the Atlantic Ocean to coastal rivers to spawn is from the Roanoke and Chowan River tributaries of Albemarle Sound in North Carolina to the St. Lawrence River in Canada (Raney 1952; Bigelow and Schroeder 1953; Leim and Scott 1966; Scott and Crossman 1973; Rulifson et al. 1982; Fay et al. 1983; Hill et al. 1989; Richkus 1990; Rulifson and Dadswell 1995). In general, juveniles migrate downstream in summer and fall, while adults migrate upriver to spawn in spring, later returning downstream to the lower river, estuary, or ocean (Shepherd 2006). Additionally, inland spawning migration extent has been altered by construction of dams that prevent access to some historic spawning habitats (W. Laney, personal observation).

Spawning location (geographical)

Documented U.S. and Canadian spawning ground locations used by Atlantic migratory striped bass can be found on the DVD supplement. The principal spawning areas for migratory striped bass along the Atlantic coast are located in the Chesapeake Bay and its tributary rivers and the Hudson River (Merriman 1941; Raney 1957; Berggren and Lieberman 1978; Kernehan et al. 1981; Setzler-Hamilton and Hall 1991; Wirgin et al. 1993; Richards and Rago 1999). Additional migratory stock spawning habitats located in the Delaware River, Roanoke River, and Canadian Atlantic rivers (Rulifson and Dadswell 1995), are believed to make smaller contributions to coastal fisheries (Richards and Rago 1999). Riverine stocks in North Carolina south of Albemarle Sound (Tar-Pamlico, Neuse, Cape Fear, and Northeast Cape Fear) are believed to make minor, if any, contributions to the coastal migratory stock. However, fish tagged in the Atlantic Ocean have been recaptured in Pamlico Sound during the spring, which suggests that some exchange historically occurred (Holland and Yelverton 1973). As noted by Richards and Rago (1999), however, composition of the coastal stock varies, and is a function of variable reproductive success in given spawning areas, spawning adult year-class strength, and season.

In the southern portion of the range, the Roanoke River's contribution to the coastal migratory stock has historically been a small percentage, with some authors stating the stock was less migratory than others (Hassler et al. 1981; Boreman and Lewis 1987; Haeseker et al. 1996). However, the Roanoke stock was historically fished at a high rate, and fish were harvested at an early age such that from 1956 through 1990, the recruited fish consisted predominantly of individuals aged two and three (NC SBSMB 1991). Few fish from the stock were surviving to an age when migratory behavior would typically be initiated. Current management measures for the stock entail a delayed harvest and lower fishing rate that provide for a broadened age structure. Under this management scheme, the percentage of migratory fish is likely to increase. The Roanoke River-Ablemarle Sound stock was declared recovered by the ASMFC in 1997 (ASFMC 1998).

Farther north, the Chesapeake Bay tributaries are thought to be the most productive spawning grounds (Merriman 1941), and have contributed as much as 90% of Atlantic coastal landings (Berggren and Lieberman 1978; Van Winkle et al. 1988). In fact, Chesapeake Bay fish make a major contribution to the fishery in the lower Hudson River and New York Bight (Berggren and Lieberman 1978). Spawning habitats in Virginia tributaries to Chesapeake Bay were documented by Tresselt (1950, 1952), Mansueti (1961b), Rinaldo (1971), McGovern and Olney (1988, 1996), Grant and Olney (1991), Olney et al. (1991), and Bilkovic et al. (2002). The only direct observations of striped bass eggs and larvae in major Virginia rivers through 1991 were made by Tresselt (1952), Rinaldo (1971), McGovern and Olney (1988), and Grant and Olney (1991).

Tresselt (1952) surveyed the Pamunkey, Mattaponi, Chickahominy, James, and Rappahannock rivers in Virginia, to determine the location of striped bass spawning grounds. Eggs were collected in appreciable numbers only on the Mattaponi River. In the Pamunkey, Mattaponi, and Chickahominy rivers, the regions of greatest egg abundance coincided with the regions of largest commercial catch. These areas were located in the first 25 miles of freshwater, and usually had high turbidity during the spawning season. The largest numbers of eggs were located 27 km above the mouth of the Pamunkey and 14 km above the mouth of the Mattaponi. Only a few eggs were collected over a wide section of the James and Rappahannock rivers (Tresselt 1952).

Similarly, Mansueti (1961b) depicted the following Virginia rivers as spawning habitat: James, Chickahominy, Pamunkey, Mattaponi, Rappahannock, and Potomac. Surveys of spawning grounds on the Chickahominy and James rivers during 1950 were conducted late, but provided the first direct documentation of striped bass spawning in those systems (Grant and Olney 1991). Additionally, Rinaldo (1971) surveyed the Pamunkey River, Virginia, during the 1966 spawning season, and determined that spawning occurred 8 to 48 km above West Point. Olney et al. (1991) documented striped bass egg mortality, production, and female biomass in Virginia rivers from 1980 to 1989. Sampling was conducted in the James, Pamunkey, Mattaponi, and Rappahannock rivers during April and May. In the Pamunkey River, eggs were collected from river kilometers (rkm) 62 to 72 and 58 to 66 in April 1987. The Pamunkey River was also sampled during 1980, 1983 to 1985, 1988, and 1989, presumably within reach 45.6 to 88.1 rkm (Olney et al. 1991).

Kernehan et al. (1981) suggested that previous, inadequate sampling underestimated the importance of the Upper Chesapeake Bay as striped bass spawning grounds. Phillips (1990) and Mansueti (1961b) identified the following Upper Chesapeake Bay spawning habitats: Potomac River, Patuxent River, Susquehanna River, Northeast River, Elk River, Chesapeake and Delaware (C&D) Canal, Bohemia River, Sassafras River, Chester River, Choptank River, Blackwater River, Honga River/Fishing Bay, Nanticoke River, Wicomico River, Monokin River, and Pocomoke River.

The Susquehanna River was historically the area of greatest egg production, and spawning was recorded as far upriver as Northumberland, Pennsylvania, or beyond (Baird 1855; Dovel 1971). However, following construction of the Conowingo Dam near the mouth (river km 16.1) of the Susquehanna River in 1928, the principal area of egg production appeared to be the main channel of Chesapeake Bay between Western Point and Chesapeake City (Dovel 1971). In the Potomac River, spawning historically occurred as far upriver as Great Falls (Baird 1855;

Hildebrand and Schroeder 1928; Shannon and Smith 1968), but in 1978 was limited to Whitestone Point and below (Nichols and Miller 1967).

In the 1960's and 1970's, major spawning activity centered in the C&D Canal (Beitch and Hoffman 1962; Johnson 1972), which led some researchers around that time to state that this canal was the most important mid-Atlantic region spawning area (Hollis 1967; Dovel 1971; Dovel and Edmunds 1971; Warsh 1977).   However, based on total eggs spawned in an area, Kernehan et al. (1981) demonstrated that from 1973 to 1977, the Upper Chesapeake Bay from Turkey Point southeast to Worton Point was far more important to spawning than the relatively small C&D Canal.

For much of the 20th century, the Delaware River exhibited poor water quality and striped bass production was low (Chittenden 1971a). Murawski (1969a) reported larvae over a distance of 108 km in the Delaware, but this distance included a 45 km void in the vicinity of Philadelphia. With improvements in riverine water quality, the Delaware River began to contribute striped bass to the coastal migratory stock (Albert 1988; USDOI and USDOC 1994), and the stock was declared recovered (ASMFC 1998).

In some years, the Hudson River contributes a significant proportion of the coastal stock (Fabrizio 1987; Van Winkle et al. 1988). The Hudson River's primary contribution to the stock occurs north and east of the river (Waldman et al. 1990; Dorazio et al. 1994). Spawning occurs in the fresh-brackish reach of the river and is concentrated between rkm 54 and 88 (Boreman 1981).

In New England, spawning was historically documented in the Thames River, Connecticut (Maltezos 1960), and ripe females were taken in the Mouson River, Maine (Towne 1940). Spawning may have occurred in the past in the Connecticut River (Merriman 1937a), but at least several decades ago no spawning was evident (Whitworth et al. 1968; Thomson et al. 1978), despite the fact that adult fish annually entered the river (Talbot 1966; Whitworth et al. 1968). However, Hardy (1978) reported (based on Neville (1939) and Raney (1952)) that there was no evidence of successful spawning in coastal areas of New Jersey, or in the rivers of New England. Currently, striped bass are apparently spawning in the Kennebec River, since the Maine Division of Marine Fisheries is catching juveniles there on a regular basis (Lew Flagg, Maine Department of Natural Resources, Division of Marine Fisheries, personal communication).

Although there are no documented striped bass spawning runs in the Connecticut River, juvenile striped bass are occasionally taken in the lower river (downstream of river km 12) by electro-shock and beach seine. For the first time in 2004, eight to ten striped bass juveniles were taken in the Connecticut River by electro-fishing during July from above the salt wedge (upstream of river km 40). Although the exact origin of these striped bass juveniles could not be determined with confidence, the juvenile striped bass occasionally taken in the lower river (downstream of river km 12) are believed to have originated from the Hudson River stock. The Hudson River juvenile striped bass survey conducted annually by the New York Department of Environmental Conservation often captures juvenile striped bass as far east as Orient Point, New York (some 100 miles east of the Hudson River). However, the recent capture of juvenile striped bass from the upper Connecticut River (above river km 40) in 2004 probably resulted from limited striped bass spawning in the Connecticut River. Each spring (April to June), there are thousands of adult striped bass in the upper river (above river km 50) that use it as a primary

feeding area for pre-spawned American shad and blueback herring. Large (greater than 80 cm) female striped bass are often sampled in the river during the spring, but a ripe female has not been documented in the upper river. Furthermore, during the juvenile alosine beach seine surveys from July through October (1976-2008), researchers have yet to capture a single juvenile striped bass. Given the record size of the current Atlantic coast striped bass stock, limited and occasional spawning in the Connecticut River would be expected. At this time, ecologists regard the Connecticut River as primarily an important spring feeding area for Atlantic coast striped bass, but not a primary spawning area (V. Crecco, Connecticut Bureau of Marine Fisheries, personal communication).

Farthest north, spawning in Canada was historically believed to occur in the Miramichi and Saint John rivers, New Brunswick, Annapolis and Shubenacadie rivers, Nova Scotia, and St. Lawrence River, Quebec (Leim 1924; Leim and Scott 1966; Scott and Crossman 1973). Rulifson and Dadswell (1995) reported that ten Canadian rivers were known or believed to sustain spawning striped bass populations, including:: the St. Lawrence River (where the spawning stock was stated to perhaps be extirpated); the Nepisiguit River in Chaleur Bay; the Tabusintac, Miramichi, Kouchibouguac, and Richibucto rivers in the western Gulf of St. Lawrence; the Saint John, its tributary the Kennebecasis, and the Annapolis rivers in the outer Bay of Fundy; and the Shubenacadie-Stewiacke river system in the inner Bay of Fundy.

Striped bass spawning in Canadian rivers is stated to occur in tidal streams a few weeks after ice leaves the system, and occurs near the head of tide (Rulifson and Dadswell 1995). In Bay of Fundy rivers, spawning is near, or a relatively short distance above, the head of tide. In the Saint John River, spawning occurs in tributaries of Belleisle Bay, approximately 64 km above Reversing Falls (Dadswell 1976). Historically, the main spawning area was at the head of tide around Hart Island above Fredericton, about 65 km upriver from the limit of saltwater excursion (Adams 1873), but it is thought spawning no longer occurs at this site (Jessop 1990).

The Shubenacadie-Stewiacke striped bass population is the only one documented as successfully reproducing in a tidal bore river. The Annapolis River population may also have historically spawned in a tidal bore river, but the tidal bore phenomenon was eliminated by construction of a causeway (Rulifson and Dadswell 1995). In western Gulf of St. Lawrence rivers, spawning occurs just above the head of tide (Hogans and Melvin 1984; Madden 1984). In the St. Lawrence River, spawning is believed to have occurred at, and upstream of, Trois Rivieres (Rulifson and Dadswell 1995). Other possible spawning grounds are alluded to in the literature (Vladykov 1946, 1947; Beaulieu 1962, as cited in Rulifson and Dadswell 1995), but no study of spawning habitats was ever conducted (Magnin and Beaulieu 1967, as cited in Rulifson and Dadswell 1995).

Spawning location (ecological)

There are a number of key components of striped bass spawning habitats necessary to retain functionality and remain hospitable for striped bass adult use, and egg and larval production and survival. These components include: appropriate flow regimes at various temporal scales, including suitable spring attractant flows for stocks migrating to inland spawning grounds and suitable flows during the spawning season; appropriate temperature regimes; appropriate dissolved oxygen levels; absence of adverse levels of turbidity, pH, and contaminants; and suitable prey resources for larvae (W. Laney, personal observation). Setzler et

al. (1980) indicated that maintenance of adequate spawning areas with good water quality is the most critical necessity for continued survival of striped bass.

Migratory striped bass mostly spawn in groups in freshwater near the heads of Atlantic coast estuaries, or far inland up major tributaries, depending upon the estuary. Hardy (1978) summarized the general characteristics of spawning habitats used by striped bass. Spawning occurs in fresh, turbid waters in relatively shallow reaches of rivers, streams, and creeks (0.3 to 6.1 m) (Abbott 1878; Tresselt 1950; Murawski 1969b). Some populations spawn in the upper tidal freshwater portions of rivers (Raney 1952, 1956; Tresselt 1952; Humphries 1966; Talbot 1966) in areas just above tidal influence (Bigelow and Schroeder 1953), or hundreds of kilometers inland in turbulent, turbid rapids (Raney 1954; McCoy 1959). These latter areas are frequently associated with the Fall Zone (the relatively narrow belt between the Coastal Plain and Piedmont provinces, where elevation changes more rapidly), and are characterized by boulders and strong currents (Norney 1882; Raney 1952; McCoy 1959; Mansueti and Hollis 1963; Talbot 1966). Stocks in estuarine systems lacking pronounced tidal cycles tend to travel further upstream to spawn (Bain and Bain 1982). Striped bass have never been documented to spawn in lakes, within reservoirs, or in the sea (Goode et al. 1884; Bigelow and Schroeder 1953).

Striped bass spawning runs begin earlier in the southern end of the range, and occur progressively later as the season advances and water temperatures warm (Raney 1952; Bigelow and Schroeder 1953; Leim and Scott 1966; Scott and Crossman 1973; Bain and Bain 1982; Rulifson et al. 1982; Fay et al. 1983; Crance 1984; Hill et al. 1989; Richkus 1990). Pre-spawning aggregations arrive in the Chesapeake Bay during January, February, and March (Dovel 1968). However, nearly all spawning activities in the mid-Atlantic region occur in April, May, and June (Fay et al. 1983). Striped bass appear on spawning grounds in the Cape Fear River, North Carolina, from mid-April to mid-May (Sholar 1977; Fischer 1980). Other North Carolina river striped bass spawning seasons are: April to early May in the Northeast Cape Fear River (Sholar 1977); April to mid-May, or late March to late May, in the Neuse River (Baker 1968; Hawkins 1979); mid-April to mid-May in the Tar-Pamlico River, with a peak of May 3-11 (Humphries 1966); and April 15 to May or June in the Roanoke River (Chapoton and Sykes 1961; Shannon and Smith 1968; Shannon 1970; Street 1975). Spawning runs on the Roanoke River begin when water temperatures reach 7 or 8°C, typically during March, and terminate at the spawning grounds near Weldon around April 1-15 (Merriman 1941; Dickson 1958; Fish and McCoy 1959; NC WRC 1962). Peak spawning on the Roanoke River was reported by Hill et al. (1989) to be May 10-20.

Temporal periods of striped bass spawning are similar throughout Chesapeake Bay (Grant and Olney 1991). Spawning periods for Virginia rivers were reported by Grant and Olney (1991), McGovern and Olney (1996), and Bilkovic et al. (2002). Peaks in spawning were generally sharp and of limited duration. In the York River tributaries (Mattaponi and Pamunkey), peaks occurred in the fourth week of April in both years surveyed (1980 and 1983), and in the Rappahannock River in 1983. In 1982, the peak spawning in the Rappahannock occurred one week earlier. Spawning in the James River was later, peaking the first week in May in both 1981 and 1983, which is also typical in the Potomac (Setzler-Hamilton et al. 1981) and in the upper Chesapeake Bay and the C&D Canal (Johnson and Koo 1975; Kernehan et al. 1981).

The spawning season in the Potomac River was reported as mid-April to mid-June, with a peak from April 23 through May 8 (Baird 1855; Setzler-Hamilton et al. 1981). The spawning

season in the Chesapeake Bay was reported as April, May, and early June (Chapoton and Sykes 1961; Dovel 1971). Similarly, in the Chesapeake and Delaware Canal, spawning occurred from mid-April to mid-June, with a peak from April 20 to May 10 (Kernehan et al. 1981). Spawning in the Delaware River was reported as occurring from late May to mid-June, with a peak in June (Raney 1952). In the Hudson River, the spawning period was reported as mid-May to mid-June, with peak activity in the last two weeks of May (Raney 1952; Rathjen and Miller 1957; Boreman and Klauda 1988).

In Canadian populations, including Bay of Fundy rivers, spawning occurs in May and June (Leim and Scott 1966; Scott and Crossman 1973; Meadows 1991; Rulifson and Dadswell 1995). In the Stewiacke, spawning begins in the fourth week of May, and depending upon the weather, continues until about June 20 (Meadows 1991). In the Annapolis River, spawning begins in late May and continues through June. Spawning in the Saint John River occurred in May, beginning May 13th and terminating May 20th (Dadswell 1976).

The exact timing of spawning activity in western Gulf of St. Lawrence rivers is not well documented (Rulifson and Dadswell 1995). Spawning may occur shortly after ice leaves the rivers. Overwintering fish migrate downstream and spawn in May and early June (Vladykov and Brousseau 1957; Hogans and Melvin 1984; Meagher et al. 1987). Spawning in the Kouchibouguac River lasts about three days (Hogans and Melvin 1984). In the Tabusintac River, local fishermen report that spawning occurs in the late summer-early fall, because large adults in reproductive condition are caught during that period. If the fishermen are correct, this would represent the only known fall-spawning population. More information is needed on the Tabusintac population to determine whether these fish are fall spawners, or are just approaching maturity for overwintering (Rulifson and Dadswell 1995).

Maturation and spawning periodicity

In U.S. rivers, males precede females to the spawning grounds in the spring, while females remain offshore or in downstream estuaries until shortly before spawning (Vladykov and Wallace 1952; Trent and Hassler 1968; Holland and Yelverton 1973). After the females arrive on the spawning grounds, characteristic mating behavior consists of a single female surrounded by up to 50 males, at or near the surface (Setzler et al. 1980). Eggs are broadcast loosely in the water, and normal spawning duration for a single female is less than four hours (Lewis and Bonner 1966). Based on the behavior of radio-tagged females in the Choptank and Nanticoke Rivers, Maryland, Hocutt et al. (1990) think that the brackish estuary downstream of spawning habitat is more important than previously recognized for females.

 Striped bass appear to be repeat spawners (iteroparous) throughout their migratory range. Raney (1952) reported that striped bass spawn more than once, but not necessarily every year. Hocutt et al. (1990) reported homing of radio-tagged females to the Nanticoke River, Maryland, and believed this constituted strong evidence for annual spawning, as well as strong evidence of natal river fidelity by females.

Studies of mitochondrial DNA (mtDNA) suggest that there is higher female than male fidelity to the natal spawning grounds, at least for Chesapeake Bay populations (Chapman 1987, 1989). Chapman (1989) drew the following conclusions for Chesapeake Bay striped bass: 1) distinct matriarchal groups occurred on spawning grounds of the Choptank River, Potomac

River, and Upper Chesapeake Bay; 2) after age two, mixed aggregations of males and females formed during winter, probably derived from populations not surveyed in 1984; 3) males from the mixed aggregations appeared to have dispersed randomly to spawn in 1986, which suggested that males did not have a strong homing instinct; and 4) females appeared to return to their natal areas, as their mtDNA frequencies in 1987 matched closely the 1984 distributions. The conclusion presumes that striped bass tend to remain in their natal areas until age two (Chapman 1989).

Based on egg collections, the diel timing of striped bass spawning activity appears to be variable among and between systems, and little specific information was found for many of the systems used for spawning. Spawning activity has been noted in late afternoon and early evening (Morgan and Gerlach 1950; NC WRC 1962), as well as late evening and early morning (Hardy 1978). Extensive studies of the vertical, horizontal, and temporal distribution of eggs during the spawning season were conducted in the Roanoke River, North Carolina, by McCoy (1959) and Cheek (1961). McCoy (1959) did not detect a statistically significant daily pattern of egg deposition, although there did appear to be some trends in the adjusted egg data showing higher deposition in the evening (22:00) during mid-May and in the early morning (06:00) during late May. Sampling conducted by Rulifson (1989, 1992) on the Roanoke River suggested that egg deposition occurred more frequently near dusk. Similarly, eggs were taken in the Susquehanna River early in the night (Pearson 1938).

Spawning and the saltwater interface

Salinity and total dissolved solid (TDS) concentrations are thought to be important factors during the striped bass spawning period, and may be responsible for deterring spawning or reducing spawning success (Bain and Bain 1982). In the naturalized population of the Sacramento-San Joaquin River, California, the number of eggs deposited reached a maximum when salinity was less than 0.18 ppt (Farley 1966), and spawning migrations did not occur at a critical salinity concentration of 0.35 ppt (Radtke and Turner 1967). Highly successful spawning was observed in the C&D Canal at salinities of 0.70 to 1.5 ppt (Johnson and Koo 1975). Additionally, Stevens (1979) reported that striped bass might not spawn where salinities exceed 5 ppt.

Salinities in some Canadian spawning sites are reported by Rulifson and Dadswell (1995). In the Bay of Fundy tributaries, the Shubenacadie and Stewiacke rivers, salinities during spawning in 1992 and 1994 ranged from 0.0 to 22.8 ppt. Spawning areas in the Annapolis and Saint John rivers were both located upstream of the salt wedge, and therefore presumably in freshwater (Rulifson and Dadswell 1995).

Spawning substrate associations

Rulifson and Dadswell (1995) reported the bottom composition of some spawning habitats in Canadian rivers. Tidal spawning areas in the Stewiacke River have silty bottoms, and at low tide are lined by mud and sand flats (Rulifson et al. 1987). Prior to the operation of the Annapolis Tidal Generating Station, the primary spawning area above Bridgetown (km 32 through 40, measured from the Annapolis River causeway), Nova Scotia, was characterized primarily by sand interspersed between basalt and granite rocks and boulders (Williams et al.

1984). In the Kennebecasis River, potential spawning habitats include a gravel substrate (Hooper 1967; Melvin 1991). The Kouchibouguac River spawning habitat substrate consists of cobble-sized shale rubble covered by a layer of mud, with eelgrass (*Zostera marina*) as the dominant submerged vegetation (Hogens and Melvin 1984).

Spawning depth associations

Limited information is available regarding the depth of striped bass spawning habitats. Striped bass spawning occurs at, or near, the water surface in some Atlantic coast rivers (Merriman 1941; Raney 1952). In the Shubenacadie and Stewiacke rivers in Canada, historic and present spawning habitats are tidally influenced. At high tide, the areas are deep and relatively wide. At low tide, both areas become narrow, shallow channels (Rulifson et al. 1987). Former spawning areas in the Annapolis River, Nova Scotia, were approximately 30 m wide and consistently 1.5 to 2 m deep. Western Gulf of St. Lawrence spawning habitats also are tidally influenced (Williams et al. 1984).

Spawning water temperature

Water temperature is a key variable influencing the activities of striped bass adults prior to, and during, spawning migrations (Bain and Bain 1982; Crance 1984). Spawning generally occurs in water temperatures ranging from 10.0 to 25.0°C (Nichols 1966; Hardy 1978; Merriman 1941). Spawning peaks are apparently triggered by a noticeable increase in water temperature, generally beginning at temperatures of at least 14°C (Fay et al. 1983). Mature adults usually initiate spawning runs when temperatures reach 14.4°C, exhibit peak activity from 15.8 to 19.4°C, and cease spawning at 20 to 25°C (W. Laney, personal observation). Other temperature extremes reported for spawning were a low of 10°C (IEM 1973) and a high of 26.5°C (Combs 1979).

Dickson (1958) reported that striped bass spawning on the Roanoke River, North Carolina, usually began after temperatures reached 15°C. Optimal spawning temperatures for the Roanoke River were reported as approximately 17 to 19°C, with no spawning observed below 12.8°C or above 22°C (Shannon and Smith 1968). McCoy (1959) reported a minimum spawning temperature of about 15°C and a maximum of about 22°C for the Roanoke River.

In Chesapeake Bay, the striped bass spawning water temperature range was from 10.4 to 23.9°C. However, most spawning occurred between 14.4 and 21.1°C, with peak activity from 17.8 to 20.0°C (based on egg presence) (Raney 1952; Sheridan et al. 1961; Hollis 1967; Rinaldo 1971). Peak spawning activity was observed to follow a rise of 3.5°C (Tiller 1955). Kernehan et al. (1981) collected striped bass eggs in the vicinity of the C&D Canal in temperatures ranging from 8.4 to 29.0°C, but the researchers noted that most larvae produced in the area resulted from intensive spawning in water with temperatures from 13.5 to 18.0°C. Peak egg densities in Virginia tributaries to Chesapeake Bay were limited to rapidly rising water temperatures in the range 13.7 to 19.5°C, with eggs found in a wide range of 8.0 to 21.2°C. Eggs were nearly always in freshwater (Grant and Olney 1991).

Spawning temperatures have been documented for striped bass in most of the Canadian rivers where they are present. In Canadian rivers entering the Bay of Fundy, spawning is

initiated when temperatures reach 11 to 11.5°C, and ceases above 22°C (Rulifson and Dadswell 1995). In the Shubenacadie-Stewiacke system, males in spawning condition were found at 14°C, and ripe females were common at 16°C. In 1992, eggs were present at temperatures from 15.5 to 22.0°C (Rulifson and Dadswell 1995). In 1994, major spawning activity occurred when water temperatures reached 18°C (K. Tull and R. A. Rulifson, East Carolina University, unpublished data). Spawning in the Annapolis River occurred at water temperatures from 15 to 24.4°C (Williams 1978; Parker and Doe 1981; Williams et al. 1984). Spawning in the Saint John River began at about 11.5°C, and maximum activity was observed at 13.5°C (Dadswell 1976). In western Gulf of St. Lawrence rivers, spawning temperatures were: Kouchibouguac River, 12 to 14.5°C, with three-day duration (Hogans and Melvin 1984); Miramichi River, spawning condition individuals of both sexes present at 12 to 14°C; Richibucto River, both sexes ripe at 16 to 16.5°C (Rulifson et al. 1987).

Initiation and duration of spawning are both temperature-dependent (Calhoun et al. 1950; Rathjen and Miller 1957), and sudden drops in temperature as a result of the passage of cold fronts, or flood-control or hydropower operations, may interrupt spawning in U.S. rivers (Calhoun et al. 1950; Chadwick 1958; Mansueti and Hollis 1963; Farley 1966; Combs 1979; Hawkins 1979). In contrast to this pattern, in the Annapolis River, Nova Scotia, peak egg production was observed after temperature drops (Parker and Doe 1981); however, rapid temperature drops to 15 or 16°C resulted in temporary cessation of spawning (Williams 1978). Spawning generally occurs on rising temperatures (Neal 1967, 1971). Depending on the estuary, there may be one to three peaks in spawning each season. Such peaks are thought to result from major increases in temperature (Hardy 1978).

Spawning dissolved oxygen associations

Dissolved oxygen concentrations greater than 5 mg/L are recommended for all life stages of striped bass (USEPA 1976; Setzler-Hamilton and Hall 1991). Given that spawning adults are present in the spring of the year when river temperatures are usually lower, oxygen concentration is not generally a limiting factor. However, historically, striped bass spawning areas in the Delaware River were eliminated due to low oxygen concentrations. Collections of fish throughout the freshwater sections of the Delaware River from 1963 to 1966 contained no striped bass. Gross pollution of the tidal freshwater zone of the river destroyed its utility as a spawning area, and resulted in the extirpation of the striped bass from the tidal fresh and freshwater portions of the river. Restoration of striped bass was deemed possible if pollution was decreased so that the tidal freshwater portion of the river was functionally restored (Chittenden 1971a). Such restoration did in fact occur, and the striped bass once again spawns in the Delaware River (W. Laney, personal observation).

Spawning and water velocity/flow

Water flow discharge and timing in striped bass spawning rivers are significant factors determining spawning habitat suitability (Fish and McCoy 1959; Turner and Chadwick 1972; Mihursky et al. 1981). Some authors note that the suitability of a spawning area appears to increase with greater river discharge (expressed as the percent of natural flow). Consequently, Fish and McCoy (1959) found that a sustained minimum flow was necessary for suitable

spawning conditions in the Roanoke River, North Carolina, and that rapid fluctuations in stream flow were detrimental to spawning.

Differences in the area and spatial extent of striped bass spawning habitat can occur in years of drought (Grant and Olney 1991). Data from the James River, Virginia, contrasted a year of severe drought (1981) with one of near-average rainfall (1983). Estimated discharge from the James River system into Chesapeake Bay during 1981 (March-May) averaged only 69,000 cfs, compared with 180,000 cfs in 1983. The peak egg production zone was displaced 15 km upriver in 1981, upstream of advancing saltwater, whereas inter-annual differences in the location of peak spawning in other river systems where drought was not a factor were not significant (less than 2 km) (Grant and Olney 1991).

Spawning suspended solid associations

Total dissolved solid (TDS) concentrations above 350 mg/L are reported to have blocked striped bass spawning runs (Radtke and Turner 1967).

Spawning feeding behavior

Spawning striped bass are reported to eat little or nothing, but the fasting process is thought to be brief (Raney 1952; Trent and Hassler 1968). Fish are reported to refrain from feeding only immediately before and during spawning (Morgan and Gerlach 1950; Hollis 1952).

Part B. Striped Bass Egg and Larval Habitat

Survival of striped bass eggs to hatching is primarily associated with relatively narrow tolerances to certain physicochemical factors, including temperature, dissolved oxygen, and current velocity. Development rates of striped bass egg and larval stages are temperature-dependent, within the range of temperatures at which the stages remain viable. Appropriate dissolved oxygen levels and current velocities are also required to maintain viability and keep egg and early larval stages in suspension (Cooper and Polgar 1981).

Survival of the striped bass larval stage is considered to be most crucial for future population abundance of mid-Atlantic striped bass stocks (Fay et al. 1983). Survival rate of larvae, in combination with environmental conditions during early life stages, probably determines the occurrence of occasional dominant year classes so evident in striped bass populations. Given the importance of the larval survival rate to the production of dominant year classes, larval habitats are especially important for sustainability of striped bass populations from individual spawning rivers (Bain and Bain 1982).

Geographical and temporal movement patterns

The habitats occupied by eggs and larvae overlap the spawning areas to a great degree, with larvae occurring further downstream than eggs. Eggs generally hatch in one to three days from fertilization, depending upon temperature (see below). Eggs are transported downstream after extrusion and fertilization, hatch as fry, and subsequently develop into post-larval and juvenile stages. In some river systems, transition from the post-larval to juvenile stage occurs in, or near, the river delta at the head of the adjacent estuary (Rulifson 1984; Rulifson et al. 1992a, 1992b).

The larval yolk-sac phase lasts three to nine days, depending upon water temperature (see below; Albrecht 1964; Eldridge et al. 1977; Rogers et al. 1977). The remaining larval development is variously reported as requiring 22 to 65 days (including an 11-day finfold stage) (Polgar et al. 1976; Rogers et al. 1977), or 35 to 50 days (Bain and Bain 1982). As summer progresses, larger larvae move downstream and by autumn some individuals have reached the mouths of estuaries (Mansueti 1954; Hassler 1958). Striped bass larvae rapidly become very motile, positively phototaxic, and continuously self-suspended (Doroshev 1970). Larvae hatched in relative proximity to estuarine nursery areas characteristically remain in the open surface waters of the estuary (Raney 1952).

In the Chesapeake Bay region, larval nursery areas are the same as the spawning areas (Rinaldo 1971). Larvae are found in both fresh and brackish waters, often in association with white perch (*Morone americana*), although the two species do not spawn in the same locations (Flemer et al. 1968). In the Delaware River, larvae have been recorded within a 103 to 107 km reach, but are found primarily in the first 13 km above Delaware Bay (Murawski 1969a).

Eggs, larvae, and the saltwater interface

Bain and Bain (1982) stated that salinity did not appear to be an important determinant of striped bass egg survival because salinities typically encountered by eggs are not detrimental to survival. However, low salinity is considered optimal for water hardening (Albrecht 1964; Morgan et al. 1981).

Eggs have been found at salinities of up to 12.0 ppt (Tresselt 1950; Hollis 1967; Bason 1971; Dovel 1971). Hollis (1967) found that larvae hatched from eggs at 4.7 to 9.7 ppt survived. Under experimental conditions, survival decreased at salinities above 4.74 ppt (Johnson 1972). Salinity tolerance of striped bass eggs has been reported as 0 to 10 ppt (Mansueti 1958a), 0 to 9 ppt (Albrecht 1964), and 0 to 8 ppt (Morgan and Rasin 1973). Optimum salinities for egg development were reported as 1.5 to 3.0 ppt (Mansueti 1958a) and 1.7 ppt (Albrecht 1964). Maximum survival occurs at 0.900 to 0.948 ppt (Albrecht 1964; Talbot 1966). In addition, development will proceed at 20 ppt, but larvae hatched die within 48 hours (Doroshev 1970).

Larvae have been documented present from 0.0 to 32.0 ppt (de Sylva et al. 1962; Albrecht 1964; Regan et al. 1968; Doroshev 1970; Dovel 1971; Rinaldo 1971; Rogers and Westin 1978). However, greatest density was found at salinity levels less than 2.0 ppt (Dovel 1971), and highest survival occurred up to 10 ppt (Doroshev 1970). Optimal salinities for yolk-sac and post-yolk-sac larvae were reported as 5 to 15 and 5 to 25 ppt, respectively (Rogers and Westin 1978). Optimal range for growth and survival as reported by Lal et al. (1977) was 3 to 7 ppt. Optimal salinities for various-aged striped bass larvae were also reported as follows: 1 to 6 day-old larvae, 3.4 ppt; 7 to 13 day-old larvae, 6.7 ppt; 14 to 20 day-old larvae, 13.5 ppt; 21 to 29 day-old larvae, 20.2 ppt; and 30 to 35 day-old larvae, 33.7 ppt (Lal et al. 1977).

In addition, salinity interactions with temperature affect egg and larval striped bass. Morgan et al. (1981) reported that a temperature-salinity interaction affected percent hatch of eggs and percent survival of newly hatched striped bass larvae, but not larval length at 24 hr of age. Equations were presented for percent hatch and percent survival as functions of temperature ("T", in °C) and salinity ("S", in ppt):

Percent hatch = $-0.83T^2 + 30.64T - 0.12 (S \times T) + (2.22 \times S) - 205.8$

Percent survival = $-1.03T^2 + 35.86T + (0.54 \times S) - 246.63$

The optimal temperature-salinity combination for percent survival was given as 10 ppt at 18°C (Morgan et al. 1981).

Egg and larval substrate associations

Stevens (1966b) suggested that in the absence of current, partially developed eggs or larvae perhaps required sandy or rocky areas with highly oxygenated water in order to escape suffocation and survive. Additionally, Bayless (1968) observed the following percent hatch rates on various substrates: coarse sand 35.7%; plastic 36.4%; silt 13.1%; silty clay 3.2%; and muck-detritus 0.0%.

Egg and larval depth associations

Striped bass eggs are deposited near the surface during spawning activity (Raney 1952). They are buoyant (Mansueti 1958a) or semi-buoyant (Merriman 1937a; Raney 1958), and are found at varying levels within the water column from the surface to the bottom. Egg distribution in the water column appears to be random at velocities in excess of 30 cm/s (Bain and Bain 1982). Yolk-sac larvae either lie horizontally on the bottom (Rinaldo 1971), or perpendicular in the water column with their heads toward the surface (Mansueti 1958a). They may even attempt to swim to the surface, dropping back to the bottom between efforts (Pearson 1938; Dickson 1958; Mansueti 1958a). At one to two days of age, larvae stay near the surface, sometimes attached to floating objects (Mansueti 1958a). By day three, they exhibit continuous swimming ability (Tatum et al. 1966; Doroshev 1970). At about four to five days, yolk-sac larvae are able to swim horizontally, exhibit positive phototaxis, and form schools in laboratory aquaria (McGill 1967). At lengths of 5.5 to 5.8 mm, larvae remain suspended in the water column, never sinking to the bottom (Sandoz and Johnston 1966; Doroshev 1970).

There are indications of a general dispersal of striped bass larvae toward the bottom as feeding begins (Rathjen and Miller 1957; Mansueti 1958a). Two weeks after hatching, larvae forage at the bottom (Rathjen and Miller 1957; Mansueti 1958a), sometimes settling over silt and mud (Hassler 1958). When individuals are about 12 mm long, schools move shoreward and remain in the shore zone throughout the first summer (Raney 1958; Nichols 1966).

Extensive information regarding the horizontal and vertical distribution of striped bass egg and larval stages (yolk-sac, finfold, and post-finfold) was provided for the Potomac River Estuary and the Chesapeake and Delaware Canal, respectively, by Setzler-Hamilton et al. (1981) and Kernehan et al. (1981). Generally, larval stages remained in, or near, the area spawned, although an apparent change in location occurred upstream in the Potomac Estuary, despite a new downstream flow of water (Setzler-Hamilton et al. 1981). The mechanisms proposed to explain this observation (Polgar et al. 1976) were: 1) the active spawning stock continually migrated upstream over time, and therefore, samples indicated an "upstream movement" of larvae; or 2) there was a differential (higher) mortality of early-spawned versus late-spawned larvae. In the C&D Canal, post yolk-sac stages tended to be mid-channel oriented and in highest concentrations near the river or estuary bottom (Kernehan et al. 1981).

Egg and larval water temperature

There is some discrepancy over temperature tolerance for striped bass eggs. Table 9-1 reviews the various findings of researchers on the topic. Morgan and Rasin (1973) and Rogers et al. (1977) indicated that egg survival rapidly declines as water temperature approaches 23°C, and gradually declines as temperature drops below 17°C.

Characterization	Temperature Range (°C)	Citation
Present	8.0 to 25.0	Dovel (1971)
Tolerance	14 to 23	Mansueti (1958a)

Characterization	Temperature Range (°C)	Citation
Optimal	17 to 20	Barkuloo (1970); Doroshev (1970); Morgan et al. (1981); Bain and Bain (1982); Fay et al. (1983)
Optimal	18 to 21	Rogers et al. (1977)
Optimal	19.9 to 20.5	Albrecht (1964)
Maximum	22.2	Barkuloo (1970)
Maximum	21.1	Stevens and Fuller (1965)
Maximum	23	Shannon and Smith (1968)
Maximum	24	Morgan and Rasin (1973)
Minimum	12.8	Albrecht (1964)
Minimum	12	Shannon and Smith (1968); Morgan and Rasin (1973)

Table 9-1. Water temperature tolerance ranges for striped bass eggs

The egg life stage is brief in comparison to other striped bass life stages (Bain and Bain 1982). In general, lower temperatures lead to longer incubation periods (Hardy 1978). Several authors documented hatching at approximately 48 hours after fertilization at a temperature of 18°C (Bain and Bain 1982). In other studies, hatching time varied from 29 hr at 22°C to 80 hr at 11°C (Pearson 1938; Raney 1952; Mansueti 1958a; Hardy 1978).

Two equations have been reported for calculating hatching time of striped bass eggs (Polgar et al. 1976; Rogers et al. 1977). Polgar et al. (1976) gave the following equation based on their observations in the Potomac River: $I = -4.6 T + 131.6$, where I is incubation time in hours and T is incubation temperature in degrees Celsius. However, Rogers et al. (1977) began their analysis of hatching time with a regression equation: $Log_{10}length = bx + a$. Then they gave this equation for time to hatching as a function of temperature: $T_h = ae^b$, where T_h = time to hatching in hours, a = y-intercept of regression equation, b = slope of regression equation, e = base of natural logarithms, and x = temperature in degrees Celsius.

There is also some discrepancy over temperature tolerance for striped bass larvae. Table 9-2 shows the findings of researchers. Additionally, temperature was found to interact with first larval feeding time to determine survival (Rogers and Westin 1981). Time to death for unfed larvae was longer at lower temperatures, within the range of 15 to 24°C (Rogers and Westin 1981).

Characterization	Temperature Range (°C)	Citation
Tolerance	14 to 23	Mansueti (1958a)
Tolerance	11 to 22	Murawski (1969a)
Tolerance	10 to 25	Davies (1970)
Tolerance	12 to 23	Doroshev (1970)
Optimum	16 to 19	Murawski (1969a)
Optimum	15 to 22	Davies (1970)
Optimum	18 to 21	Rogers et al. (1977)
Maximum	23	Shannon and Smith (1968); Doroshev (1970)
Maximum	28	Kelly and Chadwick (1971)
Minimum	10	Davies (1970)
Minimum	11	Doroshev (1970)
Minimum	12	Rogers et al. (1977)

Table 9-2. Water temperature tolerance ranges for striped bass larvae

Duration of the various striped bass larval stages is temperature-dependent (Table 9-2). For example, the yolk-sac stage lasts three days at 23.9°C and six days at 16.7 to 17.8°C (Albrecht 1964). Finfold and post-finfold stages are also temperature-mediated, and last from 11 to 65 days, with higher temperatures shortening duration of the phase (Polgar et al. 1976; Rogers et al. 1977).

Very young striped bass have lower preferred and optimal survival temperatures (less than or equal to 20°C) (Doroshev 1970; Westin and Rogers 1978; Setzler et al. 1980; Coutant 1985). These preferences and physiological optima correspond with spring spawning temperatures, but do not stay that low for very long (Coutant 1985). In the Hudson River, for example, post-yolk-sac larvae were concentrated at depths below 6 m in the main channel during June and July, but migrated to shoal and shore zones as the water temperature increased (McFadden 1977).

Egg and larval dissolved oxygen associations

Sufficient dissolved oxygen is an important factor in ensuring the survival of striped bass eggs and larvae. Low dissolved oxygen concentrations (2.0 to 3.5 mg/L) were determined to be responsible for the absence of eggs and larvae in the Delaware River (Murawski 1969a; Chittenden 1971a). Turner and Farley (1971) reported that even moderate reductions in dissolved oxygen concentrations (from 5 to 4 mg/L) decreased the survival of eggs. Lethal limits for eggs were reported as oxygen concentrations of less than 1.5 mg/L (Mansueti 1958a) and less than 5.0 mg/L (Turner and Farley 1971). The latter value was given as "predisposing to other mortality sources," rather than being directly lethal.

Dissolved oxygen requirements for larvae are essentially identical to those required for eggs (Bain and Bain 1982). Striped bass larvae need a minimum of 3 mg/L dissolved oxygen to survive (Chittenden 1971a). Moderate reductions in dissolved oxygen concentration (from 5 to 4 mg/L) also reduced the survival of larvae (Turner and Farley 1971). Rogers and Westin (1978) reported that lethal limits were below 2.3 mg/L for yolk-sac, and below 2.4 mg/L for post-yolk-sac larvae.

Egg and larval pH associations

Striped bass egg tolerance limits for pH were reported as 6.6 to 9.0 by Bowker et al. (1969). Regan et al. (1968) reported a pH tolerance of 6 to –9, with an optimum of 7 to 8 for striped bass larvae. In addition, Davies (1970) derived a calculated optimal pH range of 7.46 to 7.85 for striped bass fry.

The effects of pH on larval striped bass, and pH trends in Chesapeake Bay spawning rivers were reviewed by Rago (1992). Laboratory tests indicated that exposure of larval striped bass (less than 50 days old) to pH less than or equal to 6.0 caused rapid mortality, which was amplified as the toxicity of total aluminum increased with decreasing pH (Rago 1992). However, increased water hardness (290 ppm) and increased salinity (5 ppt) reduce the toxic effects of low pH and inorganic contaminants (Palawski et al. 1985). Furthermore, the toxicity of low pH declines after 50 to 80 days post-hatch (Buckler et al. 1987).

Studies of contaminants conducted in association with the Emergency Striped Bass Study (see Rago 1992) demonstrated that low pH rainfall, episodic pH depressions, and extended periods of low pH conditions can occur in some poorly buffered rivers of Chesapeake Bay. Rago (1992) noted that determining the importance of such conditions to the long-term trend (in the case of the late 1970's and early 1980's, a decline) of striped bass populations is much more difficult. For acid deposition to have been a primary cause of the striped bass decline that occurred subsequent to 1970, there must have been a decreasing pH trend in spawning rivers, an increase in the frequency of low pH events during spawning periods, or a combination of both. A thorough survey of data sets on striped bass spawning habitats in Chesapeake Bay indicated no statistically significant ($p<0.10$) changes in the frequency or magnitude of extreme pH events (defined as levels of pH less than 6.5) since 1970 (Janicki et al. 1986). In the Choptank River, extreme pH events were relatively common both before and after 1970, with frequencies of 32% and 39%, respectively. In the Rappahannock River, pH events below 6.5 did not increase after 1970, but pH events in the 6.8 to 7.0 range were more frequent ($p<0.05$). In the York, James and Potomac rivers, extreme pH events were infrequent or absent in the data record (Janicki et al. 1986).

Monte Carlo simulations indicated that daily sampling would have been necessary to detect statistically significant changes ($p<0.10$) in the frequency of low pH events that were observed in the Choptank and Rappahannock rivers (Rago 1992). Low pH and high aluminum concentrations are detrimental to striped bass larvae, but there was no evidence of systematic changes in frequency or magnitude of extreme pH events in any of the Chesapeake Bay spawning rivers in the 1970's. In most locations, the historical monitoring programs were inadequate to detect all but exceptionally large changes in the frequency and magnitude of extreme pH events (Janicki et al. 1986).

Egg and larval water velocity/flow

A critical factor for egg survival and hatching success is sufficient current velocity to maintain eggs in suspension as they drift downstream (Mansueti 1958a; Albrecht 1964; Talbot 1966; Regan et al. 1968; Bain and Bain 1982). Either tidal turbulence or river discharge can provide sufficient water movement to suspend the eggs (Bain and Bain 1982).

Eggs are slightly heavier than fresh water (Raney 1952; Mansueti 1958a), with an average specific gravity of 1.0005 (Albrecht 1964; Talbot 1966), but are easily floated by agitation (Merriman 1937a; Mansueti 1958a). Specific gravity levels change during early development, and consequently alter the amount of current necessary for suspension. Eggs are less buoyant immediately after fertilization (vertical water movement of 125 cm/sec required for suspension), compared with buoyancy two to three hours later (60 cm/sec required for suspension). Additionally, after 12 hours, unfertilized eggs become opaque and more buoyant than fertilized eggs (Tatem et al. 1966).

Striped bass eggs drift with currents downstream from the spawning areas, sometimes at speeds up to 2.06 km/hr and for distances up to 150 km (Neal 1964). Eggs have been recorded in water flow rates of 54.4 to 269.6 m^3/sec (Sheridan et al. 1960), tolerate current velocities of 30.5 to 500 cm/sec, and survive best at optima of 100 to 200 cm/sec (Mansueti 1958a). In current velocities of less than 30 cm/sec, eggs are generally concentrated near the bottom, and often experience mortality (Albrecht 1964). Talbot (1966) also thought that unsuspended eggs had a poor chance of survival. Similarly, yolk-sac larvae require enough turbulence to keep them from settling to the bottom where they are often smothered (Pearson 1938; Raney 1952; Mansueti 1958a). Larvae tolerate 0 to 500 cm/sec current velocity, but survive optimally at 30 to 100 cm/sec (Regan et al. 1968). In contrast, Bayless (1968) demonstrated that eggs would hatch without any period of suspension, although the percent hatch increased with length of suspension during the first 15 hours post-fertilization.

Egg and larval suspended solid associations

Striped bass eggs appear to be adapted to high turbidity and heavy suspended sediment loads (Bain and Bain 1982). Although neither turbidity nor suspended sediments have been observed to significantly decrease hatching success (Talbot 1966; Schubel and Auld 1974), Auld and Schulbel (1978) reported tolerance limits of 0 to 500 mg/L for larvae, and a lethal limit of 1,000 mg/L for eggs. Auld and Schubel (1978) reported lethality of yolk-sac larvae at levels over 500 mg/L. The lethal dose at which 50% of larvae died after two days (i.e., 48 hr LD_{50}) was reported as 3411 mg/L by Morgan et al. (1973).

Egg and larval feeding behavior

Feeding is generally thought to begin within four to ten days post-hatch (Mansueti 1958b; Tatum 1966). Doroshev (1970) indicated that feeding began at approximately eight days and 6 to 7 mm total length (TL). In laboratory studies, Doroshev (1970) found that first-feeding larvae (less than 10 mm) preferred *Cyclops* nauplii and copepodites. In the Potomac River, Beaven and Mihursky (1980) reported positive electivity of larger copepods and cladocerans, and negative electivity of copepod nauplii and rotifers in a sample of 605 striped bass larvae. At lengths

greater than 10 mm, larvae feed primarily on larger zooplankton and macroinvertebrates (Humphries and Cumming 1973).

The availability of sufficient concentrations of suitable prey (i.e., abundant zooplankton) during the first several days of feeding is a critical factor influencing larval survival (Setzler et al. 1980; Cooper and Polgar 1981; Eldridge et al. 1981; Bain and Bain 1982) and subsequent year class strength (Heinle et al. 1975; Eldridge et al. 1981). Miller (1977) estimated that a minimum concentration of 1,864 nauplii per liter was required for successful initiation of feeding. Although zooplankton abundance fluctuates widely over time in any estuary, the potential for an abundance of zooplankton appears to be related to estuarine productivity (Bain and Bain 1982). The level of productivity in an estuary is a function of both freshwater nutrient (detritus) input to the estuary (Biggs and Flemer 1972; Hobbie et al. 1973; Saila 1975; Day et al. 1975; Polgar et al. 1976), and detritus production in adjacent salt marshes (Teal 1962; Odum and Heald 1973; Reimold et al. 1973; Stevenson et al. 1975). Detrital input to the estuary from freshwater inflow is typically greatest during the late winter and early spring (Bain and Bain 1982). Heinle et al. (1975) and others have proposed that certain climatic events, which affect nutrient release from the salt marsh and nutrient contribution by freshwater input, can influence plankton abundance during the critical larval stage and consequently affect year-class size in striped bass.

The complex relationship of estuarine productivity, zooplankton abundance, and larval striped bass survival appears to have important ramifications for the success of striped bass in estuarine systems and for the evaluation of habitat suitability (Bain and Bain 1982). Apparently, even optimal habitat will only occasionally produce ideal conditions for striped bass survival, and hence produce a strong year class. However, the loss of habitat suitability, which would diminish the potential to produce strong year classes, might ultimately have serious consequences for maintenance of a viable striped bass population.

Egg and larval competition and predation

Predation may have a significant effect on egg and larval survival; however, quantitative estimates of the magnitude of predation are lacking for fish eggs and larval fish in general (May 1974; Dahlberg 1979), and for striped bass in particular (Setzler et al. 1980, McGovern and Olney 1988). Setzler et al. (1980) and Fay et al. (1983) speculated that adult and juvenile white perch probably consume large numbers of striped bass larvae. In addition, Dendy (1978) reported predation on striped bass larvae by sessile freshwater hydra polyps, *Crespedacusta sowerbyi*. Smith and Kernehan (1981) found significant predation on striped bass larvae by the free-living copepod *Cyclops bicuspidatus.*

McGovern and Olney (1988) assessed predation by fish and invertebrates on the early life history stages of striped bass in the Pamunkey River, Virginia. Field surveys during the spawning season indicated that numerous fish and invertebrate predators varied in spatiotemporal coincidence with eggs and larvae of striped bass. Neither eggs nor larvae of striped bass were positively identified in gut samples (235 stomachs of 14 species) from field-collected fishes, although various fish species did consume many white perch eggs. In the laboratory, striped bass larvae were either attacked and killed, or eaten, by the cyclopoid copepod *Acanthocyclops vernalis,* and juvenile or adult satinfin shiner *Notropis analostanus*, spottail shiner *N. hudsonius*, tessellated darter *Etheostoma olmstedi*, white perch *Morone*

americana, striped bass, bluegill *Lepomis macrochirus*, pumpkinseed *L. gibbosus*, channel catfish *Ictalurus punctatus*, and white catfish *I. catus*. Consumption of larvae by spottail and satinfin shiners increased with larger prey densities to maximal ingestion of 150 and 81 larvae per predator per hour, respectively. At prey concentrations simulating ambient densities in the Pamunkey River (20 to 100 larvae/m^3), consumption by both fish species ranged from zero to five larvae per hour. Those estimates were considered to be lower limits because prey densities were not maintained during the experiments (McGovern and Olney 1988).

Based on their work and that of previous authors (Dendy 1978; Kohler and Ney 1980; Smith and Kernehan 1981), McGovern and Olney (1988) concluded that six fish families (i.e., clupeids, cyprinids, ictalurids, percids, centrarchids, and moronids) and at least two invertebrate species (copepod *A. vernalis*, and hydra *C. sowerbyi*) were potential predators on striped bass early life stages. Furthermore, based on population abundance at the spawning grounds and knowledge of feeding behavior, the researchers indicated that bay anchovy *Anchoa mitchelli*, Atlantic menhaden *Brevoortia tyrannus*, other clupeids, yellow perch *Perca flavescens*, inland silverside *Menidia beryllina*, other percids, other cyploid copepod species, and insect larvae, should all be considered potential predators (McGovern and Olney 1988). McGovern and Olney (1988) noted that some of the additional fish species, as well as six cyclopoid copepod species, are abundant on striped bass spawning grounds in the Chesapeake Bay region and may be predators of striped bass larvae. Juvenile bay anchovy were viewed as a potential key striped bass larval predator in tidal freshwater systems, given their abundance in mid-channel areas of the Pamunkey River, and documented laboratory predation rates on other species (Dowd 1986).

Cannibalism and predation by moronid larvae or juveniles have been suggested as additional sources of striped bass larval mortality. Larval fish replaced insect larvae as the dominant food item of juvenile striped bass (25 to 100 mm SL) in some areas of the Potomac River, suggesting that slow-growing or late-spawned striped bass larvae could be cannibalized (Boynton et al. 1981). McGovern and Olney (1988) reported predation on striped bass larvae in the laboratory by 29-day-old striped bass (15 to 18 mm SL), and concluded that cannibalism could occur within the duration of a normal spawning period.

Effects of contaminants on eggs and larvae

Several authors have summarized the effects of various pesticides, heavy metals, pharmaceutical drugs, and other commonly discharged chemical substances on striped bass eggs, larvae, and juveniles (Bonn et al. 1976; Middaugh et al. 1977; Morgan and Prince 1977; Westin and Rogers 1978). Three classes of substances have been the primary focus of study: 1) monocyclic aromatic hydrocarbons (e.g., benzene); 2) chlorinated hydrocarbons (e.g., polychlorinated biphenyls, or PCBs); and 3) residual chlorines.

Relatively low levels of residual chlorine produced pronounced impacts to striped bass eggs and larvae (Middaugh et al. 1977; Morgan and Prince 1977). Eggs experienced 100% mortality at 0.21 mg/L residual chlorine; 3.5% hatch and scoliosis (spinal curvature) at 0.07 mg/L residual chlorine; and 23% hatch, with difficult chorion detachment, at 0.01 mg/L residual chlorine (Middaugh et al. 1977). There were no significant effects at levels below 0.01 mg/L residual chlorine (Middaugh et al. 1977).

The incipient lethal concentration (level at which mortality is first observed) of residual chlorine for yolk-sac larvae was 0.04 mg/L (Middaugh et al. 1977). Eggs less than 13 hours old suffered 100% mortality at 0.43 mg/L residual chlorine (Morgan and Prince 1977). Eggs that were 24 to 40 hours old experienced 100% mortality at 0.50 mg/L residual chlorine. The LC_{50} values of residual chlorine for less than 13 hour and 24 to 40 hour-old eggs, respectively, were 0.22 and 0.27 mg/L. The corresponding value for yolk-sac larvae was 0.2 mg/L residual chlorine (Morgan and Prince 1977).

Other factors involving eggs and larvae

Striped bass populations in the past have been characterized by dramatic fluctuations in year-class size (Bain and Bain 1982). Merriman (1941) first noted the occasional occurrence of unusually large year classes of striped bass, and suggested a relationship between certain climatic conditions and strong year classes. Larval survival is traditionally considered a crucial factor in the determination of adult population size in species exhibiting large fluctuations in abundance (May 1974; Dahlberg 1979). Research indicates that this concept is particularly applicable to striped bass (Polgar et al. 1976; Cooper and Polgar 1981; Eldridge et al. 1981).

In addition, Chesapeake Bay spawning and nursery habitats for striped bass may be of primary importance in sustaining the coastal migratory stock of striped bass, given that a majority (variously stated as from 50 to 90%) of the Atlantic coastal catch of migratory striped bass originates from spawning grounds in Chesapeake Bay (Setzler et al. 1980). Spawning success and young-of-the-year survival in the Chesapeake Bay area may largely determine subsequent striped bass catches from Long Island to Maine (Tiller 1950; Raney 1952; Mansueti 1961b).

Part C. Striped Bass Juvenile and Adult Riverine and Estuarine Habitat

Geographical and temporal movement patterns

Juvenile striped bass from coastal populations generally reside in riverine and estuarine habitats on a year-round basis, and rarely complete coastal migrations. Juveniles of various (or unspecified) sizes are documented from streams (Abbott 1878), rivers (Rathjen 1955), bays (Raney 1958), sounds (King 1947), sheltered coves (Rathjen and Miller 1957), flats (Howarth 1961), and freshwater ponds (Alperin 1966b). Apparently juvenile striped bass will use various nearshore microhabitats and do not appear to require specific microhabitat conditions (Bain and Bain 1982). Older juveniles may begin to move offshore in the fall (Carlson and McCann 1969; Texas Instruments 1974), or remain in the lower river (Ritchie and Koo 1968).

Young-of-year striped bass generally move downstream to higher salinity estuarine areas during their first summer (Markle and Grant 1970; Mihursky et al. 1976; Setzler et al. 1980; Raney 1952; Carlson and McCann 1969; Texas Instruments 1974; Kernehan et al. 1981). In general, there is a downstream movement of juveniles with age (Rinaldo 1971), but this movement may be more pronounced in the second summer for fish of 150 mm or more in length. Such fish may be present in the lower reaches of rivers, or may have already entered bays and sounds (Rathjen and Miller 1957; Raney 1958; Trent 1962; Alperin 1966b; Nichols 1966).

Initiation and extent of juvenile striped bass migrations vary with location (Westin and Rogers 1978), and most juveniles remain in the river and estuarine areas where they were spawned. Fay et al. (1983) reported that little evidence exists for coastal migrations of fish less than two years old, based on Vladykov and Wallace (1938), Merriman (1941), Mansueti (1961a) and Massman and Pacheco (1961). However, many do leave their natal rivers when they are two or more years of age (Atlantic Striped Bass Plan Development Team 2003).

Both juvenile and adult striped bass form schools. Small schools of juveniles are maintained into the second year of life (Abbott 1878; Raney 1952, 1954; Bigelow and Schroeder 1953; Mansueti and Hollis 1963). Westin and Rogers (1978) reported that juvenile striped bass are found in groups of a few fish to thousands in riverine and estuarine areas. Schooling is typical for striped bass as large as 4.5 kg. Larger fish school at various times, but individuals over 13.6 kg are more often found singly or in small groups (Raney 1952; Bigelow and Schroeder 1953). Additionally, striped bass appear to school by size rather than age (Westin and Rogers 1978). Vladykov and Wallace (1938) concluded that striped bass schooling patterns were based on schooled prey fish movements, rather than isotherms or salinity variations.

In the upper Chesapeake Bay, juvenile striped bass primarily remain in nursery areas, although some move through the C&D Canal into Delaware Bay (Ritchie 1970). Juveniles in deep water in lower Chesapeake Bay estuaries in November and December move into holes over 30.5 m deep in February and March (Mansueti 1954). Additionally, young-of-year striped bass released at the mouth of the Patuxent River, Maryland, remained in shoal areas during the summer (Ritchie and Koo 1968). Five to sixteen months later, some of the fish were captured 80 km or more up the Chesapeake Bay. Young-of-the-year released 27 to 53 km up the Patuxent River in fall and winter remained more or less stationary, although there were indications of a net upriver movement extending virtually into freshwater. In the following summer (second year of life), there was a definite movement downriver and into the Chesapeake Bay (Ritchie and Koo

1968). Ritchie (1970) found that fish hatched in the Patuxent River moved up the Chesapeake Bay in their second to forth year. Similarly, Setzler-Hamilton et al. (1981) found some upstream movement of 1976 juvenile striped bass during their first summer in the Potomac River estuary.

Coutant and Benson (1990) evaluated summer water temperatures, dissolved oxygen concentrations, distribution of striped bass sub-adults and adults, and juvenile abundance indices in Chesapeake Bay to discern any influences of summer habitat suitability on historical changes in populations. Criteria used to define summer habitat suitability were the same as identified for freshwater reservoirs (temperatures below 25°C and dissolved oxygen above 2 to 3 mg/L), and were confirmed for the York-Pamunkey estuary in the lower Chesapeake Bay. Habitat suitability in the upper central basin of Chesapeake Bay declined significantly from 1962 to 1987, as did striped bass juvenile abundance indices (i.e., mean catch per standard seine haul). Thickness of the suitable temperature-oxygen habitat correlated significantly with Maryland juvenile indices the following year. Relative reproduction performance of upper bay (Maryland) and lower bay (Virginia) striped bass changed between 1967 to 1973 and 1980 to 1988, in parallel with reductions in upper bay summer habitat (Coutant and Benson 1990).

Two key habitat areas for striped bass sub-adults and adults were identified in Chesapeake Bay, based on the annual temperature-oxygen cycle (Coutant and Benson 1990). The areas were: 1) a zone of cool water in north-central Chesapeake Bay near Annapolis, Maryland, where sub-adult and adult fish congregate in summer; and 2) a shallow sill across the lower bay near the mouth of the Rappahannock River, Virginia, where warm surface waters (greater than 25°C) in summer impinge on the bottom and may block egress from the bay. Reduced juvenile production at the head of the bay and subsequent population decline would be consistent with limitation of historically important habitat in summer. The resultant physiological stresses of high temperature and low dissolved oxygen would affect reproductive competence the following year (Coutant and Benson 1990).

Local movements of adult striped bass within estuaries have also been investigated. Results from sonic tracking by Koo and Wilson (1972) in the C&D Canal indicated that movements were made in a "rest and go" manner, often with lengthy rest periods. If currents moved in a desirable direction, the fish swam or drifted with the current. In an opposing current, fish remained stationary. There was little difference between day and night movements (Koo and Wilson 1972).

Great South Bay, Long Island, New York, was determined to constitute an important post-spawning habitat for young striped bass aged two to four from multiple localities, based on tagging conducted by Alperin (1966a). Of 1,917 fish seined and tagged in May and June, 281 were recovered, with 63% from New York, 11% from New England, and the remaining 26% from areas south of New York. In New York, more recoveries came from eastern than western Long Island waters, and only 2.4% were from the Hudson River. The author speculated that the irregular presence of large numbers of striped bass in Great South Bay was related to the appearance of fish from dominant year classes originating outside the state, probably from the Delaware and Chesapeake bays. If emigration from the south diminishes, Alperin (1966a) suggested that the principal source of striped bass using the bay might be of Hudson River origin.

Juveniles/adults and the saltwater interface

Characterization	Salinity (ppt)	Life Stage	Size (mm)	Citation
Tolerance	0.2 to 16.0	Juvenile		Merriman (1937a); Dovel (1971)
Tolerance	0 to 35	Juvenile	50 to 100	Bogdanov et al. (1967)
Tolerance	0 to 20	Juvenile	20 to 50	Bogdanov et al. (1967)
Tolerance	0 to 33.7	Adult		Rogers and Westin (1978)
Optimal	10 to 20	Juvenile	50 to 100	Bogdanov et al. (1967)
Optimal	10 to 15	Juvenile	20 to 50	Bogdanov et al. (1967)
Summer	> 2.0	Sub-adult		Bason (1971)
Overwinter	< 10	Resident riverine		Bason (1971)

Table 9-3. Juvenile and adult riverine and estuarine salinity ranges

Various salinity ranges are given in Table 9-3 for juvenile and adult striped bass. In the Potomac River, large fish were found upstream in salinities of 7 to 11 ppt, beyond which body size decreased with decreasing salinity (Mansueti 1959). Mason (1882) found that young fish appeared less able to adapt to abrupt salinity changes than adults.

A significant interaction between salinity and temperature tolerance has been reported for striped bass by several authors (Tagatz 1961; Otwell and Merriner 1975; Morgan et al. 1981). Tagatz (1961) reported that a transfer of juveniles from saltwater directly to freshwater was only lethal below an acclimation temperature of 12.8°C. Otwell and Merriner (1975) reported that the highest mortality of test groups occurred for the highest salinity/lowest temperature test combination. Fish younger than 28 days had significantly lower mortality rates than fish over 28 days old, for any given temperature-salinity combination. Additionally, Doroshev (1970) found that fish 100 to 170 mm in length died when transferred from freshwater at 21.1°C to 35 ppt at 7°C, but survived the reciprocal transfer.

Juvenile/adult substrate associations

Juvenile striped bass are generally found over clean, sandy bottom (Merriman 1937a; Raney 1952, 1954; Rathjen and Miller 1957; Wolcott 1962; Smith 1971). However, juveniles with an average length of 50 mm have also been found over gravel beaches (Raney 1952), and those 71 to 85 mm have been found over a mixture of mud, sand, gravel, and rock (Merriman 1941). Rathjen and Miller (1957) found them rarely over soft bottom. Additionally, juveniles in the Hudson River used shoals as nursery habitat (Carlson and McCann 1968).

Juvenile/adult depth associations

Information on juvenile striped bass depth associations is limited. Juveniles were generally found in shallow water, although "fingerlings" were recorded from deep water (over

30.5 m) in Chesapeake Bay (Mansueti 1954). Merriman (1941) recorded specimens 71 to 85 mm long in water 2.4 m deep. Overwintering occurred in depths of up to 37 m (Mansueti 1956).

Juvenile/adult water temperature

Water temperature is a key variable in determining the distribution of juvenile and adult striped bass, as a consequence of the species' thermal niche (defined by Coutant (1985) as near 24 to 26°C) (Coutant 1985, 1986). Many field studies documented that first-year migratory juveniles occupied shallow, inshore waters of estuaries during summer (Merriman 1937a; Raney 1952, 1954; Rathjen and Miller 1957; Mansueti 1961a; Smith 1971; McFadden 1977). Water temperatures in these habitats often are above 24°C. In fact, juvenile growth in temperate estuaries is always fastest in midsummer, when temperatures are highest (Vladykov and Wallace 1952; Rathjen and Miller 1957; Koo and Ritchie 1973; Texas Instruments 1975, 1976). Indices of juvenile striped bass abundance in estuaries typically are obtained by seining or trawling in shallow nursery areas. Examples include Albemarle Sound (Hassler et al. 1981), Chesapeake Bay (Scott and Boone 1973), and the Hudson River (McFadden 1977).

The high thermal affinities of juvenile striped bass have also been observed experimentally. Davies (1973) acclimated first-year striped bass to 26.7°C, from which they tolerated transfers to 32.2°C, but died at temperatures above 35°C. Bowker et al. (1969) reported that juveniles tolerated temperatures as high as 29°C in ponds. Growth and feeding rates of one-month-old juveniles increased with temperature between 12 and 24°C (Otwell and Merriner 1975). In gradient studies in the laboratory, temperature preference of juveniles was near 25°C (Meldrim and Gift 1971). Short-term preferred temperatures were determined by Texas Instruments (1976) to be 29 to 31°C, 26 to 27°C, 23 to 24°C, and 14 to 17°C for acclimation temperatures of 24, 21 to 22, 17, and 6°C, respectively.

High summer-fall abundances of age-0 striped bass in the Patuxent River estuary, Maryland, were positively associated with low winter temperatures (Wingate and Secor 2008). Winter temperature at the site (near the mouth of the river) ranged from 3.3 to 6.6°C. Wingate and Secor (2008) did not directly address the mechanisms by which winter conditions affect the summer-fall nursery fish assemblage. However, the researchers noted that winter conditions can affect subsequent spring and summer estuarine production, spawning and recruitment phenology, and distributions of juvenile striped bass. Winter temperatures and flow may affect nursery stability and production differentially among species (i.e., anadromous versus coastal-spawning species) (Wingate and Secor 2008). It is well-documented that nursery zones expand for anadromous species during years with cold, wet winter conditions (Secor 2000; Jung and Houde 2003; North and Houde 2003).

The thermal niche of adult striped bass, based on a literature review by Coutant (1985), was 18 to 25°C (centered around 20°C). However, such a niche would be a realized one, rather than the fundamental niche (Coutant 1990) because temperature selection by large striped bass in the studies reviewed was constrained by a lack of alternative temperatures or by hypoxia. In contrast, Bettoli (2005) conducted a study in a reservoir where a broad range of temperatures were present and hypoxia uncommon; they suggested that the fundamental thermal niche of adult landlocked striped bass might be lower than literature estimates.

Characterization	Temperature Range (°C)	Citation
Tolerance	10 to 27	Bogdanov et al. (1967); Davies (1970); Hester and Stevens (1970)
Optimal	14 to 21	Krouse 1968; Doroshev 1970)
Maximum	35	Talbot (1966); Gift and Westman (1972)
Overwintering	3.55 to 6	Vladykov and Wallace (1952)

Table 9-4. Water temperature ranges for juvenile riverine and estuarine striped bass

Temperature is an important factor affecting growth and survival of juvenile striped bass (Bain and Bain 1982). Water temperature ranges are presented for juvenile striped bass in Table 9-4. Meldrim and Gift (1971) found that specimens 80 to 149 mm in length preferred temperatures varied from about 15°C, in December, to around 26°C, in October. In addition, Loeber (1951) found that juveniles acclimated to higher temperatures exhibited higher lethal limits than those acclimated to lower temperatures. For example, those acclimated at 15.6°C had an LD_{50} of 31.0°C; those acclimated at 11.0°C had an LD_{50} of 29.4°C (Loeber 1951). In another study, Tagatz (1961) found that transfers of juvenile striped bass from 12.8 and 21.1°C to 7.2°C were lethal, but the reciprocal transfer (cold to warm) was not (Tagatz 1961). These findings agree with the premise that fish acclimate more easily to rising temperatures than to falling temperatures. Furthermore, juveniles appeared to be less tolerant of abrupt temperature changes than adults (Tagatz 1961).

In comparison, adult striped bass were reported by Tagatz (1961) to tolerate temperatures from 0 to 30°C with no apparent ill effects. However, adult striped bass temperature preferences depend on ambient acclimation temperatures. The maximum upper avoidance temperature is 34°C for striped bass acclimated to 27°C in late August, while striped bass acclimated to 5°C in December avoid 13°C water (Meldrim and Gift 1971). Maximum preferred temperature of 25 to 27°C was reported during the growing season (Merriman 1941).

An important hypothesis regarding striped bass adult temperature and dissolved oxygen limitations was postulated by Coutant (1985). Coutant (1985) noted that striped bass had a "paradoxical record" of distribution and abundance, including population declines in coastal waters and variable success when introduced in various freshwater reservoirs. He analyzed the record for consistency with his hypothesis that striped bass could be "squeezed" into areas delimited by their thermal and dissolved oxygen preferences or requirements. He suggested that field and laboratory observations confirmed striped bass possessed an inherent thermal niche that changed to lower temperatures as the fish aged. Such a shift could cause local conditions, especially warm surface waters and deoxygenated deep water, to be incompatible with the success of large fish. Crowding due to temperature preferences alone, or coupled with avoidance of low oxygen concentrations, could lead further to pathology, or overfishing, and thereby be a contributing factor to population declines. The thermal niche-dissolved oxygen hypothesis was proposed as a unified perspective of the habitat requirements of striped bass that could aid in study and management of the species (Coutant 1985). Furthermore, subsequent analysis of changes in habitat quality within Chesapeake Bay (Coutant and Benson 1990) suggested that "habitat squeeze" may have been a factor in past population declines.

Juvenile/adult dissolved oxygen associations

Mansueti (1961a) was among the first to recognize that striped bass could tolerate waters having marginal water quality. This observation was supported by studies of respiratory metabolism (Neumann et al. 1981). One factor, the sensitivity of striped bass to different dissolved oxygen levels, was reviewed by Coutant (1985). Striped bass field observations and experimental results in both freshwater and estuarine systems support the statement that striped bass become physiologically stressed as the dissolved oxygen content decreases to around 3 mg/L, and that concentrations near 2 mg/L are considered uninhabitable (Coutant 1985). In Chesapeake Bay, Chittenden (1971a, 1971b) found the 3 mg/L dissolved oxygen isopleth approximately restricted the distribution of striped bass. In addition, Talbot (1966) suggested that 4 mg/L might be too low for successful reproduction.

Several studies reviewed by Coutant (1985) attempted to experimentally define the limit of striped bass performance in reduced dissolved oxygen conditions (Hoff et al. 1966; Krouse 1968; Dorfman and Westman 1970; Chittenden 1971b; Meldrim et al. 1974). Restlessness was observed at about 3 mg/L (at 16 to 19°C), followed by inactivity, loss of equilibrium, and finally death as dissolved oxygen was further lowered (Chittenden 1971b). Ventilation rate (measured in gulps per second) was maximized at 2 to 3 mg/L, but declined at lower dissolved oxygen concentrations.

Survival among juvenile striped bass transferred from ambient concentrations to 2 mg/L (at 20°C) and 3 mg/L (at 25.6°C) was 80% (Dorfman and Westman 1970). Krouse (1968) concluded, based on experiments in the water temperature range of 13 to 25°C, that striped bass were capable of surviving dissolved oxygen concentrations of 3 mg/L, but not 1 mg/L. Similarly, Bogdanov et al. (1967) reported that striped bass tolerated dissolved oxygen concentrations of 3 to 20 mg/L, and optimal values were stated as 6 to 12 mg/L. Fish acclimated at 32.8°C experienced lethality after long exposure to concentrations less than 2.4 mg/L (Dorfman and Westman 1970).

Meldrim et al. (1974) found that juvenile striped bass acclimated at 18°C generally avoided dissolved oxygen concentrations of 3.8 to 4 mg/L in experimental gradients. Furthermore, adult striped bass are known to avoid areas of 44% dissolved oxygen saturation or less (Meldrim et al. 1974). Additionally, low dissolved oxygen was determined to adversely affect appetite, and the effects of prolonged poor feeding were serious (Hoff et al. 1966).

Juvenile/adult pH associations

Davies (1970) calculated an optimal pH range for juveniles (cultured fingerlings) of 7.06 to 8.35.

Juvenile/adult water velocity/flow

Young-of-the-year striped bass are generally more abundant in areas with pronounced currents (Rathjen and Miller 1957; Wolcott 1962). Striped bass were reported to tolerate current velocities from 0 to 500 cm/sec, but optimum values were given as 0 to 100 cm/sec (Bogdanov et al. 1967).

Wingate and Secor (2008 found that high summer-fall abundances of young-of-the-year striped bass near the mouth of the Patuxent River, Maryland, were positively associated with high winter flows. In addition to expanding the nursery zone for anadromous species, high flow also intensifies the formation of the maximum turbidity zone and provides favorable early foraging conditions for estuary-spawning fishes (North and Houde 2003). Ross (2003) found that fish population abundances were correlated with availability of nursery habitat that promotes growth and survival. The lower Patuxent River study site used by Wingate and Secor (2008) was situated in a transitional zone along the river-estuary gradient and the upper-lower Chesapeake Bay gradient, and distributional shifts related to availability of nursery habitats might occur in this zone. Winter conditions might also influence the timing of migration and spawning of adult fish, as well as survival of eggs and larvae to juvenile stages (Wingate and Secor 2008). As an example, the timing of spring spawning is a critical determinant of recruitment success in anadromous species, such as striped bass and American shad, *Alosa sapidissima* (Limburg 1995; Secor and Houde 1995).

Juvenile/adult suspended solid associations

Striped bass were reported to avoid areas were total dissolved solid concentrations exceeded 180 mg/L (Farley 1966; Murawski 1969a).

Juvenile/adult feeding behavior

Young striped bass feed on zooplankton, macroinvertebrates, and fish. Stevens (1966a) reported that juveniles in the Sacramento and San Joaquin River estuary fed mainly on invertebrates in winter and spring, and switched to small fish prey when they became available in the summer. They began feeding on larger fish in their second summer (Stevens 1966a). In general, early studies determined that juveniles were not highly selective and consumed food items based on availability (Heuback et al. 1963; Stevens 1966a; Hester and Stevens 1970; Manooch 1973; Boynton et al. 1981).

In Albemarle Sound, the nursery area for the Roanoke River and the Meherrin and Nottoway tributaries of the Chowan River in North Carolina, Manooch (1973) sampled 1,094 yearling and adult striped bass for food habit analysis. The striped bass sampled ranged in size from 125 to 714 mm TL. Approximately 77% of the stomachs contained food organisms. Fifteen species of fish and ten invertebrate prey taxa were identified. Food habits varied substantially with size of fish, area of collection, and season of collection. Striped bass were capable of consuming clupeids approximately 60% of their length, but generally fed on smaller fish averaging 20% of their length. Striped bass preferred soft-rayed species, which generally occurred as juveniles in the Sound (Manooch 1973).

Manooch (1973) noted that fish were the main striped bass prey in Albemarle Sound, and occurred in 93% of the stomachs containing food. Predominant species identified were: Atlantic menhaden (*Brevoortia tyrannus*), blueback herring (*Alosa aestivalis*), and bay anchovy (*Anchoa mitchelli*). Atlantic menhaden constituted 54% of the diet (50% of volume), with medium-sized striped bass commonly containing 20 to 30 Atlantic menhaden. Approximately 12.5% of the striped bass contained bay anchovy, the only other soft-rayed fish that contributed significantly to the diet. American eel (*Anguilla rostrata*) were present in four stomachs (0.12%). Two of the

American eel were adults, and two were immature individuals, all found in spring-collected large striped bass (Manooch 1973).

Manooch (1973) noted that American eel elvers were available during the spring season and thousands were observed migrating up the Roanoke River in North Carolina, but no specimens were noted in striped bass stomachs from that area. Spiny-rayed fish occurred in only 6.7% of the stomachs, but comprised approximately 18% of the volume. Spiny-rayed species present included: spot (*Leiostomus xanthurus*), weakfish (*Cynoscion regalis*), silver perch (*Bairdiella chrysura*), Atlantic croaker (*Micropogonius undulatus*), and white perch (*Morone americana*). Species that occurred only rarely included: Atlantic silversides (*Menidia menidia*), striped bass, pumpkinseed (*Lepomis gibbosus*), and yellow perch (*Perca flavescens*).

Manooch (1973) thought that cannibalism was a rare event, with only two yearling striped bass of 229 examined containing juveniles of the same species. No striped bass were found in larger specimens. Manooch (1973) noted that the low incidence of cannibalism agreed with other prior studies on the East Coast (Hollis 1952; Stevens 1958; Schaefer 1970; Ware 1970).

Invertebrates were of secondary importance to striped bass in Albemarle Sound, and consisted primarily of blue crabs (*Callinectes sapidus*), penaeid shrimp, and gammarid amphipods. Invertebrates occurred in less than 10% of the striped bass stomachs, although they were relatively numerous (18.1% frequency) when present. Total crustacean volume was less than 3% of the stomach bulk, and frequency of occurrence values ranged from 0 to 41% during the study period. The blue crab (18 to 30 mm carapace width) was the most frequently encountered crustacean, yet it occurred in only 5.4% of stomachs (Manooch 1973).

Small striped bass (125 to 304 mm) in Albemarle Sound consumed predominantly juvenile fish. Fish occurred in approximately 96% of the stomachs. Clupeids were the principal forage group present, occurring in 59.58% of the stomachs with food. Bay anchovy was a main component of the yearling diet, present in 28.5% of the stomachs, with frequency of occurrence increasing during late summer and fall. Blueback herring (28%) and Atlantic menhaden (27.5%) were also important prey species. Spiny-rayed fish were infrequently encountered and invertebrates were of little importance, occurring in only 8 of 229 stomachs, and comprising less than 1% of the food volume (Manooch 1973).

Large striped bass (305 to 714 mm) also predominantly consumed fish (91.9%) in Albemarle Sound, but invertebrates were of more importance than they were for small striped bass, occurring in 10.43% of stomachs with food. The predominant prey of large striped bass was clupeid fish, which were identified in 75% of the stomachs. Atlantic menhaden was the primary prey species (62%), although adult blueback herring and alewife contributed to the diet during the spring. Blue crabs were the most frequent invertebrate recovered, but amphipods were of numerical importance. As many as 126 amphipods were removed from a single stomach, suggesting they were selectively ingested (Manooch 1973).

Manooch (1973) found that there were significant differences in the striped bass diet correlated with the area of collection. Chi-square tests indicated that fish taken in the eastern, more saline portion of Albemarle Sound, contained significantly more blue crabs, penaeid shrimps, and gammarid amphipods (25.4%) than those in the western Sound (2.8%). Bay anchovy, spot, Atlantic croaker, silver perch, and weakfish were also more abundant in striped bass stomachs taken from fish in the more saline waters (Manooch 1973).

Manooch (1973) also found significant seasonal variations in Albemarle Sound striped bass food habits. Striped bass collected in summer and fall fed almost exclusively on fish, and specimens collected in winter and spring contained higher percentages of invertebrates. Bay anchovy was more prevalent in fall and spring, Atlantic menhaden in summer and fall, spiny-rayed fishes in fall and winter, adult blueback herring and alewife in spring (during their spawning season), juveniles of those species during the summer (during their nursery residence), and gizzard shad in winter and spring (Manooch 1973).

Researchers at North Carolina State University sampled striped bass (aged 1, 2, and 3+ years) again in western Albemarle Sound, including the Chowan River, in May through October of 2002 and 2003, to characterize diet, prey type, and size selectivity. In this updated study, *Alosa* spp., Atlantic menhaden, bay anchovy, silversides (*Menidia* spp.), and yellow perch dominated the diets of age-1 striped bass, while Atlantic menhaden dominated the diets of older striped bass. Striped bass of all ages consumed predominantly fish prey, regardless of month or year. Each age category of striped bass selected for one or more species of prey from among the clupeids. Averaged by year, striped bass of all ages displayed strong selection *for* Atlantic menhaden and *against* spiny-rayed fish prey. Striped bass demonstrated either neutral size selectivity, or selected for relatively small prey (J. A. Buckel, Center for Marine Sciences and Technology, North Carolina State University, personal communication).

Working in the summer in the York and James Rivers, Virginia, Markle and Grant (1970) found that striped bass less than 70 mm consumed primarily mysid shrimp and insects, respectively. Juveniles between 70 and 150 mm fed primarily on naked gobies (*Gobiosoma bosci*) in the York River and grass shrimp (*Palaemonetes* spp.) in the James River (Markle and Grant 1970).

In the Chesapeake Bay, juvenile striped bass (40 to 100 mm) fed on mysid shrimp (*Neomysis americana*), as well as amphipods (*Gammarus* spp.) and *Corophium* spp. Fish from 100 to 270 mm fed on bay anchovy and various invertebrates (Bason et al. 1975). In addition, Markle and Grant (1970) and Bason (1971) stated that prey selection by juvenile striped bass varied with the salinity of the nursery environment and the corresponding food item availability.

Hollis (1952) examined the stomachs of 1,736 striped bass from Chesapeake Bay during the period June 1936 through April 1937. Striped bass were primarily piscivorous, with fish comprising 95.5% of the diet by weight. Similar to other studies, it was determined that the most common prey during summer and fall were Atlantic menhaden and bay anchovy. Young spot and Atlantic croaker were also prominent prey, dominating the diet during the winter. Less than 2% of Chesapeake Bay striped bass stomach content weight was comprised of crustaceans (0 to 46% frequency). White perch and river herring (*Alosa aestivalis* or *A. pseudoharengus*) were common in spring and early summer. Freshwater organisms were dominant in samples taken from the head of the Bay. Furthermore, there was a tendency toward reduction of feeding in late May and early June, corresponding to the spawning season (Hollis 1952).

Diet of striped bass (aged 0, 1, and 2+) in Chesapeake Bay, and its Choptank, lower Patuxent, and Potomac River tributaries, was also examined between January 1990 and March 1992 by Hartman and Brandt (1995). In general, the contribution of invertebrates to the diet declined with increases in age. Age-0 striped bass ate mostly invertebrates, predominantly polychaetes, gammarid amphipods, and to a lesser degree grass shrimp and mysid shrimp, and also juvenile naked gobies. Age-1 striped bass ate mostly invertebrates (gammarids, softshell

clams (*Mya arenaria*), and mysid shrimp) for the first half of the year, but contributions of fish (bay anchovy and Atlantic menhaden) to the diet increased thereafter. Age-2 and older striped bass ate mostly fish (primarily Atlantic menhaden). However, during spring and early summer, age-0 spot, age-1 and older white perch, and polychaetes represented as much as 74 to 89% (combined) of the diets (Hartman and Brandt 1995).

In the Delaware River, juvenile striped bass 50 to 100 mm fed primarily on mysid shrimp (*Neomysis americana*) and sand shrimp (*Crangon septemspinosa*). In low salinity tidal creeks of the Delaware drainage, fish, decapod crustaceans, amphipods, and mysid shrimp were the most important food items (Bason 1971).

In Long Island Sound, Schaefer (1970) found that striped bass between 275 and 399 mm FL fed primarily on *Gammarus* spp., *Haustorius canadensis*, and *Neomysis americana*. Fish from 400 to 599 mm fed equally on fish (bay anchovy, Atlantic silverside (*Menidia menidia*), and scup (*Stenotomus chrysops*)) and amphipods. Striped bass between 600 and 940 mm FL fed more on fish than other length groups (65% of dietary volume), but still consumed amphipods, mysid-shrimp, and lady crabs (*Ovalipes ocellatus*) (Schaefer 1970).

In the Hudson River, striped bass up to 75 mm preferred *Gammarus* spp., calanoid copepods, and chironomid larvae. Juveniles from 76 to 125 mm preferred *Gammarus* sp. and calanoids, and those from 116 to 200 mm preferred Atlantic tomcod (*Microgadus tomcod*) (Texas Instruments 1976).

Sampling by Buckel and McKown (2002) was conducted in three New York Bight embayments (Manhassett Bay, Little Neck Bay, and Jamaica Bay) from May to November in 1997 and 1998. A total of 602 juvenile striped bass were examined for dietary analysis (224 age-0; 378 age-1). Diets of age-0 and age-1 striped bass were dominated by sand shrimp (*Crangon septemspinosa*). Other important prey included mysid shrimp, amphipods, horseshoe crab eggs and juveniles, and polychaete worms (mostly *Nereis* spp.). Similar to other areas of the coast, fish prey (Atlantic silversides, killifish, and bay anchovy) became important for age-1 striped bass starting in mid-summer and fall. These same fish prey made up 25 to 30%, by weight, of age-0 striped bass diets (Buckel and McKown 2002).

Other stomach contents reported for striped bass by Smith and Wells (1977) included the following: alewife (*Alosa pseudoharengus*); blueback herring (*Alosa aestivalis*); mummichog (*Fundulus heteroclitus*); striped mullet (*Mugil cephalus*); rainbow smelt (*Osmerus mordax*); weakfish (*Cynoscion regalis*); silver hake (*Merluccius bilinearis*); American eel (*Anguilla rostrata*); American lobster (*Homarus americanus*); squid (*Ilex* and *Loligo*); and various crab, clam, and mussel species.

Juvenile/adult competition and predation

Mihursky et al. (1976) noted that larval and juvenile striped bass shared common nursery areas with white perch (*Morone americana*), which are usually more abundant than striped bass. The researchers speculated that some competition for food resources probably occurred between the two species (Mihursky et al. 1976).

Bluefish (*Pomatomus saltatrix*) co-occur with striped bass as juveniles and adults in coastal bays and estuaries during the summer and fall, and biotic interactions between the two species have been hypothesized to explain opposite trends in historic landings data (Buckel and

McKown 2002). Fay et al. (1983) noted that direct evidence for competition with adult striped bass was lacking, but speculated that other large piscivores, such as bluefish and weakfish, probably compete with them for schooling forage species.

The diets of young striped bass, bluefish, and weakfish from Chesapeake Bay, and its Patuxent and Choptank River tributaries, were defined and compared across seasons by Hartman and Brandt (1995). Dietary overlap among species and across cohorts within a species was low (24 to 51% bimonthly average range, Schoener's index). Bluefish often had higher dietary overlap values with striped bass and weakfish than with other bluefish cohorts. However, dietary overlap between striped bass and weakfish cohorts was usually low due to disparity in the use of bay anchovy by striped bass (less than 31% in all months) and weakfish (greater than 50% for most age-0 and age-1) (Hartman and Brandt 1995).

In New York, Buckel and McKown (2002) recently examined the potential for competition to influence the population dynamics of bluefish and striped bass through interactions at the juvenile stage. Juvenile bluefish and striped bass were seldom captured together during the summer and early fall, suggesting low habitat overlap at the scale of a beach seine haul. Diet overlap was low also, with age-0 bluefish (both spring- and summer-spawned cohorts) having a more piscivorous diet than age-0 and age-1 striped bass. Laboratory experiments tested for interference competition between age-0 bluefish (spring-spawned) and age-1 striped bass in both mixed and single-species treatments. Results showed that bluefish grew significantly faster than striped bass, but there was no significant difference in growth between mixed and single-species treatments. Long-term field-monitoring data showed that annual estimates of growth rate for bluefish and striped bass were not correlated with annual estimates of their potential competitor's density. Both the field and laboratory data provided no evidence for competitive dietary interactions between juvenile striped bass and bluefish (Buckel and McKown 2002).

Buckel et al. (1999) also studied predation by age-0 bluefish upon age-0 striped bass in the Hudson River estuary. Researchers measured bluefish weight, density, prey size, and diet in order to estimate loss of young-of-the-year striped bass to young-of-the-year bluefish predation. Predation mortality was compared with the total loss of striped bass in the system. Data from sampling surveys conducted since the mid-1970's were used to examine relationships between bluefish abundance and striped bass recruitment levels. Bluefish diets were dominated by young-of-the-year striped bass, bay anchovy, Atlantic silverside, and *Alosa* spp. Bluefish avoided striped bass at low densities, but selected for them at high densities, suggesting a density-dependent feeding response. In the early summer of 1993, bluefish predation accounted for 50 to 100% of the total estimated loss of young-of-the-year striped bass. A significant negative correlation existed between the relative magnitude of striped bass recruitment and bluefish abundance. The authors concluded that young-of-the-year bluefish were important predators of estuarine fish and could have a substantial impact on their recruitment (Buckel et al. 1999).

Effects of contaminants on juveniles/adults

Acute toxicities (24-hour LC_{50}) of monocyclic aromatic hydrocarbons (often present in oil spills) to 6 gram juvenile striped bass were reported by Benville and Korn (1977). The acute levels were: benzene 6.9 mg/L; toluene 7.3 mg/L; ethylbenzene 4.3 mg/L; metaxylene 9.2 mg/L;

orthoxylene 11.0 mg/L; and paraxylene 2.0 mg/L (Benville and Korn 1977). Chronic effects of exposure to sub-lethal benzene concentrations of 3.5 and 6.0 μg/L were tested for four weeks by Korn et al. (1976). The initial reaction was pronounced hyperactivity. Chronic reaction to both levels included an inability to locate and consume food properly, lower percent body fat, and lower dry and wet weight at the end of the test period (Korn et al. 1976).

The relationship between bone strength, bone health and development, and levels of organic and inorganic contaminants was examined in young-of-the-year striped bass from the Nanticoke, Potomac, and Hudson Rivers, and from a North Carolina hatchery (Mehrle et al. 1982). PCBs were the most prevalent organic contaminant found. Hudson River fish had the highest PCB levels, and both Potomac and Hudson River fish had significantly higher levels of total organic contaminant residues than the Nanticoke and hatchery groups. Arsenic, lead, selenium, and cadmium were the most prevalent inorganic contaminants found. Levels of organochlorine residues and heavy metals were highly correlated with bone strength, stiffness, toughness, and stress tolerance. Hudson River fish had the highest residue levels and the poorest bone quality and bone health of the four groups evaluated (Mehrle et al. 1982).

Additionally, the incipient lethal concentration of total residual chlorine to juvenile striped bass was 0.04 mg/L (Middaugh et al. 1977).

Effects of parasites and diseases on juveniles/adults

Parasites and diseases of striped bass are reported in Paperna and Zwerner (1976) and Bonn et al. (1976). Summary tables of parasite and disease literature are provided in Westin and Rogers (1978), Smith and Wells (1977), and Setzler et al. (1980). Commonly reported diseases of striped bass include: fin rot diseases, pasteurellosis, columnaris, lymphocystis, and epitheliocystis (Setzler et al. 1980).

Since the late 1990's, mycobacterial infections in Chesapeake Bay fish have become a concern (Ottinger and Jacobs 2006). Blazer (2006) gives a thorough review on what is known about the disease and how it affects migratory striped bass. Mycobacteriosis in striped bass is characterized by cutaneous reddish lesions, visceral lesions, or a combination of the two. It is a chronic disease that develops slowly. Pathogenesis is dependent on the species of *Mycobacterium* involved, is temperature dependent (e.g., more prevalent at high temperatures), and may be dose dependent. Furthermore, striped bass can be infected without exhibiting pathology (Blazer 2006).

Part D. Late Stage Striped Bass Juvenile and Adult Marine Habitat

Geographical and temporal patterns at sea

Migration of Atlantic coast striped bass occurs during the juvenile and adult life history stages (Atlantic Striped Bass Plan Development Team 2003), and transits habitats in the nearshore Atlantic Ocean from Topsail Island, North Carolina, to Nova Scotia, Canada. Individuals migrating to the ocean generally are found moving north along the coast in summer and fall, and south during the winter, with the extent of migration varying with age, gender, and population (Vladykov and Wallace 1938, 1952; Truitt 1940; Merriman 1941; Bigelow and Schroeder 1953; Chapoton and Sykes 1961; Sykes et al. 1961; Mansueti and Hollis 1963; Nichols et al. 1966; Clark 1968; Miller 1969; Hamer 1971; Hill et al. 1989; Morris et al. 2003; Zlokovitz et al. 2003). However, some individuals are thought to overwinter in New England (Merriman 1938; Bigelow and Schroeder 1953; Raney 1958; Saila and Pratt 1973). These migrations involve fish age-2 and older, and may include some immature females (Raney 1954). These coastal migrations are not associated with spawning and usually begin in early spring, but this time period can be prolonged by the migration of striped bass that are spawning (Bain and Bain 1982).

Striped bass tagged in U.S. waters may overwinter both with Bay of Fundy fish (in freshwater) and with Gulf of St. Lawrence fish (in estuaries). The natal origin of these fish is unknown. The incidence of U.S. fish mixing with Canadian stocks likely fluctuates with population size of the U.S. migratory stock as well as the prevailing water temperatures in the Gulf of Maine, Bay of Fundy, and Gulf of St. Lawrence (Rulifson and Dadswell 1995).

Areas along the central and south Atlantic coast are used as wintering grounds by some portion of migratory adult striped bass. The inshore zones between Cape Henry, Virginia, and Topsail Island, North Carolina, serve as winter habitat for a migratory segment of the Atlantic coast striped bass population (Holland and Yelverton 1973; Setzler et al. 1980; Laney et al. 2007; Welsh et al. 2007; U.S. Fish and Wildlife Service, Maryland Fisheries Resources Office, (Annapolis, Maryland), and South Atlantic Fisheries Coordination Office (Morehead City, North Carolina), unpublished data). At least three groups of striped bass were historically present in this area, including fish from: 1) Albemarle and Pamlico Sounds, North Carolina; 2) Chesapeake Bay; and 3) New Jersey and further north, including some fish from the Hudson River population (Holland and Yelverton 1973; U.S. Fish and Wildlife Service, Maryland Fisheries Resources Office and South Atlantic Fisheries Coordination Office, unpublished data). Based on tagging studies conducted under the auspices of the North Carolina Division of Marine Fisheries from 1968 to 1971 (Holland and Yelverton 1973), and the Atlantic States Marine Fisheries Commission from 1988 to present, striped bass wintering off Virginia and North Carolina range widely up and down the Atlantic coast, at least as far north as Nova Scotia, and represent all major migratory stocks (U.S. Fish and Wildlife Service, Maryland Fisheries Resources Office and South Atlantic Fisheries Coordination Office, unpublished data; Striped Bass Tagging Committee 2003; Welsh et al. 2007).Larger, typically female, striped bass tend to migrate greater distances (Mansueti 1961a; Fay et al. 1983). Bigelow and Schroeder (1953) found that 90% of all striped bass caught in northern waters were female. Striped bass catches from the Rhode Island (Oviatt 1977) and North Carolina (Holland and Yelverton 1973) coasts, and from Long Island's south shore (Schaefer 1968b) consisted of 90%, 90%, and 85.7% females, respectively.

The extent of involvement of North Carolina striped bass in the migration was at one time not well known (Saila and Pratt 1973). At least a portion of the North Carolina population was documented to remain in North Carolina throughout the summer (Dickson 1958). Few, if any, striped bass from south of Cape Hatteras, North Carolina, were thought to take part in the annual coastal migration (Bigelow and Schroeder 1953).

In the Chesapeake Bay drainage, estimates of percent migration vary from 1.5% of the Potomac River striped bass population to around 10% of the total Bay population (W. Laney, personal observation). Koo (1970) concluded that Chesapeake Bay striped bass between ages 2 and 3 contributed significantly to the entire Atlantic coast fishery. In support, Kohlenstein (1981) showed that approximately 50% of the age-3 female striped bass in Chesapeake Bay, and a smaller percentage of age-2 and age-4 females, moved to the coast to join the migration annually. However, few males of that age exhibited migratory behavior. In addition, Berggren and Lieberman (1978) used discriminant function analysis to show that 90.2% of the coastal striped bass landed from southern Maine to Cape Hatteras, North Carolina, were derived from fish spawned in Chesapeake Bay. Other studies (Schaefer 1968a; Austin and Custer 1977) also support the importance of Chesapeake Bay-derived fish to the migratory striped bass stock.

The principal migration route for striped bass might be up the Chesapeake Bay, through the C&D Canal and Delaware Bay, and northward along the coast (Vladykov and Wallace 1938; Mansueti and Hollis 1963; Schaefer 1968a; Shubart and Koo 1968; Nichols and Miller 1967; Miller 1969). Fish returning from northern waters apparently enter Chesapeake Bay through the C&D Canal (Whitworth et al. 1968). Additionally, a winter migration from the upper Chesapeake Bay to the James River, Virginia, was reported by Truitt (1938), but he noted it did not occur in all years.

Results of studies using tagging, parasites, meristic and morphometric characters, and biochemical genetics suggest that Canadian and U.S. striped bass populations mix during their annual migrations (Rulifson and Dadswell 1995). The extent of this mixing is undetermined (Melvin 1978; Dadswell et al. 1984; Hogans 1984; Boreman and Lewis 1987; Rulifson et al. 1987; Harris and Rulifson 1988; Waldman et al. 1988). Boreman and Lewis (1987) reported that striped bass tagged by American Littoral Society members along the U.S. eastern seaboard as far south as New Jersey have been recaptured in the Canadian maritimes. Potomac River (Maryland) and Hudson River (New York) releases have been recaptured in the Annapolis River (New Brunswick) and in Minas Basin (Rulifson and Dadswell 1995). Striped bass tagged in Nova Scotia and New Brunswick waters have been recaptured as far south as New Jersey, Delaware, Virginia, and North Carolina (Nichols and Miller 1967; Rulifson and Dadswell 1995).Striped bass populations in southeastern Canada, the Hudson River, and possibly, the Delaware River, were thought to be more or less isolated and not move great distances after spawning (Bigelow and Schroeder 1953; Morris et al. 2003; Zlokovitz et al. 2003). The Hudson River population was reported to move only as far as Long Island Sound and the New York Bight (Lyman and Woolner 1954; Clark 1968; Whitworth et al. 1968). This non-migratory behavior is supported by tagging studies in the Hudson River-Long Island Sound area (Raney 1954; Clark 1968; Schaefer 1968b; Texas Instruments 1974), southern New Jersey (Hamer 1971), Chesapeake Bay (Mansueti 1961a; Moore and Burton 1975), Potomac River (Nichols and Miller 1967; Miller 1969), and Virginia Rivers (Massman and Pacheco 1961). In almost every one of these studies, some tagged striped bass appeared to remain in the same area all year, while

others were recaptured 1,000 km or more from the release area. The basis for migratory versus non-migratory behavior is unknown (W. Laney, personal observation).

McLaren et al. (1981) found that most striped bass tagged on the Hudson River remained all year within 50 km of tagging sites. Most fish that moved out of the Long Island Sound area moved northeastward. The most northerly recapture area, which encompassed over two years of study, was Provincetown, Massachusetts. No dependence on age, size, or sex was found for the migratory segment of the Hudson River population. Evidence indicated that the Hudson River population was most likely self-perpetuating and self-contained within the river and immediate surrounding coastal area. Little evidence existed for mingling of Chesapeake and Hudson stocks, either during migrations or within overwintering populations (McLaren et al. 1981).

In contrast, there was evidence to suggest that the Delaware River population moved down the coast after spawning (Whitworth et al. 1968; Bason 1971). Furthermore, some portion of the migratory striped bass population enters and overwinters in mid-coastal rivers such as the Hudson, Mullica, and Delaware River. However, these fish are thought to make up only a small percentage of the total migratory population (Westin and Rogers 1978).

From a more ecological perspective, larger, older juveniles and adults typically remain nearshore. In fact, striped bass are usually not found more than six to eight kilometers offshore (Bigelow and Schroeder 1953; Holland and Yelverton 1973), with few beyond 16 km (Raney 1954). The maximum recorded distance offshore is around 97 to 113 km, but such individuals were viewed as strays (Raney 1952). Striped bass are found into the surf zone along ocean beaches, primarily while foraging for prey (Rosko 1966; Schwind 1972; Reiger 1997; DiBenedetto 2003). In addition, striped bass exhibit schooling behavior (Raney 1952; Raney and DeSylva 1953) throughout their range.

Salinity associations at sea

Striped bass have been recorded from 0.0 to 35.0 ppt (Bigelow and Schroeder 1953; Smith 1971). In general, the species is able to withstand abrupt salinity changes (Tagatz 1961).

Substrate associations at sea

Striped bass occur over a wide variety of substrates in oceanic inshore and nearshore areas, including: rock and boulders (Raney 1952; Bigelow and Schroeder 1953); gravel (Haddaway 1930); sand (Bigelow and Schroeder 1953); eelgrass (Haddaway 1930); and mussel beds (Bigelow and Schroeder 1953). In both estuarine and marine habitats, striped bass have been documented often along sandy beaches (Bigelow and Schroeder 1953) and rocky shores (Pearson 1931). They have been recorded from shallow bays, troughs, and gullies hollowed out by wave action, sand bars (Bigelow and Schroeder 1953), in the surf (Schaefer 1967), and sometimes under rafts of floating rockweed (Bigelow and Schroeder 1953).

Depth associations at sea

Most depths recorded for striped bass range from 0.6 to 46 m (Haddaway 1930; Nichols 1966), with larger fish generally in deeper water. As noted in many angling publications and

fishing accounts, large fish also frequent the surf zone of beaches from Maine through North Carolina (Rosko 1966; Schwind 1972; Reiger 1997; DiBenedetto 2003). Off the North Carolina Outer Banks during the winter, striped bass are commonly present from the surf zone out to depths of 18.3 m. As an exception, Bigelow and Schroeder (1953) reported the capture of an 18-inch striped bass taken by trawl in 128 m during February 1949, about 111 km south of Martha's Vineyard. The researchers believed this constituted evidence for possible wintering by some striped bass on the bottom, "...well out on the continental shelf in localities where the otter trawlers do not ordinarily operate..." (Bigelow and Schroeder 1953).

Water temperature associations at sea

Striped bass have been recorded in the ocean at water temperatures ranging from 0.1°C (Clark 1968) to around 27°C, although kills may occur at the higher temperatures (Raney 1952; Bigelow and Schroeder 1953; Talbot 1966). Adult striped bass normally avoid temperatures higher than about 18.5 to 21.0°C (Goode et al. 1884). However, they can remain active down to 1.0°C, and can withstand abrupt temperature changes (Mansueti 1959; Tagatz 1961).

Water velocity/flow associations at sea

Striped bass are typically found where some current is running (Raney 1952; Bigelow and Schroeder 1953). Bigelow and Schroeder (1953) give a good description of typical striped bass haunts in Gulf of Maine habitats:

> The best spots along rocky shores are in the surf generally, and in the wash of breaking waves behind offlying boulders and among them, or where a tidal current flows most swiftly past some jutting point. In the mouths of estuaries they are apt to hold to the side where the current is the strongest, and in the breakers out along the bar on that side. In shallow bays, they often pursue small fry among the submerged sedge grass when the tide is high, dropping back into the deeper channels on the ebb. And they frequent mussel beds, both in enclosed waters and on shoal grounds outside, probably because these are likely to harbor an abundance of sea worms (*Nereis*).

Feeding behavior at sea

Schaefer (1970) examined the food habits of striped bass taken from the surf on Long Island, New York. He noted that the bay anchovy was the dominant vertebrate prey item, and that amphipods and mysid shrimp were the dominant invertebrate prey. Relative occurrence of invertebrates in the diet of striped bass in the Long Island surf decreased significantly between spring and fall, but no-significant differences were attributable to fish size. The relative occurrence of vertebrate prey increased significantly between spring and fall, as well as with increases in fish size (Schaefer 1970). In contrast, Oviatt (1977) reported that striped bass feeding inshore ate Atlantic menhaden, while those captured offshore fed on sand lance (*Ammodytes* spp.).

Food habits of striped bass in Massachusetts coastal waters were determined by Nelson et al. (2003). Striped bass were collected from four different habitats (i.e., estuaries, ocean-facing

beaches, rocky shorelines, and ocean waters more than 900 m offshore) in the North Shore, Cape Cod Bay, and Nantucket Sound regions. Sampling was conducted from June through September of 1997 through 2000. Stomach contents of 3,006 striped bass were examined for patterns in prey composition and body size related to coastal region, time period of capture, foraging habitat, and length of striped bass. Length of striped bass sampled ranged from 290 to 1,162 mm TL. Over 48 prey species representing 55 families from six phyla were found in the stomachs. Fish (mostly clupeids, silversides (*Menidia* sp.), and sand lance (*Ammodytes* sp.)) and crustaceans (mostly sand shrimp (*Crangon septemspinosa*), rock crab (*Cancer irroratus*), and American lobster (*Homarus americanus*)) dominated the diet of striped bass by both weight (91 to 95%) and number (87 to 97%), and had a high frequency of occurrence (42 to 66%) in the stomachs. Similarity in prey taxa among coastal regions was moderate to high (58 to 74%) (Nelson et al. 2003).

In addition, stomach contents of striped bass larger than 675 mm TL collected during August and September from estuaries and rocky shoreline habitats in the North Shore and Cape Cod Bay regions had a higher average percentage of Atlantic menhaden (*Brevoortia tyrannus*) by weight than similar-sized striped bass collected during June and July from the same habitats. Also, in the North Shore area, striped bass larger than 675 mm TL sampled in rocky shorelines contained a higher average percentage of rock crabs by weight than similar-sized bass taken in estuaries. Striped bass larger than 675 mm TL in rocky habitats consumed more American lobster than smaller striped bass residing in this same habitat. Furthermore, the size distribution of the dominant fishes and crabs (i.e., sand lance, American menhaden, rock crab, and green crab (*Carcinus maenus*)) consumed by striped bass was related to bass body size. In summary, benthic prey types were found to be a major component of the diet of striped bass in Massachusetts coastal waters (Nelson et al. 2003).

Section II. Identification and Distribution of Habitat Areas of Particular Concern for Striped Bass

Since migratory striped bass are not a species managed jointly with a federal Fishery Management Council, and since there is no formal federal Fishery Management Plan for the species, Essential Fish Habitat (EFH) has not been formally described or designated. Therefore, the definition of a Habitat Area of Particular Concern (HAPC) is modified to be areas within the species' habitat that satisfy one or more of the following criteria: 1) provides important ecological function; 2) is sensitive to human-induced environmental degradation; 3) is susceptible to coastal development activities; or 4) is considered to be rarer than other habitat types. Any HAPC designated by the ASMFC for a species solely under its management is not subject to the consultant requirements of the Magnuson-Stevens Act. Any HAPC described for Atlantic migratory striped bass will be a subset of the habitats described in Section I. There are four habitat types that might qualify as HAPCs for Atlantic migratory striped bass, and they are discussed below.

Spawning sites occur in the freshwater portions of estuaries, or their tributaries, along the Atlantic coast. Such sites provide the critical ecological function of reproduction; are sensitive to anthropogenic impacts such as dam emplacement, nutrient and sediment loading, and pollution; are susceptible to navigational dredging and other coastal development activities; and are relatively small in extent and extremely rare in comparison to the areal extent of other migratory striped bass habitats.

Nursery areas are much broader in extent. These areas include the freshwater and low-salinity portions of tributaries and their receiving estuaries for age 0 to 2 striped bass, and the higher salinity bays, estuaries, and the nearshore ocean for older juveniles. These sites provide the critical ecological function of growth to maturity; are sensitive to anthropogenic impacts such as navigational dredging and port development, sedimentation, toxic and hypoxic conditions, nutrient loading, and hypoxia; are highly susceptible to coastal development impacts from recreational and commercial vessel traffic, and receive all terrestrial runoff; and are limited in extent, although less rare than *spawning habitats*.

Inlets provide the only means of ingress and egress for striped bass adults and older juveniles migrating to and from riverine spawning and estuarine nursery habitats. They provide the critical ecological function of access to habitats necessary for reproduction and growth to maturity; they are sensitive to human-induced environmental degradation as a result of channel alterations, such as deepening and stabilization; they are all coastal and highly susceptible to coastal development activities, both commercial and recreational; and they are perhaps rarer (smaller in extent) than *spawning habitats*.

Finally, *wintering grounds* occur in the nearshore Atlantic Ocean from Long Island Sound south to at least Topsail Island, North Carolina. These habitats provide the critical ecological function of foraging and cover for adults most of the year; are sensitive to human-induced environmental degradation due to fishing activities, commercial navigation, offshore oil and gas exploration, and construction of offshore liquid natural gas (LNG) facilities; they are all coastal and subject to the aforementioned coastal development activities; and they are restricted to a relatively narrow band of nearshore ocean, although not as rare as *spawning habitats* and *inlets*.

Section III.   Present Condition of Habitats and Habitat Areas of Particular Concern for Striped Bass

Concerns regarding the condition of, and threats to, habitats used by striped bass and other Atlantic coast estuarine-dependent fish species are not new. As an example, the following quotations are provided:

> *The warning here is that fish resources of the seashores and bays of the Atlantic coast are in danger of extreme depletion from poorly planned community and industrial development* (Clark 1967, page 1).

> *It is the sad truth that our Atlantic estuaries are not being maintained in good condition—instead they are being systematically demolished, altered or poisoned in almost every town and county along the sea coast. Big and small projects are gradually eating away this precious environment and wreaking havoc with fish and shellfish resources harvested there* (Clark 1967, page 11).

> *Frequently, the real effects of estuarine projects are masked until the last moment and then the work is steamrollered in the face of the last minute opposition* (Clark 1967, page 17).

The statements, just as applicable today as when they were written almost forty years ago (despite the intervening passage of the Clean Water Act and National Environmental Policy Act), are from John Clark's *Fish and Man: Conflict in the Atlantic Estuaries* (1967). Clark (1967) summarized the threats to Atlantic estuaries (e.g., filling to establish upland, navigational dredging, gravel and sand mining, mosquito control, marsh impoundment, highway construction, and water control), and described measures the fourteen coastal states were taking to mitigate their impacts.

Two authors have addressed the long-term impacts on striped bass of human alterations to habitats in Chesapeake Bay (Mansueti 1961b; Rago 1992). Rago (1992) reviewed the consequences of habitat degradation for striped bass, and the difficulty of linking such degradation to striped bass population trends. It was noted that estuarine environments, such as Chesapeake Bay, are dynamic ecosystems, and organisms that persist in them must be highly adaptable to change (Saila 1975).

Due to the large amount of information and time constraints, portions of Section III of this chapter are based on information provided in the two National Coastal Condition reports (Summers 2001, 2004), which address habitat quality in most of the estuaries occupied by migratory striped bass juveniles and adults. Additional information was derived from the State Comprehensive Wildlife Conservation Strategies, and individual state efforts such as the North Carolina Coastal Habitat Protection Plan (CHPP) (Street et al. 2005), which contain evaluations of habitats used by striped bass.

Habitat quantity

Striped bass spawning migrations formerly extended much farther upstream in a number of rivers than they do at present, at least under conditions of high spring river discharges. In many systems, access to former spawning habitats above the Fall Line is either totally or

partially blocked by dams and locks, or dams equipped with fish passage devices that allow only partial passage of spawning striped bass. On a range-wide basis there has been an overall net loss of, and/or reduction of, access to spawning habitat. For example, in the Cape Fear River, North Carolina, navigation locks and associated low-head dams constructed by the U. S. Army Corps of Engineers, as well as upstream dams constructed for cooling water supply (e.g., Progress Energy Carolinas, Inc., Buckhorn Dam) and flood control (e.g., U. S. Army Corps of Engineers, B. Everett Jordan Dam and Reservoir), reduce or prevent access to historic spawning habitats. The same situation exists for many dams and locks along the Atlantic coast.

Habitat quality

The quality of striped bass spawning habitats has been compromised by reduced access, the presence of contaminants, adverse flow regimes due to hydropower generation from upstream dams, and functional loss due to changes induced by navigational dredging and filling to create uplands for development.

Section IV. Significant Environmental, Temporal, and Spatial Factors Affecting Distribution of Striped Bass

Table 9-5. Significant environmental, temporal, and spatial factors affecting distribution of striped bass. This table summarizes the current literature on striped bass habitat associations. For most categories, optimal and tolerable ranges have not been identified, and the summarized habitat parameters are listed under the category reported. Please note that, although there may be subtle variations between systems, the following variables may include a broad range of values to encompass the different systems that occur along the East Coast. NIF = No Information Found.

Life Stage	Time of Year and Location	Depth (m)	Temperature (°C)	Salinity (ppt)	Substrate	Current Velocity (m/sec)	Dissolved Oxygen (mg/L)
Adult (Spawning)	Migration into natal rivers along the Atlantic coast, from North Carolina to Quebec in late February to June (south to north progression)	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** 14-20 **Optimal:** 15.8-20 **Reported:** 10-26.5	**Tolerable:** NIF **Optimal:** 0-1.5 **Reported:** 0-23	**Tolerable:** NIF **Optimal:** NIF **Reported:** Silt, mud/sand, cobble, or rock bottom	**Tolerable:** NIF **Optimal:** NIF **Reported:** Suitability increases with increased flow	**Tolerable:** >5 **Optimal:** NIF **Reported:** >3
Egg and Larval	Spawning grounds of individual rivers in late February to June (south to north progression)	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF	**Tolerable:** 14-23 (egg); 10-25 (larvae) **Optimal:** 17-21 (egg); 15-22 (larvae) **Reported:** 12-24 (egg); 10-28 (larvae)	**Tolerable:** NIF **Optimal:** 1.5-3 (egg); 3-7 (larvae) **Reported:** 0-10 (egg); 0-32 (larvae)	**Tolerable:** NIF **Optimal:** NIF **Reported:** Sandy/rocky	**Tolerable:** 7.3 (egg); 0-5 (larvae) **Optimal:** 0.3-1 (larvae) **Reported:** NIF	**Tolerable:** >5 **Optimal:** NIF **Reported:** NIF

Life Stage	Time of Year and Location	Depth (m)	Temperature (°C)	Salinity (ppt)	Substrate	Current Velocity (m/sec)	Dissolved Oxygen (mg/L)
Early Juvenile – Riverine Environment	Year round residency; move downstream with age	**Tolerable:** NIF **Optimal:** NIF **Reported:** 0-37	**Tolerable:** 10-27 **Optimal:** 14-21 **Reported:** 3.55-35	**Tolerable:** 0-35 **Optimal:** 10-20 **Reported:** NIF	**Tolerable:** NIF **Optimal:** NIF **Reported:** Mixture of mud, sand, gravel, rock; rarely soft bottom	**Tolerable:** 0-5 **Optimal:** 0-1 **Reported:** NIF	**Tolerable:** 3-20 **Optimal:** 6-12 **Reported:** >3
Juvenile and Adult (At-sea)	Coastal migrations beginning in spring	**Tolerable:** NIF **Optimal:** NIF **Reported:** 0.6-46	**Tolerable:** 1-18.5 **Optimal:** NIF **Reported:** 0.1-27	**Tolerable:** NIF **Optimal:** NIF **Reported:** 0-35	**Tolerable:** NIF **Optimal:** NIF **Reported:** Rock, boulder, sand, gravel, mussel beds	**Tolerable:** NIF **Optimal:** NIF **Reported:** Swift flow	**Tolerable:** NIF **Optimal:** NIF **Reported:** NIF

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Chapter 10

THREATS TO DIADROMOUS SPECIES

Please Note: Due to broad geographic ranges, diadromous species are susceptible to varied threats throughout different life stages. The threats identified under this section occur during the freshwater and/or estuarine portion of species life histories.

PART I. IDENTIFICATION OF THREATS

THREAT #1: BARRIERS TO UPSTREAM AND DOWNSTREAM MIGRATION

Section 1.1A: Dams and Hydropower Facilities

Issue 1.1A.1: Blocked or restricted upstream access

There has been considerable loss of historic spawning habitat for shad and river herring due to the dams and spillways impeding rivers along the East Coast of the United States. Permanent man-made structures pose an ongoing barrier to fish passage unless fishways are installed or structures are removed. Low-head dams can also pose a problem, as fish are unable to pass over them except when tides or river discharges are exceptionally high (Loesch and Atran 1994). Historically, major dams were often constructed at the site of natural formations conducive to water power, such as natural falls. Diversion of water away from rapids at the base of falls can reduce fish habitat, and in some cases cause rivers to run dry at the base for much of the summer (MEOEA 2005).

Example: American shad

Many dams have facilities that are designed to provide upstream passage to spawning habitat for migratory species. However, dams without adequate upstream fish passage facilities prevent, or significantly reduce, the numbers of migratory fish that return to available habitat (Quinn 1994). Suboptimal fish passage at a low-head dam on the Neuse River, North Carolina, resulted in limited production of American shad in that system (Beasley and Hightower 2000). Subsequent removal of the dam in 1998 facilitated the return of American shad and striped bass to historic spawning habitats above the dam (Burdick 2005).

American shad likely spawned in most, if not all, rivers and tributaries in their range prior to dam construction along the Atlantic coast (Colette and Klein-MacPhee 2002). Precise estimates are not possible, but scientists speculate that at least 130 rivers supported historical runs; now there are fewer than 70 spawning systems for American shad. Furthermore, individual spawning runs at one time may have numbered in the hundreds of thousands, but current runs may provide less than 10% of historic spawning habitat (Limburg et al. 2003). Dams built from the 19th century through the mid-20th century on several major tributaries to the Chesapeake Bay have substantially reduced the amount of spawning habitat available to American shad (Atran et al. 1983; CEC 1988), and likely contributed to long-term stock declines (Mansueti and Kolb 1953).

Example: American eel

Aside from presence/absence studies of American eel, no other methods are currently used to determine anthropogenic impacts on American eel populations. Since American eel randomly disperse throughout Atlantic coastal tributaries, it is assumed that they will colonize all available inland areas. Thus, an absence of American eel is often attributed to anthropogenic impacts. However, this theory is complicated by the fact that this speciesnaturally decreases in density as individuals migrate upriver from the sea, making it difficult to quantify the true impact that dams are having on American eel populations (Smogor et al. 1995; Richkus and Whalen 1999; Wiley et al. 2004). Using GIS analysis, Busch et al. (1998) found that 84% of Atlantic coastal tributary access has been lost or restricted from use by American eel. Habitat loss was greatest from Maine to Connecticut (91%), with a reduction in stream access from 111,482 km to 10,349 km. In the mid-Atlantic region (New York through Virginia), stream habitat has been reduced (88%) from 199,312 km to 24,534 km. From North Carolina to Florida, stream habitat has been reduced (77%) from 246,007 km to 55,872 km (Busch et al. 1998). In the St. Lawrence watershed, Verreault et al. (2004) identified 151 hydrodams greater than 2.5 m equipped with turbines and 8260 dams greater than 2.5 m without turbines. They estimate that these dams block 12,140 km^2 of suitable habitat within the St. Lawrence watershed, and that this loss of habitat represents 836,545 fecund females (Verreault et al. 2004).

Most fishways, particularly those designed for salmonids, are not adequate for American eel (Verdon et al. 2003) because they do not accommodate the unique swimming mode of anguillid eels (Knights and White 1998). Because American eel can climb damp substrates, they are sometimes able to ascend dams without passage structures (Legault 1988; Haro et al. 2000). Unfortunately, many coastal rivers do not have provisions for the upstream passage of American eel (Haro et al. 2000).

Barriers that impede or restrict upstream passage for American eel may cause a significant decrease in recruitment to upstream habitats. Because phenotypic sex in American eel may be determined by environmental factors, such as population density (Krueger and Oliveira 1999), a decline in recruitment could ultimately reduce the numbers of females that migrate out to sea to spawn (Verdon et al. 2003). River impoundments that lack fish passage structures could contribute to imbalanced sex ratios dominated by males, if high concentrations of American eel become trapped below the barriers (Richkus and Whalen 1999; Haro et al. 2000). Wiley et al. (2004) found that in Maryland, stream sites within 5 km of impassable or semi-passable structures had a mean American eel density twice that of sites located further away, demonstrating that American eel congregate in areas below artificial structures. American eel densities below taller structures were some of the highest recorded throughout the study (Wiley et al. 2004). American eel trapped below barriers could also incur mortality by increased predation, competition for food, and the spread of disease (Haro et al. 2000).

Machut (2006) found that an increase in the number of barriers on the Hudson River significantly lowered American eel condition (measure of fat content). American eel condition may be affected when their natural migratory behavior is altered. This can occur because dams may hamper movements between different habitat types (Cairns et al. 2004), and American eel have been documented moving between freshwater and estuarine habitats (Morrison et al. 2003). As brackish waters tend to lead to increased productivity and growth

(Helfman et al. 1987; Morrison and Secor 2003), a decreased ability to migrate back and forth into this type of habitat may lead to decrease in fat content. Svedäng and Wickstrum (1997) found that eels with a lower fat content (lower condition) might stall their migrations downstream in order to increase fat content to a level that will sustain them.

<u>Example: Atlantic sturgeon</u>

The most significant threat to Atlantic sturgeon is the loss of upstream spawning habitat from dams (Hill 1996; NRC 1996; Secor et al. 2002). The construction of dams is considered to be a factor in the reduction and elimination of Atlantic sturgeon in New England (ASMFC 1998; USFWS-NMFS 1998). Atkins (1887) noted a large decrease in the abundance of Atlantic sturgeon after a dam was built on the Kennebec River in Augusta, Maine. Before construction of the dam, the fish spawned between Augusta and Waterville, Maine (Atkins 1887; Colette and Klein-MacPhee 2002). The Roanoke River, North Carolina, historically a spawning ground of the Atlantic sturgeon, now restricts migration of Atlantic sturgeon with a 22 m high dam (Armstrong and Hightower 2002). Armstrong and Hightower (2002) estimate that 58% to 73% of the historic Atlantic sturgeon spawning habitat in the Roanoke River is now located above this dam. Additionally, the Wilson Dam in the Santee River Basin, South Carolina, eliminated almost all spawning habitat along this river (P. Brownell, NOAA Fisheries, Southeast Regional Office, personal communication).

Issue 1.1A.2: Impacts during downstream migration

Another impact of dams on diadromous species migration is their potential to cause mortality to young fish that pass over sluices and spillways during out-migration. Potential effects to fish passing through spillways or sluices may include injury from turbulence, rapid deceleration, terminal velocity, impact against the base of the spillway, scraping against the rough concrete face of the spillbay, and rapid pressure changes (Ferguson 1992; Heisey et al. 1996).

<u>Example: Shad and river herring</u>

Prior to the early 1990s, it was thought that migrating shad and river herring suffered significant mortality going through turbines during downstream passage (Mathur and Heisey 1992). One study estimated that mortality of adult American shad passing through a Kaplan turbine was approximately 21.5% (Bell and Kynard 1985).

Juvenile shad emigrating from rivers have been found to accumulate in larger numbers near the forebay of hydroelectric facilities, where they become entrained in intake flow areas (Martin et al. 1994). Relatively high mortality rates were reported (62% to 82%) at a hydroelectric dam for juvenile American shad and blueback herring, depending on the power generation levels tested (Taylor and Kynard 1984). In contrast, Mathur and Heisey (1992) reported a mortality rate of 0% to 3% for juvenile American shad (55 to 140 mm fork length), and 4% for juvenile blueback herring (77 to 105 mm fork length) through Kaplan turbines. Mortality rate increased to 11% in passage through a low-head Francis turbine (Mathur and Heisey 1992). Other studies reported less than 5% mortality when large Kaplan and fixed-blade, mixed-flow turbines were used at a facility along the Susquehanna River

(RMC 1991, 1994). At the same site, using small Kaplan and Francis runners, the mortality rate was as high as 22% (NA 2001). At another site, mortality rate was about 15% where higher revolution, Francis-type runners were used (RMC 1992).

Additional studies reported that changes in pressure had a more pronounced effect on juveniles with thinner and weaker tissues as they moved through turbines (Taylor and Kynard 1984). Furthermore, some fish may die later from stress, or become weakened and more susceptible to predation, so losses may not be immediately apparent to researchers (Gloss 1982).

Example: American eel

Risks to fish passing through turbines during downstream migration vary depending on the species. For American eel, greater mortality may be associated with the Kaplan turbine than with the Francis turbine (Desrochers 1994). Berg (1986) reported that 15% to 50% of European eel experienced lethal injuries in a Kaplan turbine. Monten (1985) reported injury rates of 9%, 65%, and 100% for European eel 50 to 52 cm in length from a Francis turbine operating under generator load conditions of 61%, 80%, and 100%. However, Hadderingh and Bakker (1998) reported disparate results for a Kaplan turbine. They found that European eel of comparable sizes had injury rates of 23%, 10%, and 6%, as flow increased. RMC (1995) reported that American eel (average size 86 cm) sustained a 9% injury rate in a small Francis turbine, but results may have been underestimated (McCleave 2001). At the Robert Moses Power Dam in New York, survival rates of American eel passing through the turbine were estimated at 84% for 1 hour and 73.5% for 88 hours. Seventy-six of 207 American eel had a visible injury after passage. Furthermore, 24.1% of the American eel had debilitating or fatal injuries, including severed bodies, hemorrhaging, lacerations, damaged eyes, scrapes, loss of equilibrium, and internal or vertebral damage (NA and Skalski 1998). American eel injured while passing through one turbine face a greater chance of mortality at the next facility downstream (McCleave 2001). Horizontal bulb turbines have the least impact on American eel due to the greater distance between bulb vanes and runner blades, and fewer blades than on vertical turbines (Hadderingh and Bakker 1998). Additionally, female American eel are generally longer than males of the species (Helfman et al. 1987; Krueger and Oliveira 1997; Oliveira and McCleave 2000), which may contribute to increased risk of mortality (Travade and Larinier 1992). Another potential threat of hydroelectric facilities is an alteration of natural lighting. Silver-phase American eel migrate downstream primarily at night and are strongly photophobic, so lighting may alter normal downstream migratory behavior (Haro and Castro-Santos 2000). Artificial lighting has also been found to impact yellow-phase American eel migrating upstream (Verdon et al. 2003).

Example: Atlantic sturgeon

For Atlantic sturgeon, larger fish may be more prone to mortality from hydroelectric dams. Dadswell and Rulifson (1994) found three dead Atlantic sturgeon (1.5 m to 2.0 m length) below the Annapolis River STRAFLO turbine in the Bay of Fundy. The cause of death was a mechanical strike, which is common for fish of larger sizes (Dadswell and Rulifson 1994).

Issue 1.1A.3: Delayed migration

Example: Alosines

When juvenile alosines delay outmigration, they may concentrate behind dams, making them more susceptible to actively feeding predators. They may also be more vulnerable to anglers that target alosines as a source of bait. Delayed outmigration can also make juvenile alosines more susceptible to marine predators that they may have avoided if they had followed their natural migration patterns (McCord 2005a). In open rivers, juvenile alosines gradually move seaward in groups that are likely spaced according to the spatial separation of spawning and nursery grounds (Limburg 1996; J. McCord, South Carolina Department of Natural Resources, personal observation).

Issue 1.1A.4: Changes to the river system

In addition to physically impeding fish migration, dams can have other impacts on anadromous fish habitat. Releasing water from dams and impoundments (or reservoirs) may lead to flow alterations, altered sediment transport, disruption of nutrient availability, changes in water quality downstream (including both reduced and increased changes in temperatures), streambank erosion, concentration of sediment and pollutants, changes in species composition, solubilization of iron and manganese and their absorbed or chelated ions, and hydrogen sulfide in hypolimnetic (release of water at low level outlets) releases (Yeager 1995; Erkan 2002). Many dams spill water over the top of the structure where water temperatures are the warmest, which essentially creates a series of warm water ponds rather than a natural stream channel (Erkan 2002). Conversely, water released from deep reservoirs may be poorly oxygenated, below normal seasonal water temperature, or both, thereby causing loss of suitable spawning or nursery habitat in otherwise habitable areas.

Reducing minimum flows can dehydrate otherwise productive habitats causing increased water temperature or reduced dissolved oxygen levels (ASMFC 1985, 1999; USFWS et al. 2001).

Pulsing or "hydropeaking" releases typically produce the most substantial environmental alterations (Yeager 1995), including reduced biotic productivity in tailwaters (Cushman 1985).

During low flow periods (typically summer and fall), gases, dissolved oxygen in particular, may be depleted (Yeager 1995). Storing water at hydropower facilities during times of diminished rainfall can also lead to low dissolved oxygen conditions downstream. Such conditions have occurred along the Susquehanna River at the Conowingo Dam, Maryland, from late spring through early fall, and have historically caused large fish kills below the dam (Krauthamer and Richkus 1987).

Example: Alosines

Disruption of seasonal flow rates in rivers has the potential to impact upstream and downstream migration patterns for adult and juvenile alosines (ASMFC 1985, 1999; Limburg 1996; USFWS et al. 2001). Changes to natural flows can also disrupt natural productivity and

availability of zooplankton, which is nourishment for larval and early juvenile alosines (Crecco and Savoy 1987; Limburg 1996).

Although most dams that impact diadromous fish are located along the length of rivers, fish can also be affected by hydroelectric projects at the mouths of rivers, such as the large tidal hydroelectric project at the Annapolis River in the Bay of Fundy, Canada. Dadswell et al. (1983) found that this particular basin and other surrounding waters are used as foraging areas during summer months by American shad from all runs along the East Coast of the United States. Because the facilities are tidal hydroelectric projects, fish may move into and out of the impacted areas with each tidal cycle. Although turbine mortality is relatively minor with each passage, the repeated passage into and out of these facilities may cumulatively result in substantial overall mortalities (Scarratt and Dadswell 1983).

Example: American eel

River flow changes from dams may be especially stressful for poor swimming American eel elvers during their upstream migration. In fact, changes to the pattern and/or volume of discharge from a dam may delay or halt elver migration (Jessop and Harvie 2003). Jessop and Harvie (2003) found that flow changes at the Mactaquac hydroelectric dam on the St. Johns River, New Brunswick, may have caused a decrease in recruitment of American eel elvers through the dam. Prior to 1980, a large number of American eel elvers migrated through the Mactaquac dam. In 1980, two turbines were installed at the dam; following this change, migration of elvers ceased and has not since been documented. Researchers concluded that changes to hourly and daily discharge patterns could possibly account for the failure of elver migration (Jessop and Harvie 2003).

Example: Atlantic sturgeon

Atlantic sturgeon can be affected by altered dissolved oxygen concentrations, temperatures, water flow, destratification, water withdrawal, modified sediment load and channel morphology, and contaminated water as a result of dams that change the river system (Hill 1996; NRC 1996; Secor et al. 2002). Deepwater releases in late winter through early spring are often below habitable temperatures for Atlantic sturgeon ($13.3\,^{o}C$ to $17.8\,^{o}C$) (Borodin 1925). Dams can alter hard substrates that are utilized by Atlantic sturgeon for spawning habitat (Parsley et al. 1993; Beamesderfer and Farr 1997); impoundments can also be a limiting factor in Atlantic sturgeon recovery (Secor et al. 2000; Secor and Niklischek 2001). Furthermore, loss of access to substrate, as well as the degraded quality of substrate, may be the largest factors hampering the recovery of Atlantic sturgeon (P. Brownell, NOAA Fisheries, Southeast Regional Office, personal communication).

Issue 1.1A.5: Secondary impacts

Blocked migratory paths can reduce the diadromous species contribution of nutrients and carbon to riparian systems. Riverine habitats and communities may be strongly influenced by migratory fauna that provide a significant source of energy input (Polis et al. 1997). Furthermore, many freshwater mussels are dependent upon migratory fishes as hosts

for their parasitic larvae (Neves et al. 1997; Vaughn and Taylor 1999); loss of upstream habitat for migratory fish is a major cause of mussel population declines (Williams et al. 1993; Watters 1996).

Example: Alosines

It is estimated that the annual biomass contribution of anadromous alosines to the nontidal James River, Virginia, was 155 kg/ha (assumes 3.6 million fish with 70% post-spawning mortality) in the 1870s, before dams blocked upstream migration (Garman 1992). Based on the estimated 90% reduction in alosine abundance in the Chesapeake Bay over the past 30 years, Garman and Macko (1998) concluded that, "the ecological roles hypothesized for anadromous *Alosa* spp. may now be greatly diminished compared to historical conditions."

Section 1.1B: Avoiding, Minimizing, and Mitigating Impacts of Dams and Hydropower Facilities

Approach 1.1B.1: Removing dams

Not all projects are detrimental to fish populations, so each site should be evaluated separately to determine if fish populations will be (or are being) negatively impacted (Yeager 1995). Wherever practicable, tributary blockages should be removed, dams should be notched, and bypassing dams or installing fish lifts, fish locks, fishways, or navigation locks should be considered. Full dam removal will likely provide the best chance for restoration; however, it is not always practicable to remove large dams along mainstem rivers. Removing dams on smaller, high-order tributaries is more likely to benefit ascending river herring than shad, which spawn in the larger mainstem portions of rivers (Waldman and Limburg 2003).

Example: Successful Dam Removals

Along the large, lower-river tributaries of the Susquehanna River, Pennsylvania, at least 25 dams have either been removed or fitted with fishways, which has provided a total of 350 additional stream kilometers for anadromous fish (St. Pierre 2003). In addition, some dams within the Atlantic sturgeon's range have been removed, including the Treat Falls Dam on the Penobscot River, Maine, and the Enfield Dam on the Connecticut River, Connecticut. In 1999, the Edwards Dam at the head-of-tide on the Kennebec River was removed, which restored 18 miles of Atlantic sturgeon spawning and nursery habitat and resulted in numerous sightings of large Atlantic sturgeon from Augusta to Waterville (Squires 2001).

Unfortunately, many waterways along the Atlantic coast host impoundments constructed during the Industrial Revolution that originally were a source of inexpensive power; many of these structures are no longer in use and should be removed (Erkan 2002).

Approach 1.1B.2: Installing or modifying fish passage facilities

Approach 1.1B.2A: For upstream passage

Approach 1.1B.2A.1: Fishways

Fish passage facilities, or fishways, allow fish to pass around an impoundment they would otherwise be unable to negotiate. Vertical slot fishways are commonly used to provide upstream access around dam structures. They are designed to draw fish away from the turbulent waters at the base of the dam toward the smooth flowing waters at the entrance of the fishway. Once fish enter the fishway, they negotiate openings, or vertical slots, in the baffle walls. Fish move from pool to pool as they advance up the fishway, using the pools as rest areas (VA DGIF 2006).

Another type of fishway is the fish ladder. Fish ladders consist of a series of baffles, or weirs, that interrupt the flow of water through the passage structure. As with vertical slot fishways, a series of ascending pools is created.

A third type of fishway, the Denil fishway, is the most common type in the northeast and reliably passes shad and river herring. In fact, construction of fish ladders in coastal streams of Maine resulted in rapid and noticeable increases in the number of adult alewife returning to these streams (Rounsefell and Stringer 1943).

It is important to note that although fish passage facilities are instrumental in restoring fish to historical habitat, they are not 100% efficient because some percentage of target fish will not find and successfully use the fishway (Weaver et al. 2003). At sites where bypass facilities are in place, but are inadequate, efficiency of upstream and downstream fish passage should be improved. Furthermore, passage facilities should be designed specifically for passing target species; some facilities constructed for species such as Atlantic salmon, have proven unsuitable for passing shad (Aprahamian et al. 2003).

Example: American shad

In 1999, a vertical slot fishway was opened at Boshers Dam on the James River, Virginia, ending nearly 200 years of blocked access to upstream areas. As a result, 221.4 km of historical spawning habitat on the main stem of the river and 321.9 km on tributaries was restored. By 2001, an increasing trend of relative abundance of American shad in the fall zone was strongly correlated with an increasing trend of American shad passage (Weaver et al. 2003).

Example: American eel

For American eel, upstream passage is facilitated with the use of ramps and substrates to take advantage of the anguillid's natural climbing abilities. A channel or ramp is provided with low water flow and substrate (gravel or nylon brushes or mesh) to assist the American eel elvers and yellow-phase individuals in passing an obstruction (Richkus and Whalen 1999; Solomon and Beach 2004).). In New Zealand, 250 m gravel-lined pipes have been used to pass anguillid eels over dams as

high as 68 m (Clay 1995). Portable elver passages may also be an option in some areas (Boubee and Barrier 1996; Wippelhauser et al. 1998). While the effectiveness of most designs has not been evaluated, there are several accounts of increased American eel biomass moving upstream after passes have been installed (Liew 1982; Clay 1995; Laffaille et al. 2005).

American eel passage is complicated by the fact that yellow-phase American eel require different substrates than elvers. Desrochers (1996) demonstrated an 85% efficiency rate of passage for migrating yellow-phase American eel, when approximately 450 mm of rods and tubes were placed on a ladder. The installation of tubes along the 58 m eel ladder at the Beauharnois Station on the St. Lawrence River resulted in a 77% ascent rate. Vegetation and matricial substrates proved ineffective for yellow-phase American eel (Desrochers 1996). Because the efficiency of passage of upstream migrants depends partly on the substrate size used in the ladder, it is important to determine the size and age of the American eel that are most likely to use the upstream passage for each river, as it will vary on a river-to-river basis (Richkus and Whalen 1999).

The stalling of fish at the exitways of ladders could potentially lower a ladder's efficiency (Verdon 1998; Richkus and Whalen 1999). Suitable exit passageways should be designed to assist with the exit and upstream migration of American eel, as well as to decrease the likelihood of fall-back entrainment (Verdon 1998; Richkus and Whalen 1999; Richkus and Dixon 2003).

Approach 1.1B.2A.2: Pipe passes

Pipe passes consist of a pipe below the water level that passes through a barrier. Substrate is provided in the pipe to decrease water velocity and to allow American eel to crawl through the pipe. Although this design creates a direct passage, it is flawed because the pipe often becomes blocked with debris, rendering it ineffective. Pipe passes are most efficient at the outflow of large impoundments that act as a sediment trap for debris so that water entering the pipe is clear of material that might cause a blockage (Solomon and Beach 2004).

Approach 1.1B.2A.3: Locks and lifts

For locks, fish swim into a lock chamber with an open lower gate. The gate periodically closes and the chamber is filled with water, bringing it up to level with the headpond. The upper gate is then opened and the fish swim out. This type of fish passage involves a great deal of engineering and can be expensive. This solution is ideal for very high head situations where conventional passes are impractical (Solomon and Beach 2004).

Alternatively, a lift involves a chamber that fish swim into. The chamber is lifted up to or above the head pond level and the fish swim out. The amount of lifts may also be important. At the Conowingo Dam on the Susquehanna River, a second fish lift has contributed to successfully restoring American shad to that system. Over 200,000 fish passed at both lifts in 2001, compared to 1990, when only 15,000 fish

returned (St. Pierre 2003). Moffitt et al. (1982) noted that blueback herring responded quite favorably to improved lift facilities at the Holyoke Dam on the Connecticut River, with passage increasing tremendously. Despite these improvements, stocks have declined considerably in recent years (R. St. Pierre, United States Fish and Wildlife Service retired, personal communication).

Approach 1.1B.2A.4: Easements

American eel often pass obstructions using irregularities in flow caused by edge effects, growth of algae and other plants, or features such as cracks and rubble. Providing these types of features is beneficial to areas where a full-scale engineering solution is not a viable option. Additionally, this enhancement is beneficial for sites with non-vertical barriers (weirs). It is both effective and inexpensive to maintain (Solomon and Beach 2004).

Approach 1.1B.2B: For downstream passage

Fish migrating downstream may pass through turbines, spillage, bypass facilities, or a combination of the three. One comparison between spillways and efficiently operated turbines found that the two systems were comparable in reducing fish mortality (Heisey et al. 1996).

Downstream passage of spent adult American shad through large turbines at the Safe Harbor project along the Susquehanna River, Pennsylvania, found that survival rate was 86% (NA and Skalski 1998). Survival rates would likely not be as favorable at facilities that employ smaller, high-speed turbines. Additional measures to help facilitate survival rates include controlled spills during peak migration months (St. Pierre 2003).

At some sites it is not desirable to move fish through turbines, so fish can be moved through a bypass facility. Creating a strong attraction flow helps guide fish to the bypass system and away from the intake flow areas of the turbines (Knights and White 1998; Verdon et al. 2003). Additionally, barrier devices can help deter fish away from flow intake areas. Barrier devices used to deter fish include lights, high-frequency sound, air bubble curtains, electrical screens, water jet curtains, and chemicals. Mechanical barrier devices include hanging chains, louvers, angled bars, and screens (Martin et al. 1994; Richkus and Whalen 1999; Richkus and Dixon 2003). Submerged strobe lights were found to be quite effective at directing fish away from turbines through a sluiceway (Martin et al. 1994).

Example: American eel

Studies of anguillid eels have stressed the importance of installing deterrents near the turbines and attraction mechanisms to direct the fish to the bypass facility (Hadderingh et al. 1992; Knights and White 1998; Verdon et al. 2003). Research suggests that light may be the best deterrent for American and European eels, with several studies demonstrating strong avoidance reactions to high intensity light

(Patrick et al. 1982; Hadderingh et al. 1992). One facility using underwater and overwater high intensity lights had a 51% deflection rate of yellow-phase anguillid eels and a 25% deflection rate of silver-phase anguillid eels (Hadderingh et al. 1992). Another facility on the St. Lawrence River created a "wall of light" by suspending 90 m long, surface to bottom 84 1000W halogen lamps in an area where the depth was 10 m. It was estimated that 85% of the downstream migrant American eel avoided the light (McGrath et al. 2003). Other researchers have found that silver-phase European eel showed no reaction to strobe lights (Adams and Schwevers 1997). Additional research is needed to find effective methods for deterring American eel from turbine entries (Richkus and Whalen 1999).

Example: Atlantic sturgeon

Atlantic sturgeon are not known to successfully use existing fish passageways (USFWS-NMFS 1998; Secor et al. 2002), but a lot of work has been done recently to identify promising upstream and downstream passage engineering designs for Atlantic sturgeon (P. Brownell, NOAA Fisheries, Southeast Regional Office, personal communication).

Approach 1.1B.3: Operational modifications

Hydroprojects operate more closely to the natural flow patterns of a stream when water moves through them with a fairly constant flow. Consequently, storage-release projects are more likely to alter both daily and seasonal flow patterns (Yeager 1995). Adjusting instream flows to more closely reflect natural flow regimes may help increase productivity of alosines, especially during summer to early fall when large, deep reservoirs stratify, and anoxic water releases are possible (McCord 2003).

Power generation can also be reduced, or ceased altogether, during prime downstream migration periods. This option might be cost-effective if migratory behavior coincides with off-peak rate schedules (Gilbert and Wenger 1996). Flows can be re-regulated at dams downstream of the primary dam to stabilize flows further downstream (Cushman 1985). Additionally, some studies have found that the most efficient operating flows for small turbines may not result in the best fish survival rates, but that operation at higher flows may pass fish more safely (Fisher et al. 1997).

Where hydrological conditions have been modified, additional measures can be implemented to help mitigate impacts on the river. For example, operational changes can be made to accomplish a number of improvements, such as reducing the upper limit of variability of one or more of the physical or chemical characteristics of the river. For example, incorporating turbine venting into major dams has proven useful for increasing dissolved oxygen concentrations. Alternatively, aerating reservoirs upstream of hydroelectric plants (Mobley and Brock 1996), as well as aerating flows downstream from the plants using labyrinth weirs and infuser weirs have also proven reliable for increasing the dissolved oxygen concentration in the water (Hauser and Brock 1994).

Example: Alosines

For alosines that migrate downstream during early evening hours, maintaining peak efficiency flows through selected turbines during these hours, as well as employing turbines that reduce mortality, may be effective (St. Pierre 2003).

Example: American eel

In simulation studies, Haro et al. (2003) showed that turbine mortality decreased significantly for American eel during days when there was substantial rainfall. Furthermore, the simulations showed that if power generation were suspended on days when eel catch was 25% to 75% of the total catch for all days, eel mortality was reduced by two-thirds to one-half relative to normal operations. Mortality was further halved when limits were set on generation using a combination of rainfall events and eel run timing factors (Haro et al. 2003).

Approach 1.1B.4: Streambank stabilization

States that have significant problems with streambank erosion have turned to stabilization to help further prevent erosion. Projects should maintain vegetated riparian buffers, making use of native vegetation wherever possible (MEOEA 2005). Habitat modification, including manipulating the cross-sectional geometry of the stream channel, may also serve to mitigate effects (Cushman 1985).

Example: Blueback herring

Loesch (1987) found that blueback herring responded favorably to changes in physical and hydrological conditions, becoming re-established and even increasing in abundance once favorable conditions were established or restored.

Approach 1.1B.5: Fish transfers

When populations have been extirpated from their habitat due to dam blockage, it may be necessary to transfer sexually mature pre-spawning adults or hatchery-reared fry and fingerlings above obstructed areas.

Example: American eel

Transplanting American eel may lead to a decrease in predation because the eels can disperse in the river instead of concentrating below the obstruction (Soloman and Beach 2004). Anguillid eels have also been successfully trapped upstream of a dam in New Zealand and released on the downstream side (Charles and Mitchell Associates 1995; Richkus and Whalen 1999).

Example: Alosines

Transplanting of fertilized alosine eggs has had limited success; eggs are now collected mostly for use in culture operations. Culture operations have focused primarily on American shad, and to a lesser degree blueback herring, alewife, and hickory shad (Hendricks 2003). Transplanting adult American shad, blueback herring, and alewife has been highly successful. Adult gravid shad can be trapped in the river where they originate, or other rivers, and trucked to upstream sites where they can be expected to spawn in areas that are otherwise not accessible. This may be an effective means for supplementing the river population until fish passage facilities are improved (both in the upstream and downstream direction), or fish passage facilities are constructed where they currently do not exist. As the return populations grow, further modifications may be necessary to accommodate larger runs (St. Pierre 1994).

For example, the release of hatchery-reared American shad in the James River, Virginia, in the mid-1990's, resulted in greater than 40% of hatchery-reared fish spawning several years later. This percentage greatly exceeded the percentage of the hatchery contribution (3 to 8%). If the offspring of hatchery-reared fish survive to reproduce, this should provide a significant boost to this severely depressed population (Olney et al. 2003).

At the Conowingo Dam on the Susquehanna River, Pennsylvania, 70 to 85% of the adult American shad returning from 1991 through 1995 were hatchery-reared. By 2003, the hatchery-to-wild ratio had been reversed, and naturally produced adults comprised 40 to 60% of returning fish (St. Pierre 2003).

Additionally, Maryland reported that over 80% of the 142 adults captured in the Patuxent and Choptank rivers in 2000 were of hatchery origin. It appears that shad stock enhancement, through the release of hatchery-reared fish, has proven to be beneficial when accompanied by other management measures including habitat restoration and water quality protection (Hendricks 2003).

Finally, pre-spawning adult American shad were taken from the Connecticut River and transplanted in the Pawcatuck River, Rhode Island, where they had been absent for 100 years. Six years later, in 1985, a population of over 4,000 fish existed (Gibson 1987).

Section 1.2: Road Culverts and Other Sources of Blockage

Issue 1.2A: Road culverts

While dams are the most common obstructions to fish migration, road culverts are also a significant source of blockage. Culverts are popular, low-cost alternatives to bridges when roads must cross small streams and creeks. Although the amount of habitat affected by an individual culvert may be small, the cumulative impact of multiple culverts within a watershed can be substantial (Collier and Odom 1989).

Roads and culverts can also impose significant changes in water quality. Winter runoff in some states includes high concentrations of road salt, while stormwater flows in the summer cause thermal stress and bring high concentrations of other pollutants (MEOEA 2005).

Example: Alosines

Sampled sites in North Carolina revealed river herring upstream and downstream of bridge crossings, but no herring were found in upstream sections of streams with culverts. Additional study is underway to determine if culverts are the cause for the absence of river herring in these areas (NCDENR 2000). Even structures only 20 to 30 cm above the water can block shad and river herring migration (ASMFC 1999).

Issue 1.2B: Other man-made structures

Additional man-made structures that may obstruct upstream passage include: tidal and amenity barrages; tidal flaps; mill, gauging, amenity, navigation, diversion, and water intake weirs; fish counting structures; and earthen berms (Durkas 1992; Solomon and Beach 2004). The impact of these structures is site-specific and will vary with a number of conditions including head drop, form of the structure, hydrodynamic conditions upstream and downstream, condition of the structure, and presence of edge effects (Solomon and Beach 2004).

Issue 1.2C: Natural barriers

Rivers can also be blocked by non-anthropogenic barriers, such as beaver dams, waterfalls, log piles, and vegetative debris. These blockages may be a hindrance to migration, but they can also be beneficial since they provide adhesion sites for eggs, protective cover, and feeding sites (Klauda et al. 1991). Successful passage at these natural barriers is often dependent on individual stream flow characteristics during the fish migration season.

THREAT #2: WATER WITHDRAWAL FACILITIES

Section 2.1A: Hydropower, Drinking Water, Irrigation, and Snow-making Facilities

Issue 2.1A.1: Impingement and entrainment

Large volume water withdrawals (e.g., drinking water, pumped-storage hydroelectric projects, irrigation, and snow-making), especially at pumped-storage facilities, can drastically alter local current characteristics (e.g., reverse river flow). Withdrawals may also alter other physical characteristics of the river channel, including stream width, depth, current velocity, substrate and temperature. This can cause delayed movement past the facility, or entrainment where the intakes occur (Layzer and O'Leary 1978). Planktonic eggs and larvae entrained at water withdrawal projects experience high mortality rates due to pressure changes, shear and mechanical stresses, and heat shock (Carlson and McCann 1969; Marcy 1973; Morgan et al. 1976). Well-screened facilities are unlikely to cause serious mortality to juveniles; however, large volume withdrawals can entrain significant numbers (Hauck and Edson 1976; Robbins and Mathur 1976).

Impingement of fish can trap them against water filtration screens, leading to asphyxiation, exhaustion, removal from the water for prolonged periods of time, or removal of protective mucous and descaling (DBC 1980).

For Example: American eel

Impingement and entrainment in water intakes and turbines have been identified as sources of mortality of seaward-migrating American eel at facilities in South Carolina (McCord 2005b).

Example: Alosines

Studies conducted along the Connecticut River found that larvae and early juveniles of alewife, blueback herring, and American shad suffered 100% mortality when temperatures in the cooling system of a power plant were elevated above 28°C; 80% of the total mortality was caused by mechanical damage and 20% was due to heat shock (Marcy 1976b). Ninety-five percent of the fish near the intake were not captured by the screen, and Marcy (1976b) concluded that it did not seem possible to screen fish larvae effectively. Results from earlier years led Marcy (1976a) to conclude that although mortality rates for eggs and larvae entrained in the intake system were very high, given the high natural mortality rate and the number of eggs produced by one adult shad, the equivalent of only one adult shad was lost during that study year as a result of egg and larval entrainment. Furthermore, there was no evidence that adult shad had changed the location of their spawning areas in the river as a result of plant operation (Marcy 1976a).

Another study of juvenile American shad emigrating from the Hudson River found that impingement at power plants was an inconsequential source of mortality; however, when added to other more serious stresses, it may possibly contribute to increased mortality rates (Barnthouse and Van Winkle 1988).

Example: Striped bass

Withdrawals or diversions of water can cause direct mortality of egg, larval, and juvenile striped bass due to entrainment (eggs, larvae, juveniles) or impingement (eggs, larvae, or juveniles depending upon the size of screen openings). Striped bass eggs may be able to survive impingement velocities up to 24 cm/s (0.8 ft/s) for 6 min, but test results have been highly variable (Skinner 1974). Survival of juvenile striped bass less than 40 mm was significantly affected at impingement velocities over 15 cm/s (0.5 ft/s) (Skinner 1974). Juveniles 40 to 50 mm long could withstand 6 minute periods at 24 cm/s (0.8 ft/s), but not 49 cm/s (1.6 ft/s). Skinner (1974) concluded that water velocity was a more important factor than time of exposure, but both were related to survival.

Kerr (1953) showed that 80% of striped bass 19 to 38 mm could avoid an impingement velocity of 30.5 cm/s (1 ft/s). However, only 5% of this size class could avoid 43 cm/s (1.4 ft/s). For juveniles 26 to 76 mm, 95% were able to avoid an impingement velocity of 61 cm/s (2 ft/s), and all juveniles 127 to 178 mm avoided 84 cm/s (2.7 ft/s) (Kerr 1953).

Issue 2.1A.2: Alteration of stream physical characteristics

Water withdrawals can also alter physical characteristics of streams, including: decreased stream width, depth, and current velocity; altered substrate; and temperature fluctuations (Zale et al. 1993). In rivers that are drawn upon for water supply, water is often released downstream during times of decreased river flow (usually summer). Additionally, failure to release water during times of low river flow and higher than normal water temperatures can cause thermal stress, leading to fish mortality. Consequently, water flow disruption can result in less freshwater input to estuaries (Rulifson 1994), which are important nursery areas for many anadromous species.

For Example: American shad

Cold water releases often decrease the water temperature of the river downstream, which has been shown to cause juvenile American shad to abandon their nursery areas (Chittenden 1969; 1972). At the Cannonsville Reservoir on the West Branch of the Delaware River, cold water releases from the dam resulted in the elimination of nursery grounds below the dam for American shad (DBC 1980).

Section 2.1B: Avoiding, Minimizing, and Mitigating Impacts of Water Withdrawal Facilities

Approach 2.1B.1: Use of technology and water velocity modification

Impacts resulting from entrainment can be mitigated to some degree through the use of the best available intake screen technology (ASMFC 1999), or through modifying water withdrawal rates or water intake velocities (Lofton 1978; Miller et al. 1982). Devices have also been used at hydroelectric projects to deter fish from intake flows, including: electrical screens, air bubble curtains, hanging chains, lights, high-frequency sound, water jet curtains, chemicals, visual keys, or a combination of these approaches (Martin et al. 1994). Promoting measures among industry that use reclaimed water, instead of freshwater from natural areas, can help reduce the amount of freshwater needed (FFWCC 2005). Location along the river was also found to be a significant factor affecting impingement rates in the Delaware River (Lofton 1978).

THREAT #3: TOXIC AND THERMAL DISCHARGES

Section 3.1A: Industrial Discharge Contamination

Issue 3.1A.1: Chemical effects on fish

Industrial discharges may contain toxic chemicals, such as heavy metals and various organic chemicals (e.g., insecticides, solvents, herbicides) that are harmful to aquatic life (ASMFC 1999). Many contaminants have been identified as having deleterious effects on

fish, particularly reproductive impairment (Safe 1990; Longwell et al. 1992; Mac and Edsall 1991). Chemicals and heavy metals can be assimilated through the food chain, producing sub-lethal effects such as behavioral and reproductive abnormalities (Matthews et al. 1980). In fish, exposure to polychlorinated biphenyls (PCBs) can cause fin erosion, epidermal lesions, blood anemia, altered immune response, and egg mortality (Post 1987; Kennish et al. 1992). Furthermore, PCBs are known to have health effects in humans and are considered to be human carcinogens (Budavari et al. 1989).

A number of common pollutants have been found to disturb the thyroid gland in fish, which plays a role in the maturation of oocytes. These chemicals include: lindane (organochlorine) (Yadav and Singh 1987); malathion (organophosphorus compound) (Lal and Singh 1987; Singh 1992); endosulfan (organochlorine) (Murty and Devi 1982); 2,3,7,8-PCDD and –PCDF (dioxin and halogenated furane); some PCBs (particularly 2,3,7,8-TCDD *para* and *meta* forms) (Safe 1990); and PAHs (polycyclic aromatic hydrocarbons) (Leatherland and Sunstegard 1977, 1978, 1980).

Steam power plants that use chlorine to prevent bacterial, fungal, and algal growth present a hazard to all aquatic life in the receiving stream, even at low concentrations (Miller et al. 1982). Pulp mill effluent and other oxygen-consuming wastes are discharged into a number of streams.

Example: Alosines

Lack of dissolved oxygen from industrial pollution and sewage discharge can greatly affect abundance of shad and prevent migration upriver or prevent adults from emigrating to sea and returning again to spawn. Everett (1983) found that during times of low water flow when pulp mill effluent comprised a large percentage of the flow, river herring avoided the effluent. Pollution may be diluted in the fall when water flow increases, but fish that reach the polluted waters downriver before the water has flushed the area will typically succumb to suffocation (Miller et al. 1982).

Effluent may also pose a greater threat during times of drought. Such conditions were suspected of interfering with the herring migration along the Chowan River, North Carolina, in 1981. In past years, the effluent from the pulp mill had passed prior to the river herring run, but drought conditions caused the effluent to remain in the system longer. Toxic effects were indicated, and researchers suggested that growth and reproduction may have been disrupted as a result of eutrophication and other factors (Winslow et al. 1983).

Even thermal effluent from power plants can have a profound effect on fish, causing disruption of schooling behavior, disorientation, and death. Researchers concluded that 30°C was the upper natural temperature limit for juvenile alosines (Marcy et al. 1972).

Example: Atlantic sturgeon

Due to their benthic feeding behavior and long life span, Atlantic sturgeon may be particularly sensitive to contaminants (USFWS-NMFS 1998). Although few studies have documented the impact of environmental contaminants on Atlantic sturgeon, PCBs have been detected in Atlantic sturgeon in the St. Lawrence and Hudson Rivers. Levels of PCBs in

these fish were above the upper limit set by the U.S. Environmental Protection Agency (EPA) for edible fish (Spagnoli and Skinner 1977). Other contaminants reported in Atlantic sturgeon include cadmium, mercury, and lead (Rehwoldt et al. 1978).

Example: American eel

Impacts to American eel from contamination include: impaired osmoregulation, direct mortality, decreased growth rate, reduced fecundity, and impaired reproductive success (Dutil et al. 1987; Brusle 1991, 1994; Castonguay et al. 1994a, b; Hodson et al. 1994; Knights 1997; Casselman et al. 1998; Haro et al. 2000; Robinet and Feunteun 2002).

American eel are particularly vulnerable to contamination by lipophilic compounds (Robinet and Feunteun 2002). Due to their high lipid content and benthic habitat preference, American eel have a high bioaccumulation rate (Couillard et al. 1997; Richkus and Whalen 1999). American eel might be a good indicator species for bioaccumulation studies (Van der Oost et al. 1988) because they transfer most of their somatic lipid stores to the gonads and gametes making them particularly sensitive to water contamination (Robinet and Feunteun 2002; Ashley et al. 2003). For example, in the St. Lawrence River, the highest concentrations of contaminants were found in the gonads of migrating silver eels. These chemical levels could be toxic to larvae, and since migrating females do not feed, the chemical levels in the eggs could be toxic at hatching (Hodson et al. 1994). In another instance, American eel exposed to fenithrothion (an organophosphorus insecticide) were found to have significantly lower fat contents (Sancho et al. 1998). Calow (1991) estimated that energy costs resulting from chemical stress have some minor consequences on growth and reproduction. These pesticides disturb fat accumulation, and are likely to reduce migration efficiency and breeding success in American eel.

Other suspected impacts from pesticides and contaminants include changes in behavior and migratory orientation, and performance and successful mating of American eel. These suspected threats have not yet been documented in the literature (Haro et al. 2000).

There is also some evidence that the contamination of eels may have an impact on the food chain. Studies of dead beluga whales (*Delphinapterus leucas*) in the estuary and the Gulf of St. Lawrence show levels of PCBs, DDT, pesticides, and mirex far above those found in Beluga whales in the Arctic (Massé et al 1986; Martineau et al. 1987). Marine mammals are exposed to contaminants by the prey they consume over their lifetime. The mirex found in the beluga whales is thought to be unique to the fish in the upper St. Lawrence River and Lake Ontario (Kaiser 1978; Castonguay et al. 1989). American eel contain 10 times as much mirex as estuarine fish in these areas. Thus, the eels may be a source of toxic chemicals to the beluga whales (Hodson et al. 1994).

Issue 3.1.2: Sewage effects on fish

Sewage can have direct and indirect effects on anadromous fish. Minimally effective sewage treatment during the 1960s and early 1970s may have been responsible for major phytoplankton and algal blooms in tidal freshwater areas of the Chesapeake Bay, which reduced light penetration (Dixon 1996), and ultimately reduced SAV abundance (Orth et al. 1991). Some of Massachusetts' large to mid-sized rivers receive raw sewerage into their

waters, and during summer low flows, are composed primarily of sewerage treatment effluent (MEOEA 2005).

Issue 3.1.3: Thermal effects on fish

Example: Striped bass

Reductions or increases in temperature as a function of industrial discharges or hydropower operations can affect spawning activity of striped bass. A sharp rise in temperature occurring during the spawning run may cause premature spawning in normally unsuitable areas (Farley 1966). Sudden drops in water temperature during the spawning run, or during the spawning act, have caused complete cessation of spawning activities (Calhoun et al. 1950; Mansueti and Hollis 1963; Boynton et al. 1977). Adult striped bass may overwinter in thermal discharge areas along the Atlantic coast, provided that they do not have to remain in the plume too long (Marcy and Galvin 1973).

Additionally, reductions or increases in temperature as a function of industrial discharges or hydropower operations can affect survival of eggs, larvae, and juveniles. Striped bass early life stages show significantly elevated mortality rates when exposed to rapid changes in water temperature (such as that in a thermal discharge plume) (Schubel et al. 1976). Eggs were able to sustain 15°C temperature elevation for 4 to 60 minutes, but an elevation of 20°C above acclimation temperature killed all eggs in 2 minutes. Yolk-sac larvae survival was significantly affected at a temperature elevation of 15°C. Furthermore, slightly lower temperature elevations, on the order of 10°C, significantly affected survival of 8 mm and 24 mm striped bass (Chadwick 1974). Mortality was not over 50% unless the absolute test temperature was 32.2°C or higher, regardless of the temperature elevation. Kelly and Chadwick (1971) presented 48 hour LC_{50} values from 30°C to 33°C for various acclimation temperatures.

Section 3.1B: Avoiding, Minimizing, and Mitigating Impacts of Toxic and Thermal Discharges

Approach 3.1B.1: Proper treatment of facility discharge

Although there has been a general degradation of water quality coastwide, the levels of sewage nutrients discharged into coastal waters during the past 30 years have decreased as a result of the Clean Water Act, passed in 1972. This has led to a decrease in organic enrichment, which has benefited water quality conditions. A reduction of other types of pollutant discharges into these waters, such as heavy metals and organic compounds, would not be expected (ASMFC 1999).

In many northern rivers, such as the Kennebec, Penobscot, Connecticut, Hudson, and Delaware Rivers, dissolved oxygen levels approached zero parts per million in the 1960s and 1970s. Since then, water quality has greatly improved as a result of better point-source treatment of municipal and industrial waste (USFWS-NMFS 1998). In 1974, secondary and tertiary sewage treatment was initiated in the Hudson River, which led to conditions where dissolved oxygen was greater than 60% saturation. There was a return of many fish species to

this habitat (Leslie 1988), including a high abundance of juvenile shortnose sturgeon (Carlson and Simpson 1987; Dovel et al. 1992).

Additionally, although poor water quality is often identified as a barrier to fish migration, it should be noted that poor water quality can be caused by both point and non-point sources of pollution. In fact, it may be difficult, if not impossible, for water quality standards to be achieved in some regions due to the effects of non-point sources of pollution (Roseboom et al. 1982).

Example: American shad

The estimated lost spawning habitat for American shad in 1898 was 5.28×10^3 river km, and in 1960 it was estimated at 4.49×10^3 km. The most recent estimate is now 4.36×10^3 river km. This increase in available habitat has been largely attributed to restoration efforts and enforcement of pollutant abatement laws (Limburg et al. 2003).

In compliance with the Clean Water Act, proper treatment of large city domestic sewage at treatment plants has dramatically improved the poor water quality conditions that persisted in the Delaware River for many years. Water quality problems were dramatically manifested in a "pollution block," including severely depressed levels of dissolved oxygen in the early 1900s in the Philadelphia/Camden area. There were very few repeat American shad spawners in this river, compared with other mid-Atlantic rivers (Miller et al. 1982). The situation had greatly improved by the late 1950s, due to a reduction in point-source pollution entering tidal waters, which led to an increase in dissolved oxygen by the 1980s (Maurice et al. 1987). This has led to a large enhancement of the American shad population in this river (Ellis et al. 1947; Chittenden 1969; Miller et al. 1982).

Similarly, improvements to water quality in the Potomac River in the 1970s led to increased water clarity and subsequently an increase in SAV abundance in 1983 (Dennison et al. 1993). In addition, pulp mill effluent was thought to have limited American shad survival in the Roanoke River (Walburg and Nichols 1967), but compliance with water quality standards in recent years has resulted in improved spawning habitat in this system (Hightower and Sparks 2003). Additional measures to improve habitat include reducing the amount of thermal effluent into rivers and streams, and discharging earlier in the year to reduce impacts to migrating fish (ASMFC 1999).

Example: American eel

While contaminated areas still exist in the United States, environmental toxin levels have decreased in many watersheds, which will potentially minimize the impacts on American eel. A decrease in American eel recruitment was not observed until well after the introduction of pollutants into watersheds, therefore contaminants cannot be the main cause of eel decline (Castonguay 1994a). Further research is needed to determine the impacts of these remaining contaminants on the eel population (Richkus and Whalen 1999).

THREAT #4: CHANNELIZATION AND DREDGING

Section 4.1A: Impacts of Dredging on Fish Habitat

Issue 4.1A.1: Primary environmental impacts of channelization

Channelization has the potential to cause significant environmental impacts (Simpson et al. 1982; Brookes 1988), including bank erosion, elevated water velocity, reduced habitat diversity, increased drainage, and poor water quality (Hubbard 1993). Dredging and disposal of spoils along the shoreline can also create spoil banks, which block access to sloughs, pools, adjacent vegetated areas, and backwater swamps (Frankensteen 1976). Dredging may also release contaminants resulting in bioaccumulation, direct toxicity to aquatic organisms, or reduced dissolved oxygen levels (Morton 1977). Furthermore, careless land use practices may lead to erosion, which can lead to high concentrations of suspended solids (turbidity) and substrate (siltation) in the water following normal and intense rainfall events. This can displace larvae and juveniles to less desirable areas downstream and cause osmotic stress (Klauda et al. 1991).

Spoil banks are often unsuitable habitat for fishes. Sand areas are an important nursery habitat to YOY striped bass. This habitat is often lost when dredge disposal material is placed on natural sand bars and/or point bars. The spoil is too unstable to provide good habitat for the food chain. Mesing and Ager (1987) found that electrofishing CPUE for gamefish was significantly greater on natural habitat than on "new (75%)," recent (66%)," or "old (50%)" disposal sites. Old sites that had not been disposed on for 5 to –10 or more years had not recovered to their natural state in terms of relative abundance of gamefish populations. The researchers also found that placement of rock material on degraded sand disposal sites had significantly greater electrofishing CPUE for sportfish than these sites had prior to placement of the rock material (Mesing and Ager 1987).

Example: Alosines

Draining and filling (or both) of wetlands adjacent to rivers and creeks in which alosines spawn, has eliminated spawning areas in North Carolina (NCDENR 2000).

Example: Striped bass

Reinert et al. (2005) published a case history documenting the impact of harbor modifications on striped bass in the Savannah River, Georgia, and South Carolina. During the 1980's, Savannah River striped bass suffered a population decline. The CPUE declined by 97% and egg production declined by 96%. Loss of freshwater spawning habitat through harbor modifications was identified as the primary cause (Reinert et al. 2005).

Spawning habitat deterioration in the St. Lawrence River was thought to be caused by construction of the St. Lawrence Seaway (1954-1959), island construction for the International World's Fair of 1967 in Montreal (1963-1964), and creation of Sterns Island from dredged sediments in 1965 (Beaulieu 1985).

Issue 4.1A.2: Secondary environmental impacts of channelization

Secondary impacts from channel formation include loss of vegetation and debris, which can reduce habitat for invertebrates and result in reduced quantity and diversity of prey for juveniles (Frankensteen 1976). Additionally, stream channelization often leads to altered substrate in the riverbed and increased sedimentation (Hubbard 1993), which in turn can reduce the diversity, density, and species richness of aquatic insects (Chutter 1969; Gammon 1970; Taylor 1977). Suspended sediments can reduce feeding success in larval or juvenile fishes that rely on visual cues for plankton feeding (Kortschal et al. 1991). Fish species that rely on benthic invertebrates within sediments may also experience decreased food availability if prey numbers are reduced. Sediment re-suspension from dredging can also deplete dissolved oxygen, and increase bioavailability of any contaminants that may be bound to the sediments (Clark and Wilber 2000).

Issue 4.1A.3: Impacts of channelization on fish physiology and behavior

Example: Alosines

Migrating adult river herring have been found to avoid channelized areas with increased water velocities. Several channelized creeks in the Neuse River basin in North Carolina have reduced river herring distribution and spawning areas (Hawkins 1979). Frankensteen (1976) found that the channelization of Grindle Creek, North Carolina removed in-creek vegetation and woody debris, which served as substrate for fertilized eggs.

Channelization can also reduce the amount of pool and riffle habitat (Hubbard 1993), which is an important food-producing area for larvae (Keller 1978; Wesche 1985). American shad postlarvae have been found concentrated in riffle-pool habitat (Ross et al. 1993).

Dredging can negatively affect alosine populations by producing suspended sediments (Reine et al. 1998), and migrating alosines are known to avoid waters of high sediment load (ASMFC 1985; Reine et al. 1998). It is also possible that fish may avoid areas where there is ongoing dredging due to suspended sediment in the water column. This was believed to have been the cause of a diminished return of adult spawning shad in a Rhode Island river, although no causal mechanism could be established (Gibson 1987). Filter-feeding fishes, such as alosines, can be negatively impacted by suspended sediments on gill tissues (Cronin et al. 1970). Suspended sediments can clog gills that provide oxygen, resulting in lethal and sub-lethal effects to fish (Sherk et al. 1974, 1975).

Nursery areas along the shorelines of the rivers in North Carolina have been affected by dredging and filling, as well as by erection of bulkheads; however, the degree of impact has not been measured. In some areas, juvenile alosines were unable to enter channelized sections of a stream due to high water velocities caused by dredging (ASMFC 2000b). Despite findings by Miller et al. (1982) that the effects of river dredging on fish populations were insignificant, they suspected that migrating juvenile shad could potentially be impacted by increased suspended solids, lowered dissolved oxygen concentration, and release of toxic materials.

Example: American eel

Dredging can also entrain seaward-migrating adult American eel, increase turbidity or suspended sediments that may negatively affect migrating adults, glass eels, and elvers, and cause changes in salinity regimes that could impact eel distribution and prey availability (McCord 2005b; ASMFC 2000).

Example: Atlantic sturgeon

Some studies have noted that dredging and filling operations alter habitat characteristics important to Atlantic sturgeon, including: disturbance of benthic flora and fauna; elimination of deep holes through establishment of uniform depth profiles; alteration of the rock substrate; and increased sedimentation (Smith and Clugston 1997; IAN 1999; Stein et al. 2004). Indirect impacts include destruction of feeding areas, disruption of migrations, and re-suspension of sediments in the spawning habitat (USFWS-NMFS 1998; Bushnoe et al. 2005). Siltation from dredging can also reduce spawning success by smothering eggs and covering suitable substrates for adhesive eggs (USFWS-NMFS 1998).

Dredging, and the removal of a rock outcropping, in the Rocketts, James River, Virginia, may have destroyed historic Atlantic sturgeon spawning grounds. Before dredging occurred in this area, Rocketts had substrate that was the exact configuration of known spawning sites (Bushnoe et al. 2005). Other studies have found no direct link between dredging and impacts to Atlantic sturgeon habitat, but it has been cited as a potential threat to the recovery of the species (Beamesderfer and Farr 1997; USFWS-NMFS 1998; Caron and Tremblay 1999; Collins et al. 2000; Bushnoe et al. 2005).

In addition to altering habitat, mechanical and hydraulic dredging can cause physical harm to Atlantic sturgeon by entraining fish through the drag arms and impeller pumps. Mortalities of this nature have been documented in King's Bay (Georgia) and Charleston (South Carolina) (M. Collins, South Carolina Department of Natural Resources, personal communication), and in the Cape Fear River (North Carolina) (USFWS-NMFS 1998). Dredging operations can also potentially have an impact on larval sturgeon. Veshchev (1981) documented the impact of dredges on *Acipenser guldenstadti* and *A. stellatus* larvae in the Volga River, Russia. He found that dredging caused 68.0% to 76.8% mortality of the total larvae caught upstream of the suction unit. Veshchev (1981) reported that 1,000 larva were destroyed by the dredges. Khodorevskaya (1972) also recorded the entrainment of fingerling sturgeon by dredges.

Example: Striped bass

Larval striped bass consumed 40% less prey when suspended solids exceeded 200 mg/L (Breitburg 1988).

Issue 4.1A.4: Increase in boat strikes

Example: Atlantic sturgeon

Dredging creates areas of safe passage for ships and boats. Increased boating traffic is likely to occur in dredged areas, which in turn may increase the risk of Atlantic sturgeon propeller strikes. To date, there is only one documented case of propeller strike mortality to Atlantic sturgeon in the Delaware River (USFWS-NMFS 1998), however, Delaware Fish and Wildlife staff consistently find adult and juvenile Atlantic sturgeon that wash ashore in the Delaware River during the spring or historical spawning season with obvious propeller wounds (C. Shirey, Delaware Division of Fish and Wildlife, personal communication). In May 2005, there were six confirmed reports of dead Atlantic sturgeon in the Delaware River, three of which had obvious external injuries. Dead sturgeon ranged from juvenile to adult, with one fish aged at 45 to 49 years (G. Murphy, Delaware Division of Fish and Wildlife, personal communication). The Delaware River may pose a bigger collision threat than other areasdue to the high volume of ship traffic and the narrow, shallow nature of the port (D. Fox, Delaware State University, personal communication).

Boat strikes do not appear to be an issue in the Hudson River because the channel is not routed through prime spawning habitat (J. Mohler, U.S. Fish and Wildlife Service, personal communication). Little is known about the extent of sturgeon mortalities in other areas due to ship strikes and more research is needed in this area.

Section 4.1B: Avoiding, Minimizing, and Mitigating Impacts of Channelization

Approach 4.1B.1: Seasonal restrictions and proper material disposal

Dredging restrictions are already in place in many rivers including the Kennebec, Connecticut, Cape Fear, Cooper, and Savannah Rivers (USFWS-NMFS 1998), to help curtail the impacts of dredging to anadromous fish. Seasonal restrictions on dredging in areas where anadromous fish are known to occur should be established until there is irrefutable evidence that dredging does not restrict the movement of fish (Gibson 1987). It is recommended that dredge material be disposed of in the most ecologically beneficial way possible that will prevent harm to existing natural habitats (FFWCC 2005).

THREAT #5: LAND USE CHANGE

The effects of land use and land cover on water quality, stream morphology, and flow regimes are numerous, and may be the most important factors determining quantity and quality of aquatic habitats (Boger 2002). Studies have shown that land use influences dissolved oxygen (Limburg and Schmidt 1990), sediments and turbidity (Basnyat et al. 1999; Comeleo et al. 1996), water temperature (Hartman et al. 1996; Mitchell 1999), pH (Osborne and Wiley 1988; Schofield 1992), nutrients (Basnyat et al. 1999; Osborne and Wiley 1988; Peterjohn and Correll 1984), and flow regime (Johnston et al. 1990; Webster et al. 1992).

Siltation, caused by erosion due to land use practices, can kill submerged aquatic vegetation (SAV). SAV can be adversely affected by suspended sediment concentrations of less than 15 mg/L (Funderburk et al. 1991) and by deposition of excessive sediments (Valdes-Murtha and Price 1998). SAV is important because it improves water quality (Rybicki and Hammerschlag 1991), and provides refuge habitat for migratory fish and planktonic prey items (Maldeis 1978; Killgore et al. 1989; Monk 1988).

Section 5.1A: Agriculture

Issue 5.1A.1: Sedimentation and irrigation

Decreased water quality from sedimentation became a problem with the advent of land-clearing agriculture in the late 18th century (McBride 2006). Agricultural practices can lead to sedimentation in streams, riparian vegetation loss, influx of nutrients (e.g., inorganic fertilizers and animal wastes), and flow modification (Fajen and Layzer 1993). Agriculture, silviculture, and other land use practices can lead to sedimentation, which reduces the ability of semi-buoyant eggs and adhesive eggs to adhere to substrates (Mansueti 1962).

In addition, excessive nutrient enrichment stimulates heavy growth of phytoplankton that consume large quantities of oxygen when they decay, which can lead to low dissolved oxygen during the growing season (Correll 1987; Tuttle et al. 1987). Such conditions can lead to fish kills during hot summer months (Klauda et al. 1991).

Another factor, chemical contamination from agricultural pesticides, has a significant potential to impact stream biota, especially aquatic insects, but is difficult to detect (Ramade et al. 1984).

Furthermore, irrigation can cause dewatering of freshwater streams, which can decrease the quantity of both spawning and nursery habitat for anadromous fish. Dewatering can cause reduced water quality as a result of more concentrated pollutants and/or increased water temperature (ASMFC 1985).

Example: American eel

American eel habitat may be further reduced in areas that already have poor water quality where dewatering only exacerbates the problem (McCord 2005b).

Example: River herring

Uzee (1993) found that in some Virginia streams, there was an inverse relationship between the proportion of a stream's watershed that was agriculturally developed and the overall tendency of the stream to support river herring runs. In North Carolina, cropland alteration along several creeks and rivers has significantly reduced river herring distribution and spawning areas in the Neuse River basin (Hawkins 1979).

Issue 5.1A.2: Nutrient loading

Atmospheric nitrogen deposition in coastal estuaries of states such as North Carolina, has had an increasingly negative effect on coastal waters, leading to accelerated algal production (or eutrophication) and water quality declines (e.g., hypoxia, toxicity, and fish kills). The primary source of atmospheric nitrogen in these areas comes from livestock operations and their associated nitrogen-rich (ammonia) wastes, and to a lesser degree, urbanization, agriculture, and industrial sources (Paerl et al. 1999). Animal production farms have greatly contributed to deteriorating water quality in other areas, including the Savannah, Ogeechee, and Altamaha Rivers (Georgia), and the Chesapeake Bay (USFWS-NMFS 1998; Collins et al. 2000; McBride 2006).

From the 1950s to the present, increased nutrient loading has made hypoxic conditions more prevalent (Officer et al. 1984; Mackiernan 1987; Jordan et al. 1992; Kemp et al. 1992; Cooper and Brush 1993; Secor and Gunderson 1998). Hypoxia is most likely caused by eutrophication, due mostly to non-point source pollution (e.g., industrial fertilizers used in agriculture) and point source pollution (e.g., urban sewage).

Example: Atlantic sturgeon

Eutrophic conditions pose a serious threat to Atlantic sturgeon because the species does not have the physiological or behavioral ability to cope with hypoxic conditions (Niklitschek 2001; Secor and Niklitschek 2001), and oxygen squeezes can cause direct mortality. Reduced dissolved oxygen levels are thought to be the cause of extirpation of Atlantic sturgeon populations in the St. Mary's River, Georgia (USFWS-NMFS 1998; Collins et al. 2000). Furthermore, degraded habitat in southern estuaries may have contributed to decreased spawning populations of juvenile shortnose sturgeon (Collins et al. 2000). Particularly, summer nursery habitats for juvenile Atlantic sturgeon are at risk from water quality deterioration, specifically hypoxic conditions (Secor and Gunderson 1998; Secor et al. 2000; Secor and Niklitschek 2002; Niklitschek and Secor 2005).

Issue 5.1A.3: Hypoxia

Example: Atlantic sturgeon

Niklitschek and Secor (2005) evaluated how temperature, dissolved oxygen, and salinity influence Atlantic sturgeon production in the Chesapeake Bay. They determined that summer was the most critical season for Atlantic sturgeon, and that low tolerance for high temperatures (greater than 28ºC) is a limiting factor during the first two summers of life. Using models, Niklitschek and Secor (2005) predicted that as temperatures increase to sub-lethal levels in the summer, YOY would utilize deeper and cooler waters as a thermal refuge. However, in the Chesapeake Bay, deeper and cooler areas are located down-estuary and do not have suitable salinities for early juvenile Atlantic sturgeon, which are restricted to lower salinity regions. This model also predicted that summer habitat would include 0 to 35% of the modeled bay area, but in drought years almost no summer habitat was available for juvenile sturgeon. Niklitschek (2001) found that summer hypoxic conditions and high temperatures in the mainstem and tidal sections of the tributaries of the Chesapeake Bay caused habitat fragmentation and restricted usable habitat to a small portion of the bay. The

total area of suitable habitat under average July conditions corresponded to 1586 km^3 and 1076 km^3, which was only 8.5% and 5.8%, respectively, of the total surface area of the mainstem and tidal sections of the tributaries (Niklitschek 2001).

Hypoxic conditions in Narragansett Bay (Rhode Island), Chesapeake Bay (Virginia and Maryland); Cape Fear River, Neuse River estuary, and Pamlico Sound (North Carolina), and the Savannah and Cooper Rivers (Georgia) threaten juvenile sturgeon (Mallin et al. 1997; Leathery 1998; Collins et al. 2000; Secor and Niklitschek 2001; C. Powell, formerly Rhode Island Division of Fish and Wildlife, personal communication). A secondary threat is mortality to their benthic prey organisms (McBride 2006; W. Laney, U.S. Fish and Wildlife Service, personal communication). Evidence of the effects of hypoxia on sturgeon populations remains circumstantial, but trends show that hypoxia may affect populations, and spawning is absent in many estuaries where hypoxic conditions prevail (Collins et al. 2000).

Section 5.1B: Avoiding, Minimizing, and Mitigating Agricultural Impacts

Approach 5.1B.1: Erosion control and best management practices

Erosion control measures and best management practices (BMPs) can reduce sediment input into streams, which can reduce the impact on aquatic fauna (Lenat 1984; Quinn et al. 1992). Agricultural BMPs may include: vegetated buffer strips at the edge of crop fields, conservation tillage, strip cropping, diversion channels and grassed waterways, soil conservation and water quality planning, nutrient management planning, and installing stream bank fencing and forest buffers. Animal waste management includes: manure storage structures, runoff control for barnyards, guttering, and nutrient management (ASMFC 1999). Programs to upgrade wastewater treatment at hog and chicken farms should be promoted (NC WRC 2005). Additionally, restoring natural stream channels and reclaiming floodplains in areas where the channel or shoreline has been altered by agricultural practices can help mitigate impacts (VA DGIF 2005).

Example: Atlantic sturgeon

Improved water quality in the Hudson River, New York, has accompanied a recovery in shortnose sturgeon populations (Secor and Niklitschek 2001). Models designed to address how to meet new EPA dissolved oxygen criteria for the Chesapeake Bay found that achieving the EPA dissolved oxygen criteria would increase habitat by 13% per year and that an increase in temperature by 1ºC would reduce habitat by 65%. The models were used to help identify four areas in the Chesapeake Bay that require special consideration to aid in the restoration of Atlantic sturgeon (Niklitschek and Secor 2005).

Section 5.2A: Logging/Forestry

Issue 5.2A.1: Logging

Logging activities can modify hydrologic balances and instream flow patterns, create obstructions, modify temperature regimes, and input additional nutrients, sediments, and toxic substances into river systems. Loss of riparian vegetation can result in fewer refuge areas for fish from fallen trees, fewer insects for fish to feed on, and reduced shade along the river, which can lead to increased water temperatures and reduced dissolved oxygen (EDF 2003). Potential threats from deforestation of swamp forests include: siltation from increased erosion and runoff; decreased dissolved oxygen (Lockaby et al. 1997); and disturbance of food-web relationships in adjacent and downstream waterways (Batzer et al. 2005).

In South Carolina, forestry BMPs for bottomland forests are voluntary. When BMPs are not exercised, plant material and disturbed soils may obstruct streams, excessive ruts may force channel-eroded sediments into streams, and partially stagnated waters may become nutrient-rich, which can lead to algal growth. These factors contribute to increased water temperature and reduced dissolved oxygen (McCord 2005c).

Example: Striped bass

For striped bass, warmer water temperatures may decrease the amount of summertime refuge habitat, which can negatively impact reproduction (Sessions et al. 2005).

Example: Atlantic sturgeon

In many systems, like the Chesapeake Bay, hard substrate has been buried under sediments resulting from erosion caused by deforestation, agriculture, and urbanization (Secor et al. 2002; Bushnoe et al. 2005). For the past two centuries, the hard substrate used by Atlantic sturgeon for spawning purposes has been lost from burial by sedimentation and siltation (Secor et al. 2000). Lack of suitable spawning substrate has been cited as one of the limiting factors for Atlantic sturgeon in the Chesapeake Bay (USFWS-NMFS 1998).

Section 5.2B: Avoiding, Minimizing, and Mitigating Logging Impacts

Approach 5.2B.1: Best management practices

Virginia advocates working with private, small foresters to implement forestry BMPs along rivers to reduce the impacts of forestry practices (VA DGIF 2005). Florida discourages new bedding on public lands where there is healthy groundcover (FFWCC 2005).

Section 5.3A: Urbanization and Non-Point Source Pollution

Issue 5.3A.1: Pollution impacts on fish and fish habitat

Urbanization can cause elevated concentrations of nutrients, organics, or sediment metals in streams (Wilber and Hunter 1977; Kelly and Hite 1984; Lenat and Crawford 1994). Recent studies conducted in Charleston Harbor, South Carolina, found that crustacean prey of estuarine fishes are directly affected by urbanization and related water quality parameters, including concentrations of a variety of toxicants (especially petroleum-related materials) (EDF 2003). Furthermore, the amount of developed land may influence use of a habitat, but other factors such as size, elevation, and habitat complexity are important as well, and in some cases may outweigh the negative effects of development (Boger 2002). More research is needed on how urbanization affects diadromous fish populations.

Example: Alewife

One study found that when the percent of land in areas increased to about 10% of the watershed, the number of alewife egg and larvae decreased significantly in tributaries of the Hudson River, New York (Limburg and Schmidt 1990).

Example: American eel

Machut (2006) found that American eel density and condition were negatively affected by urbanization in a tributary of the Hudson River. American eel from this tributary also had a higher parasite load (Machut 2006).

Section 5.3B: Avoiding, Minimizing, and Mitigating Impacts of Urbanization and Non-Point Source Pollution

Approach 5.3B.1: Best management practices

Urban BMPs include: erosion and sediment control; stormwater management; septic system maintenance; and forest buffers (ASMFC 1999). Siting stormwater treatment facilities on upland areas is recommended where possible (FFWCC 2005). Wooded buffers and conservation easements should be established along streams to protect critical shoreline areas (ASMFC 1999), and low impact development should be implemented, where practicable (NCWRC 2005).

Example: Alosines

Since the abundance of SAV is often used as an indirect measure of water quality, and there is a correlation between water quality and alosine abundance, steps should be taken to halt further reduction of underwater sea grasses (especially important in the Chesapeake Bay) (B. Sadzinski, Maryland Department of Natural Resources, personal communication).

Regarding cumulative effects on river herring spawning habitat, Boger (2002) suggested that land use and morphology within the entire watershed should be considered, and that the cumulative effects within the entire watershed may be as important as the type of land use within buffer zones. This is an important point to consider when establishing required widths of buffer zones in an effort to balance anthropogenic activities in the watershed and maintain biological integrity of streams (Boger 2002).

THREAT #6: ATMOSPHERIC DEPOSITION

Section 6.1A: Atmospheric Deposition

Issue 6.1A.1: Acid rain and low pH

Atmospheric deposition occurs when pollutants are transferred from the air to the earth's surface. This occurrence inputs a significant source of pollutants to many water bodies. Pollutants can get from the air into the water through rain and snow, falling particles, and absorption of the gas form of the pollutants into the water. Atmospheric deposition that causes low pH and elevated aluminum (acid rain) can contribute to changes in fish stocks. When pH declines, the normal ionic salt balance of the fish is compromised and fish lose body salts to the surrounding water (Southerland et al. 1997).

Example: American shad

American shad stocks that spawn in poorly buffered Eastern Shore Maryland rivers, like the Nanticoke and Choptank, were found to be vulnerable to storm-induced, toxic pulses of low pH and elevated aluminum. These stocks, therefore, may recover at a much slower rate than well-buffered Western Shore stocks, even if all other anthropogenic stressors are removed (Klauda 1994; ASMFC 1999). Streams often experience their highest levels of acidity in the spring, when adult shad are returning to spawn (Southerland et al. 1997).

There is speculation that recent precipitous declines in American shad populations may partly be due to acid rain (Southerland et al. 1997). Fertilized eggs, yolk-sac larvae, and to a lesser degree, young feeding (post yolk-sac) larvae of American shad have the highest probability for exposure to temporary episodes of pH depressions and elevated aluminum levels in, or near, freshwater spawning sites (Klauda 1994). Klauda (1994) suggests that even infrequent and temporary episodes of critical or lethal pH and aluminum exposures in the spawning and nursery areas could contribute to significant reductions in egg or larval survival of American shad and thereby slow stock recovery. Juvenile fish are more susceptible to the effects of low pH, which may effectively prevent reproduction (Klauda 1994).

Threats may be seasonal, ongoing, or even sporadic, all of which can have long-term effects on the recovery of stocks. For example, Hurricane Agnes in 1972 is suspected of causing the 1972 year-class failure for American shad, hickory shad, alewife, and blueback herring, as well as altering many spawning habitat areas in the Chesapeake Bay. Almost

twenty years later, these impacts were suggested to be contributing to the slow recovery of stocks in this area (Klauda et al. 1991).

Section 6.1B: Avoiding, Minimizing, and Mitigating Impacts of Atmospheric Deposition

Approach 6.1B.1: Reduction of airborne chemicals

Supporting the reduction of airborne chemical releases from power plants, paper mills, and refineries is one way to decrease the levels of toxins in the air that eventually settle into riverine habitat. Incentives can be promoted at the state level and through cooperative interstate agreements (FFWCC 2005).

THREAT #7: REDUCED DISSOLVED OXYGEN

Section 8.1A: Reduced Dissolved Oxygen

Issue 8.1A.1: Hypoxia and anoxia

Dissolved oxygen is a fundamental requirement for all aquatic life (Summers 2001). Many states have set a threshold concentration of 4 to 5 ppm as their water quality standard. Concentrations below approximately 2 ppm are stressful to many estuarine organisms (Diaz and Rosenberg 1995; Coiro et al. 2000). Eutrophication associated with urbanization (see Threat #5 above) can lead to reduced levels of dissolved oxygen in habitats used by diadromous juveniles and adults. Many riverine and estuarine habitats occupied by migratory diadromous fish are experiencing hypoxia (reduced oxygen) and/or anoxia (absence of oxygen) more often, and over more extensive areas, than in the past (Coutant and Benson 1990; Summers 2001; Bales and Walters 2003; Chesapeake Bay Foundation 2004; Summers 2004).

For example, Summers (2001) reported that in Long Island Sound, low dissolved oxygen occurs primarily during the summer months in the central and western portions. The water in Long Island Sound tends to be highly stratified in the late summer months and has probably always experienced some periods of low dissolved oxygen. However, human inputs of nutrients have added to the problem, resulting in more significant damage to ecologically and economically important organisms. A time series of average dissolved oxygen concentrations in Long Island Sound shows generally decreasing measurements from 1963 to 1993. Conditions appeared to improve from 1987 to 1993, but remained substantially degraded with respect to measurements made prior to 1970. The number of days for which conditions were hypoxic (below 3 ppm) ranged from 35 in 1995 and 1996, to 82 in 1989 (Summers 2001).

Although overall estuarine oxygen levels are reported in the National Coastal Condition Reports (Summers 2001, 2004) as "good" coastwide, both reports note that levels in a number of East Coast estuaries are problematic. Coastwide "good" conditions are

defined as meaning that less than 5% of coastal waters have "poor" dissolved oxygen concentrations (i.e., less than 2 ppm). "Fair" dissolved oxygen conditions coastwide mean that only 5% to 15% of the coastal waters have poor dissolved oxygen, and "poor" means that more than 15% of the coastal waters have poor dissolved oxygen concentrations. Specific estuaries mentioned as having dissolved oxygen problems were the Neuse River estuary, Chesapeake Bay, and Long Island Sound (Summers 2001, 2004).

A recent review of dead zones and their consequences for estuarine and marine ecosystems was conducted by Diaz and Rosenberg (2008). The researchers indicated that such dead zones in estuaries and the coastal oceans have, "...spread exponentially since the 1960's and have serious consequences for ecosystem functioning." Dead zones have formed from the increase in primary production, and consequent worldwide eutrophication, fed by riverine runoff of fertilizers and the combustion of fossil fuels. Enhanced primary production results in an accumulation of particulate organic matter, which facilitates microbial activity and the consumption of oxygen in bottom waters. Diaz and Rosenberg (2008) compiled data on dead zones globally, and documented about 101 dead zones on the East Coast of the United States, within ASMFC jurisdiction. Each Atlantic coastal state, with the exception of Pennsylvania, had from one (e.g., in Connecticut and New Hampshire) to as many as 24 (e.g., in South Carolina) dead zones. On a positive note, some of the documented dead zones were historic and have been alleviated through positive management actions (Diaz and Rosenberg 2008).

Diaz and Rosenberg (2008) also defined the degrees of hypoxia present. The most common form, *seasonal hypoxia*, occurs once per year in the summer after spring blooms, when water is warmest and stratification strongest, and usually lasts until autumn. Seasonal hypoxia is responsible for about one-third (33 of 101) of the dead zones on the East Coast. The usual ecosystem response to seasonal oxygen depletion is mortality of benthic organisms, followed by some level of recolonization when normal conditions return (Diaz and Rosenberg 2008).

Diaz and Rosenberg (2008) noted that *periodic hypoxia* was reported in almost half (46 of 101) of the East Coast dead zones. Periodic hypoxia might occur more often than seasonally, but tends to be less severe, lasting from days to weeks. Many smaller systems, such as the York River tributary to the Chesapeake Bay, experience this form of hypoxia when local weather events and spring neap-tidal cycles influence stratification intensity (Diaz and Rosenberg 1995).

Other causes of hypoxia are: 1) diel cycles, which influence production and respiration, and cause hypoxia that lasts only hours on a daily basis (Tyler and Targett 2007); and 2) wind and tides influencing areas on the margins of seasonal dead zones (Breitburg 2002). Diaz and Rosenberg (2008) noted that these are known as *episodic hypoxia* events, and are infrequent. Episodic hypoxia might occur less than once per year, sometimes with years lapsing between events. Episodic oxygen depletion is the initial sign that a system has reached the critical point of eutrophication, which in combination with stratification of the system tips it into hypoxia. Fifteen of the East Coast dead zones (or 14.8%) documented by Diaz and Rosenberg (2008) were classified as episodic.

Lastly, *persistent hypoxia* can occur where systems are prone to persistent stratification. None of the dead zones on the East Coast were classified as persistent (Diaz and Rosenberg 2008).

Example: Striped bass

The Neuse River estuary, Chesapeake Bay, and Long Island Sound are important nursery and foraging areas for migratory striped bass juveniles and adults. Eileen Setzler-Hamilton and Lenwood Hall, Jr. (1991) wrote that, "There is increasing concern that low dissolved oxygen in the deeper waters of the upper Chesapeake Bay and its tributaries has eliminated much of the summer habitat for sub-adult and adult striped bass." Those words were written seventeen years ago and remain just as true today.

Striped bass of all ages avoid waters with dissolved oxygen concentrations less than 3 to 4 mg/L. From 1984 through 1987, there was no suitable habitat (defined as water with temperature below 25°C and dissolved oxygen above 2 to 3 mg/L) for striped bass remaining in late July in the north-central segments of the Chesapeake Bay (Coutant and Benson 1990). Additionally, CBF (2004) reported that during the summer of 2004, approximately 35% of the water in the mainstem Chesapeake Bay had unhealthy dissolved oxygen levels for many forms of aquatic life, including striped bass. A "dead zone" with dissolved oxygen concentrations of less than 2 mg/L extended from off the mouth of the Rappahannock River in Virginia, to well above the Patuxent River in Maryland. Most Chesapeake Bay-associated rivers (e.g., York, Rappahannock, Potomac, and Patuxent, among others) experienced similar problems in their lower reaches at that same time (CBF 2004).

The primary cause of reduced oxygen in Chesapeake Bay and its tributary rivers was stated as nitrogen pollution that fueled large algal blooms, which in turn were decayed by oxygen-consuming bacteria. In particular, during 2004, algal blooms early in the year caused dissolved oxygen levels well below average at many locations in the Bay during February and March. While the overall size of the 2004 dead zone was smaller than the historic one of 2003, the volume of anoxic water was greater (CBF 2004).

Similarly, Wiley and Tsai (1990) reported that the Broomes Island area of the Patuxent River was no longer acceptable summer habitat for striped bass. White perch, hogchokers, and striped bass dominated monthly trawl catches at Broomes Island from 1965 through 1968, but by 1988 and 1989, striped bass were caught rarely. Eutrophication and the resulting increase in hypoxic bottom waters were stated as the probable causes of the deterioration of this summer habitat (Wiley and Tsai 1990).

THREAT #8: GLOBAL WARMING

Section 8.1A: Global Warming

In a demonstration of great foresight, the American Fisheries Society held a symposium on the effects of climate change on fish during the 1988 annual meeting (Regier et al. 1990).

The conveners noted that, "there is growing consensus that climate change will result from the continuing buildup of heat-trap gases in our atmosphere." They further noted that, "...efforts to adapt the scientific method to forecast some potential effects of climate change on fish and fisheries...," were well along and reflected by the symposium papers (Reiger et al. 1990).

Regier et al. (1990) perceived three types of causal or relational connections between atmospheric phenomena and fish in hydrological systems, including: 1) quite direct ecological pathways from the local climate to the local stock or association of fish; 2) more general, looping ecosystemic pathways involving linkages between climatological, hydrological, and biotic subsystems; and 3) even more complex pathways that involve, in addition to the above, human activities that change with climate change and the effects of those cultural changes on the natural parts of our ecosystems and biosphere (Regier et al. 1990).

The emphasis in the symposium papers was mostly on some simpler examples of the second type of causal connection. Due to the fact that fish live in water, and water temperature is a complex function of multiple factors, researchers thought that fish might offer relatively few instances of simple direct connections of the first type. As yet, they reported, few scientists knowledgeable about fish had given thought to examples of the third type; however, they did note that Coutant (1981) had suggested that a major regional effect could occur through the construction of water-storage reservoirs in areas of increased aridity (Regier et al. 1990). That prediction appears to be coming true in the wake of several recent prolonged and severe drought years, at least in the southeastern United States (W. Laney, personal observation).

In a recent study by Lassalle et al. (2008) using models to predict diadromous species distributions, the researchers found that temperature was the most explanatory variable in six of the twenty-one individual species models. In addition, longitude was the most explanatory variable in fifteen of the other species. The researchers claimed that their models could be used to predict changes in species distribution under global warming conditions. They used Allis shad (*Alosa alosa*), which is a declining European species, as an example. Biogeographical history proved to be an important component in the evaluation of these models. The researchers noted that the models could be used to predict whether a particular area would be suitable for restoration under global warming scenarios, and thought models should be used as a decision support tool to assess the suitability of conservation units (Lassalle et al. 2008).

Issue 8.1A.1: Habitat modifications

Coutant (1990) noted that a fish population's "habitat" could be defined as the volume of water that provides suitable conditions over time for sustained high performance, linked to the physiological performance of fishes under different environmental regimes. As climate changes occur, modification of such habitat is expected in local environments. Such modifications could result in changes in large-scale distribution patterns for fish species, and consequent changes in the thermal niche space available. As noted by Coutant (1990), the linkage between fish production and thermal niche space is confounded when the habitat is made unsuitable by a low dissolved oxygen concentration.

Example: Striped bass

The implications of continued global habitat warming and predicted outcomes for Atlantic migratory striped bass were discussed in detail by Coutant (1990). Based on the results of environmental change scenarios produced by two general circulation models (U.S. National Aeronautics and Space Administration's Goddard Institute for Space Studies model, GISS, and Princeton University's Geophysical Fluid Dynamics Laboratory model, GFDL) that each assume a doubling of carbon dioxide, Coutant (1990) predicted changes in habitat use and distribution of Atlantic migratory striped bass. For striped bass, the predicted outcome of continued global warming is alteration in distribution, both locally and geographically. Both climate models predicted a pronounced upward shifting of estimated annual coastal temperatures (Coutant 1990).

Issue 8.1A.2: Temperature change

Example: Striped bass

Since temperatures on striped bass spawning grounds are predicted to rise, Coutant (1990) indicated that annual events that seem related to the seasonal cycle of water temperature might increase in frequency. He noted that once day-length sets the annual maturation cycle of temperate-zone fishes, temperature plays a dominant role in keying the actual spawning events. Based on data in Westin and Rogers (1978), the average temperature at which striped bass spawning was maximized across the species' range was 16°C, and first spawning occurs at an average temperature of 14.8°C. However, Coutant (1990) noted that it was debatable whether either of those temperatures represented indices of successful recruitment because multiple spawning periods were common in many rivers, and, according to Polgar (1982), survival of eggs and larvae was dependant upon the relative timing of egg depositon and environmental vagaries within the spawning period (Coutant 1990).

Coutant (1990) noted that if the temperatures are used as indicators regardless, then the climate models predict that spawning temperatures will be reached much earlier in the season. Spawning times for the Hudson River and Chesapeake Bay were estimated to differ from three to four weeks. In more northern latitudes, exemplified by coastal water temperatures at Bar Harbor, Maine, the two models differed by nearly a month in the estimated time at which spawning temperatures would be reached. In addition, because striped bass in Canada were reported to spawn over a wide range of temperatures, it was difficult to estimate a timing change. Furthermore, river temperatures might influence spawning more than the modeled coastal temperatures (Coutant 1990).

Additionally, temperature changes might be accompanied by rising sea levels with attendant flooding of spawning habitats in estuaries and wetland nursery areas (Orson et al. 1985). Coutant (1990) noted that predictions of how the coastal environment necessary for striped bass spawning and juvenile rearing would respond to a rising sea level requires consideration of many coastal processes, including tidal ranges, storm surges, intrusion of groundwater and surface water, and sedimentary processes, as well as the response by the plant communities of coastal ecosystems to changes in these processes. Resultant impacts

are likely to be highly site-specific and to include changes both in temperature and dissolved oxygen structure and in physiographic features.

As climate warms, estuaries used by striped bass as nursery areas or adult foraging areas may no longer provide suitable thermal niche space, especially in the summer. The Roanoke River-Albemarle Sound striped bass, which exist at the boundary between the coastal migratory habitats of more northern stocks and the riverine habitats of the more southern stocks, could become strictly riverine and congregate in the summer in the cooler tailwaters upstream of hydroelectric dams (Coutant 1990).

In contrast, stocks in the Bay of Fundy, the Saint John estuary (New Brunswick), the Northumberland Strait, and estuaries entering the Gulf of St. Lawrence that now live under suboptimal thermal conditions and have sporadic year classes, could benefit as global warming produces conditions closer to optimal. The juvenile striped bass thermal niche of $24^{o}C$ to $28^{o}C$ would be more likely to occur in shallow estuaries, establishing warmer conditions for juvenile rearing. Projected expansion of the striped bass range, or any increase in population abundance in the Gulf of St. Lawrence region, would depend greatly on the configuration of coastal currents there (Rulifson et al. 1987).

Issue 8.1A.3: Sea level rise

Example: Striped bass

Accompanying predicted temperature changes could be rising sea levels with attendant flooding of spawning habitats in estuaries and wetland nursery areas (Orson et al. 1985). Predictions of how striped bass spawning and juvenile rearing environments will respond to a rising sea level requires consideration of many coastal processes, including: tidal ranges, storm surges, intrusion of groundwater and surface water, sedimentary processes, and the response by the plant communities of coastal ecosystems to changes in these processes. Resultant impacts are likely to be highly site-specific and to include changes both in temperature and dissolved oxygen structure and in physiographic features (Coutant 1990).

PART II. EFFECTS OF HABITAT DEGRADATION ON HARVESTING AND MARKETABILITY

Effects of habitat degradation that result in non-natural mortality can affect the size of the population and ultimately the size of the allowable harvest. Some threats may not increase mortality, but can reduce or eliminate marketability. These threats include non-lethal limits of contaminants that may render fish unfit for human consumption, or changes in water quality that may reduce fish condition or appearance to a point where they are unmarketable (ASMFC 1999).

Example: Alosines

Table 12-1 lists threats that have been identified for shad and river herring habitat. Because the magnitude of an impact may vary locally or regionally, the degree to which each impact may occur is not specified. Instead, the likelihood to which each impact may occur within each geographical area (riverine waters, territorial waters, or EEZ) is provided. The categories are as follows: Present (P) denotes a threat that has been specifically identified in the literature; No Information Found (NIF) indicates that no information regarding this threat was found within the literature, but there is a possibility that this threat could occur within the specified geographical area; and Not Present (NP) indicates that the threat could not possibly occur within that geographical area (e.g., dam blockage in the EEZ).

THREAT	Riverine Waters	Territorial Waters	EEZ
Chemical			
Acid/aluminum pulses	P	NIF	NIF
Sedimentation	P	NIF	NIF
Suspended particles	P	NIF	NIF
Inorganic inputs	P	P	NIF
Organic chemicals	P	P	NIF
Thermal effluent	P	P	NP
Urban stormwater pollution	P	P	NIF
Sewage/animal waste	P	P	NIF
Non-point source pollution	P	P	NIF

THREAT	Riverine Waters	Territorial Waters	EEZ
Physical			
Dams/spillways	P	NP	NP
Other man-made blockages (e.g., tide gates)	P	P	NP
Non-anthropogenic blockages (e.g., vegetative debris)	P	NP	NP
Culverts	P	NP	NP
Inadequate fishways/fish-lifts	P	NP	NP
Water releases from reservoirs	P	P	NP
Non-hydropower water withdrawal facilities (e.g., irrigation, cooling)	P	P	NP
Channelization	P	NIF	NP
Dredge and fill	P	P	NP
Urban and suburban sprawl	P	NIF	NP
Land-based disturbances (e.g., de-forestation)	P	NIF	NP
Jetties	NP	P	NP
Overharvesting	P	P	P
Biological			
Excessive striped bass predation	P	P	NIF
Nuisance/toxic algae	P	NIF	NIF

Table 12-1. Threats to shad and river herring habitat

PART III. DIADROMOUS THREATS IDENTIFIED IN STATE WILDLIFE ACTION PLANS

Purpose and Scope

Congress created the State Wildlife Grants Program in 2001 to provide every state and territory with federal dollars to support conservation efforts and prevent wildlife from becoming endangered. This program supports projects that protect and restore important lands and waters, collect information on the status of wild populations, and develop partnerships with landowners to protect declining species and habitats on public and private lands. The idea is that states should take a proactive approach to protect wildlife and habitats before they become too rare and costly to protect.

To make the best use of the State Wildlife Grants Program, Congress charged each state and territory with developing a State Wildlife Action Plan (SWAP). As a result of this mandate, state fish and wildlife agencies developed strategic plans that were submitted to the U.S. Fish and Wildlife Agency by October 1, 2005. These SWAPs are intended to be tools for adaptive management, and thus, will undergo additional revisions.

Congress identified eight required elements that each state must address in their SWAP. Strategies should also identify and focus on the "species in greatest need of conservation," yet address the "full array of wildlife" and wildlife-related issues. The eight required elements are summarized as follows:

(1) Information on the distribution and abundance of fish and wildlife species

(2) Description of locations and relative condition of key habitats and community types

(3) Description of problems that may adversely affect species identified in element (1) above or their habitats, and priority research and survey efforts

(4) Description of conservation actions

(5) Proposed plans for monitoring

(6) Descriptions of procedures to review the strategy

(7) Plans for coordinating the development, implementation, review, and revision of the plan with Federal, State, and local agencies, and tribal governments

(8) Broad public participation

For most states, many of the diadromous fish species are identified under element (1) above as species of greatest conservation need (SCGN). States used multiple sources to identify SGCN, such as federally listed species, as well as state-listed Endangered, Threatened, and Species of Concern. Many states ranked species according to the Natural Heritage methodology, maintained by NatureServe. NatureServe is a non-profit conservation organization that provides scientific information to local, national, and global interests to help them collect and manage information on their natural resources. The following "S," or state rankings, have the same standards used to classify species in each state, but wording may vary slightly from state-to-state. Note that these are not the same as federal or global rankings.

S1 – Critically imperiled in the state

S2 – Imperiled in the state

S3 – Vulnerable to extirpation or extinction in the state

S4 – Apparently secure in the state

S5 – Secure, common, widespread, and abundant in the state

It is important to note that some species may have an S4 or S5 state ranking, but were still included in the plan. It could be that habitats in which they are present are known to have problems that could threaten their ranking. Regardless, the purpose of this document is not to note the state status of each diadromous fish species, but rather to identify threats, wherever possible. State rankings are provided simply as an additional reference.

Identifying Threats

Element (3) above requires states to identify problems or threats to species and their habitats. Because plans are required to identify SGCN, it is possible that some diadromous fish species were not identified in some state plans because they are not SGCN within that state. Some states noted the presence of other diadromous fish species within certain habitats that were not listed as SGCN within their state. Wherever possible, if a diadromous fish species is known to occur within a given state but was not listed in the SWAP, it is noted. Information about presence of species within these states was obtained from tables included on the accompanying DVD.

The threats identified may, or may not, be an inclusive list for each species. Some states are better suited to comprehensively identify diadromous fish species-habitat associations and threats because of better availability of information and greater funding. In some states, identified threats may be overarching threats to a particular habitat (e.g., upland rivers) or a broad species category (e.g., fish). The threats may apply to many, but not all, of the species within the habitat. Thus, it is possible that some of the threats may not apply to American eel, Atlantic sturgeon, American shad, hickory shad, alewife, blueback herring, or striped bass. If there is a question about a specific threat within a state, please contact a representative from the state fish and wildlife agency.

Finally, it should be emphasized that the format and content of information will vary, according to how it was presented in each individual SWAP. The text has been adapted from each plan in an effort to best summarize the data. Consult each plan for additional information (for full text, see http://www.wildlifeactionplans.org).

SWAP Information By State

Maine

The following diadromous fish species were ranked, general descriptions of primary and secondary habitats provided, and threats to individual species identified. Please note that

although alewife and blueback herring were not identified as SGCN within the state, they occur within many rivers and streams throughout the state. Thus, it is likely that some of the threats identified may also apply to these species.

Species

American eel - S1 state ranking

Primary habitats are lakes and ponds, and rivers and streams, and secondary habitat is estuaries and bays. Threats to habitat include: dams; poorly functioning fish passage facilities for upstream and downstream movement; and habitat loss or degradation.

American shad - S2 state ranking

Primary habitat is rivers and streams, and secondary habitat is estuaries and bays. Threats to habitat include: dams and other physical obstructions; and land use (e.g., farming, logging, and urbanization).

Atlantic sturgeon - S1 state ranking

Primary habitat is rivers and streams, and secondary habitat is estuaries and bays. Threats to habitat include: habitat loss or degradation.

Striped bass - S1 state ranking

Primary habitat is rivers and streams, and secondary habitat is estuaries and bays. Threats to habitat include: habitat loss or degradation.

Citation

Maine Department of Inland Fisheries and Wildlife. 2005. Maine's Comprehensive Wildlife Conservation Strategy.

New Hampshire

New Hampshire's state ranking for each species is listed below. In addition to the state rankings, New Hampshire also compiled a list of all the associated risk factors relevant to each species and habitat, then scored, ranked, and categorized those factors. Rankings range from 1-4, with 1 being species with the least risk factors and 4 being species with the greatest risk factors. Also noted below is a brief justification for inclusion in the SWAP, but is not necessarily a comprehensive list of threats for the species.

Species

Alewife - S5 state ranking; level 2 risk

Justification for inclusion: presence of dams, which reduce access to spawning habitat.

American eel - S5 state ranking; level 3 risk

Justification for inclusion: dams, unfavorable environmental conditions in freshwater and marine habitats, pollution, and climate change.

American shad - S5 state ranking; level 3 risk

Justification for inclusion: dams and pollution.

Atlantic sturgeon - S1 state ranking; level 4 risk

Justification for inclusion: habitat degradation and barriers.

Blueback herring - S4 state ranking; level 2 risk

Justification for inclusion: dams, which severely limit access to spawning habitat.

Habitats

Although threats for these individual species are not identified in the SWAP, diadromous species are identified within their respective watershed groupings, with the most challenging threats identified for the entire watershed grouping.

Coastal Transitional Watershed

This grouping contains American eel. The most challenging threat facing coastal transitional watersheds is introduced species.

Connecticut River Mainstem Watersheds

This grouping contains American eel, American shad, and blueback herring. The most challenging threats facing Connecticut River mainstem watersheds include non-point source pollution and agriculture.

Northern Upland Watershed

This grouping contains American eel. No critical threats have been identified for northern upland watersheds. However, development and altered hydrology are likely to become problematic over time.

Non-tidal Coastal Watersheds

This grouping contains alewife, American eel, American shad, Atlantic sturgeon, and blueback herring. The most challenging threats facing this watershed include development and non-point source pollution.

Tidal Coastal Watersheds

This grouping contains alewife, American eel, American shad, Atlantic sturgeon, and blueback herring. The most challenging threat facing this watershed is development,including: urbanization, habitat loss and conversion, non-point source pollution, and other factors.

Citation

New Hampshire Fish and Game Department. 2005. New Hampshire's Wildlife Action Plan.

Massachusetts

Species

The Massachusetts SWAP lists the following diadromous fish species as SGCN:

Alewife - unranked (state conservation status not yet assessed)

Specific threats to this species include: dams, pollution, development, over-fishing, and poorly maintained fishways.

American eel - S5 state ranking

Specific threats to this species include: water pollution, dams that hinder migration, changes in ocean circulation patterns, and possibly overfishing.

American shad - S3 state ranking

Specific threats to this species include: dams, inadequately or poorly maintained fishways, and pollution.

Atlantic sturgeon - S1 state ranking

This is a SGCN in large and mid-sized rivers. Specific threats to this species include: dams, water pollution, historic over-fishing, and bycatch and the associated mortality rates. The late age at which Atlantic sturgeon begin spawning, and a requirement for freshwater, estuarine, and coastal habitats to complete their life cycle, make them particularly vulnerable.

Blueback herring - S4 state ranking

Specific threats to this species include: dams and pollution.

Habitats

There are four habitats that contain some or all of the SGCN listed above. Threats to the overall habitat, but not necessarily to the individual species, have been identified and are discussed below.

Connecticut and Merrimack Rivers- Mainstem Habitat

A) *Species:* Atlantic sturgeon, American shad, blueback herring, American eel, and alewife

B) *Threats*:

1) <u>Water quality deterioration</u>: Specific threats include toxins in the river (e.g., PCBs), combined sewer overflows (CSOs), bio-accumulation of contaminants, and non-point source pollution (e.g., agricultural run-off).

CSOs in the state regularly cause temporary Class C water quality conditions in urban areas after storms. The Massachusetts Department of Public Health issued fish consumption advisories recommending that children under 12, pregnant women, and nursing mothers not consume any fish from specified areas of the Connecticut River, and the general public should not consume American eel because of elevated levels of PCBs.

2) <u>Habitat loss and fragmentation</u>: Specific threats include: impoundments, filling of wetlands bordering the rivers, and urbanization of the river corridor. Disconnection of the rivers from their floodplains by channelization has led to dramatic changes in habitat.

3) <u>Air pollution</u>: Specific threats include: acid precipitation and atmospheric deposition of mercury and other contaminants. Some sources are local, but the majority of pollution originates from sources outside of the region.

4) <u>Hydroelectric dams</u>: The Connecticut and Merrimack Rivers are some of the most developed rivers in the Northeast. The Massachusetts sections of each of these rivers contain two major hydroelectric dams, including the first dam upstream from the sea on each system. These large dams with operating hydroelectric facilities create unique threats to fish and wildlife populations, including:

   i. *Impoundments*: About one third of the mainstem Connecticut River, and most of the freshwater portion of the Merrimack River, is impounded. The habitat found in these impoundments is far different from that of free-flowing rivers.

   ii. *Bypasses*: Large hydroelectric dams divert much of the river flow away from the rapids habitat. This often results in rapids below both the Turners Falls dam on the Connecticut River, and the Pawtucket dam on the Merrimack River, being dry for much of the summer.

   iii. *Population fragmentation*: Dams form barriers to migration, which can dramatically reduce the habitat available to anadromous fish and may fragment resident fish populations.

   iv. *Flow alteration*: The Turners Falls Hydroelectric Project on the Connecticut River is a "peaking" project. It stores water over a period of several hours, and then releases it all at once, dramatically changing the river flow. These daily changes in flow below the dam and reservoir level above the dam disrupt fish and wildlife habitat and lead to large-scale riverbank erosion.

5) <u>Invasive species</u>: A number of invasive species have taken hold in these watersheds and threaten native species. These include: common reed (*Phragmites australis*), purple loosestrife (*Lythrum salicaria*), Eurasian watermilfoil (*Myriophyllum spicatum*), and water chestnut (*Trapa natans*), as well as Mute Swans, Asiatic clams (*Corbicula fluminea*), and hemlock woolly adelgid (*Adelges tsugae*).

6) <u>Human usage</u>: Recreational use of these rivers, whether by boat or on foot, can degrade habitat and sometimes cause outright destruction of these species of concern.

Large and Mid-size Riverine Habitat

A) *Species*: Atlantic sturgeon, American shad, blueback herring, American eel, and alewife

B) *Threats*:

1) <u>Physical habitat alterations:</u> Channelization, particularly near urban centers, has resulted in massive habitat loss in all watersheds, but especially in the Charles, Concord, Blackstone, North and South Coastal, and Merrimack watersheds. Portions of some rivers, for example, the Hoosic River in Adams and North Adams, have actually been completely culverted and run through food chutes instead of natural channels.

2) <u>Dams</u>: Dams impact all watersheds in the state. The only mainstem in Massachusetts considered to be free-flowing is the Taunton River. These dams all result in a loss of physical habitat suitable for fluvial species within the impoundment, but other habitat impacts are also apparent. Stream flow downstream of almost all impoundments is severely restricted during low flow times of the year or when lakes are being refilled after an artificially induced lake drawdown. Minimum streamflow criteria are not regulated for most reservoir situations. Likewise, maximum streamflow is not regulated during artificial drawdowns when spring-like (or greater) flows are allowed to take place in times other than spring. These dams also cause a buildup of sediment, sometimes severely contaminated, within the impoundment and result in incised channels downstream of the impoundment. Incised channels further isolate the river channel from the surrounding floodplain.

    i. *Hydroelectric power*: The Deerfield, Westfield, and Swift Rivers have the majority of hydroelectric generation (excluding the Connecticut and Merrimack River mainstems, discussed above).

    ii. *Flood protection*: Large-scale flood control projects exist on the Quinebaug, Westfield, and Millers Rivers.

    iii. *Reservoirs*: Water supply reservoirs are common statewide and range in size from the 25,000-acre Quabbin Reservoir to smaller secondary or backup water supply impoundments.

3) <u>Sewerage treatment effluent:</u> Many of Massachusetts's large to mid-sized rivers are impacted by effluent from centralized sewerage treatment plants. In some cases, raw sewerage continues to be released into our waters. The Blackstone, Charles, Concord, and Nashua Rivers are particularly impacted. During summer low flows, the Blackstone and Assabet rivers (a tributary to the Concord River) are composed primarily of sewerage treatment effluent.

4) <u>Stormwater runoff</u>: Runoff has caused substantial changes to water quality and causes erosion issues. Winter runoff often includes high concentrations of

road salt, while stormwater flows in the summer cause thermal stress and bring high concentrations of other pollutants. Road, culvert, and public water and sewer have created pathways, both intentional (CSO flows) and unintentional (inflow and infiltration), that have expedited the movement of rainfall and runoff into stream channels.

5) <u>Water withdrawal and surface water diversion:</u> These activities result in impacts to all of the basins to some extent, as illustrated in the Stressed Basins Report published by the Massachusetts Department of Conservation and Recreation, but especially to some of the higher quality rivers in the state. The Ipswich River continues to serve as the model for environmental degradation caused by water withdrawal. The Ipswich River is impacted by both surface water diversion and groundwater withdrawal, and was listed by American Rivers in 2003 as one of the ten most endangered rivers in America, due to worsening flow conditions.

Marine and Estuarine Habitat

A) *Species*: Atlantic sturgeon, American shad, blueback herring, American eel, and alewife

B) *Threats*:

1) <u>Shoreline development</u>: This is the greatest threat to the coastal bays and estuaries in the state. Massachusetts has lost close to 30% of its coastal wetlands due to development. The loss of coastal wetlands reduces the filtration ability provided by such wetlands to waters entering bays and estuaries. Shoreline development results in more impervious surface with increased stormwater runoff and accompanying potential for sedimentation and toxic contamination.

2) <u>Wastewater treatment</u>: Overflows and leaks from wastewater treatment plants and faulty septic systems can result in bacterial and pathogenic contamination and increase nitrogen loading in Massachusetts's coastal waters. This, in turn, promotes algal growth on eelgrass beds to the detriment of this valuable aquatic food and cover source for fish, shellfish, marine invertebrates, and waterfowl and other aquatic birds.

3) <u>Boating</u>: Increased commercial and recreational boat traffic re-suspends sediments, further shading submerged vegetation. Direct discharge of waste from recreational boating, and accidental oil spills from commercial shipping, have been threats in the past and will continue in the future.

4) <u>Invasive species</u>: A number of invasive species have taken hold in these habitats and threaten native species. These include common reed (*Phragmites australis*) and purple loosestrife (*Lythrum salicaria*).

Lake and Pond Habitat

A) *Species:* American eel and alewife

B) *Threats*:

    1) Eutrophication: Accelerated eutrophication due to watershed activities is one of the greatest threats to Massachusetts's lakes. These activities can include input from: nutrient-rich effluents from sewage treatment plants, agricultural run-off, stormwater run-off from impervious surfaces, leaching from septic systems, and soil erosion from construction and timbering activities. Currently, hundreds of waters in Massachusetts do not meet their designated water quality standards. This accelerated eutrophication can contribute to an increase in the abundance of aquatic vegetation, increased turbidity, decreased dissolved oxygen levels, and increased sedimentation which ultimately decreases the depth of a lake. Most Massachusetts lakes are particularly susceptible to accelerated eutrophication due to their small watersheds.

    2) Invasive species: The introduction of non-native invasive plants that can create monocultures and eliminate open water habitat is another major threat to the lakes. As with aquatic plants, the introduction of non-native animals, such as zebra mussels (*Dreissena polymorpha*) or snakeheads (*Channa* sp.), can have a devastating effect on the aquatic ecosystem.

Citation

Commonwealth of Massachusetts, Executive Office of Environmental Affairs (MEOEA). 2005. 2005 Massachusetts Comprehensive Wildlife Conservation Strategy.

Rhode Island

Species

The Rhode Island SWAP lists the following diadromous fish species as SGCN:

Alewife – S3 state ranking

American eel – S5 state ranking

American shad – S1 state ranking

Atlantic sturgeon – SH (state historical) ranking

This species is listed by Rhode Island as a Species of Special Concern, and is imperiled.

Blueback herring – S1 state ranking

Habitats

Marine and Estuarine Habitats

A) *Species*: Marine/estuarine fish, which includes diadromous fish (individual species are not indicated)

B) *Threats*:

1) Wetland loss: Direct loss and fragmentation of wetlands has been caused by shoreline development, recreational use, bulkheads, poor urban development, dredging, dredge disposal, ditching and draining, and other benthic disturbances.

2) Changing water regime: Changes in the freshwater regime have resulted from freshwater diversion, dam removal and waterway restoration, and ditching wetlands.

3) Pollution: Pollution has caused sedimentation and contamination of marshes.

i. *Point source*: Direct contamination has come from industrial discharge, heavy metals, sediment, oil spills, marine accidents, ocean dumping, and other contaminants.

ii. *Non-point source*: Sedimentation and contamination has come from erosion, agriculture run-off, pesticides, and septic systems.

4) Nutrient loading: Nutrient loading originating from sewage pollution (e.g., combined sewage overflow, failing and inadequate systems, and boat waste) has caused algal blooms and other issues.

5) Temperature: Temperature changes and regulation have caused problems for native species survival.

6) Invasive species: These species directly affect habitat, competitors, predators, pathogens or parasites, and/or changes in the native species dynamics, or by directly competing with the native species.

River and Stream Habitats

A) *Species*: Diadromous species (individual species associations are not identified)

B) *Threats*:

1) Habitat fragmentation: This has been caused by a lack of conservation planning capabilities and coordination, a lack of a focal area approaches to conservation, human disturbance, chemical contaminants and disease, and road effects.

2) Habitat degradation: This has been caused by chemical contaminants and disease, human disturbance, and impairment of water quality.

3) Habitat loss: This has been caused by inadequately sized preserves, plant succession, invasive species, and impairment of aquatic contiguity.

4) Lack of research: There has been a lack of information from research to address habitat and taxonomic issues.

Citation

State of Rhode Island Department of Environmental Management, Division of Fish and Wildlife. 2005. Rhode Island's Comprehensive Wildlife Conservation Strategy.

<u>Connecticut</u>

Species

The Connecticut SWAP lists the following diadromous fish species state rankings:

Alewife – S3 state ranking

American eel – S5 state ranking

American shad – S3 state ranking

Atlantic sturgeon – S1 state ranking

Blueback herring – S5 state ranking

Hickory shad – S2 state ranking

Striped bass – S3 state ranking

Habitats

General

A) *Species*: All fish and wildlife species collectively

B) *Threats*:

1) Insufficient knowledge: In general, there is insufficient scientific knowledge regarding wildlife, as well as freshwater, diadromous, and marine fish species, and their habitats (distribution, abundance and condition).

2) Habitat fragmentation, degradation, and loss: These problems have resulted from development or changes in land use, a lack of resources to maintain/enhance wildlife habitat, a lack of landscape-level conservation efforts, public indifference toward conservation.

3) Invasive species: These species (e.g., *Phragmites australis*, *Lythrum salicaria*, and Mute Swan) have caused problems for species and habitat in many areas.

4) Species limitations: Some species with depressed populations have experienced delayed recovery due to limited reproductive potential, dispersal ability, or other factors.

Freshwater Habitats

A) *Species*: Diadromous species (individual species associations are not identified)

B) *Threats*:

1) Habitat fragmentation, degradation, and loss: These problems have resulted from stream channel modifications, dams, channelization, filling, dredging, development, sedimentation, vegetation control, and shoreline modification. There has also been a loss of coldwater habitat due to decreased groundwater input or increased warming (e.g., wetlands filling, impoundment, removal of riparian vegetation).

2) Predation: There have been impacts to prey species from predation by striped bass in the Connecticut River.

3) Fish passage: Populations have been fragmented and access has been lost to upstream and spawning habitat due to impediments to fish movements, such as dams, barriers, culverts, and tide gates.

4) Pollution: There have been impacts of point and non-point source pollution on diadromous fish populations.

5) Boating: Excessive boat activity has lead to wake wash, sediment suspension, and propeller scarring.

6) Water withdrawal and surface water diversion: Instream flow alterations and increasing temperatures have been caused by consumptive withdrawals of surface or ground water and wetland loss. Water diversions that reduce stream flows have also resulted in fish mortality, loss of habitat, and interference with migration.

7) Regulations: Ineffective or insufficient land use regulations among towns have impacted fish habitats.

8) Lake manipulations: There have been adverse impacts to fish from lake manipulations (e.g., excessive vegetation control, water level manipulation, and dredging).

9) Nutrient loading: Excessive nutrient run-off and vegetation control has lead to a loss of the oxygenated hypo-limnetic and meta-limnetic zone.

10) Migration disruption: Dredging and development have lead to disrupted migration of diadromous fish.

11) Natural barriers: Beaver dams have impacted coldwater habitats, resulting in ponding and warming, fragmentation of habitat, and increased sedimentation and nutrient loading.

Marine Habitats

A) *Species*: All aquatic species in marine areas

B) *Threats*:

1) <u>Habitat fragmentation, degradation, and loss</u>: Disturbance, destruction, alteration, or loss of critical habitat structure or function is a major problem.

2) <u>Residual contaminants</u>: Residual contaminants in sediments and water, such as nutrients and pesticides, effect marine species in many ways.

3) <u>Temperature</u>: There are adverse impacts from temperature shifts, including widespread long-term (e.g., global warming) and local short-term impacts (e.g., temporary power plant shutdowns).

4) <u>Non-native species</u>: Predation, competition, displacement from habitat, and/or disease transmission are associated issues.

5) <u>Fishing</u>: Unintentional damage, injury, or mortality due to fishing (e.g., incidental catch, or injuries from fishing gear) is also a problem.

Citation

State of Connecticut Department of Environmental Protection, Bureau of Natural Resources. 2005. Connecticut's Comprehensive Wildlife Conservation Strategy.

<u>New York</u>

Species and Habitats

Critical habitats and sub-habitats are identified in the SWAP, including a breakdown of breeding, feeding, and nursery/juvenile life history stages. Threats to individual species within these habitats have also been identified.

Alewife – no state ranking, and has unprotected state status

Critical habitats and sub-habitats for alewife include:

1) Breeding and nursery/juvenile:
  i. Estuarine
    a) Shallow subtidal
  ii. Riverine
    a) Coastal plain stream
    b) Deepwater river
2) Breeding only:
  i. Riverine
    a) Warmwater stream
3) Feeding:
  i. Marine
    a) Deep subtidal

Specific threats to this species include: loss of historic spawning grounds and degradation of spawning and juvenile habitat, primarily in inshore areas.

American eel – S5 state ranking, and has unprotected state status

Critical habitats and sub-habitats for American eel include:

1) Nursery/juvenile:
    i. Estuarine
        a) Cultural
        b) Shallow subtidal
        c) Deep subtidal
        d) Intertidal
    ii. Riverine
        a) Coastal plain stream
        b) Deepwater river
    iii. Lacustrine
        a) Coastal plain
2) Breeding:
    i. Marine

Specific threats to this species include: barriers to migration, especially dams, which can cause upstream and downstream passage to migration to be inadequate or absent. Contamination from industrial pollution also threatens the American eel, and may contribute to the suppression of female development. Due to their wide range of life history cycle, American eel recruitment may also be affected by climate and weather.

American shad – S4 state ranking, and has protected state status

Critical habitats and sub-habitats for American shad include:

1) Nursery/juvenile:
    i. Estuarine
        a) Intertidal
    ii. Riverine
        a) Deepwater river
2) Breeding:
    i. Estuarine
        a) Shallow subtidal
    ii. Riverine
        a) Deepwater river

Specific threats to this species include: continued shoreline development and related dredging activities due to increased commercial boat traffic in the Hudson Shallow spawning habitat. Dams located in Pennsylvania along the Susquehanna River are still a threat to migratory spawning stocks, but fish passage improvements continue.

Atlantic sturgeon – S1 state ranking, and has threatened state status

Critical habitats and sub-habitats for Atlantic sturgeon include:

1) Breeding and nursery/juvenile:
    i. Estuarine
        a) Deep subtidal

Specific threats to this species include: dredge and development activities in spawning and nursery areas. The effect of contaminants on juveniles is unknown at this point.

Blueback herring – no state ranking, and has protected state status

Critical habitats and sub-habitats for blueback herring include:

1) Breeding and nursery/juvenile:
    i. Estuarine
        a) Shallow subtidal
2) Breeding only:
    i. Riverine
        a) Warmwater stream

No specific threats to this species have been identified.

Citation

New York State Department of Environmental Conservation. 2005. A Strategy for Conserving New York's Fish and Wildlife Resources.

New Jersey

Species

Currently, the SGCN identified within New Jersey include the following categories: endangered, threatened, special concern and regional priority species, species of unknown status, and species identified as extirpated. Additionally, species that have not been reviewed through the Delphi Status Review, but hold a global element rank of G1-G3, and/or a state element rank of S1-S3, have been included among the species of special concern and regional priority. At this time, only the following diadromous species have been listed as SGCN in New Jersey:

Atlantic sturgeon – S3 state ranking, and a state species of special concern

Hickory shad - S3 state ranking

Habitats

Threats are identified for the five different landscapes in the state. Landscape regions are ecoregions within the state that were delineated based on land forms, soils, vegetation, and hydrological regimes. Landscape regions are further divided into conservation zones, based on the variable habitats that exist within these regions. Specific habitat threats and conservation goals for each conservation zone are also identified. Listed below are the landscapes and conservation zones that contain Atlantic sturgeon or hickory shad, and associated threats within these zones. Please note that although American shad, alewife, blueback herring, American eel, and striped bass were not identified as SGCN, they occur within many rivers and streams throughout the state. Thus, it is likely that some of the threats identified may also apply to these species.

Atlantic Coastal Landscape Ecoregion

    A) Atlantic Ocean Conservation Zone

        1) *Species*: Atlantic sturgeon and hickory shad

        2) *Threats*:

            i. Habitat loss and degradation: This results from commercial fishing practices, such as gillnetting for monkfish and dogfish sharks impacting sturgeon.

            ii. Oil spills: In particular, large events always loom as a threat due to the large amount of oil routinely transported to ports in the Delaware River near Philadelphia and New York Harbor are. Oil spills have potentially serious short and long-term impacts on all marine species.

            iii. Aquaculture: The impacts of this practice are largely unmeasured and poorly understood.

            iv. Hydraulic crab dredging: The impacts of this practice are largely unmeasured and poorly understood.

    B) Atlantic Coastal Cape May Zone; Atlantic City Area Zone; Brigantine – Great Bay Area Zone; Barnegat Bay – Little Egg Harbor Zone; and Northern Atlantic Coastal Conservation Zone

        1) *Species:* Atlantic sturgeon and hickory shad

        2) *Threats*:

            i. Aquaculture: The impacts of this practice are largely unmeasured and poorly understood.

            ii. Hydraulic crab dredging: The impacts of this practice are largely unmeasured and poorly understood.

Delaware Bay Landscape Ecoregion

    A) Tuckahoe River Watershed Conservation Zone

        1) *Species:* Atlantic sturgeon

        2) *Threats*:

            i. Invasive species: These species threaten the ecological integrity of habitats in the region.

    B) Delaware Bay Shoreline Conservation Zone

        1) *Species:* Atlantic sturgeon and hickory shad

2) *Threats*:

    i. <u>Oil and hazardous materials spills</u>: Delaware Bay is the second largest port for oil transport on the East coast, so oil spills are a real threat to habitats and animal populations.

C) Cape May Peninsula Conservation Zone

1) *Species:* Atlantic sturgeon and hickory shad

2) *Threats*:

    i. <u>Development</u>: This can lead to habitat fragmentation, water quality declines, and pressure on groundwater resources.

Piedmont Landscape Ecoregion

A) Central Piedmont Plains Conservation Zone

1) *Species:* Atlantic sturgeon and hickory shad

2) *Threats*:

    i. <u>Development</u>: This removes upland buffers and wetlands.

    ii. <u>Chemical contamination</u>: Pesticides and herbicides are potential threats.

B) Southern Piedmont Plains Conservation Zone

1) *Species*: Atlantic sturgeon and hickory shad

2) *Threats*:

    i. <u>Chemical contamination</u>: Run-off of pesticides and other contaminants (e.g., PCBs) from residential and agriculture areas into waterways is problematic.

    ii. <u>Physical habitat alterations</u>: Ditching, draining, and filling of marshes eliminates habitat and degrades the remaining surrounding areas. Clearing of vegetation along rivers and streams is a leading cause of habitat loss, fragmentation, and degradation of riparian and aquatic ecosystems. Loss of vegetated buffers along streams and rivers increases runoff of contaminants from roads and developed areas, impacting aquatic communities.

    iii. <u>Oil spills</u>: This zone is situated entirely within the ports of Wilmington, Delaware, and Philadelphia, Pennsylvania, which together support some of the largest petro-chemical facilities in the United States. This results in potentially catastrophic spill and contaminants-related threats.

    iv. <u>Dredging</u>: Shipping channel expansion or deepening in the Delaware River could have significant implications on salinity levels in tidal freshwater emergent marshes.

> v. <u>Invasive species</u>: Aquatic nuisance species may render some freshwater systems unsuitable for many fish and aquatic invertebrate species.
>
> vi. <u>Natural barriers</u>: In riparian areas, North American beavers can create wetland habitat suitable for many species by damming up streams, but may, in turn, alter riparian habitat downstream from the dam.

Skylands Landscape Ecoregion

A) Southern Highlands Conservation Zone

> 1) *Species*: Hickory shad
>
> 2) *Threats*:
>
> > i. <u>Development</u>: This practice causes disturbance, culvert construction, habitat loss, fragmentation, and degradation.
> >
> > ii. <u>Chemical contamination</u>: The use of pesticides, mowing, and other agricultural practices may impact species in this area. The effects of contamination and alteration of waterways and wetlands, is exascerbated by increased human encroachment into riparian areas.
> >
> > iii. <u>Non-point source pollution</u>
> >
> > iv. <u>Unrestricted livestock access to waterways</u>
> >
> > v. <u>Reduction in stream flows</u>
> >
> > vi. <u>Stream cleaning activities</u>
> >
> > vii. <u>Dams</u>

Citation

New Jersey Department of Environmental Protection, Division of Fish and Wildlife. 2008. New Jersey Wildlife Action Plan for Wildlife of Greatest Conservation Need.

<u>Delaware</u>

Species

Due to staff and funding limitations, the Natural Heritage Program does not track many of Delaware's species, especially estuarine and marine fish. Although SGCN were divided into two tiers, mapping of SGCN did not allow the original intent of the use of the tiers to be realized. Thus, for the first iteration of the SWAP, all SGCN are treated as being in equal need of conservation. The SGCN received the following state rankings, and are associated with these primary and secondary habitats:

Atlantic sturgeon –S2 state ranking on Tier 1, and is state endangered

Primary habitats for Atlantic sturgeon include: Freshwater Aquatic Habitat and Brackish and Marine Habitat. Secondary habitats for the species include: Coastal Plain Streams Habitat and Nearshore Habitat (note that this habitat is listed as a habitat of conservation concern).

Hickory shad –S2 state ranking on Tier 2

Primary habitats for hickory shad include: Freshwater Aquatic Habitat and Brackish and Marine Habitat. Secondary habitats for the species include: Coastal Plain Streams Habitat and Nearshore Habitat (note that this habitat is listed as a habitat of conservation concern).

Habitats

Delaware uses the term "conservation issues" synonymously with "threats" or "stresses," and defines it as, "human actions that adversely impact wildlife, native plants, and natural communities, and the ecological processes that sustain them." Listed below are the habitats that contain Atlantic sturgeon or hickory shad, and associated threats within these zones. Please note that although American shad, alewife, blueback herring, American eel, and striped bass were not identified as SGCN, they occur within many rivers and streams throughout the state. Thus, it is likely that some of the threats identified may also apply to these species.

Non-tidal Coastal Plain Streams Habitat

A) *Species*: Atlantic sturgeon, hickory shad, and American eel

B) *Threats*:

1) Development: Residential and commercial development practices, including altered hydrology, nutrients and sediments in the water, and the use of pesticides all influence this habitat.

2) Agricultural and forestry operations: Practices from these industries, including ditching and draining, altered hydrology, nutrients and sediments in the water, and use of pesticides, impact this habitat.

3) Shoreline protection: Practices, including artificial shoreline hardening, impact this habitat.

4) Industrial operations: These operations cause air pollution, accidental spills of toxins and sewage, chronic water pollution, impingement, entrapment, and entrainment at water intakes, and sedimentation from sand and gravel quarrying.

5) Transportation and utility operations and maintenance: These operations cause transportation infrastructure, altered hydrology, commercial ships and boats, road salt, and channel dredging.

6) Invasive species: These species (e.g., Snow Goose, resident Canada Goose, Asiatic clam, and invasive plants) cause problems in this habitat.

7) Water use: Problems associated with water use include: dams, dam operations, groundwater withdrawals, and surface water withdrawals.

8) <u>Recreational activities</u>: Issues with these activities include: recreational use on foot and with boats, personal watercraft, and off-road vehicles.

9) <u>Wildlife harvest</u>: This includes inappropriate hunting and fishing activities.

10) <u>Resource management</u>

Nearshore Habitat

A) *Species*: Atlantic sturgeon and hickory shad

B) *Threats*:

1) <u>Development</u>: Residential and commercial development practices, including altered hydrology, nutrients and sediments in the water, and the use of pesticides all influence this habitat.

2) <u>Agricultural and forestry operations</u>: Practices from these industries, including ditching and draining, altered hydrology, nutrients and sediments in the water, and use of pesticides, impact this habitat.<u>Shoreline protection</u>: Practices, including beach nourishment, impact this habitat.

3) <u>Industrial operations</u>: These operations cause air pollution, accidental spills of toxins and sewage, chronic water pollution, impingement, entrapment, and entrainment at water intakes, and sedimentation from sand and gravel quarrying.

4) <u>Transportation and utility operations and maintenance</u>: These operations cause commercial ships and boats and channel dredging.

5) <u>Solid waste disposal</u>: This is a problem because of trash ingestion.

6) <u>Invasive species</u>: The species most impacting this habitat are green crab and Japanese shore crab.

7) <u>Energy production</u>: Concerns with energy production involve wind farm facilities, tidal turbines, and thermal pollution from power plants.

8) <u>Recreational activities</u>: These activities are problematic with recreational use on foot and with boats, personal watercraft, and off-road vehicles.

9) <u>Wildlife harvesting</u>: Issues with harvest include: inappropriate hunting and fishing, fishing gear entanglement, fisheries bycatch, and commercial fisheries dredging.

10) <u>Resource management</u>

Citation

Delaware Division of Fish and Wildlife and Delaware Department of Natural Resources and Environmental Control. 2006. Delaware Wildlife Action Plan 2007-2017.

Maryland

Species

The Maryland SWAP lists the following diadromous fish species as SGCN:

Atlantic sturgeon – S1 state ranking

American shad – S3 state ranking

Hickory shad – S3 state ranking

Habitats

Maryland has identified SGCN found within specific habitats and threats to these habitats, which are as follows:

Coastal Plain Streams Habitat

A) *Species*: American eel, American shad, and hickory shad

B) *Threats*:

1) Development: Urban land use and impervious surfaces can result in chemical and hydrologic changes and fragmentation and isolation.

2) Sedimentation

3) Habitat loss and degradation: Issues associated with this threat include: Removal or degradation of riparian buffers; loss of headwater areas; deforestation that results in loss of forested watershed; and bank erosion.

4) Atmospheric deposition

5) Invasive species

6) Agricultural and forestry operations: Issues associated with this threat include: Pesticide/herbicide application results in pollution or degradation of water quality; liming practices; livestock and grazing practices; inappropriate timber harvest practices impact water quality or cause loss of coarse woody debris; and nutrient enrichment.

7) Stream blockages: This issue includes blockages caused by dams.

8) Dumping

9) Recreational activities

10) Point-source pollution

11) Reduction in stream flows: Activities associated with this issue include: Groundwater and stream water withdrawals; stream channelization

12) Sea-level rise

Coastal Plain Rivers Habitat

 A) *Species*: American shad, hickory shad, alewife, and blueback herring

 B) *Threats*:

  Threats include #1-12 listed above in the ***Coastal Plain Streams Habitat***, as well as the following:

   1) Oil and chemical spills

Piedmont Rivers Habitat

 A) *Species*: American eel, American shad, hickory shad, alewife, and blueback herring

 B) *Threats*:

  Threats include #1-12 listed above in the ***Coastal Plain Streams Habitat***, as well as the following:

   1) Hydroelectric power generation

Highland Rivers Habitat

 A) *Species*: American eel

 B) *Threats*:

  Threats include #1-12 listed above in the ***Coastal Plain Streams Habitat***, as well as the following:

   1) Acid mine drainage

   2) Hydroelectric power generation

Oligohaline Estuarine Habitat

 A) *Species*: Atlantic sturgeon, American eel, American shad, hickory shad, alewife, and striped bass

 B) *Threats*:

   1) Invasive species: This includes: ballast water release.

   2) Dredge spoil dumping

   3) Habitat loss and degradation: This includes: development, agriculture, human activities, recreation, and environmental contaminants result in habitat degradation.

   4) Oil and chemical spills

   5) Pollution: This issue includes: metalloids, changes in pH, thermal and toxic discharges, nutrients (especially nitrogen and phosphorus), and sedimentation that result in water quality degradation.

   6) Loss of submerged aquatic vegetation

    7) <u>Hydrologic and ground water alterations</u>: These alterations result in changes in salinity.

Mesohaline Estuarine Habitat and Polyhaline Estuarine Habitat

A) *Species*: Atlantic sturgeon, American eel, American shad, hickory shad, alewife, and striped bass

B) *Threats*:

Threats include #1-7 listed above in the ***Oligohaline Estuarine Habitat***, as well as the following:

    1) <u>Loss of dissolved oxygen</u>: This can lead to fish kills.

    2) <u>Oyster reef extraction</u>: This results in habitat loss for diadromous species.

    3) <u>Dredges and scrapes</u>: This issue impacts: SAV and bottom sediments.

Watersheds

Maryland has also listed the following threats that may be present for every watershed within the state. For a more complete listing of the extent, trend, severity, persistence, reversibility, prevention, and restoration factors for each watershed, refer to the state wildlife action plan. Threats include:

    1) <u>Non-point source pollution</u>: Problems are caused by chemical changes from acid deposition/low pH, acid mine drainage, excess nitrates, excess phosphorus, mercury deposition, and organic matter retention.

    2) <u>Point source pollution</u>: Problems are caused by chemical changes from agricultural pesticides, dissolved oxygen, industrial sources, and pathogens.

    3) <u>Habitat alteration</u>: This is caused by channelization, forest fragmentation, ground water withdrawal, migration barriers, runoff/baseflow/down cutting, sedimentation, surface water withdrawal, and wetland loss.

    4) <u>Invasive species</u>: Changes are caused by invasive riparian plants and non-native aquatic plants.

    5) <u>Future changes</u>: Concerns are from land conversion and sea level rise.

Citation

Maryland Department of Natural Resources. 2005. Maryland Wildlife Diversity Conservation Plan.

District of Columbia

Species

       The following anadromous fish species have been identified as SGCN within the District of Columbia. In addition to including species that are globally ranked as G1-G3 species, selection criteria also included declining species, species that are SGCN in Maryland or Virginia, and species with small, localized "at-risk populations." Based on the criteria selected by the District, the following species have been identified:

Atlantic sturgeon – Ranked G1-G3, possibly extirpated from the District

Alewife – Stable population in the District

American eel – Declining population in the District

American shad – Increasing population in the District

Blueback herring – Stable population in the District

Hickory shad – Increasing population in the District

Habitats

       Species-habitat associations have been identified, as well as threats within each habitat. Threats have also been ranked #1 through 3, with 1 being the lowest threat and 3 being the highest threat. The species-habitat associations and threats are as follows:

Rivers and Streams Habitat

A) *Species*: Atlantic sturgeon, alewife, American eel, American shad, blueback herring, and hickory shad

B) *Threats*:

    1) Sedimentation: (2.3 rank) Sedimentation in the District is mainly a function of activities occurring in jurisdictions bordering the Potomac and Anacostia Rivers outside of the District. Due to land disturbance caused by housing and road construction, changes in the hydrologic regime caused by development, and the concurrent increase in impervious surfaces, stormwater runoff during rain events move large quantities of soil from land surfaces into the waterways. Once the rivers begin to widen and slow in the District, the sediment which had been transported downstream with the swift upstream current begins to settle out as sediment. Sedimentation is also caused by water moving oil from disturbed sites in the District.

    2) Hydrologic regime changes: (3 rank) Changes to hydrologic regimes have a number of sources. Urban development with associated draining, paving, topography changes, and other changes in land use can either increase or

decrease the quantity of water flow. Converting forests to lawns, roadways, driveways, or rooftops changes the hydrologic regime by removing the effect of water uptake and transpiration by the trees. The water not normally taken up and transpired by the trees may flow overland and directly into a receiving waterbody. Changing hydrologic regimes in the District are generally leading to reduced recharging of the aquifers and more runoff directly into creeks, streams and rivers. The runoff also tends to lead to increased rates or erosion, increased pollutant loads, and sedimentation.

3) Stormwater erosion: (3 rank) Increases in stormwater erosion occur concurrently with increases in impervious surfaces and changes in land use that occur during development. Due to the highly developed character of the District, stormwater has a tendency to produce a lot of erosion even in naturally vegetated areas. When stormwater is unregulated, or improperly directed to a receiving pond, it leads to sedimentation, transport of pollutants, and dramatic changes in water temperature in the District's creeks, streams, and rivers into which the water flows.

4) Pollution: (2.5 rank) Pollution can enter a habitat in a variety of ways ranging from urban runoff to air pollution. Nutrient loading can create conditions in which native plants cannot compete with invasive and alien species. Airborne pollutants, such as nitrogen and carbon dioxide, can contribute to this excess nutrient loading. The District, as an urban center, is especially vulnerable to both point and non-point source water pollution. Point source pollution includes municipal wastewater and stormwater discharges. For example, millions of gallons of raw sewage are released into the Anacostia River every year. Non-point source pollution results from vast urban development and road construction. Urban development in the District and upstream in Maryland brings pollutants from building and streets into the Anacostia River.

5) Erosion of rivers and streams: (2.9 rank) Erosion is caused both by high flows, typically caused by heavy rains, in the spring falling on frozen ground incapable of absorbing the precipitation, and in the summer and fall associated with passing hurricanes or other large scale meteorological events. It can also occur in the winter, caused by the scouring of river and stream bottoms and banks by ice flows. This type of erosion is believed to be partially responsible for the loss of submerged aquatic vegetation in the District.

6) Invasive species: (2.3 rank)

7) Recreation: (1 rank)

8) Hardened shorelines: (1.9 rank)

9) Migration barriers: (1.4 rank)

10) Piped streams/channelization: (2.4 rank)

Emergent Tidal Wetlands Habitat

A) *Species:* American eel

B) *Threats*:

1) Sedimentation: (2.8 rank) See *Rivers and Streams Habitat* for more information.

2) Hydrologic regime changes: (1.5 rank) Low-lying habitats, such as emergent tidal wetlands are impacted by changes in hydrologic regimes when their associated upland habitats are developed. Riparian woodlands are impacted by changes in hydrologic regimes when the channelization of streams lowers the water table. This eliminates the connection between streams and riparian woodlands, except during floods. This, in turn, increases sedimentation in floodplain forests due to floods.

3) Stormwater erosion: (1.8 rank) See *Rivers and Streams Habitat* for more information.

4) Pollution: (2.7 rank) See *Rivers and Streams Habitat* for more information.

5) Erosion of rivers and streams: (1.3 rank) See *Rivers and Streams Habitat* for more information.

6) Invasive species: (2.5 rank)

7) Hardened shorelines: (1.3 rank)

8) Habitat loss: (1.8 rank) Habitat loss is a threat most closely linked to resident Canada geese. The overly abundant resident geese enter these wetlands to feed, but due to their numbers, end up destroying the habitat.

Tidal Mudflats Habitat

A) *Species*: American eel

B) *Threats*:

1) Sedimentation: (2.6 rank) See *Rivers and Streams Habitat* for more information.

2) Hydrologic regime changes: (2 rank) Low-lying habitats, such as tidal mudflats are impacted by changes in hydrologic regimes when their associated upland habitats are developed. Riparian woodlands are impacted by changes in hydrologic regimes when the channelization of streams lowers the water table. This eliminates the connection between streams and riparian woodlands, except during floods. This, in turn, increases sedimentation in floodplain forests due to floods.

3) Stormwater erosion: (2.2 rank) See *Rivers and Streams Habitat* for more information.

4) Pollution: (2.6 rank) See *Rivers and Streams Habitat* for more information.

5) <u>Erosion of rivers and streams</u>: (1.8 rank) See *Rivers and Streams Habitat* for more information.

6) <u>Invasive species</u>: (2.8 rank)

Submerged Aquatic Vegetation Habitat

A) *Species*: Alewife, American eel, American shad, blueback herring, and hickory shad

B) *Threats*:

1) <u>Sedimentation</u>: (2.1 rank) See *Rivers and Streams Habitat* for more information.

2) <u>Hydrologic regime changes</u>: (1.4 rank) See *Tidal Mudflats Habitat* for more information.

3) <u>Stormwater erosion</u>: (2.4 rank) See *Rivers and Streams Habitat* for more information.

4) <u>Pollution</u>: (2.1 rank) See *Rivers and Streams Habitat* for more information.

5) <u>Invasive species</u>: (2.2 rank)

6) <u>Recreation</u>: (1 rank)

7) <u>Habitat loss</u>: (2.6 rank) Habitat loss is caused by poor water quality and physical erosion and scouring. High turbidity, often caused by wind and wave induced erosion in aquatic systems, and overland stormwater erosion in terrestrial environments, prohibits light penetration needed for vegetative growth. Physical erosion and scouring of stream and river bottoms by either high flows or ice can cause the uprooting of established plants. All of these processes are negatively affecting SAV in the District.

Citation

Government of the District of Columbia, Department of Health, Environmental Health Administration, Fisheries and Wildlife Division. 2005. District of Columbia Comprehensive Wildlife Conservation Strategy.

<u>Virginia</u>

Species

Virginia categorized SGCN into four tiers, which are rankings separate from the Federal and Virginia endangered species lists. There are also six section-level ecoregions within Virginia. Within these ecoregions, ecoregional drainage units (EDU) have been delineated. The diadromous fish species have been identified as occurring within the following ecoregions (listed first) and EDUs (in parentheses). Because none of the diadromous fish species in Virginia are categorized as tier I species, individual threats are not identified for these species within their EDUs. The following diadromous fish species were listed as SGCN:

Atlantic sturgeon – Tier II

This species has a very high conservation need because it has a high risk of extinction or extirpation from the state. Ecoregional delineations include: Coastal Plain (James EDU; York EDU; Rappahannock EDU; Potomac EDU).

Alewife - Tier IV

This species has a moderate conservation need because the species may be in rare parts of its range, particularly on the periphery. Ecoregional delineations include: Coastal Plain (James EDU; York EDU; Rappahannock EDU; Potomac EDU; Delmarva EDU; Chesapeake Bay EDU; Albemarle Sound EDU) and Piedmont (James EDU; York EDU; Rappahannock EDU; Potomac EDU).

American shad – Tier IV

This species has a moderate conservation need because the species may be in rare parts of its range, particularly on the periphery. Ecoregional delineations include: Coastal Plain (James EDU; York EDU; Rappahannock EDU; Potomac EDU; Delmarva EDU; Chesapeake Bay EDU; Albemarle Sound EDU) and Piedmont (Chowan EDU; James EDU; York EDU; Rappahannock EDU; Potomac EDU).

American eel – Tier IV

This species has a moderate conservation need because the species may be in rare parts of its range, particularly on the periphery. Ecoregional delineations include: Coastal Plain (James EDU; York EDU; Rappahannock EDU; Potomac EDU; Delmarva EDU; Chesapeake Bay EDU; Albemarle Sound EDU), Piedmont (Chowan EDU; James EDU; York EDU; Rappahannock EDU; Potomac EDU; Pee Dee EDU), Blue Ridge (Roanoke EDU; Pee Dee EDU; James EDU; Rappahannock EDU; Potomac EDU), and Ridge and Valley (Roanoke EDU; James EDU; Potomac EDU).

Habitats

In addition to identifying SCGN contained within EDUs, habitat groups have been identified for SGCN, as well as the associated threats for those habitats. The following lists contain the source of threat, the threat itself, and the scope and severity of the threat in parenthesis (scale = 1 (least severe) to 4 (most severe); U is for unknown). Please note that although blueback herring and hickory shad were not identified as SGCN within the state, they occur within many rivers and streams throughout the state. Thus, it is likely that some of the threats identified may also apply to these species.

Chowan River Habitat

A) *Species*: Alewife, American shad, and American eel

B) *Sources of threat*:

       1) Industrial – mineral extraction: This causes turbidity alteration (2, 3).

2) <u>Industrial – other</u>: This causes hydrologic regime alteration from water supply dams (1, 3), nutrient input regime alteration from paper mills (1, 3), turbidity alteration from paper mills (1, 3), dissolved oxygen regime alteration from paper mills (1, 2), and organic pollutants from paper mills (1, 2).

3) <u>Forestry</u>: This causes sediment load alteration (4, 3).

4) <u>Municipal development</u>: This causes nutrient input regime alteration (from wastewater treatment plants, straight pipes, septic systems from Franklin and Emporia (1, 2), sediment load alteration from Franklin and Emporia (1, 2), toxins from Franklin and Emporia (1, 2), and hydrologic regime alteration from water supply extraction (1, 2).

5) <u>Agriculture</u>: This causes herbicides and fungicides (4, 2), insecticides (4, 2), toxins from pig farm lagoon spills (3, 4), sediment load alteration (3, 3), dissolved oxygen regime alteration from pig farms (3, 2), and nutrient input regime alteration from pig farms (3, 2).

Delmarva Peninsula Habitat

A) *Species*: Alewife, American shad, and American eel

B) *Sources of threat*:

1) <u>Industrial – rights-of-way</u>: This causes organic pollutants from roads and railways (2, 1), and herbicides and fungicides from roads and railways (2, 1).

2) <u>Industrial – other</u>: This causes toxins from spills on roads and rails (2, 1).

3) <u>Municipal development</u>: This causes nutrient input regime alteration from septic systems (2, 3), and channel and shoreline alteration from installation of bulkheads (2, 2).

4) <u>Agriculture</u>: This causes herbicides and fungicides from poultry and tomatoes (4, 3), insecticides from tomatoes and other crops (4, 3), dissolved oxygen regime alteration from poultry and tomatoes (4, 3), nutrient input regime alteration from poultry and tomatoes (4, 4), and organic matter input regime alteration from poultry and tomatoes (4, 4).

James River Habitat

A) *Species*: Alewife, American shad, American eel, and Atlantic sturgeon

B) *Sources of threat*:

1) <u>Industrial – mineral extraction</u>: This causes sediment load alteration from sand mining in Coastal Plain (1, 1), and turbidity alteration from sand mining in Coastal Plain (1, 1).

2) <u>Industrial – power generation</u>: This causes habitat fragmentation from dams (3, 3), and metals (2, 3).

3) <u>Industrial – rights-of-way</u>: This causes organic pollutants from roads and railways (3, 3), and herbicides and fungicides from roads and railways (3, 2).

4) <u>Industrial – other</u>: This causes toxins from industry particularly around Hopewell (2, 4), toxins from spills on roadways and rails, and accidents at industrial sites (1, 4), and habitat fragmentation from remnant mill dams) (3, 3).

5) <u>Forestry</u>: This causes organic matter input regime alteration (1, 1), sediment load alteration (3, 3), and turbidity alteration (2, 1).

6) <u>Municipal development</u>: This causes nutrient input regime alteration (4, 3), nutrient input regime alteration from wastewater treatment plants and straight pipes (2, 4), dissolved oxygen regime alteration (2, 3), channel or shoreline alteration (2, 4), other toxins from pharmaceuticals and drugs in wastewater (U, U), hydrologic regime alteration from water withdrawal (1, 3), hydrologic regime alteration from dam installation for water sources (1, 3), and turbidity alteration from road building and bridges (1, 2).

7) <u>Other land management</u>: This causes channel or shoreline alteration from landowners in the stream (1, 3).

8) <u>Agriculture</u>: This involves herbicides and fungicides (4, 3), insecticides (4, 3), sediment load alteration (4, 3), dissolved oxygen regime alteration (2, 3), channel or shoreline alteration (4, 2), turbidity alteration (4, 3), and organic matter input regime alteration (2, 1).

9) <u>Atmospheric deposition</u>: This causes pH regime alteration (2, 3).

Piankatank River Habitat

A) *Species*: Alewife

B) *Sources of threat*:

1) <u>Atmospheric deposition</u>: This causes toxins from aerial mercury from power plants) (4, 2).

2) <u>Forestry</u>: This causes sediment load alteration (3, 2).

3) <u>Agriculture</u>: This causes sediment load alteration (2, 2).

Potomac River Habitat

A) *Species*: Atlantic sturgeon, Alewife, American shad, and American eel

B) *Sources of threat*:

1) <u>Industrial – mineral extraction</u>: This causes sediment load alteration (1, 1), and turbidity alteration (1, 1).

2) <u>Industrial – power generation</u>: This causes habitat fragmentation from dams (3, 3), metals from atmospheric deposition (2, 3), pH regime alteration from acid precipitation (2, 3), and unintentional capture or killing of eels killed in turbines (2, 2).

3) <u>Industrial – rights-of-way</u>: This causes organic pollutants from roads and railways (3, 3), and herbicides and fungicides from roads and railways (3, 2).

4) <u>Industrial – other</u>: This causes toxins (2, 4), toxins from Shenandoah spills and others (3, 2), toxins from spills and accidents at industrial sites (1, 4), and habitat fragmentation from remnant mill dams (3, 3).

5) <u>Forestry</u>: This causes turbidity alteration (2, 1), organic matter input regime alteration (2, 1), and sediment load alteration (3, 3).

6) <u>Municipal development</u>: This causes nutrient input regime alteration (3, 2), nutrient input regime alteration from wastewater treatment plants and straight pipes (2, 4), channel or shoreline alteration (3, 4), dissolved oxygen regime alteration (3, 3), herbicides and fungicides (3, 2), insecticides (3, 2), turbidity alteration from road building/bridges (1, 2), toxins from pharmaceuticals and their by-products (U, U), hydrologic regime alteration from impervious surfaces (3, 4), hydrologic regime alteration from water withdrawal (2, 3), and hydrologic regime alteration from dam installation for water sources (2, 3).

7) <u>Other land management</u>: This causes channel or shoreline alteration from landowners bulldozing in streams (1, 3).

8) <u>Agriculture</u>: This involves herbicides and fungicides (4, 3), insecticides (4, 3), toxins from poultry farms and other livestock (3, 2), sediment load alteration (4, 3), dissolved oxygen regime alteration (3, 3), nutrient input regime alteration from poultry farms and other livestock (3, 4), channel or shoreline alteration (4, 2), turbidity alteration (4, 3), and organic matter input regime alteration (2, 1).

9) <u>Exotic or introduced species</u>: This causes competition from zebra mussels (1, 4), competition from snakehead (1, 2), and predation from snakehead (1, 2).

Rappahannock River Habitat

A) *Species*: Atlantic sturgeon, Alewife, American shad, and American eel

B) *Sources of threat*:

1) <u>Industrial – mineral extraction</u>: This causes sediment load alteration from sand mines in Coastal Plain (1, 1).

2) <u>Industrial – power generation</u>: This causes habitat fragmentation from dams (1, 1), metals from atmospheric deposition (2, 3), and pH regime alteration from acid precipitation (2, 3).

3) <u>Industrial – rights-of-way</u>: This causes organic pollutants from roads and railways (3, 2), and herbicides and fungicides from roads and railways (2, 2).

4) <u>Industrial – other</u>: This causes toxins from spills and accidents at industrial sites (1, 4), toxins from various industry in and below Fredericksburg (1, 2), and habitat fragmentation from remnant mill dams (3, 3).

5) Forestry: This causes organic matter input regime alteration (1, 1), and sediment load alteration (2, 2).

6) Municipal development: This causes nutrient input regime alteration from wastewater treatment plans and straight pipes (2, 4), channel or shoreline alteration (2, 4), dissolved oxygen regime alteration (2, 2), turbidity alteration from road and bridge building (1, 2), toxins from pharmaceuticals and their by-products (U, U), and hydrologic regime alteration from water withdrawal (1, 1).

7) Other land management: This causes channel or shoreline alteration from landowners bulldozing in stream (1, 3).

8) Agriculture: This involves herbicides and fungicides (4, 3), insecticides (4, 3), sediment load alteration (4, 3), dissolved oxygen regime alteration (2, 2), nutrient input regime alteration (4, 3), channel or shoreline alteration (4, 2), and turbidity regime alteration (4, 3).

9) Exotic or introduced species: This causes competition from blue catfish (2, 1), and predation from blue catfish (2, 1).

Roanoke River Habitat

A) *Species*: American eel

B) *Sources of threat*:

1) Industrial – mineral extraction: This causes sediment load alteration from sand mines in Coastal Plain (2, 2), and turbidity alteration from sand mines in Coastal Plain (2, 2).

2) Industrial – power generation: This causes habitat fragmentation from dams (4, 3), hydrologic regime from dams (4, 3), metals (2, 3), and water temperature regime alteration from Philpott Dam operations (1, 4).

3) Industrial – rights-of-way: This involves organic pollutants from roads and railways (2, 2), and herbicides and fungicides from roads and railways (3, 2).

4) Industrial – other: This causes toxins (2, 3), toxins from spills on roads and rails and accidents at industrial sites (1, 4), and habitat fragmentation from remnant mill dams (3, 3).

5) Forestry: This causes organic matter input regime alteration (1, 1), and sediment load alteration (3, 3).

6) Municipal development: This causes nutrient input regime alteration from wastewater treatment plans and straight pipes (2, 4), channel or shoreline alteration (2, 4), channel or shoreline alteration from alteration of Roanoke River at Roanoke (1, 2), dissolved oxygen regime alteration (2, 2), turbidity alteration from road and bridge building (2, 2), other toxins from pharmaceuticals/drugs and their by-products (U, U), and hydrologic regime alteration from water withdrawal (1, 2).

7) Other land management: This causes channel or shoreline alteration from landowners bulldozing in streams (1, 3).

8) Agriculture: This involves herbicides and fungicides (4, 3), insecticides (4, 3), sediment load alteration (4, 3), dissolved oxygen regime alteration (2, 2), nutrient input regime alteration (4, 3), nutrient input regime alteration from aquaculture (1, 1), channel or shoreline alteration (4, 2), turbidity alteration (4, 3), organic matter input regime alteration (2, 1), and parasitism (1, 1).

9) Introduced/exotic species: This causes competition from blue and flathead catfish (2, 1; scope of effects on mainstem species is higher, 3), and predation from blue and flathead catfish (2, 1; scope of effects on mainstem species is higher, 3).

York River Habitat

A) *Species*: Alewife, American shad, American eel, and Atlantic sturgeon

B) *Sources of threat*:

1) Industrial – mineral extraction: This causes sediment load alteration from sand mines in Coastal Plain (1, 1).

2) Industrial – power generation: This causes habitat fragmentation from Lake Anna (1, 2), and metals from atmospheric mercury (2, 3).

3) Industrial – rights-of-way: This involves organic pollutants from roads and railways (3, 2), and herbicides and fungicides from roads and railways (2, 2).

4) Industrial – other: This causes toxins from paper mill and oil refinery at mouth (2, 3), toxins from spills on roadways and rails and accidents at industrial sites (1, 4), and habitat fragmentation from remnant mill dams (3, 3).

5) Forestry: This causes turbidity alteration (3, 2), organic matter input regime alteration (3, 2), and sediment load alteration (2, 2).

6) Municipal development: This causes nutrient input regime alteration from wastewater treatment plans, straight pipes (1, 2), channel or shoreline alteration (2, 4), turbidity alteration from road and bridge building (1, 2), dissolved oxygen regime alteration (1, 1), and hydrologic regime alteration from water withdrawal and the proposed King William reservoir (1, 2).

7) Municipal: This involves other toxins from pharmaceutical/drugs and their by-products (U, U).

8) Agriculture: This involves herbicides and fungicides (4, 3), insecticides (4, 3), sediment load alteration (4, 3), dissolved oxygen regime alteration (2, 2), nutrient input regime alteration (4, 3), channel or shoreline alteration (4, 2), turbidity alteration (3, 2), and organic matter input regime alteration (3, 2).

9) Invasive species: This causes competition from blue catfish (2, 1), and predation from blue catfish (2, 1).

Citation

Virginia Department of Game and Inland Fisheries (VA DGIF). 2005. Virginia's Comprehensive
    Wildlife Conservation Strategy.

North Carolina

Habitats

Threats have been identified for freshwater and marine habitats, but only priority
conservation status species-habitat associations are listed. At this time, ***Atlantic sturgeon*** is the
only diadromous fish species identified as having priority conservation status within the state.
Since habitats were grouped by river basins, additional sources (including tables on the DVD
supplement to this document) were used to list known presence of other diadromous fish species
within these habitats. These species include: American shad, hickory shad, alewife, blueback
herring, and Atlantic sturgeon. Therefore, it is likely that some of the threats identified for
Atlantic sturgeon under the various habitats may also apply to these species. The following
species-habitat associations have been identified, and relevant threats are presented below:

Roanoke River Basin Habitat

A) *Species*: Atlantic sturgeon is a priority aquatic species that is present in this habitat; also
    present are American shad, hickory shad, alewife, blueback herring, and striped bass

B) *Threats*:

  1) Sedimentation: Agriculture, forestry, and construction have degraded water
      and habitat quality.

  2) Contamination: Dioxin, selenium (from historic discharge from ash pond
      basins), and mercury levels are degrading aquatic habitats.

  3) Water withdrawls: Current and future water withdrawals have the potential to
      reduce flows to the lower Roanoke River and increase salinity levels
      downstream.

  4) Non-point source pollution

  5) Point source pollution: Sources include: municipal wastewater treatment
      plants, selenium ash pond discharge, industrial facilities, small package
      treatment plants, and urban and industrial stormwater systems. Wastewater
      treatment plants can cause elevated nitrogen, phosphorus, copper, and fecal
      coliform levels. They have also led to elevated ammonia nitrogen (NH_3)
      concentrations at San Souci.

  6) Growth: Especially in Stokes and Granville counties, growth will affect land
      use, cover, and water quality.

  7) Dams: Amount and timing of water releases from dams, particularly along
      the Roanoke River, can alter downstream aquatic and riparian flora and fauna.
      Changes in flow regime in the lower mainstem Roanoke River, and associated

draining of the backswamps, is likely to be partially responsible for increased frequency of low dissolved oxygen (less than 5 mg/l), primarily in June.

Cape Fear River Basin Habitat

A) *Species*: Atlantic sturgeon is a priority aquatic species that is present in this habitat; also present are American shad, hickory shad, alewife, and blueback herring

B) *Threats*:

    1) Water quality degradation: This impairment has been caused by: sediment, fecal coliform, ammonia, chlorides, low dissolved oxygen, turbidity, nutrients, mercury, and other point and non-point source pollutants.

    2) Sedimentation: This issue comes from agriculture, forestry, construction, and stormwater discharge in urbanized areas.

    3) Locks and dams: These obstructions block migration routes for diadromous and resident species, reduce recolonization and dispersal potential, and create unnatural flow regimes.

Neuse River Basin Habitat

A) *Species*: Atlantic sturgeon is a priority aquatic species that is present in this habitat; also present are American shad, hickory shad, alewife, and blueback herring

B) *Threats*:

    1) Animal waste: The byproducts from animals and fertilizers increase levels of nitrates and phosphates; this, in turn, can lead to excess growth of aquatic plants (such as algae), and decreased dissolved oxygen levels (especially during summer months), resulting in fish kills.

    2) Channelization: Channelization of streams for agriculture can cause bank erosion.

    3) Forestry: This activity contributes 13% and 6% of nitrogen and phosphorus, respectively.

    4) Dams and other impoundments: These structures affect aquatic species by altering water hydrology and habitat, reducing flows and dissolved oxygen, and causing erosion. Modification of flow regimes by upstream impoundments impact various life history characteristics of downstream migratory fishes and other aquatic fauna, such as limiting dispersal and recolonization.

    5) Water withdrawals: Withdrawals for irrigation reduce the quantity of available habitat and alter water hydrology.

    6) Water demands and wastewater discharges: These issues have increased from the growing population. There are over 400 point source waste discharge permits for the basin from municipal wastewater treatment plants, industrial

facilities, small package treatment plants, and large urban and industrial stormwater.

7) <u>Sedimentation</u>: Losses of natural areas and increases in impervious surfaces from construction lead to high sediment runoff. there is also increased lawn fertilizer runoff from more homes, and heavy metal runoff, which contributes to elevated mercury levels in fish tissue.

8) <u>Atmospheric deposition</u>: This comes from nitrogen in cars and factories, which can lead to decreased water quality.

9) <u>Non-point source pollution</u>: Large quantities of nutrients, especially nitrogen, from non-point sources are considered the greatest threat to water quality of the Neuse River estuary.

Tar-Pamlico River Basin Habitat

A) *Species*: Atlantic sturgeon is a priority aquatic species that is present in this habitat; also present are American shad, hickory shad, alewife, and blueback herring

B) *Threats*:

1) <u>Sedimentation</u>: This results from land clearing activities, streambank erosion, and channelization associated with construction and agriculture.

2) <u>Agriculture</u>: Activities including swine, dairy, and poultry, contribute to nutrient inputs, erosion, and sedimentation. Influxes of sediment reduce the quality and quantity of necessary habitat for aquatic organisms.

3) <u>Water withdrawals</u>: These activities, plus inter-basin transfers, reduce the quantity of available habitat for aquatic species.

4) <u>Growth</u>: Increased drinking water, wastewater discharge, and stormwater control from a growing population cause problems for aquatic species.

5) <u>Urban expansion</u>: Cumulative and secondary impacts due to urban expansion (e.g., greater Raleigh and Rocky Mount) will cause increased impervious surfaces, which in turn may lead to increased stream sedimentation.

6) <u>Point source pollution</u>: Discharges from municipal wastewater treatment plants, industrial facilities, small package treatment plants, large urban and industrial stormwater systems, degrade water quality. Wastewater treatment plant effluent increases conductivity, elevates nitrogen levels, and lowers dissolved oxygen.

Chowan River Basin Habitat

A) *Species*: Atlantic sturgeon is a priority aquatic species that is present in this habitat; also present are American shad, hickory shad, alewife, and blueback herring

B) *Threats*:

1) The Chowan River was classified as "nutrient sensitive waters" in 1979 (NCDWQ 2002) as a result of excessive levels of nitrogen and phosphorus in

wastewater and runoff. Chronic episodes of hypoxia exist in the river and its tributaries from late June through September during most years. Dissolved oxygen levels frequently fall below 3.0 mg/l, which negatively affects aquatic biota. Cyclonic events and their accompanying rainfall, storm surge, inundation and flushing of bottomland swamp habitats have occurred repeatedly within the basin since 1995.

2) <u>Non-point source pollution</u>: Degradation of water quality results from: agriculture, animal operation, urban development, forestry, stormwater discharge, rural residential development, hydrologic modifications, and septic systems.

3) <u>Point source pollution</u>: Point sources may include: municipality waste water treatment plants, industrial facilities, and urban and industrial stormwater systems. As of 2001, there were 11 permitted wastewater discharges and 34 registered animal operations in the basin.

4) <u>Water withdrawals</u>: These withdrawals are made for agriculture purposes.

Pasquotank River Basin Habitat

A) *Species*: Atlantic sturgeon is a priority aquatic species that is present in this habitat; also present are alewife and blueback herring

B) *Threats*:

1) <u>Physical habitat destruction</u>: This is the primary threat within this basin, and results from loss of riparian vegetation, straightening of streams, erosion of banks, and reductions of aquatic vegetation.

2) <u>Water withdrawals</u>: These withdrawals are made for agriculture purposes.

3) <u>Non-point source pollution</u>: The point sources that degrade water quality include: agriculture, animal operation, urban development, forestry, stormwater discharge, rural residential development, hydrologic modifications, and septic systems.

4) <u>Point-source pollution</u>: The non-point sources that degrade water quality may include: municipal wastewater treatment plants, industrial facilities, reverse-osmosis water treatment facilities, and urban and industrial stormwater systems. As of 2001, there were 34 permitted wastewater discharges, 51 general stormwater permits, and 29 registered animal operations in the basin .

5) <u>Growth</u>: Increasing population growth in the basin will continue to put more pressure and demand on wastewater treatment systems.

White Oak River Basin Habitat

A) *Species*: Atlantic sturgeon is a priority aquatic species that is present in this habitat; also present are American shad, alewife, and blueback herring

B) *Threats*:

    1) <u>Eutrophication</u>: Excessive nutrient input from such things as wastewater treatment plants, industry, agriculture, and hog/chicken farms degrade water quality.

    2) <u>Wastewater discharge</u>: In the White Oak River basin there are 50 permitted discharges, four of which are major discharges with greater than, or equal to, 1 million gallons per day.

Marine Habitat

A) *Species*: Atlantic sturgeon is a priority aquatic species that is present in this habitat; also present are American shad, blueback herring, and striped bass

B) *Threats*:

    1) <u>Vessel interaction</u>: This includes collisions; higher frequencies occur in areas that have heavy boating and vessel traffic.

    2) <u>Oil and gas exploration</u>: Oil deposits on the ocean floor can reduce food sources for all marine species and result in ingestion of tar balls.

    3) <u>Dredging</u>: Dredging in navigation channels and boat basins, especially areas with fine sediment and low flushing, can cause direct destruction or degradation of habitat and/or incidental take of marine species. Additionally, channelization of inshore and nearshore habitats can result in the disposal of dredge material in shallow habitats, impacting foraging grounds. Channelization of streams and ditching can also lead to hydrologic modifications.

    4) <u>By-catch</u>: By-catch of marine organisms occurs in a number of different fisheries, some of which may cause injury or kill fish.

    5) <u>Entrainment</u>: Saltwater cooling intake systems at coastal power plants have been reported to entrap marine species.

    6) <u>Explosives</u>: Use of underwater explosives to remove abandoned oil platforms, for military activities, or for oil exploration can result in injury or death to marine species in the vicinity of the explosion.

    7) <u>Dams and other impoundments</u>: These structures obstruct and modify water flow to the coast; there are over 2,000 dams in North Carolina.

    8) <u>Water withdrawals</u>: These withdrawals result in hydrologic changes.

    9) <u>Road fill and culverts</u>: These activities cause obstructions and flow alterations.

    10) <u>Forestry</u>: Log salvage operations may impact anadromous fish nursery areas.

    11) <u>Growth</u>: Development and excessive impervious cover degrades water quality.

12) Eutrophication: Loading of nutrients from sources such as sewage treatment facilities, land disposal sites, onsite wastewater treatment facilities, agricultural sources, homeowners, and golf courses, has the potential to degrade water quality.

13) Sedimentation: This occurs from erosion along the coast.

14) Contamination: Fecal coliform bacteria from sewage treatment facilities, stormwater outfalls, and possibly oceanfront septic systems, contaminate the water supply. Additionally, toxic chemicals from sources such as roads, parking lots, associated transportation, marine wood preservatives, dredging, and marina development, all impact habitat.

15) Invasive species

Citation

North Carolina Wildlife Resources Commission (NC WRC). 2005. North Carolina Wildlife Action Plan.

South Carolina

Species

American eel – SNR ranking; highest priority ranking for greatest conservation concern

American shad – S5 state ranking; highest priority ranking for greatest conservation concern

Atlantic sturgeon – S3 state ranking; highest priority ranking for greatest conservation concern

Blueback herring – S3 state ranking; highest priority ranking for greatest conservation concern

Hickory shad – S4 state ranking; highest priority ranking for greatest conservation concern

Striped bass – SNR ranking; moderate priority ranking for greatest conservation concern

Threats

In addition to discussing the general effects of different threats that challenge species present in the state, the SWAP identifies specific threats to the following species and species groupings:

Alosines (includes American Shad, Hickory Shad, and Blueback Herring)
   A) *Watersheds:* Waccamaw-Pee Dee, Santee-Cooper and Savannah River Basins

B) *Threats*:

1) <u>Dams</u>: Dams restrict migrations, and have eliminated populations of alosines from historical habitats. The result has been a general reduction in alosine populations, even in currently accessible river reaches. The Santee Basin has nearly 45 dams in the South Carolina portions of the basin alone.

2) <u>Water withdrawal</u>: Tidal freshwater marshes along the Cooper River (many of which are relic rice impoundments with breached or eroded dikes), which were used extensively as spawning habitat by blueback herring prior to rediversion of flows into the Santee River, are less extensive under reduced flows, and many are now partly dewatered or influenced by brackish water. The flow regimens in both the Cooper and Santee Rivers is typically in highs and lows (with more abrupt changes from peaked power generation and flood releases) than are characteristic of more gradual river flow changes that occur in open rivers where waters expand into, and withdraw from, floodplains.

3) <u>Fish passage</u>: Fish passage efficiency for blueback herring at the St. Stephen Dam is low. Fish passage designs and flow protocols currently used at dams on the lower Santee-Cooper Basin were initially designed for passing blueback herring into the lakes for forage and do not maximize passage efficiency for alosines in either direction. Dams on the Santee-Cooper Basin that currently incorporate passage for alosines, do not employ methodologies that accommodate timely outmigration and maximized survival of post-spawning adults or emigrating juveniles.

4) <u>Predation</u>: Large concentrations of double-crested cormorants (*Phalacrocorax auritus*) occur immediately below dams. The cormorant population has increased dramatically over the past decade, and these birds have been shown to feed heavily on alosines (up to 64.5% of diet). Although the impact of cormorant predation on alosine populations has not been quantified, it appears that cormorants have to potential to negatively impact both upstream passage success for blueback herring and out-of-lake passage for all juvenile alosines.

5) <u>Invasive species</u>: Competition and predation from non-native species, in particular flathead catfish (*Pylodictis olivaris*) and blue catfish (*Ictalurus furcatus*), may be additive to 'more natural' sources of mortality, and may be particularly problematic below dams where catfish density is often high.

American Eel

<u>Note</u>: Due to its complicated life cycle, the American eel population faces a broad range of challenges, some of which are specific to a particular growth stage. Since males and females largely utilize separate habitats, impacts in a given region may affect the sex ratio of the eel population.

A) *Watersheds:* Pee Dee, Edisto, and Santee River Basins

B) *Threats*:

    1) <u>Dams</u>: Dams and causeways obstruct access to a diversity of habitats, which may limit basin-specific and statewide populations. The Pee Dee, Edisto, and Santee coastal drainages have suffered an 83% reduction in unobstructed stream habitat.

    2) <u>Invasive species</u>: Issues exist, particularly with flathead catfish (*Pylodictis olivaris*) and blue catfish (*Ictalurus furcatus*). Both of these catfish are piscivorous and opportunistic; they will feed on any fish that can fit in their mouths. Additionally, non-indigenous pathogens or parasites such as the Asian swimbladder nematode (*Anguillicola crassus*), has been shown to have significant negative impacts on the European eel (*Anguilla anguilla*) and on captive American eels in South Carolina and Texas.

Atlantic Sturgeon

A) *Watersheds*: Waccamaw-Pee Dee, Santee-Cooper, and Savannah River Basins

B) *Threats:*

    1) <u>Dams</u>: Obstructed access to a diversity of habitats may limit basin-specific populations of both Atlantic and shortnose sturgeon. Dams can block spawning migrations and severely restrict the availability of spawning and nursery habitat, particularly in large river systems when dams are near the coast, as in the Santee River. Dams and other impediments to migration have eliminated sturgeons from many historical habitats in South Carolina; the result being a general reduction in sturgeon populations in even currently accessible river reaches. Reduced flows caused by dams can also reduce dissolved oxygen to levels unsuitable for sturgeon.

    2) <u>Fish passage</u>: Both the Pinopolis navigational lock and St. Stephen fish passage facility provide passage for blueback herring and American shad. However, these facilities do not effectively pass sturgeons, nor do they incorporate efficient outmigration technologies, even for alosines. Effective passage designs for sturgeons have not yet been determined. In fact, poorly designed fish passage facilities may negatively impact sturgeon populations by increasing mortality.

    3) <u>Contamination</u>: Bioaccumulation of contaminants, such as dioxin, in parts of Winyah Bay, may reduce productivity or increase susceptibility to diseases or stress.

Striped Bass

A) *Watersheds*: Savannah and Pee Dee Rivers

B) *Threats*:

    1) <u>Sedimentation</u>: Clearing forests and riparian areas of coastal rivers and their tributaries have caused this problem.

2) Water temperature: Increased temperatures have resulted from clearing forests and riparian areas of coastal rivers and their tributaries. Warmer water temperatures may decrease the amount of summertime refuge habitat for striped bass and negatively impact reproduction.

3) Hydrologic modification

4) Overfishing

5) Dams: Dams disrupt migrations and altering thermal and hydrologic regimes. The presence of impoundments along the Savannah and Pee Dee Rivers may partially account for limited reproduction in those systems.

Citation

South Carolina Department of Natural Resources. 2005. South Carolina Comprehensive Wildlife Conservation Strategy.

Georgia

Species

At this time, none of the diadromous fish species are listed as SGCN by the state of Georgia and no habitat associations have been identified in their wildlife action plan. However, the designation of high priority waters in the Southern Coastal Plain Ecoregion was based, in part, on suitable habitat for diadromous fish species, as well as others.

Habitat

Given that diadromous species can be found in the Southern Coastal Plain Ecoregion, threats identified for that region are highlighted below. However, no individual species are identified with any particular threat.

Threats:

1) Development: This has resulted in habitat loss and fragmentation.

2) Water withdrawals

3) Dams: These obstructions result in altered hydrological regimes and sediment transport processes.

4) Eutrophication: These impacts on systems from human activities include: increased flow variability, reduced dissolved oxygen, and increased silt loads.

5) Invasive species

6) Global warming

Citation

Georgia Department of Natural Resources, Wildlife Resources Division. 2005. A
Comprehensive Wildlife Conservation Strategy for Georgia.

Florida

Species

Many of the diadromous fish species are listed as SGCN under Florida's Comprehensive
Wildlife Conservation Strategy. Although state ranking information is not provided, a number of
criteria were used to determine eligibility. The following species have been listed, with their
status and trend indicated:

American eel – low status; unknown trend

American shad – low status; declining trend

Atlantic sturgeon – low status; declining trend

Blueback herring – low status; unknown trend

Hickory shad – low status; declining trend

Striped bass – low status; stable trend

Habitats

Species-habitat associations have been identified, as well as threats and sources of threat.
The sources of threat are ranked and their corresponding threats are also ranked. The threat levels
are as follows: VH=very high; H=high; M=medium; and L=low.

Calcareous Stream Habitat

Statewide Threat Rank of Habitat: High

   A) *Species*: Striped bass

   B) *Sources of Threat*:

      1) Nutrient loads (H in urban and agriculture areas): This has caused altered
species composition/dominance (H), and altered water quality of surface water
or aquifers by nutrients (H).

      2) Invasive plants (H): This has caused altered species composition/dominance
(H).

      3) Invasive animals (M): This has caused altered species
composition/dominance (H), and erosion/sedimentation (H).

4) <u>Development</u> (M): Conversion to housing and urban development has caused altered water quality of surface water or aquifers by nutrients (H), erosion/sedimentation (H), and altered landscape mosaic or content (M).

5) <u>Chemicals and toxins</u> (M): This has caused altered water quality of surface water or aquifer by contaminants (M).

6) <u>Roads</u> (M): This has caused erosion/sedimentation (H).

7) <u>Forestry</u> (L): This has caused altered species composition/dominance (H), and erosion/sedimentation (H).

8) <u>Agriculture</u> (L): This has caused altered water quality of surface water or aquifers by nutrients (H), and erosion/sedimentation (H).

9) <u>Mining/drilling</u> (L): This has caused erosion/sedimentation (H).

Coastal Tidal River or Stream Habitat

Statewide Threat Rank of Habitat: Very High

A) *Species*: Atlantic sturgeon, American eel, blueback herring, hickory shad, American shad, and striped bass

B) *Sources of Threat*:

1) <u>Water withdrawal</u> (H): This has caused altered species composition/ dominance (H), altered hydrologic regime (H), altered landscape mosaic or content (H), altered water salinity, pH, conductivity, or other physical water quality characteristics of surface water or aquifers (M), and altered community structure (M).

2) <u>Channel modifications/shipping lanes</u> (H): This has caused altered species composition/dominance (H), altered hydrologic regime (H), habitat destruction or conversion (M), altered water salinity, pH, conductivity, or other physical water quality characteristics of surface water or aquifers (M), and altered community structure (M).

3) <u>Dam operations</u> (H): This has caused altered species composition/dominance (H), altered hydrologic regime (H), altered water salinity, pH, conductivity, or other physical water quality characteristics of surface water or aquifers (M), altered community structure (M), and fragmentation of habitats, communities, and ecosystems (M).

4) <u>Conversion to housing and urban development</u> (H): This has caused altered hydrologic regime (H), altered landscape mosaic or content (H), and habitat destruction or conversion (M).

5) <u>Shoreline hardening</u> (H): This has caused altered species composition/ dominance (H), habitat destruction or conversion (M), fragmentation of habitats, communities, and ecosystems (M), and altered community structure (M).

6) <u>Management of nature – vegetable clearing/snagging for water conveyance</u> (M): This has caused altered species composition/dominance (H), altered hydrologic regime (H), fragmentation of habitats, communities, and ecosystems (M), and altered community structure (M).

7) <u>Roads</u> (M): This has caused habitat destruction or conversion (M).

8) <u>Chemicals and toxins</u> (M): This has caused altered species composition/ dominance (H), and altered water quality of surface water or aquifer by contaminants (M).

9) <u>Conversion to commercial and industrial development</u> (M): This has caused habitat destruction or conversion (M).

10) <u>Nutrient loads</u> (M): This has caused altered species composition/dominance (H), and altered water quality of surface water or aquifer by nutrients (M).

11) <u>Invasive plants</u> (M): This has caused altered species composition/dominance (H), and altered community structure (M).

12) <u>Invasive animals</u> (L): This has caused altered species composition/dominance (H).

13) <u>Sea level rise</u> (L): This has caused altered hydrologic regime (H).

Marine and Estuarine Habitats

Statewide Threat Rank of Habitat: Very High

A) *Sources of Threat*:

1) <u>Coastal development</u> (VH): This has caused altered hydrologic regime (VH), altered species composition (VH), habitat destruction (VH), missing key communities or functional guilds/trophic shift (H), and sedimentation contamination (M).

2) <u>Dam operations/incompatible release of water</u> (VH): This has caused altered hydrologic regime (VH), altered species composition (VH), altered water quality by contaminants (VH), altered water quality/physical chemistry (VH), habitat disturbance (VH), and altered water quality by nutrients (H).

3) <u>Channel modifications/shipping lanes</u> (VH): This has caused altered hydrologic regime (VH), altered water quality/physical chemistry (VH), habitat destruction (VH), habitat disturbance (VH), and sedimentation contamination (M).

4) <u>Inadequate stormwater management</u> (VH): This has caused altered hydrologic regime (VH), altered species composition (VH), altered water quality by contaminants (VH), altered water quality/physical chemistry (VH), habitat disturbance (VH), altered water quality by nutrients (H), and sedimentation contamination (M).

5) <u>Shoreline hardening</u> (VH): This has caused altered hydrologic regime (VH), and habitat destruction (VH).

6) <u>Management of nature (beach nourishment, impoundment)</u> (H): This has caused altered hydrologic regime (VH), altered species composition (VH), altered water quality by contaminants (VH), altered water quality/physical chemistry (VH), habitat disturbance (VH), and missing key communities or functional guilds/trophic shift (H).

7) <u>Chemicals and toxins</u> (H): This has caused altered water quality by contaminants (VH), and sedimentation (M).

8) <u>Industrial spills</u> (H): This has caused altered water quality by contaminants (VH), habitat disturbance (VH), and sedimentation (M).

9) <u>Incompatible industrial operations</u>: This has caused altered hydrologic regime (VH), altered species composition (VH), altered water quality by contaminants (VH), and missing key communities or functional guilds/trophic shift (H).

10) <u>Surface water withdrawal</u>: This has caused altered hydrologic regime (VH), altered species composition (VH), and altered water quality/physical chemistry (VH).

11) <u>Invasive animals</u> (H): This has caused altered species composition (VH), and habitat disturbance (VH).

12) <u>Invasive plants</u> (H): This has caused altered species composition (VH), and sedimentation contamination (M).

13) <u>Incompatible resource extraction: mining/drilling</u> (H): This has caused altered water quality/physical chemistry (VH).

14) <u>Climate variability</u> (H): This has caused altered weather regime/sea level rise (H).

15) <u>Nutrient loads</u> (H): This has caused altered water quality by nutrients (H).

16) <u>Utility corridors</u> (M): This has caused altered hydrologic regime (VH), and habitat destruction (VH).

17) <u>Vessels impacts</u> (M): This has caused habitat destruction (VH), and habitat disturbance (VH).

18) <u>Boating impacts</u> (M): This has caused habitat destruction (VH), and habitat disturbance (VH).

19) <u>Incompatible recreational activities</u> (M): This has caused altered species composition (VH), and habitat disturbance (VH).

20) <u>Groundwater withdrawal</u> (M): This has caused altered hydrologic regime (VH), altered species composition (VH), and altered water quality/physical chemistry (VH).

21) <u>Incompatible fishing pressure</u> (M): This has caused altered species composition (VH), and missing key communities or functional guilds/trophic shift (H).

22) <u>Solid waste</u> (M): This has caused habitat disturbance (VH).

23) Roads, bridges, and causeways (M): This has caused altered hydrologic regime (VH), habitat destruction (VH), and sedimentation contamination (M).

24) Thermal pollution (M): This has caused altered water quality/physical chemistry (VH).

25) Fishing gear impacts (M): This has caused habitat disturbance (VH).

Inlet Habitat

Statewide Threat Rank of Habitat: Very High

A) *Species*: American eel, Atlantic sturgeon, American shad, and blueback herring

B) *Sources of Threat*:

1) Channel modification/shipping lanes (H): This has caused habitat disturbance (H), altered water quality/physical chemistry (M), erosion (M), habitat destruction (M), altered hydrologic regime (M), and sedimentation (M).

2) Shoreline hardening (H): This has caused altered structure (M), erosion (M), habitat destruction (M), and sedimentation (M).

3) Dam operation/incompatible release of water (H): This has caused habitat disturbance (H), altered water quality/physical chemistry (M), altered hydrologic regime (M), and sedimentation (M).

4) Disruption of longshore transport of sediments (H): This has caused erosion (M), and sedimentation (M).

5) Coastal development (H): This has caused altered species composition (M), altered structure (M), altered water quality/physical chemistry (M), habitat destruction (M), and altered hydrologic regime (M).

6) Management of nature (beach nourishment, impoundments) (H): This has caused habitat disturbance (H), altered species composition (M), and sedimentation (M).

7) Boating impacts (H): This has caused habitat disturbance (H).

8) Incompatible boating activities (H): This has caused habitat disturbance (H)

9) Light pollution (H): This has caused altered species composition (M).

10) Industrial spills (M): This has caused habitat disturbance (H).

11) Harmful algal blooms (M): This has caused altered species composition (M).

12) Roads, bridges, and causeways (M): This has caused altered structure (M), habitat destruction (M), and altered hydrologic regime (M).

13) Inadequate stormwater management (M): This has caused altered species composition (M), altered water quality/physical chemistry (M), and altered hydrologic regime (M).

14) Incompatible industrial operations (M): This has caused altered species composition (M), and habitat destruction (M).

15) Invasive plants (M): This has caused altered species composition (M).

16) Acoustic pollution (M): This has caused habitat disturbance (H).

17) Vessel impacts (M): This has caused habitat disturbance (H), and habitat destruction (M).

18) Utility corridors (M): This has caused habitat disturbance (H).

19) Fishing gear impacts (M): This has caused habitat disturbance (H).

20) Military activities (M): This has caused habitat disturbance (H).

21) Invasive animals (M): This has caused habitat disturbance (H), and altered species composition (M).

22) Surface water withdrawal (M): This has caused altered water quality/physical chemistry (M).

Large Alluvial Stream Habitat

Statewide Threat Rank of Habitat: High

A) *Species*: American eel and striped bass

B) *Sources of Threat*:

1) Dam operations (H): This has caused altered species composition/dominance (M), altered community structure (M), habitat destruction or conversion (M), fragmentation of habitats, communities, and ecosystems (M), altered hydrologic regime (M), and erosion/sedimentation (M).

2) Management of nature-water control structures (H): This has caused altered species composition/dominance (M), altered community structure (M), habitat destruction or conversion (M), fragmentation of habitats, communities, and ecosystems (M), altered hydrologic regime (M), and erosion/sedimentation (M).

3) Channel modification/shipping lanes (H): This has caused altered species composition/dominance (M), altered community structure (M), habitat destruction or conversion (M), fragmentation of habitats, communities, and ecosystems (M), altered hydrologic regime (M), and erosion/sedimentation (M).

4) Invasive animals (M): This has caused altered species composition/dominance (M), altered community structure (M), habitat destruction or conversion (M), and erosion/sedimentation (M).

5) Surface water withdrawal (M): This has caused fragmentation of habitats, communities, and ecosystems (M), and altered hydrologic regime (M).

6) Groundwater withdrawal (L): This has caused altered hydrologic regime (M).

7) Incompatible forestry practices (L): This has caused altered species composition/dominance (M), altered community structure (M), habitat destruction or conversion (M), fragmentation of habitats, communities, and

ecosystems (M), altered hydrologic regime (M), and erosion/sedimentation (M).

8) Chemicals and toxins (L): This has caused altered species composition/dominance (M).

9) Incompatible recreational activities (L): This has caused altered species composition/dominance (M), altered community structure (M), habitat destruction or conversion (M), and erosion/sedimentation (M).

Softwater Stream Habitat

Statewide Threat Rank of Habitat: Very High

A) *Species*: Atlantic sturgeon and striped bass

B) *Sources of Threat*:

1) Surface water withdrawal (H): This causes fragmentation of habitats, communities, and ecosystems (H), altered hydrologic regime (H), altered landscape mosaic or context (H), and altered community structure (M).

2) Conversion to agriculture (H): This causes fragmentation of habitats, communities, and ecosystems (H), altered landscape mosaic or context (H), and altered community structure (M).

3) Nutrient loads from agriculture (H): This causes altered water quality of surface water or aquifer by nutrients (H).

4) Roads (H): This causes fragmentation of habitats, communities, and ecosystems (H), erosion/sedimentation (H), altered water quality of surface water or aquifer by nutrients (H), and habitat destruction or conversion (M).

5) Conversion to housing and urban development (H): This causes fragmentation of habitats, communities, and ecosystems (H), altered landscape mosaic or context (H), erosion/sedimentation (H), and habitat destruction or conversion (M).

6) Dam operations (M): This causes fragmentation of habitats, communities, and ecosystems (H), and altered hydrologic regime (H).

7) Nutrient loads from urban (M): This causes altered water quality of surface water or aquifer by nutrients (H).

8) Incompatible resource extraction: mining/drilling (M): This causes erosion/sedimentation (H), and habitat destruction or conversion (M).

9) Chemicals and toxins (M): This causes altered water quality of surface water or aquifer by contaminants (M).

10) Conversion to commercial and industrial development (M): This causes erosion/sedimentation (H), and habitat destruction or conversion (M).

11) Invasive species (M): This causes altered species composition/dominance (M).

12) Incompatible recreational activities (L): This causes erosion/sedimentation (H), and habitat destruction or conversion (M).

13) Incompatible forestry practices (L): This causes altered hydrologic regime (H), erosion/sedimentation (H), and habitat destruction or conversion (M).

14) Groundwater withdrawal (L): This causes altered hydrologic regime (H).

15) Incompatible agricultural practices (L): This causes altered hydrologic regime (H), and erosion/sedimentation (H).

Spring and Spring Run Habitat

Statewide Threat Rank of Habitat: Very High

A) *Species*: Atlantic sturgeon and striped bass

B) *Sources of Threat*:

1) Nutrient loads from urban (VH): This has caused altered species composition/dominance (VH), altered water quality of surface water or aquifer by nutrients (VH), altered community structure (H), and habitat destruction or conversion (H).

2) Invasive plants (VH): This has caused altered species composition/dominance (VH), altered community structure (H), and habitat destruction or conversion (H).

3) Nutrient loads from agriculture (H): This has caused altered species composition/dominance (VH), altered water quality of surface water or aquifer by nutrients (VH), altered community structure (H), and habitat destruction or conversion (H).

4) Invasive animals (H): This has caused altered species composition/dominance (VH), and altered community structure (H).

5) Incompatible recreational activities (M): This has caused altered species composition/dominance (VH), altered water quality of surface water or aquifer by nutrients (VH), altered community structure (H), habitat destruction or conversion (H), and erosion/sedimentation (M).

6) Surface water withdrawal (M): This has caused altered hydrologic regime (H).

7) Groundwater withdrawal (M): This has caused altered community structure (H), habitat destruction or conversion (H), and altered hydrologic regime (H).

8) Conversion to recreation areas (L): This has caused altered species composition/dominance (VH), altered community structure (H), and habitat destruction or conversion (H).

9) Incompatible forestry practices (L): This has caused altered community structure (H), and habitat destruction or conversion (H).

10) Conversion to commercial and industrial development (L): This has caused habitat destruction or conversion (H).

Subtidal Unconsolidated Marine/Estuary Sediment Habitat

Statewide Threat Rank of Habitat: High

A) *Species*: Atlantic sturgeon and striped bass

B) *Sources of Threat*:

1) Dam operation/incompatible release of water (H): This has caused altered water quality of surface water or aquifer by contaminants (H), habitat disturbance (H), altered water quality by nutrients (M), altered water quality physical and chemical (M), and altered hydrologic regime (M).

2) Inadequate stormwater management (H): This has caused altered water quality of surface water or aquifer by contaminants (H), habitat disturbance (H), altered species composition (M), altered water quality by nutrients (M), altered water quality physical and chemical (M), and altered hydrologic regime (M).

3) Coastal development (H): This has caused altered water quality of surface water or aquifer by contaminants (H), habitat disturbance (H), habitat destruction (M), and altered hydrologic regime (M).

4) Chemicals and toxins (H): This has caused altered water quality of surface water or aquifer by contaminants (H), habitat disturbance (H), and altered species composition (M).

5) Incompatible industrial operations (H): This has caused altered water quality of surface water or aquifer by contaminants (H), habitat destruction (M), and altered hydrologic regime (M).

6) Channel modification/shipping lanes (M): This has caused habitat disturbance (H), habitat destruction (M), and altered hydrologic regime (M).

7) Fishing gear impacts (M): This has caused habitat disturbance (H), and habitat destruction (M).

8) Incompatible recreational activities (M): This has caused habitat disturbance (H).

9) Roads, bridges, and causeways (M): This has caused habitat disturbance (H).

10) Management of nature (beach nourishment, impoundments) (M): This has caused altered water quality physical and chemical (M).

11) Boating (L): This has caused habitat disturbance (H).

12) Nutrient loads (L): This has caused altered species composition (M).

13) Invasive animals (L): This has caused habitat disturbance (H).

14) Thermal pollution (L): This has caused habitat disturbance (H), and altered water quality physical and chemical (M).

15) Solid waste (L): This has caused habitat disturbance (H).

16) Surface water withdrawal (L): This has caused altered water quality physical and chemical (M).

Submerged Aquatic Vegetation Habitat

Statewide Threat Rank of Habitat: Very High

A) *Species*: American eel

B) *Sources of Threat*:

1) Coastal development (VH): This has caused altered water quality physical and chemical (VH), habitat destruction (VH), altered species composition (VH), sedimentation (VH), altered water quality by contaminants (H), altered water quality by nutrients (H), altered structure (H), erosion (H), altered hydrologic regime (H), and habitat fragmentation (M).

2) Harmful algal blooms (VH): This has caused altered water quality physical and chemical (VH), habitat destruction (VH), altered species composition (VH), altered water quality by nutrients (H), and altered primary productivity (H).

3) Inadequate stormwater management (VH): This has caused altered water quality physical and chemical (VH), habitat destruction (VH), altered species composition (VH), sedimentation (VH), altered water quality by contaminants (H), altered water quality by nutrients (H), erosion (H), and altered primary productivity (H).

4) Channel modification/shipping lanes (VH): This has caused altered water quality physical and chemical (VH), habitat destruction (VH), sedimentation (VH), altered structure (H), erosion (H), altered hydrologic regime (H), altered primary productivity (H), and habitat fragmentation (M).

5) Nutrient loads (all sources) (H): This has caused altered water quality physical and chemical (VH), habitat destruction (VH), altered species composition (VH), sedimentation (VH), altered water quality by contaminants (H), altered water quality by nutrients (H), altered structure (H), altered primary productivity (H), and habitat fragmentation (M).

6) Incompatible industrial operations (H): This has caused altered water quality physical and chemical (VH), habitat destruction (VH), altered species composition (VH), sedimentation (VH), altered water quality by contaminants (H), altered structure (H), erosion (H), altered primary productivity (H), and habitat fragmentation (M).

7) Dam operation/incompatible release of water (H): This has caused altered water quality physical and chemical (VH), habitat destruction (VH), altered species composition (VH), sedimentation (VH), altered water quality by contaminants (H), altered water quality by nutrients (H), erosion (H), altered hydrologic regime (H), and altered primary productivity (H).

8) <u>Climate variability</u> (H): habitat destruction (VH), altered species composition (VH), altered structure (H), erosion (H), altered hydrologic regime (H), and altered primary productivity (H).

9) <u>Surface water withdrawal</u> (H): This has caused altered water quality physical and chemical (VH), habitat destruction (VH), altered species composition (VH), altered hydrologic regime (H), and altered primary productivity (H).

10) <u>Invasive plants</u> (H): This has caused habitat destruction (VH), altered species composition (VH), altered water quality by nutrients (H), altered structure (H), and altered primary productivity (H).

11) <u>Groundwater withdrawal</u> (H): This has caused altered water quality physical and chemical (VH), habitat destruction (VH), altered species composition (VH), altered hydrologic regime (H), and altered primary productivity (H).

12) <u>Roads, bridges, and causeways</u> (H): This has caused altered water quality physical and chemical (VH), habitat destruction (VH), altered species composition (VH), sedimentation (VH), altered water quality by contaminants (H), altered water quality by nutrients (H), altered structure (H), erosion (H), altered hydrologic regime (H), altered primary productivity (H), and habitat fragmentation (M).

13) <u>Shoreline hardening</u> (H): This has caused altered water quality physical and chemical (VH), habitat destruction (VH), altered species composition (VH), sedimentation (VH), altered water quality by contaminants (H), altered water quality by nutrients (H), erosion (H), and altered primary productivity (H).

14) <u>Invasive animals</u> (H): This has caused habitat destruction (VH), and altered species composition (VH).

15) <u>Destruction of longshore transport of sediments</u> (H): This has caused altered water quality physical and chemical (VH), altered species composition (VH), sedimentation (VH), altered water quality by nutrients (H), erosion (H), and altered primary productivity (H).

16) <u>Management of nature (beach nourishment, impoundments)</u> (M): This has caused altered water quality physical and chemical (VH), habitat destruction (VH), altered species composition (VH), sedimentation (VH), erosion (H), altered hydrologic regime (H), altered primary productivity (H), and habitat fragmentation (M).

17) <u>Boating impacts</u> (M): This has caused altered water quality physical and chemical (VH), habitat destruction (VH), altered species composition (VH), sedimentation (VH), altered water quality by contaminants (H), altered water quality by nutrients (H), altered structure (H), erosion (H), altered primary productivity (H), and habitat fragmentation (M).

18) <u>Chemicals and toxins</u> (M): This has caused altered water quality physical and chemical (VH), habitat destruction (VH), altered species composition (VH), and altered primary productivity (H).

19) Incompatible recreational activities (M): This has caused altered water quality physical and chemical (VH), habitat destruction (VH), altered species composition (VH), sedimentation (VH), altered water quality by contaminants (H), altered water quality by nutrients (H), altered structure (H), and erosion (H).

20) Key predator/herbivore losses (M): This has caused habitat destruction (VH), altered species composition (VH), and altered primary productivity (H).

21) Utility corridors (M): This has caused habitat destruction (VH), altered structure (H), and habitat fragmentation (M).

22) Fishing gear impacts (M): This has caused habitat destruction (VH), altered species composition (VH), altered structure (H)

23) Industrial spills (M): This has caused altered water quality physical and chemical (VH), habitat destruction (VH), altered species composition (VH), altered water quality by contaminants (H), and altered primary productivity (H).

24) Incompatible aquaculture operations (M): This has caused altered water quality physical and chemical (VH), habitat destruction (VH), altered species composition (VH), sedimentation (VH), altered water quality by nutrients (H), altered structure (H), erosion (H), altered primary productivity (H), and habitat fragmentation (M).

25) Vessel impacts (M): This has caused habitat destruction (VH), altered water quality by contaminants (H), and altered structure (H).

26) Placement of artificial structure (M): This has caused habitat destruction (VH), altered species composition (VH), sedimentation (VH), altered structure (H), and altered primary productivity (H).

27) Thermal pollution (M): This has caused habitat destruction (VH), and habitat fragmentation (M).

28) Solid waste (L): This has caused habitat destruction (VH), altered structure (H), and altered primary productivity (H).

Canal/Ditch Habitat

A) *Species*: American eel

B) *Sources of Threat*:

1) Conversion to housing and development (north region): This has caused habitat destruction/conversion (including loss of existing ditch or swale habitat to curb and gutter underground storm-sewer-type drainage systems associated with more intensive urban or suburban development) (applies only in north region), and loss of riparian cover along canals/ditches as a result of canal maintenance practices (applies to central and south regions).

2) <u>Intensification of surface water diversion/drainage associated with more intensive development</u> (north region): This has caused habitat destruction/conversion (including loss of existing ditch or swale habitat to curb and gutter underground storm-sewer-type drainage systems associated with more intensive urban or suburban development) (applies only in north region), and loss of riparian cover along canals/ditches as a result of canal maintenance practices (applies to central and south regions).

3) <u>Incompatible canal maintenance practices</u> (e.g., removing all canal bank vegetation through herbicide applications, etc.) (all regions): This has caused habitat destruction/conversion (including loss of existing ditch or swale habitat to curb and gutter underground storm-sewer-type drainage systems associated with more intensive urban or suburban development) (applies only in north region), and loss of riparian cover along canals/ditches as a result of canal maintenance practices (applies to central and south regions).

4) <u>Conversion to housing and development</u> (north region): This has caused an altered landscape mosaic (including destruction or conversion of wet flatwoods adjacent to roadside ditches) (north region).

5) <u>Nutrient loads</u> (all regions): This has caused altered water quality by contaminants.

6) <u>Chemicals/toxins</u> (e.g., oil/grease and heavy metals from roads) (north region): This has altered water quality from contaminants.

7) <u>Incompatible agricultural practices</u> (e.g., pesticides in runoff or drainage water) (all regions): This has altered water quality from contaminants.

8) <u>Incompatible residential practices</u> (e.g., pesticides in runoff) (all regions); <u>mosquito control</u> (north region): This has altered water quality from contaminants.

9) <u>Management of dams/control structures</u> (central/south regions):

10) <u>Incompatible agricultural practices</u> (e.g., management of runoff) (all regions): This has caused altered hydrologic regime (e.g., large pulses of flood water or storm runoff that disrupts life cycle requirements or alters or removes physical habitat).

11) <u>Incompatible residential practices</u> (e.g., management of runoff) (all regions): This has caused altered hydrologic regime (e.g., large pulses of flood water or storm runoff that disrupts life cycle requirements or alters or removes physical habitat).

Citation

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PART IV. THREATS LITERATURE CITED

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Chapter 11

DIADROMOUS FISH HABITAT CONSERVATION AND RESTORATION RECOMMENDATIONS AND/OR REQUIREMENTS

Group I. Recommendations for All Commission-Managed Diadromous Species

Dams and Other Obstructions

General Fish Passage

1) States should work in concert with the United States Fish and Wildlife Service (USFWS) and the National Oceanic and Atmospheric Administration Fisheries Service (NOAA Fisheries) to identify hydropower dams that pose significant impediment to diadromous fish migration, and target them for appropriate recommendations during Federal Energy Regulatory Commission (FERC) relicensing.

2) States should identify and prioritize barriers in need of fish passage based on clear ecological criteria (e.g., amount and quality of habitat upstream of barrier, size, status of affected populations, etc.). These prioritizations could apply to a single species, but are likely to be more useful when all diadromous species are evaluated together.

3) A focused, coordinated, well supported effort among federal, state, and associated interests should be undertaken to address the issue of fish passage development and efficiency. The effort should attempt to develop new technologies and approaches to improve passage efficiency with the premise that existing technology is insufficient to achieve restoration and management goals for several East Coast river systems.

4) Where obstruction removal is not feasible, install appropriate passage facilities, including fish lifts, fish locks, fishways, navigation locks, or notches (low-head dams and culverts).

5) At sites with passage facilities, evaluate the effectiveness of upstream and downstream passage; when passage is inadequate, facilities should be improved.

6) Dams/obstructions where upstream passage structures will be installed should be evaluated for effectiveness of downstream passage. Upstream passage structures should not be installed at these sites, unless downstream passage can be made safe, effective, and timely.

7) Facilities for monitoring the effectiveness of the pass should be incorporated into the design where possible.

8) Before designing and constructing fish passage systems, determine the behavioral response of each species of interest to major physical factors so that effectiveness can be maximized.

9) Protection from predation should be provided at the entrance, exit, and throughout the pass.

10) The passage facility should be designed to work under all conditions of head and tail water levels that prevail during periods of migration.

11) Passages are vulnerable to damage by high flows and waterborne debris. Techniques for preventing damage include robust construction, siting facilities where they are least exposed to adverse conditions, and removing the facilities in the winter.

Upstream Fish Passage

1) Diadromous fish must be able to enter the passage facility with little effort and without stress.

2) To prevent fish from becoming entrained in intake flow areas of hydropower facilities, construct behavioral barrier devices and re-direct them to safer passage areas.

3) Fish ascending the pass should be guided/routed to an appropriate area so that they can continue upstream migration, and avoid being swept back downstream below the obstruction.

Downstream Fish Passage

1) To enhance survival at dams during emigration, evaluate survival of fish passed via each route (e.g., turbines, spillage, bypass facilities, or a combination of the three) at any given facility, and pass fish via the route with the best survival rate.

Other Dam Issues

1) Where practicable, remove obstructions to upstream and downstream migration.

2) Locate facilities along the river where impingement rates are likely to be lowest.

3) Alter water intake velocities, if necessary, to reduce mortality to diadromous species.

4) To mitigate hydrological changes from dams, consider operational changes such as turbine venting, aerating reservoirs upstream of hydroelectric plants, aerating flows downstream, and adjusting in-stream flows.

5) Natural river discharge should be taken into account when alterations are being made to a river because it plays a role in the migration patterns of diadromous fish.

Water Quality and Contamination

1) Maintain water quality and suitable habitat for all life stages of diadromous species in all rivers with populations of diadromous species.

2) Non-point and point source pollution should be reduced in diadromous fish habitat areas.

3) Implement best management practices (BMPs) along rivers and streams, restore wetlands, and utilize stream buffers to control non-point source pollution.

4) Implement erosion control measures and BMPs in agricultural, suburban, and urban areas to reduce sediment input, toxic materials, and nutrients and organics into streams.

5) Upgrade wastewater treatment plants and remove biological and organic nutrients from wastewater.

6) Reduce the amount of thermal effluent into rivers. On larger rivers, include a thermal zone of passage.

7) Provide management options regarding water withdrawal and land use to minimize the impacts of climate change on temperature and flow regimes.

8) Discharge earlier in the year to reduce impacts to migrating fish.

9) Conduct studies to determine the effects of dredging on diadromous habitat and migration; appropriate best management practices, including environmental windows, should be considered whenever navigation dredging or dredged material disposal operations would occur in a given waterway occupied by diadromous species.

10) Introduction of new categories of contaminants should be prevented.

Habitat Protection and Restoration

1) When states have identified habitat protection or restoration as a need, state marine fisheries agencies should coordinate with other agencies to ensure that habitat restoration plans are developed, and funding is actively sought for plan implementation and monitoring.

2) Any project resulting in elimination of essential habitat (e.g., dredging, filling) should be avoided.

3) Substrate mapping of freshwater tidal portions of rivers should be performed to determine suitable diadromous fish habitat, and that habitat should be protected and restored as needed.

4) States should notify in writing the appropriate federal and state regulatory agencies of the locations of habitats used by diadromous species. Regulatory agencies should be advised of the types of threats to diadromous fish populations, and recommended measures that should be employed to avoid, minimize, or eliminate any threat to current habitat quantity or quality.

5) Each state encompassing diadromous fish spawning rivers and/or producer areas should develop water use and flow regime guidelines protective of diadromous spawning and nursery areas to ensure the long-term health and sustainability of the stocks.

Permitting

1) Develop policies for limiting development projects seasonally or spatially in spawning and nursery areas; define and codify minimum riparian buffers and other restrictions where necessary.

2) Projects involving water withdrawal (e.g., power plants, irrigation, water supply projects) should be scrutinized to ensure that adverse impacts resulting from impingement, entrainment, and/or modifications of flow and salinity regimes due to water removal will not adversely impact diadromous fish stocks.

3) State fishery regulatory agencies should develop protocols and schedules for providing input on Federal permits and licenses required by the Clean Water Act, Federal Power Act, and other appropriate vehicles, to ensure that diadromous fish habitats are protected.

Other

1) Promote cooperative interstate research monitoring and law enforcement. Establish criteria, standards, and procedures for plan implementation as well as determination of state compliance with management plan provisions.

2) Diadromous fish may be vulnerable to mortality in hydrokinetic power generation facilities, and such projects should be designed and monitored to eliminate, or minimize, fish mortality.

3) The use of any fishing gear that is deemed by management agencies to have an unacceptable impact on diadromous fish habitat should be prohibited within appropriate essential habitats (e.g., trawling in spawning areas or primary nursery areas should be prohibited).

Group II. Alosine-Specific Recommendations

Dams and Other Obstructions

Fish Passage

1) Passage facilities should be designed specifically for passing alosines for optimum efficiency at passing these species.

2) Conduct studies to determine whether passing migrating adults upstream earlier in the year in some rivers would increase production and larval survival, and opening downstream bypass facilities sooner would reduce mortality of early emigrants (both adult and early-hatched juveniles).

Other Dam Issues

1) Ensure that decisions on river flow allocation (e.g., irrigation, evaporative loss, out of basin water transport, hydroelectric operations) take into account flow needs for alosine migration, spawning, and nursery use, and minimize deviation from natural flow regimes.

2) Ensure that water withdrawal effects do not impact alosine stocks by impingement/entrainment, and employ intake screens or deterrent devices as needed to prevent egg and larval mortality.

3) When considering options for restoring alosine habitat, include study of, and possible adjustment to, dam-related altered river flows.

Habitat Protection and Restoration

1) States should identify and quantify potential shad and river herring spawning and nursery habitat not presently utilized, including a list of areas that would support such habitat if water quality and access were improved or created, and analyze the cost of recovery within those areas. States may wish to identify areas targeted for restoration as essential habitat.

2) Resource management agencies in each state shall evaluate their respective state water quality standards and criteria to ensure that those standards and criteria account for the special needs of alosines. Primary emphasis should be on locations where sensitive egg and larval stages are found.

3) ASMFC should designate important shad and river herring spawning and nursery habitat as Habitat Areas of Particular Concern (HAPCs).

Permitting

1) All state and federal agencies responsible for reviewing impact statement for projects that may alter anadromous alosine spawning and nursery areas shall ensure that those projects will have no impact or only minimal impact on those stocks. Of special concern are natal rivers of newly established stocks or stocks considered depressed or severely depressed.

Stock Restoration and Management

1) When populations have been extirpated from their habitat, coordinate alosine stocking programs, including:

   a. reintroduction to the historic spawning area

   b. expansion of existing stock restoration programs, and

   c. initiation of new strategies to enhance depressed stocks.

2) When releasing hatchery-reared larvae into river systems for purposes of restoring stocks, synchronize the release with periods of natural prey abundance to minimize mortality and maximize nutritional condition. Determine functional response of predators on larval shad at restoration sites to ascertain appropriate stocking level so that predation is accounted for, and juvenile out-migration goals are met. Also, determine if night stocking will reduce mortality.

3) Manage alewife and blueback herring separately given that management actions will affect them differently due to their life history differences (currently, these species are managed as a single stock and lumped together in commercial catch records; this hinders understanding of fishery impacts to populations of river herring species).

River-Specific Habitat Recommendations

River-specific habitat recommendations for American shad can be found in:
Atlantic States Marine Fisheries Commission. 2007. American shad stock assessment report for
    peer review, volumes II and III. Atlantic States Marine Fisheries Commission Stock
    Assessment Report No. 07-01 (Supplement), Washington, D.C.

<u>Group III. American Eel-Specific Recommendations</u>

Dams and Other Obstructions

Upstream Fish Passage

1) Passage facilities should be designed specifically for passing American eel for optimum efficiency at passing this species.

2) Eel-specific passage structures should be installed on rivers where eels will gain access to habitat. Those areas which will gain large amounts of habitat due to the installation of a fish passage should be given first priority over those rivers which will only open a small portion of habitat.

3) Passages should be constructed so eels can locate the appropriate starting point for ascent (lower entrance of the pass). This can be achieved by placing the entrance where the eels naturally congregate or by providing an attracting mechanism. If eels gather naturally in more than one place, multiple passes, or multiple entrances to a single pass should be designed.

4) Eel ladders should be constructed so that the outflow of the holding tank or resting flow is directed towards the ramp. This may make it easier for eels to find the ladder, as they are attracted to the scent of other eels.

5) A strong flow should be provided close to the entrance of the bypass system to attract American eel to the ladder.

6) Eel swimming ability is important when considering the design of a pass. The maximum swimming speed and the ability to maintain that speed, as well as the influence of temperature on swimming ability should be taken into account.

7) To overcome the head difference at the facility without expending too much effort, the volume and velocity of the water flow within a pass should be restricted. A substrate should also be provided which slows and disorganizes the flow, and allows the eels to ascend the pass by crawling as much as swimming.

8) Facilities at or near the tidal zone should be designed to primarily pass elvers (90-130mm); however, larger fish will pass through the tidal zone, therefore facilities should be designed to accommodate eels up to 300 mm length range.

9) The size range of American eel that require passage increases with distance upstream. Facilities higher in the watershed should be designed for a greater size range.

10) The size of the American eel should be taken into account when selecting substrates for fish ladders for upstream migration. Smaller elvers require different substrates than larger yellow eels.

11) Natural substrates should not be used in eel passes, as they tend to deteriorate quickly and require constant replacement. Furthermore, some natural materials will not accommodate the size range of all eels because they are very selective. This does not apply to natural emergent vegetation, which can represent an important aspect of passage based on easement.

12) The exit of the pass should be extended into quieter water where there is a rough or weedy bottom. This will help the eels to escape and will provide cover.

13) Where funding is limited, facilities should be designed to pass a limited size range of eels, instead of all size ranges. It is therefore important to research the size range and other conditions of eels migrating through a particular site.

14) Vandalism and theft of eels are potential problems. To overcome this, robust construction and locked covers can help, as well as building facilities where the general public does not have access.

15) Artificial light should not be placed near eel ladders to ensure that migrations are not disrupted, and covers should be placed on passages to protect eels from direct sunlight.

Downstream Fish Passage

1) Important design criteria for bypass facilities targeting silver eels include:

   a. Size of the migrating fish

   b. Seasonal and diurnal timing of migration

   c. Environmental conditions that stimulate migration, and

   d. Behavior of the fish.

2) Bypass structures should include deterrents from turbine entry to attract and/or facilitate the downstream passage of eels via bypass facilities. Deterrents can either be behavioral or mechanical.

3) Cost-effective mitigation measures should be considered, including trap and transport of American eel downstream of the dam.

4) Where possible, turbine operations should be suspended to provide spill flows during times of peak downstream migration.

5) To reduce mortality in American eel, generation of hydropower should be reduced or ceased wherever practicable. This can be achieved by using criteria based on a combination of rainfall events and eel run timing factors.

6) Physical screens to exclude fish from intakes should be used where the obstruction is small relative to the flow of the river. Any screen that is effective for excluding salmon smolts where gaps are 12.5 mm or less is also effective for excluding all silver

eels. Approach velocities (i.e., calculated velocity component perpendicular to the screen face) used for salmonids should allow silver eels to avoid impingement on the screen.

Other Dam Issues

1) Where obstruction removal is not feasible, install passage facilities for American eel (e.g., wetted surfaces, ramps, bucket lifts, etc.) that provide optimum efficiency.

Water Quality and Contamination

1) Steps should be taken to eliminate or limit the contamination of American eel habitat from compounds that are known to accumulate in American eel.

Group IV. Atlantic Sturgeon-Specific Recommendations

Dams and Other Obstructions

Fish Passage

1) Passage facilities should be designed specifically for passing Atlantic sturgeon for optimum efficiency at passing this species.

2) Fish passage facilities should be designed to aid in the upstream and downstream passage of Atlantic sturgeon. Most fish ladders in Atlantic coast streams and rivers are designed to pass alosines, and the specific needs of sturgeon will need to be considered as passage facilities are improved or constructed.

3) The removal of dams, or the consideration of passage efforts, should be focused on those systems where Atlantic sturgeon historical habitat loss through blockage is greatest.

Habitat Protection and Restoration

1) Protection or restoration of critical habitat is considered the most beneficial conservation method for the restoration of sturgeons. Restore degraded historical habitat wherever possible. Also, habitat improvements that increase the survival of young-of-the-year are likely to make a strong contribution to population growth.

2) Water flows should be restored to appropriate levels during spawning season.

3) New spawning habitat should be created with the use of artificial reef materials in areas where hard substrate has been degraded.

4) ASMFC should designate important habitats for Atlantic sturgeon spawning and nursery areas as HAPCs.

Group V. Striped Bass-Specific Recommendations

Dams and Other Obstructions

Fish Passage

1) Passage facilities should be designed specifically for passing striped bass for optimum efficiency at passing this species.

2) Conduct studies to determine whether passing migrating adults upstream earlier in the year in some rivers would increase striped bass production and larval survival, and opening downstream bypass facilities sooner would reduce mortality of early emigrants (both adult and early-hatched juveniles).

Water Quality and Contamination

1) Federal and state fishery management agencies should take steps to limit the introduction of compounds which are known to be accumulated in striped bass tissues and which pose a threat to human health or striped bass health.

2) Every effort should be made to eliminate existing contaminants from striped bass habitats where a documented adverse impact occurs.

3) Water quality criteria for striped bass spawning and nursery areas should be established, or existing criteria should be upgraded to levels that are sufficient to ensure successful striped bass reproduction.

Habitat Protection and Restoration

1) Each state should implement protection for the striped bass habitat within its jurisdiction to ensure the sustainability of that portion of the migratory stock. Such a program should include:

    a. Inventory of historical habitats

    b. Identification of habitats presently used

    c. Specification of areas targeted for restoration, and

    d. Imposition or encouragement of measures to retain or increase the quantity and quality of striped bass essential habitats.

2) States in which striped bass spawning occurs should make every effort to declare striped bass spawning and nursery areas to be in need of special protection; such declaration should be accompanied by requirements of non-degradation of habitat quality, including minimization of non-point source runoff, prevention of significant increases in contaminant loadings, and prevention of the introduction of any new categories of contaminants into the area. For those agencies without water quality regulatory authority, protocols and schedules for providing input on water quality

regulations to the responsible agency should be identified or created, to ensure that water quality needs of striped bass stocks are met.

3) Each state should survey existing literature and data to determine the historical extent of striped bass occurrence and use within its jurisdiction. An assessment should be conducted of those areas not presently used for which restoration is feasible.

4) ASMFC should designate important habitats for striped bass spawning and nursery areas as HAPCs.

Permitting

1) All state and federal agencies responsible for reviewing impact statements and permit applications for projects or facilities proposed for striped bass spawning and nursery areas shall ensure that those projects will have no or only minimal impact on local stocks, especially natal rivers of stocks considered depressed or undergoing restoration.

Chapter 12

FUTURE HABITAT RESEARCH INFORMATION NEEDS FOR DIADROMOUS SPECIES

Group I. Research Needs for All Commission-Managed Diadromous Species

Dams and Other Obstructions

Fish Passage

1) Evaluate performance of conventional fishways, fish lifts, and eel ladders, and determine features common to effective passage structures and those common to ineffective passage structures.

2) Conduct basic research into diadromous fish migratory behavior as it relates to depth, current velocity, turbulence, entrained air, light, structures, and other relevant factors.

3) Use information from (1) and (2) to conduct computer fluid dynamics (CFD) modeling to develop more effective fishway designs.

4) Research technologies (barriers, guidance systems, etc.) for directing emigrating fish to preferred passage routes at dams.

5) Identify low-cost alternatives to traditional fishway designs.

6) Develop effective downstream passage strategies to reduce mortality.

Other Dam Issues

1) Document the impact of power plants and other water intakes on larval, postlarval, and juvenile mortality in anadromous fish spawning areas, and calculate the resultant impacts to adult population sizes.

2) Evaluate the upstream and downstream impacts of barriers on diadromous species, including population and distribution effects.

Water Quality and Contamination

1) Determine effects of change in temperature and pH for all life stages of all diadromous species. Use this information to model impacts of climate change on species.

2) Develop studies to document which contaminants have an impact on the various life stages of each diadromous species; also note the life stages that are affected, and at what concentrations.

3) Determine unknown optima and tolerance ranges for depth, temperature, salinity, dissolved oxygen, pH, substrate, current velocity, and suspended solids.

Habitat Protection and Restoration

1) Use multi-scale approaches (including GIS) to assess indicators of suitable habitat, using watershed and stream-reach metrics if possible (it should be noted, that where site-specific data is lacking, it may not be appropriate to assess at this scale).

2) Use multi-scale approaches for restoring diadromous fish habitat, including vegetated buffer zones along streams and wetlands, and implementing measures to enhance acid-neutralizing capacity.

3) Conduct studies on the effects of land use change on diadromous species population size, density, distribution, health, and sustainability.

4) Examine how deviation from the natural flow regime impacts all diadromous species. This work should focus on key parameters such as rate of change (increase and decrease), seasonal peak flow, and seasonal base flow, so that the results can be more easily integrated into a year-round flow management recommendation by state officials.

5) Investigate consequences to diadromous stocks from wetland alterations.

Other

1) Determine survival and mortality rates for all life stages of all diadromous species.

2) Investigate predator-prey relationships for all life stages of all diadromous species.

3) Determine the effects of channel dredging, shoreline filling, and overboard spoil disposal in the Atlantic coast on diadromous species.

4) Define restrictions necessary for implementation of energy projects in diadromous species habitat areas, and develop policies on limiting development projects seasonally or spatially.

Group II. Alosine-Specific Research Needs

Water Quality and Contamination

1) Review studies dealing with the effects of acid deposition on anadromous alosines.

2) Determine if intermittent episodes of pH depressions and aluminum elevations (caused by acid rain) affect any life stage in freshwater that might lead to reduced reproductive success of alosines, especially in poorly buffered river systems.

3) Determine if chlorinated sewage effluents are slowing the recovery of depressed shad stocks.

Habitat Protection and Restoration

1) Conduct research on habitat requirements for all life stages of hickory shad.

Migration

1) Determine factors that regulate and potentially limit downstream migration, seawater tolerance, and early ocean survival of juvenile alosines.

2) Conduct research on hickory shad migratory behavior.

Other

1) Focus research on within-species variation in genetic, reproductive, morphological, and ecological characteristics, given the wide geographic range and variation at the intraspecific level that occurs in alosines.

2) Research predation rates and impacts on alosines.

3) Evaluate the effect of bycatch on alosines.

4) Ascertain how abundance and distribution of potential prey affect growth and mortality of early life stages of alosines.

Group III. American Eel-Specific Research Needs

Dams and Other Obstructions

Fish Passage

1) Research the behavior of American eel approaching hydropower dams to determine searching behavior and preferred routes of approach to confirm best siting options for upstream passage.

2) Investigate, develop, and improve technologies for American eel passage upstream and downstream at various barriers for each life stage.

3) Investigate how river flow, lunar phase, water temperature, and behavior near artificial lighting impact the behavior of American eel, and influence the amount of time that the eels spend at a dam.

4) Research the behavior of silver eels at downstream passages; determine specific behavior of eels migrating downstream, and research how they negotiate and pass hydropower facilities.

Water Quality and Contamination

1) Determine the effects of contaminant bioaccumulation on American eel, including impacts on survival and growth (by age), maturation, and reproductive success.

2) Research the ability of contaminated eels to carry out successful breeding.

3) Examine the environmental conditions required for the hatching success of American eel.

Habitat Protection and Restoration

1) Establish characteristics and distribution of American eel habitat (using conventional methods as well as GIS), and the value of that habitat with respect to growth and sex determination.

2) Determine the effects of loss of historic habitat to potential American eel population and reproductive capacity.

3) Investigate the impact of seaweed harvesting on American eel.

4) Research the changes in ocean climate and environmental quality that might influence larval and adult eel migration, spawning, recruitment, and survival, including oceanic heat transport and interactions with the atmosphere and greenhouse gas warming.

5) Determine the importance of coastal lakes and reservoirs to American eel populations.

Migration

Silver-phase

1) Identify migratory routes and guidance mechanisms of silver eels migrating to the ocean.

2) Determine mechanisms for the recognition of the spawning area by silver eels, mate location in the Sargasso Sea, spawning behavior, and gonadal development in maturation.

3) Identify verify specific American eel spawning locations in the Sargasso Sea.

4) Research the factors that cause American eel to initiate downstream migration and affect their patterns of movement.

Leptocephalus

1) Identify the precise mechanisms of larval transport for American eel.

2) Examine the mechanisms for leptocephalus exit from the Sargasso Sea and transport across the continental shelf.

3) Determine mechanisms of recruitment of leptocephali and glass eels to coastal areas.

Glass Eel

1) Investigate the impact of stream velocity/discharge and stream morphology on upstream migration of glass eel and elvers.

Yellow-phase

1) Research behaviors and movements of American eel during their freshwater residency.

Parasitism

1) Evaluate the occurrence and impact of the nematode parasite, *Anguillicola crassus*, on all life stages.

Feeding

1) Examine the mode of nutrition for leptocephali in the ocean.

2) Examine food habits for glass eels at sea.

Other

1) Research all aspects of the leptocephalus life history stage.

Group IV. Atlantic Sturgeon-Specific Research Needs

Dams and Other Obstructions

Fish Passage

1) Fish passage requirements and appropriate structures for Atlantic sturgeon are largely unknown. Research all fish passage requirements for Atlantic sturgeon.

Bycatch

1) Determine levels of bycatch and compare to F_{50} target levels for individual Atlantic sturgeon populations.

2) Characterize Atlantic sturgeon bycatch in various fisheries by gear and season; include data on fish size, health condition at capture, and number of fish captured.

3) Develop markers that permit identification of bycatch of Atlantic sturgeon by population origin.

Population Status

1) Conduct assessments of population abundance and age structure in various river systems, with particular emphasis on documenting occurrence of age 0-12 juveniles and spawning adult Atlantic sturgeon as indicators of natural reproduction.

2) Continue to determine the extent to which Atlantic sturgeon are genetically differentiable among rivers, and interpret biological significance of findings.

3) Conduct further analyses to assess the sensitivity of F_{50} to model inputs for northern and southern stocks of Atlantic sturgeon.

Culture and Stock Enhancement

1) Further develop techniques for capture, transport, and long-term holding of wild Atlantic sturgeon brood stock.

2) Refine maturation-induced spawning procedures, and sperm cryo-preservation techniques for Atlantic sturgeon to assure availability of male gametes.

3) Continue basic cultural experiments at all life stages of Atlantic sturgeon to provide information on:

    a. Efficacy of alternative spawning techniques

    b. Egg incubation and fry production techniques

    c. Holding and rearing densities

    d. Prophylactic treatments

    e. Nutritional requirements and feeding techniques, and

    f. Optimal environmental rearing conditions and systems.

4) Identify suitable stocking protocols for hatchery-reared Atlantic sturgeon (e.g., individual size, time of year, site, and marking technique).

5) Conduct and monitor pilot-scale Atlantic sturgeon stocking programs before conducting large-scale efforts that encompass a broad geographic area.

6) Establish Atlantic sturgeon stocking goals and success criteria prior to development of large-scale stock enhancement or recovery programs.

Tagging and Tissues

1) Standardize collection procedures, and develop a suitable long-term repository for Atlantic sturgeon biological tissues for use in genetic and other studies.

2) Establish coordinated tagging programs to delineate migratory patterns and stock composition, giving priority to marking juveniles in important sturgeon rivers before they migrate to the ocean.

3) Maintain database for tagged Atlantic sturgeon.

4) Identify rates of tag loss and tag reporting for Atlantic sturgeon.

5) Analyze existing sea sampling data to characterize at-sea migratory behavior. Use electronic tagging to model coastal migrations of juvenile and adult Atlantic sturgeon.

Maturity and Aging

1) Develop methods to determine sex and maturity of captured Atlantic sturgeon.

2) Evaluate aging techniques for Atlantic sturgeon with known-age fish, with emphasis on verifying current methodology based on fin rays.

3) Determine length, fecundity, and maturity at age for all Atlantic sturgeon stocks.

4) Develop a protocol for ageing validation in Atlantic sturgeon.