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Project Summary

Development of Techniques and Methodology for the Laboratory Culture of Striped Bass, *Morone saxatilis* (Walbaum)

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This summary describes the research undertaken to develop laboratory culture techniques for striped bass *(Morone saxatilis)* that could be used to provide an adequate supply of various life stages of this important fish species for water quality and hazard evaluation testing.

For each of the four life stages defined here (egg, larval, juvenile, and adult) the upper and lower lethal levels where applicable and an approximation of optimum conditions were defined with regard to physical characteristics of the environment including temperature, salinity, dissolved oxygen, light, and turbidity. Satisfactory laboratory diets were defined and verified for each life stage. A comprehensive set of procedures was developed and described in a step-by-step manner for use by research personnel wishing to maintain laboratory populations of striped bass for physiological and toxicological use.

This Project Summary was developed by EPA's Environmental Research Laboratory, Narragansett, RI, to announce key findings of the research project that is fully documented in a separate report of the same title (see Project Report ordering information at back).

Introduction

Striped bass, *Morone saxatilis,* is an important commercial and sport fish species with a center of distribution between the Hudson River and the mouth of Chesapeake Bay. Individuals of this species ascend major rivers to spawn, use coastal estuaries as nursery grounds, and as adults make seasonal migrations along the coast rarely straying more than five miles from the shoreline. Because it passes its entire life cycle in the waters immediately adjacent to the Boston-Washington, D.C., megalopolis, it is subjected to the most intense effects of man-made pollution and environmental alteration. In spite of these abuses, the Atlantic population of striped bass has until recently enjoyed great abundance. Although in the past a considerable amount of research has been done on the culture of the species for stocking into southern reservoirs, no reliable culture methodology has been developed for maintaining all of the life stages of the striped bass in the laboratory where the effects of various pollutants may be determined in physiological studies and bioassay experiments.

This study was undertaken to develop a reliable culture protocol for all life stages of the striped bass. Armed with such a protocol, researchers will be in a better position to examine the effects of water borne pollutants on this resilient but vulnerable species.

Discussion

The striped bass is a desirable candidate for toxicological investigations in the United States for the following reasons. The species inhabits a wide range and is distributed along all three coasts. It is a commercially and recreationally important species throughout its range. It is also an ecologically important member of the community it inhabits, not only along the coasts, but also within the coastal plain rivers. Its life stages are euryhaline and eurythermal, making them extremely useful in studies to determine sublethal differences in the physiology of toxicants over broad salinity and/or temperature ranges. In addition to being easy to culture, a great deal of the background research on this species has been reported.

To date, striped bass culture has been undertaken primarily by federal or state fish hatcheries to stock lakes, reservoirs and impoundments for sport fishing and shad (*Dorosoma* sp.) control. This work is done almost exclusively in fresh water, either in a hatchery or in ponds (Bonn et al., 1976). The culture methodology recommended in this report for the life history stages of the striped bass, however, utilized sea water wherever possible. This was the case not only because of its availability and cost-effectiveness of use, but primarily to keep disease problems to a minimum. Although some fresh water (to reduce salinity) is needed during spawning and early larval stages, juveniles and adults feed and grow in sea water.

Using the methods recommended in this research and summarized below, striped bass life history stages can be cultured which are representative of the species. Figure 1 and Table 1 describe developmental stages of striped bass larvae to metamorphosis.

Culture Methods Outlined

The outline of recommended methods to secure batches of larvae or juveniles for toxicological studies that follows is based on the details presented in Sections 8-11 of the full report available from NTIS. In general, if 20,000 prolarvae are required for studies, then a minimum of 40,000 eggs are needed. This estimate is based on 50% survival, although egg survival varies from 10-20% for artificially spawned to 60-90% for naturally spawned (see Table 2) and fertilized eggs. This survival rate can be increased to 40-50% by using antibiotics in the rearing water. If 2,000 post-larvae are required, then 2,500 prolarvae (80% survival through initial feeding), or 5,000 eggs are required. These estimates are provided as a guide and may vary with broodstock, investigator, facilities and other variables.

A. Fertilized eggs can be obtained from natural or artificial spawnings.
   1) Artificially spawned eggs require maintenance of broodfish (mature adults) in culture system equipped with temperature, sa-

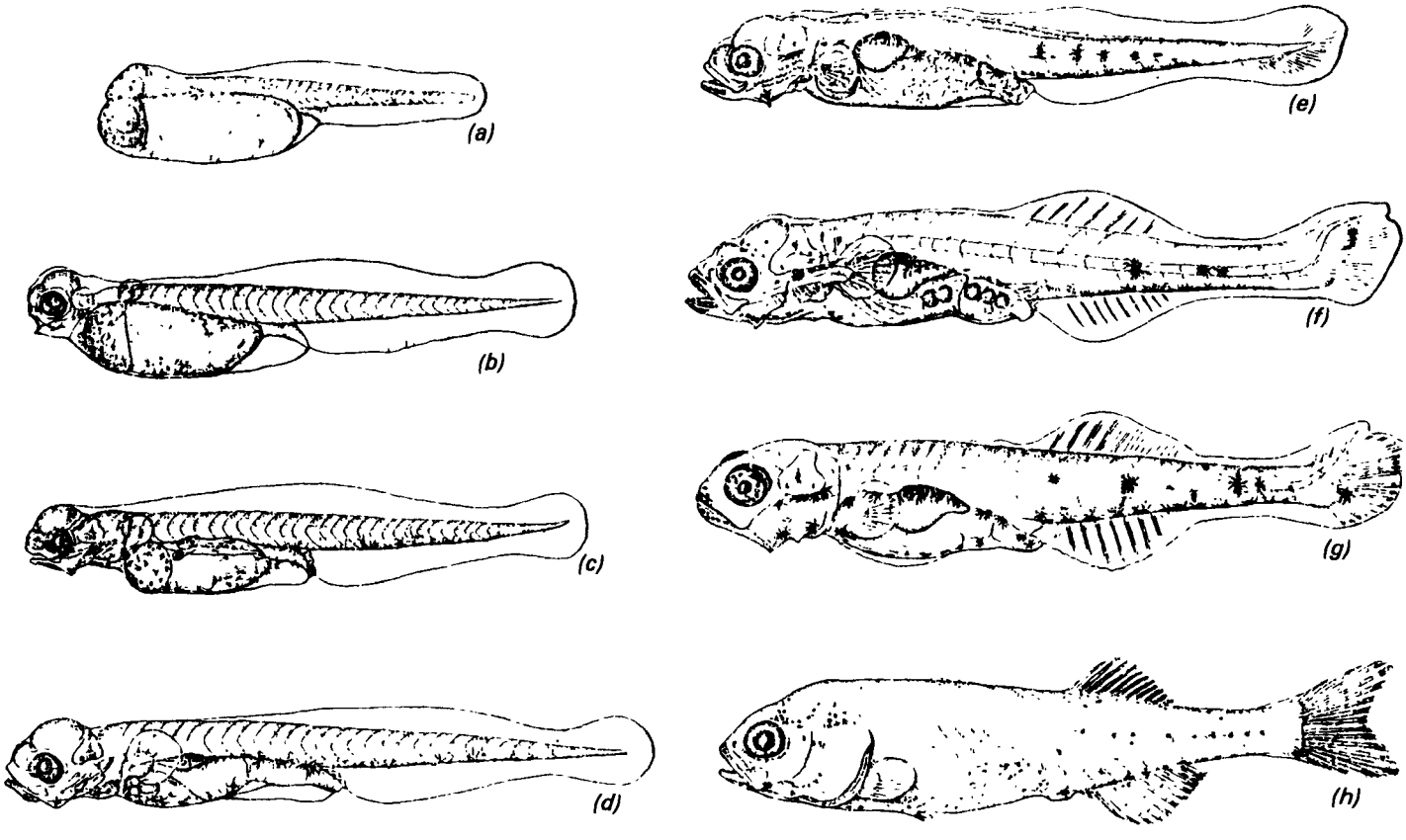

Figure 1. *Developmental stages (after Manuseti, 1958) of striped bass larvae to metamorphosis. Refer to Table 1 for further description of stages.*

Table 1. *Developmental Stages of Striped Bass, Reared at About 17°C, Unless Otherwise Stated, Through Transformation*

Age	Length mm TL[a]	Characteristics
25.8 hours after fertilization (4)[b]	3.25-4.06	Hatching completed for eggs at 24°C. (a)[c]
36-48 hours after fertilization (2)	2.5-3.7	Hatching occurs. (a)
51.8 hours after fertilization (4)	3.25-4.71	Hatching completed for eggs at 18°C. (a)
1st day after hatching (4)	3.58-5.12	Eyes almost fully pigmented; pigmented ventrally; one-third yolk reabsorbed at 24°C.
	4.23-5.20	Eyes only partially pigmented; yolk slightly reabsorbed at 18°C.
2-5th day after hatching (1,2)	4.5-5.2	Yolk sac partly absorbed, eyes pigmented yellow, black & orange, differentiation of jaws and digestive tract begun, pectoral buds formed fan-like fin, 21-23 myotomes. (b)
3rd day after hatching (3)	5.2	Eyes pigmented, jaws developing, pectoral fins become differentiated.
(4)	4.71-6.23	Eyes pigmented; mouth parts moving; pigmented ventrally jaw to oil; yolk three-fourths reabsorbed; pectoral buds present at 24°C.
	5.04-5.77	Eyes pigmented; gut differentiated; ventrally pigmented; pectoral buds visible at 18°C.
4th day after hatching (3)	5.8	Small chromatophores along ventral edge of entire yolk sac.
(4)	5.5-7.5 (live)	Yolk absorbed at 24°C.
5th day after hatching (1)	5.5-5.8	One-third yolk reabsorbed, commencement of intestinal peristalsis, 23-24 myotomes. Swimming pelagically. (c)
6th day after hatching (3)	6.0	Oil globule and yolk nearly absorbed, pigmentation ventrally. (c)
6th-7th day after hatching (4)	5.5-7.5 (live)	Yolk absorbed at 18°C. (d)
8th day after hatching (1)	5.8-6.5	Teeth on jaws, orange pigment in caudal (heteroceral) area, differentiation of stomach, three-fourths yolk reabsorbed, 25 myotomes. Transition to active pelagic feeding. (d)
(3)	6-9	Second dorsal and anal slightly differentiated, well-developed mouth parts. (d)
10-15th day after hatching (2)	7.5	Yolk sac fully absorbed and no oil globule visible, pectorals only fins visible, teeth visible, generally pigmented on body. (e)
10th day after hatching (3)	9.0	Pectorals only fins developed, ready for food.
15th day after hatching (1)	10-12.5	Division of fin fold into three divisions, complete reabsorption of oil globule, single-chamber gas bladder filled with air. Feeding on plankton. (e)
18th day after hatching (3)	13.0	Dorsal and anal fin rays well differentiated and rudimentary spines observed. (f)
20-30th day after hatching (1,2)	10, 12-16	Differentiation of rays in caudal, anal and dorsal fins. First dorsal elements and pelvic fins absent, myotomes correlated with number of vertebrae. (g)
30 days after hatching (4)	13.1-15.4	Metamorphosis at 24°C.
30-40th day after hatching (2)	15 (stunted)	Soft dorsal, anal and caudal (homocercal) fins well differentiated, spinous and pelvic fins not well developed and well ossified, no stripes visible yet. Initial formation of lateral-line scales (Murawski, 1958). (h)
40 days after hatching (4)	11.9-20.4	Metamorphosis at 18°C.
40-50th day after hatching (1)	22-35	Differentiation of rays in first dorsal and pectoral fins. Full complement of lateral-line scales by 30 mm (Murawski, 1958).
50-70th day after hatching (1)	35-45	Scales
(2)	20	Scales observed for first time, fins except larval pelvic in various stages toward full meristic count, pigmentation stronger.
60-80th day after hatching (2)	25	Covered with scales, 3 anal spines and full complement of meristic characters, body covered with melanopores.

Table 1. *(continued)*

Age	Length mm TL[a]	Characteristics
80-90th day after hatching (1)	50-80	Appearance of longitudinal stripes.
70-100th day after hatching (2)	30	Meristic counts complete except for fin rays, body pigmentated.
3-4 weeks after hatching (3)	36	Fully developed fins and rays, pigmentation of black dots.

[a] *Total length measured on preserved samples unless otherwise stated.*
[b] *Numbers in parenthesis refer to source, i.e., (1) Doroshev (1970); (2) Mansueti (1958); (3) Pearson (1938); and (4) Rogers* et al. *(1977).*
[c] *Letters in parenthesis refer to Figure 11.*

Table 2. *Percent Survival Through Hatching of Striped Bass Eggs from Artificial and Natural Spawnings*

Incubation Salinity* (‰)	Incubation Temperature (°C) 16	18	20	21
Artificially induced spawning				
0	58.5 (561)	64.3 (280)	—	4.7 (536)
5	1.2 (249)	—	7.4 (244)	5.4 (185)
10	19.3 (165)	—	11.6 (215)	11.6 (205)
15	31.2 (160)	—	0	0
Natural matured spawning				
0	77.9 (384)	71.0 (473)	—	71.5 (421)
5	90 (10)	—	90 (10)	—
10	90 (10)	—	90 (10)	—
15	90 (10)	—	80 (10)	—

* *Percent survival at 0‰ and 16°C (60°F), 18°C (65°F), and 21°C (70°F) reported by Shannon (1970). Survivals at the other salinity-temperature combinations are results of this study.*
+( ) = *number of eggs per treatment.*

linity, and photoperiod control with subsequent controlled spawning.

2) Naturally spawned eggs may be obtained easily by plankton or neuston net fishing in spawning rivers at the time of spawning (February-May).
3) Collection of naturally spawned eggs insures genetic diversity not available among progeny of a mating under controlled spawning.

B. Handling of fertilized eggs to maximize survival and hatching.

1) Eggs collected from plankton tows must be separated before transporting them to rearing containers.
2) Eggs secured from artificial spawnings can be stocked directly into rearing containers at a rate of approximately 100 per liter.
3) Handle eggs only in water, i.e., dip or pipette or siphon. Do not use dip nets.
4) When transferring shipped eggs to rearing containers check that water temperature of two are within 1 °C of each other and that rearing water quality is optimum for egg survival (Table 3).
5) Water quality, especially temperature, dissolved oxygen and salinity, should be monitored daily and maintained at optimum levels (Table 3).

C. Handling of larvae to maximize survival and growth.

1) Easiest method of securing prolarvae is from eggs on hand that hatch.
2) The recommended larval rearing system is static prepared tank system modified from Houde & Ramsey (1971) and described in greater detail in the full report available from NTIS.
3) Larvae can be stocked at 100 per liter until actively feeding, when densities should be reduced to approximately 50 per liter.
4) Growing larvae should be graded to nearly equal size fish to reduce cannibalism.
5) The water quality in larval rearing containers should be monitored daily and maintained at optimum level (Table 3).
6) Larvae should be fed 10-20% of their dry body weight at least twice daily beginning about 4-5 days after hatching an approved strain of newly-hatched brine shrimp nauplii. Table 4 lists biochemical characteristics of some life diets for larval bass.
7) Growth rates at various temperatures are detailed in Figure 2.
8) As larvae reach metamorphosis other foods such as ground squid or prepared diets (moist pellets) can be added to adult brine shrimp.

Table 3. *Summary of Optimal Rearing Conditions for the Various Striped Bass Life Stages*

	Eggs	Prolarva	Larvae	Postlarvae	Juveniles & Subadults	Adults
ABIOTIC FACTORS						
Temperature	16-20°C	16-21°C		18-22°C	>10 and <25°C	>10 and <24°C
Salinity	2-10‰	5-15‰		10-20‰	10-30‰	10-30‰
Dissolved Oxygen	air saturated		air saturated		air saturated	air saturated
Light	natural photoperiod		natural photoperiod		natural photoperiod	natural photoperiod
Turbidity	500 mg/l[a]		≤100 mg/l[a]		<4 mg/l[b]	—
BIOTIC FACTORS						
Diet	not applicable	not applicable		15-20% body weight (dry) twice daily	5-8% body weight (wet) per day	3-5% body weight (wet) daily
Density	50-75 per liter	50-25 per liter		30-10 per liter	10-2 bass per 100 liters	2.4 g/l maximum

[a] *Fine grained sediment.*
[b] *Bentonite.*

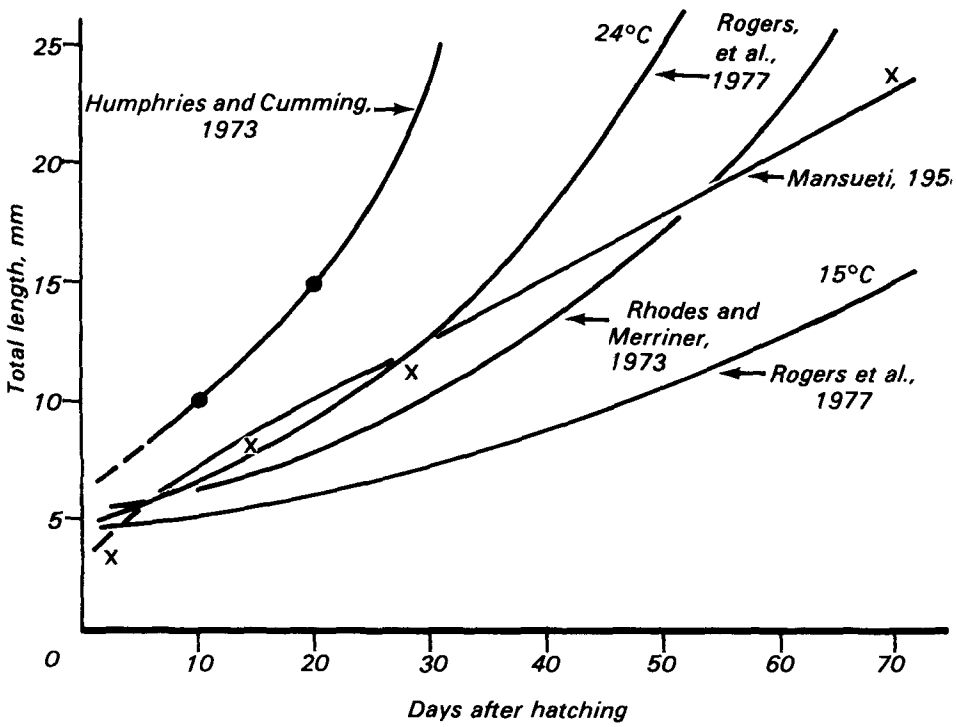

Figure 2. *A comparison of growth rates observed under fixed temperature regimes (Rogers et al., 1977) with those obtained in earlier studies under conditions of increasing temperature.*

D. Handling juveniles.

1) Juveniles, if needed for research, can be reared from eggs or larvae, or collected by seining in spawning rivers.
2) Juveniles collected from the field should be kept separate from any reared or other collected bass already in the culture system.
3) Water quality should be monitored daily and maintained at optimum conditions (Table 3).
4) Juveniles can be fed frozen brine shrimp, ground squid, prepared diets, or commercial trout feeds; the first is generally preferred.

Conclusions

During the course of this study all of the life stages of the striped bass from egg to adult were successfully maintained under laboratory conditions. The temperature, salinity, dissolved oxygen, light and turbidity requirements of all life stages were either determined empirically, approximated from environmental data, or where reported by other workers corroborated in our laboratory. Optimum and survival limits for each of these parameters were, where appropriate, specified. By maintaining conditions within these bounds, striped bass eggs were repeatedly reared through to the juvenile stage. A population of striped bass adults was successfully maintained in captivity for five years. Despite repeated attempts, we were unable during the course of this study to successfully induce spawning in the laboratory. Sexually mature adults of both sexes, however, did occur among our captive population. A step-by-step culture methodology has been prepared for use by future workers.

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Table 4. *Caloric and Percent Composition of Some Live Larval Food Items*

Food Item	*Calories/gram (ash-free, dry)*	*Percent of Dry Weight Lipid*	*Protein*
Artemia salina			
nauplii	*5800-6000(1)* 5454-5953(3)*	*15.04-27.24*	*42.5-50.2(1)*
adults	*5115-5854(3)*	*6.51*	*62.78(1)*
Acartia clausi		*5.8*	*82.6(4)*
Acartia tonsa	*5664 ± 86(2)*		
Calanus finmarchicus	*6835 ± 191(2)*	*10.5-47.0*	*30-77(4)*
Calanus helgolandicus	*5515 ± 277(5)*	*11.5*	*75.2(4)*

**Numbers in parentheses refer to source: (1) Helfrich* et al. *(1973); (2) Laurence (1977); (3) Paffenhofer (1967); (4) Raymont* et al. *(1963); and (5) Slobodkin and Richman (1961).*

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The complete report, entitled "Development of Techniques and Methodology for the Laboratory Culture of Striped Bass, Morone saxatilis*," (Order No. PB 82-217 795; Cost: $22.50, subject to change) will be available only from:*
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