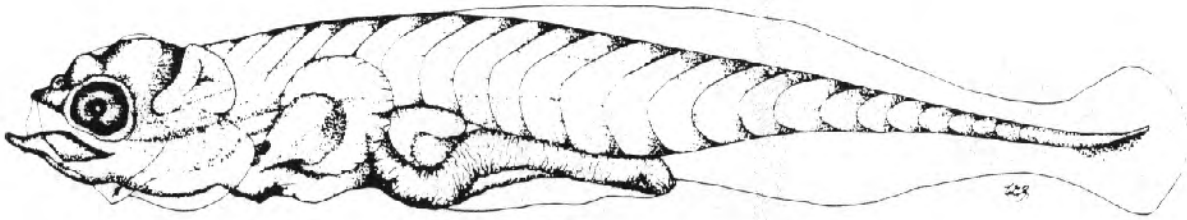
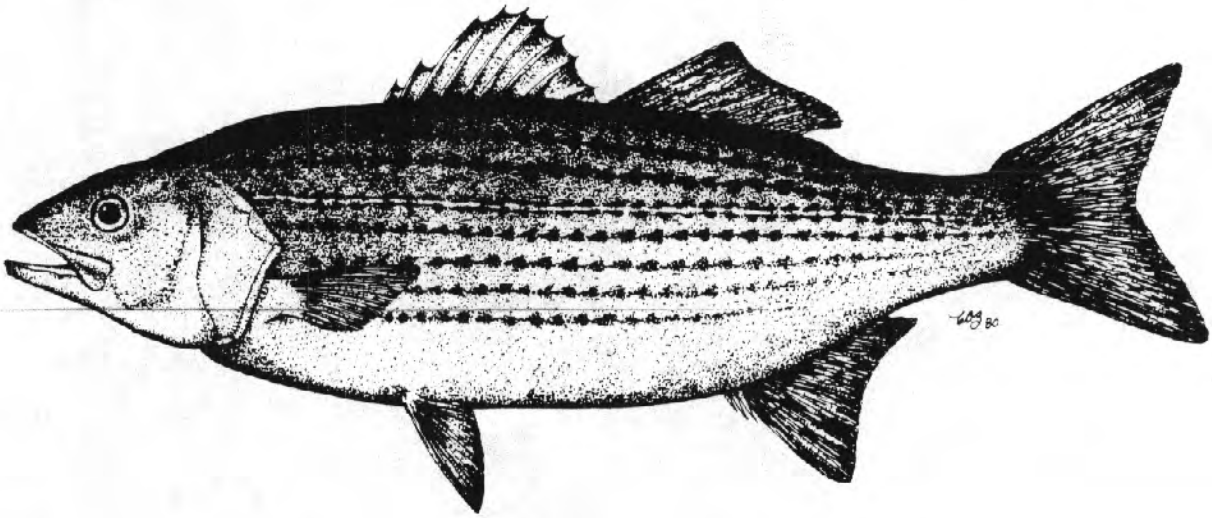


Tank Culture of Striped Bass Production Manual



Illinois Striped Bass
IDC F-26-R

Drs. William M. Lewis and R. C. Heidinger
Principal Investigators

Bruce L. Tetzlaff
Research Project Director in Charge of Production

Fisheries Research Laboratory
Southern Illinois University at Carbondale

TANK CULTURE OF STRIPED BASS

Illinois Striped Bass Project

IDC F-26-R

Drs. William M. Lewis and Roy C. Heidinger

Principal Investigators

Bruce L. Tetzlaff
Research Project Director
in Charge of Production

Fisheries Research Laboratory
Southern Illinois University
Carbondale, Illinois 62901

Photographs by Bruce L. Tetzlaff
(except where noted)

March 1981

DEDICATION

This manual is dedicated to the memory of

Roger E. Burrows

who did so much to further the development of water reuse in fishery production, and who made major contributions to the design of the facility described here.

Table of Contents

Part I: System design

1. Pilot facility design 1
2. Rearing tanks 4
3. Water supply and pumps 12
4. Supply and drain lines 13
5. Aeration 14
6. Biofilters 18
7. Brine shrimp facility 22
8. Makeup water 26
9. Temperature control 26

Part II. Rearing procedures

1. Biofilter activation 29
2. Filter maintenance 32
3. Transport of larvae 34
4. Stocking of larvae 34
5. Feeding the larvae 38
6. Training to accept a dry diet 44
7. Transfer to production tanks 48
8. Growth, production, and carrying capacity 52
9. Mortality and diseases 59
10. Water quality 64
11. System monitoring 71
12. Fingerling harvest 76
13. Transport and stocking 81

References..... 88

Appendices

A. Historical development of striped bass tank culture.. 95
B. Biofiltration in fish culture 102

PREFACE

The purpose of this manual is to present various criteria and techniques for the tank culture of striped bass in a water reuse system. These techniques have been developed over a period of seven years. The results of the previous work are summarized in Appendix A, while specific results of the various experiments can be found in Lewis and Heidinger 1974 and 1975, Lewis, Heidinger, and Tetzlaff 1977a, and in the 1977, 1978, 1979, and 1980 Annual Performance Reports for this project (Illinois Striped Bass F-26-R). Appendix B presents basic information on water reuse and biofiltration in fish culture.

During the seven years of development, survival of striped bass from one-day-old larvae to 5- to 10-cm fingerlings has increased from less than 1 percent in 1974 to 46 percent in 1980 (5,000 to 105,000 fingerlings). This survival compares very favorably with rates obtained in pond culture. A high percentage of these tank reared fish, sometimes 50 percent, did not have an inflated gas bladder. This phenomenon, however, is not unique to tank reared striped bass. In 1979, we obtained a small sample of fish from five ponds at the time of harvest from a southeastern hatchery; 0 to 55 percent of these fish did not have an inflated gas bladder.

When comparing survival rates between various culture systems, one should be careful to compare survivals only during the same developmental time frame. For example, most survival rates reported for open pond culture refer to rearing striped bass from 7 to 14 days

of age to 1-2 inches (approximately 50 days of age). Considering this time frame, the survival of our fish in 1980 was approximately 80 percent.

PART I: SYSTEM DESIGN

Pilot Facility Description[1]

The SIUC striped bass rearing facility at Gorham, Illinois, consists of thirty-four 1,730-liter (457-gallon) circular fiberglass rearing tanks. Water for these tanks is supplied by gravity from a 4,500-liter (1189-gallon) elevated constant-head tank. A schematic diagram of the water treatment system can be found in Figure 1. The constant-head tank is also utilized for mixing and water aeration. Water is pumped to the tank from the biofilter effluents, the pressure sand filter-UV sterilizer system, and the electronic boilers. Water passing through the pressure-sand filters and UV sterilizer can originate from either a well water pretreatment facility or from the biofilter effluents.

After passing through the rearing tanks, waste water flows by gravity through a parallel set of 4 biofilter modules (Figure 2). Each biofilter module consists of 4 concrete basins with a volume of 1,090-liters(288-gallons) for each basin (17,400-liters/4,597-gallons total volume for all biofilters). See page 17 for a description of the medium. These biofilters are designed as upflow units. Water enters a lower chamber, which acts as a sedimentation basin, and flows upward through the biofilter substrate. After passing through the biofilters the water is pumped to the constant-head tank.

---

[1] With minor alterations, the facility (consisting of the culture tanks, biofilters, and associated support equipment) used during the course of this investigation was designed and installed by UMA Engineering, Portland, OR.

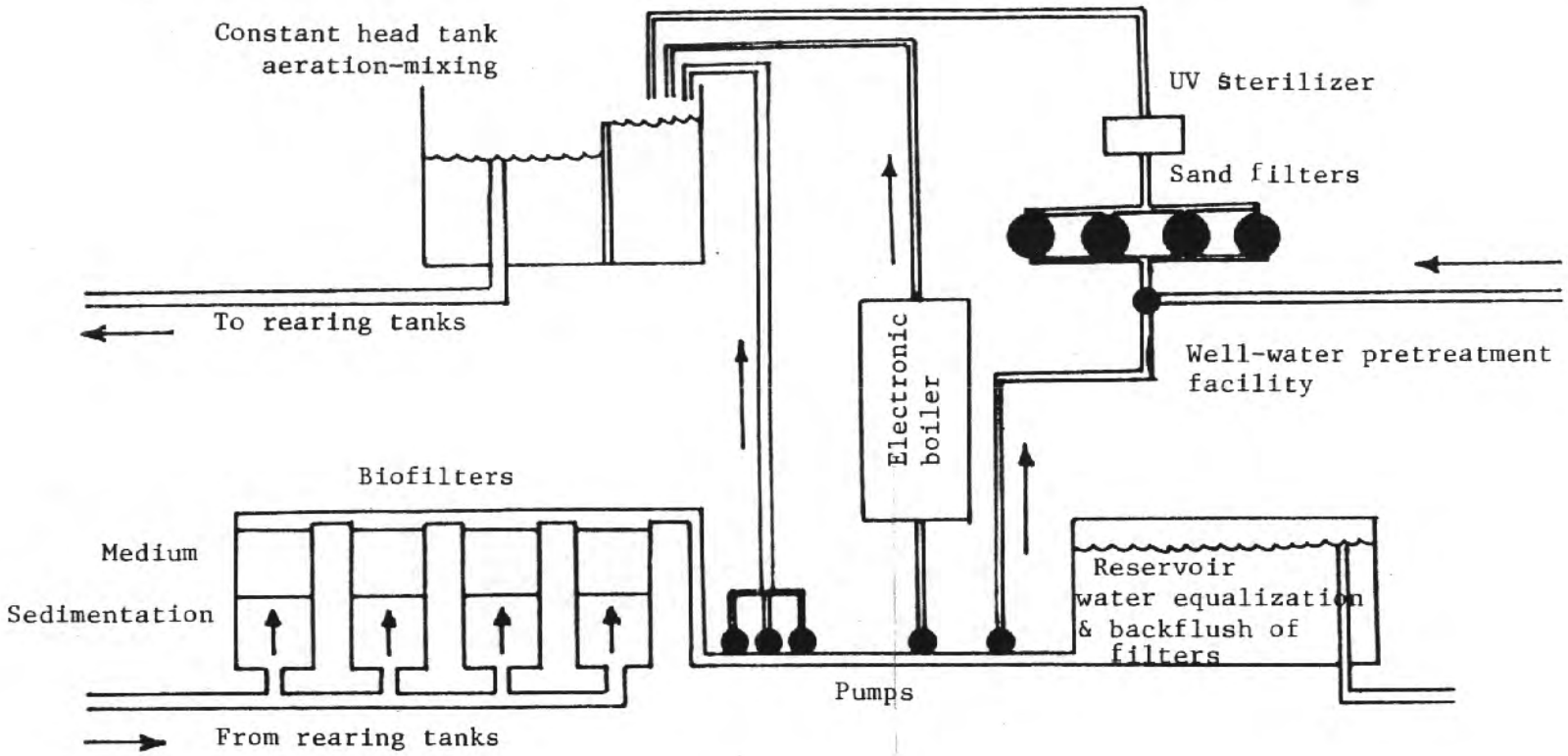


Figure 1. Schematic diagram of the water treatment facility of the striped bass rearing facility. (Air supply and emergency electrical power not shown).

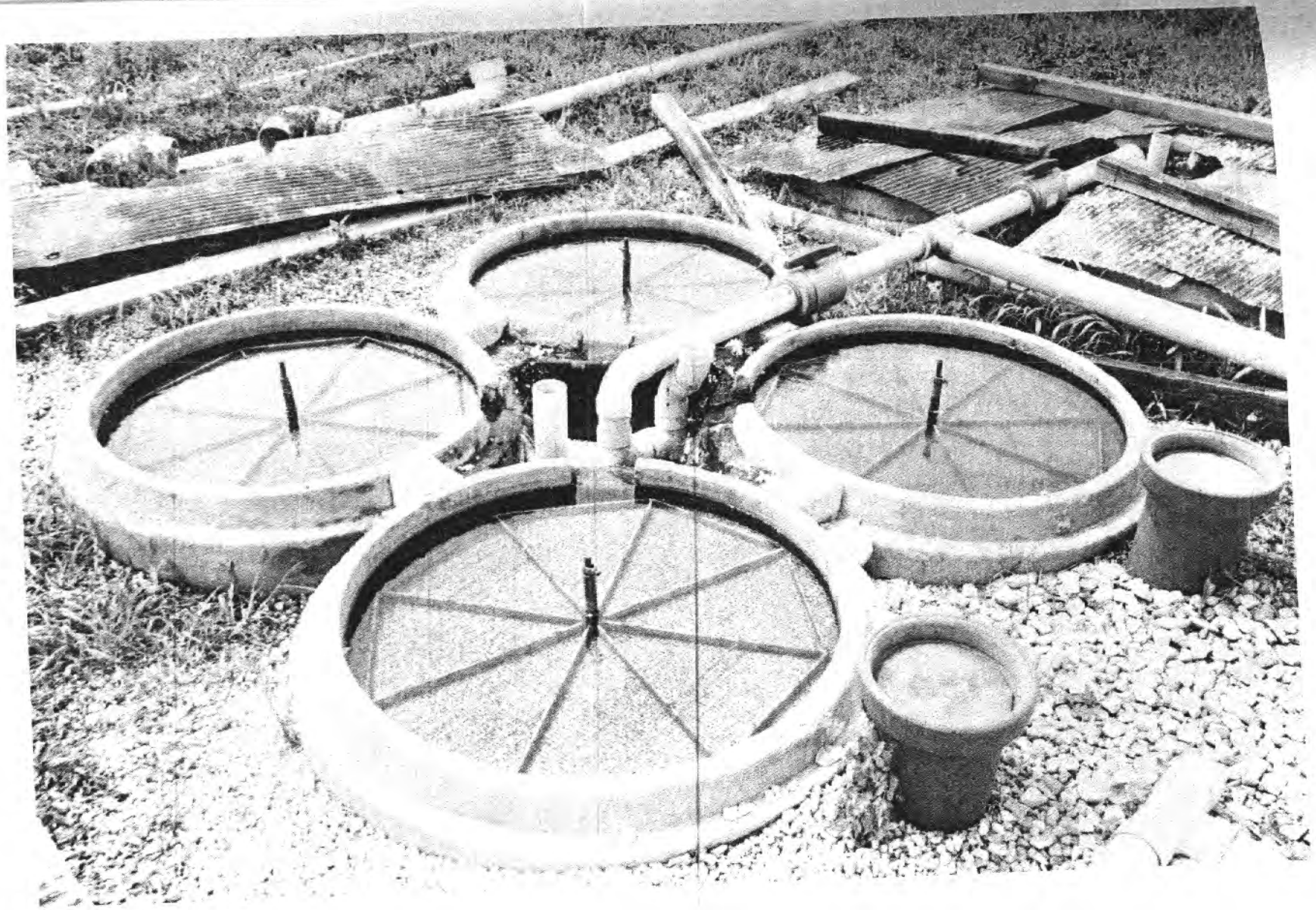


Figure 2. Upflow biofilters used at the SIUC Striped Bass Facility at Gorham, Illinois (substratum 1-cm polystyrene beads). Note the covers on the distant module to prevent algal growth.

Makeup water is supplied from a sand point well located at a depth of 50 feet. Prior to entering the system, the well water is aerated and passed through a sedimentation chamber with a one-hour retention time for the flocculation of iron. The water is then passed through a rapid sand filter which removes a large percentage of the flocculated iron. The remaining iron is removed by pressure filters of the type used for swimming pools.

All rearing tanks are enclosed in an uninsulated sheetmetal building to protect the striped bass fry and fingerlings from bright sunlight and inclement weather, and to prevent the growth of algae in the system.

Rearing Tanks

Production Tanks

Insulated circular fiberglass tanks with volumes of 1,730 liters and diameters of 1.8 meters (6 feet) and 0.8 meters (30 inches) deep are used as culture tanks (Figure 3). Circular tanks of this size provide ease of access for in-use cleaning. Ready access to the center of the tanks also facilitates harvesting the fingerling fish. Other tank shapes have also been used to successfully rear striped bass fingerlings, including concrete and metal raceways and rectangular tanks.

The standpipe is 69 cm (27 inches) high, giving a freeboard of 10 cm (4 inches). The freeboard prevents striped bass fingerlings from jumping from the tanks and lessens the possibility of the tanks overflowing if the fry retention screens become blocked.

The tubular standpipe covers (Figure 4) which are supplied with

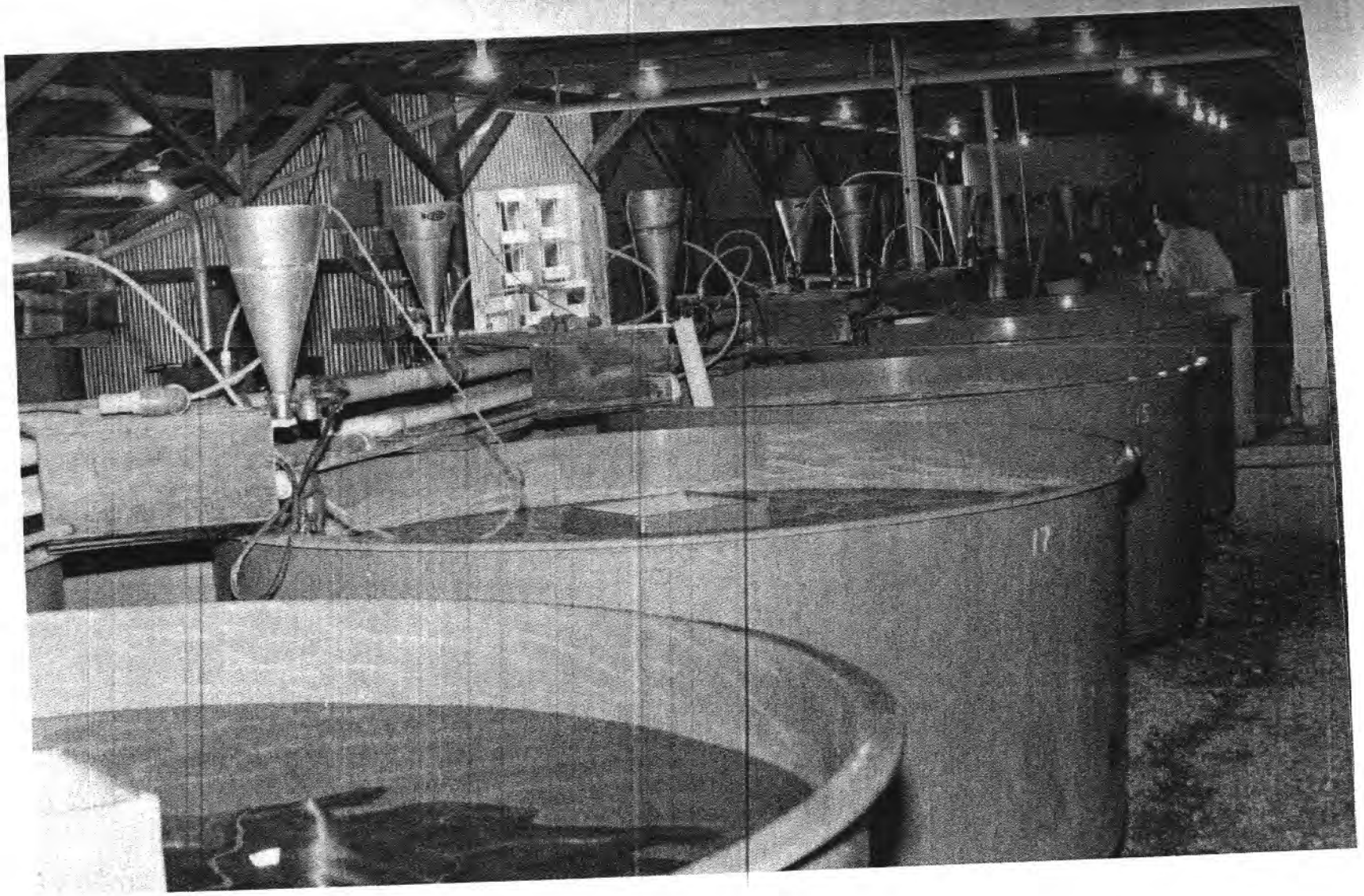


Figure 3. Circular rearing tanks used for striped bass fingerling production.

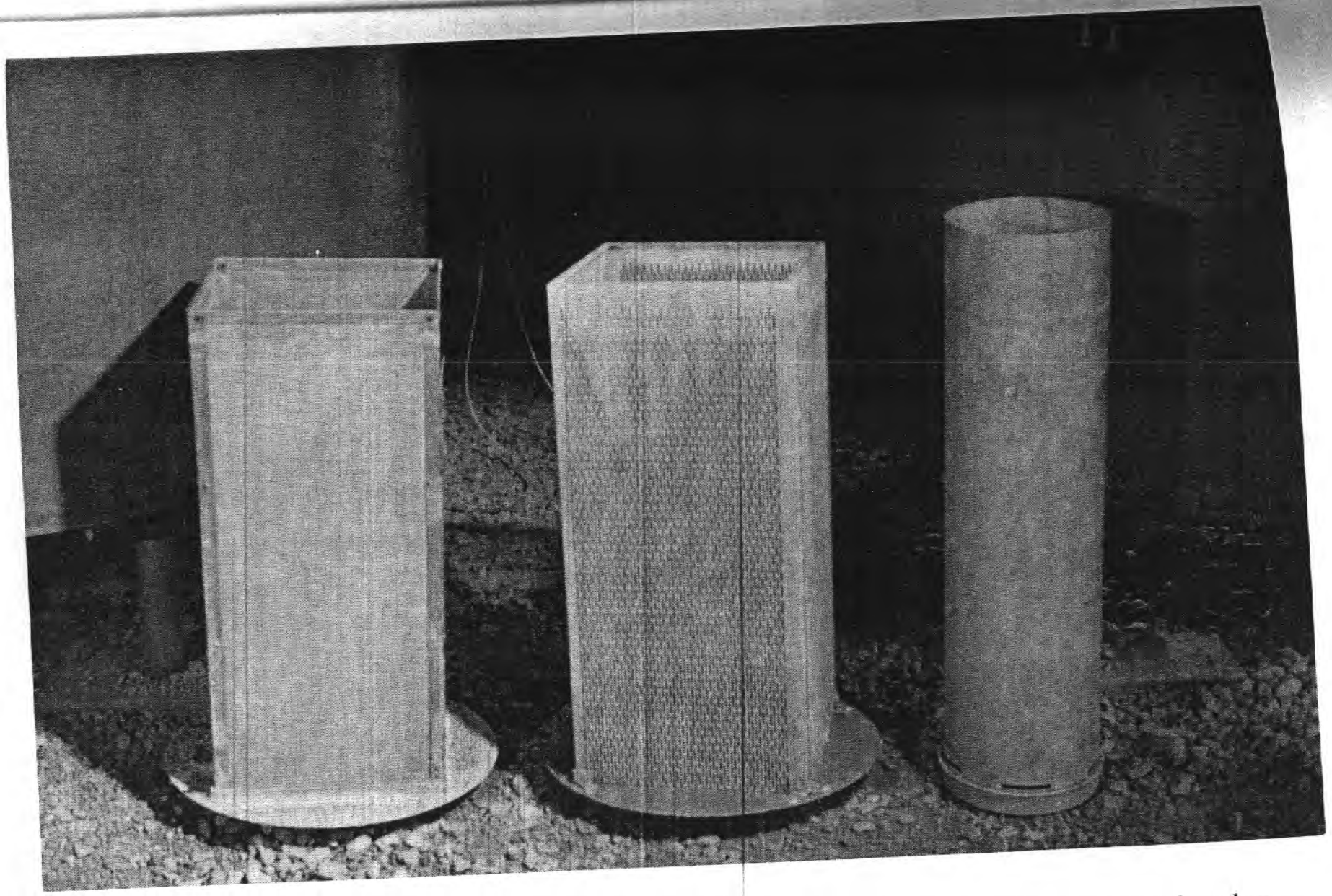


Figure 4. Fry retention screens used in the 1.8-meter circular tanks; 0.5 and 3.2-mm bar mesh screens and slotted standpipe cover.

the circular tanks are useful only when the striped bass attain 7.6 cm (3 inches) or more in length. Standpipe screens with smaller meshes and greater surface area are necessary to retain striped bass fry and small fingerlings. If the screen affords insufficient screening surface, the striped bass larvae can be impinged on the screen due to high velocity across the screen. A standpipe cover measuring 25 cm on a side and 76 cm high (12 inches x 30 inches) provides 9,290 cm^2 of surface area (1,440 $inches^2$) (Figure 4).

For striped bass fry and fingerlings less than 20 mm long (0.75 inches), a 0.5 mm bar-mesh screen (50 mesh/inch) is needed. Screen with this mesh rapidly becomes blocked with particulate matter and organic growth, thus necessitating frequent cleaning. The screens can be cleaned while in use by brushing. Due to the abrasion of frequent brushing, stainless steel or hard aluminum screen is recommended rather than fiberglass or nylon.

Similar standpipe covers with screen mesh sizes of 2 mm (12 mesh/inch) and 3.2 mm (8 mesh/inch) should also be available for use when the fish are 20 mm to 7.6 cm in size. After the striped bass reach 7.6 cm (3 inches) in length a standpipe cover with a slot size of 6.3 mm (0.25 inches) can be used. While using the standpipe screens (Figure 4) we have used these tubular standpipe covers by placing them loosely over the standpipe inside the screens. This will insure that a portion of the wastewater leaving the tank will be pulled from the bottom of the tank and thus prevent a layer of uncirculated water from forming. At various times the tubular

standpipe cover can be removed to allow surface film to pass through the screen.

Because of the high oxygen requirement and small initial size of striped bass, rearing tank water supplies must cover a wide range of flush rates and independently control velocity of circular flow. The latter control is accomplished by adjusting the angle of the inflow nozzle. The rate of flush is controlled by individual valves for each tank. A minimum diameter of 3.2 to 3.8 cm (1.25 to 1.5 inches) is also advised to eliminate the need for high water pressure in the influent manifolds. Flush rates for small fry are approximately 7.6 liters per minute (2 gpm), while large fingerlings may require a flush of 76 liters per minute (20 gpm) in each of the 1,730-liter tanks. The water supply pipe is directed vertically upward until the quantity of water prohibits this, after which the pipe is directed vertically downward. When fingerling striped bass surpass 6 cm in length, they tolerate circular currents. The circular current helps to avoid the accumulation of sediment in the tanks and assures uniform circulation.

Upflow Rearing Tanks

Striped bass larvae stocked into the circular fiberglass tanks before they are 6 days old have not survived (Lewis et al. 1977a). It is, therefore, necessary to use another type of rearing tank for the larval fish. The tank design which has given the best survival of larval striped bass utilized upflowing water currents. The tanks used at the SIUC facility are box-shaped, measuring 91 cm on a side,

and have a rearing volume of 550-liters(145-gallons). These tanks are shown in Figure 5a and diagrammed in Figure 5b.

The tanks are constructed of marine plywood. An aluminum plate containing 100 3.2-mm holes is installed approximately 10 cm (4 inches) above the bottom of the tank to serve as a diffuser. Twenty-eight cm (11 inches) below the upper rim of the tank a stainless steel screen, angled inward at 60°, measures 91 cm (36 inches) at the base, 61 cm (24 inches) at the top, and 33 cm (13 inches) on each side, thus providing 10,064 cm^2 of surface area (1,560 $inches^2$). Water velocity through the screen should not exceed 0.163 mm/second in order to prevent impingement of the larvae. Foam rubber strips are glued to the screen bottom to assure a complete seal with the retention screen bracket. All other possible gaps and holes in the screen and tank are sealed with silicone sealant. Considerable numbers of striped bass fry will escape through holes as small as 2 to 3 mm. It is also recommended that stainless steel bolts be used rather than steel or aluminum pop rivets in the screen construction. Corroded rivets have resulted in the loss of almost all fish in some tanks (Lewis et al. 1977a). Above the level of the screen base, a 7.6 cm (3 inch) overflow is installed in the side of the tank (Figure 5a and 5b). This overflow has a swivel joint, which permits adjusting the water level within the upflow tank to maintain a water-to-water interface across the screen. This design permits the use of the full screening surface and eliminates the danger of impinging the larvae. Since the upward flow of water exerts pressure on the screen, it must

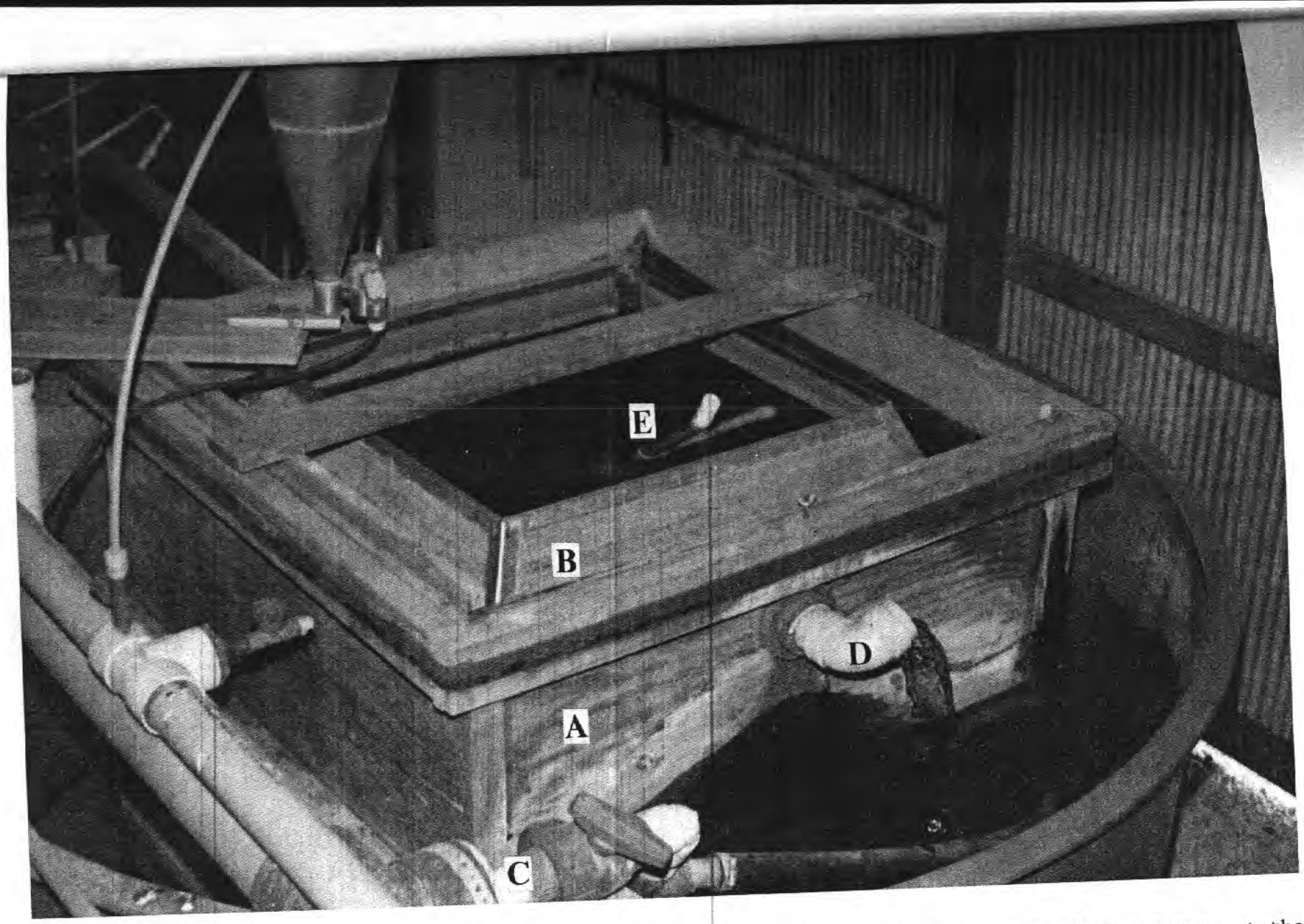


Figure 5a. Upflow tank inserted in a circular production tank used to raise striped bass larvae at the SIUC facility. A) Upflow tank, a 91-cm cube constructed of marine plywood with a false bottom of aluminum plate perforated with 100 3.2-cm holes. B) Larvae retention screen, 50 mesh (0.5mm) stainless steel screen. C) Water supply connects to tank below false bottom (diffuser plate). D) Overflow from behind larvae retention screen. E) Swivel pipe used to remove surface film.

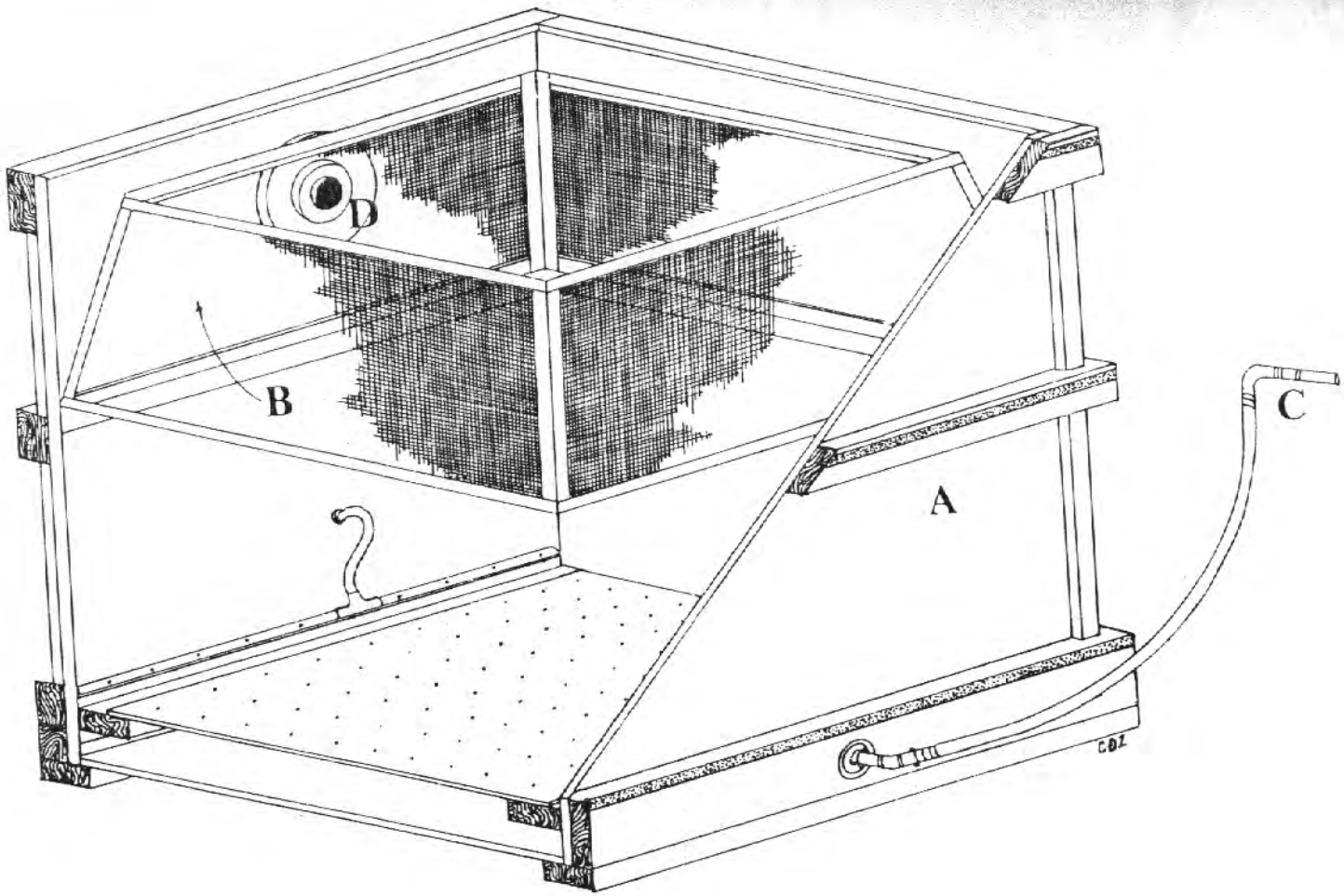


Figure 5b. Schematic diagram of upflow insert. (See 5a for description of components).

be firmly attached to the tank. A wooden frame which presses the screen edge against the screen bracket is bolted to the rearing tank.

Flush rates in the upflow are kept at the minimum to insure adequate levels of dissolved oxygen (7.0 ppm to saturation). The size of the holes in the water diffusion plate limits the flush rate to 26 lpm (7 gpm). However, a flush rate of 7.5 lpm (2 gpm) appears to be optimum for striped bass fry at the time of initial introduction into the upflow tank.

Once the striped bass are 10 days old, they can be harvested from the upflow tanks and stocked into the main rearing tanks. After harvest the upflow inserts can be removed, thus permitting the tank containing the upflow to be used as a rearing tank.

Water Supply and Pumps

Characteristics of the pumps which raise water to the constant-head tank will depend on the requirements of each rearing facility. If possible all pumps should be three phase. The SIUC facility uses a series of three 3-hp centrifugal pumps with a conbined pumping rate of 1,134 lpm (300 gpm). An additional 190 lpm is added to the constant-head tank by a 1.5 hp centrifugal pump which circulates a portion of the total flow through the electric heaters. This combined pumping capacity supplies 38 lpm (10 gpm) to each of the 34 rearing tanks. With this flush rate, 46 minutes are required for one complete turnover in the rearing tanks (1.3 turnovers per hour). We have found a turnover rate of 1.3 per hour to be inadequate to maintain water quality in the rearing tanks when they are heavily loaded with fish. Turover rates of 3 to 4 per hour would be more desirable. However,

this rate cannot be attained in the SIUC facility due to inadequate drain lines. Nickum (1978) found that walleye fingerlings require a minimum of two turnovers per hour when cultured intensively, while Pecor (1978) reports turnover rates ranging from 2.4 to 4.1 per hour when rearing northern pike X muskellunge hybrids. When the turnover rate is low, the tanks are not being used efficiently. To maximize the efficiency of the rearing tanks, the fish should be crowded into them (with corresponding increase of flush rates) up to the point where water velocity is not excessive or growth of the fish is not inhibited by the high density. To date the optimum density (kg fish/m^3) has yet to be determined for striped bass.

Although less efficient in energy use and cost, a series of smaller pumps can be more beneficial than fewer large-capacity pumps. During the initial stages of rearing, when minimal flush rates are desired, the system can be operated with one or two pumps. Later, if mechanical difficulty occurs in one pump, the remaining pumps can still provide considerable water to the fish. In a system operated with few pumps, the loss of a pump can result in inadequate flushing of the system.

A separate pump should be installed for backflusing the various filters. In the SIUC pilot facility, water from this pump can also be directed through pressure sand filters to the constant-head tank. A second backflush pump is also used at this facility to rapidly drain the biofilters during backflushing.

Water Supply Lines and Drain Lines

To avoid heavy metal toxicity, PVC pipe and valves should be used

for all water lines. Pipe containing zinc or copper can be toxic to both the striped bass and the nitrifying bacteria. However, new PVC pipe may contain toxic residues. Therefore, the system should be thoroughly flushed prior to the introduction of fish or the activation of the biofilter.

Generally, the water input lines and headers should be 1.5 times the size that is required for maximum flow. During the rearing season calcium deposits and biological growth can restrict the flow.

Drain lines should be twice the size that is hydrodynamically necessary. Considerable amounts of biological growth can develop which will reduce the flow rate by the time of harvest, when the greatest need for maximum tank flush rates exist. Further, harvest of the fingerling striped bass is complicated if individual tanks cannot be rapidly drained. To restore flow lost to biofouling, consideration should be given to installing duplicate drain lines. Alternate use of drain lines results in death of the fouling organisms during periods when a line is not in use. As a further precaution for maintaining full flow, drain lines should have a number of readily accessible cleanouts.

Aeration

System Water Aeration

At the SIUC rearing facility, recirculated water is pumped to the constant-head tank through aspirators as described in Burrows and Combs (1968). The aspirators consist of a 3.8 cm (1.5 inch) diameter pipe and an open 1.9 cm (0.75 inch) air intake pipe (Figure 6). Water flowing through the outer pipe draws in air and

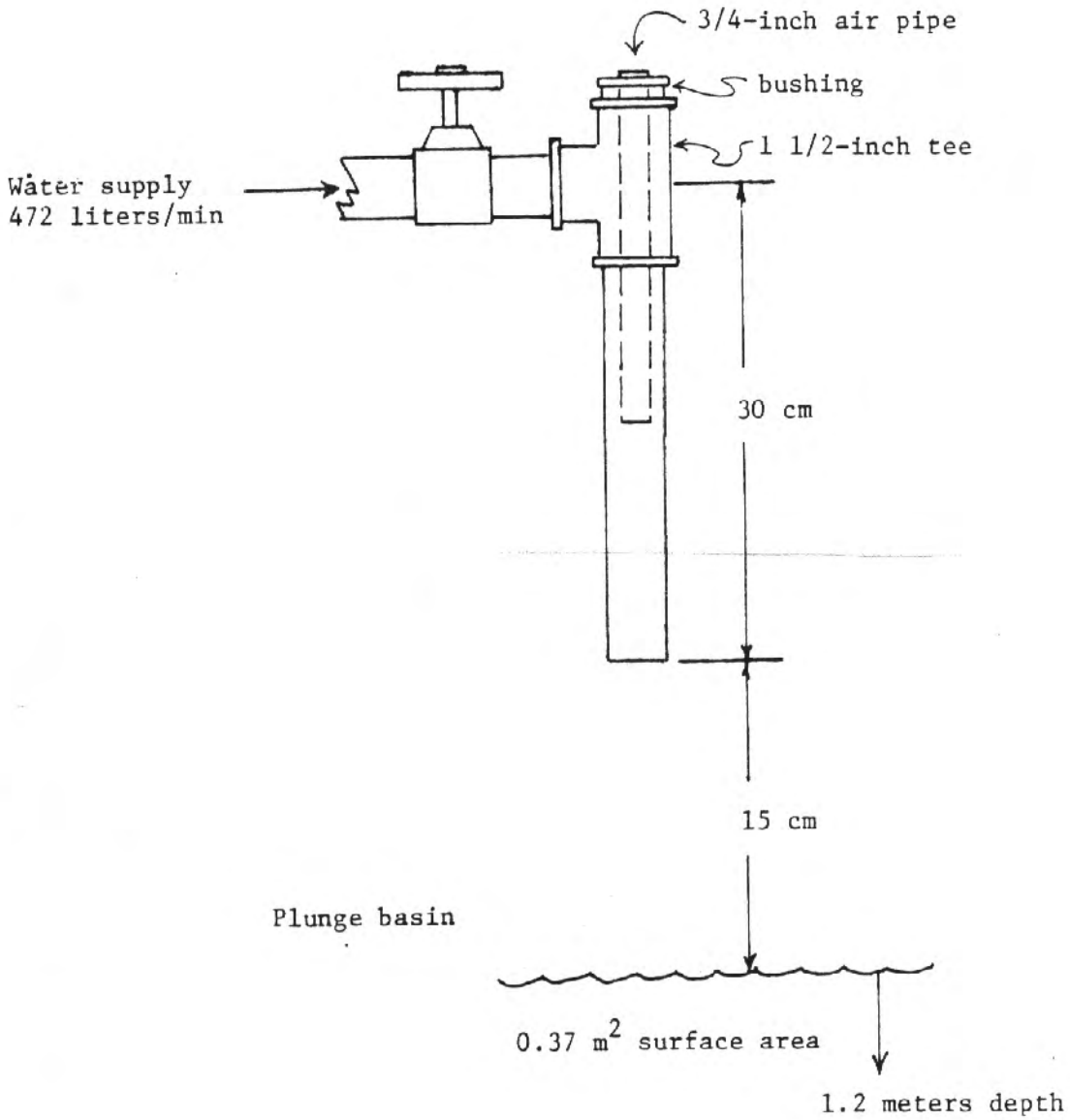


Figure 6. Aspirator water aerator (after Burrows and Combs 1968).

plunges into a basin 1.5 m (5 feet) deep. As described by Burrows and Combs, each aspirator requires a flow of 472 lpm (125 gpm), at a pressure of 0.7 kg/cm^2 (10 psi), a surface area of 0.37 m^2 (4 $feet^2$), and should be placed 15 cm (6 inches) above the water surface (Figure 6). Burrows and Combs (1968) state that this system maintains oxygen concentrations within 90 to 100% of saturation These authors prefer plunge basin aspirators over closed pipe venturi systems, since the latter may produce super-saturation of nitrogen while failing to remove carbon dioxide. The boiling action produced by the aspirators (Figure 7) is reported to adequately remove carbon dioxide without nitrogen supersaturation. The addition of pure oxygen into the rearing tanks has been considered. Additional oxygenation in the rearing tanks could increase the carrying capacity of the tanks to the point that ammonia became the limiting factor.

Compressed Air

Supplemental aeration in the rearing tanks is not necessary until artificial feed is utilized. After this time, the addition of compressed air will help to mix the water, and, if placed near the standpipe screens, reduce the rate at which the screens become blocked with particulate waste.

During the period in which the striped bass fry are being fed brine shrimp nauplii, compressed air is needed in hatching the brine shrimp cysts and to keep the hatched nauplii in suspension while in the automatic feeders. At the SIUC facility brine shrimp are hatched

Figure 7. Boiling action of aspirator type water aerator.

in small individual units (11.3-liter containers). Ninety-six of these containers, each with one aquarium airstone, are required to produce sufficient shrimp. An additional 20 to 40 airstones are used in the shrimp feeders.

A large quantity of compressed air may also be used to upwell the biofilter medium during backflushing operations. Compressed air boiling up through the medium during back washing will loosen heavy bacterial coatings and particulate matter A 3-hp Roots® rotary blower provides sufficient compressed air for our system (125 cubic feet/minute).

Since relatively low pressure air is used for these operations, PVC plastic pipe can be used for air supply lines. However, this pipe will transmit air blower sounds which can be disturbing to personnel.

Biofilters

The biofilters in the SIUC facility (Figure 2) are of the submerged upflow design. Styrene beads 1 cm in diameter are used as a substrate for bacterial growth. Depth of the medium in the filters is 46 cm (18 inches) which provides a surface area of 2,500 m^2. To determine the maximum theoretical carrying capacity of the biofilter, the ammonia oxidation rate must be known. Biofilter ammonia oxidation rates reported in the literature are highly variable. Liao and Mayo (1974) were able to achieve rates up to 1,407 mg NH_3-N/m^2 media surface/day when experimenting with salmon at 12C. However, Davis (1977) recommends that calculated ammonia loading rates not exceed 200 mg/m^2/day to maintain good growth in channel catfish. Thus,

for the following calculations, we assumed the approximate oxidation rate of the biofilter at the SIUC facility to be 600 mg NH_3-N/m^2/day x 2,500 m^2 = 1.5 kg NH_3-N/m^2/day. Based on an estimated pollution rate of striped bass (0.0270 kg NH_3-N/kg feed/day), a theoretical 55.5 kg feed could be used each day. To convert the quantity of food to numbers of fish, the feeding rate is divided into this quantity. The feeding rate for 5-cm (1.5 g) fingerlings is 10 percent of body weight per day, while large fingerlings receive approximately 5 percent per day. The theoretical carrying capacity is thus 555 kg of 5-cm fingerlings and 1,100 kg of larger fingerlings. By dividing these carring capacities by average weights, the numbers of fingerlings can be determined. The theoretical carrying capacity of our system is thus 370,000 5-cm (1.5 g) or 84,100 10-cm (13.2 g) fingerlings. However, water quality problems have risen with densitites far below these maxima. During 1976, ammonia-nitrogen levels increased to levels above 2 mg/l with a density of 21,100 10-cm fingerlings weighing 205 kg (Lewis et al. 1977a). In 1978, ammonia-nitrogen again approached 2 mg/l with densities of 64,100 fingerlings weighing 123 to 216 kg (Lewis et al. 1978).

There are several reasons for this apparent discrepancy. As discussed in Appendix B, three water quality parameters affect the rate of ammonia oxidation in the biofilter; namely, temperature, initial ammonia influent level, and dissolved oxygen. We feel that oxygen is the limiting factor in our biofilter, inasmuch as the temperatures and ammonia influent in our system are comparable with those in other

systems. There are two factors in the design of our system which inhibit oxygenation of the biofilter: clogging of the medium, and low water flow to the biofilter through pipes preventing the effluent to be exposed to the air.

Striped bass require a prolonged training period before they will readily accept commercial feeds. During the training period the striped bass fry are supplied with food at 25% of their body weight daily. Much of this food enters the biofilters as undigested particulate waste. The small size of the substratum for bacteria attachment used in our facility results in much of this waste being trapped in the filter. Heterotrophic bacteria then act upon this waste resulting in the production of additional ammonia and the consumption of oxygen. In addition, as the heterophic bacteria multiply, they smother the nitrifying bacteria thus considerably reducing the effective surface area of the substratum.

The low water flow of our system (38 l/minute to each tank) results in turnover times in the tanks of 45 to 50 minutes. During this time the fish utilize much of the available oxygen, resulting in the effluent oxygen concentrations of 5 mg/l. In the closed pipes, the tank effluents enter the biofilters without regaining oxygen. With biofilter influent concentrations of 5 mg/l, the amount of oxygen available to the bacteria is only 1 to 2 mg/l.

A more efficient design would employ a method of removing the particulate matter prior to biofiltration. The lower portions of the biofilters at the SIUC facility were designed to act as sedimentation basins for this purpose. However, the small particle size of the feeds needed for striped bass results in waste which does not

efficiently settle out in these basins. To effectively remove these wastes, sand filtration would be required.

Biological clogging of the biofilters is also common. Filamentous bacteria of the genus Sphaerotilus can develop to the level at which large portions of the biofilter substrate will adhere to itself and form large mats (Burrows and Combs 1968; Combs 1974). Although bacterial clogging can be controlled chemically, a biofilter with easy access to facilitate mechanically agitating the medium would simplify the removal of the mats. Clogging and matting on our biofilters was alleviated by cleaning one of the four modules daily.

In the SIUC system to reoxygenate the effluent from the rearing tanks, pure oxygen is injected through a stone diffuser into the drain-lines between the rearing tank effluents and biofilter influents. Sufficient oxygen is added to bring the biofilter influent concentration to saturation.

Based on ammonia levels, the carrying capacity of our biofilter exceeds 375 kg of 5 to 7-cm fingerlings when oxygen is maintained in the filter. This is equivalent to an ammonia oxidation rate of 202.5 mg NH_3-N/m^2/day. During 1979 ammonia-nitrogen never exceeded 1 mg/l with this density of fingerlings (Lewis et al. 1979). It is therefore, possible that with the improved maintenance our biofilter would support 555 kg of striped bass fingerlings. However, if the space is available for a large biofilter, an ammonia oxidation rate of 200 mg NH_3-N/m^2/day should be used when designing the facility. by over-filtering the system a margin of safety would be included.

In addition to mechanical agitation, facilities for upwelling the

biofilter medium with compressed air would expediate the cleaning process. Low density medium such as styrene beads do not lend themselves to this method of cleaning, since they float above the water surface. Brittle materials, such as rock and oyster shell, tend to chip and form fine particles during this process and thus have to be replaced periodically.

During the backflush procedure the total flow of the system must pass through the filters which are not being serviced, hence an additional load is placed on the remaining filters. The size of pipes and valves used for the biofilters should be adequate to accommodate this added flow.

Brine Shrimp Rearing Facility

At the present time, live brine shrimp nauplii (*Artemia*) are an essential component of the diet of striped bass larvae. The striped bass are supplied with brine shrimp from an age of 4.5 days after hatching until the fish readily accept a powdered dry feed (20 to 30 days old when reared at 20 to 25 C).

At the SIUC rearing facility, 77 to 125 g of brine shrimp cysts are hatched in 11.3-liter containers (6.8 to 11.1 g/liter). The hatching medium used is a solution of 3.0 to 3.2% salt (uniodized rock salt).

The brine shrimp cysts are allowed 72 hours to hatch at a temperature of 24 C (75 F). To maintain temperature the rearing containers are kept in a heated, uncirculated water bath. Ninety-six of these containers are kept in operation, with 32 being used each day. Because

of the importance of brine shrimp nauplii to the success of rearing striped bass, the shrimp rearing facility should be protected from changes in weather, preferably enclosed in an insulated building.

The brine shrimp are supplied to the striped bass at hourly intervals, 24 hours per day. At the SIUC facility Neilson® Brine Shrimp Feeders are used (Figure 8). Each feeder is supplied with an airstone to maintain the live nauplii and to keep them evenly distributed in the solution. These solenoid valve feeders have a feeding rate of approximately 150 ml/second. An Eagle Signal Cycl-Flux® poultry timer system is used to control the feeders. The feeders used at the SIUC facility have a capacity of 4 liters (1.1 gallon). To supply sufficient nauplii to 50,000 striped bass fry for a period of 6 hours, two of the feeders are required. A shrimp feeder with a capacity of 8 to 10 liters would increase the efficiency of the feeding procedure.

Various methods have been developed to separate the live brine shrimp nauplii from the cyst shells, and to concentrate the brine shrimp. We use a series of 10-liter separatory funnels (Figure 9). Cylinders with an inside diameter of 15 cm (6 inches) are equipped with a 1.2-cm diameter drainpipe. The drainpipe is installed in a sleeve with an O-ring seal, thus it can be raised to various levels in the water column to drain off the nauplii as they separate from the shells and unhatched eggs. When the hatching medium containing the brine shrimp nauplii is held in the cylinder, three layers form. The shells from the hatched cysts sink to the bottom, the unhatched

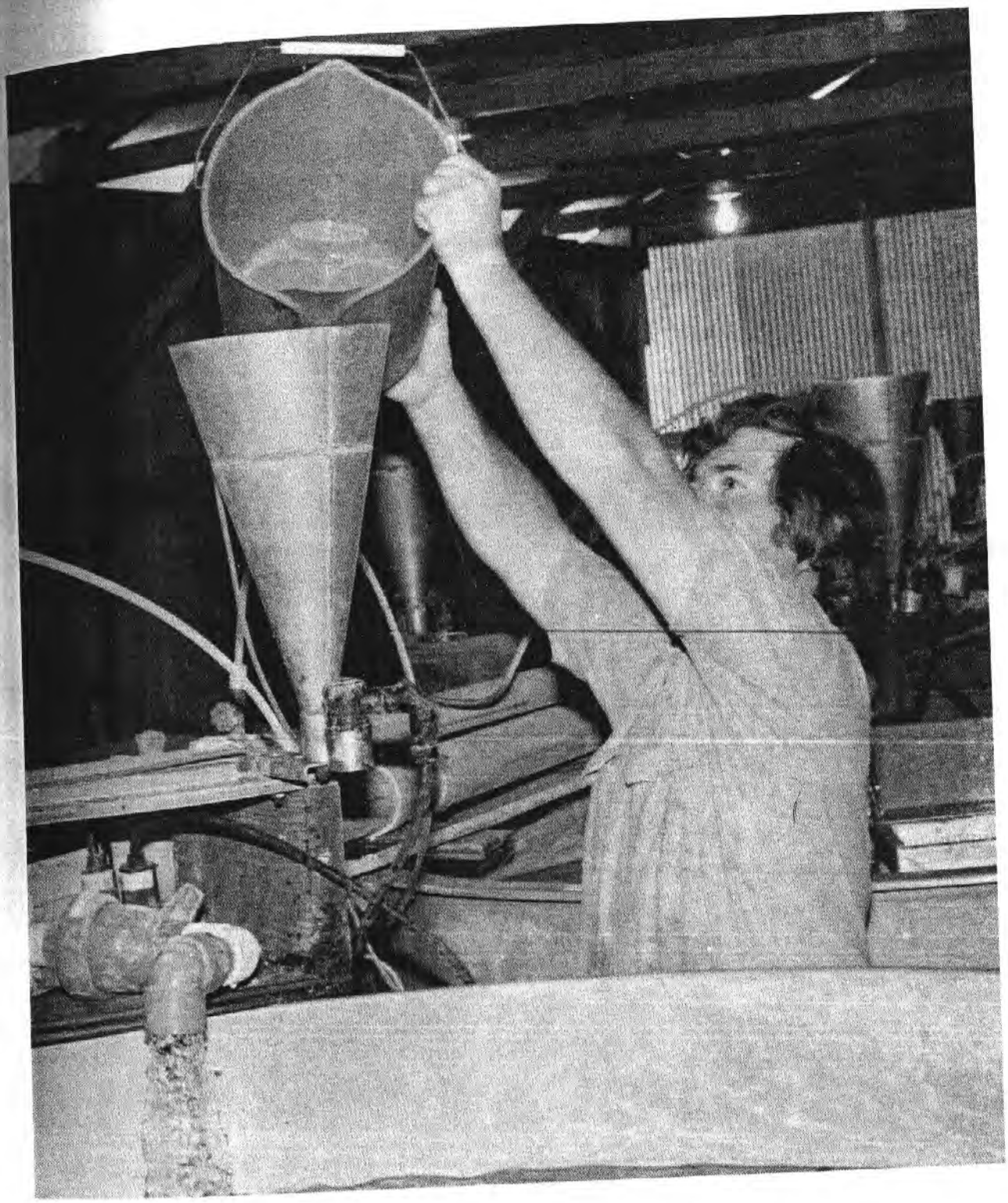


Figure 8. Four-liter brine shrimp feeder used at the SIUC facility.

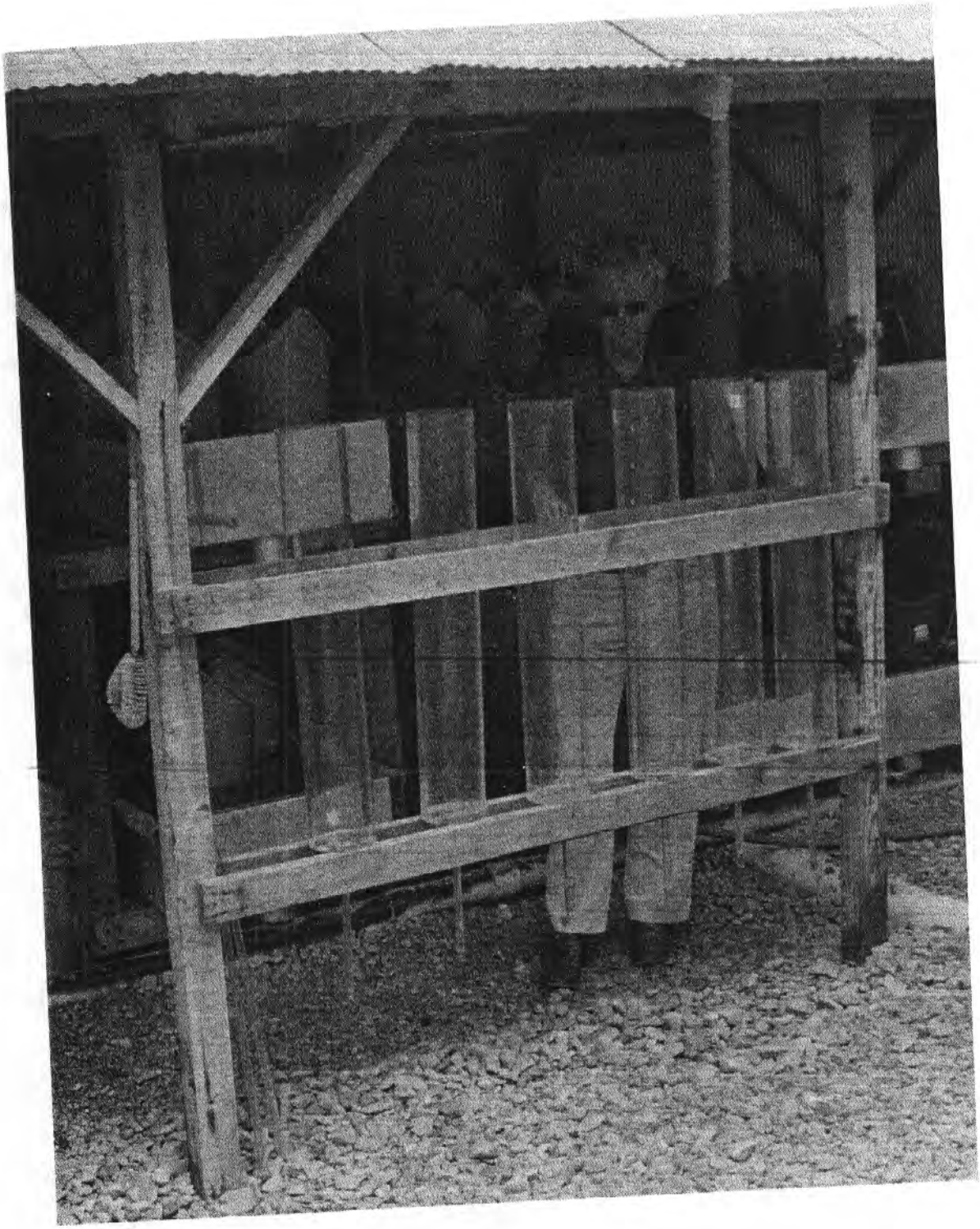


Figure 9. Brine shrimp separators. The drain tube extending from the bottom of the large cylinder can be moved vertically to the level above the shells and where the nauplii are concentrated. (Photo by John Richardson)

cysts float to the surface, and the live nauplii remain in the water column. Over a period of 5 to 15 minutes the nauplii begin to sink, permitting them to be concentrated in a layer over the cyst shells. By varying the settling time, various concentrations of nauplii can be obtained.

Makeup and Flush Water

In order to replace water used for filter backflushing and to prevent the accumulation of nitrate and phosphates, a supply of makeup and flush water is needed. In many recirculating systems a constant flush equalling 2 to 5 percent of the total flow is used. In the SIUC facility flush water is pumped only for 2 to 3 hours per day to replace water used for filter backwashing or occasionally to cool the system. However, this facility incorporates an in-line storage reservoir. The makeup water is supplied from a sand point located 50 feet below the ground surface. Before use, the water is aerated and subjected to a 60-minute retention period, then passed through a rapid sand filter and a pressure sand filter.

Temperature Control

The use of recirculated water in the culture of fingerling fishes provides an opportunity to control water temperature which is not economical in pond or flow-through rearing systems. Once the temperature of the water has been adjusted, relatively small amounts of energy are required to maintain constant temperatures. The majority of the energy is used to adjust the temperature of the make and flush water.

At the SIUC rearing facility, striped bass fry are introduced into the system at water temperatures of 15 to 18 C. When the fry reach an age of 10 days, the temperature is raised to 25 C at a rate of 1 to 2 C per day. The warmer temperature increases the growth rate of the fish. Also, in warm water the fingerlings appear to be more easily trained to accept an artificial diet. Brandenburg et al. (1979) had greater success in training largemouth bass fingerlings to accept an artificial diet at 27 C than at the lower temperatures tested. Although no empirical data are available, striped bass fry appear to behave in a similar manner.

The heating requirements of a system using recirculated water depend upon several factors, including the ambient temperature, surface area of water exposed to the air, amount and type of auxiliary aeration used, and the amount and temperature of makeup water.

The SIUC striped bass rearing facility is small enough (101,000 liters) to make the use of electric heaters practical. In this system, 189 liters per minute (14% of the pumped water) is passed through a set of three heaters having a total output of 85 kilowatts. This amount of heat is adequate to maintain the system at 25 C $\pm$ 2 C, while adding 14 C makeup water at an average rate of 2% of total flow (26 liters per minute). However, when the flush is increased to 10% of total flow (132 lpm), the system temperature drops 2 to 5 C, depending upon ambient temperature.

Burrows and Combs (1968) have had success using a stainless steel heat exchanger and heat pump to control the temperature in

larger systems. These authors found that maintaining 10% of the water flow (the makeup water) at the desired temperature offers adequate control. Changes of 4 C in the ambient air temperature will result in changes of 1 C in the rearing unit.

Another possible source of heat for a rearing system using recirculated water is the waste heat of electric power generating plants. This would then be a factor in the choice of location for a rearing facility. The large volume of water available for heat storage, and the fact that the water is circulated suggest that solar heating may also be practical.

Depending on the latitude of the rearing facility, a method of cooling the water may also be needed. During some years the rearing water temperature at the SIUC facility can surpass 31 C and thus approach the lethal temperature of striped bass (see the section on water temperature). When the system temperature reaches 30 C we pump 14 C well-water to lower the temperature to 25 to 27 C. Well-water is also used to lower the water temperature to 18 to 20 C for harvest of the fingerlings.

PART II: STRIPED BASS REARING PROCEDURES

The procedures detailed below are summarized in Figure 10. This figure shows the time sequence of the events based on the age of the striped bass.

Filter Activation

Prior to the introduction of fish into the system the biofilters should be activated. Activation (Meade 1974) assures that sufficient nitrifying bacteria are present in the filters when the fish are introduced. During activation the biofilters can be inoculated with Nitrosomonas and Nitrobacter, either from pure cultures, or by adding a small amount of garden soil to the filters. These bacteria are common in most soils, as well as in stream and lake sediment. To make activation more complete, sources of ammonia and other nutrients needed by the nitrifying bacteria can be added to the system. During activation one can limit circulation of water to passage through only the biofilters, as opposed to circulating the water through the entire system. Both organic and inorganic sources of ammonia have been used for activation of biofilters. Siddall (1974) found that 3.3 mg per liter of $(NH_4)_2SO_4$ or 3 to 6 mg/l of NH_4Cl were 2.4 to 2.9 times more efficient as sources of ammonia than organic sources. Meade (1974) developed a mixture of $(NH_4)_2HPO_4$ and other nutrients which he used in several systems. He recommends a stock solution of:

Tap water	1,000 liters
$(NH_4)_2HPO_4$	40 grams
Na_2HPO_4	40 grams
Artificial sea salt	40 grams
$CaCO_3$	250 grams

Age of fish (days)	Rearing tank	Diet	System operation
-21			Fill system and active biofilter[1]
- 5			Equilibrate system temperature to hatchery[2]
1	Upflow		Stock fish, commence incubating brine shrimp cysts
4.5		Live brine shrimp	
10		Brine shrimp and ground Tetra SM[3]	Start to raise water temperature to 25 C at 1 C per day
12	Begin transfer to circular tanks	Begin adding Salmon diet	
15		Increase brine shrimp ration	Temperature should be approaching 25 C
20	Circular		
30		Terminate brine shrimp and Tetra	Begin daily cleaning of biofilters. Grade out cannibles if necessary.
45		Salmon feed	Begin oxygenation of biofilter

[1] 21 days before fish arrive

[2] 5 days before fish arrive

[3] Tetra SM, a commercial grade of Tetra-Min®

Figure 10. Time series diagram of the striped bass rearing procedure.

Meade maintains an ammonia concentration of 10 to 20 mg/l during the activation period, and a pH of 7.5. He also activates the filters above the temperature at which the system is to be operated. In a system to be used for rearing salmon at temperatures of 13 to 14 C, Meade activates the filters at 23 C. Inasmuch as striped bass are reared at 23 to 25 C, an activation temperature of 25 C should be adequate to develop sufficient densities of Nitrosomonas and Nitrobacter in the biofilters.

Activation of the biofilters can occur slowly. Kawai et al. (1964) found that densities of the various bacteria required up to two months to reach equilibrium in marine aquariums. However, we, as well as Sidall (1974) and Spotte (1970), have found that an activation period of two weeks is adequate to develop sufficient populations of nitrifying bacteria in fresh water systems.

Burrows and Combs (1968) have had success in stocking fish at low densities into recirculated systems without activation of the biofilters. Spotte (1970) discusses this technique and recommends several ammonia-tolerant species of fishes that can be used to activate a new biofilter.

At the SIUC striped bass rearing facility, an activation period of 2 to 3 weeks is used. Ammonia (NH_4Cl) is added initially at concentrations of 1 to 3 mg/l. Longer activation periods are recommended with higher initial ammonia levels if sufficient time is available. Sufficient time should also be given to allow the water temperature to be lowered to that of the hatchery producing the larvae.

During activation with one initial load of ammonia there is generally a gradual decrease in ammonia and simultaneous increase in nitrite (Kawai et al. 1964). Nitrite is not converted to nitrate until most of the ammonia has been oxidized to nitrite. Growth of the nitrite-oxidizing bacterium Nitrobacter is inhibited by high ammonia concentrations (Lees 1952). This phenonmenon is shown graphically in Appendix B, Figure B-2.

Filter Maintenance

The submerged upflow biofilters used at the SIUC striped bass rearing facility require no maintenance until the fish are 30 days old. At 30 days, the fish are being trained to accept artificial feeds. The training results in large quantities of fine particulate waste entering the filters. It therefore becomes necessary to backwash the filter. From day 30 to the end of the rearing season one of the four biofilter modules is backflushed daily. The biofilter wash procedure entails draining the module and washing out the accumulated sludge (Figure 11). Mechanical agitation of the polystrene beads is also required.

When the fish are 40 days old, bacterial activity in the biofilters is sufficient to deplete the dissolved oxygen in the filters. Therefore, from this time onward, as indicated previously, pure oxygen is added to the culture tank effluent in sufficient quantities to maintain dissolved oxygen in the biofilter influents near saturation.

Figure 11. Hand stirring of the biofilter substratum while backwashing to remove particulate matter.

Transport of Larvae

The striped bass larvae raised in the SIUC facility are air shipped from the East Coast. For shipment the larvae are packaged in plastic bags with an atmosphere of pure oxygen. The bags are then placed in insulated shipping cartons. Larvae are usually shipped when they are 1 to 2 days old. The fish we have received have in most cases exhibited good survival for at least 8 hours of shipping and holding in the plastic bags. Although little mortality occurs during shipping, 2.5- to 4-day old larvae have shown poor survival after stocking; therefore, we recommend obtaining either 1-day-old or preferably 5-day-old larvae. Bonn et al. (1976) recommended shipping striped bass fry at densitities up to 10,600 per liter of water (40,000 per gal). Due to variations in shipping container size among the various hatcheries, larvae are shipped in quantities ranging from 50,000 to 400,000 per container. Arrangements should be made with the hatchery to ship the fry in quantities that would stock one or two rearing units (50,000 or 100,000 per bag for the 550-liter upflow tanks described earlier). Counting the fry after they have been stressed by transport may result in mortality.

Stocking Larvae into the Upflow Tanks

Striped bass larvae should not be handled at ages of 2.5 through 4.0 days old. When larvae of this age are transported and stocked into the rearing system, 90 to 100 percent mortality occurs within 48 hours (Lewis et al. 1977b, 1979, 1980). Little initial mortality occurs with 1-day-old larvae or larvae older than 4 days. At the time the larvae

are to be stocked into the upflow tanks, the water temperature should be adjusted to the temperature of the hatchery where the fish are obtained. The shipping bags containing the larvae should be placed in the rearing tanks to temper the fish to the rearing water temperature. Most hatcheries double-bag the larvae. Removal of the outer bag will reduce the time needed to temper the fish. Tempering times of one-half hour per degree centigrade difference appear to be adequate. The temperature of the shipping water can be estimated by placing a thermometer between the shipping bag and insulation while the bag is still in the shipping carton.

The method we have used to rapidly divide the fry from one shipping bag between two rearing tanks utilizes a string noose (Figure 12). The shipping bag is placed in the rearing rank and rocked from side to side to evenly distribute the larvae. A slipknot noose is placed around the vertical center of the bag and tightened. Once the bag is sufficiently constricted to divide the fry, one side of the bag is opened while the noose is tightened completely. The divided bag is then quickly cut near the noose on the other side.

Water tempering the striped bass fry by the addition of tank water directly to the shipping bags is not recommended unless some water quality parameter in the rearing facility is abnormal; for example,very high or low methyl-orange alkalinity, very high salinity or sulfates, etc. In attempts to water temper larvae at the SIUC facility no differential mortality was noted between tempered and untempered larvae (Lewis et al. 1977a).

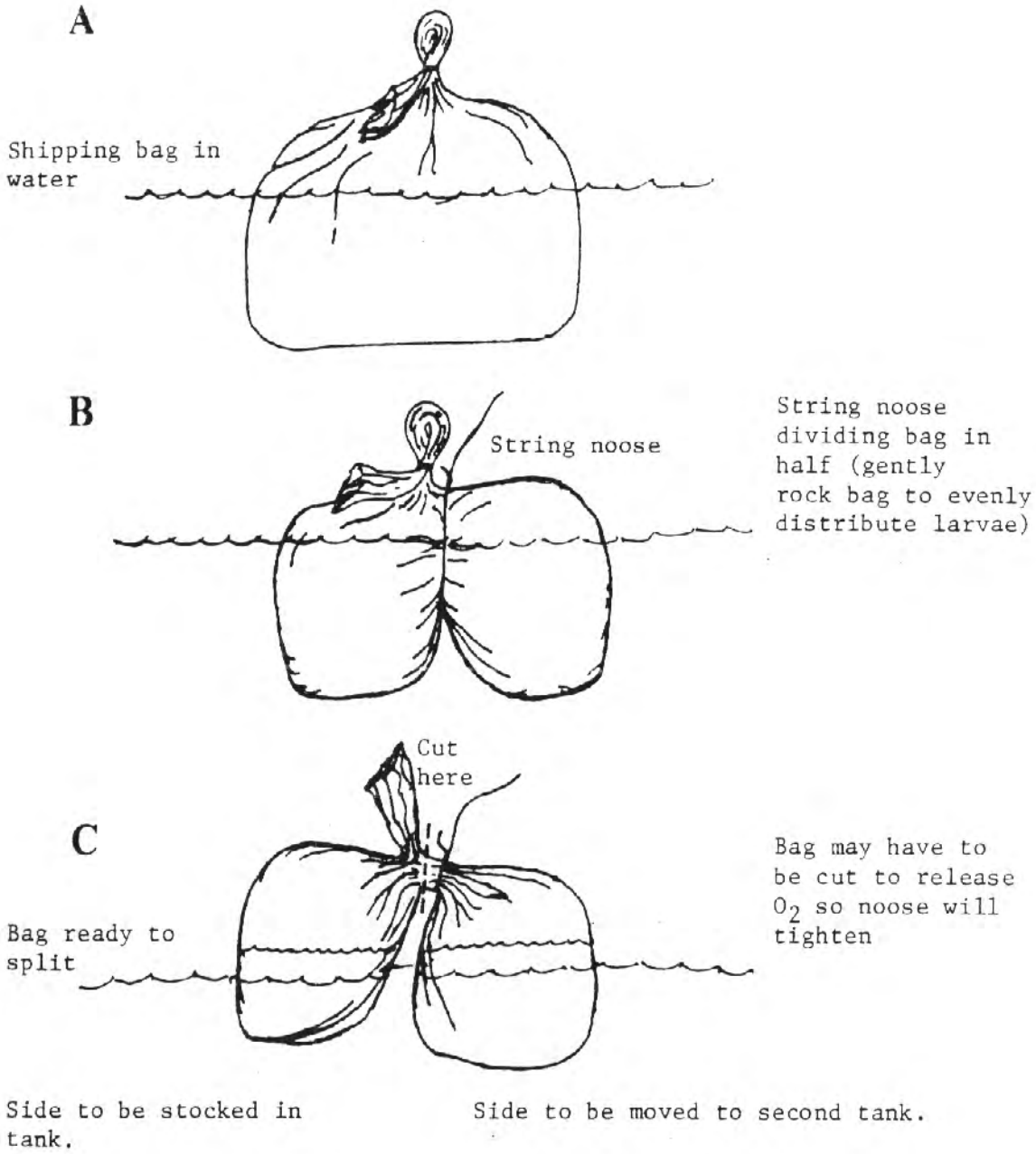


Figure 12. Dividing a shipping bag of striped bass larvae with a string noose. A. Shipping bag in rearing tank. B. String noose placed over the vertical center of the bag. C. Tightened noose.

We have experimentally stocked larvae into the upflow tanks at rates of 100 to 200 larvae per liter. On the basis of this experience, we recommend stocking the larvae into the upflow tanks at a density of 100 larvae per liter.

The shipping cartons containing the striped bass fry should be opened in subdued light. McHugh and Heidinger (1978) found that sudden exposure to bright light can result in mortality of striped bass larvae.

When the shipping bags are opened, oxygen and the carbon dioxide that has built up will be released. The sudden loss of carbon dioxide will increase the pH, converting the ammonia to the toxic non-ionized form. Transfer of the fry to the rearing tanks should therefore be rapid.

Since the striped bass fry will not feed until they are 4.5 to 5 days old, flush rates in the upflow rearing tanks may be kept to a minimum without risk of lowering water quality. Flush rates of 7.6 liters per minute (2 gal/min) should maintain water quality for the first 6 or 7 days after stocking. In the 550-liter upflow rearing tanks this flush produces on upward flow of 1.1 cm/min. This flush rate is sufficient to produce a water velocity of 15.9 cm/sec in the orifices of the upflow tank diffuser plate and thus prevent the fry from entering the 3.2-mm holes in the diffuser plate.

Care must be taken to maintain an oxygen level of at least 6.5 ppm and to avoid the accumulation of waste material in the upflow tanks. The feeding of brine shrimp nauplii will result in an accumulation of bottom sediment and some associated fungal growth. After 3 to 5 days of feeding it is necessary to begin cleaning the bottom of the upflow tanks by siphoning. Siphoning the bottoms and brushing the screens must

be done daily, or in some cases, twice daily as feeding rates increase. In addition, every one or two days it is necessary to lower the overflow to drain off the surface film. A flashlight is useful to monitor progress while siphoning. The light produced by the flashlight also appears to help repel the striped bass fry from the area being cleaned. However, the larvae should not be exposed to any bright light source for the first week after hatching. Braid (1977) found that striped bass showed the greatest attraction to a light with an intensity of 80.7 Lux (7.5 foot-candles), whereas lesser attraction occurred to light sources with lower and higher intensities. Attraction to reflective surfaces was also noted. The attraction to reflective surfaces may contribute to the mortality which occurs when striped bass larvae less than 6 days old are stocked into circular fiberglass tanks.

Feeding the Larvae

As of this time, artificial feed cannot be substituted for brine shrimp as the first food for the larvae. Braid (1977) and Carraeon (1978) consistently obtained greater survival of striped bass larvae fed brine shrimp nauplii than in fish fed dry diets in experiments lasting 10 to 25 days. Bowman (1979) compared survival and growth of striped bass larvae fed eight different feeds. Survival of striped bass fed Skretting Salmon Starter or Tetra Min Baby Fish Food "E"® were comparable with or even slightly higher than the survival of larvae fed brine shrimp nauplii. However, at the end of his 15-day

experiments, larvae fed brine shrimp nauplii demonstrated significantly greater growth than fish fed other diets.

Rotifers, as an alternative to live brine shrimp nauplii, were examined by Al-Ahmad (1978). Striped bass larvae accepted the rotifers well and commenced to feed upon them. The maximum density of rotifers obtained in the medium used to culture them was 32 per ml. An initial density of 20 rotifers per ml was required to approach satiation of the fish larvae after one hour of feeding. Thus it appears that the space required for rearing the food supply would exceed the space required to rear the fish. Using the methods of rearing brine shrimp described previously, densities of 1400 to 2500 nauplii per ml are obtained.

Once the striped bass larvae reach 6 days of age and water flow patterns no longer produce major mortalities, the quantity of brine shrimp supplied to the larvae is the single most critical factor in their survival.

The number of striped bass which survive to stockable size is directly related to the quantity of brine shrimp cysts used to feed the larvae. Using the data from Lewis et al. (1977a) and from three years of production data from the Illinois Striped Bass Project (Lewis et al. 1978, 1979, 1980) a predictive equation was developed to estimate the number of striped bass produced from a given weight of brine shrimp cysts. The number of striped bass fingerlings which would be produced from a given quantity of brine shrimp cysts is estimated by:

$$F = 8974 + 1.5279\ (S)$$

where F is the number of striped bass and S is the quantity of brine shrimp cysts in grams. The reverse equation which can be used to

estimate the quantity of brine shrimp cysts required to produce a given number of striped bass is:

$$S = -2643.9 + 0.6073\ (F)$$

with F and S representing the same parameters as in the preceding equation. The R^2 of this equation is 0.9279 which produces a 95 percent confidence interval of $\pm$ 12067 grams of brine shrimp cysts (Figure 13).

Striped bass larvae begin to feed on live brine shrimp nauplii when they are 4.5 to 5 days old, and the brine shrimp feeders must be filled at 6-hour intervals to provide sufficient quantities of nauplii to the fish. Of course, it is necessary to commence incubation of brine shrimp cysts 72 hours (incubation period for the cysts) in advance of the anticipated need for nauplii and to commence subsequent batches that will be available to maintain brine shrimp in the feeders. The methods and the facility for hatching brine shrimp cysts have been described earlier.

Brine shrimp nauplii are automatically supplied to the fish at hourly intervals, 24 hours per day. We use a 24 hour-per-day feeding schedule combined with 24 hour illumination. However, striped bass will both initiate feeding and continue to feed in total darkness, although their success at capturing the brine shrimp nauplii is lower than in light (McHugh and Heidinger 1977; Lewis et al. 1977b). Bonn et al. (1976) recommend supplying brine shrimp continuously. However, when using an hourly feeding schedule in the upflow rearing units brine shrimp nauplii are continuously available. The density of brine shrimp in the rearing tanks should be high. During the initial days of feeding the limited swimming ability of the striped bass larvae

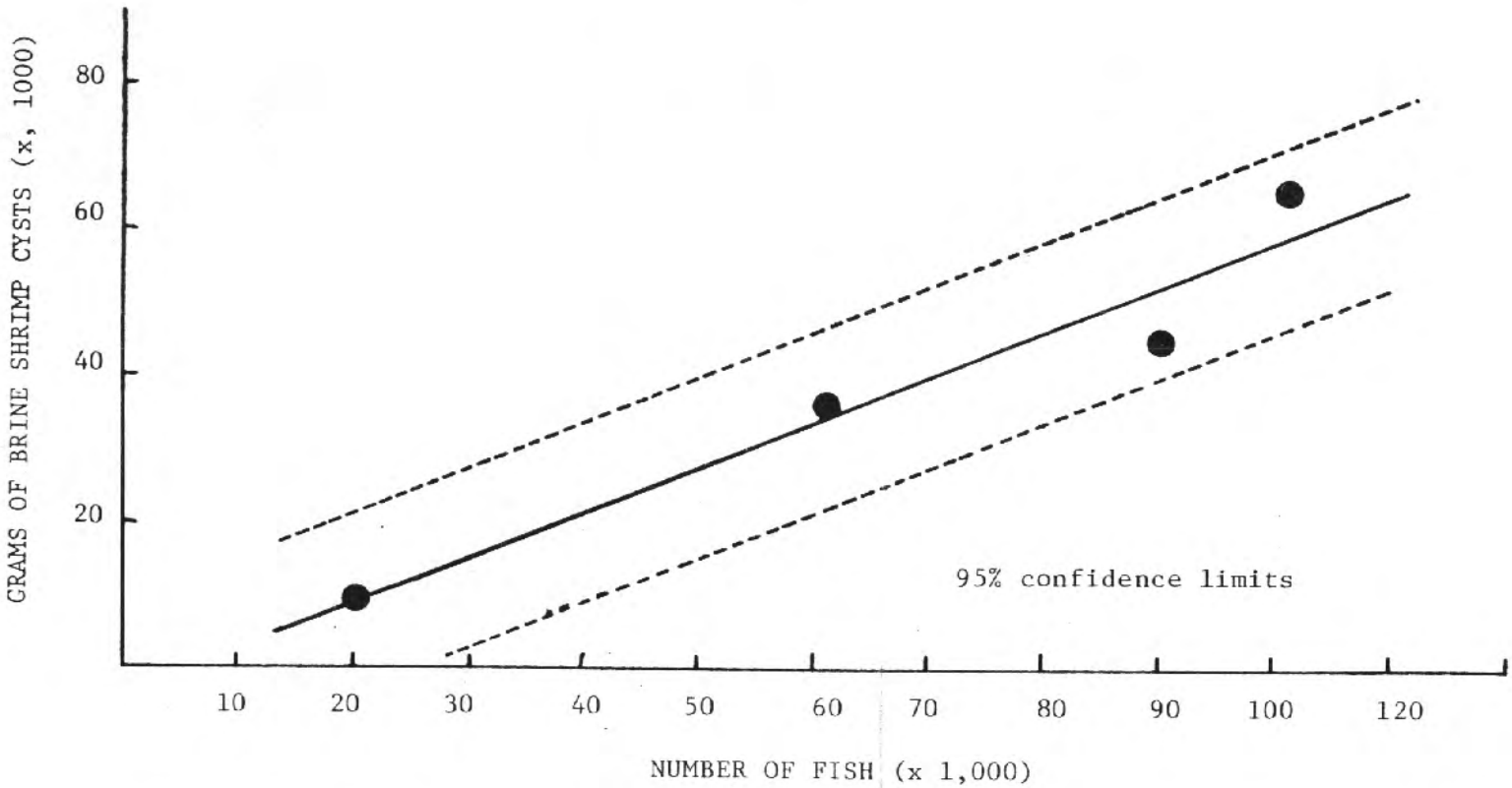


Figure 13. Relationship between the quality of brine shrimp and the number of striped bass fingerlings produced. (Note: The number of fingerlings produced is more closely correlated with the quantity efficiency of brine shrimp fed than with initial stocking rates).

prevents them from actively seeking out the nauplii. Later in the rearing period a high density of nauplii is needed to discourage cannibalism. The brine shrimp nauplii should be a more available prey than other striped bass larvae. Brine shrimp cysts are expensive ($50.35/kg in 1980) and large quantities are needed. Although this price amounts to only $0.04 per fingerling, large quantities of cysts are often difficult to obtain. Therefore, arrangements to obtain the brine shrimp cysts should be made well in advance (one year is not unreasonable). Upon receipt of the cysts, tests on their hatchability should be made. Hatch rates above 80 percent are considered excellent, while rates of 60 to 70 percent should be expected.

Live brine shrimp nauplii should be supplied to the striped bass fry for 25 days. This time span permits a prolonged period for training the fish to accept commercial foods and provides the fish with an added nutrient supplement during the sensitive period when they are transferred to the circular tanks. The length of time which striped bass require brine shrimp has not been determined. Lewis et al. (1977a) found 13 to 14 mm striped bass begin to accept finely ground dry feed when it is fed in conjunction with brine shrimp. However, Lewis et al. (1978) concluded that brine shrimp should be included in the diet of striped bass until they reach 19 mm in total length in order to assure high survival and uniform growth.

While the quantity of brine shrimp nauplii fed to the larvae is based in part on maintaining a minimum density of nauplii, the minimum quantity fed to the fry after 10 days is based on the ration required by the fish, inasmuch as they are capable of seeking out the nauplii

and consume essentially all of the nauplii made available.

The age at which striped bass larvae begin to actively seek out brine shrimp nauplii, and the optimum densities of nauplii in the rearing tanks have yet to be determined. However, based on the data of Al-Ahmad (1978), 12-day-old larvae can actively seek out and capture prey. At initial densities of 10 and 20 per ml, the numbers of rotifers found in the stomachs of seven-day-old striped bass larvae were significantly greater than when the prey were supplied at an initial density of 5 per ml. However, at 12 days of age, the densities of prey in the stomachs were similar for all densities suggesting that at the lower density the larvae were able to seek out the prey.

The time required for complete egestion of brine shrimp nauplii was determined to be 9 hours at 25 C and 11 to 12 hours at 20 C (McHugh 1975). As a minimum, each striped bass larva must thus receive sufficient nauplii to fill its digestive tract every 9 hours. However, other factors also determine the quantity of brine shrimp nauplii required. Brine shrimp can be lost through escapement through the screens and through natural death. Also some of the nauplii can settle to the bottom of the rearing unit and become unavailable to the striped bass larvae. The upflow rearing tanks reduce the settling effect.

At SIUC, nauplii are supplied to the fish at a rate of 50-60 nauplii per ml of rearing tank volume per day from age 5 through 15 days. After the fish reach 15 days, the density of nauplii is increased to 100 to 120 per ml per day in the upflow tanks.

In the circular fiberglass tanks, the density of nauplii is much less. However, at a length of 15 mm, the fish will feed upon the

nauplii which have settled to the bottom.

When the shrimp have been introduced into the tank, the fish will aggregate around the cloud of nauplii and follow it as it settles to the tank bottom.

Training the Fry to Accept a Dry Diet

Training the fry to accept artificial feed should be started when the fish are approximately 12 days old. This training overlaps by 18 days the feeding of brine shrimp. At this time it appears desirable to transfer the fry from the upflow to the circular flow units when they are about 12 days old. Hence feeding dry feed in combination with brine shrimp begins immediately after transfer of the fish to the circular flow tanks.

In the SIUC pilot rearing facility, Tetra® commercial flake feed is used as the training diet. This feed has several desirable charactistics. A major constituent of the feed is invertebrates, giving it an odor and color similar to the brine shrimp nauplii. Also, the low oil content of the flakes permits them to be finely ground. Other training diets have been tested, including salmon starters (Lewis et al. 1977), and special mixtures of trout feed, pasteurized fish and vitamins (McIlwain 1976). Bonn et al. (1976) also mentioned the use of semi-moist feeds.

The striped bass fry will commence to eat dry feed earlier if the particle size is small. Bonn et al. (1976) recommend a particle size similar to the size of the brine shrimp nauplii (approximately 0.7 mm). However, a smaller particle size (0.2 to 0.5 mm) is more acceptable to striped bass larvae (Lewis et al. 1977). The fry will attack larger particles, but will often reject them. Their acceptance of a smaller

particle may be due to the coarse texture of a dry feed. The Tetra® should, therefore, be ground to a flow consistency. We have found a ball-mill satisfactory for pulverizing the feed.

After the flake feed has been given to the striped bass for five days, a high protein salmon starter is added to the feed. The mixture is prepared by pulverizing together equal parts of the Tetra® and salmon starter. After the striped bass begin to eat the dry feed they are offered a series of salmon feeds, with periods of overlap when changing from one feed to another. The following is the feeding schedule we have used:

Age of the fish[1] (days)	Food type[2]
5-11	live brine shrimp nauplii
12-17	pulverized Tetra® and brine shrimp nauplii
18-22	pulverized Tetra®, pulverized starter, and live brine shrimp nauplii
23-30	pulverized starter, starter, and live brine shrimp nauplii
31-35	starter
36-40	starter and 2/64 salmon
41-45	2/64 salmon
46-50	2/64 salmon and 3/64 salmon
51-60	3/64 salmon
61-75	3/64 salmon and 4/64 salmon
76-	4/64 salmon

The first three dry feeds are fed at frequent intervals (12 to 16 times per day). During this period the fry are also receiving brine shrimp nauplii. Bonn et al. (1976) also recommends presenting the fry

[1]When reared at 23 to 25° C.
[2]Combination diets mixed in equal proportions.

with the training diet in conjunction with the brine shrimp nauplii. A small quantity of the ground Tetra® can be added to the brine shrimp feeders each time they are refilled. We have added 10 ml (4 grams) to the 4-liter feeders with little problem, whereas 25 ml would tend to clog the solenoid valves. The striped bass fry are fed at an approximate rate of 25 to 50% of body weight per day during training. At a total of 16 to 18 mm the fry should be actively eating the dry feed.

Once the fish are taking the unpulverized salmon starter the quantity of feed is held constant until, as a result of growth of the fish, it becomes approximately 15% of the body weight per day. The quantities of 2/64 salmon starter mixture are again held constant until the rate approaches 5 to 10% of the body weight per day. Due to the rapid increases in weight of the fish, the feeding levels should be adjusted every 3 to 4 days.

When the fish are approximately 40 to 45 days old, a feeding schedule based on an average of 5% of body weight per day is used. The quantity of feed actually given the fish is dependent upon the temperature of the rearing system water (Figure 14). At 20 C the fish are given 3.2 percent of their body weight in food per day, while at 30 C the fish receive 6.4 percent per day. The ration is divided among 10 feedings throughout a 24 hour period.

Greater food utilization will result from a feeding frequency of 10 feedings per day than from 6 feedings per day (Lewis et al. 1978). The feed should be gradually spread over the tank to insure even utilization. This necessitates hand feeding, or an automatic feeder with a very slow delivery rate such as the auger feeder described by

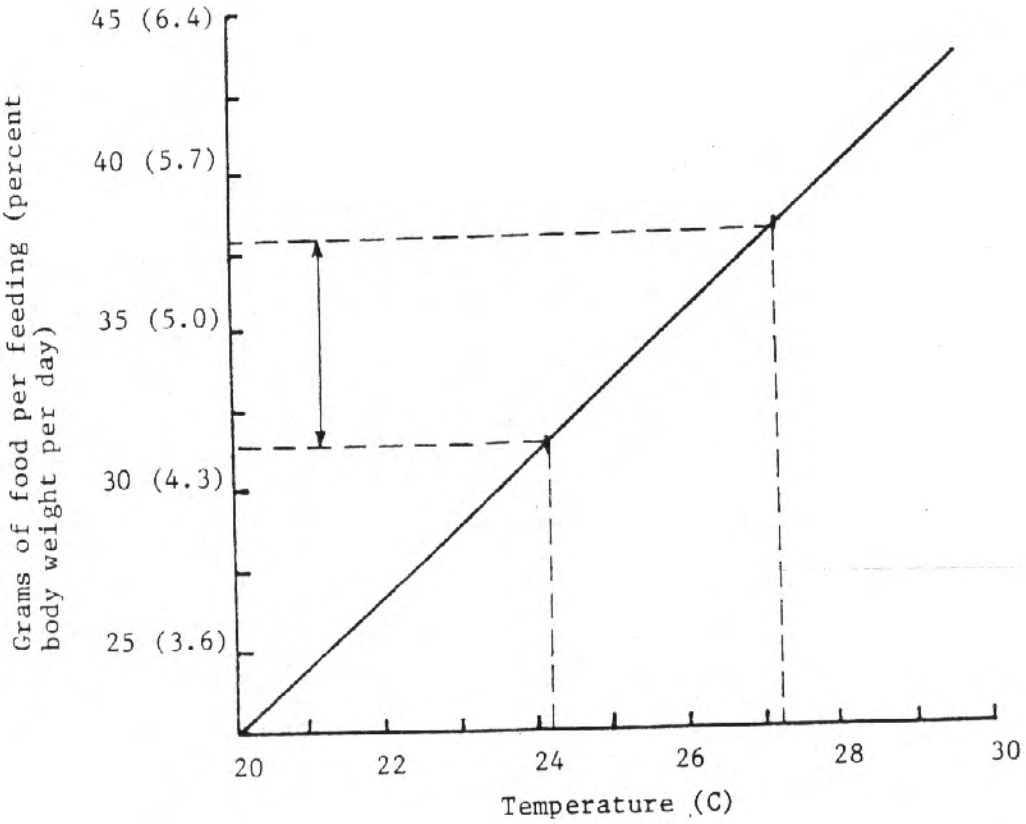


Figure 14. Feeding schedule for each tank of 3500 2-g striped bass fingerlings with 10 feedings per day, and daily feeding rate based on the temperature of the rearing tank water and an average rate of 5 percent of body weight per day.

Wehr and Lewis (1974).

When hand feeding, the fish are actually feed 20 times per day. A half ration is given to each tank of fish, then repeated. We feel that this procedure may permit the more aggressive fish to satiate themselves during the first feeding, thus giving the less aggressive fish the opportunity of obtaining food during the second feeding. Generally, the more aggressive fish will first feed on food near the surface, then once the surface supply is depleted, they feed off the tank bottom.

Transfer of Larvae to Circular Tanks

While the fry will survive in the upflow tanks beyond 12 days, once the use of dry feed commences it is very difficult to keep the upflow tanks free of sediment and prevent the screens from becoming clogged. Further, there is some indication that maintaining the fish beyond 12 days in the upflow tanks is unfavorable to gas bladder inflation.

Transferring the fish presents several problems. Striped bass fry at this age are 8 to 12 mm in total length and are capable of swimming through the 3.2 mm diameter holes in the diffuser plate. This necessitates continuing the flush while capturing the fry. Striped bass fry at this age also may be subject to handling shock. Removing 11-day-old striped bass from the upflow tanks with dip nets can result in considerable mortality (Lewis et al. 1977a). Nevertheless, McHugh and Heidinger (1978) reported mortalities of only 2 to 2.5% when handling 5- to 21-day-old striped bass fry. Utilization of a method of capturing, transferring, and grading the fish without removing them from the water would reduce the possibility of handling mortality.

Several methods of transferring striped bass without removing them from the water have been attempted. The use of a siphon was unsuccessful, as was an attempt to allow the fish to escape through the overflow once the retaining screens were removed (Lewis et al. 1978). Currently, a small bag seine is used at the SIUC pilot facility (Figure 15). The seine measures 100 cm on a side (slightly larger than the 91-cm size of the rearing tank), and funnels back to an open bag 25 cm in diameter and 50 cm deep. The seine should be constructed of a smooth material. With a coarse material, some striped bass larvae will adhere to the sides of the seine during harvest, and thus be removed from the water. These fish frequently do not survive. To capture the fry, the open end of the bag is closed and the tank is seined. Once the fry are concentrated in the bag portion of the seine, the seine is lifted from the water until only the bag containing the fry is left submerged. A large, water-filled bucket is brought up around the bag and the fish are released into the bucket. This method has been successful for 12-to 25-day-old fry (Lewis et al. 1978, 1979) but is slow. Three people are needed for this procedure (one seiner, one to handle the bucket, and one to transfer the fry). Approximately one hour is required to harvest each upflow tank. The fry should be lowered into the receiving tank rather than poured. Due to the higher activity level of the fish at higher water temperatures, the system temperature should be lowered to 19 to 21 C prior to all harvests or gradings.

Stocking rates for the circular tanks depend upon the flush capability of the system and the availability of supplemental oxygenation in the rearing tanks. With flushing capabilities of 3 to 4 turnovers per hour in the rearing tanks, each circular tank will support 7,500

Figure 15. Harvesting of upflow rearing tanks with bag seine.

to 10,000 fingerlings. With turnover rates of 1.5 per hour, we have maintained 7,500 6-cm fingerlings per 1730-liter tanks. However, stopping the flush to four tanks each holding 10 to 12 thousand 4.5-cm fingerlings resulted in heavy mortalities in two of the fish tanks after 3 hours (Lewis et al. 1980). The use of artificial means to reoxygenate water in the rearing tanks should raise the carrying capacity of the tanks. Methods of oxygenation may include aeration or the addition of pure oxygen. The use of surface agitators should not be used with fingerlings less than 5 to 6 cm long. With the use of oxygenation, the carrying capacity of the rearing tanks could be increased several times. (See section on carrying capacity, page 52).

Flush rates in the circular tanks at the time the fry are stocked should be the minimum required to maintain water quality. We used a flush rate of 11 to 19 liters per minute. The flush should be gradually increased as the striped bass grow and their swimming capability increases. When the fish are first transferred into the circular flow tanks the circular flow of water should be held to a minimum. After the fish are 3 to 4 weeks old the circular flow can be gradually increased by altering the angle at which the water enters the tank.

Daily, or as required, the bottoms of the circular flow tanks must be siphoned to remove accumulated sediment. Siphoning is facilitated by the use of a swimming pool type vacuum head on a 3/4" garden hose. In addition to the tank bottom, the area between the standpipe and the retaining screen should also be siphoned daily and occasionally the sides of the tank should be cleaned. As needed the screens must also be cleaned by brushing. This is needed about once every 3 days when the fish are initially stocked, increasing to 3 to 4 times

daily until the screen mesh size can be increased.

Growth, Production, and Carrying Capacity

Larval striped bass measure 4.5 to 5.5 mm at one day of age. During the period of yolk-sac absorption and during the first few days of feeding, increases in length are minimal. Bowman (1979) obtained increases in length of 2.0 to 7.3 mm when rearing striped bass from age 5 days to age 20 days on a live brine shrimp diet. After feeding on a diet of live brine shrimp and dry food from an age of 4.5 to 27 days, striped bass attained average lengths of 14.8 to 25.0 mm (Lewis et al. 1977a). These data are presented in Figure 16. The large variability in length shown in this figure was attributable to the various feeding rates for the live brine shrimp nauplii.

Striped bass larvae begin to accept dry feed when they attain a length of 13 to 14 mm and will readily accept the dry diet when 18 mm in length (Lewis et al. 1977a). As shown in Figure 16, this length of 18 mm corresponds to the point where growth in length becomes exponential. Growth rates exceeding 2 mm per day can be obtained after this period (approximately 25 to 30 days of age). The growth rate of striped bass based on these data can be expressed by the equation:

$$\ln(TL) = 1.240 + 1.289 \ln(A)$$

where TL is the total length of the fish in mm and A represents the age in days. Using this equation, the time required to produce various sizes of fish can be estimated (with the understanding that the equation is based on fish reared at 25 to 27 C). To produce 51-mm fingerlings (2 inches), approximately 55 days are required,

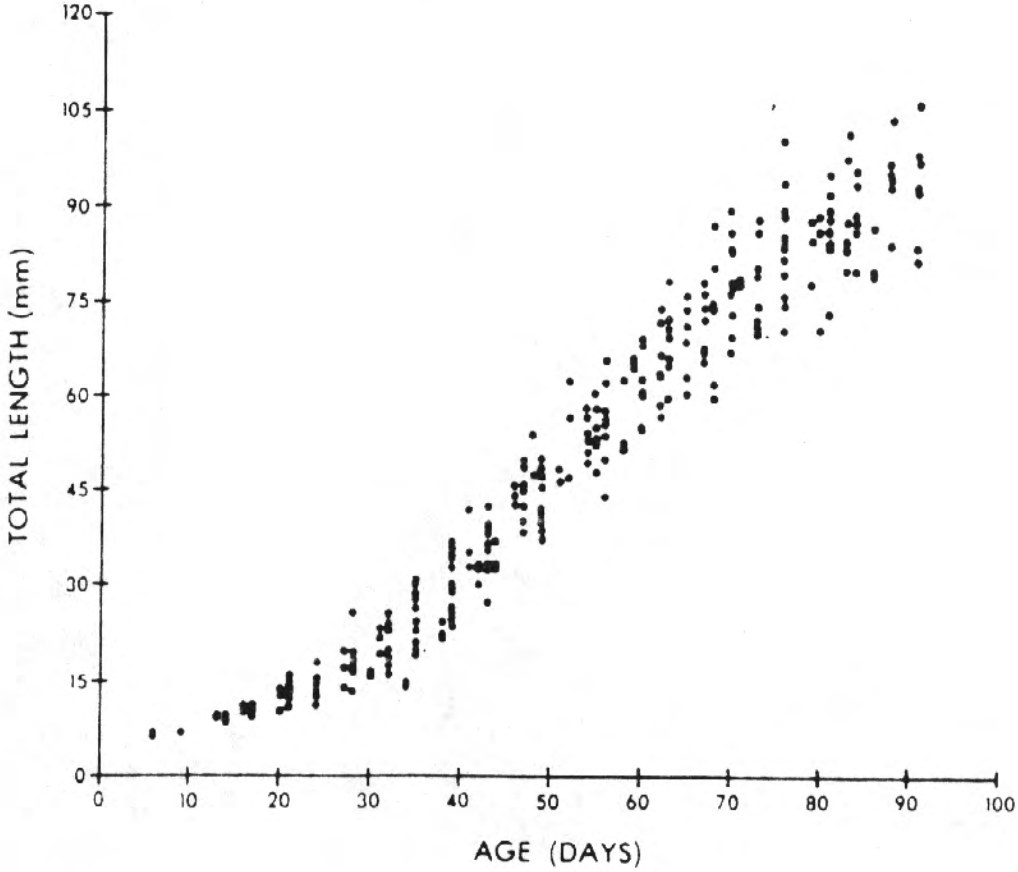


Figure 16. Growth of tank-reared striped bass fingerlings. (Data from Lewis et al. 1977a).

whereas 95 days are required to produce 120 mm (4 inch) fish.

Although increase in length is relatively slight when the striped bass are less than 20 days old, their weight increase is rapid. Between 5 and 10 days of age, we calculate that these fish will double their weight 6.5 times. Therefore, assuming a food conversion of 5:1, the larvae must ingest 340 percent of their body weight in brine shrimp nauplii daily.

The relationship between length and weight for larval and fingerling striped bass is shown in Figure 17. This relationship can be described by the equation:

$$W = (5.77 \times 10^{-6}) \cdot (L^{3.180})$$

where W represents weight in grams and L is total length in mm. Based on the coefficient of 3.18, it can be seen that striped bass exhibit near isometric growth over this range of lengths.

For ease of calculation, a single regression equation was calculated on the data in Figure 17. However, as this figure shows, two separate regression lines could be drawn, one of which would include fish with total lengths less than 30 mm and one for fish 18 mm and longer. The intersection of these lines at an approximate fish length of 25 mm corresponds to the point at which the striped bass undergoes metamorphosis from postlarvae to fingerling.

Knowledge of the weight of the fish is useful in determining feeding rates. Fish weight is also the principal parameter required when estimating the carrying capacity of the rearing system.

In order to estimate the carrying capacity of the rearing tanks, the tolerance levels of striped bass to low oxygen levels and ammonia must be known.

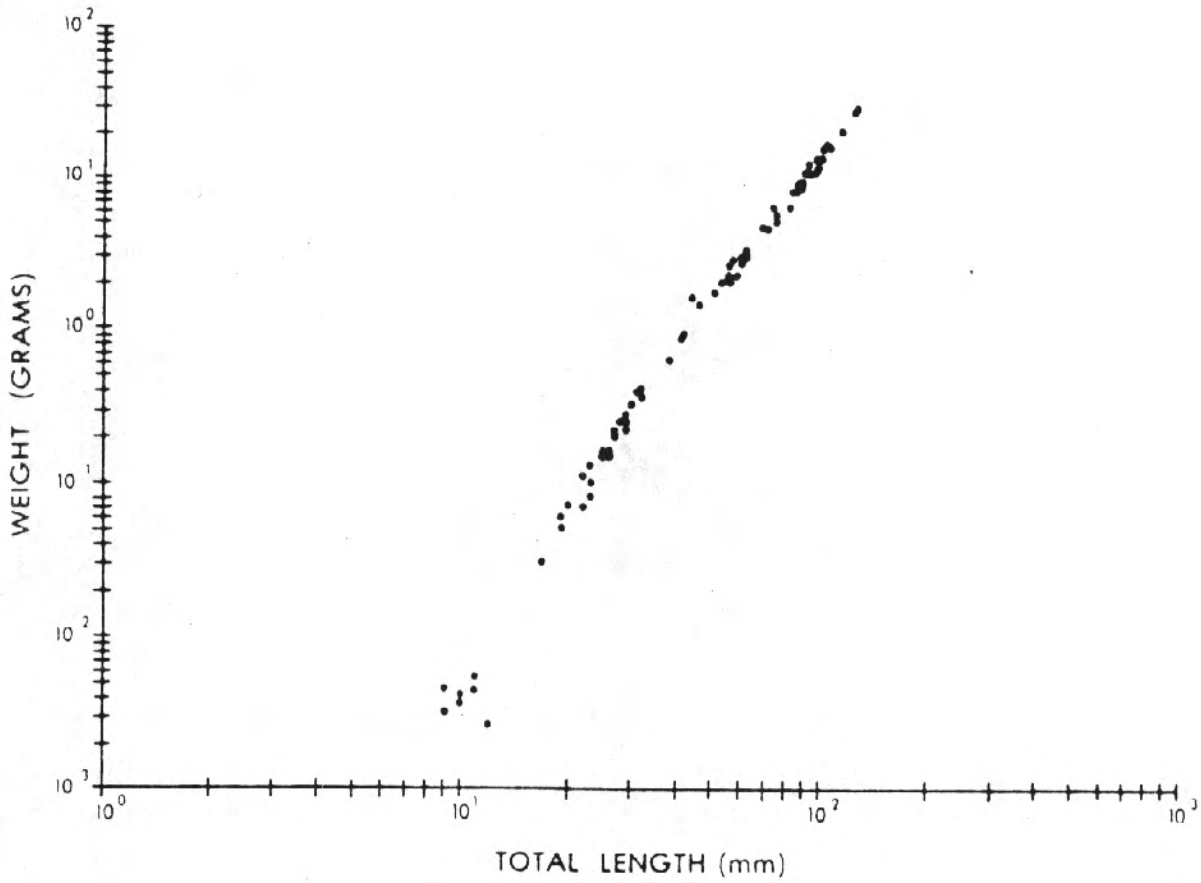


Figure 17. Length-weight relationship of striped bass with total lengths of 9 to 125 mm. (Data pooled over three years combining Hudson River and South Carolina strains).

With striped bass larvae less than 20 days old, oxygen levels should not fall below 6.5 mg/l. Older fish can tolerate levels as low as 4.8 to 5.0 mg/l with no apparent effects. At this level, fingerlings continue to feed extremely well. Oxygen consumption of 1- to 22-gram fingerlings has been determined by Klyashtorin and Yarzhombek (1972). They present an equation for determining oxygen consumption based on weight of fish reared at 20-22 C. This equation can be expressed as:

$$C = 640\ W^{-9.25}$$

where C represents consumption in mg oxygen per kg of fish per hour and W is the average weight of the fish in grams.

Klyashtorin and Yarzhombek (1972) also examined the effects of temperature on the oxygen consumption of 1- to 3-g fingerlings. Their results can be expressed in the equation:

$$C = 121.36 \exp (0.07117T)$$

where C is again oxygen consumption, T represents temperature (C), and exp means raise the natural base(e) to the given power.
Although these authors did not examine the effects of temperature on various sizes of striped bass fingerlings, we combined the two equations, giving:

$$C = 144.32W^{-0.25} \exp (0.07114T)$$

based on the data presented by Klyashtorin and Yarshombek (1970) this equation will closely approximate the oxygen consumption of fingerlings ranging in weight from 1 to 10 g at temperatures of 10 to 30 C.

We therefore have the minimum oxygen tolerance level of the fingerlings (5 mg/1) and the oxygen consumption rate of the fingerlings. Thus, to estimate the flow of water to supply oxygen to fingerlings at 25 C, we first determine the consumption rates (converted to minutes).

1 g fish:	854.5 mg/kg/hr =	14.24 mg/kg/min
5 g fish:	571.4 mg/kg/hr =	9.52 mg/kg/min
10 g fish:	480.5 mg/kg/hr =	8.01 mg/kg/min

Then, assuming the water entering the rearing tanks is saturated with oxygen (8.4 mg/1 at 25 C at sea level), we can determine the relative available amount of oxygen: 8.4 mg/1 - 5 mg/1 tolerance = 3.4 mg/1 available. To determine flow required, divide the available oxygen into the amount of oxygen required, giving:

1 g fish:	4.2 liter/min/kg fish
5 g fish:	2.8 liter/min/kg fish
10 g fish:	2.4 liter/min/kg fish

It should be noted that these are rough estimates and do not take into account numerous other factors. Oxygen consumption at various feeding rates has not been determined for striped bass. Also no allowance was made in the calculation to provide for the oxygen consumption resulting from the reuse of the water (BOD, COD, biological growth on the tank sides). Therefore these values should be considered as minimal.

Determination of the flush rate required to remove ammonia is calculated in a similar fashion. Hazel et al. (1971) determined the 96-hour LC_{50} for striped bass to be 1.9 mg/1 NH_4OH (0.76 mg/1 as NH_4OH-N) at 23 C in fresh water. We assume that the maximum safe

level needed to prevent sub-lethal effects is 10 percent of the lethal level. Therefore by knowing the pH of the rearing water, the safe total ammonia level can be determined using the calculations of Trussell (1972). At 25 C and a pH of 8.0, 0.076 mg/1 NH_4OH-N (one-tenth the lethal) would result from a total ammonia-nitrogen concentration of 1.35 mg/1.

The ammonia production rates for striped bass fingerlings have yet to be determined. Based on data from our rearing system, we estimate the values to be approximately 0.0270 kg NH_3-N/kg feed/day. Feeding rates at our facility are approximately 10 percent of body weight for 1 g fingerlings and 5 percent for fingerlings 2 g and larger. Thus converting ammonia production to a "per fish-weight" basis.

1 g fish: 0.0270 kg/kg/day x 0.10 = 0.0027 kg/kg fish/day
2g+ fish: 0.0270 kg/kg/day x 0.05 = 0.00135 kg/kg fish/day

which is equivalent to:

1 g fish: 1.875 mg NH_3-N/kg fish/min
2g+ fish: 0.938 mg NH_3-N/kg fish/min

The quantity of ammonia-free water required to remove these levels is found by dividing the above by the maximum allowable concentration (1.35 mg/1) giving:

1 g fish: 1.39 1/min/kg fish
2g+ fish: 0.69 1/min/kg fish

If the input water contains ammonia, the allowable increase in ammonia would be determined by subtracting the input concentration from the maximum allowable level.

As can be noted by comparing the flushes required for oxygen maintenance and ammonia removal, oxygen is the first limiting factor

in the rearing tanks. Thus by adding oxygen to the rearing tanks, the flow index (kg fish/lpm) can be increased by a factor of 3.

Mortality and Diseases

With the use of the upflow rearing tanks to eliminate the mortality which occurs in the circular fiberglass tanks, the major loss of striped bass fry is attributed to problems related to feeding. Mortality from deteriorated water quality can also result in considerable losses. Tolerance of striped bass to various water quality parameters can be found in the section on water quality.

During several years noticeable mortality of 30 to 40 mm fingerlings has been observed (Lewis et al. 1977a, 1978). The expected cause of this mortality is nutrition, although no data are available to support this conclusion. The fingerling fish "grow" into this mortality with rearing tanks containing the larger fish experiencing the mortality first. During this period 2 to 5% of the fingerlings have been lost. However, in most years this mortality has not been observed, suggesting that the cause was possibly a vitamin difficiency in one batch of food.

Feeding and Cannibalism

Cannibalism can result in the loss of 70 to 80% of the striped bass larvae. The incidence of cannibalism has been greatly reduced by supplying the larvae with adequate amounts of acceptable food. Reducing the food supply for 3 days has initiated problems with cannibalism. Cannibalism, once begun, will continue even after adequate amounts of food are available.

The mortality of fingerlings which fail to convert to the artificial diet begins three days after brine shrimp nauplii are eliminated from the diet and continues for 5 to 7 days. During this period we

expect to lose 2 to 5 percent of the fingerlings.

As stated in the section on feeding, severe mortality can result from the use of a dry food having a particle size which is too large. A 4 percent mortality of 5-cm fingerlings was attributed to an increase in pellet size of 0.4 mm and a decrease in crude protein content from 52% to 46% (Lewis et al. 1978).

Diseases and Parasites

Fish reared intensively are more vulnerable to diseases and parasites, since they are confined in crowded conditions. However, in a closed system the fish are also isolated from sources of infection and outbreaks of disease can be quickly observed. Isolation and precautions to insure this isolation have reduced the outbreaks of parasites or diseases during the five years of rearing striped bass at the SIUC facility.

The only pathogens which have been encountered were a *Trichodina* infection which was eliminated without loss of fish and an outbreak of *Aeromonas* in two tanks which contained fish which suffered mechanical damage during grading. The *Trichodina* was eliminated by treating the system with 35 mg/l formalin. The use of a moist pelleted feed to which 0.3 percent nitrofurazone (as active ingredient) had been added eliminated the *Aeromonas* infection (Lewis et al. 1979).

The major precautions needed to maintain a disease-free environment in the rearing facility include the maintenance of separate equipment for use in the facility, and avoiding the introduction of other fish. Dipnets, pumps, buckets, etc., which are used in the rearing facility should not be used for any other purpose. Other equipment,

such as fish graders and weighing scales, should be sterilized with Roccal® or 500 ppm chlorine prior to their use in the facility.

To further isolate the striped bass fry and fingerlings from disease, well water is preferred over surface water as a source of makeup water in the system. Surface water can, however, be used if properly treated before being introduced into the system. Burrows and Combs (1968) recommend the use of pressure-sand filters in combination with ultraviolet sterilization for the treatment of surface water. Pressure sand filtration will, according to these authors, remove particles larger than 15 microns. To eliminate pathogens smaller than 15 microns, Burrows and Combs recommend UV sterilization. They state that UV sterilization will destroy organisms less than 30 microns in size.

When fish are reared intensively in tanks not supplied from surface waters, the incidence of metazoan infections (monogenetic trematodes such as Gyrodactylus or copepods such as Lernaea) are greatly reduced. However, the potential for outbreaks of bacterial and protozoan infections still exist.

Striped bass are susceptible to all of the diseases and parasites common to cultured fish. While striped bass fingerlings exhibit reasonable tolerance to the therapeutic chemicals used to treat these pathogens, the fry are often more sensitve to the dosages effective for most pathogens. Also, several of the chemicals which are not toxic to the fish may severely affect the nitrifying bacteria in the biofilters. Collins et al. (1975) examined the effects which several chemicals used in fish culture have on nitrification. He found no effects on nitrification in systems treated with formalin

at 25 mg/l and malachite green at 0.10 mg/l. Several of the antibiotics and bacteriostats can be expected to affect the filter bacteria, although no evidence is available. However, incorporating these chemicals into the feed should not produce concentrations sufficiently high to affect the filters.

Some of the chemicals used to treat common diseases and parasites of warm-water fish are found in Table 1. Diagnosis and identification of specific infections are beyond the scope of this manual. For additional information on specific infections, the numerous texts on parasites, fish diseases, and fish pathology can be consulted. Malachite green (zinc free) has been successfully used in recirculated systems. Burrows and Combs (1968) have used this chemical at a concentration of 1 mg/liter for 1 hour to control heterotrophic bacteria and algae in their biofilters. The 96-h LC_{50} for malachite green on striped bass fingerlings has been shown to be 0.2 mg/liter in both static (Hughes 1969) and flow through (Lewis et al. 1978) bioassays.

In 1979, the striped bass rearing system was treated with 35 mg/l formalin to remove a Trichodina infection. Twelve hours after the treatment, the ammonia-nitrogen levels in the system had risen from 0.25 to 1.20 mg/l. However, nitrification rates were unaffected, as ammonia levels were significantly reduced by the biofilters. We feel that the sudden increase in ammonia resulted from the decay of fungal and some bacterial growths which had been killed by the formalin. Interference with the ammonia determination was also expected. Konikoff (1973) showed that the Nesslerization technique of ammonia determination cannot be used when formalin is present in the water. However, later studies showed that no interference occurs with selective

Table 1. Common chemicals used for the treatment of diseases and parasites of warmwater fish and their toxicity to striped bass.

Disease & medication	Effective dose	24-hr LC_{50}	Approval [1]
Bacterial infections			
Furinace	0.1 mg/l indefinite	1.5 mg/l(Bonn et al. 1976)	non-foodfish
Furacin	5.0 mg/l active indefinite	10+ mg/l(Hughes 1973)	not approved
(Nitro furazone)	10-22 mg/l dip for 8 hrs	not applicable	
Oxyteratacycline (Terramycin)	1.8 mg/kg food for 10 days	150 mg/l(Hughes 1973)	food fish
Roccal	1.0 mg/l dip for 5 hrs	not applicable	not approved
Protozoan infections			
Formalin	15-35 mg/l indefinite	35 mg/l(Hughes 1969)	requirements completed
Malachite green	0.1-0.2 mg/l indefinite	0.2 mg/l(Hughes 1969)	not approved
Monogenetic trematodes			
Formalin	100-150 mg/l dip for 1 hr	not applicable	requirements completed
Masotin (Dylox)	0.25 mg/l indefinite	16-30 mg/l(Hughes 1971)	non-foodfish

[1] Schnick, Meyer, and Van Meter 1979.

ion probes, thus permitting accurate ammonia determination.

Masoten® has also been used successfully in a system involving recirculated water. Largemouth bass fingerlings were treated with 0.25 mg/liter to control Gyrodactylus in the SIUC facility without any apparent effect on the nitrifying bacteria (Alan Brandenburg, SIUC Fisheries Research Laboratory, personal communication). However, at the time of treatment the system was not heavily loaded with fish. Caution should therefore be exercised when using this chemical.

Water Quality

Inasmuch as the culture of striped bass in systems using recirculated water is in the developmental stage, optimal ranges for many of the water quality paramenters have yet to be determined. In a recirculating system the water quality requirements of the nitrifying bacteria must also be considered.

Dissolved Oxygen

In a system using recirculated water, falling levels of oxygen not only affects the fish directly but also affect function of the biofilters. When the dissolved oxygen of the biofilters concentration is dropping there is generally a simultaneous increase in ammonia and nitrite.

Striped bass fry require moderately high levels of oxygen (greater than 6 mg/l) during their early development. Fortunately, during this period the load on the culture system is very light, since the fry are small and quantity of food entering the system is low. The lower temperature (15 to 18 C) at which the striped bass fry are reared also raises the saturation level for oxygen in the water.

At fingerling size, striped bass are fairly tolerant of short-term low levels of dissolved oxygen. When the oxygen level was decreased at a rate of 1 mg/liter/hour, striped bass did not begin to die until the dissolved oxygen was 1.04 mg/liter, while a dissolved oxygen of 0.74 mg/liter was needed before 50% of the fish had died (Chittenden 1971). However, Chittenden reports that the fish began to show symptoms of stress at a dissolved oxygen level of 3.12 mg/liter. For sustained periods the level of oxygen should be maintained about 5 mg/liter (Bonn et al. 1976). A dissolved oxygen value of 5.0 to 4.5 mg/liter was determined to be critical for striped bass fingerlings reared at 22 C (Klyashtorin and Yarzombek 1972).

The level of dissolved oxygen can also affect the nitrifying bacteria. Meade (1974) determined from the formulas of various authors that 4.0 to 4.6 kg of oxygen is required to oxidize 1 kg of NH_3-N to NO_3-N. Therefore, if the ammonia (NH_3-N) concentration in the biofilter influents equals 1 mg/liter, the minimum level of oxygen required is 4.0 mg/liter.

Pure oxygen is now supplied to the submerged upflow biofilters at the SIUC striped bass rearing facility. In past years, oxygen levels in the biofilters frequently fell below the minimum level acceptable for nitrification. Therefore, in 1979, gaseous oxygen was added to the discharge from the culture tanks at a rate sufficient to maintain dissolved oxygen near saturation in the biofilter influents. The principal effect of this procedure was stabilization of ammonia concentrations in the rearing system.

Temperature

The optimum range of temperature for hatching striped bass eggs and rearing fry is 16 to 18 C. Dorshev (1970) obtained higher survival of fry to an age of 6 days at these temperatures than at the other temperatures tested. Total mortality of the fry occurred at low temperatures and high temperatures (8.5 to 10 C, and 26 to 27 C). Shannon (1970) also reports 100% mortality of striped bass fry hatched at 27 C.

Bonn et al. (1976) recommend maintaining the striped bass fry at temperatures of 14 to 21 C for the first 9 days after hatching, after which they can be acclimated to temperatures anywhere from 18 to 32 C. We maintain water temperatures at 15 to 18 C until the fry are 10 days old, then raise the temperature to 23 to 25 C at a rate of 1 to 2 C per day.

At the SIUC facility 5 to 7 cm striped bass fingerlings were exposed to temperatures of 31.5 C with no mortality (Lewis et al. 1979). Advanced fingerlings (300 to 400 g) have been maintained for 24 hours at a temperature of 32 C without mortality. Cox and Coutant (1980) have determined the temperature of no growth (the temperature at which the metabolic activity will equal the energy acquired from food) to be 33.5 C. Although the rearing temperatures of 25 C used at SIUC promote more rapid growth, Bonn et al. (1976) report that more efficient food conversion occurs at temperatures fo 18 to 19 C. However, Cox and Coutant (1980) found maximum food conversions of 100- to 286-gram striped bass to range from 24 to 26 C. In addition, these authors also found that striped bass fed to satiation once per day exhibited maximum growth rate (weight gain) and maximum food consumption at 25 C.

Higher water temperatures also promote more rapid oxidation of ammonia in the biofilter. Kawai et al. (1965) found that nitrification was most efficient at 30 C in freshwater systems, with lower rates occurring at both lower and higher temperatures.

pH and Alkalinity

Bonn et al. (1976) give optimum pH values of 7.5 to 8.5 for striped bass. A pH 5.3 has been reported to be lethal to striped bass (Tatum et al. 1965). In our unit striped bass have been reared at pH values of 6.7 to 8.4 with no deleterious effects. However, a pH of 7.8 to 8.0 is preferred to insure optimum conditions for biofiltration. Large amounts of free hydrogen ions are released during nitrification. Nitrite and nitrate are released by the bacteria as nitrous and nitric acids. Therefore, to compensate for this addition of acid, maintaining alkalinities greater than 150 mg/liter (as $CaCO_3$) is recommended. Various methods are available for alkalinity control. Burrows and Combs (1968) utilize a layer of crushed oyster shell 30 cm deep in their biofilter. They state that they could maintain the pH of their system at 7.8 with this method. As reported by Siddall (1974) calciferous materials which also contain magnesium (such as dolomite gravel or oyster shell) are better buffers than pure calcium carbonate (calcite). However, in a system using recirculated water, the buffering capabilities of these materials can also be reduced by coatings of either organic or inorganic materials. Burrows and Combs (1968) prevented this occurrence by frequently agitating the oyster shell during backwash procedures. Therefore, some method should be available to agitate the buffering material at frequent intervals. Siddall (1974) found that tumbled gravel had greater buffering capacity than new gravel

which had been washed. In our unit crushed oyster shell is added to the aeration basin below the aspirators. During the fingerling production season the alkalinity never falls below 218 mg/liter as $CaCO_3$ and pH in the rearing tanks has ranged from 7.5 to 8.1 with no change greater than 0.1 pH per day.

Although the oxidation of ammonia to nitrite occurs most rapidly at a pH of 7.8 in fresh water, the optimum pH for the nitrite to nitrate reaction occurs at 7.1 (Saeki 1958). Therefore a pH between these values can be considered optimum.

Maintaining high alkalinity in the rearing water with a calcium (magnesium) carbonate-bicarbonate buffering system will result in hard water. A high total hardness may enhance survival of striped bass. Bonn et al. (1976) recommend a hardness in excess of 150 mg/liter. Hazel et al. (1971) had difficulty keeping fingerling striped bass alive in water with a hardness of 25 to 30 mg/liter as $CaCO_3$, but were successful with a hardness of 150-200 mg/liter.

High alkalinity and pH will also reduce the toxicity and solubility of some of the heavy metals. Burrows and Combs (1968) have found no measurable amounts of copper and zinc in their system although they use bronze and brass pump impellers and valves. However, higher pH also increases the percentage of unionized ammonia, the toxic form.

Salinity

In a study by Albrecht (1964) survival of striped bass fry less than 5 days old was inversely related to salinity (diluted sea water). Fingerling striped bass, however, tolerate increasingly greater amounts of salinity as they increase in size, and slightly saline water may

increase survival. However, due to the increased corrosion of pipes and pumps with saline water, it is not recommended for a culture facility using recirculated water. Also, exposure to 0.4% saline water (as NaCl) greatly reduced the efficiency of freshwater nitrifying bacteria (Kawai et al. 1965).

Ammonia

Ammonia is second only to oxygen as a cause of mortality in cultured fishes. Ammonia is excreted by the fish and produced by the mineralization of organic waste. In water, ammonia takes two forms: ammonium ion (NH_4+), and undisassociated ammonia (NH_3 or NH_4OH). The undisassociated form is toxic ammonia. Temperature and pH determine the relative amounts of undisassociated ammonia in the water, with pH having the greater effect (Trussell 1972). Downing and Merkins (1955) determined that there was ten times more toxic ammonia in the water at pH 8 than at pH 7. The proportion of ammonia in the unionized form can be found in published tables such as those in Emerson et al. (1975). However, with the availability of hand-held calculators, these can easily be calculated from the equation of Trussell (1972). The pK_a or disassociation constant is first calculated as a function of temperature by:

$$pK_a = 0.0918 + \frac{2729.92}{T}$$

where T is in Kelvin (Celsius +273).

The proportion of ammonia in the unionized form is then calculated from the pH and pK_a using:

$$\text{proportion unionized} = \frac{1}{(10^{pK_a - pH} + 1)}$$

This proportion can then be multiplied by the total ammonia concentration to give the concentration of unionized ammonia. Other water quality parameters also affect the toxicity of ammonia. Decreases in oxygen increase the toxicity of ammonia to trout (Merkins and Downing 1957). Salinities at the concentration of seawater have also been shown to increase the toxicity of ammonia to striped bass fingerlings (Hazel et al. 1971).

For striped bass the 96-h LC_{50} for undisassociated ammonia (NH_4OH-N) was determined to be 1.7 mg/liter at 15 C, and 0.8 mg/liter at 23 C (Hazel et al. 1971). At the pH of their test water (7.3 to 8.0) the total ammonia concentration (NH_4-N) would be 20 to 100 times these values. The highest ammonia concentration obtained in the SIUC system was 2.6 mg/l NH_3-N. At a pH of 7.63, only 0.0695 mg/l was in the unionized form.

Sublethal effects of ammonia in fishes include reduced feeding, reduced growth, gill damage and reduced resistance to diseases. Therefore, ammonia levels should be kept to a minimum. Bonn et al. (1976) recommend maintaining total ammonia concentrations below 0.6 mg/liter. An efficient biofilter should keep ammonia concentrations below this level.

Nitrite

Nitrite (NO_2-N) is not an excretory product of fishes, but is formed when Nitrosomonas oxidizes ammonia. At present, safe limits for nitrite have not been determined for striped bass fry or fingerlings. Bonn et al. (1976) recommend keeping nitrite levels below 0.2 mg/liter. In our unit, 5-cm fingerlings have been exposed to nitrite

levels as high as 1.4 mg/liter (as NO_2-N, 4.6 mg/liter as NO_2) for short periods without mortality. However, the normal operating level is less than 0.10 mg/liter as NO_2-N (0.33 mg/liter as NO_2).

Nitrate

Nitrate is the end product of the oxidation of ammonia. As with nitrite, no safe or lethal limits have been determined for striped bass. However, according to Bonn et al. (1976) striped bass will tolerate nitrate levels in excess of 800 mg/liter, although levels below 38 mg/liter are recommended. In a system using recirculated water a 2% flush will maintain nitrate levels at 7.5 mg/liter (Burrows and Combs 1968). In the work at SIUC, the highest level of nitrate attained was 18.5 mg/liter as NO_3-N (81.4 mg/liter as NO_3), as reported by Lewis et al. (1980).

System Monitoring

Intensive culture of fish necessitates monitoring of the mechanical system, including water pumps, automatic feeders, and heaters. It also permits the condition of the fish to be closely watched.

The success of a production year depends on the ability to maintain water flow through the rearing tanks. Even with automatic emergency oxygen systems, the cessation of water flow through the rearing tanks for a period of several hours can result in loss of fish from ammonia toxicity. Water flow can be easily monitored with flow switches controlling alarms. Backup pumps should be available and personnel with the ability to activate these pumps must be available.

During the 25 to 30 days in which the striped bass are receiving

live brine shrimp nauplii, the solenoid btine shrimp feeders must be monitored regularly. A frequent fault of these feeders is the jamming of the solenoid mechanism by hard-shelled insects which have fallen into the feeder.

Other mechanical systems which must be watched include the compressed air system and the water heating system for brine shrimp rearing. Failure of the aeration system in the brine shrimp rearing facility can result in the loss of two consecutive days of brine shrimp, whereas failure of the water heating system can result in greatly reduced hatches of brine shrimp. Both of these factors can result in an interruption in the feeding schedule which may result in cannibalism and lower production.

Monitoring Water Quality

Monitoring of the water quality parameters must be done regularly. Once the biofilter is activated, water quality generally remains excellent until the fish are being trained to accept commercial diets. Therefore, before training begins water quality need not be closely monitored.

During the training period and the period in which the fingerlings are less than 25 mm in total length, both the general system chemistry and the rearing tank chemistry should be monitored. At this time there is the greatest accumulation of uneaten food and organic material in the rearing tanks, providing the conditions for oxygen depletion and ammonia buildup in individual rearing tanks. These two parameters should therefore be monitored every other day in each of the rearing tanks.

Once the fish surpass 20 to 25 mm, the flush rates can be increased to remove excreted ammonia. Also at this time the feeding rates can be adjusted to prevent overfeeding, after which the monitoring of ammonia levels in the rearing tanks can be eliminated. However, the high activity level of the fish and midsummer fluctuations in temperature can result in oxygen depletions in individual tanks. Therefore, monitoring of this parameter should continue.

Monitoring of general water quality in the rearing system should include ammonia, nitrite and dissolved oxygen in both the biofilter influents and effluents. These parameters will give a good indication of the condition of the biofilters. Periodically, pH and alkalinity should be determined to insure that pH is within the proper range and that sufficient buffering capability exists.

Monitoring of Fish

The striped bass fry and fingerlings should also be monitored. Periodically, samples of fish should be taken and examined. From these fish, the growth rate, the amount of food in the stomach, the presence or absence of fat in the peritoneal cavity, and the presence or absence of any deformities can be determined. Microscopic examination of these fish should be made to determine whether any parasites are present.

General observation of the fry and fingerlings should also be made at frequent intervals. Aberrant behavior can suggest the development of a problem. Reduced feeding rates can be the result of deteriorating water quality or parasite infection. Fish scratching ("flashing") can also mean that a parasite is present. Other aberrant conditions which

can readily be observed include the presence of damaged or diseased fish, erratic swimming, or piping at the surface.

When water temperatures surpass 27 C, striped bass fingerling have a tendency to damage one another when feeding. This condition can be determined by the presence of damaged fish; fish with frayed fins, or missing eyes. Fish which have lost scales from fighting can suffer severe osmotic stress. These fish will appear pale and will often die.

Striped bass fingerlings which have been graded suffer some mechanical damage. The larger fish which have been retained by the grader can develop bacterial infections (Aeromonas or Pseudomonas) from this damage. During the initial phases of infection, infected fish rapidly develop pale areas in the caudal region. If this is observed, the fish in that tank should immediately be fed a medicated food.

Erratic swimming behavior by some of the fish can be caused by the lack of an inflated gas bladder. Fish lacking an inflated gas bladder usually swim at or near the surface with their heads toward the surface. These fish will also tend to be smaller than those which possess an inflated gas bladder.

Fish which are piping at the surface are suffering from a lack of oxygen, resulting either from a lack of dissolved oxygen in the rearing tank or from damage or disease in individual fish. Moribund fish which have suffered mechanical damage and are in osmotic shock can also be found piping at the surface.

Healthy striped bass fry exhibit certain predictable behavior patterns. While in the upflow inserts, the striped bass larvae can at times be seen in tight clusters, either near the surface or at

other depths in the tanks. This condition may be a passive act. The larvae may be accumulating in certain areas of low water currents. At other times these aggregations appear to be reacting to light intensity, with the fish concentrating in areas of lower light intensity.

On several occasions we have observed striped bass larvae swimming at the surface, apparently attempting to break the surface film. This behavior may be related to gas bladder inflation, although we have no firm data to support this hypothesis. However, we do recommend that the surface film on the upflow tanks be periodically removed during the period of gas bladder inflation (age 5 days until the fish are harvested from the upflow tanks).

Striped bass fingerlings are strongly rheotaxic, swimming against even slight circular currents. Some fish will also orient ventrally to the sides of the tank, suggesting that they are not strongly geotaxic or that they lack a strong dorsal light response. They will frequently orient to the tank sides or the sides of retention screens rather than the tank bottoms. If the fish are swimming laterally or searching for food, this behavior should be considered normal. However, if the head is pointed toward the surface, these fish may lack inflated gas bladders.

Cannibalism can be monitored by direct observation. Before a significant size differential will identify fish as cannibals, their behavior can often be observed. The cannibals will frequently not participate during feeding, but will remain in the lower half of the tank, often swimming from fish to fish as if to determine whether a given fish is susceptible to being eaten. When these fish are observed, attempts should be made to remove them by grading. Once removed from smaller fish, the cannibals will readily take the commercial feed.

Circular Tank Harvest and Grading Fingerlings

When highly predatory fish such as the striped bass are confined at high densities, the potential for cannibalism exists. Losses from cannibalism of 70 to 80% can occur among striped bass fry and fingerlings (Lewis et al. 1977a). Therefore, these fish must be graded at an early age and frequently. Striped bass with total lengths of 11 mm have been observed to cannibalize 10 mm striped bass. However, due to the difficulty in handling striped bass fry of this size, and to the lack of sufficient difference in size between the cannibals and other fry, grading becomes difficult until the cannibals are 22 to 25 mm long. Also due to the lack of a large size differential, the earlier gradings tend to be incomplete, thus necessitating frequent grading. Bonn et al. (1976) recommend grading striped bass fingerlings at 3-week intervals. This frequency requires considerable time and labor, but will significantly reduce the losses from cannibalism.

To assist in the capture of striped bass from the circular tanks, a rotating fish concentrator can be constructed. The concentrator consists of two plywood frames connected to a 30-cm long section of plastic pipe with hinges (Figure 18). The plywood frames are covered with nylon screen (3.2 mm mesh). When the two arms of the concentrator are in line, they should be within 2.5 cm of the tank sides. The concentrator is 25 cm (10 in) high. Flexible rubber is attached to the outer edges of the concentrator arms and between the inner edges and the center pipe to prevent fish from escaping. The diameter of the center pipe is large enough to be slipped over the tank

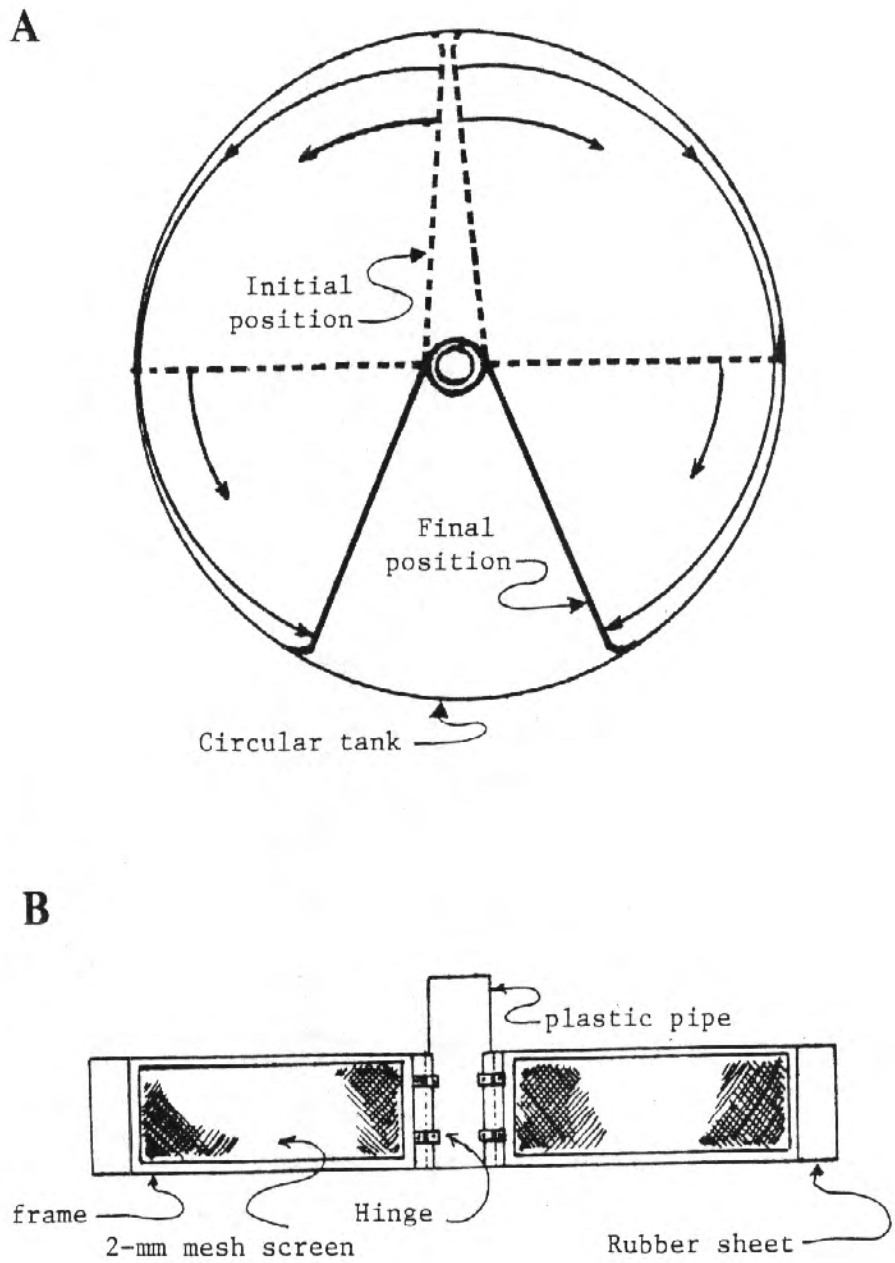


Figure 18. Principle of use (A), and design (B), of a fish concentrator for use in circular tanks with a center standpipe.

standpipe.

To use the concentrator the water level in the rearing tank is lowered to a depth of 15 to 20 cm (6 to 8 inches). The standpipe screen is then removed and the concentrator is placed over the standpipe (Figure 19). By rotating the arms of the concentrator toward one another, the fish can be concentrated in one portion of the tank and removed with a dip net. When used with 8-cm striped bass fingerlings, 85 to 100% of the fish can be captured in a single pass of the concentrator.

For striped bass less than 25 mm long it is necessary to construct a bar grader; for fish above 25 mm commercial minnow graders can be used. As a reference value, striped bass 25 mm (1 inch) in total length are retained by a bar spacing giving a clearance of 4 mm (0.16 inches). Prior to grading the fingerlings, the system temperature is lowered to 20 C to reduce the activity level of the fish.

Weights and numbers of striped bass in the rearing tanks can be determined during the grading process. Samples of fish are netted from the rearing tank, counted, and weighed in water (Figure 20). Average weights calculated from the samples are then used to estimate the number of fish in the rearing rank from the total weight of fish in the tank.

Figure 19. Harvesting of striped bass fingerlings with the circular concentrator (second pass through the rearing tank).

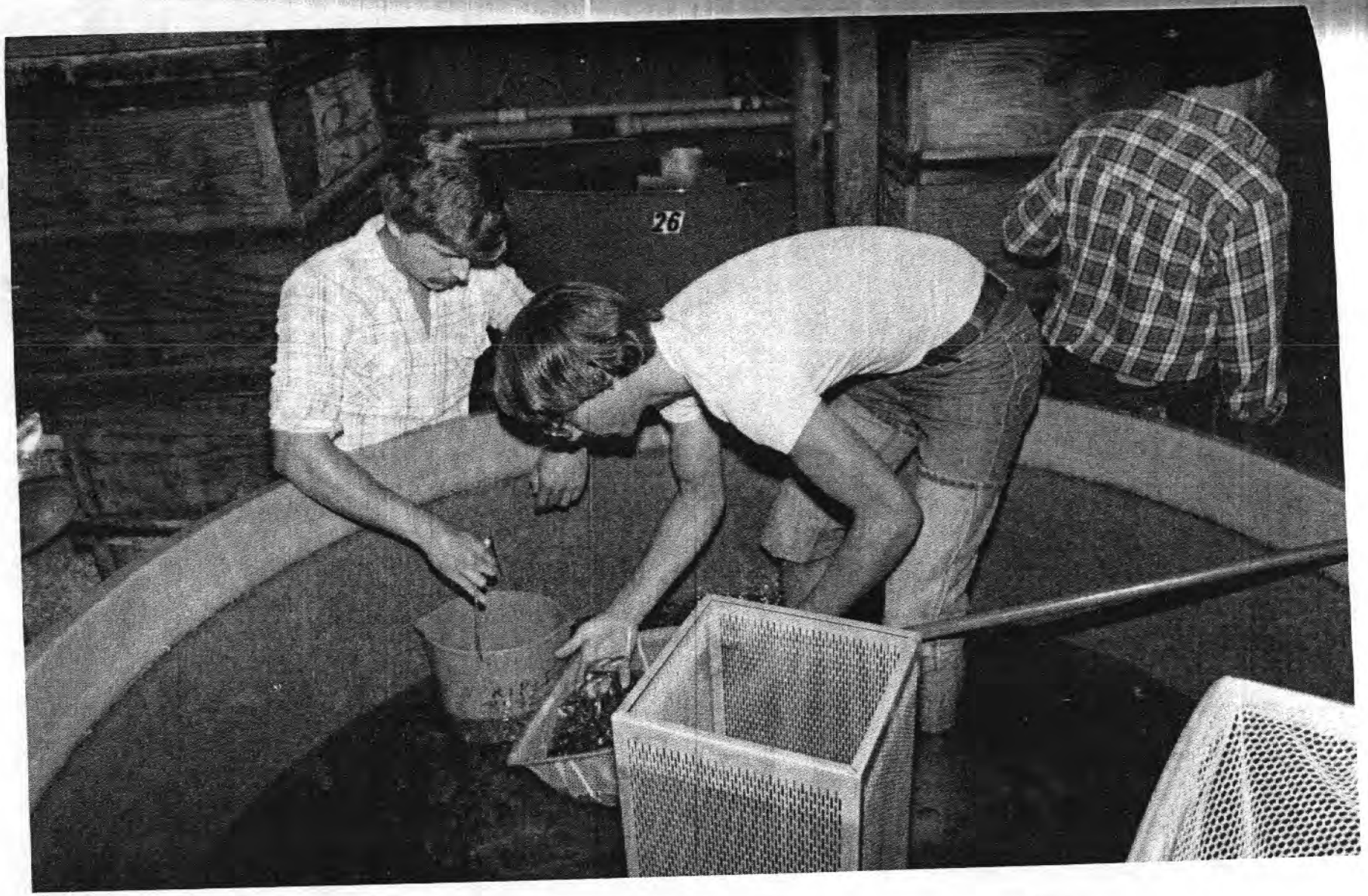


Figure 20. Sampling of 5-cm striped bass fingerlings prior to grading.

Transport and Stocking of Fingerlings

The system by which striped bass fingerlings are transported to a lake for stocking varies with the size of the fingerlings and the distance they are to be transported. For distances which require 4 hours or less of travel, we haul the fingerlings in 0.75% salt (uniodized rock salt) and 0.5 to 1.25 ppm quinaldine (Lewis et al. 1978). Bonn et al. (1976) recommend 1% reconstituted sea water with 0.25 quinaldine. The concentration of quinaldine used should not totally anesthetize the fish. Five minutes after being introduced into the well-mixed hauling medium, the fingerlings should be able to maintain equilibrium and exhibit swimming ability, yet be sedated enough to be captured by hand (Figure 21). The use of salt reduces the osmotic effects of shock which are produced by the stress and handling and mechanical damage. For trips of 4 hours or less, striped bass fingerlings have been transported at temperatures ranging from 17 to 27 C (63 to 81 F), with the lower temperatures used for longer trips.

For fingerlings 5 cm or less in length the transport water should be oxygenated by use of compressed oxygen dispensed through air stones or millipore tubing. Compressed oxygen may also be used for fingerlings larger than 5 cm, but agitators may be more satisfactory for extended distances of transport. If agitators are used, the level of sedation of the fish should be held to a minimum. Agitators cannot be used for fish smaller than 5 cm, since this size fish will suffer mechanical damage from the agitators. Striped bass transported in perforated minnow hauling cans without sedation suffered 100 percent mortality.

Figure 21. Properly sedated fingerlings ready for transport.

For hauling the fingerling striped bass we use filtered, oxygenated, well water to which enough recirculated water from the system is added to adjust the temperature. The use of 100% recirculated water limits the distance which striped bass can be transported, since recirculated water will have a biological oxygen demand prior to the beginning of transport.

Other propyhlactic chemicals which can be added to the hauling medium include antibiotics or antibacterials and antifoaming agents. Furacin at 5 to 10 ppm (active ingredient) has been used successfully as a bacteriostat. With hauling temperatures below 22 C (72 F) foaming has not occurred when hauling striped bass at SIUC. However, when transporting fingerlings at higher temperatures foaming has occurred, necessitating the use of antifoam chemicals.

When transporting 5-cm fingerlings, Bonn et al. (1976) recommend not exceeding densities of 60 g of fish per liter for travel times of 1 to 4 hours, 40 g per liter for trips of 4 to 8 hours, and 30 g per liter for trips over 8 hours (0.5, 0.33, and 0.25 lb/gal). Ice which has been made from chlorine-free water can be used to maintain temperatures when necessary.

The fingerling striped bass should be tempered with lake water prior to release. The addition of lake water to the hauling tank will adjust the temperature of the fish to that of the lake, and flush the anesthetic from the water. Striped bass fingerlings appear to rapidly sedate and rapidly recover from anesthetization with quinaldine. However, tempering times of 30 minutes to 1 hour (5 to 10 turnovers of water in the hauling tank) are recommended to insure that the fingerlings

are able to orient after release. We use a 12-volt submersible pump with a capacity of 150 lpm (40 gal/min) for flushing the hauling tank during tempering (Figure 22).

For release of the fingerlings into a lake, direct, rapid, gravity draining of the fish and hauling water is superior to netting the fingerlings. To facilitate direct draining of the fingerlings into the lake, an aluminum or plastic pipe 6 to 8 inches in diameter and 10 to 15 feet long can be used (Figure 23). Continuous pumping with the tempering pump during release prevents any fish from being stranded in the transport tank.

The particular habitat of a lake into which the fingerling fish are stocked may affect their survival. Areas with moderate turbidity may reduce the amount of predation (Bonn et al. 1976). We chose areas which are immediately adjacent to a large expanse of open water. The fingerlings may then reach open water without encountering shoreline predators. In Texas, striped bass are stocked directly into open water (Burkett et al. 1977).

To reduce the possibility of light-induced shock, the site of stocking should be shaded or the stockings made on overcast days. However, stocking the fingerlings by direct draining eliminates the need for shade, since the fingerlings are not exposed to direct daylight when this method is used. We have attempted to reduce the potential for predation and light-induced stress and disorientation by stocking at night (Lewis et al. 1978). Night stocking also reduces the need for temperature regulation during transport. When stocking at night the time periods around dusk and dawn should be avoided, since these are prime feeding periods for some predatory fishes. There is

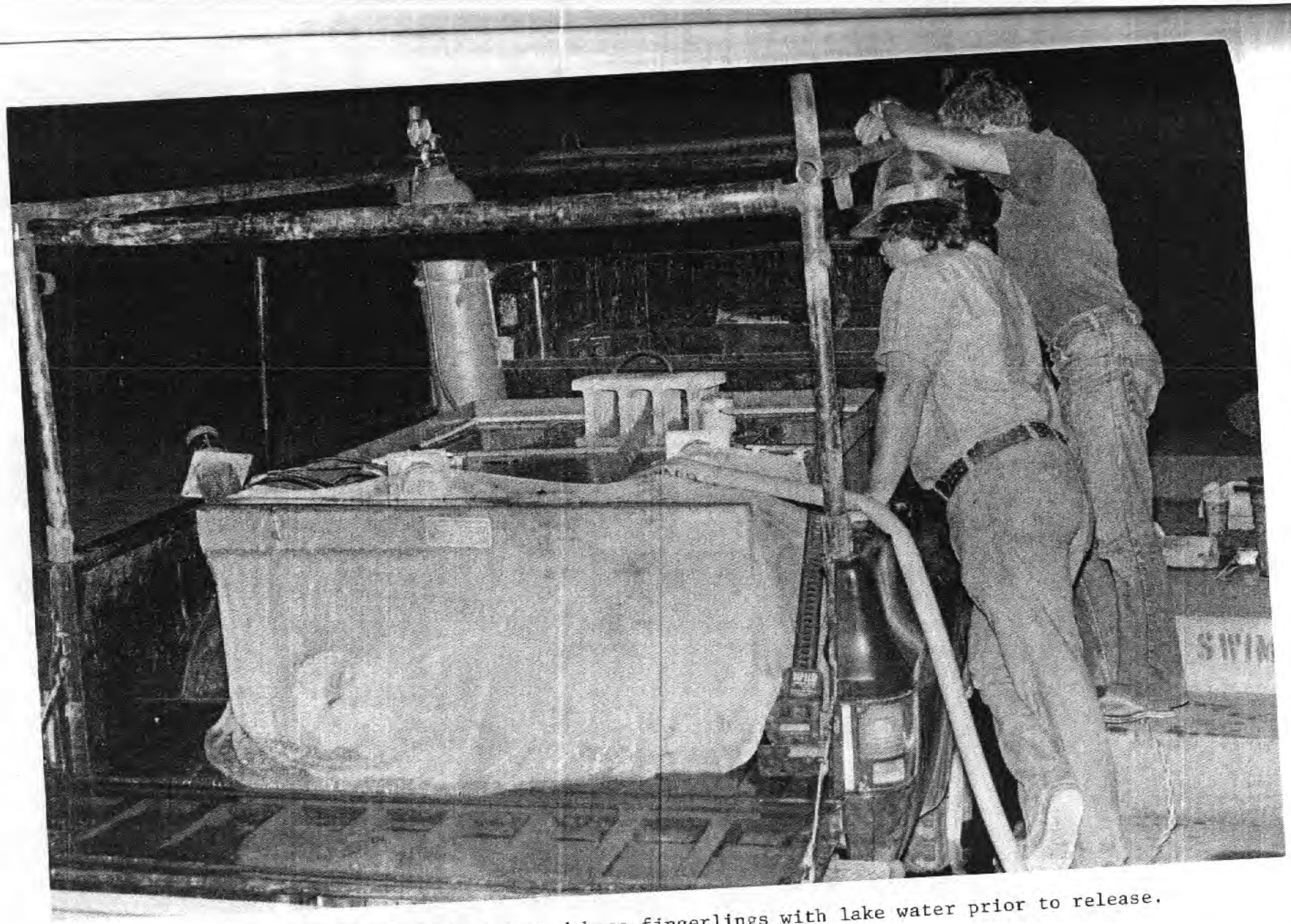


Figure 22. Acclimating striped bass fingerlings with lake water prior to release.

Figure 23. Release of striped bass fingerlings by direct draining. (These fish were stocked at 11:00 p.m. to eliminate light-induced stress and to reduce the possibility of predation).

also some evidence which suggests that striped bass fingerlings are attracted to the lights found around some boat ramps, causing the fish to remain in shallow water.

References

Al-Ahmad, T.A. 1978. Evaluation of rotifers as food for striped bass, Morone saxatilis (Walbaum), fry under controlled condition. M.S. Thesis, Auburn University. 62 pp.

Albrecht, A.B. 1964. Some observations on factors associated with survival of striped bass eggs and larvae. Calif. Fish Game. 50(2):100-113.

Bonn, E.W. (Chairman) and the Striped Bass Committee of the Southern Division of the American Fisheries Society. 1976. Guidelines for striped bass culture. Publ. Striped Bass Comm. So. Div. Amer. Fish. Soc. 103 pp.

Bowman, J.R. 1979. Survival and growth of striped bass, Morone saxatilis (Walbaum), fry fed artificial diets in a closed recirculating system. M.S. Thesis, Auburn University. 59 pp.

Braid, M.R. 1977. Factors affecting the survival and growth of striped bass, Morone saxatilis, fry in recirculating systems. Ph.D. Thesis, Auburn University. 86 pp.

Brandenburg, A.M., M.S. Ray, and W.M. Lewis. 1979. Use of carp eggs as a feed for fingerling largemouth bass. Prog. Fish-Cult. 41(2):97-98.

Burkett, H.B., E.G. Simmons, and R.J. Kemp. 1977. Statewide fisheries research. Statewide striped bass study. Texas Parks and Wildl. Fed. Aid Proj. F-31-R-3. Final Rep. 30 pp.

Burrows, R.E., and B.D. Combs. 1968. Controlled environments for salmon propagation. Prog. Fish-Cult. 30(3):123-136.

_____, and J.R. Galat. 1974. Development of upflow expanded polystyrene bead filter with pre-settling. pp. 99-102 In Water reuse systems for fish hatcheries. Workshop Proceedings.

Carreon, J.A. 1978. Studies on the culture of larval striped bass, Morone saxatilis (Walbaum) in closed recirculating systems. Ph.D. Thesis, Auburn University. 101 pp.

Chitterden, M.E., Jr. 1971. Effects of handling and salinity on oxygen requirements of striped bass, Morone saxatilis. J. Fish. Res. Bd. Can. 28(12):1823-1830.

Collins, M.T., J.B. Gratzek, D.L. Dawe, and T.G. Nemetz. 1975. Effects of parasiticides on nitrification. J. Fish. Res. Bd. Can. 32(11): 2033-2037.

Combs, B.D. 1974. Operational data for the production-recycle system at Abernathy. pp. 19-21 In Water reuse systems for fish hatcheries. Workshop Proceedings.

Cox, David K. and Charles C. Coutant. 1980. Growth efficiences of juvenile striped bass, Morone saxatilis, as a function of temperature and ration. Draft report of research sponsored by the Office of Health and Envir. Research, U.S. Dept. Energy under contract W-7405-eng-26 with Union Carbide Corp. Environ. Sci. Div., ORNL. 24 pp.

Davis, J.T. 1977. Design of water reuse facilities for warm water fish culture. Ph.D. Thesis, Texas A & M University. 109 pp.

Dorshev, S.I. 1970. Biological features of the eggs, larvae, and young of the striped bass, Roccus saxatilis (Walbaum) in connection with the problem of its acclimation in the U.S.S.R. J. Ichth. 10:235-248.

Downing, K.M., and J.C. Merkins. 1955. The influence of dissolved oxygen concentrations on the toxicity of unionized ammonia to rainbow trout (Salmo gairdnerii Richardson). Ann. Appl. Biol. 43:243-246.

Emerson, K., R. C. Russo, R.E. Lund, and R.V. Thurston. 1975. Aqueous ammonia equilibrium calculations: effects of pH and temperature. J. Fish, Res. Bd. Canada 32(12):2379-2380.

Hazel, C.R., W. Thomson, and S.J. Meith. 1971. Sensitivity of striped bass and stickleback to ammonia in relation to temperature and salinity. Calif. Fish Game 57(3):138-153.

Hughes, Janice S. 1969. Toxicity of some chemicals to striped bass (Roccus saxatilis). Proc. 22nd Ann. Conf. S.E. Assoc., Game Fish Comm. 22:230-234.

_____. 1971. Tolerance of striped bass, Morone saxatilis (Walbaum) larvae and fingerlings to nine chemicals used in pond culture. Proc. 24th Ann. Conf. S.E. Assoc. Game Fish. Comm. 24:431-438.

_____. 1973. Acute toxicity of thirty chemicals to striped bass (Morone saxatilis). Western Assoc. Game Fish Comm. 53:399-413.

Kawai, A., Y. Yoshida, and M. Kinata. 1964. Biochemical studies on the bacteria in aquariums with circulating systems. I. Changes in the qualities of breeding water and bacterial population of the aquarium during fish cultivation. Bull. Jap. Soc. Sci. Fish. 30:55-62.

_____. 1965. Biochemical studies on the bacteria in aquariums with circulating systems. II. Nitrifying activity of the filter

sand. Bull. Jap. Soc. Sci. Fish. 31:65-71.

Klyashtorin, L.B. and A.A. Yarzhombek. 1972. Some aspects of the physiology of the striped bass, Morone saxatilis. J. Ichth. 15(6): 985-989.

Konikoff, Mark. 1973. Comparison of clinoptilolite and biofilters for nitrogen removal in recirculating fish culture system. Ph.D. Thesis. Southern Illinois University, Carbondale. 98 pp.

Liao, P.B., and R.D. Mayo. 1974. Intensified fish culture combining water reconditioning with pollution statement. Aquaculture 3:61-85.

Lees, H. 1952. The ammonia-oxidizing systems of Nitrosomonas. Biochem. J. 52:134-139.

Lewis, W.M., and G.L. Buynak. 1976. Evaluation of a revolving plate type biofilter for use in recirculated fish production and holding units. Trans. Amer. Fish. Soc. 105(6):704-708.

Lewis, W.M. and R.C. Heidinger. 1974. Tank culture of striped bass fingerlings. Third Progress Rep. to UMA Engineering Pacific, Inc. Portland Oregon. 13 pp (mimeo).

_____, and_____. 1975. Striped bass rearing experiments 1975. Rep. to Consolidated Edison Company of New York. UMA Engineering, Inc. and Coop. Fish. Res. Lab., Southern Illinois University, Carbondale. 89 pp.

_____, and _____, and B.L. Tetzlaff. 1977a. Striped bass rearing experiments 1976. Rep. to Consolidated Edison Company of New York. Coop. Fish. Res. Lab., Southern Illinois University, Carbondale. 197 pp.

Lewis, W.M., R.C. Heidinger, and B.L. Tetzlaff. 1977b. Illinois striped bass project. Dept. Conserv. and S. Ill. Univer. Fed. Aid Proj. F-26-R-1. Ann. Performance Rep. 56 pp.

_____,_____,_____, A. Brandenburg, M. Paller, and L. Wawrononwicz. 1978. Illinois striped bass project II. Dept. Conserv. and S. Ill. Univ. Fed. Aid Proj. F-26-R-2. Ann. Performance Rep. 108 pp.

_____,_____,_____,_____, L. Wawronowicz, K. Kursawski, J. Stoeckel, E. Forss, and R. Sheenan. 1979. Illinois striped bass project III. Dept. Conserv. and S. Ill. Univ. Fed. Aid Proj. F-26-R-3. Annu. Performance Rep. 105 pp.

_____,_____,_____, K. Kurzawski, J. Stoeckel, and W. Davin. 1980. Illinois striped bass project IV. Dept. Conserv. and S. Ill. Univ. Fed. Aid Project. F-26-R-4. Annu. Performance Rep. 99 pp.

McHugh, J.J. 1975. Laboratory investigations of the mortality of striped bass less than eight weeks of age. M.A. Thesis. So. Ill. Univ. 41 pp.

_____, and R.C. Heidinger. 1977. Effects of light on feeding and egestion time of striped bass fry. Prog. Fish-Cult. 39(1):33-37.

_____,and_____. 1978. Effects of light shock and handling shock on striped bass fry. Prog. Fish-Cult. 40(2):82.

McIlwain, T.D. 1976. Closed circulating system for striped bass production. Proc. 7th Ann. Meeting World Mariculture Soc. 523-534.

Meade, T.L. 1974. The technology of closed culture of salmonids. Univ. Rhode Island Mar. Tech. Rep. 30. 30 pp.

Merkins, J.C. and K.M. Downing. 1957. The effect of tension of dissolved oxygen on the toxicity of unionized ammonia to several species of fish. Annu. Appl. Biol. 45:521-526.

Nickum, J.G. 1978. Intensive culture of walleyes: The state of the art. Selected coolwater fishes of North America. Amer. Fish. Soc. Spec. Publ. 11:187-194.

Pecor, C.H. 1978. Intensive culture of tiger muskelleunge in Michigan during 1976 and 1977. Selected coolwater fishes of North America. Amer. Fish. Soc. Spec. Publ. 11:202-209.

Presol, G. 1974. Nitrification in upflow biofilters. pp. 35-38 In Water reuse systems for fish hatcheries. Workshop Proceedings. KCM Consulting Engineers, Seattle.

Saeki, A. 1958. Studies on fish culture in filtered closed-circuit aquaria. I. Fundamental theory and system design. Bull. Jap. Soc. Sci. Fish. 23:684-695.

Schnick, R.A., F.P. Meyer, and H.D. Van Meter. 1979. Compounds registered for fisheries uses. Fisheries 4(5):18-19.

Semmens, M.J. 1976. A feasibility/development study for the removal of ammonia from wastewater using biologically regenerated clinoptilolite. Univ. Illinois Water Resources Center Res. Rep. 115. 141 pp.

Shannon, E.H. 1970. Effect of temperature changes upon developing striped bass eggs and fry. Proc. 23rd Annu. Conf. S.E. Assoc. Game Fish. Comm. 23:265-274.

Siddall, S.E. 1974. Studies of closed marine culture systems. Prog. Fish-Cult. 36(1):8-14.

Spotte, S. 1970. Fish and invertebrate culture. John Wiley and Sons, Inc., N.Y. 145 pp.

Tatum, B.L., J.D. Bayless, E.G. McCoy, and W.B. Smith. 1965. Preliminary experiments in the artificial propagation of striped bass, Roccus saxatilis. Proc. 19th Annu. Conf. S.E. Assoc. Game Fish. Comm. 19:374-389.

Trussell, R.P. 1972. The percent unionized ammonia in aqueous ammonia solutions at different pH levels and temperature. J. Fish. Res. Bd. Can. 29(10):1505-1507.

Wehr, L.W. and W.M. Lewis. 1974. An electrically operated automatic fish feeder. Prog. Fish-Cult. 36(2):117-118.

A P P E N D I C E S

Appendix A: Historical Development of Tank Culture for Striped Bass

The Fisheries Research Laboratory at SIUC began investigating the culture of striped bass in the Spring of 1974. During 1974 and 1975 experiments were conducted under contract with UMA Engineering of Portland, Oregon. UMA was conducting a feasibility study on a hatchery design for Consolidated Edison Company of New York. The major goal of the project was to develop the facilities and techniques necessary for rearing striped bass in tanks from newly hatched larvae to stockable sized fingerlings.

During the 1974 experiments, attempts were made to rear the larvae in 3.0 m diameter circular tanks, 0.6 m x 3.0 m aluminum raceways, and 1.8 m diameter fiberglass tanks. The 3.0 m tanks were supplied with water from earthen ponds, while the troughs and fiberglass tanks were supplied with filtered and ultra-violet sterilized well water. While in the tanks, the larvae were fed live brine shrimp nauplii and Abernathy's Salmon Starter, either separately or in combination. In addition, the striped bass in the circular tanks were receiving zooplankton with the water supply.

Three major periods of mortality were experienced during the 1974 experiments. Striped bass stocked into the circular fiberglass tanks at 2 and 3 days of age were suffering 98 to 100 percent mortality three days after stocking. However, larvae stocked into these tanks at 6 days of age did not experience this mortality. No reason for the mortality could be proven, although water velocity in the rearing tanks and/or toxicity of the new fiberglass tanks was suspected. Later studies suggested that tank toxicity was not the source of mortality.

A second period of mortality began when the fish were 10 days old.

This mortality was not as sudden and affected only the fish being held in the aluminum troughs and the fiberglass tanks. It was concluded that the feeding schedule was the cause of this mortality. Fish in the troughs and fiberglass tanks were being fed at 15-minute intervals for 12 hours approximating daylight and then not fed for 12 hours approximating night time. However, the fish in the 3 m circular tanks were fed continuously with natural food entering with the fresh water. By changing the feeding schedule of the fish in the troughs and fiberglass tanks to a 24 hour per day regime, the mortality was reduced.

The third period of mortality affected fish in the 3.0 m circular tanks and was related to deteriorating water quality. Insufficient screen surface area was designed into these tanks, thus preventing adequate flushing. Therefore, oxygen concentrations decreased while ammonia levels increased, resulting in the loss of fish.

In this first examination of the culture techniques needed to rear striped bass in tanks several major problems were defined. Laboratory studies determined that the food egestion time for striped bass is 5-6 hours. Also, striped bass will commence to feed and continue to feed in total darkness (McHugh and Heidinger 1977). Thus a 24-hour per day feeding schedule is necessary. The 1974 experiments indicated that larval striped bass will not survive when offered only salmon starter as food. Also, due to the fluctuating water temperatures experienced during the 1974 experiments, a rearing system which provided temperature control was designed.

To provide better environmental control for the 1975 rearing experiments UMA Engineering constructed the present system at Gorham,

Illinois. During the 1975 experiments, the system was operated on a semi-closed circuit. Aerated, filtered well water was supplied to two circular fiberglass tanks and the 10 aluminum raceways used in 1974. Thirty-two circular fiberglass tanks were supplied with recirculated water which used submerged upflow biofilters for ammonia control. The five 3-m diameter circular tanks were supplied with aerated water from an earthen pond and the effluents of the 10 aluminum raceways.

The experiments were designed to examine the effects of various water flow patterns on the survival of striped bass larvae, whether striped bass could be fed salmon starter initially, whether a 24 hour versus 12 hour feeding schedule was necessary, and how long striped bass could survive on brine shrimp nauplii alone. Four water flow patterns were examined: circular flow in the 1.8 m fiberglass tanks, linear flow in the aluminum troughs, downward flow in which the water passed through a gravel and rock substrate, and upflow in wooden tanks utilizing a perforated plate in the tank bottoms as the water inlet.

The results of the 1975 experiments demonstrated that the survival of striped bass larvae less than 5 days old was 3 to 19 times greater in the upflow tanks than their survival in the circular flow and downflow fiberglass tanks, and linear flow aluminum raceways. Additionally, the work confirmed the 1974 study which showed that the striped bass would not survive when initially fed salmon starter alone.

The 1975 study also suggested that brine shrimp nauplii is an adequate diet for the first 3 weeks, and that fish greater than 5 weeks old will readily accept the commercial salmon feeds. During the period when live brine shrimp nauplii become inadequate, cannibalism

became a major problem which continued until the fish were graded at 8 weeks of age. Striped bass reared during the 1975 experiments appeared in good condition but grew slowly. This suggested that the rearing temperature was less than optimum for growth of the larval and fingerling fish.

For the 1976 experiments, which were funded by Consolidated Edison Company of New York, modifications were made to the rearing system so that it could be run on closed cycle and the water heated. The goals of the 1976 experiments were to again show that prefeeding larvae placed in the circular flow tanks would not survive, and to examine several training diets to supplement live brine shrimp nauplii. The diets tested included Silver Cup Salmon starter, ground Tetra-Min® aquarium fish food, fresh frozen unfertilized fish eggs, and live grindal worms (Enchytrae). During the 1975 experiments, samples of fish greater than two weeks old showed a frequent occurrence of brine shrimp cysts in the gut samples. This suggested that the quality of brine shrimp supplied to the larvae was limiting. Therefore, the brine shrimp ration was increased 5 to 7 fold.

The results of the 1976 experiments confirmed that larvae stocked into the circular tanks at ages less than 6 days will not exhibit good survival. Also, the results suggested that cannibalism in striped bass larvae and fingerlings are functions of food supply. Once the fish are trained to readily accept commercial diets cannibalism will stop if the few very large individuals are graded out.

The examination of the various training diets showed variable results. Fish eggs were readily accepted by 10 mm striped bass. However, it was difficult to obtain sufficient quantities of small

eggs. The eggs of gizzard shad (0.7 mm diameter) were much more readily taken than were the 1.0 to 1.2 mm eggs of the common carp. Also, although 10- to 12-mm striped bass were observed striking the salmon starter, the food particles were usually rejected. We conclude that this rejection was a function of the texture of the food particles rather than an olfactory response of the fish. By grinding the food to a particle size less than 0.2 mm acceptance of the food was increased. However, the high oil content of the salmon starter made grinding difficult. Therefore, we use finely ground Tetra® flake food as our training diet.

The controlled temperature regime of the rearing water also appeared to be beneficial. By maintaining water temperature at 14-17 C during the first 10 days, very high oxygen content in the water was assured. By raising the water temperature to 25 C at the time the fish were being trained (15 to 17 days old), increased training success was attained which was probably a result of the increased activity of the fish. This temperature also produced good growth, resulting in 10 to 12 cm fingerlings after 100 days. Cox and Coutant (1980) later found that a temperature of 25 C produced the maximum growth and the maximum food intake in advanced striped bass fingerlings.

During 1977, 1978, and 1979 the experiments on improving the tank culture method of striped bass were included as part of a comprehensive striped bass project funded by Dingell-Johnson funds through the Illinois Department of Conservation. The 1977 studies were severely restricted due to a nation-wide shortage of striped bass fry. However, in 1977 studies were conducted to examine the ability of striped bass larvae to feed under dark conditions. This was a duplication of work

originally performed in 1974 except that younger larvae (5 to 9 days old) were used. Results from these studies showed that striped bass will both initiate feeding and continue to feed in total darkness. However, fish fed in light conditions had better success at capturing the live brine shrimp nauplii. In another experiment, we further examined the use of fish eggs as an alternate feed. Eggs from snook (0.4 to 0.5), white bass (0.6 mm), and common carp (0.9 to 1.2 mm) were tested. Results from this study indicated that striped bass larvae will not accept fish eggs until they reach 10 to 12 mm in total length, irregardless of size of egg tested.

In 1978, experiments were conducted to further develop knowledge on the feeding of striped bass less than 25 mm in length. The results of these studies confirmed the importance of brine shrimp nauplii. We found that cannibalism and differential growth of striped bass can be remarkedly reduced by increasing the quantities of brine shrimp given to the fish and that a shortage of sufficient brine shrimp for a period of three days can induce cannibalism. Secondly, the survival of striped bass larvae is directly related to the quantity of brine shrimp utilized. Also, a rapid conversion from brine shrimp to commercial feeds can produce cannibalism and differential growth in 19 mm larvae.

During 1979, the first attempt was made to predict the number of surviving striped bass fingerlings from the quantity of brine shrimp cysts utilized. During that year 101 thousand fish were produced, 92 percent of the predicted 110 thousand. However, later examinations of the brine shrimp cysts showed considerable variation in the weights of the cysts utilized. Therefore, some of this error may be attributed to underestimates of the weight of shrimp supplied to the fish.

In 1980, further data were collected on the relation between the quantity of brine shrimp cysts utilized and the number of striped bass produced. Data from 1980 and previous years have been included in this manual.

Additional research is needed on many aspects of striped bass culture. The factors affecting gas bladder inflation have yet to be identified. The causes of the mortality of striped bass fry shipped and stocked into the rearing system at 2.5 to 4 days of age are also unknown. In addition, technical data which would be useful include exact ammonia excretion rates of striped bass fingerlings, determination of the nutritional requirements of striped bass fry and fingerlings and whether live brine shrimp nauplii and the commercial feeds meet these requirements, and the effects of the intensity and periodicity of illumination on striped bass.

Appendix B: Biofiltration in Fish Culture

The use of a tank facility supplied with recirculated water for the intensive culture of fishes is primarily concentrated in the northern and western United States, where such systems are used for the production of salmonids. However, due to the rising cost of land and more stringent regulations on water quality of hatchery discharges, it is anticipated that the use of these systems will increase. This type of facility enables the fish culturist to more easily maintain optimum water temperature. Once the temperature of the water is adjusted, the energy requirement for maintaining optimum temperature is trivial compared to that which would be required to maintain a single pass system. Optimum temperature favors accelerated growth of the fingerling fish and thus reduces the time required for them to reach marketable or stockable size. Since the fish are confined, any outbreak of disease or pathogens can be discovered at an early stage and treatment is less costly and easier to accomplish. A number of species of fish must be trained to accept artificial feed. Tank confinement of the fish facilitiates the training process; in fact, some species cannot be trained except when they are confined at high density. Harvesting and grading the fish is also made easier by their confinement. In addition, cannibalism of piscivorous fish can be minimized in a tank system.

Tank culture of fish involving reuse of water does have certain limitations. The foremost limitation is cost, both for initial investment and for operations. Operational costs consist primarily of the energy required for pumping and heating the water, and the cost of

of feed for the fish. Another major concern when using a water reuse system is the danger of mechanical failure. In such a system oxygen is supplied to the fish by mechanical means, thus power failure can result in the complete loss of fish. A source of emergency power and backup pumps is therefore necessary. More extensive training is also required for hatchery technicians, since equipment such as pumps, automatic feeders and timing circuits and filters must be maintained, and water quality must be monitored.

A generalized water reuse system is shown in Figure B-1. The arrangement and design of the components may vary, but all tank systems contain the various components shown. In operation, the water from the culture tanks, carrying dissolved waste and part of the particulate matter, drains by gravity to a settling basin (in some systems the biofilter precedes the settling basin), where a large percentage of the particulate matter is removed, in part by sedimentation and in part by flotation. The clarified water then passes through the biofilter, where the dissolved waste materials, especially ammonia, are acted upon by bacteria growing on the substrate. The bacteria convert the highly toxic ammonia to less toxic nitrates. The water is then pumped to an elevated reservoir, being aerated as it enters the reservoir. A second reservoir at ground level is used to equalize the volume of water in the system. To prevent the accumulation of nitrate and other end products of waste digestion, a 5 to 10% replacement of water volume must be added to the system each day.

In biological filtration the various organic components excreted by fishes are digested by a succession of heterotrophic bacteria.

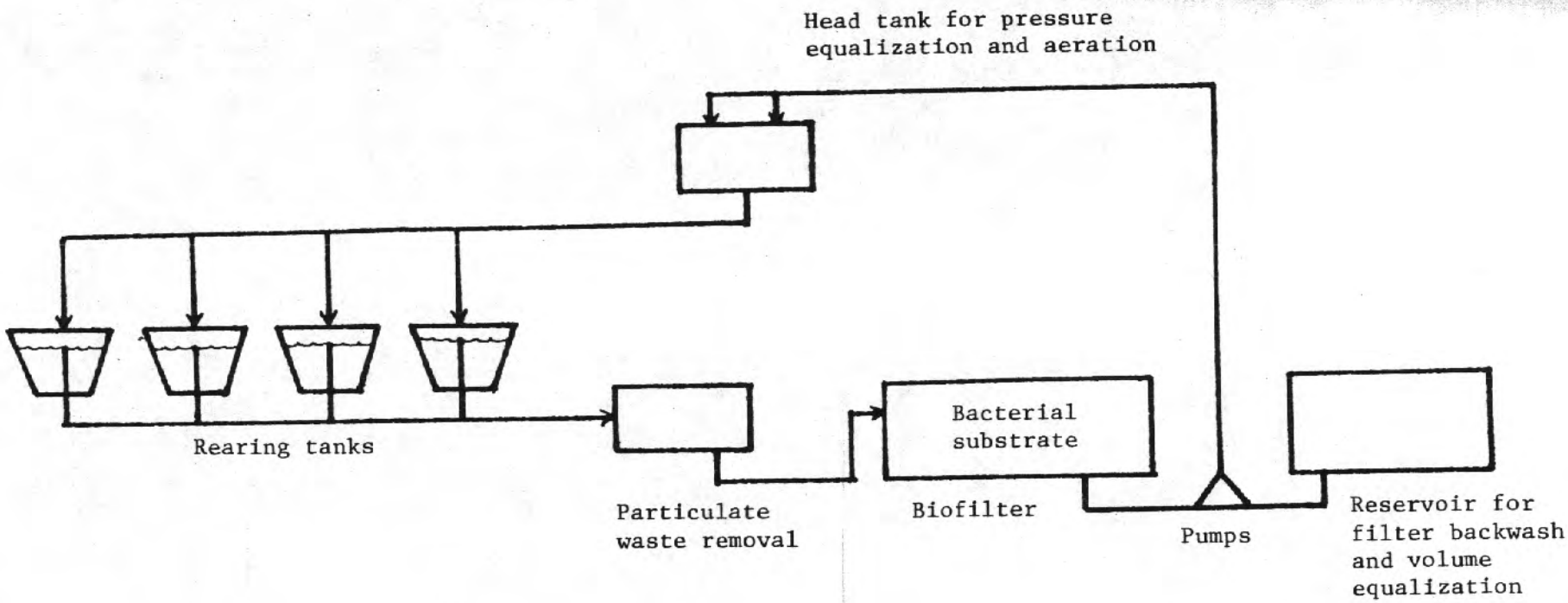


Figure B-1. Generalized water reuse system for fish culture. (Emergency electrical power and air supply not shown.)

The digestion process results in the production of carbon dioxide (CO_2) and ammonia (NH_3). This ammonia, and a large amount of the ammonia that is excreted directly by the fish is oxidized by bacterial action, first to nitrites and then to nitrates. This nitrification process is principally accomplished by two aerobic autotrophic groups of bacteria, Nitrosomonas and Nitrobacter. Nitrosomonas oxidizes ammonia to nitrite, which is then oxidized to nitrate by Nitrobacter. These bacteria are found in all aerobic garden soils, stream and lake sediments, and in flowing or standing water.

The nitrification process produces about two moles of hydrogen ions for each mole of ammonium that is oxidized. Meade (1974) examined several of the proposed chemical formulas for the nitrification process and reported that 4.0 to 4.6 grams of oxygen are required to oxidize 1 gram of ammonia to nitrate. The two products of nitrification, nitrite and nitrate, are actually released by the bacteria as nitrous acid and nitric acid (H_2NO_2 and HNO_3). Since these acids can cause a major reduction in pH, well buffered water, or the addition of a buffer in salt form, is required.

Although the nitrifying bacteria are found in the culture water, the biofilter offers numerous sites for the bacteria to attach and thus permits large populations to develop. Considerable work has been done on the design of biofilter systems. Three principal designs of biofilters have been used in fish culture systems: submerged, trickle, and revolving plate. A submerged filter may be either downflow or upflow. The advantage of an upflow filter is that less particulate material infiltrates the filter bed. In the trickle filter

the water cascades over the filter medium. The advantage of this filter is that it favors oxygen replenishment. The revolving plate filter has been used in fish culture only on an experimental basis (Lewis and Buynak 1976). This type of filter favors oxygen replenishment and is self-cleaning.

Several design features of biofilters have been found to affect nitrification, including: surface area of the biofilter substratum amount of void space in the medium retention time, flow rate, and depth of the filter. All of these design features are interrelated and essentially determine the exposure time of the water to the nitrifying bacteria. The surface area of the filter medium controls the amount of bacteria which can be present in the filter, while the surface area of the filter (the actual dimension) will control flow rate through the filter. The generally accepted rate of flow is 40 liters per minute per square meter (1 gallon per minute per square foot) for submerged downflow and trickling filters, and twice that rate for submerged upflow filters. Burrows and Combs (1968) recomend not exceeding 40 liters per minute per m^2 for submerged downflow filters. However, in their system particulate waste was filtered from the water in fine material placed above the biofilter. Liao and Mayo (1974) found that ammonia removal was independent of the rate of flow for rates between 61 and 102 liters per minute per m^2 of filter substrate. However, they used sedimentation, rather than filtration to remove particulate wastes. Optimum flow rate for the revolving plate biofilters when used in fish culture have yet to be determined.

The retention time in a biofilter is equivalent to the turnover time and is equal to the time required to fill the dry filter at the

given rate of flow. Retention time is related to flow rate, volume of the filter, and void space provided by the filter medium. In a filter with a given volume, increasing the size of the filter medium will increase the void space (space not occupied by the medium), and thus will increase the retention time. However, increases in the medium size will reduce the surface area of the medium available for bacterial attachment. Submerged filters with large void space frequently utilize forced circulation to increase the amount of contact between the water and bacterial substrate. The smallest medium size which will not become blocked is most desirable, providing the water remains in the filter for a long enough time for the bacteria to act on the ammonia. Increased retention time is to insure complete oxidation, and is usually most easily obtained by increasing the depth of the filter. Liao and Mayo (1974) recommend a filter depth of 1.2 m (4 ft), as do Burrows and Combs (1968). With a filter medium size of 1 to 9 cm, a filter depth of 1.2 m gives the required surface area for bacterial attachment and retention time for oxidation.

When one design feature is altered, a second feature must also be varied to compensate for the change in contact time. Thus when the depth of the filter is specified, the flow rate is also defined. In a deep filter (1.5 to 2.5 m) greater flow rates can be used. Also the water will mix sufficiently as it passes through the substratum. However, oxygen levels can drop within the filter thus affecting nitrification. When a shallow filter is used, the flow rate must be reduced to allow sufficient time for oxidation of the ammonia. Also, mechanical means of circulating the water within the filter may be necessary to assure that the ammonia has made contact with the bacteria.

If the surface area of the media is reduced (either by reducing the dimensions of the filter or increasing the size of the media), the flow rate must be reduced to again increase the contact time.

The size of the medium chosen for a filter is governed by the quantity of ammonia which must be oxidized, the physical space available for the filter, and the quantity of solid wastes which will enter the filter. Various water quality parameters affect the nitrification process; these include temperature, pH, dissolved oxygen, and ammonia concentration. The rate of nitrification increases as temperature increases. Kawai et al. (1965) found that the optimum temperature for nitrification in fresh water is 30 C, while a temperature of 30 to 35 C appears more suitable for salt water systems. However, cooler water temperatures had a less repressive effect in fresh water than in sea water. As discussed above, the nitrification process requires large amounts of oxygen. In fact, oxygen appears to be rate limiting (Semmens 1976). When oxygen levels in the filter bed fall below 3 mg per liter, nitrification virtually ceases and anaerobic conditions, even for a short period, may eliminate the nitrifying bacteria. Presol (1974) found that after a period of anaerobic conditions 25 to 30 days were required before nitrification reached normal levels at 12 C. It is thus evident that stopping the flow through a submerged biofilter, even for a short time, can have serious consequences.

Numerous authors have determined that increases in the influent ammonia concentration will increase nitrification. Davis (1977) found a linear relationship between the proportion of ammonia removed

and the influent ammonia concentration. In his experimental system, 25 percent of the ammonia was removed at an influent concentration of 0.4 mg/l ammonia, whereas 72 percent was removed at an influent concentration of 1.2 mg/l. However, due to the low level of ammonia tolerated in a fish culture system, this is of limited application. Nitrification is favored by pH levels above 7.0, with a pH of 7.8 being optimum for Nitrosomonas and 7.1 for Nitrobacter (Saeki 1958). Values above 7.0 insure the availability for inorganic bases such as Ca^+ and Mg^+ to neutralize the acid produced during nitrification. However, it should be noted that higher pH also increases the toxicity of ammonia by increasing the proportion of ammonia in the unionized form.

The development of Nitrosomonas populations in the filter bed is favored by high ammonia concentrations. Nitrobacter, however, is inhibited by high levels of ammonia, thus requiring the oxidation of much of the ammonia to nitrite prior to the development of Nitrobacter (Less 1952). This is most evident in new rearing systems initially stocked with moderate loads of fish (Figure B-2). Ammonia levels will increase due to the presence of fish, then decline as Nitrosomonas populations develop and oxidize the ammonia to nitrite. Only after much of the ammonia is converted to nitrite will Nitrobacter populations develop and begin oxidizing the nitrite to nitrate. Therefore, the weight of fish initially stocked into a new system must be very low unless the system has been preactivated. This condition can also develop in operating filters. A surge of ammonia produced by over feeding or improper tank cleaning can inhibit Nitrobacter, and

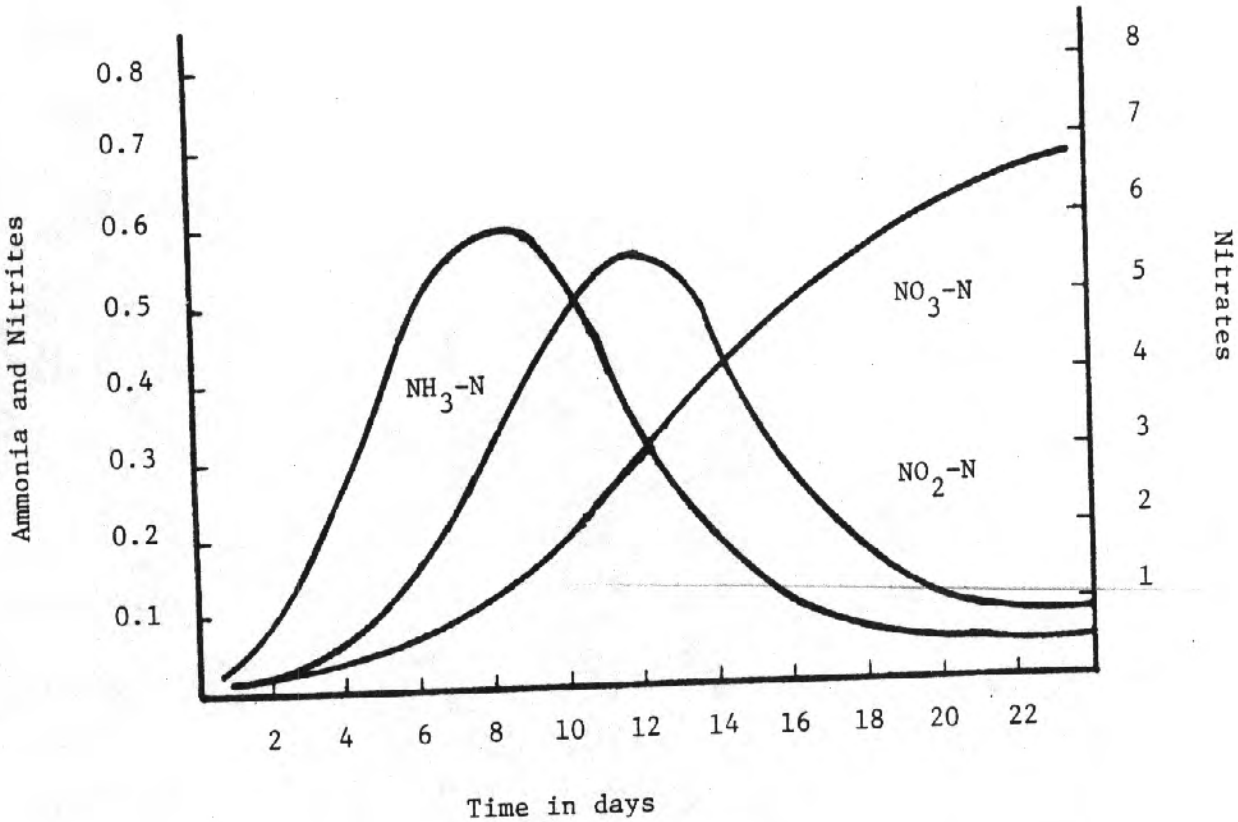


Figure B-2. Typical response of nitrifying bacteria in a new culture system with 2% makeup flush.

nitrite resulting from oxidation of some of the ammonia can cause loss of fish, while ammonia levels remain below the lethal level.

Tank culture systems involving biofiltration and reuse of water have definite carrying capacities. The carrying capacity of such a system is governed principally by the ability of the filter to oxidize ammonia. In the filter this oxidation capacity is controlled by the various physical dimensions and water quality parameters discussed above. With knowledge of these parameters, the carrying capacity (in terms of grams of ammonia per day) can be determined. However, to date little work has been done on the nitrification rates of fish culture biofilters at the temperatures at which warm water fishes are cultured (23 to 30 C). To convert carrying capacity expressed as grams of ammonia per day to carrying capacity in terms of weight of fish, knowledge of pollution rate by the species being cultured is necessary. As used here, "pollution rate" is defined as change in water quality resulting from the maintenance of the fish including pollutants excreted by the fish and released by the fish food.

Pollution rate (in grams of ammonia per unit weight of fish per day) is affected by species, size, and age of the fish, feeding rate, concentration of dissolved oxygen, temperature, and level of activity for the species (active metabolism).

Temperature will directly affect the metabolic activity of the fish. The efficiency of food conversion (weight gain of fish divided by weight of food utilized) may also be affected by temperature. The temperature which produces the most rapid growth may not be the temperature at which maximum food conversion occurs. Therefore, when temperature is controlled for maximum growth, a reduced percentage

of the food is assimilated and an increased percentage of it contributes to pollution of the water.

The activity level of the fish being cultured affects the quantities of pollutants entering the biofilter. Activity level (active metabolism) is a function of species, temperature, and rearing facility. Sedentary species, such as the channel catfish and largemouth bass, spend more time in an inactive state then pelagic species such as the striped bass, or stream species such as the rainbow trout. The increased activity of the latter two results in more energy being expended, and thus more metabolic pollutants entering the system. For a given species, rearing the fish in a system with greater water current requires them to expend more energy. Burrows and Combs (1968) note the difference in water velocities between rectangular circulating ponds and raceways. These authors consider the ponds with greater water velocities beneficial, since fish with more stamina are produced.

Two factors will limit the carrying capacity of the culture system; namely, water flow through the rearing tanks, and the ability of the biofilter to convert organic pollutants to less toxic minerals. The quantity of water flowing through the rearing tanks must be sufficient to both supply the fish with oxygen and remove accumulating toxic metabolites. However, the rate of flush cannot create water currents greater than those which the fish can tolerate.

The second limit on the carrying capacity of the system, the biofilter, is discussed in the section on biofiltration in Part I: System Design.

In addition to ammonia, other organic pollutants are produced by

the fish, principally dissolved and suspended organic materials. Some dissolved organics can be oxidized chemically and thus produce a chemical oxygen demand (COD) on the system, while other materials are utilized by heterotrophic bacteria, protozoa and fungi, and there is an associated biological oxygen demand (BOD). Growth of the heterotrophs can reduce the oxygen levels in the system and compete with the nitrifiers for sites of attachment on the filter substrate in the biofilter. Liao and Mayo (1974) believe that high quantities of organic material can limit ammonia removal. Their filters were achieving complete removal of ammonia until a pollution rate of 0.98 g NH_3-N per square meter of medium surface per day developed, after which the organic load prevented the filter from completely removing the ammonia. Rearing fish in a system with a high BOD and COD also adds to the risk of oxygen depletion during power outage or pump failure. The addition of pure oxygen to the biofilter to satisfy the BOD accelerates the mineralization of these organics.

Particulate organic matter suspended in the water can clog the biofilter and interfere with the function of the filter. The particulate matter adheres to the biofilter substrate, causing the flow of water through the filter to be channelized. This results in the development of anoxic conditions in portions of the filter, which can cause the production of toxic gases such as hydrogen sulfide that destroy the nitrifying bacteria. Suspended organic matter contributes to the growth of heterotrophs, which also tend to clog the filter, and the action of the heterotrophs on the organic material may result in the release of additional metabolic wastes. Liao and Mayo (1974)

determined that 70% of the ammonia (NH_3-N) was associated with organic solids. They reported that 20% of the ammonia load on the biofilter could be removed by the use of a clarifier.

Various methods of removing suspended solids have been employed. Burrows and Combs (1968) used a 25-cm layer of crushed oyster shells (5 to 20 mm in size) over their submerged downflow biofilters. Meade (1974) used a screen to filter out large particles prior to biofiltration. The biofilter used by Meade had a large void space and was thus less likely to become blocked. He used a sedimentation basin after the biofilter to serve as a clarifier. Liao and Mayo (1974) also recommend retention times of 15 to 30 minutes, a basin depth of 1 meter, and maintaining dissolved oxygen above 5 mg per liter in the basin. Davis (1977) examined the use of both pre-biofilter and post-biofilter settling basins in recirculating systems. He recommends a primary (pre-biofilter) settling basin which utilizes a 25 to 30 minute retention time, an overflow rate of 100 m^3 water per m^2 of basin surface per day and a basin depth of 1.3 m. He also recommends using the trout raceway cleaning technique in which the water level is lowered and the flush increased to allow fish activity to remove wastes. Davis determined that the solids produced by this method exhibit even (linear) settling with time. In the system used by Davis, a trickling filter was utilized, thus necessitating a post-biofilter clarifier. He determined that a clarifier with a retention time of 20 minutes, an overflow rate of 175 m^3 water per m^2 of surface per day and a depth of 1.8 m reduced solids by 80 percent.

Other methods which have been utilized to concentrate particulate

matter include sludge traps at the drain end of raceways and the sedimentation basin-sludge scraper system proposed by Burrows and Galat (1974).

The success of these various methods of concentrating particulate matter depends upon the settling rate of the material. During the process of training striped bass fry to accept dry feed in the SIUC system, particulate matter is produced which remains in suspension. In situations such as this, filtration would most likely be more effective than sedimentation.

On infrequent occasions, growth of the nitrifying bacteria will be great enough to cause blockage and channelization in the biofilter. Occasional backwashing will correct this condition. Trickling filters characteristically slough off much of their bacterial growth when the growth is excessive. If a large part of the growth leaves at one time, nitrification can be affected until a new growth attaches to the medium. Revolving plate biofilters demonstrate a continuous sloughing which eliminates this problem.