CHAPTER 4
INDUSTRY PROFILES

4.1 OVERVIEW OF THE INDUSTRY

Aquaculture in the United States began in the 1850s as a commercial enterprise when fish culturists developed the technology needed to spawn and grow brook trout. Several culturists who became proficient in fish raising techniques found that they could sell their fish for a profit (Stickney, 2000b). Today, the aquaculture industry in the United States encompasses the production of finfish, shellfish, crustaceans, reptiles, other aquatic animals, and aquatic plants. These plants and animals are produced for a variety of reasons, including as food, pets, bait, and sportfish; for ornamental and display purposes; as research and test organisms; and to enhance natural populations. EPA has broadly defined aquatic animal production (AAP) to include any production of aquatic animals and is not including aquatic plant production in the definition. The following chapter describes AAP in the United States, including systems used to produce aquatic animals and many of the aquatic animals produced.

As valuable commercial fisheries began to decline in the latter half of the 19th century, there was a growing concern about the stock depletion. Spencer F. Baird, who was affiliated with the Smithsonian Institute, worked with Congress to create a federal fisheries agency (Stickney, 2000b). The nation’s first federal conservation agency, the U.S. Fish & Fisheries Commission, was established in 1871. Known today as the National Marine Fisheries Service, the agency is responsible for marine commercial fisheries, while the U.S. Fish and Wildlife Service (USFWS) is responsible for freshwater fish, whose use is primarily recreational. The two agencies share responsibility for anadromous fish such as chinook and coho salmon.

After the U.S. Fish & Fisheries Commission was established, fish culture activities developed quickly. By the end of 1871, 11 states had established fish commissions; by 1877 there were fish commissions in 26 states. To further expand fish culture activities, Baird instructed Livingston Stone to set up an egg collection facility in California on the McCloud River. Prior to this facility, fish culturists transported fish via the newly established transcontinental railway from the east to the west coast. The striped bass and the American shad became most successful (Hartman and Preston, 2001). These species were exported around the world as well. Many people were optimistic that the work of artificial propagation of foodfishes, the introduction of promising exotic species, and the redistribution of native fishes to new waters could ensure an increased and sustainable food production in the nation’s natural water bodies, in particular the Great Lakes and the oceans.

The goals of public fish hatcheries, often referred to as conservation hatcheries, differ from the goals of private commercial fish hatcheries. Conservation hatcheries produce fish for stocking in public waters to enhance or restore recreational or commercial fisheries. Private, for-profit hatcheries produce fish for several purposes, including food, bait, use in the aquarium trade, and use in stocking private waters (Westers, 2001). Generally, public hatcheries focus on the “wild” qualities of the fish produced. Fish produced for enhancement purposes are produced to retain genetic integrity and characteristics needed to survive in the wild. On the other hand, most private hatcheries focus on maximum production to meet economic goals. Commercial producers emphasize genetic selection for fast growth and adaptation to culture conditions. These differences in goals are reflected in the variety of production strategies generally applied by public and private programs.

4.1.1 Development of Federal, State, and Local Hatchery Programs

Expansion continued and by 1949, 46 of the states counted a total of 522 hatcheries, while the federal system had 99 hatcheries in 43 states. At the same time, however, stocking programs came under scrutiny. Stocking of fingerling-size trout had replaced the early fry stocking programs, but even fingerling stocking, in most instances, produced dismal returns. At the same time, angling pressure increased. To meet angler demand many states launched into stocking catchable-size trout. It was now possible to feed the fish dry prepared diets, giving hatcheries the opportunity to greatly expand production in terms of biomass and numbers. This expansion required greater hatchery capacity.

Public fish hatcheries became extremely popular with the public at large, as many became favored places to visit. Such facilities became firmly entrenched in local communities, making it politically difficult to discontinue their operations. Hatcheries attracted not only tourists, but also people interested in sportfishing opportunities, especially the stocking of catchable trout.

Both state and federal governments established research facilities, which made significant contributions to the advancement of fish culture in the United States. State fish hatchery facilities made significant advances in developing of prepared feeds, identifying diseases and treatments, advancing engineering design for water systems, and identifying or developing methods to measure and control water quality (Stickney, 2000b).

In 1973, the Endangered Species Act became a catalyst for shifting the goals of some public hatcheries from stocking sport fish to propagating endangered and threatened species. Today the USFWS considers its primary responsibility to be fish resource restoration and maintenance, while the states’ responsibility is to supply fish for the enhancement of sportfishing opportunities. As a consequence, many federal hatchery facilities have either been closed or transferred to the state.

Currently, 28 USFWS hatcheries are involved in the restoration of threatened or endangered species. Maintaining the genetic integrity of these aquatic organisms is considered a high priority (Hartman and Preston, 2001). Despite this goal, 49 states use non-native sport fish species, and some states rely entirely on non-native species for recreational sportfishing (Schramm and Piper, 1995).

An example of a successful state hatchery program is the restoration of red drum in the Gulf of Mexico. The Texas Parks and Wildlife Department releases 20–30 million juvenile red drum fingerlings annually into coastal bays (Pennell et al., 2001). It has been estimated that since 1990, the abundance of red drum 1 to 5 years of age is double the population prior to the 1980s. The success of this fishery might also have been affected by the closing of the commercial fishery in 1981 when red drum was declared a game fish.

Both federal and state hatcheries serve as a tool for fisheries management to develop and maintain recreational, commercial, and tribal fisheries; supply year-classes to supplement natural reproduction; introduce new species; and restore endangered or threatened species.

Put-and-take stocking, or stocking that increases angler opportunities through the release of harvestable-size fish, is still an important activity in many state hatchery programs. State hatcheries stocked more than 60 million catchable trout in 1980. That same year the federal hatchery system released 11.9 million catchable trout. Coldwater fish make up the largest stocking program; 252 state and 36 federal hatcheries produce salmonids. More than 500 million coldwater fish are stocked annually, and according to Radonski and Martin (1986), this number falls 38 million short of the amount required to meet angler demands. Most salmonids stocked are Pacific salmon species released as smolts into various river systems connected to the Pacific Ocean. In the Columbia River Basin, more than 90 state and federal hatcheries raise and release some 190 million juvenile Pacific salmon annually (Schramm and Piper, 1995).

Total estimated production of all salmonid species for stocking in public waters is 35–42 million pounds annually. In terms of numbers of fish stocked on an annual basis, coolwater fish, including walleye, northern pike, and yellow perch, are the most abundant. At least 47 states have programs for stocking coolwater fish. In the 1983–1984 season, over 1 billion walleye fry were stocked in the United States, followed by 42 million northern pike and 13 million yellow perch. Finally, approximately 43 states have warmwater stocking programs for primarily largemouth bass. Other warmwater fish species stocked to restore or enhance fisheries are smallmouth bass, bluegill, sunfish, crappies, striped bass, hybrid striped bass, channel catfish, flathead catfish, and blue catfish (Smith and Reeves, 1986).

4.1.2 Development of Commercial Aquatic Animal Production

Commercial foodfish production in the United States began to grow in the 1960s. Before that time, AAP was generally limited to trout production. The trout industry in Idaho began to expand, as did production of warmwater species in southern states, particularly catfish production (Stickney, 2000b). Interest in commercial AAP gained popularity at several universities, including the University of Washington and Auburn University. The expansion of faculties' expertise and research activities in commercial AAP led to an increased body of knowledge about fish life cycles, production methods, and husbandry practices. Commercial AAP benefited from new research activities and strong university programs. In the mid-1980s, the U.S. Department of Agriculture (USDA) created five regional aquaculture centers that represent the Western, North Central, Southern, Northeastern, and Tropical/Subtropical Regions. Building on academic interest in commercial AAP and state and federal hatchery experiences, commercial foodfish production in the United States has grown over the past 30 years.

Idaho dominates trout production with cultured rainbow trout. Relying on cold spring water, the trout industry has been developed primarily around the Magic Valley region of Idaho using water from subterranean rivers (Stickney, 2000b). Initiated by research at USFWS laboratory facilities in Stuttgart, Arkansas, and in Marion, Alabama, channel catfish became the dominant species for production in the southern United States. Although the catfish industry was originally centered in Arkansas, falling water tables in the early 1970s limited expansion potential. Instead, catfish farmers moved to the Mississippi Delta region with its flat topography and shallow water table. Today Mississippi leads catfish production in the United States; however, catfish are produced in all southern states. Limited catfish production occurs in other states, such as California and Idaho. Though once considered of interest as food only in southern states, today the catfish industry has developed a national market through an aggressive marketing campaign (Stickney, 2000b). In addition to trout and catfish, other freshwater fish and shellfish are also raised commercially in the United States, including hybrid striped bass, tilapia, and crawfish.

Salmon production developed in the 1970s in Puget Sound, Washington, with the production of pan-sized coho. Research to expand net pen production originally focused on coho and chinook. Researchers, charged with maintaining threatened native stocks of Atlantic salmon in Maine, experimented with producing Atlantic salmon in the Pacific Northwest and discovered that Atlantic salmon were better suited for production in captivity than salmon species indigenous to the Pacific. Today, salmon culture continues to grow in Maine, whereas in Washington salmon production has leveled off and even declined in recent years. Salmon net pen culture is illegal in Oregon and Alaska; however, salmon ranching, the production of smolts for release and recapture as adults, is permitted.

Commercial marine fish production in the United States remains limited. A few commercial facilities produce red drum in Texas, and there are a few commercial operations for the production of summer flounder (Stickney, 2000b). Marine shrimp culture is well established in the United States; Texas is the leading producer. Some mollusc production, including oysters, mussels, and clams, occurs on the Atlantic, Gulf of Mexico, and Pacific coasts of the United States.

4.2 SYSTEM TYPES

4.2.1 Ponds Systems

4.2.1.1 Levee Ponds[1]

Regions of the United States with relatively flat land and sufficient clay in the soils are usually well suited for constructing levee ponds for producing aquatic animals. A levee pond is constructed by creating earthen levees from excess soil that is covering the future pond bottom. It can be constructed as a single unit or as a singular part of a group of ponds in which the levees often serve as common walls for more than one pond. The tops of levees are maintained, at least on one side, so that the operator can move equipment and vehicles along the pond bank for feeding and harvesting. Assistance in pond design and construction is sometimes available from local offices of the USDA's Natural Resources Conservation Service (NRCS).

Water supplies for levee ponds are typically wells, located on-site at a facility. Some facilities rely on pumped or free-flowing water from surface water bodies such as lakes, streams, or coastal waters. Those relying on surface waters, however, must be careful not to introduce undesirable species or organisms into the culture ponds. Water might need to be screened or filtered as it is pumped into the pond. Rainwater falling directly on the pond is also captured and can be a source for maintaining water levels. For those systems that rely on well water, water conservation and rainwater capture are important management tools to minimize pumping costs.

Like watershed ponds, the size and shape of levee ponds are determined by the available land, its topography, and its underlying soils. Levee pond size varies from less than 1 to more than 25 acres, but most ponds for foodfish production are 4 to 16 acres. Smaller ponds may be used for broodstock holding and fry or seed production because they are easier to manage for these purposes than larger ponds. Larger levee ponds are typically more difficult to manage and harvest than smaller ones, but they are more economical to construct. The average depth of a levee pond is about 4 to 5 feet.

Drainage structures on a levee pond have two functions. The first is to provide a conveyance for overflow, which regulates the water level in the pond. If a pond captures excessive rainfall, the overflow structure allows the excess water to drain before it overflows the levees that enclose the pond. In some pond facilities (e.g., baitfish facilities), overflow pipes connect the ponds so water can be transferred between adjacent ponds to conserve water.

The second function of drainage structures is to allow the complete draining of the pond. The drainpipe is located in one of the levee walls just below the grade of the pond bottom. Some ponds have a drainage structure that functions as both an overflow control and a drain. For example, the structure can be in the form of a standpipe that swivels or a

---

[1] Some of the information for this section was adapted from T. Wellborn and M. Brunson, *Construction of Levee-type Ponds for Fish Production*, publication no. 101 (Southern Regional Aquaculture Center, Stoneville, Mississippi, 1997).

riser structure. Other ponds have separate overflow pipes and drains. If the drain has a valve, the valve remains closed at all times until the pond is drained.

In catfish ponds, which represent more than half of the ponds in production in the United States, as well as other high-density production ponds such as ponds for hybrid striped bass and shrimp, the use of mechanical aeration is common throughout the growing season. Stationary mechanical aerators are strategically positioned in the pond to maintain sufficient dissolved oxygen (DO) levels throughout the entire pond. In the event of extreme low-oxygen conditions, supplemental emergency aeration might be required. Emergency aeration is usually provided by using tractor-driven mechanical aerators.

Fish harvest takes place using seines that can be stretched across the entire pond. The mesh size of the seine allows smaller fish to escape to be harvested at a later date. After being seined into a section of the pond, the fish are removed from the pond with a net attached to a scale and boom. After being simultaneously removed and weighed, the fish are loaded into live haul trucks for shipment to a processing facility.

Levee ponds are the most commonly used method of production for channel catfish. Hybrid striped bass and shrimp are also commonly grown in levee ponds. Any species amenable to pond culture can be grown in a levee pond; for example, crawfish, shrimp, baitfish, ornamentals, sport fish, and perch. The following are some examples of different production practices in levee ponds:

> *Channel catfish*. Channel catfish fingerlings are produced in nursery ponds, which are smaller than production ponds. Feed-trained fry are stocked into the ponds, usually in the spring of the year. These ponds are managed to ensure that plankton blooms are also available as a source of natural food until the fry become proficient at using the artificial diet as their sole source of food. Fingerlings are grown in the ponds for about 5 to 9 months and then harvested by seining during the colder seasons and transferred to growout ponds. The nursery pond is eventually drained, and any remaining fish are killed to prevent cannibalism of the fry by larger fish.
>
> Foodfish production varies among farms, but it can involve crops of single cohorts or multiple cohorts. For the single-cohort cropping system, fingerlings are stocked, grown to market size, and then harvested. The pond is cleaned of all fish (by draining or killing the remaining fish), and a new cohort is put into the pond to repeat the cycle. Multiple cohorts can be cropped by selectively harvesting larger fish and understocking with fingerlings. This approach allows the operator to use most of the water for many years between draining events.
>
> Both fingerlings and foodfish are typically fed with mechanical feeders that blow the feed across the surface of the pond. With respect to stocking density, producers usually try to achieve a maximum biomass of about 6,000 pounds/acre. Mechanical aeration is required to maintain adequate water quality and oxygen

levels in the ponds. Most catfish farmers use paddlewheel aerators to supply sufficient aeration for production.[2]

Penaeid shrimp. Levee ponds are also commonly used for the production of penaeid shrimp. The ponds are filled in the spring of each year, and the larval shrimp are stocked in the ponds. The shrimp are fed by broadcasting feed into the ponds with mechanical feeders or by hand feeding out of a boat criss-crossing the pond. Shrimp production ponds are also aerated to maintain sufficient levels of DO. After the shrimp are harvested in the fall, the ponds are drained and left to dry. This oxidizes the organic matter and reduces the likelihood of disease problems from growing season to growing season. Most shrimp facilities use surface water as a source and screen the inlets to prevent predators from entering the ponds. Because many of the shrimp grown in the United States are non-native species, escapement and disease are concerns for regulatory agencies. Outlets are screened to prevent escapement. Water is reused by draining it to ditches and pumping or conveying it back into the ponds from the ditches.

Crawfish. Levee ponds are also used in crawfish production. Managing crawfish production ponds is different from managing other pond production systems. Crawfish ponds are shallow, with an average depth of 18 to 24 inches. They are drained every spring to begin the reproduction process. As the water is drained from the ponds, the crawfish burrow into the pond bottom and produce their young. A forage crop is planted to provide food for the crawfish when the ponds are flooded in the fall; rice is a common forage crop. After the growing season, the rice is harvested, and the rice stubble is left in the field. The field is then flooded to a depth of about 1.5 feet. The crawfish come out of their burrows and feed on the decaying vegetation. Crawfish are harvested by using baited traps.

4.2.1.2 Watershed Ponds[3]

In much of the United States, watershed ponds are built to capture storm water runoff, which serves as the primary water supply for the pond. Although often not ideal for use as AAP ponds, watershed ponds can be constructed in hilly areas that are not suitable for levee ponds. Watershed ponds are constructed by building earthen dams, or levees, to trap water in a topographic depression within the landscape. Another construction technique uses two- or three-sided ponds that are constructed parallel to hills bordering creeks. Watershed ponds constructed for AAP may sometimes differ from those used as general farm ponds or those used to control large volumes of runoff from agricultural or other types of watersheds. The goal of AAP watershed pond site selection and construction is to have a pond that allows the owner ease of management and harvesting. The USDA's NRCS has design criteria for watershed ponds, and local offices often offer site-specific design assistance.

---

[2] Information adapted from C. Tucker, Channel Catfish Culture, in the *Encyclopedia of Aquaculture*, 2000. ed. R.R. Stickney, pp. 153-170. John Wiley and Sons, NY.

[3] Some of the information for this section was adapted from J. Jensen, *Watershed Fish Production Ponds: Site Selection and Construction*, publication no. 102 (Southern Regional Aquaculture Center, Stoneville, Mississippi, 1989).

Like levee ponds, the local topography determines the size and shape of watershed ponds constructed for AAP. On gently sloping or rolling landscapes, the watershed pond is sited and constructed to capture enough water to maintain adequate water levels throughout the year and to minimize the need for water sources other than runoff. On steeper slopes or if available land permits, one or more ponds can be constructed in series to capture larger volumes of runoff during rainy seasons. Another technique for steeply sloped terrain is to divert excess water around the watershed pond. The ponds are constructed with relatively flat bottoms for ease of harvest with seines. The levees are constructed with top widths that are sufficient to drive trucks and other farm equipment on, primarily for feeding and harvesting. Costs for watershed pond construction depend primarily on the amount of soil moved to create levees and smooth pond bottoms.

Depending on the contributing watershed, these ponds could be rather large (in excess of 20 acres). Experience has shown, however, that ponds smaller than 20 acres are easier to manage and harvest than larger ponds. Ponds that are too small (less than about 5 acres for foodfish production) also are not as desirable, especially from a harvesting perspective. Extra labor is required to harvest multiple small ponds to collect enough fish to make centralized processing efficient. The pond size is a function of the watershed, annual and seasonal rainfall, available land, and production goals. Pond depths are kept below 10 feet to facilitate harvesting, enhance aeration and mixing, and meet other pond management needs.

Drains are usually installed in the watershed pond to allow the operator to completely drain the pond when the production strategy requires draining. Watershed ponds are also equipped with overflow pipes to drain smaller volumes of excess water from the ponds during runoff events. The overflows may be piped to adjacent ponds that are constructed and operated in series. At sites in Alabama, for example, up to five watershed ponds were observed in series. A properly designed watershed pond also includes an emergency spillway, which is a low spot along a levee that is grassed and maintained to control runoff. The emergency spillway is sized according to expected runoff volumes, depending on local climatic conditions and the size of the watershed.

The quantity of water available from runoff events for a watershed pond depends on the size of the contributing watershed, frequency and duration of rainfall events, and land use characteristics of the watershed. These factors also greatly influence the quality of water entering the pond during rainfall events. Large watersheds typically collect more water than smaller ones and might present the opportunity for more pollutants to accompany the runoff into the ponds. The frequency and duration of rainfall events have obvious implications on the quantity of water available for the ponds and the amounts that might overflow. (Heavier and more frequent rainfall produces more water.) Watersheds with land uses like roads, houses, and agricultural cropland present different water quality inputs to watershed ponds. For example, roads contribute oil and other petroleum products, metals, and potentially large amounts of suspended solids to watershed ponds.

Management strategies for watershed ponds for AAP depend primarily on the size and type of fish. Watershed ponds are used primarily for the production of catfish, as well as other warmwater and coolwater species such as hybrid striped bass, sunfish, yellow perch, ornamental fish, baitfish, and many sport and game fish. The species and life stage (e.g., fry, fingerling, or food-sized fish) will determine relative densities and many management practices, as shown in the following examples:

> *Catfish food-sized fish.* These fish are often stocked to achieve maximum densities of about 5,000 to 6,000 pounds/acre. They can be harvested and understocked with smaller fish to maintain higher biomass and longer periods between draining; complete draining usually occurs once every 7 to 10 years. Ponds are aerated to maintain DO and water quality. Fish are fed once or twice daily with mechanical feeders.
>
> *Hybrid striped bass food-sized fish.* These fish are often stocked to achieve maximum densities of about 5,000 to 6,000 pounds/acre. They must be completely harvested before restocking. (The ponds are drained between harvest and stocking or are treated with a piscicide to remove remaining fish.) Ponds are usually drained annually or biennially, depending on stocking size, and are aerated to maintain DO and water quality. Fish are fed once or twice per day with mechanical feeders.
>
> *Baitfish.* Baitfish are often stocked to achieve a desired number of fish per acre to maintain size requirements at harvest. The overall densities are typically less than 300 to 500 pounds/acre. Ponds must be completely harvested before restocking, and they are usually drained annually for maintenance; aeration is used to assist in harvest. Fish are fed minimally to supplement natural food as well as provide nutrients to the pond for natural food production. They are fed by hand or with mechanical feeders. Feeding may also be used to concentrate the fish to facilitate harvesting.

4.2.2 Flow-through Systems[4]

Flow-through systems consist of single- or multiple-pass units with constantly flowing culture water, and they commonly use raceways or tanks (circular or rectangular). Raceways typically are long rectangular tanks constructed of earth, concrete, plastic, or metal. Sizes vary depending on topography and the operational goals of the facility. Some sizes commonly used are 80 feet long, 8 feet wide, and 2.5 feet deep (trout); 100 feet long, 10 feet wide, and 3 feet deep (trout and catfish); or a series of cells 30 feet long, 10 to 20 feet wide, and about 3 feet deep. Many raceways are constructed to reuse the flowing water several times by passing the water through multiple units before discharging it.

Circular or rectangular tanks are also used with constantly flowing water, and they are made from concrete, plastic, or metal. They can be above the ground or placed in the ground, and most use gravity to maintain flows. The primary difference between raceways and tanks is the flow pattern within the containment structure. Raceways tend to have plug flows of water along the length of the raceway. Tanks establish varying flow patterns, depending on the inlet and drain configurations, and the volume of water used.

---

[4] Information for this section was adapted from J. Avault, 1996a. *Fundamentals of Aquaculture* (AVA Publishing, Baton Rouge, Louisiana).

Circular tank systems are operated to enhance solids removal, while raceways allow settling of solids within a portion of the rearing unit.

Flow-through systems are found throughout the United States, wherever a consistent volume of water is available. Most flow-through systems use well, spring, or stream water as a source of production water. The water source is chosen to provide a constant flow with relatively little variation in rate, temperature, or quality.

Flow-through systems are the primary method used to grow salmonid species, such as rainbow trout. These species require high-quality coldwater with high levels of DO. Flow-though systems are located where water is abundant, which enables farmers to efficiently produce these types of fish. Some other species cultured using flow-through systems are hybrid striped bass, tilapia, and ornamentals.

Facility size for flow-through systems can vary tremendously. Facilities can range from small earthen or concrete raceway systems producing about 2,000 pounds of fish per year to much larger facilities with production levels in the millions of pounds per year.

Most flow-through systems require supplemental oxygen or aeration to maintain sufficient levels of DO in the culture water. The source water might require oxygenation to be suitable for production, or as water is reused in serial units, oxygenation or aeration might be required. In some cases, facilities use mechanical or passive aeration devices to increase the DO concentration of the culture water. Other facilities might add on-site generated or liquid oxygen to supplement DO levels.

Because many flow-through systems have relatively constant temperatures all year, the fish can be fed year-round. Feeding systems for flow-through systems vary significantly by size and management objectives. Small operators might choose to hand-feed all fish, use demand feeders in different areas of the production facility, or have a mechanical system to deliver feed to the different raceways. Large operators typically use some kind of mechanical feeding system to distribute feed at the desired intervals to meet production goals.

Flowing water in flow-through systems is expected to carry away accumulating waste products, including feces, uneaten feed, and other metabolic wastes. The flowing water and swimming fish help move solids down through the raceway. Raceway systems typically have quiescent zones at the tail ends of the raceways. The quiescent zones allow solids to settle in an area of the raceway that is screened off from the swimming fish. Baffles, or other solids-flushing enhancements, help move solids to the quiescent zones without breaking them into smaller particles. The settled solids are then regularly removed from the quiescent zone by vacuuming or gravity. Flow-through systems with tanks sometimes use self-cleaning or concentrating devices to collect solids and allow them to be efficiently removed from the system. Most facilities store the collected solids in settling basins, convey the solids to a dewatering process, or hold the solids in a storage tank for future disposal.

4.2.3 Recirculating Systems

Recirculating systems are highly intensive culture systems that actively filter and reuse water many times before it is discharged. These systems typically use tanks or raceways to hold the growing animals and have extensive filtration and support equipment to maintain adequate water quality. Recirculating systems use biological filtration equipment to remove ammonia from the production water. Solids removal, oxygenation, temperature control, pH management, carbon dioxide control, and disinfection are other common water treatment processes used in recirculating systems. The size of the recirculating system depends primarily on available capital to fund the project and can be designed to meet the production goals of the operator.

Recirculating systems can be used to grow a number of different species. They can be used anywhere in the country because a relatively small volume of water is needed to produce a unit of product. Thus, the facility can economically temper the water to optimal production temperatures. Recirculating systems grow various species of fish in controlled environments year-round. Species commonly grown in such systems include hybrid striped bass and tilapia.

Feeding regimes in recirculating systems vary significantly from operation to operation. Some operators feed by hand once or twice per day, whereas other operators use automatic feeders to feed the fish at specified intervals throughout the day.

The water treatment processes are designed to minimize water requirements, which leads to small-volume, concentrated waste streams. A typical recirculating facility has one or more discrete waste streams. Solids and backwash water removed from the production system create an effluent that is high in solids, nutrients, and biochemical oxygen demand (BOD). Most systems add make-up water (about 5% to 10% of the system volume each day) to dilute the production water and to compensate for evaporation and other losses. In addition, some overflow water, which is dilute compared to the solids water, is discharged.

Recirculating system facilities use a variety of methods to treat, hold, or dispose of the solids collected from the production water. Some facilities send the collected solids, and some overflow water, directly to a publicly owned treatment works (POTW) for treatment. Other facilities pretreat in settling ponds or other primary treatment systems to concentrate solids and send a more dilute effluent to the POTW. Still others concentrate solids and then land-apply the solids slurry when practical. The overflow water may be directly discharged, land-applied, or otherwise treated.

4.2.4 Net Pens and Cages

Net pens and cages are suspended or floating holding systems in which some cultured species are grown. These systems may be located along a shore or pier or may be anchored and floating offshore. Net pens and cages rely on tides, currents, and other natural water movement to provide a continual supply of high-quality water to the cultured animals. In most locations, net pens are designed to withstand the high-energy environments of open waters and are anchored to keep them in place during extreme weather events. Strict siting requirements typically restrict the number of units at a given site to ensure sufficient flushing to distribute wastes and prevent degradation of the bottom below and near the net pens.

Net pens use a floating structure to support nets, which are suspended under the structure in the water column. The net pens vary in shape but are typically circular, square, or rectangular on the water surface. Their size also varies, depending on the available surface area and depth. For example, a net pen facility that EPA visited in Maine had 10 adjoining square units, each with a surface area of about 250 square feet and a depth of about 40 feet.

A common practice in net pen culture is to use two nets—a containment net on the inside and an outer predator net to keep out predators, such as seals. The predator net also adds protection to minimize the risk of underwater escapement. At the surface, jump nets are used to keep fish from jumping out of the net pen. The jump nets extend several feet above the surface around the perimeter of the net pen. Bird nets are also suspended above the surface of the net pens to prevent bird predation. Cage culture uses floating cages or baskets that are usually much smaller than net pens. The shape of cages varies, and plastic and other corrosion-resistant materials are usually used to construct them.

For cage and net pen culture, the mesh size of the netting used to contain the fish should be large enough to prevent critically reduced water flows when fouling occurs, but small enough to keep the cultured fish inside the structure. Most nets and cages are cleaned mechanically with brushes and power washers. Antifoulants have limited use in the United States. A few have been approved for foodfish production, but those typically show minimal effectiveness.

Net pens and cages are used primarily in the coastal areas of the United States to grow anadromous or near-coastal species of finfish. The species most commonly cultured in net pen and cage operations are anadromous salmonid species like Atlantic salmon *(Salmo salar)*. Other Pacific salmon species, including pink (*Oncorhynchus gorbuscha*), chum (*Oncorhynchus keta*), chinook (*Oncorhynchus tshawytscha*), sockeye (*Oncorhynchus nerka*), and coho (*Oncorhynchus kisutch*), are either grown in net pens for part of their life cycle, prior to release into the open ocean for final growout, or grown to food-size (chinook and coho). Other species, such as steelhead trout (*Oncorhynchus mykiss*), cobia *(Rachycentron canadum),* and redfish (*Sciaenops ocellata),* also can be cultured in net pen operations.

Feeding practices include hand feeding and use of a variety of mechanical feeders. Operators of small cages with a low biomass of fish mostly rely on hand feeding, which necessitates placing the cages near shore with access from land, a dock, or a small boat. Most net pen systems contain a large biomass of fish (e.g., 30,000 fish with a harvest weight of about 8 to 10 pounds each) and require the use of mechanical feeders. For net pens that are single structures without supporting walkways, barges and boats with feed blowers are used to take feed to the net pens and dispense feed, usually once or twice a day. Bad weather can impede this method of feeding. Other facilities may use a stationary blower to deliver feed to each net pen in a group of pens. To control overfeeding, many facilities use underwater cameras to monitor feed consumption.

Most net pens are regularly inspected by divers. The divers look for holes in the nets, dead fish, and fouling problems. State regulatory programs require benthic monitoring at many net pen sites to ensure that degradation is not occurring under or around the net pens.

4.2.5 Floating and Bottom Culture Systems[5]

The production of bivalves in the United States involves several different methods, which are selected based on variables such as species, location, and legal or political issues. The commercial growout of bivalves always relies on naturally occurring foods that are present in the water in which the bivalves are placed. The key to successful floating and bottom culture is sufficient tides and currents to move water containing natural food to the shellfish. The water movement must also move wastes away from the growing shellfish and minimize the accumulation of sediment. Harvests can be made with divers, lifting gear, or conventional shellfishing techniques. The basic growout techniques use the intertidal areas above mean low water (but within the tidal reach) and the subtidal areas (areas always submerged). Those techniques can be further subdivided into techniques that use the bottom and those that use the water column. Some species are better suited for off-bottom culture (e.g., mussels); other species (e.g., clams and oysters) may be grown in either bottom or off-bottom growout systems. The specific locations of a growing area and that area's tidal characteristics (e.g., whether it is intertidal or subtidal) dictate the choice of intertidal versus subtidal growout. Other factors, such as legal restrictions, social pressure, waterway use, and aesthetics, might dictate the culture method.

One popular bottom culture technique places the shellfish directly on the bottom in beds. Clams tend to dig into the bottom substrate, while oysters and mussels remain on top of the substrate. When predation is a problem, the shellfish are placed in mesh bags or covered with mesh to keep the predators away from the growing crop. Bottom culture techniques require a relatively firm bottom to keep the shellfish from sinking too deep into the substrate. Bottom culture does not work when excessive sediment settles over the shellfish beds and smothers the crop. Shellfish can also be placed in trays, nets, or racks positioned directly on the bottom.

Off-bottom culture techniques include suspending shellfish from longlines on strings or racks. Longlines can also be used to suspend the shellfish in bags or racks. Floats are sometimes used to suspend strings, bags, or trays of shellfish in the water column. Racks of strings are a popular off-bottom method of growing mussels.

4.2.6 Other Systems: Alligator Farming

The only species of alligator commercially produced in the United States is the American alligator *(Alligator mississipiensis)*. Alligator production, which takes place primarily in Louisiana and Florida, is a relatively new business that is still undergoing many changes.

---

[5] The information for this section was adapted from J. Kraeuter, et al., 2000, Preliminary Response to EPA's Aquaculture Industry Regulatory Data Development Needs, Molluscan Shellfish Technical Subgroup.

Alligator production facilities usually consist of corrugated metal buildings constructed on top of concrete slabs with walls that form a tank. The buildings are insulated to reduce heating costs during the winter. To maintain the desired temperature, heated water is circulated through a piping network encased in the concrete floor. The drainage structures for alligator production facilities differ greatly from facility to facility, but most have a single drain for each alligator pen in the production area. These pen drains usually combine to form a main drain, which conveys wastewater to the wastewater treatment operations for the facility.

Alligator feeding regimes have changed significantly since alligator farming first began. Currently, most alligators are fed a manufactured diet consisting of pelleted feed with the same feedstocks used for finfish feeds.

Cleanliness of the growout areas is important to the production of high-quality skins for eventual sale. Most alligator pens are cleaned every other day using a high-pressure hot-water spray, sometimes combined with small amounts of bleach to reduce the risk of bacterial infection. Water drained from the growout areas is usually discharged to a singular treatment lagoon or a series of lagoons before it is land applied for its fertilizer value.

4.3 PRODUCTION DESCRIPTION BY SPECIES

4.3.1 Catfish

Representing nearly half of the total AAP in the United States for all species, production of channel catfish (*Ictalurus punctatus*) is the largest AAP enterprise in the country. In 2000, more than 656 million pounds of channel catfish were produced commercially. In 2001, sales increased to over 670 million pounds (USDA, 2002). Production is concentrated in the southeastern United States: Mississippi, Alabama, Arkansas, and Louisiana account for 97% of the total domestic catfish production (USDA, 2002). Catfish growers in 13 select states had sales of $443 million in 2001, down 12% from the previous year (USDA, 2002). Prices per pound dropped from $0.75 in 2000 to $0.65 in 2001.

The original range of channel catfish extended from northern Mexico through the states bordering the Gulf of Mexico and up the Mississippi River and its tributaries (Tucker, 2000). Today, the channel catfish can be found throughout the world as a sport fish and an AAP product. A native North American freshwater fish, the channel catfish is a bottom dweller with a preference for a substrate of sand and gravel. Its natural habitat is sluggish to moderately swift rivers and streams; however, channel catfish also thrive in ponds and lakes.

Between 1955 and 1965 most of the growth in commercial catfish culture occurred in southeast Arkansas. Farmers discovered that raising fish could be a profitable alternative to growing traditional crops like rice and cotton. By 1975, the industry began to expand quickly, particularly in Mississippi, where profits from traditional agriculture were in decline. Aquaculture offered farmers an opportunity to diversify their crop production and use land that did not successfully support row crops. Cooperation among farmers helped create the infrastructure needed to support catfish production, including the development of large feed mills and fish processing plants. In 1968, the creation of a national grower's association, the Catfish Farmers of America, also enhanced the growth of the industry. In 1986 the Catfish Institute, an association of catfish farmers, processors, and feed manufacturers, launched a national marketing campaign, further strengthening the industry.

Today most catfish farms are family farms or partnerships. According to the USDA, about 88% of catfish farms are small businesses with annual sales of less than $750,000 (USDA, 2000). Of the 1,370 catfish farms in the United States, 38% reported annual revenues of less than $25,000. Catfish production plays a significant role in the southeastern United States, a region that continues to be one of the more economically challenged regions in the country.

4.3.1.1 Production Systems

Facilities and culture practices vary within the southeast region. Many studies on catfish farming have focused on practices in northwest Mississippi (Tucker et al., 1996; Tucker and van der Ploeg, 1993) and west-central and central Alabama (Boyd et al., 2000; Schwartz and Boyd, 1994b). There are fewer studies on catfish farming practices in Louisiana and Arkansas, the other two leading producers of commercial catfish, or on practices in other states with catfish farms.

In the southeastern United States, the two major catfish-producing areas are (1) the Mississippi River Alluvial Valley, which includes northwest Mississippi, southeast Arkansas, and northeast Louisiana, and (2) west-central Alabama and east-central Mississippi (JSA, 2000a). Because of the flat topography and an available groundwater source, many catfish farms in the Mississippi River Alluvial Valley use levee (embankment) ponds. Levee ponds are built by removing dirt from the area that will become the pond bottom and using that dirt to build levees around the pond perimeter. In west-central Alabama and east-central Mississippi, some catfish farms use watershed ponds. Watershed ponds take advantage of hills and sloping terrain to build a pond by damming an existing drainage area to capture rainwater and runoff from the watershed. Many watershed ponds also require an additional source of water to supplement rainwater and runoff.

Overall, by operation size in acres, about 90% of all commercial catfish ponds in production in the United States are levee ponds; the remaining 10% are watershed ponds (USDA, 1997).

Levee Ponds

Ponds in northwest Mississippi are predominantly levee ponds. Most ponds are rectangular with about a 3:1 to 5:1 ratio of length to width with an average pond size of between 8 and 15 acres of water surface. For ease of harvest, most pond depths range from 3 to 5 feet. The height of the levee is 1 to 2 feet above normal water stage (freeboard and storage) (JSA, 2000a).

Watershed Ponds

In west-central Alabama and east-central Mississippi, commercial catfish farms use both levee and watershed ponds. The average size of ponds in this region is 10 to 12 acres. The average maximum depths are 7 feet at the pipe and 3 feet on the shallow end. The height of the levee for a watershed pond is around 3 feet above normal water stage. Watershed ponds can expect more input from rainwater and runoff because a larger natural watershed area drains into the pond. A levee pond has a smaller "watershed" contained within the slopes of the levee.

About 75% of the commercial catfish ponds in west-central Alabama are watershed ponds. The remaining 25% of the ponds in this region are levee ponds, filled with water pumped mainly from groundwater wells (JSA, 2000a). About half of the ponds in east-central Mississippi are watershed ponds, and the other half are levee ponds or hybrid watershed-levee ponds that primarily use water pumped from nearby streams or other surface water supplies rather than from groundwater supplies (JSA, 2000a).

4.3.1.2 Culture Practices

Catfish AAP in ponds involves four phases: (1) broodfish production, (2) hatchery production, (3) fry nursery production, and (4) growout production (JSA, 2000a). Broodfish are held in ponds and allowed to randomly mate each spring. Spawning occurs when the water temperature rises above 70 °F. Fertilized eggs are then taken to a hatchery, where they hatch under controlled conditions. The fry are raised in the hatchery for 5 to 15 days and are then transferred to a nursery pond, where they are fed a manufactured feed throughout the summer and fall. Fingerlings weighing 0.7 to 1.4 ounces are seined from the nursery pond and transferred to the foodfish growout ponds in winter or spring, where they are fed a manufactured feed until they reach the size desired for processing, usually 1 to 2 pounds. In the southeastern United States, 18 to 30 months (two or three growing seasons) are required to produce a food-size channel catfish from an egg (JSA, 2000a). Within the industry, some farmers specialize in producing fingerlings. The fingerlings are then sold to farmers who specialize in growing food-size fish. Many farmers combine all aspects of production by having broodfish ponds, a hatchery, fry nursery ponds, and growout ponds. In the catfish industry, fish are usually harvested from growout ponds with long seine nets pulled by tractor-powered reels. The fish are transferred to live-haul trucks in a basket connected to a crane. Using different mesh sizes, the seines are designed to capture market-sized fish and allow smaller fish to remain in the pond. The captured fish are then transported to processing plants or directly to market.

Broodfish ponds represent about 2% of the total pond area devoted to catfish production. Although some farmers harvest and drain broodfish ponds every fall to replace poor breeders and adjust the sex ratios, most broodfish ponds in northwest Mississippi are drained only every 1 to 5 years (Tucker, 1996). Instead of draining the pond every year, broodfish are inspected by seining the pond. In Alabama very few commercial hatcheries remain in operation (Boyd et al., 2000). Most fingerlings stocked in Alabama ponds are imported from Mississippi.

After a short stay in the hatchery, the fry are moved to a nursery pond for further growth. Nursery ponds are stocked with approximately 100,000 to 300,000 fry/acre. Because recently transferred fry are weak swimmers, farmers prepare a natural plant food source for fry that are too weak to swim to the areas where feed is offered (Tucker, 2000). After a month or so, as the fry approach 2 inches in length, they are referred to as fingerlings. Fingerlings ranging in age from 5 to 9 months and weighing 0.7 to 1.4 ounces are harvested from the nursery ponds and placed in growout ponds. The nursery ponds are harvested by seining each pond several times over 1 to 3 months The mesh size of the seine grades the fish by size, releasing smaller fingerlings back into the nursery pond for further development.

Nursery ponds are usually drained each year to remove all fish from the pond. Fingerlings are removed from the pond to prevent cannibalism of fry in the next cycle of fingerling production (Tucker, 2000). Nursery ponds represent approximately 10% of the total pond area in commercial production. Because these ponds are drained each year between crops, water use is higher in nursery ponds than in broodfish or foodfish growout ponds (Tucker and Hargreaves, 1998).

Broodfish and nursery pond practices remain fairly constant throughout the industry, but foodfish culture practices often vary among different farms based on production goals and the economics of different production strategies. There are two fundamental production variables in foodfish growout: fish stocking density and cropping system (Tucker and Robinson, 1990). Stocking densities in growout ponds range from 4,000 to more than 12,000 fish/acre and average about 6,000 fish/acre. The cropping system refers to the stocking-harvest-restocking schedule. The two cropping systems in commercial catfish production are clean harvest and understocking (or multiple-batch). In the clean harvest system, farmers keep only one year-class of fish in the pond at one time. Fingerlings are stocked and grown to the desired harvest size (1 to 2 pounds/fish). Faster-growing fish are selectively removed by seining the pond in two to four separate harvests over several months until all of the fish are removed. After the harvest, the pond is often restocked without draining in order to conserve water and to reduce time lost between crops (Tucker, 2000).

The understocking or multiple-batch system has more than one year-class of fish (with three or four distinct size-classes of fish) after the first year of production. Multiple-batch harvesting is the predominant production type, accounting for 89.2% of foodfish harvest (USDA, 1997). At first the pond is stocked with a single year-class of fingerlings. Faster-growing fish are selectively harvested using large-mesh seines, and fingerlings are added to replace the harvested fish. Most commercial catfish ponds in Alabama use multiple-batch systems and harvest with seines (Boyd et al., 2000). This process of selective harvest and understocking (adding fingerlings) continues for years without draining the pond. After several cycles, the pond contains several year-classes of fish with a range of sizes from recently stocked fingerlings to fish that might be several years old.

The clean harvest system produces fish more uniform in size than fish from understocked ponds, and processors prefer uniform sizes (Tucker and Robinson, 1990). Inventory records are also easier to keep with the clean harvest system because populations are reset at zero after each crop cycle. With the clean harvest system, feed conversion efficiencies are better because larger fish, which convert feed to flesh less efficiently, are not carried over into the next production cycle. The advantage of the understocking system is that more ponds will have market-size fish at any one time than with clean harvest crops. This is important because it provides a farmer with other harvest options if a pond is temporarily unacceptable for processing because of factors like algae-related off-flavors or ongoing losses due to infectious disease.

Water use practices have shifted in the catfish industry in recent years. Today farmers use water more conservatively. Before 1985, many catfish ponds in northwest Mississippi were regularly refilled with pumped water (Tucker and Hargreaves, 1998). Farmers believed that "flushing" the pond improved productivity. Research by McGee and Boyd (1983), however, showed that "flushing" was generally not beneficial. Today almost all catfish ponds in northwest Mississippi are managed as "static" systems with very little water exchange except from heavy rain creating overflow. In another study in Alabama (Seok et al., 1995), in a period of 3 years, three ponds were harvested annually by draining and three were harvested without draining. There were no differences in net production, average fish size at harvest, or feed conversion rates; however, in the undrained ponds, concentrations of chlorophyll *a* and total ammonia nitrogen were higher. This study has reinforced the practice of harvesting without draining, a management practice that is now common throughout the catfish industry.

Daily management practices for both crop systems are similar. Today, foodfish ponds are usually drained only when a levee needs to be repaired or when there is a need to adjust the inventory by completely removing all fish. Table 4.3–1 shows that most commercial ponds remain in production for 3 to 10 years between renovations before being drained, and the average time between pond drainings is over 6 years (USDA, 1997). On average, producers drained ponds less often (every 6.4 years) at operations where 90% or more of the ponds were levee ponds than at operations with a smaller percentage of levee ponds (every 4.7 years). Smaller operations (measured by acreage) drained ponds more often regardless of predominant pond type. During renovation the pond bottom is dried and the dried clay is broken by disking the bottom. Dried material is scraped from the bottom and used to rebuild the levee and restore the proper pond slope.

Table 4.3–1. Number of Years Between Drainings By Pond Type and Operation Size

Operation Size (Ac)	*Pond Type*[a]					
	Levee Ponds	*Standard Error*	*Watershed/ Mixture Ponds*	*Standard Error*	*All*	*Standard Error*
1–19	3.1	(± 0.4)	2.4	(± 0.5)	2.9	(± 0.3)
20–49	5.9	(± 0.5)	2.6	(± 0.8)	5.1	(± 0.5)
50–149	6.1	(± 0.3)	8.4	(± 1.7)	6.5	(± 0.4)
150 or more	8.7	(± 0.4)	9.7	(± 0.7)	8.8	(± 0.4)
All	6.4	(± 0.2)	4.7	(± 0.8)	6.1	(± 0.2)

[a] Pond type for the operation was classified levee if at least 10% of the operation's ponds were reported as levee ponds. Otherwise, the pond type was classified as "Watershed/Mixture."

Source: USDA, 1997.

Feed Management

Feed allowances in growout ponds average between 75 to 125 pounds/acre/day during late spring and early summer (Tucker, 2000). Feeding activity declines as water temperatures drop in late fall, with feeding rates declining to less than 25 pounds/acre/day during midwinter; however, feeding allowances may be higher during unusually mild winters. A report from the USDA's Animal and Plant Health Inspection Service (APHIS) found that 87.5% of operations with fish on hand during winter fed their foodfish during winter, with 62.8% feeding 3 or more days per month (USDA, 1997). Operators identified water temperature and levee condition as being very important criteria in determining winter feeding schedules.

The cost of feed depends on its quality and contents. The conversion of feed protein to fish protein is important because protein is the most expensive feed ingredient, based on the amount of protein in the feed and the cost of the protein used. In most catfish feed, a portion of the protein comes from fish meal and sometimes other animal sources. In recent years, the industry has improved upon earlier catfish feeds. Modern feeds contain less crude protein and a much smaller percentage of animal protein (Boyd and Tucker, 1995).

The feed conversion ratio (FCR) is a measure of the feeding efficiency. It is calculated as the ratio of the weight of feed applied to the weight of the fish produced:

$$\text{FCR} = \frac{\text{Dry weight of feed applied}}{\text{Wet weight of fish gained}}$$

Commercial catfish farms in Mississippi typically achieve an FCR of 2.04 to 2.40 (Boyd and Tucker, 1995). Much lower FCRs (in the 1.3 to 1.5 range) can be reached in research ponds under conditions where fish are less crowded, have less wasted food, and live in water with better aeration than is found on most commercial farms (Boyd and Tucker, 1995). The FCR is an important tool that operators use for measuring the efficiency of the system. If stocking rates are too low, efficient feeding becomes more difficult (fish are too spread out), and thus increasing the stocking density would improve FCR. When stocking and feeding rates are increased to the point where water quality is negatively impacted, however, FCR increases (poorer efficiency). As the growing season progresses, the fish grow and require more feed. As feeding rates increase, water quality tends to deteriorate as a result of excessive phytoplankton (microscopic algae), increased oxygen demand, and high concentrations of nutrients, including total ammonia nitrogen. In ponds that use the multiple-batch system, removing marketable fish and adding new fingerlings, the feeding rate might remain more constant because the number of pounds of foodfish per acre levels out as large fish are removed and small fish are added.

Health Management

High fish densities and stressful environmental conditions can lead to the outbreak and rapid spread of infectious diseases in channel catfish ponds. Bacterial diseases account for most of the losses of fingerlings in nursery ponds, whereas foodfish in growout ponds are most often affected by proliferative gill disease (PGD), caused by the myxosporean parasite, and "winter-kill syndrome," a disease associated with external fungal infections

(Tucker, 2000). PGD occurs most often in spring and autumn when temperatures are between 60 and 68 °F. There is no treatment for the disease, but farmers can reduce losses by maintaining high DO levels during an outbreak. "Winter-kill syndrome" is common when temperatures fall below 60 °F. Mortality rates from this fungal infection can be high, and the conditions that contribute to its outbreak are not well understood. There is no cost-effective treatment available for fungal infections in large commercial ponds.

The channel catfish virus (CCV) affects young catfish and can lead to large losses in hatcheries or nursery ponds. The virus causes channel catfish virus disease (CCVD), and fish less than 1 month old are most susceptible. There is no cure for CCVD, but losses can be reduced by controlling water temperature in hatcheries and reducing stress in fry or fingerling populations by maintaining relatively low stocking densities, avoiding stressful handling, and preventing adverse environmental conditions (Plumb, 1994a; Winton, 2001).

Three bacterial diseases are significant to channel catfish AAP because they can cause large losses: enteric septicemia of catfish, columnaris disease, and motile aeromonad septicemia.

Enteric septicemia in catfish (ESC) is one of the leading bacterial diseases in commercial catfish production. This disease costs the industry millions of dollars annually in fish mortalities and expenditures for preventive measures and therapeutic treatments (Plumb, 1994b; Winton, 2001). Only two Food and Drug Administration (FDA)-approved drugs, oxytetracycline (Terramycin) and sulfadimethoxine-ormetroprim (Romet), are effective against ESC. Today farmers rely more on vaccination and management practices to reduce stress to prevent ESC rather than drug treatments.

Two other bacterial diseases are often encountered in channel catfish production: motile *Aeromonas* septicemia (MAS), a ubiquitous disease of many freshwater fish species, and columnaris, caused by *Flexibacter columnaris*. MAS is typically caused by one of several gram-negative, motile bacteria that are members of the genus *Aeromonas*, such as *A. hydrophila*, *A. sobria*, and *A. cariae*. Occasionally, various species of *Pseudomonas*, especially *Pseudomonad fluorescen*s, can cause a form of disease that is indistinguishable from MAS (Winton, 2001).

Most columnaris infections in channel catfish are mixed infections with other bacteria, especially ESC and MAS. Initial columnaris infections are usually the result of mechanical or physiological injuries or environmental stress. MAS is also a stress-mediated disease. Treatment with a 1% to 3% salt solution or 2 to 4 milligrams/liter of potassium permanganate reduces the incidence of post-handling infections.

Infectious disease is a significant problem in catfish production that is primarily controlled by preventing the poor water quality conditions that lead to outbreaks. Pond culture of catfish prohibits the use of most drugs and pesticides for treatment because of the high cost of treating the large water volume. Sick fish tend not to eat, so the few FDA-approved medicated feeds are limited in their effectiveness.

Some algae and bacteria that grow in catfish ponds produce odorous organic compounds that can give the fish undesirable off-flavors. Synthesized by blue-green algae, geosmin, an earthy-smelling compound, and 2-methylisoborneol, which has a musty smell, are the two most common causes of off-flavors in pond-raised catfish (Tucker, 2000). To prevent off-flavored fish from reaching the market, fish are taste-tested before harvest. In Alabama it is a common practice to treat ponds with copper sulfate to control blue-green algae and off-flavor in ponds. Studies show that copper precipitates rapidly in ponds and is unlikely to be a concern in effluents (Boyd et al., 2000).

4.3.1.3 Water Quality Management and Effluent Treatment Practices

Water Quality in the Production System

In catfish ponds, the most important constituents of potential effluents are nitrogen, phosphorus, organic matter, and settleable solids (JSA, 2000a). These materials are a direct or indirect product of feeds added to the ponds to promote rapid fish growth. Farmers need relatively high stocking and feeding rates to reach profitable levels of production. Although catfish are able to convert more feed into flesh than warm-blooded animals, nutrient use is not as efficient. Less than 30% of the nitrogen and phosphorus added to the pond in feed is recovered in the harvested fish (JSA, 2000a). The remainder of the nutrient load stays in the pond system as fish waste. Inorganic nutrients in fish waste stimulate the growth of phytoplankton, which in turn stimulate the production of more organic matter through photosynthesis. For both watershed and levee ponds, nitrogen and phosphorus compounds and organic matter are present in the pond water throughout the growout period and represent potential pollutants if discharged.

Fish wastes contain nitrogen, phosphorus, and other nutrients required for plant growth. The input of these nutrients, particularly in the summer growing season, stimulates the growth of plant communities in catfish ponds. Although some ponds may develop rooted aquatic plants, the most common plant form is phytoplankton (Tucker, 1996). Phytoplankton are producers as well as users of oxygen. They also assimilate ammonia as a nitrogen source of growth (Tucker, 1996). Phytoplankton can be beneficial to the catfish pond system; however, a pond with high levels of phytoplankton biomass might use more oxygen than it produces, resulting in a community deficit of DO.

Catfish need sustained levels of DO. Ideally, minimum DO concentrations need to be between 4 and 5 milligrams/liter to maintain the health of the fish (Tucker, 1996). Aerators are one of the most common control technologies used in the catfish industry to improve water quality. Mechanical aerators improve the quality of the water in the pond by continually mixing the water and preventing thermal stratification. Aeration also adds DO to the system. By enhancing DO concentrations, aeration increases the capacity of ponds to assimilate organic matter through aerobic processes. Higher DO concentrations also increase the nitrification rate of ammonia to nitrate, which is then lost from the pond through denitrification. In addition, aeration and water circulation influence rates of phosphorus loss from the system. The interface between water and sediment in aerated ponds appears to be sufficiently oxidized to enhance rates of inorganic phosphorus removal from pond water and reduces the availability of phosphorus for phytoplankton (JSA, 2000a). Furthermore, circulation can also improve water quality by increasing nutrient uptake by phytoplankton. Water circulation increases the aggregate exposure of phytoplankton cells to light, resulting in an increase in phytoplankton growth rates, which in turn increases the nutrient uptake.

Over time natural processes in the pond lower the concentrations of nitrogen, phosphorus, and organic material. If water is retained in catfish ponds over a period of time, biological, chemical, and physical processes remove some of the waste generated by fish. Some of the organic matter from phytoplankton production and fish waste is oxidized in the natural process of microbial decomposition (JSA, 2000a). Total nitrogen levels in catfish pond waters are lowered as nitrogen is lost from the water column as organic matter with nitrogen particulates is decomposed on the bottom of the pond. Nitrogen is also lost from the water as a gas through denitrification and volatilization. Finally, total phosphorus concentrations in the water are lowered as phosphorus is lost to the pond bottom soils as particulate organic phosphorus and precipitates of calcium phosphates.

Effluent Characteristics

The major components of concern from catfish pond effluents are solids, organic matter, phosphorus, and nitrogen. Based on these components, the major potential impact on receiving waters is the possibility of eutrophication. The impact on the receiving waters will depend on the volume and concentration of substances in the effluent in relation to the flow rate of the receiving body of water and the timing of the effluent discharge (JSA, 2000a).

Watershed ponds and levee ponds, as well as the different production practices used by different facilities, influence water use practices and water quality in the ponds. In turn, water quantity and quality affect the discharge volume and the characteristics of the water discharged, or effluent, from catfish production. Effluent from a pond may be discharged intentionally. For example, a pond might be periodically drained for harvest or maintenance. Ponds might also discharge water though unplanned events, such as overflow due to excessive rainwater and runoff.

General characteristics of overflow from catfish ponds in northwest Mississippi are described in a study (Table 4.3–2) that examines long-term changes in the quality of effluents from typical commercial catfish ponds (Tucker et al., 1996). Water samples were taken from 20 ponds in Washington County in northwest Mississippi over a 2-year period beginning in summer 1991. These ponds represented typical culture practices of ponds used to produce catfish in the area. Samples were taken in August (summer), November (autumn), February (winter), and May (spring). Samples were collected from the top 12 inches of the surface of the pond and the bottom 12 inches of the pond at a site adjacent to the discharge pipe. Samples were taken at two different depths because water can be discharged from ponds at either the surface or the bottom, depending on the type of discharge pipe. Samples were analyzed for BOD, chemical oxygen demand, total ammonia, total nitrogen, nitrite, nitrate, total phosphorus, soluble reactive phosphorus, suspended solids, and settleable solids.

Table 4.3–2. Means and Ranges of Potential Effluents Parameters from 20 Commercial Channel Catfish Ponds in Northwest Mississippi from Summer 1991 Through Spring 1993

Season	*Settleable Solids (mL/L)*	*Suspended Solids (mg/L)*	*Total Nitrogen (mg N/L)*	*Total Ammonia (mg N/L)*	*Total Phosphorus (mg P/L)*	*Biochemical Oxygen Demand (mg O_2/L)*
Summer 1991	0.20 (0–0.90)	127 (40–225)	6.1 (2.1–14.1)	1.22 (0.01–3.19)	0.54 (0.23–1.24)	26.1 (14.6–41.2)
Autumn	0.02 (0–0.25)	80 (20–225)	6.1 (2.9–10.8)	2.63 (0.05–6.35)	0.26 (0.14–0.58)	9.7 (1.9–29.7)
Winter 1992	0.06 (0–0.70)	109 (51–194)	5.1 (2.1–8.8)	0.86 (0.04–3.85)	0.34 (0.13–0.62)	13.7 (5.7–20.3)
Spring	0.11 (0–1.35)	123 (72–204)	4.5 (1.8–6.7)	1.06 (0.04–3.04)	0.31 (0.15–0.56)	14.8 (8.2–27.1)
Summer	0.09 (0–0.58)	117 (47–175)	7.0 (2.6–10.9)	0.71 (0.03–2.02)	0.51 (0.26–0.87)	21.2 (10.5–36.4)
Autumn	0.02 (0–0.15)	93 (41–175)	6.9 (3.8–10.4)	2.76 (0.07–8.10)	0.35 (0.15–1.03)	12.3 (5.4–34.0)
Winter 1993	0.01 (0–0.03)	93 (39–165)	5.5 (0.6–8.8)	1.48 (0.02–5.14)	0.34 (0.14–0.62)	11.9 (4.8–22.9)
Spring	0.12 (0–0.70)	135 (46–289)	5.2 (1.5–7.9)	2.21 (0.03–4.44)	0.37 (0.24–0.58)	14.9 (8.5–25.5)

Source: Tucker et al., 1996.
Note: Ranges are in parentheses.

Pond effluents varied from pond to pond, season to season. Typically the quality of potential effluents was poorest in the summer, with high concentrations of solids, organic matter, total phosphorus, and total nitrogen. This same trend was confirmed by other studies of catfish pond water quality (e.g., Tucker and van der Ploeg, 1993).

Long-term changes in quality of effluents in typical commercial catfish ponds in central and west-central Alabama are described in a study (Table 4.3–3) by Schwartz and Boyd (1994b). They collected water samples during February, May, August, and November of 1991 and 1992 from 25 commercial catfish ponds using the same sampling method used in the study described above. Samples were analyzed for 5-day biochemical oxygen demand (BOD_5), total ammonia, total Kjeldahl nitrogen (TKN), total phosphorus, soluble reactive phosphorus, nitrite, nitrate, total ammonia, suspended solids, volatile solids, and settleable solids.

Table 4.3–3. Means and Ranges of Potential Effluent Parameters from 25 Commercial Channel Catfish Ponds in Central and West-Central Alabama from Winter 1991 Through Autumn 1992

Season	*Settleable Solids (mL/L)*	*Suspended Solids (mg/L)*	*Kjeldahl Nitrogen (mg N/L)*	*Total Ammonia (mg N/L)*	*Total Phosphorus (mg P/L)*	*Biochemical Oxygen Demand (mg O_2/L)*
Winter 1991	0.06 (0–0.33)	81 (22–202)	3.7 (0.9–9.2)	0.7 (0.07–2.47)	0.25 (0.04–0.57)	9.0 (1.2–21.9)
Spring	0.05 (0–0.40)	52 (5–134)	4.4 (1.8–10.6)	1.07 (0.02–3.45)	0.21 (0.07–0.37)	6.5 (2.4–21.4)
Summer	0.19 (0–1.80)	96 (14–240)	5.0 (1.7–11.3)	0.85 (0.05–4.71)	0.36 (0.12–0.75)	10.7 (4.3–20.3)
Autumn	0.03 (0–0.54)	103 (18–232)	6.1 (2.2–11.5)	1.86 (0.10–8.07)	0.46 (0.12–1.85)	18.1 (6.1–35.6)
Winter 1992	0.01 (0–0.10)	29 (1–100)	1.9 (0.6–3.7)	0.27 (0.03–1.08)	0.09 (0–0.31)	9.2 (5.5–17.5)
Summer	0.15 (0–0.28)	102 (10–308)	3.9 (1.6–8.4)	1.89 (0.06–3.30)	0.19 (0–0.47)	8.0 (1.4–15.9)
Autumn	0.03 (0–0.25)	73 (14–337)	6.0 (2.2–14.0)	1.91 (0.09–5.26)	0.27 (0.06–0.83)	7.6 (1.2–23.4)

Source: Schwartz and Boyd, 1994b.
Note: Ranges are in parentheses.

Settleable solid concentrations were highest during the summer and were generally greater in the surface waters. Phytoplankton were the major source of suspended solids in the samples. The other effluent parameters (e.g., suspended solids, TKN, BOD, total ammonia, and total phosphorus) generally cycle throughout the year. These effluent parameters are usually lower in the spring, increase through the summer, peak in the fall, and then decrease in the winter.

Overall, concentrations of settleable solids and suspended solids were similar in Alabama and Mississippi catfish ponds. Concentrations of Kjeldahl nitrogen in Alabama ponds and total nitrogen in Mississippi ponds are not directly comparable because of a difference in analytical methods; however, if nitrogen compounds not measured in the Kjeldahl analysis are accounted for, values for total nitrogen are probably similar in both studies. Concentrations for total phosphorus and BOD are somewhat higher in ponds in Mississippi. This is probably a result of the higher fish stocking and feeding rates commonly used in Mississippi, which might lead to higher standing crops of phytoplankton (Tucker et al., 1996).

Schwartz and Boyd (1994a) also conducted a study to describe the quality of effluents drained for harvest. This study was conducted in three watershed ponds at the Alabama Agricultural Experiment Station near Auburn. Ponds were stocked with 4,000 fingerling channel catfish per acre and were fed a pelleted commercial feed during the growing season and intermittently during the winter. This study showed that concentrations of TKN, BOD, and settleable solids were fairly constant throughout the draining phase. As the pond level was lowered and the seining phase began, these variables increased in concentration. Total ammonia nitrogen, soluble reactive phosphorus, and total phosphorus steadily increased during the draining phase and then sharply increased during the seining phase. Increases in phosphorus were likely a result of sediments being stirred up. The rise in total ammonia-nitrogen concentrations was likely a result of metabolic wastes, becoming more concentrated in a decreasing volume of water.

Draining a pond for harvest concentrates fish into a relatively small volume of water, causing sediments to be stirred up by the fish and the nets. Water discharged during harvest contains solids and other substances from the disturbed sediments and is, therefore, different from typical pond water (JSA, 2000a). The findings from this study suggest that the best way to minimize impacts from effluents from ponds drained for harvest is to harvest ponds as quickly as possible, and either to not discharge the water during the seining process or to discharge this highly contaminated water into a settling basin or retention pond (JSA, 2000a). As noted in the report prepared by the Technical Subgroup for Catfish Production in Ponds for the Joint Subcommittee on Aquaculture, most ponds are not drained for harvest (JSA, 2000a). Draining ponds for harvest is practiced mostly in watershed ponds that have deep areas near the dam that prevent harvest by seining. Watershed ponds are common in areas such as west-central Alabama and east-central Mississippi, but overall they constitute a small proportion of ponds used in catfish farming.

Current Industry Effluent Treatment Practices

In addition to natural processes in ponds that help improve water quality by reducing levels of organic material and concentrations of nitrogen and phosphorus, catfish farmers also play a role in improving in-pond water quality through best management practices (BMPs).

Effluent volume from levee ponds is lowered by two common management practices in the catfish industry. The practices, which include keeping the pond water level below the level of the drain and not draining water between crops, significantly reduce the volume of water discharged (JSA, 2000a).

As demonstrated in a study by Tucker et al. (1996), reuse of water for multiple crops results in significant savings in water use and also reduces overall effluent volume. This study modeled the effect of water reuse on mass discharge of nutrients and organic matter for levee ponds operated at three intervals (1, 3, and 5 years) between total pond drainings and managed with and without storage potential. Harvesting fish without draining the ponds between crops substantially reduced the average volume of water discharged each year, and the reduction was greatest when ponds were also managed to maintain storage potential. For ponds not managed to maintain surplus water storage, the model indicated that using the ponds for 5 years before draining reduced the annual average waste discharge by approximately 45% compared to annually drained ponds. When ponds were managed for surplus water storage, discharge of nutrients and organic matter was reduced relative to annually drained ponds by more than 60% when ponds were used for 5 years between drainings. Currently, the average time between production pond drainings is more than 6 years.

The following is a summary of common practices in the catfish industry and the ways in which they affect effluent quality.

> *Draining practices*. Draining practices are a function of harvest practices. Water is most commonly drained from a pond to facilitate harvests, prevent predation in fingerling ponds, or maintain pond banks and bottoms. Catfish production is characterized by infrequent drainings. Although nursery ponds are drained annually, growout ponds are drained once every 5 to 10 (or more) years. When the water is used for several years between draining events, effluent volumes are significantly reduced.
>
> *Harvest practices*. Fish raised in ponds are typically harvested using seines that can be stretched across the entire pond. Catfish are usually harvested with seine nets without draining the ponds. Some watershed ponds require partial draining before harvest to capture fish in the deeper end of the pond adjacent to the dam (Tucker et al., 2002). Ponds harvested without draining have reduced effluent volumes. Draining and seining also affect effluent pollutant loads.
>
> *Feed management*. Feed management is one of the most important practices that can influence water quality in the pond system. By managing feed, farmers manage the amount of nutrients in the form of fish waste and uneaten feed that are added to the pond system. Water quality in catfish ponds is directly related to the amount of feed added to the ponds. Uneaten feed contributes only to lowering of water quality, not to fish growth.
>
> *Water quality management*. Catfish need sustained levels of DO at 4.0 milligrams/liter or above. Most catfish farmers use paddlewheel aerators to supply sufficient aeration for production. Mechanical aeration is required to maintain adequate water quality and oxygen levels in the ponds. Mechanical aerators improve the quality of the water in the pond by continually mixing the water and preventing thermal stratification. Aeration also adds DO to the system. By enhancing DO concentrations, aeration increases the capacity of ponds to naturally assimilate organic matter through aerobic processes.
>
> *Overflow management*. Ponds can be managed to store precipitation and minimize the need for expensive pumped ground or surface water. The practice of preventing overflow by capturing rainwater is common throughout the catfish industry. By maintaining pond depths at 6 to 12 inches below the height of the overflow structure, about 160,000 to 325,000 gallons of storage capacity per surface acre of the pond is available to capture direct rainfall. When more water is stored, less water is released through overflows and smaller amounts of potential pollutants are released. Capturing rainfall and reducing the amount of overflow reduce the need for pumping additional water into a pond to compensate for water lost to evaporation and infiltration.

4.3.2 Trout

The production of trout represents the second largest sector of total AAP in the United States. In 2000, the total value of all trout sales, both fish and eggs, was $75.8 million

(USDA, 2001). Idaho leads trout production in the United States and accounted for 53% of the total value of trout sold in 2000. Pennsylvania, North Carolina, and California are the other leading trout-producing states. Trout distributed for restoration, conservation, and recreational purposes, primarily from state and federal hatcheries, had an estimated value of $60.9 million for both eggs and fish distributed.

Trout are cultured both for foodfish production and to stock recreational facilities. Rainbow trout (*Oncorhynchus mykiss*) is the most common species cultured for AAP; however, brown trout (*Salmo trutta*) and brook trout (*Salvelinus fontinalis*) are also raised in AAP facilities. Trout belong to the group of fishes called salmonids, which are coldwater fishes that also include Atlantic salmon and Pacific salmon. Rainbow trout were originally native to North American rivers draining into the Pacific Ocean. Brook trout are native to an area that extends from the northeastern coast of North America, west to the Great Lakes, and south through the Appalachian Mountains. The brown trout, a native of European waters, was first introduced into the United States more than 100 years ago. Because of their popularity as both a sport fish and a source of food, all three species of trout are now widely distributed and cultured around the world (Avault, 1996b.).

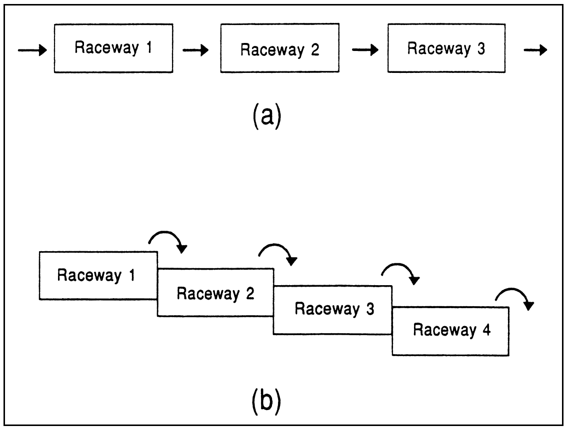
Rainbow trout culture became a farming business in the early 1900s, with a third of the farms operating as fee-fishing operations (Hardy et al., 2000). In Idaho, the first commercial trout farm was started in 1909 near Twin Falls. This area is known for its abundant spring water with a constant temperature from the Eastern Snake River Aquifer. In the early 1950s, trout farming expanded greatly, supported in part by the development of pelleted feeds. Farms no longer had to prepare their own feed, and production costs decreased. During the growth phase of the 1950s and 1960s, individual operators, including egg producers, growers, fish processors, distributors, and feed manufacturers, dominated the U.S. trout farming industry. Over the past decade, the industry has become more consolidated and vertically integrated. Today the most common trout farming businesses combine farming, processing, and sales. Egg production and feed manufacturing have remained specialized businesses.

Individuals and sport fisher groups originally began trout production to replenish wild stocks in natural waterways. These private hatcheries eventually evolved into the current state and federal hatchery system. State and federal hatcheries produce a number of species for restocking programs, while private commercial trout producers focus on food production of rainbow trout. Public hatcheries generally focus on the quality of the fish produced. Fish produced for enhancement purposes are produced to retain genetic integrity and characteristics needed to survive in the wild. Private hatcheries focus on maximum production to meet economic goals. Commercial producers emphasize genetic selection for fast growth and adaptation to culture conditions. These differences in goals are reflected in the different production strategies applied by public and private programs.

Trout production is the largest component of the inland stocking program. In 1982, some 200 million trout were stocked from more than 200 state and federal fish hatcheries, with states contributing roughly 80% of this total.

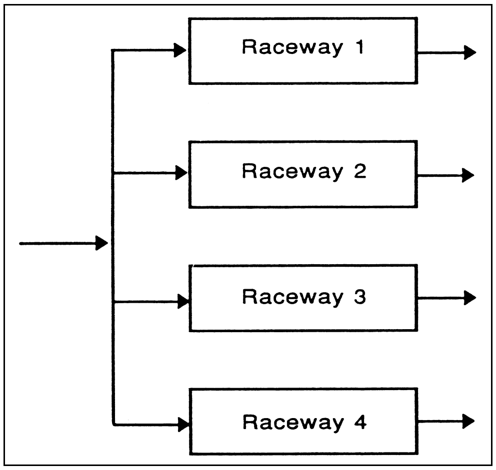
4.3.2.1 Production Systems

Most trout production facilities use flow-through systems. Flow-through systems are raceways, ponds, or tanks through which water flows continuously. Commonly, they are earthen or concrete rectangular troughs with varied dimensions and angles of pitch to allow a shallow stream of water to flow directly from one end to the other. The most common configuration for multiple raceways is either in series or in parallel. When constructed in series (Figure 4.3–1), water enters the upper raceway and then exits into a second raceway just downstream. This gravity-driven flow continues to the last raceway in the series. When raceways are constructed in parallel (Figure 4.3–2), the water source splits to flow through multiple raceways arranged parallel to each other. The water then exits the raceways into a common outflow pipe. Many large flow-through farms use a combination of the series and parallel configurations (Lawson, 1995a), shown in Figure 4.3–3. In North Carolina, raceways for trout production are typically 3 feet deep, 8 feet wide, and 40 to 60 feet long; most commercial facilities in North Carolina use concrete raceways (Dunning and Sloan, n.d.) In the southeastern United States, concrete raceways are also the most common rearing unit for commercial trout farms (Hinshaw, 2000). In Idaho the most common rearing unit is a concrete raceway with dimensions of 10 to 18 feet wide, 80 to 150 feet long, and 2.5 to 3.5 feet deep (IDEQ, n.d.).



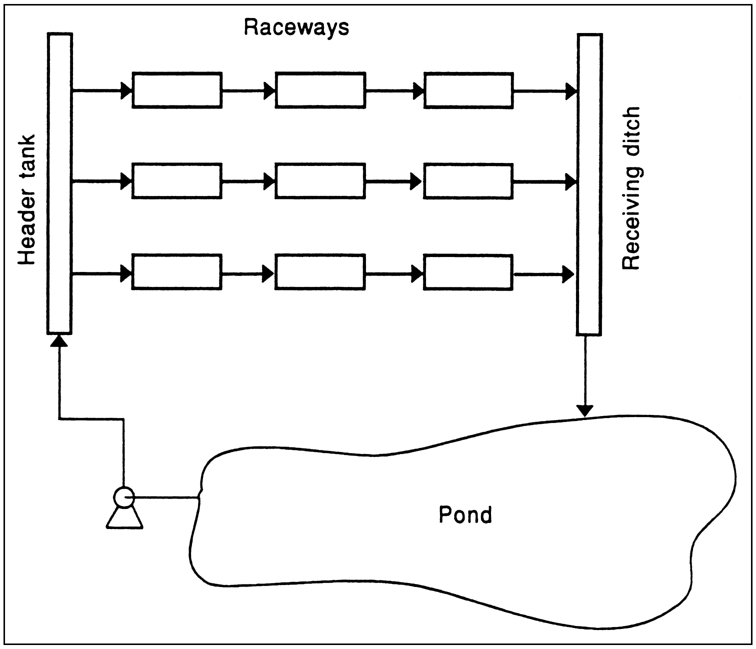
Source: Lawson, 1995a.

Figure 4.3–1. Raceway Units in Series (a) on Flat Ground and (b) on Sloping Ground



Source: Lawson, 1995a.

Figure 4.3–2. Raceway Units in Parallel



Source: Lawson, 1995a.

Figure 4.3–3. Combination Series and Parallel Raceway Units with Water Recirculation

4.3.2.2 Culture Practices

After fertilization and water-hardening, eggs are transported to incubation systems where they are incubated undisturbed until the eyed stage (about 14 days at a water temperature of 50 °F). Handling the eggs before the eyed stage damages and kills the sensitive embryos. There are several incubation methods for trout eggs. Eggs can be placed in wire baskets or rectangular trays suspended in existing hatchery troughs. Partitions between the trays force the water to flow up through the eggs from below before spilling over into the next compartment. Water is passed through the baskets or trays, and the newly hatched fry drop through the mesh to the bottom of the trough. The second method of incubation uses specially designed hatching jars placed in rows in hatchery troughs. The third method uses vertical flow incubators, which are widely used for trout eggs. Water is introduced at one end of the top tray and flows up through the screen bottom, circulating through the eggs. The water then spills over the tray below and is aerated as it drops.

Eggs hatch in the trays and remain there until they are ready to feed. Fungal growth can affect incubation. To prevent fungal growth, it is common to treat eggs with formalin (a 37% solution of formaldehyde) at a concentration of approximately 1 part formalin to 600 parts water for 15 minutes, every 1 to 3 days (Cain and Garling, 1993). Because of the specialized skill and labor involved in spawning, as well the high cost of maintaining broodstock, many trout farmers buy eggs for incubation rather than producing their own (Cain and Garling, 1993). In the North Central Region (Illinois, Indiana, Iowa, Kansas, Michigan, Minnesota, Missouri, Nebraska, North Dakota, Ohio, South Dakota, and Wisconsin), 92% of all purchased rainbow trout eggs come from outside the region, predominantly from western states. Farmers can also purchase fingerlings from hatching facilities that specialize in incubation and fry growout.

Trout emerge from eggs with a reserve of food in a yolk sac. At this stage, they are referred to as yolk-sac fry, or alevins, and they continue to live off and obtain nutrition from their yolks for approximately 20 days at 50 °F or 10 days or less at 60 °F (Hardy et al., 2000). When the fish begin to swim up to the surface, the thin yolk sac has been absorbed, and they begin to seek food actively. If incubation does not occur in a rearing trough, sac fry are transferred to a trough shortly after hatching. Troughs for raising fry are usually 12 to 16 feet long, 12 to 18 inches wide, and 9 to 12 inches deep. Fry are typically stocked at a rate of 1,000 to 2,000 fry per square foot of trough surface area. Flow rate and temperature also affect stocking rates. The water level in the fry trough should be kept shallow until the fish begin to “swim up.” When fry reach about 2 inches, they are ready for transfer to larger, deeper fingerling tanks. Fish are usually held and fed in fingerling tanks until they reach a length of about 3 inches, and then they are moved to outdoor raceways for final growout.

The maximum amount of fish in pounds that a volume of water in a raceway can support is referred to as the carrying capacity. The carrying capacity of a culture unit depends on water flow rate, water volume, water temperature, DO concentration, pH, and fish size. From the time fingerlings (about 3 inches) are stocked in raceways until they reach marketable size (12 to 16 inches), they must be graded periodically to sort the fish into similar size groups and improve feeding efficiency. Trout are typically graded four times during a production cycle. Using a rectangular frame with evenly spaced bars of aluminum tubing, PVC pipe, or wooden dowels, the grader is placed in the inflow end of the raceway and moved toward the outflow end. This crowds larger fish in the outflow area so they can be removed and stocked in another raceway with fish of similar size.

Trout are harvested by using a bar grader as described above. As the fish are crowded into a small area of the raceway, they are dipped out with a hand net or a combination of a hand net and fish pump. The ease of harvesting fish from raceways makes this type of rearing unit very popular for flow-through systems. Round tanks use crowding screens specifically designed for the tank.

Feed Management

Early life stages such as fry are usually hand fed. Fry need many regular feedings throughout the day; they are often observed and fed only what they can consume in a short amount of time to prevent overfeeding. Fish in production raceways may be fed with mechanical feeders or demand feeders (IDEQ, n.d.). Mechanical feeders typically deliver a predetermined amount of feed to the fish. Commercial feeder designs range from stationary units to truck-mounted units. Automatic designs, like spring-loaded belts or auger-driven feeders, deliver small amounts of feed at any one time. This method restricts fish to a set amount of food each day. Demand feeders allow fish to feed to satiation. This method allows fish to choose how much feed is needed and when feed is released. Fish activate the suspended feeder, dispensing small amounts of feed, by bumping a rod that extends to the water.

In the United States, consumers expect trout to have white meat, so they are fed diets lacking the carotenoid pigments that give trout and salmon fillets their typically red color (Hardy et al., 2000). In nature, these pigments are present in their food through natural sources such as krill, yeast, or algae, or through astaxanthin, the carotenoid pigment found in the wild, produced by chemical synthesis. In Europe and Chile, trout are expected to have pigmented meat, so the feed for these fish is supplemented with astaxanthin.

Feed, including its manufacture, storage, and delivery to the fish, is one of the most important aspects of trout AAP waste management (IDEQ, n.d.). Research by Boardman et al. (1998) showed that using high-energy feed may reduce the amount of solids leaving the system. The study showed that effluents of basins receiving standard trout grower feed generally contained higher levels of total suspended solids (TSS) than those receiving high-energy feed. Further analysis showed that effluents of basins receiving the standard grower trout feed had lower levels of TKN than those receiving a high-energy feed.

Health Management

Bacterial gill disease (BGD) is one of the most common diseases of cultured trout (Piper et al., 1982). Sudden lack of appetite, orientation in rows against the water current, lethargy, and riding high in the water are typical signs of BGD. Crowding, mud and silt in the water supply, and dusty starter diets are stress factors that contribute to outbreaks of the disease. The most important factor contributing to BGD is the accumulation of fish metabolic wastes due to crowding. To treat the disease, facility operators correct unfavorable water conditions, reduce stress, and use constant flow treatments with salt (NaCl), or Chloramine-T at 8 to 10 milligrams/liter (under an FDA-sponsored Investigational New Animal Drug (INAD) application) for 1 hour for 2 or 3 days. Furunculosis, another common bacterial fish disease, is generally considered a disease of salmonids. Once an infected population of trout has overcome the disease, some of the survivors become carriers. Stress and poor water quality conditions can reduce the resistance of fish, and carrier fish can experience chronic or acute infections. Healthy rearing conditions, sanitation, and use of pathogen-free fish help control furunculosis. If the bacterium is sensitive to Terramycin (oxytetracycline), facility operators can use medicated feed. Facilities may also use Romet-30. Vaccination against furunculosis can also be effective (Plumb, 1994c).

Fish health management in rainbow trout farming is based on prevention; once a disease outbreak occurs, it is difficult to treat or control (Hardy et al., 2000). Farmers keep raceways clean, use high-quality feed, prevent overcrowding, minimize disease vectors, and vaccinate stocks. Vaccination has been very effective in preventing some important diseases in rainbow trout (Hardy et al., 2000). Birds are a common disease vector because they move from farm to farm and eat diseased fish. Most farms in Idaho use netting to restrict birds' access to trout raceways. Use of antibiotics delivered in feed to treat rainbow trout is not a common practice. Antibiotic use is limited by cost and by the regulation of their use in trout farming. Only two antibiotics (Terramycin and Romet-30) have been approved for use in the United States for fish, and they are not typically effective against many trout diseases. According to reports from site visits conducted by EPA, several trout production facilities in Idaho use vaccination programs to prevent disease rather than treating sick fish with antibiotics (Tetra Tech, 2002a; Tetra Tech, 2002b).

4.3.2.3 Water Quality Management and Current Treatment Practices

Water Quality Management Practices

Flow-through systems require large inputs of high-quality, oxygenated water. In the trout culture industries in the northeast and northwest United States, freshwater springs are the most common source of water because of their relatively low and constant water temperatures (Lawson, 1995b). Water supplies may also come from surface waters such as streams, rivers, and irrigation returns. In western North Carolina, most water supplies come from surface waters that have been diverted for use by the facility (Tetra Tech, 2002a).

Concrete raceways have the advantage that there is no erosion of the sides, as happens with earthen ponds or raceways. This also means that these raceways can be operated at higher flow rates. The water flowing in delivers the needed oxygen to the fish while carrying away the dissolved metabolic waste products as the water exits the pond, or they are passed on to the pond below if raceways are positioned in series. These metabolic waste components must be kept within safe concentrations for the fish being raised. Concentrations of un-ionized ammonia-nitrogen need to be controlled to limit the impacts of this highly toxic compound.

DO is another important limiting factor in flow-through systems. These systems often use gravity aerators to supplement the oxygen supply. Gravity aerators are often called waterfall aerators or cascades (Lawson, 1995c). They use the energy released when water loses altitude to transfer oxygen. Based on local topography, if a sufficient gradient exists, gravity fall is a common method for aerating flow-through systems. Man-made gravity aerators include components such as weirs, splashboards, lattices, or screens, which break up water to increase surface area and oxygen transfer. For example, facilities may use a combination of splashboards and weirs between raceways to create gravity aerators. Aeration or oxygenation can minimize the impact of DO as a factor limiting production. The greater the flow of water through the raceway, the more oxygen is delivered and the more fish can be supported.

In a study conducted by Boardman et al. (1998), three trout farms in Virginia were selected to represent fish farms throughout Virginia (Table 4.3–4). Sampling and monitoring (Table 4.3–5) at all three sites revealed that little change in water quality between influents and effluents occurred during normal conditions at each facility. Raceway water quality, however, declined during heavy facility activity like feeding, harvesting, and cleaning. During a 5-day intensive study, high TSS values were correlated with feeding events. TKN and ortho-phosphate (OP) concentrations also increased during feeding and harvesting activities. Overall, most samples taken during this study had relatively low solids concentrations, but high flows through these facilities increased the total mass loadings.

Table 4.3–4. Site Characteristics of Trout Farms

Characteristic	***FARM***		
	A	***B***	***C***
Average production (lb/yr)	59,965–80,027	59,965	175,045–250,002
Fish type	Rainbow, brook	Rainbow	Rainbow, brook, brown
# Raceways in use (total #)	3 (7)	14 (14)	24 (31)
Feeding practice	Automated (pull string)	Hand (measured)	Hand (measured)
Reported feed conversion ratios (FCRs)	1.6	1.6–2	1.2–1.8
Concrete/earthen-lined	Concrete	Both	Both
Water source	Spring	Spring	Spring
Labor	1 person	1 person	4–6 people
Pollutants regulated	TSS, NH_3-N, SS	TSS, BOD_5, SS	TSS, BOD_5, NH_3-N, SS
Treatments	Sediment traps	None	Sediment traps

Source: Boardman et al., 1998.

Table 4.3–5. Water Quality Data

Parameter	***FARM A***			***FARM B***			***FARM C***		
	Inlet	***Within Farm***	***Outlet***	***Inlet***	***Within Farm***	***Outlet***	***Inlet***	***Within Farm***	***Outlet***
Flow (mgd)	1.03–1.54 (1.18)			4.26–9.43 (6.39)			9.74–10.99 (10.54)		
DO (mg/L)	9.2–14.2 (10.6	3.2–13.3 (7.0)	5.7–9.5 (8.5)	8.2–11.5 (10.5)	5.8–10.8 (8.6)	6.8–9.6 (7.9)	9.4–10.6 (10.5)	4.8–9.7 (7.6)	7.2–9.4 (8.1)
Temp (ºC)	10.5–13 (12.2)	11.5–15 (13)	11–15.5 (12.9)	6–12.5 (9.7)	6–14 (9.1)	5–16.5 (11.4)	8.5–13.5 (10.5)	8–14 (11.0)	8.5–14 (10.4)
pH (SU)	7.1–7.4 (7.3)	7.0–7.4 (7.2)	7.3–7.8 (7.5)	7.3–7.6 (7.5)	7.2–7.6 (7.4)	6.9	7.3	7.1–7.6 (7.3)	7.8
TSS (mg/L)	0–1.1 (0.2)	0–30.4 (3.9)	0.8–6 (3.2)	0–1.8 (0.5)	0–43.7 (5.3)	1.5–7.5 (3.9)	0–1.5 (0.3)	0–28 (7.1)	4.1–62 (6.1)[a]

Parameter	***FARM A***			***FARM B***			***FARM C***		
	Inlet	***Within Farm***	***Outlet***	***Inlet***	***Within Farm***	***Outlet***	***Inlet***	***Within Farm***	***Outlet***
SS (ml/l)	ND[b]		0–0.04 (0.02)	ND		0.01–0.08 (0.04)	ND		0.04–0.08 (0.07)
BOD_5 (mg/L)	0–1.25 (0.7)	0.5–3.9 (1.5)	0.96–1.9 (1.3)	0–1.4 (0.5)	0.3–7.2 (2.1)	0.6–2.4 (1.2)	0–2.0 (1.1)	0.4–7.5 (2.5)	0.5–1.8 (1.3)
DOC (mg/L)	0.93–4.11 (2.1)	0.9–7.9 (2.9)	1.5–2.4 (1.9)	0.91–2.56 (1.6)	1.2–8.1 (2.7)	1.2–3.1 (1.9)	1.1–2.7 (2.0)	1.1–11.1 (2.4)	1.5–3.8 (2.3)
NH_3-N (mg/L)	0.6	0.2–1.1 (0.5)	0.5–0.6 (0.6)	0.2	0.06–1.1 (0.5)	0.45	0.03	0.03–2.2 (0.4)	0.02–0.17 (0.1)

[a] Two outliers were not included in the calculation of mean.

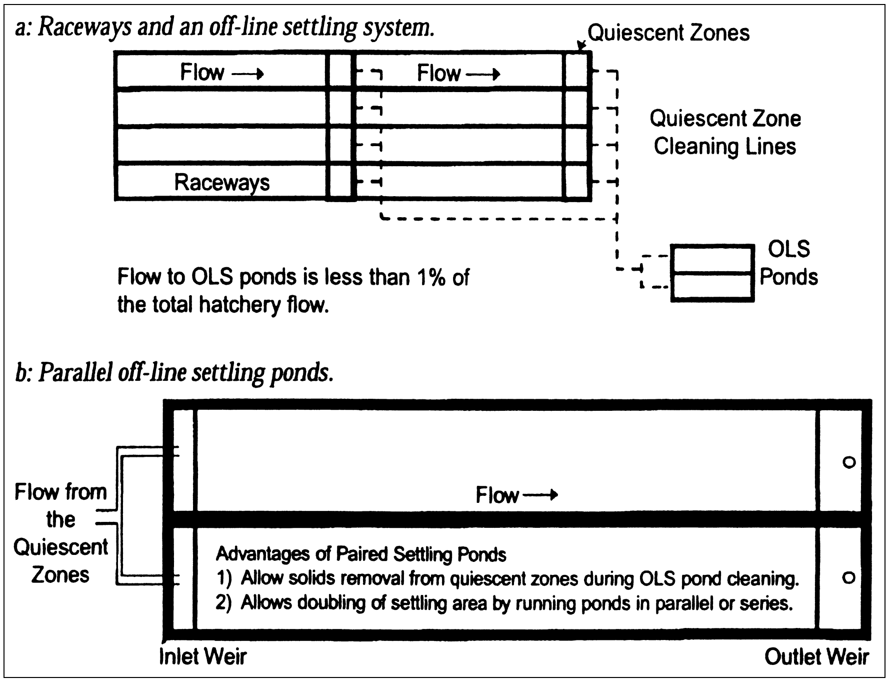
[b] ND: Non-detect

Source: Boardman et al., 1998.

Note: Averages are in parentheses.

Quiescent zones are the primary areas where solids are collected in a raceway. These zones are downstream of the rearing area, without fish, which allows bio-solids to settle undisturbed while intact and large in size (IDEQ, n.d.). Typically, quiescent zones are part of each trough or raceway; their dimensions account for the settling velocity of particles. The swimming activity of larger fish helps move solids downstream into settling zones. The most common method of solids removal from quiescent zones is through a vacuum head (IDEQ, n.d.). Usually, standpipes in each quiescent zone connect to a common 4- to 8-inch PVC pipe, which carries the slurry of water and solids to the offline destination. In *Idaho Waste Management Guidelines for Aquaculture Operations* (IDEQ, n.d.), the state recommends cleaning quiescent zones as often as possible, with a minimum of twice per month on lower raceway sets and once per month on upper raceway sets. Last-use quiescent zones should be cleaned most frequently.

Offline settling (OLS) ponds are settling zones that receive the water and solids slurry from the quiescent zones (Figure 4.3–4). These ponds can be earthen or concrete and are the second settling zone in the solids collection system. Quiescent zones, in combination with OLS ponds, are the most commonly used solids collection and removal system for trout farming in Idaho (IDEQ, n.d.). Flow to OLS ponds is usually very small when compared to the total facility flow. OLS pond effluent is typically less than 1.5% of the total flow during daytime working hours and less than 0.75% averaged over 24 hours. The depth of a typical OLS pond is 3.5 feet, but some are deeper. Depth is not required for settling efficiency but is required for solids storage. The Idaho Department of Environmental Quality recommends that, at a minimum, OLS ponds should be cleaned every 6 months In Idaho most trout production operators remove the solids from OLS ponds when TSS levels approach 100 milligrams/liter. Many facilities in the state have several OLS ponds, which are linked together to improve solids collection. When one pond is undergoing solids harvest, the other is receiving solids from the quiescent zones. To remove the solids, the inflow is diverted to another OLS pond, and the supernate from



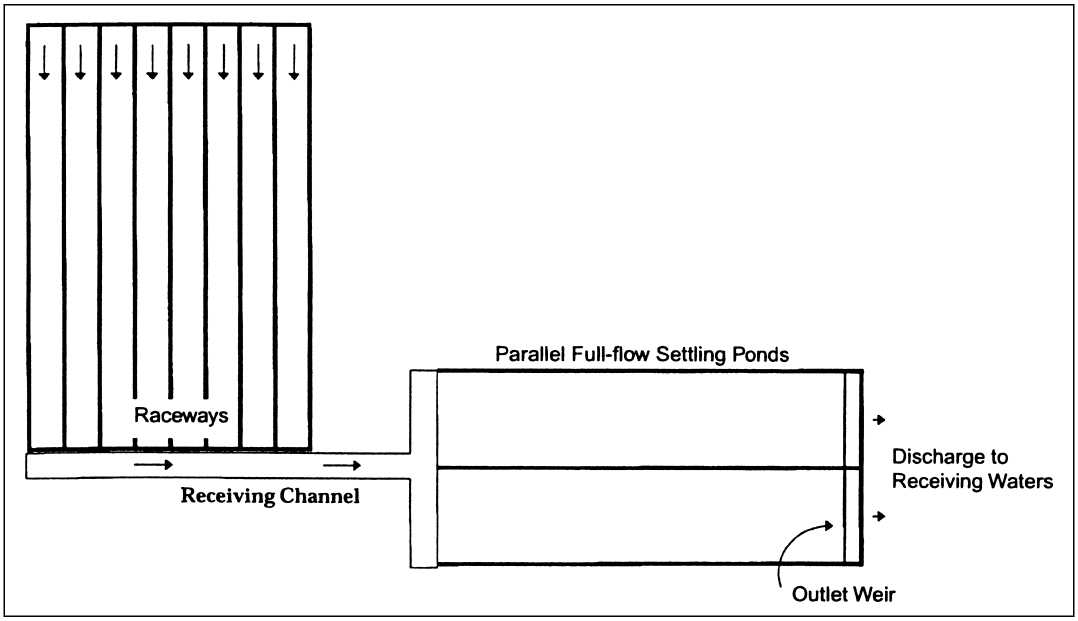
Source: IDEQ, n.d.

Figure 4.3–4. Offline Settling Ponds

the pond being harvested is moved to an adjacent pond. Earthen ponds are allowed to dry for a few days, and the solids are removed by a backhoe from the pond bank. In a concrete pond, the OLS pond has a ramp where a front-end loader can enter the pond to remove solids.

Some trout facilities use full-flow settling (FFS) ponds (Figure 4.3–5), which may not include quiescent zones or OLS ponds. The FFS system has one or two large settling zones, which collect the solids from the water flow for the entire facility. Instead of removing solids from individual raceways or troughs, the water from all of the rearing units combines and enters the FFS pond, where the solids are collected. FFS ponds are typically used by smaller facilities with low flow volumes.

In the study of Virginia trout farms by Boardman et al. (1998), waste solid accumulations in quiescent zones were monitored to quantify the capacity and trapping efficiency of the units. Solids were found to accumulate at a rapid rate (more than 7,800 centimeters/day or 256 feet/day); however, the trapping efficiency of the units was found to be extremely low when taking into account the FCRs and typical utilization rates of production fish. High overflow rates, particle degradation, flow spikes, and high sludge banks led to scouring of waste solids and a point of maximum capacity for the sediment trap.



Source: IDEQ, n.d.

Figure 4.3–5. Use of Full-Flow Settling Ponds to Treat 100% of the Flow From the Fish Farm Before it is Discharged

Sludge Treatment and Disposal

Once solids are removed from OLS ponds or FFS ponds, they are stored or used in ways that minimize their impact on groundwater or surface waters. In Idaho, land application of collected solids to cropland has become the easiest and most widely adopted technique to dispose of wastes and recycle nutrients from trout production settling ponds (IDEQ, n.d.). Regulations vary from state to state, but most allow for aquacultural solid wastes to be applied to land because of minimal concentrations of metals, pathogens, and toxic substances in the sludge. The rate at which sludge may be applied to land varies based on soil type, plant type, odor issues, and sludge nutrient content.

Composting is another popular sludge disposal and treatment option (Boardman et al., 1998). When large areas of land are not available for land application or transportation costs for disposal are high, composting represents a good alternative (IDEQ, n.d.). Because of high costs, landfills are one of the least common means of disposing of solid wastes from concentrated aquatic animal production (CAAP) facilities; however, some states are required to take their sludge to a landfill, where the states regulate the waste as industrial, rather than agricultural, waste (Boardman et al., 1998).

4.3.3 Salmon

Two distinct sectors influence salmon AAP: production for foodfish and production for stocking to restore wild stocks for conservation and recreation. In the United States, private salmon farming for foodfish production began in Washington state in the early 1970s with farms producing pan-sized coho salmon (*Oncorhynchus kisutch*) in marine net pens (Roberts and Hardy, 2000).

Public hatchery stocking programs are dominated by production of coldwater fish (salmonids). Most salmonids stocked in the United States are Pacific salmon released as smolts into various river systems connected to the Pacific Ocean. In the Columbia River Basin, more than 90 state and federal hatcheries raise and release roughly 190 million juvenile Pacific salmon annually (Schramm and Piper, 1995).

Atlantic salmon dominates commercial production in the United States. Although salmon was traditionally sold smoked or canned, today most salmon is sold frozen or fresh. According to the 1998 Census of Aquaculture (USDA, 2000), 45 farms produced salmon commercially in the United States, producing more than 110 million pounds in food-size fish. In 1998 the salmon AAP sector generated more than $103 million in revenue (USDA, 2000). The 1998 Census of Aqauculture data show that three states, Alaska with 19 farms, Maine with 12 farms, and Washington with 9 farms, are the largest producers of salmon in the United States (USDA, 2000). Alaska, which prohibits private farming of all fish species, has 19 salmon hatcheries that are operated as private nonprofit corporations. They raise smolts and release them into the wild, where they are later harvested from the ocean in a practice called ocean ranching.

Both Atlantic and Pacific salmon belong to the Salmonidae family, which also includes trout and whitefish. Atlantic salmon has its own genus, *Salmo*, while the five primary species of Pacific salmon belong to the genus *Oncorhynchus*. In the United States, there are five species of Pacific salmon: pink (*O. gorbuscha*), chum (*O. keta*), sockeye (*O. nerka*), chinook (*O. tshawytscha*), and coho (*O. kisutch*).

Wild salmon begin their life cycle as eggs in the gravel of cold, freshwater rivers and streams. When females reach freshwater spawning grounds, they use their caudal fin to excavate a nest, or redd, in the gravel riverbed. Females deposit their eggs in layers as they are fertilized by the male salmon. The female covers the eggs with gravel and guards the nest for up to 2 weeks. In 2 to 6 months, the eggs hatch into translucent hatchlings called alevins and obtain nutrition from their yolk sacs. After 3 to 4 months, the inch-long salmon fry emerge from the gravel and begin foraging for food in the river. As the fry grow into fingerlings, they move to a lake to mature as fingerlings before smoltification. Chum and pink salmon spend little time (1 to 3 months) in freshwater before moving to sea. Chinook begin to move to sea within 6 months, while coho usually stay in freshwater for up to 1 year, and sockeye salmon stay in freshwater for 1 to 3 years.

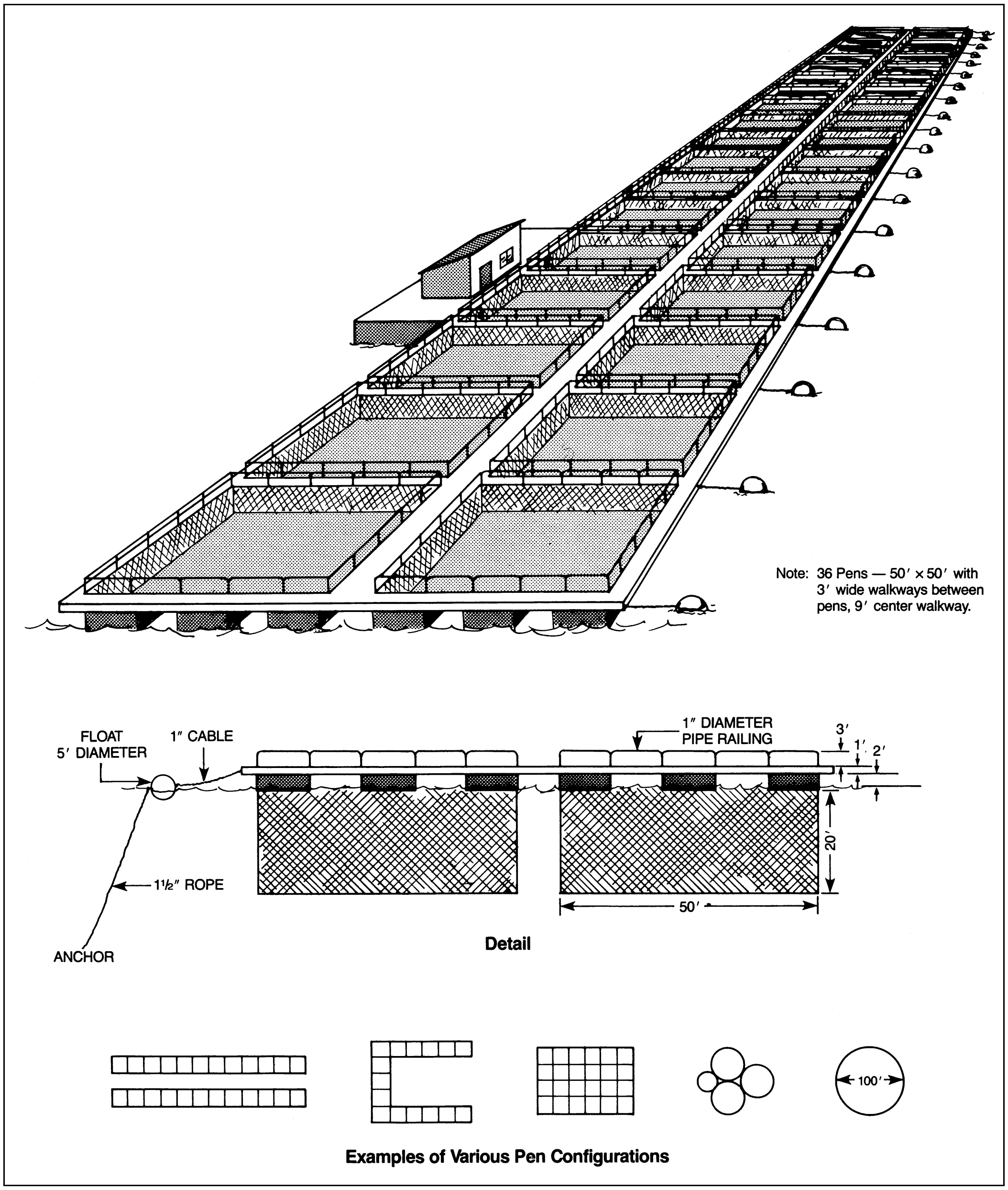
When they reach 2 inches in length, Pacific salmon begin feeding on insects, worms, and other invertebrates. As they develop dark vertical bar markings, they are called parr. At about 6 inches, Pacific salmon begin moving to sea. The physiological changes salmon make to switch from a freshwater to a saltwater environment are collectively called smoltification. After smoltification, salmon remain in the sea for 1 to 5 years, depending on the species, feeding and growing to sexual maturity and then returning to freshwater streams to spawn. Atlantic salmon parr may remain in freshwater for as long as 8 years before moving to sea (Weber, 1997). Most salmon species die after spawning, but Atlantic salmon can spawn several times, returning to the sea between events.

4.3.3.1 Production Systems

There are two types of salmon AAP, salmon farming and salmon ranching (or ocean ranching). Salmon farming involves two phases: (1) the freshwater hatchery phase for the incubation of eggs and the raising of juveniles to the smolt stage and (2) the seawater phase, in which the salmon are grown out to market size, usually in floating pens (Clarke, 2000). Salmon ranching, which is practiced primarily in Alaska, is an alternative form of AAP that involves the release of smolts from hatcheries and the harvest of adults returning from the ocean.

The hatchery or freshwater stage begins when fertilized eggs are placed in hatcheries operated with oxygenated water. Salmon hatcheries generally use flow-through systems; some partial recirculation systems are used to conserve heat during egg incubation. Stacked trays, upwelling jars, or troughs may be used as egg incubators. The salmon life cycle makes it possible for fish farmers to raise juvenile salmon in land-based tank and raceway operations before growing them out in marine environment net pens or cages. Young fish are raised in upland hatcheries until they become smolts; on the west coast, however, parr are often placed in estuarine pens of reduced salinity, and some fish are raised to maturity in freshwater. Smolts are then transferred to net pens (i.e., salmon farming), where they remain for 1 to 2 years until they reach market size. In Alaska, Pacific salmon (coho and chinook) are commonly raised in marine net pens for periods of 1 to 6 months before release by public agencies or Native American tribes for enhancement projects. These fish are stocked as late parr or smolt and released after growing in the pens (i.e., salmon ranching). Holding salmon later than their normal smolt outmigration timing causes them to residualize in the nearshore waters, a technique used to enhance the sport fishery.

Generally, flow-through systems are used in the hatchery phase for the production of smolts. Raceways, tanks, or ponds are used to grow juvenile salmon until they undergo smoltification. Saltwater production normally begins after smoltification when the salmon are moved to net pen systems, which is the dominant production mode in saltwater salmon farming in coastal waters (Figure 4.3–6). The advantages of net pen cage farm systems in marine environments are relatively low capital cost per unit of rearing volume, reduced risks of stock loss through system failure and low DO, and access to large volumes of relatively high quality water without pumping costs (Karlsen, 1993). The primary disadvantages of marine net pen systems are increased risks due to storm damage; a complicated, lengthy, and expensive permitting process; a reduced ability to manipulate environmental conditions such as water temperature; and a potentially increased risk of predation and disease transmission from wild animals.



Source: WDF, 1990.

Figure 4.3–6. Example of a Fish Farm and Various Pen Configurations

4.3.3.2 Culture Practices

Broodstock may be collected from the wild or raised at a hatchery facility. The goals of a hatchery program raising salmon to be released into the wild are different from the goals of a hatchery raising salmon for commercial production. Domestication is an important characteristic for salmon raised for commercial production, but hatcheries want to avoid the domestication of salmon that are to be released into the wild (Pepper and Crim, 1996). For enhancement production, broodstock should be chosen from wild stocks. For commercial foodfish production, broodstock may be either collected from the wild or bred and raised at a hatchery facility.

There are several types of incubators, but generally they all have a container with sufficient water flowing through it and some type of screened enclosure to prevent eggs and larvae from being washed away (Billard and Jensen, 1996). After hatching, the salmon, now called alevins, have a large yolk sac reserve. As they near the completion of the yolk absorption, alevins leave the substrate and become free-swimming fry (Pennell and McLean, 1996). The timing of emergence occurs as the alevins complete the absorption of the yolk sac. Emergence is influenced by factors such as light, substrate type, and changes in temperature and oxygen concentrations.

The initial presentation of food is a critical stage in salmon culture because it marks the transition between incubation and raising (Pennell and McLean, 1996). The fry are then transferred to rearing units. Flow-through raceways, both earthen and concrete, are the most common rearing units used for juvenile salmon culture (Pennell and McLean, 1996).

Production for Release

Pacific salmon species dominate production for release. Alaska hatcheries incubate approximately 100 million sockeye salmon eggs per year. Most of the fry are stocked into lakes not accessible to wild salmon and allowed to develop into smolts under natural conditions (Clarke et al., 1996). Atlantic salmon are more challenging to cultivate for release because of their slower growth rates and large smolt size. Most smolt production hatcheries use elevated temperature to speed incubation, advance the time of the first feeding, and optimize feeding during the summer (Clarke et al., 1996).

Production for Commercial Culture

For Atlantic salmon, smolt production for either stock enhancement or commercial AAP is most efficient when done in the shortest amount of time to minimize costs (Clarke et al., 1996). Atlantic salmon usually require 2 years of growth to reach the smolt stage in nature, but in commercial production, practices have allowed facilities to produce smolts in the first year by manipulating favorable temperatures, using high-energy feed, and applying good husbandry practices to minimize stress and disease (Clarke et al., 1996).

After smoltification, salmon for foodfish production are transferred to net pens for growout to market size. After the smolts are introduced to saltwater net pens, farmers monitor the progress of the salmon as they adjust to saltwater. Atlantic salmon, which can be especially sensitive, may need several days to resume proper feeding and acclimate to the net pens (Novotny and Pennell, 1996).

Harvest Practices

A decade ago, the growout phase in net pens required at least 2 years. Today, salmon can reach harvest size in 10 to 15 months after their transfer to net pens. Changes in feed formulation, feed pelletizing technology, the introduction of effective vaccines, and the domestication of farmed salmon stocks has shortened the time needed to grow salmon to harvest size (Roberts and Hardy, 2000). Today, after 12 to 18 months in net pens, fish are ready to harvest at weights ranging from 5 to 11 pounds (Novotny and Pennell, 1996). Because the salmon market is driven by quality of fish, farms emphasize quality control for harvest. Prior to harvesting, fish go through a period of starvation to reduce the fat content in the muscle tissue and the flora in the gut. This practice increases the shelf life of the salmon product (Novotny and Pennell, 1996). Fish are crowded into one corner of the pen and then pumped out with a fish pump or fish escalator and through a grader.

Feed Management

Feeding practices include hand feeding and a variety of mechanical feeders (Novotny and Pennell, 1996). Smaller cages with a low biomass of fish rely mostly on hand feeding. This requires cages placed nearshore with land access, a dock, or a small boat. Most net pen systems contain a large biomass of fish (e.g., 30,000 fish with a harvest weight of about 8 to 10 pounds) and require the use of mechanical feeders. For net pens that are single structures without supporting walkways, barges and boats with feed blowers take feed to the net pens and feed, usually once or twice a day. Bad weather can impede this method of feeding. Other facilities use a stationary blower to deliver feed to each net pen in a group of pens. To control overfeeding, many facilities also use underwater cameras to monitor feed consumption (Nash, 2001).

Health Management

To prevent transmission of diseases, salmonid eggs are sometimes disinfected at the time of fertilization or at the eyed stage. The common treatment used is the iodophor Povidine with a 1% to 2% concentration of active iodine, which is similar to iodine but not as corrosive (Billard and Jensen, 1996). About 1 quart of solution with 100 parts per million (active iodine) is applied to every 2,000 eggs for a period of 10 minutes, followed by a rinsing. Formalin is also used to prevent the spread of fungus (*Saprolegnia)* infections in eggs.

Freshwater salmonid diseases that have been observed in Pacific salmon hatcheries in the Pacific Northwest include furunculosis, bacterial gill disease, bacterial kidney disease, botulism, enteric redmouth disease, coldwater disease, columnaris, infectious hematopoietic necrosis, infectious pancreatic necrosis, viral hemorrhagic septicemia, and erythrocytic inclusion body syndrome. Pacific salmon hatcheries have also had outbreaks of a large number of parasitic infections like gyrodactylus, nanophyetus, costia, trichodina, ceratomyxosis, proliferative kidney disease, whirling disease, and ichthyophonis (Nash, 2001). Atlantic salmon are especially susceptible to furunculosis. The frequency of pathogen occurrences varies geographically. For example, a greater percentage of Alaska hatcheries tested positive for infectious hematopoietic necrosis, viral hemorrhagic septicemia, furunculosis, and ceratomyxosis between 1988 and 1992 than hatcheries located in other western states.

In the past, oral delivery of oxytetracycline in the feed was the standard treatment. Today, the use of vaccines is a common industry practice. Immersion and injected vaccines have been so successful and so commonly used that antibiotic treatment is infrequent (Novotny and Pennell, 1996).

Several drugs have been approved by the FDA for use in salmonid AAP (FDA, 2002). Oxytetracycline is approved for use in Pacific salmon for marking skeletal tissue and for use in salmonids to control ulcer disease, furunculosis, bacterial hemorrhagic septicemia, and pseudomonas disease. Sulfadimethoxine is approved for use in salmonids to control furunculosis. Tricaine methanesulfonate is approved for use as a sedative or as an anesthesia, and formalin is approved for use in salmon culture to control protozoa (*Chilodonella, Costia, Epistylis, Ichthyophthirius, Scyphidia, Trichodina* spp.) and monogenetic trematodes. Formalin is also approved for use on salmon eggs to control fungi of the family Saprolegniaceae.

4.3.3.3 Water Quality Management

Hatchery Water Quality Characteristics

Like other flow-through systems, hatcheries for salmon smolt production rely on a clean water supply with a consistent temperature. Water quality management in the system, including the raceways, directly affects the quality of effluents and the volume of discharge released from the rearing unit.

In a study by Kendra (1991), salmonid hatchery effluents from 20 different facilities (11 state and 9 commercial) in Washington state were monitored during the summer low-flow season. Relative to source water, effluents from salmonid hatcheries had elevated levels for temperature, pH, solids, ammonia, organic nitrogen, total phosphorus, and oxygen demand. Cleaning events elevated concentrations of solids, nutrients, and oxygen demand (Table 4.3–6). Salmonid smolts in Washington are typically released from state hatcheries through the drawdown of the rearing unit or pond. Near the completion of the release event, samples indicated increases in solids, nutrients, and oxygen demand. As the pond depth decreased, fish crowding increased the amount of disturbed accumulated sediments.

This study (Kendra, 1991) also measured the impact of effluent on receiving waters and found that benthic communities below hatchery outfalls were different from those located upstream or farther downstream. Three of the four hatchery discharges in the benthic community study caused a depression of taxa sensitive to organic pollutants. Several mayfly and stonefly species were eliminated below the outfall, as well as elmid beetles. Some invertebrates, such as mollusc families, planarians, and oligochaetes, were enhanced by the hatchery discharge (Kendra, 1991). As a result of this study, the hatchery National Pollutant Discharge Elimination System (NPDES) permit limits in Washington were revised to include primary settling of solid wastes as a minimum requirement for all hatcheries.

Table 4.3–6. Hatchery Effluent Quality During Cleaning and Drawdown Events

Cleaning Events								
Variable	*Units*	*Yakima Trout Hatchery (Single Raceway)*		*Aberdeen Trout Hatchery (Multiple Raceway Composite)*		*Drawdown Event, Naselle Salmon Hatchery (Rearing Pond)*		
		Normal	*Cleaning*	*Normal*	*Cleaning*	*Prior to Drawdown*	*Drawdown Midpoint*	*Drawdown Near End*
pH	SU	7.4	7.6	—	—	7.6	6.7	7.1
DO	mg/L	4.4	6.8	8.4	7.7	9.8	7.0	12.1
TSS	mg/L	1	88	1	12	7	30	94

Cleaning Events								
Variable	Units	Yakima Trout Hatchery (Single Raceway)		Aberdeen Trout Hatchery (Multiple Raceway Composite)		Drawdown Event, Naselle Salmon Hatchery (Rearing Pond)		
		Normal	Cleaning	Normal	Cleaning	Prior to Drawdown	Drawdown Midpoint	Drawdown Near End
Total volatile suspended solids	mg/L	0	69	<1	8	3	8	25
Settleable solids	mL/L	<0.1	2.5	<0.1	0.1	<0.1	0.3	1.1
Total Kjeldahl nitrogen	mg N/L	0.43	1.7	0.20	0.82	0.30	0.52	1.3
Total phosphorus	mg P/L	0.22	4.0	0.03	0.56	0.03	0.30	0.11
Chemical oxygen demand	mg/L	6	130	6	21	6	18	56
Biochemical oxygen demand (5-day)	mg/L	3	32	4	12	<3	3	—

Source: Kendra, 1991.

Net Pen Water Quality

In a study by the Washington Department of Fisheries (WDF, 1990) to evaluate the environmental impacts of commercial culture of fish in net pens, several water quality parameters were analyzed and potential impacts on the surrounding environment were evaluated. The EIS study by the Washington Department of Fisheries concluded that fish farms were not likely to have a significant impact on DO levels in Puget Sound except during the summer or autumn at sites that had low background DO levels and did not have adequate flushing (WDF, 1990). Overall, field measurements indicated that the area affected by low DO levels was less than 165 feet around the net pen structures.

Salmon net pens might also cause or increase phytoplankton blooms by increasing localized nutrient enrichment (Weston, 1986). Excessive phytoplankton growth can cause eutrophication. In a summary of experiments and modeling for phytoplankton impacts, the WDF assessment concluded that nutrients added by net pen operations were not likely to adversely affect phytoplankton abundance in Puget Sound. Model results for five 500,000 pounds/year farms showed an average increase of 0.0085 milligrams/liter in nitrogen concentrations in winter conditions, or less than 1% increase in total nitrogen concentrations (Table 4.3–7). During the summer, the model predicted a 2% increase in phytoplankton biomass. The study did note, however, that poorly flushed bays are more sensitive to nutrient loading and that areas identified as nutrient-sensitive should limit total fish production. The study also recommended locating farms to minimize the overlap of near-field conditions from multiple farms.

Table 4.3–7. Effect of Five Fish Farms in an Embayment on the Nitrogen, Phytoplankton, and Zooplankton Concentrations for Summer and Winter Conditions Based on the Kieffer and Atkinson Model (1988)

	Dissolved Nitrogen (mg/L)		***Phytoplankton (mg/L)***		***Zooplankton (mg/L)***	
	Ambient	***Increase***	***Ambient***	***Increase***	***Ambient***	***Increase***
Winter	1.5	0.0085	0.012	0	0.003	0
Summer	0.012	0	0.186	0.004	0.186	0.004

Source: WDF, 1990.

In a technical memorandum prepared by the National Oceanic and Atmospheric Association (NOAA) (Nash, 2001), the report identified three key issues of net pen salmon farming in the Pacific Northwest that appear to carry the most risk: the impact of bio-deposits (uneaten feed and feces), the impact on benthic communities of the accumulation of heavy metals in sediments below the net pens, and the impact on nontarget organisms from the use of therapeutic compounds (pharmaceuticals and pesticides) at net pen farms.

Sediment deposits beneath net pen operations affect benthic communities. Biodeposits from uneaten feed and fish fecal matter settle onto sediments near net pens and affect the chemistry of the sediment and the benthic community (Nash, 2001). Sedimentation from salmon farms changes the total volatile solids and sulfur chemistry in the sediments in the immediate area surrounding the net pens. At sites with poor water circulation, deposit accumulations can exceed the aerobic assimilative capacity of sediments, leading to reduced oxygen tension and significant changes in the benthic community. The accumulation of organic wastes in the sediments can also change the abundance and diversity of the benthic infaunal communities.

The impact on benthic communities of the accumulation of heavy metals in the sediments below the net pens was also identified as a significant impact from salmon farming (Nash, 2001). Both copper, from marine antifouling compounds used on net pens, and zinc, from fish feeds, can be toxic in their ionic forms to marine organisms. Higher concentrations of sulfide in the sediment reduce the availability of both copper and zinc, which could make the observed concentrations near net pens nontoxic.

Results from a sampling program in the Broughton Archipelago in British Columbia confirmed that organic waste material was accumulating at a rate faster than the rate of decomposition beneath salmon net pen farms (Deniseger and Erickson, 1998). Sediments from 30 active fish farms were surveyed for physical and chemical characteristics. Researchers found that material accumulations can be significant (greater than about 1 foot). Sedimentation affects the benthic community by creating anaerobic conditions, which can persist for up to 1.5 years or more (Erickson, 1999, personal communication). Additional information about net pen water quality is available from Mosso et al., 2003.

Current Treatment Practices in Net Pen Systems

The same advantages that make the net pen systems favorable for production are also the characteristics that limit the use of treatment practices. Net pens are open systems that use natural water currents and tides for water supplies and flushing. Relative to pond and raceway facilities, net pen systems have several advantages, including the following: land requirements are minimal, construction and capital costs are generally lower, and there are virtually no pumping costs (Weston, 1992). From an effluent treatment perspective, however, net pen culture creates unique challenges. Because the effluent is not confined, treatment of dissolved wastes does not appear possible, and the treatment or removal of solid wastes has several technical difficulties (Weston, 1992). For the most part, the industry relies on dispersal and dilution of waste by natural water currents to maintain water quality for fish production and to minimize environmental impacts (Weston, 1992). The most effective way of reducing water pollution from net pen facilities is to minimize the loss of feed (Bergheim et al., 1991)

Most net pens are inspected by divers on a regular basis. The divers look for holes in the nets, dead fish, and fouling problems. State regulatory programs require benthic monitoring at many net pen sites to ensure that degradation is not occurring under or around the net pens. Other current requirements include video recordings in the spring and fall of the bottom beneath and adjacent to the cages; biennial sediment redox layer depth determinations (which measure sediment chemistry) during the fall; monitoring and reporting monthly feed use; and monitoring and reporting water quality, nutrients, and phytoplankton at farfield sites at four separate water depths. Prior to placement in pens, Atlantic salmon smolt/juveniles must be marked to link the identity of each fish to the facility. In Maine, reproductively viable non-North American Atlantic salmon stocks and transgenic salmonids are prohibited at CAAP facilities (USEPA, 2002).

BMPs required for fish pen operations in Maine include mortality removal; prohibition of disposal of feed bags or other solid wastes into U.S. waters; prohibition of discharge associated with pressure washing of nets; operation of facilities to minimize the concentration of net-fouling organisms; prohibition of biocides, tributyltin compounds, and storage of predator control or containment nets on the sea floor; minimizing the loss of unconsumed food and food fines from pens; reporting requirements for events such as fish kills, algal blooms, and confirmation of fish infected with infectious salmon anemia or other transmittable disease; and damage to a net pen that could result in salmon escapement. BMPs for disease control include using FDA-approved drugs. Unapproved drugs, including drugs in the INAD program, are prohibited. There is also a reporting requirement for all drugs discharged within 30 days of application (USEPA, 2002).

4.3.4 Striped Bass

Striped bass (*Morone saxatilis*) were originally produced and stocked in freshwater impoundments primarily for recreational purposes. Interest in hybrid striped bass for foodfish production in the United States began in the late 1970s. Production of food-size hybrid striped bass in the United States grew from about 1 million pounds in 1990 to more than 10 million pounds in 1996 (Harrell and Webster, 1997).

One of four *Morone* species, the striped bass is a major sport and commercial species native to the Atlantic and Gulf coasts of the United States, with stockings that have expanded its range throughout much of North America (Kohler, 2000a). The other *Morone* species are white bass *(M. chrysops*), yellow bass (*M. mississippiensis*), and white perch (*M. americana*). When a reproducing population of striped bass was discovered in landlocked Santee Cooper Reservoir in South Carolina, fisheries biologists were interested in stocking striped bass in reservoirs for sport fishing and as a predator to control underutilized forage species. *Morone* hybridization programs began in the 1960s and focused on combining characteristics of recreational trophy fish with adaptability to landlocked freshwater systems (Kohler, 2000a).

In 1965, Robert Stevens, of the South Carolina Wildlife Resources Department, initiated the production of hybrid striped bass by crossing striped bass with white bass. The first hybrid striped bass cross, of the striped bass female with the white bass male, was initially called the original cross-hybrid striped bass, but it is now referred to as the palmetto bass. The reciprocal hybrid striped bass cross of the white bass female with the striped bass male is called the sunshine bass. Of the various crosses and backcrosses made, only the hybrid of a striped bass crossed with a white bass has gained wide acceptance as a cultured species.

4.3.4.1 Production Systems

The industry has two main components: fingerling production and growout production. Some farmers are involved in both sectors, but most farms focus on either fingerling or growout production.

Hybrid striped bass are frequently sorted by size, or phases, to keep fish of similar size together and prevent cannibalism. Hybrid striped bass fry and phase I (approximately 0.2 inches) fingerlings in ponds feed on zooplankton until they reach about 0.2 inches in size, when they must be trained to accept artificial feeds to decrease the chances of cannibalism. Often fish are harvested, graded, and stocked into tanks for training on feed and then are reintroduced into growout ponds as phase II fish (Harrell, 1997).

Foodfish are often stocked to achieve maximum densities of about 5,000 to 6,000 pounds/acre. They must be completely harvested before restocking. The ponds are drained between harvesting and restocking. To avoid draining the ponds, some farmers treat the ponds with a piscicide (a pesticide like Rotenone, used to kill fish) to eliminate remaining fish before restocking. Ponds are usually drained annually or biennially, depending on stocking size. Ponds are aerated to maintain DO and water quality. Fish are fed once or twice daily with mechanical feeders. Like catfish, hybrid striped bass production is concentrated in the southeastern United States and includes North Carolina, South Carolina, Florida, and Virginia.

Millions of *Morone* fingerlings are produced annually in state and federal hatcheries for stock enhancement and in private hatcheries as seed stock for foodfish production and fee fishing operations (Harrell and Webster, 1997). The fingerlings are stocked in earthen ponds, flow-through systems, closed recirculating systems, and net pens for growout. Today, foodfish production is based primarily on the production and raising of hybrid *Morone*. Although other striped bass hybrids have been created for potential foodfish production or have been used for stocking recreational programs, today only the palmetto bass and the sunshine bass are raised for production (Harrell and Webster, 1997).

In 1995 the Northeast Regional Aquaculture Center funded a survey conducted by the Striped Bass Growers Association and the University of Maryland to collect information from producers on the state of the striped bass industry (Harrell and Webster, 1997). The survey indicated that 66% of striped bass/hybrid striped bass producers use earthen ponds, 15% use tanks, 10% use net pens, and 9% use raceways for production. Of the producers culturing fish in tanks, most used flow-through systems (67%), while 22% used closed recirculating systems and 11% had the capability for both.

Stocking density for ponds differs between production of foodfish and production of fish for population enhancement efforts. Phase I fingerling ponds for population enhancement programs are stocked at a higher density, and fish are harvested at a smaller size than in ponds at foodfish growout operations. Stocking densities of striped bass larvae for population enhancement efforts range from about 50,000 to 600,000 per acre, and fish are harvested at sizes from 200 to 1,600 fish/pound (Harrell, 1997). In growout ponds stocking densities range from about 74,000 to 150,000 larvae/acre, with harvest sizes from 45 to 130 fish/pound (Harrell, 1997).

4.3.4.2 Culture Practices

Hatchery Phase

Unlike production of most cultured species, hybrid striped bass production typically relies on fertile wild broodfish to begin the production process. Striped bass broodstock are usually collected during spawning migrations in river headwaters above and below dams using electrofishing or gillnets (Kohler, 2000a). Another way to develop broodstock is to raise larvae or fingerlings in captivity until they reach reproductive age (Sullivan et al., 1997). Producers use hormones to induce spawning and then collect the eggs. Semen is then added to a mixture of eggs and water for fertilization. Embryos are incubated in aquaria, Heath trays, or MacDonald-type jars (Kohler, 2000a). Development is temperature-dependent; at 60.8 to 64.4 °F, the embryos begin to hatch 1 to 2 days after fertilization. By the fifth day, depending on the water temperature, the larvae absorb their yolk sacs. At this stage, they are known as fry until they metamorphose into juvenile phase I fish.

Phase I in Ponds

Successful phase I production requires a proper fertilization plan to ensure that the right zooplankton communities are present. Before phase I ponds are stocked with fry, they are drained, refilled, and fertilized with a mixture of organic fertilizers (such as cottonseed meal and alfalfa hay) and inorganic fertilizers (such as ammonium nitrate and phosphoric acid). The stocking density is dependent on the production goal. If the purpose of stocking is population enhancement, fry are stocked at a higher density to produce a greater number of smaller fish at harvest. Population enhancement programs need high quantities of fish to meet the management objectives of stocking a certain number of fish per acre of a reservoir or number of fish per mile of a river (Harrell, 1997). Fingerling producers stock fish at lower densities to produce larger fish. Producers buying fingerlings for growout want as large a fingerling as possible so that the fish can reach market size faster. Fry are fed salmon starter feeds by day 21 at a rate of 5 to 10 pounds/acre/day. Producers use progressively larger feed sizes and increase the ration sizes as the fish grow. Phase I usually takes 30 to 45 days when fish reach total lengths of 1.0 to 2.0 inches and weigh about 0.03 ounces (Kohler, 2000a). Survival rates greater than 15% for white bass and sunshine bass and greater than 45% for striped bass and palmetto bass are considered successful for phase I production. Phase I is the period during which the fish primarily feed on live food, mostly zooplankton; however, toward the end of this phase, the fish become more piscivorous. If supplemental feeding has not been initiated, cannibalism can cause high production losses.

Phase II in Ponds

Harvested phase I fingerlings are graded to separate out fish that are less than 1.0 inch total length (TL). Larger fish that are greater than 2.0 inches TL are also graded out to prevent cannibalism. The separated size groups are stocked in separate ponds. Unlike phase I, fertilizers are not used in phase II ponds. Because the fish are being fed manufactured feed, there is no need to stimulate zooplankton growth. Phase II describes striped bass and hybrid bass fingerlings from the time of phase I harvest until they are 1 year old (Harrell, 1997). Many growout farmers purchase phase I fish and stock them in their ponds for phase II and phase III growout. Some producers market phase II fish; these fingerlings are primarily sold to government agencies for enhancement stocking or to net pen operations. Harvesting smaller ponds (< 2.5 acres) for phase II fish is similar to harvesting phase I fish. Ponds are drained down, and producers use seine nets to harvest the fish. This is a common practice for fish used for enhancement purposes, where fish are loaded directly into a transport unit (Harrell, 1997). Larger ponds are too expensive to drain and harvest at one time, so many farmers have started using large haul seines similar to those used by the catfish industry, and fish loading pumps to move fish between ponds. The pumps can be connected to graders that sort the fish by size and return smaller fish to the pond being harvested for further growout.

Phase III in Ponds

Phase III production is not common in enhancement production efforts, so most of the available information on actual production efforts in ponds comes from the industry itself, not from scientific literature (Harrell, 1997). Phase III growout is basically the second year production of striped bass and hybrid striped bass to a market-size fish. Most of the time, fish are harvested before the beginning of the third growing season, and the ponds are prepared to receive a new crop of phase II fish to repeat the cycle. Production ponds for final growout are usually larger than phase I and phase II ponds. Most phase III ponds are about 5 to 6 acres, with a range between 1 and 10 acres (Harrell, 1997). Since most growout operations do not have the facilities to completely draw down a pond and hold the harvest in tanks until the fish can be sold, producers harvest their ponds weekly or biweekly (Harrell, 1997). Haul seines are pulled through the pond, and fish are crowded into live cars. Producers can also use boom nets and then load fish into hauling trucks for transport to a processing plant. Fish can also be quickly killed with an ice brine or electrical shock; then the individually are graded and sorted into shipping containers.

Other Systems Used to Culture Hybrid Striped Bass

Flow-through systems and recirculating systems are also used to culture hybrid striped bass. For hybrid striped bass production, the advantages of flow-through or recirculating systems include better control over water quality and the health of the fish, growing seasons that are independent of climatic influences, easier fish handling and harvests, and flexibility for extended harvests, resulting in year-round sales.

A small percentage of hybrid striped bass production relies on freshwater cage culture methods, which are generally restricted to small-scale operations where pond water resources are not conducive to seining or ponds are already inhabited by other fish. Phase II fingerlings are stocked through openings in the cage top, which also allow for feeding and harvesting. With fish confined in the cages, the culturist can readily observe their behavior and health and more easily feed, manage, and harvest.

Feed Management

When hybrid striped bass are cultured in tanks or other confined systems, automatic feeders are often used to dispense feed at regular intervals. In larger systems, such as ponds, blowers are more commonly used to dispense the food across a wider area. Finding a cost-effective feed for striped bass and hybrid striped bass is very important because feeding cost can be one of the largest variable expenses of producing these species (Gatlin, 2001). Protein is an essential element in hybrid striped bass diets. It is important to maintain the proper ratio of protein to energy to ensure that the fish synthesize the protein and use it for growth instead of metabolizing it for energy. An excess of energy can reduce intake and result in decreased growth. Because protein is the most expensive component of many AAP diets, it is not economical to supply excess protein. In a feeding trial at Kentucky State University, one group of juvenile sunshine bass raised in cages was fed a diet with 41% protein and a protein-to-energy ratio of 99 milligrams protein/kilocalorie energy, a second group was fed a diet with more protein and higher protein-to-energy ratios, and a third group was fed a diet with 41% protein and a lower protein-to-energy ratio. The results for the first two groups were similar. The decrease in protein in the third group's diet did not limit growth; however, it did cause increased fat deposition, which can cause a decreased meat yield in the final product (SRAC, 1998).

Fry and phase I fingerlings in ponds feed on zooplankton until they reach about 0.2 inches in size, when they must be trained to accept artificial feeds to decrease the chances of cannibalism. Often fish are harvested, graded, and stocked into tanks for training on feed and then can be reintroduced as phase II fish in growout ponds (Harrell, 1997). Because feeding observation is an important method of determining overall stock health, floating feed is most often preferred, except during the winter. In winter months, sinking feed is used so that fish will not have to rise to the surface for floating feed and be exposed to extreme temperature changes (Harrell, 1997).

Initially, phase II fish need to eat about 15% to 25% of their body weight per day, given in two separate feedings. Once the fingerlings reach 0.06 pounds, daily feeding rates are gradually decreased to about 2% to 3% of their body weight in two separate daily feedings (Harrell, 1997). Tractor-drawn blowers are often used to deliver the feed at large operations, but demand and automatic feeders can also be used in pond culture (Hochheimer and Wheaton, 1997).

Although hand feeding and demand feeders have been used in some flow-through systems, automated mechanical feeders are most commonly used for both recirculating and flow-through systems. These feeders include towed blowers, stationary broadcast or blower feeders, and automated feed delivery systems (Hochheimer and Wheaton, 1997).

Health Management

There appears to be no difference between pure strains of striped bass and hybrid striped bass with respect to the fishes' susceptibility to diseases. Striped bass diseases are caused by viruses, bacteria, fungi, protozoa, and metazoan parasites. Except for viruses and parasitic worms, most of the infectious agents trigger diseases only when striped bass are stressed or injured. Since striped bass and their hybrids are extremely susceptible to environmental stress, the best ways to prevent infectious diseases are to follow good AAP practices and health management practices, including an emphasis on maintenance of good water quality, use of optimum stocking densities, provision of adequate feed and good nutrition, maintenance of optimum temperature, and use of proper fish handling procedures (Plumb, 1997).

Viruses known to infect striped bass include the lymphocystis virus, infectious pancreatic necrosis virus (IPNV), and striped bass aquareovirus. Because viruses do not severely threaten striped bass, little is done to control virus outbreaks. Fish infected with lymphocystis are simply removed from a production facility; it is not practical, however, to remove fish infected with IPNV. In either case, the facility can be dried thoroughly or disinfected with chlorine (200 milligrams/liter) to kill any residual virus. There is not adequate information about striped bass aquareovirus to manage and control outbreaks (Plumb, 1997).

Bacteria cause the most serious debilitating infections of cultured striped bass. No bacterial diseases are unique to striped bass, but some bacteria have more serious effects on striped bass than on other cultured fish. Bacterial diseases affecting striped bass are MAS, *Pseudomonas* septicemia, Columnaris, Pasteurellosis, Edwardsiellosis, Vibriosis, Enterococcosis, Streptococcosis, Mycobacteriosis, and Carnobacteriosis (Plumb, 1997).

Control of bacterial diseases is best achieved through maintaining a high-quality environment and preventing conditions stressful to the fish. Sterilization of nets, buckets, and other production tools prevents cross-contamination between culture units. In recirculating water or open water supplies, ultraviolet (UV) radiation and ozone disinfection can reduce bacteria. Some drugs have proven effective in treating bacterial infections in striped bass. Although none of the therapeutic agents are FDA-approved, bathing fish in sodium chloride (0.5% to 2% for varying times) or potassium permanganate (2 to 5 milligrams/liter for an hour to indefinitely) and feeding fish medicated feed have been successful in treating bacterial infections. Medicated feed containing oxytetracycline (Terramycin) has been fed at a rate of 2.5 to 3.5 grams/45 kilograms of fish per day for 10 days for treatment, and medicated feed containing Romet-30 (sulfadimethoxine-ormetoprim) has been fed at a rate of 2 to 3 grams/45 kilograms of fish per day. Romet-30, however, might not be effective against Streptococcosis (Plumb, 1997).

Less is known about fungal diseases than about other diseases affecting striped bass because of the difficulty in identifying fungi and the fact that fungi are sometimes secondary pathogens to other diseases, injuries, or environmental stress. A few fungi that are known to cause infections in striped bass are *Saprolegnia parasitica* and related species, which cause “water mold,” and *Branchiomyces* species, which causes “gill rot.” Treatments of fungal infections with formalin, copper sulfate, and potassium permanganate have been used, but are often unsuccessful. Preventing fungal infections on eggs is possible through daily treatments of formalin at a rate of approximately 600 milligrams/liter for a 15-minute flush (Plumb, 1997).

4.3.4.3 Water Quality Management and Effluent Treatment Practices

Pond Systems

In a study in South Carolina (Tucker, 1998), water samples were collected and analyzed from 20 commercial hybrid striped bass ponds (Table 4.3–8). In an attempt to provide a broad representation of the industry, researchers included large and small operations, as well as ponds from both the coastal plain and piedmont areas of the state. Most of the commercial ponds sampled were freshwater ponds, but some saltwater ponds were also represented in this study. Overall, water quality parameters varied considerably from pond to pond. The BOD_5 of samples ranged from 2 to 60 milligrams/liter, and suspended solids and volatile suspended solids were typically high but variable. Generally, concentrations for many of the variables were higher in the pond samples than in the water source samples.

Table 4.3–8. Means and Ranges for Selected Water Quality Variables from Hybrid Striped Bass Ponds in South Carolina

Variable	*Mean*	*Range*
Suspended solids (mg/L)	49	0–370
Volatile suspended solids (mg/L)	29	0–135
Biochemical oxygen demand (mg/L)	11.5	1.4–64.4
Kjeldahl nitrogen (mg/L)	7.1	0–97.0
Total ammonia (mg N/L)	0.95	0.02–7.29
Nitrite (mg N/L)	0.07	0–2.94
Nitrate (mg N/L)	0.36	0–4.61
Total phosphorus (mg P/L)	0.31	0–1.9
Soluble reactive phosphorus (mg P/L)	0.02	0–0.18

Source: Tucker, 1998.

The South Carolina study also compared water quality in fingerling ponds and growout ponds. Fingerlings were usually produced in smaller ponds, and although average aeration rates were similar for fingerling and growout ponds, water exchange was less in fingerling production. Biomass and feeding rates were lower in fingerling ponds, as were parameters associated with particulate matter and nutrients. Overall, the quality of effluents from hybrid striped bass ponds varied greatly from pond to pond. The study did not find any significant seasonal variation in quality, but researchers noted that the sampling protocol might have affected the measure of true seasonal effects. Concentrations of suspended solids, total nitrogen (including total ammonia), and BOD were the water quality variables most elevated relative to the source water and would have the greatest impact on receiving bodies of water.

Other Production Systems

Water management in intensive systems, such as flow-through and recirculating systems, must address the full range of water quality parameters that could affect fish health and growth. Parameters to consider are continuous flow, adequate oxygen, consistent temperature, waste removal from the culture space, acceptable ranges of ammonia levels, control of parasite populations, and elimination of all other stress factors. Nearly all intensive systems include simple settling as part of the water management system to remove solids from the effluent stream, whether the water is to be reused in the system or discharged. Simple settling has proven adequate in removing the relatively dense waste solids from hybrid striped bass production (Hochheimer and Wheaton, 1997).

Because net pen culture practices rely on the water quality of the site at which the pens are located, there is little information on water management practices for hybrid striped bass production. Cages can be moved around within the pond, but generally they are of such small size that any water quality effects are negligible.

4.3.5 Tilapia

Tilapia are indigenous to Africa. In the 1940s they were introduced into Caribbean nations and, as a result, also entered Latin America and the United States. By the late 1950s the species had become the main focus of AAP research at Auburn University. Tilapia have been raised in most, if not all, U.S. states. Species cultured in the United States include Nile tilapia (*Oreochromis niloticus*), blue tilapia (*O. aureus*), Mozambique tilapia (*O. mossambicus*), Zanzibar tilapia (*O. urolepis hornorum*), and various hybrids of these species (Popma and Masser, 1999). In states where the growing season is not long enough to produce tilapia before winterkill occurs, production takes place in greenhouses or other buildings where supplemental heat is available. Since tilapia are still considered exotic, some states have restrictions on tilapia culture. In Arizona, California, Colorado, Florida, Hawaii, Illinois, Louisiana, Missouri, Nevada, and Texas, a permit may be required to culture tilapia, or the fish may be raised only if the species of interest appears on a list of approved fishes (Stickney, 2000c).

Most species of tilapia are mouthbrooders. Males construct nests in pond bottoms, females extrude eggs into the nests, males fertilize them, and females scoop them up in their mouths. Egg incubation (about 1 week) and hatching of fry take place in the female’s mouth, and fry stay in the mouth during yolk sac absorption. Once fry are ready to forage for food, they stay in a school around the female and go back into her mouth at any sign of danger. The fry remain in a school for several days after leaving the shelter of the female and stay around the edges of the pond where the water is warmest.

Mozambique tilapia can mature as early as 3 months after hatching; blue and Nile tilapia mature after approximately 6 months (Stickney, 2000c).

Although many tilapia species are produced as foodfish, some species, such as *Tilapia zilli*, are herbivorous and have been used to control aquatic vegetation in irrigation canals and sewage lagoons. Other more colorful tilapia species have been marketed as aquarium fishes in the ornamental market. Some salt-tolerant tilapia and hybrids have become the focus of new interest in tilapia production in coastal ponds and marine cages in the Bahamas and some Caribbean nations (Stickney, 2000c).

Tilapia have become one of the most commonly cultured species in the world. The 1998 Census of Aquaculture estimated that 116 farms produced 11.5 million pounds of tilapia, with a total of 137 farms producing food-size tilapia with a value of more than $23 million (USDA, 2000). The top five states for tilapia production in the United States are (in descending order) California, Maryland, Texas, Idaho, and Florida. Many culturists prefer to raise blue tilapia and Nile tilapia over the Mozambique tilapia because the former have better dress-out percentages, later maturity, and a more desirable flesh color (Stickney, 2000c).

4.3.5.1 Production Systems

Three primary types of production systems are in use at tilapia farms: ponds, flow-through production, and recirculating systems. Ponds and recirculating systems are the more common systems used for tilapia production in the United States, while flow-through systems are less common. In the southern United States, tilapia are sometimes raised in cages or net pens in lakes, large reservoirs, farm ponds, rivers, cooling water discharge canals, and estuaries; however, cage culture is a less common production system for tilapia.

Tilapia's intolerance of coldwater limits its production potential in outdoor systems throughout most of the United States. Only southern Florida, Texas, Puerto Rico, Hawaii, and other Pacific islands have climates suitable for year-round outdoor pond production (Rakocy, 1989). Enclosed greenhouses are also used in some parts of the country, and in temperate climates tilapia must be grown indoors with heated water. Operators must either heat their airspace and influent water or use alternative sources of warmwater, such as recycled wastewater that has been used to cool power plants or geothermally heated water (Rakocy and McGinty, 1989).

Ponds for tilapia production are similar to pond systems developed for other warmwater AAP species such as catfish and shrimp. Tilapia ponds require a design conducive to draining because fish harvest is difficult to perform without removing some or all water from the pond (Rakocy and McGinty, 1989). Tilapia are also cultured in flow-through systems. Circular tanks are the most common rearing unit for flow-through tilapia production because they have superior flow characteristics with fewer low-flow "dead spots" than rectangular tanks (Rakocy, 1989). Recirculating systems for tilapia production are similar to flow-through systems in terms of tank design, aeration, feeding, fish handling, and solids removal; however, water discharge is minimal with the operation of a recirculating system. Recirculating systems are widely used to produce tilapia for the live fish market because recirculating systems can be used for year-round production. Recirculating systems can also reduce water-heating costs and transportation costs because facilities can be located near large metropolitan market areas.

4.3.5.2 Culture Practices

Tilapia are often bred in recirculating systems because spawning is more easily observed and controlled in small tanks than in other systems. Ten to twenty days after tilapia broodstock spawn, fry begin to swim away from the mouth-brooding female fish. Fry can be collected with dip nets from the brood tank for stocking in nursery tanks (Rakocy, 1989).

Male tilapia are preferred for intensive food fish culture because they grow more quickly than female fish. Female fish divert energy from growth to producing eggs, and mouth-brooding females generally do not eat while holding young in the mouth. It is possible to produce all male fish with certain hybrids of *Oreochromis* species. Feeding newly hatched female fry with feed treated with male hormones inverts the sex of female tilapia to change them into reproductively functional male tilapia. Androgens such as methyl testosterone are used to invert the sex of female fry (Kohler, 2000b). Other methods include using a combination of hormones to produce “supermale” tilapia with double Y (YY) chromosomes instead of XY chromosomes. These YY males can be crossed with normal XX female fish to produce all male progeny. Researchers also have been experimenting with triploid (fish that have three sets of chromosomes and are unable to reproduce) and tetraploid fish (fish that have four sets of chromosomes that can be mated with diploid fish to produce triploids) to produce faster growing fish without the use of hormone treatments (Kohler, 2000b).

Fitzpatrick et al. (2000) treated fry with methyl testosterone at a concentration of 60 milligrams/kilogram in their feed for 4 weeks beginning at the initiation of feeding. The treated fry were raised in three 16-gallon tanks that contained no soil or gravel, 11 pounds of soil, or 11 pounds of gravel, respectively. Methyl testosterone water levels peaked at approximately 3.6 nanograms/milliliter at 28 days after the onset of feeding. The concentration of methyl testosterone in water decreased to background levels (nondetect to 0.02 nanograms/milliliter) in 1 to 2 weeks after the end of treatment with methyl testosterone-impregnated food in those tanks containing soil or gravel. The concentration of methyl testosterone in the tank containing no soil or gravel remained above background levels for 3 weeks after the end of treatment with methyl testosterone-impregnated food (Fitzpatrick et al., 2000). Methyl testosterone degrades when exposed to light or high temperatures. In addition, bacteria and fungi can metabolize methyl testosterone; therefore the light, temperature, and microbial degradation in an outdoor pond setting degrade methyl testosterone.

The soil concentration of methyl testosterone in the tank with soil was 6.1 nanograms/gram at the end of the 28-day treatment period. This level decreased to approximately 3 nanograms/gram at 8 weeks after the end of the treatment period (cessation of experiment). The methyl testosterone soil background level was 0.5 nanograms/gram at the beginning of the experiment. The methyl testosterone levels in the gravel tank ranged from 22.9 to 99.2 nanograms/gram of fine sediment at 8 weeks after the end of the treatment period. The authors suggested that the slow degradation of methyl testosterone in soil and gravel might have occurred because the sediments acted as a trap for methyl testosterone (Fitzpatrick et al., 2000).

Stocking density for tilapia fry in flow-through systems can be maintained at as high as 750 fry per square foot. Once fish reach approximately 1 pound, recommended stocking levels drop to about nine fish per square foot (Rakocy, 1989). Most tilapia raised for foodfish are harvested when they reach 1 pound. Depending on the quantity of food and aeration inputs, tilapia can be raised from fry to harvestable sizes in 7 to 8 months (Rakocy, 1989). Tilapia are more difficult to capture in seines than many other species of cultured freshwater fish because they have a tendency to jump over, or burrow under, nets (Rakocy and McGinty, 1989). Only 25% to 40% of tilapia in a small pond are usually harvested by seine nets. Complete or partial pond draining is usually necessary to harvest all the tilapia in a pond (Rakocy and McGinty, 1989). Tilapia in recirculating systems are usually harvested by crowding the fish into one part of the tank. The fish are then dipped out of the tank with nets or pumped out.

Feed Management

In pond production tilapia are able to feed on naturally occurring green algae, blue-green algae, zooplankton, benthic invertebrates, and decomposing organic matter. Many operators, however, supply tilapia with commercially prepared feeds using mechanical feeders to encourage faster growth.

Because tilapia can thrive on naturally occurring foods in ponds, they can be integrated into catfish pond culture during the summer months when water temperatures are above 50 °F. The stocked tilapia produce a second crop of fish without the producer incurring additional feed costs. Raising tilapia with catfish also might have the additional benefit of reducing off-flavor problems that can occur in traditional catfish farming because tilapia consume the blue-green algae that often cause an off-flavor problem (Rakocy and McGinty, 1989). Labor costs associated with sorting the catfish and tilapia at harvest, however, may reduce net profits for the operator.

Tilapia raised in flow-through systems are fed commercially prepared feeds using mechanical feeders. Adult fish are usually fed 3 to 6 times per day, at a rate of approximately 1% to 3% of their body weight per day. Under ideal conditions, with high-quality feeds, FCRs approaching 1.5 are possible with tank-raised tilapia (Rakocy, 1989). Tilapia in recirculating systems are also fed high-protein, commercially prepared feeds that optimize growth. Generally, the fish are fed using automatic feeders, which dispense food from above the tank.

Health Management

Three types of water-conditioning chemicals are commonly added to commercial recirculating systems for tilapia production. Sodium bicarbonate, or an alternative alkalinity source such as sodium hydroxide, is often added to replace alkalinity lost to nitrification in the biofilter (Loyless and Malone, 1997; Malone and Beecher, 2000; Tetra Tech, 2002c). Salt (sodium chloride) is added the system to prevent the occurrence of brown blood disease, which occurs in fish when water contains high nitrite concentrations. With this fish disease, nitrite enters the bloodstream through the gills and turns the blood to a chocolate-brown color. Hemoglobin, which transports oxygen in the blood, combines with nitrite to form methemoglobin, which is incapable of oxygen transport. Brown blood cannot carry sufficient amounts of oxygen, and affected fish can suffocate despite adequate oxygen concentration in the water (Tetra Tech, 2002c). Calcium chloride is used to simultaneously provide chlorides and increase calcium hardness in soft water areas.

4.3.5.3 Water Quality Management and Effluent Treatment Practices

Pond Systems

Tilapia become susceptible to disease when water temperatures are below 65 °F or when levels of ammonia, pH, and DO fall beyond recommended ranges. Tilapia are more tolerant of low DO levels than many other cultured foodfish species (Stickney, 2000c). Tilapia grown at low densities may not benefit from artificial aeration under normal pond conditions; however, supplemental aeration is recommended when growing tilapia in intensive pond culture systems with high fish densities (Papoutsoglou and Tziha, 1996; Rakocy and McGinty, 1989).

Tilapia ponds are drained to harvest fish, to adjust fish inventories, or to repair ponds. At the start of pond draining for harvest, pond water effluent characteristics can be expected to be similar to production water characteristics. Fish harvest by seining, however, stirs up sediments at the bottom of the pond. In fertilized tilapia ponds, sediments are likely to contain significant quantities of nitrogen and phosphorus. As draining and seining continue, effluent water quality can be expected to deteriorate (Tucker, 1998).

There is little mention in the literature of pond effluent treatment practices specifically for tilapia. If tilapia, however, are held in earthen ponds similar to those used for other freshwater fish, effluent management practices developed for catfish, crawfish, and hybrid striped bass can be expected to apply to tilapia culture. Tucker (1998) outlines some general pond culture effluent management guidelines: use high-quality feeds to reduce waste; provide adequate aeration and water circulation to avoid pond stratification; minimize water exchange during the growing season; leave excess storage capacity to capture rainfall and minimize overflow; harvest ponds without draining; and if draining is necessary for harvest, hold the last 10% to 20% of the water for 2 to 3 days prior to discharge to allow time for solids to settle.

Flow-through Systems

Flow-through systems must be managed to provide sufficient volumes of water to supply fish with oxygen and remove solid and dissolved wastes; therefore, these systems have a high demand for water.

There is little information concerning effluent treatment in tilapia flow-through systems; however, it is likely that common solids removal practices for other flow-through systems, including screens and settling basins, are common for tilapia flow-through production as well.

Recirculating Systems

Tilapia are hardy, disease-resistant fish, but when water temperatures are too low, they lose their resistance to disease and stop growing. In indoor recirculating systems, the optimal water temperature for tilapia production is 82 to 86 °F (Rakocy and McGinty, 1989). In temperate climates, water used in recirculating systems needs to be heated, especially during winter months. Alternatives to heating municipal or well water include using geothermically heated water (Rakocy and McGinty, 1989) or using heated effluents from electric power generating stations (Rakocy, 1989).

Many growers aerate recirculating systems with oxygen from liquid oxygen tanks leased from commercial suppliers. Tilapia grown in a recirculating system in North Carolina are supplemented with approximately 0.5 pounds of liquid oxygen per pound of food added to the system (Tetra Tech, 2002c). High-density systems that use enriched oxygen sources must also provide for a means of carbon dioxide stripping to prevent pH depression in the circulating waters (Grace and Piedrahita, 1994). Some recirculating system design guidelines advocate direct aeration of tanks (Malone and Beecher, 2000; Sastry et al. 1999) or indirect aeration through the use of airlift pumps (Parker, 1981; Parker and Suttle, 1987; Reinemann and Timmons, 1989). In these blown air systems, oxygen addition and carbon dioxide stripping are reasonably balanced, and a separate carbon dioxide stripping process is not employed (Loyless and Malone, 1998).

Some of the water in recirculating systems must be discharged daily to remove solid wastes. In general, effluents from recirculating systems are more concentrated than wastewater from flow-through or pond systems. The total daily volume of effluents from recirculating systems is typically orders of magnitude smaller than effluents from flow-through systems of similar capacity that do not reuse water. Small discharge volumes make wastewater more economical to treat and in some cases alleviate the need to discharge to receiving waters. A recirculating system used to grow tilapia in North Carolina discharged such small quantities of wastewater that evaporation from an on-site aerobic waste lagoon exceeded the rate of wastewater inflow during summer months (Tetra Tech, 2002c).

4.3.6 Other Finfish

4.3.6.1 Largemouth Bass

Largemouth bass *(Micropterus salmoides)* are said to be the most sought after freshwater sport fish in the United States. State and federal hatcheries produced 21 million largemouth bass for sport fish stocking in 1995 and 1996 (Heidinger, 2000). It is estimated that commercial hatcheries produced approximately the same amount. A limited number of adult bass are used as foodfish by some consumers (mainly centered around large cities), but it can take 2 to 3 years to grow bass to an adequate food-fish size.

The geographic range of largemouth bass is limited by temperature because they can be stressed at low temperatures (around 36 to 39 °F). These temperatures can occur in the winter in culture ponds located at the latitude of southern Illinois.

There are two subspecies of largemouth bass, the northern largemouth bass (*M. salmoides salmoides*) and southern Florida largemouth bass (*M. salmoides floridanus*). Genetic tests are required to tell the two species apart because they cannot be differentiated by a visual inspection. It is important to know which species one is working with during production because the southern subspecies is not as tolerant of low temperatures as the northern subspecies can be (Heidinger, 2000).

Production Systems

Various methods are used to produce largemouth bass. Most producers stock broodfish in ponds to spawn, although some are stocked in raceways or net pens, allowing the producer to be in greater control of production. Ponds are usually rectangular and less than 6 feet deep with no obstructions. Ponds are drained and completely dried in the fall to get rid of predacious insects, fishes, and diseases. Some operators sow winter rye in the pond to serve as an organic fertilizer after spring flooding. Agricultural lime can be added if the pond bottom soil is too acidic. Ponds should not be filled more than 14 days before stocking to prevent the buildup of predacious insects. Well water or surface water, which is filtered through 52 mesh/inch saran socks, are both acceptable for filling the ponds (Davis and Lock, 1997).

Culture Practices

Fry are left in the spawning ponds or moved to rearing ponds and fed zooplankton and aquatic insects. When the fish are fingerlings, they are raised at a low density on insects, or they can be trained in tanks to eat a prepared diet. Fingerlings (1.5 to 2.0 inches) are seined from nursery ponds, graded to uniform sizes, and stocked in round or rectangular flow-through tanks for feed training. Stocking density can be high, with a range from 200 to 500 fish/cubic foot (Tidwell et al., 2000). Fingerlings that are trained to eat the prepared diet grow faster than those feeding on insects, and the trained bass can then be moved to ponds, net pens, or raceways until they reach the desired size.

Bass are most often harvested by trapping, seining, or draining the pond. Fingerlings are generally harvested 2 to 4 weeks after stocking, when they are approximately 1.5 inches in length, to lessen the chances of cannibalism. Although cannibalism is possible at any time, it is more likely to occur if fry are stocked at different ages and sizes and if there is a shortage of food. If at any time it is found that no appropriate invertebrates are present in the pond as a food source, the bass must be harvested regardless of size (Heidinger, 2000).

During training periods in tanks, largemouth bass are extremely susceptible to external parasites and the bacterial disease columnaris (caused by *Cytophagus columnaris*). Affected fish are treated immediately through medicated feed (terramycin). The use of salt baths of 0.5% to 1.0% for up to 1 hour is another practice used to reduce stress from handling and grading and to reduce the incidence of infectious diseases (Tidwell et al., 2000).

4.3.6.2 Smallmouth Bass

Smallmouth bass *(Micropterus dolomieui)* are popular sport fish found in many parts of the United States and are essentially nonmigrating fish. The species requires growing temperatures from 50 to 70 °F and spawning temperatures of 58 to 62 °F; the upper lethal temperature reported is 95 °F (Illinois-Indiana Sea Grant, n.d.). Ponds are the most common production system used for smallmouth bass culture (Illinois-Indiana Sea Grant, n.d.).

4.3.6.3 Carp

Several species of carp *(*family Cyprinidae*)* have been cultured in the United States. The government stopped stocking common carp (*Cyprinus carpio*) in the United States in the late 1800s because of problems associated with the species, such as damage due to erosion caused by the fish digging into the pond banks. Many reproducing populations, however, became established from early stocking programs and are still plentiful today. Although common carp are cultured as foodfish in other countries, there is a very small demand for them as foodfish in the United States. The fish have many small bones and often have poor flavor. There is a very small amount of commercial production of bighead carp (*Aristichthys nobilis*) and silver carp (*Hypopthalmichthys molitrix*), but that production is insignificant. Various carp species are banned in some U.S. States because they are considered to be exotic species.

The grass carp (*Ctenopharyngodon idella*) is commercially produced in the United States primarily for use in controlling aquatic vegetation. This species is very controversial because of concerns that it might also consume desirable vegetation and reproduce and become established in areas where it is not desired. Since the species is banned in many states and controversial in others, commercial producers began producing triploid grass carp (fish that have three sets of chromosomes and are unable to reproduce). Triploid grass carp are beneficial in controlling vegetation, and they die after a few years, so the decision can then be made whether to restock. Some states that had banned carp have made exceptions and allow stocking of the sterile triploid grass carp as long as the producers can certify that the fish are 100% triploid (Stickney, 2000a).

Culture Practices

Production of sterile triploid grass carp includes subjecting fertilized eggs to a pressure treatment that makes the eggs hold onto an extra set of chromosomes. The process involves placing the eggs in a stainless steel container and subjecting them to 8,000 psi (pounds per square inch) of hydrostatic pressure. The eggs hatch after an incubation period of 2 to 3 days, and the fish feed off of their attached yolk sacs. After 3 days, the fish can be fed hard-boiled egg yolks followed by commercial fish food and brine shrimp larvae as they grow. After a week in the hatchery, the young fish should be transferred to larger ponds, which should be fertilized to encourage zooplankton growth for a food source. After the fish reach approximately 1.5 inches in length, they begin to eat green plant material. The fish can undergo blood testing to determine whether they are triploid and sterile when they are 2 to 3 inches in length (Imperial Irrigation District, 1998).

4.3.6.4 Flounder

The summer flounder (*Paralichthys dentatus*) is a foodfish found along the east coast of the United States, from Maine to Florida (Bengtson and Nardi, 2000). The winter flounder (*Pseudopleuronectes americanus*) is a foodfish found along the east coast of North America, from the state of Georgia to Labrador, Canada. The species has been exploited for more than a century and is now considered overexploited due to its decline over the past 20 years. Hatchery production of winter flounder was first attempted in the late 1800s by the U.S. Fish and Fisheries Commission in an attempt to try to rebuild wild populations that were in decline. Those hatcheries released tens of millions of larvae before closing in the 1950s. Some of the techniques developed at those hatcheries are still in use today, now that declining stocks, coupled with a demand for quality flatfish, have once again motivated attempts to culture winter flounder (Howell and Litvak, 2000).

Production Systems

Commercial hatchery production of summer flounder in recirculating or flow-through tanks began in 1996, after 6 years of government funding for research and development for cultural practices of the species. So far, only wild-caught broodstock have been used in commercial production, but hatcheries are working on domesticating them (Bengtson and Nardi, 2000).

Researchers and fish culturists of winter flounder have looked to information on production techniques for summer flounder for guidance. There are, however, some differences in culture techniques for the two species. Hormonal injections to induce spawning seem to be used more in winter flounder production than in summer flounder production. Static, flow-through, and *in situ* systems have all been used to raise winter flounder larvae, though static systems have been used only in research, not for commercial production. In larval flow-through systems, 100-liter circular tanks are supplied with seawater that has been filtered and treated with ultraviolet light and kept at ambient temperatures and salinities. One *in situ* system was tried in Rhode Island with favorable results. It consisted of an open-mesh enclosure (406 cubic feet in size) suspended from a surface flotation collar. The mesh size was small enough to keep larvae in while still allowing their natural food to enter the enclosure. The estimated time for growth to market size is 2 to 4 years. This time might be shortened in an AAP setting due to optimal fixed conditions used there, and the growout systems used would be similar to those for summer flounder (land-based tanks or raceways and net pens) (Howell and Litvak, 2000).

Culture Practices

Ideally, captured summer flounder broodstock are held for several months to allow them to adjust to their new surroundings and nonliving food diet before spawning is initiated. Some hormonal injections have been tried to induce spawning, but the most widely used method is hand-stripping the ripe fish. It is a high priority to develop methods for natural spawning since hand-stripping fish is highly stressful to the fish and might not be the best method for gathering the highest-quality eggs. After eggs and milt are stripped from the females and males, the gametes are combined in beakers where fertilization takes place. The embryos are placed in cylindrical containers of seawater. Hatched larvae can be taken from the incubation containers and put into rearing tanks, where they feed on rotifers. Survival rates are higher in rearing tanks to which algae have been added.

After larvae go through metamorphosis and settle to the bottom of the rearing tanks, they should be transferred to juvenile rearing tanks where they can become accustomed to an artificial diet and grow out to about 2 grams before being netted and graded into larger tanks. Tanks may be round or square and range in size from 106 to 212 cubic feet. Raceways should have rounded corners (known as D-ended). Regular cleaning of tanks and removal of uneaten feed and feces are extremely important.

Summer flounder can grow to about 5 grams in five months and are then ready for transfer to a growout operation. It has not been determined what systems and procedures work best for growout production, but recirculating systems and net pens have both been tested by certain companies. The U.S. government has funded some of those projects and hopes to compare growth and quality of the fish grown in the two types of systems, as well as qualitative and quantitative cost production differences for the two systems. The estimated time for growth to market size is 24 to 28 months (Bengtson and Nardi, 2000).

4.3.6.5 Paddlefish

Paddlefish *(Polyodon spathula)* are prehistoric fishes used as foodfish and as a source of eggs, or roe, for caviar. They are found in 22 states on the Mississippi River Basin and the adjacent Gulf Coast drainage. Overfishing, habitat modification, and contamination by polychlorinated biphenyls (PCBs) and chlordane have caused paddlefish numbers to decline. Paddlefish are protected against illegal roe collection through their listing on the United Nations' Convention on International Trade of Endangered Species of Wild Fauna and Flora (CITES).

Production Systems

Paddlefish can be raised in ponds or raceways. In pond production, survival rate ranges from 30% to 80%. In raceways, the survival rate increases to approximately 50% to 80%. Paddlefish broodstock are usually obtained from the wild because they take 7 to 9 years to mature. They are generally raised in circular tanks with an average diameter of 8 feet, allowing them to swim continuously and aerate their gills; however, tanks can be larger.

Culture Practices

Approximately 2 weeks before propagation, ponds to be used for paddlefish fry are completely drained and dried. After propagation, the ponds are filled with water from a well or from a reservoir that filters water through a saran sock. Organic fertilizers, such as rice bran or cottonseed, soybean, and alfalfa meals, are recommended for use in the nursery ponds to achieve a total nitrogen amount of 40 pounds/acre. During the initial fertilization period, large zooplankton such as *Daphnia* species should be inoculated into the pond at a concentration of eight *Daphnia* per gallon. It is recommended that ponds be covered with netting to prevent bird predation of fry.

Propagation of paddlefish can be achieved artificially. The fertilized eggs are placed in incubation jars, where fry hatch in approximately 6 days. The fry absorb residual yolk in 5 to 6 days, after they are ready to eat external food such as *Daphnia*. Once water temperatures are higher than 65 °F, fry can be stocked at a rate of 25,000 fish/acre in the prepared (fertilized) earthen ponds, where they feed on the *Daphnia* or insect larvae. At the age of about 5 to 6 weeks old, the fry's gill rakers develop, allowing them to filter-feed. Their diet can be supplemented during this time with trout/salmon crumbles (50% protein) at a rate of 15 pounds/acre, and after about 3 to 4 weeks, when the fish are 3 inches, they can eat 1/16-inch extruded pellets. In about 6 months, fish can grow to up to

14 inches long and 0.33 pounds in weight. The fish can be harvested easily with gill nets or seines.

Paddlefish fingerlings (less than 10 inches) can also be cultured in raceways or flow-through systems. If groundwater is used, it should be aerated and heated to more than 72 °F. Surface water may also be used, but it needs to be filtered and also aerated and heated if needed. Because strong sunlight can cause sunburn and mortality in paddlefish, outdoor raceways should be covered with 95% shade cloth, which may also offer some protection against bird predation. Like fry raised in ponds, fry in raceways can be trained to eat a sinking diet of trout/salmon crumbles (more than 50% protein), and after about 3 to 4 weeks, when the fish are 3 inches long, they can eat 1/16-inch extruded pellets. The pellets can be provided by automatic feeders every 15 to 20 minutes for about 7 to 10 days; then both automatic and hand feeding can be used to feed every 2 hours until the fish are stocked into ponds or reservoirs.

Initially, fry can be stocked in raceways at eight fish per gallon, but as they grow, fish should be reduced to lower concentrations to prevent crowding. After 2 weeks, fry should be about 2 inches in length and should be reduced to 2.5 fish per gallon. At 4 weeks after stocking, fish should be about 4 inches and should be reduced to 0.75 fish per gallon. If fish start “billing”—swimming at the surface with their paddles out of the water—they are demonstrating that they are stressed by high densities. Reducing densities generally stops this behavior (Mims et al., 1999).

4.3.6.6 Sturgeon

Atlantic, shortnose, lake, and white sturgeons (*Acipenser oxyrhynchus, A. brevirostrum, A. flurescens,* and *A. transmontanus*, respectively) are prehistoric anadromous fish used as foodfish and a source of roe for caviar. Sturgeons were once abundant, but habitat modification and overfishing, combined with the species’ slow reproductive rate, have dramatically reduced sturgeon populations (Friedland, 2000). White sturgeons are found in North America from Ensenada, Mexico, to Cook Inlet, Alaska (PSMFC, 1996), while Atlantic sturgeons are found from Florida to Labrador, Canada, and shortnose sturgeons range from Florida to New Brunswick, Canada (Friedland, 2000).

Production Systems

Sturgeon culture facilities are usually land-based tank systems. Producers can use recirculating systems during different production cycle phases. Sturgeon producers can also use gravity-flow linear raceways and discharge water to water bodies, preventing the escapement of cultured fish through the use of screens and settling ponds (Doroshov, 2000).

Bird predation is a significant problem in pond culture, especially for small sturgeons. Netting over ponds can help prevent bird predation, but recirculating systems or flow-through systems may be more economical for the growout of small sturgeons to market size. Larger sturgeons (1 pound or larger) are less vulnerable to bird predation due, in part, to the fact that the larger fish are almost entirely benthic (Bury and Graves, 2000).

Culture Practices

Sturgeon culture is difficult because of the complexity of replicating the species' natural spawning and raising activities. Minor surgery is required for internal examination of the fish to determine their sex and level of maturity, and eggs must be closely monitored to determine when they can be successfully fertilized (Government of British Columbia, n.d.).

Migrating Atlantic sturgeons are captured with gill nets, transported to hatcheries, and placed in either 0.25-acre freshwater earthen ponds or round fiberglass tanks. The fish are held for 12 to 13 days before spawning is induced by intramuscular injections of acetone-dried or fresh sturgeon pituitary gland extract. Eggs and sperm are mixed for 1 to 2 minutes, and fertilized eggs are stirred and washed for 10 to 30 minutes before being placed in MacDonald hatching jars. Yolk sacs are absorbed by fry 9 to 11 days after hatching, and the fry are then fed a diet of ground beef liver mixed with salmon mash, supplemented with live *Artemia* nauplii (Conte et al., 1988).

Lake sturgeons can be artificially spawned and then raised in floating cages or net pens. Spawning lake sturgeons are dip-netted from the Fox and Wolf Rivers in central Wisconsin. Sperm and eggs are collected from the fish, and the fish are then released. Eggs are fertilized and placed in MacDonald hatching jars. The fry absorb their yolk sacs within 10 days after hatching, after which the fry actively swim and feed on live brine shrimp nauplii. When the fry reach a length of about 1 inch, they begin feeding on larger zooplankton (Conte et al., 1988).

Shortnose sturgeons are captured with gill nets or by electro-fishing and transferred to cylindrical tanks at the hatchery. Females are held for 3 to 4 weeks, and males up to 6 weeks, before spawning is induced by intramuscular injections of acetone-dried or fresh sturgeon pituitary gland extract. Eggs and sperm are collected and mixed, and fertilized eggs are incubated in MacDonald hatching jars or Heath Techna trays. Eggs are treated daily with formalin (1,670 milligrams/liter for 10 minutes using a constant-flow method) to prevent fungus development. Larvae are raised in fiberglass and aluminum troughs. The troughs are 8 feet long, 1.5 feet wide, and 8 inches deep, and they are connected to a flow-through freshwater system, which has regular applications of formalin (1.775 milligrams/liter for 1 hour) and occasional applications of streptomycin/penicillin. After 1 week, larvae are fed live *Artemia* nauplii and salmon starter meal. After the larvae absorb their yolk sacs, ground beef liver is added to the diet as are supplemental experimental feeds and commercial semi-moist and dry rations.

Juvenile shortnose sturgeons are raised in 0.5-acre outdoor ponds (mean depth about 5 feet) where they feed on the ponds' benthic fauna and supplemental dry rations. They can also be raised indoors in 12-feet-diameter, 2.5-feet-deep fiberglass tanks connected to a freshwater recirculating system, where they feed on beef liver, squid, earthworms, polychaete worms, dry salmon and trout rations, experimental diets, and later on trout crumbles. Tanks are preferred because producers have more control of the water quality in tanks than in ponds.

Adult shortnose sturgeons are held in 0.5-acre ponds or cylindrical and raceway tanks (with a volume of 190 to 2,300 gallons) supplied with recirculated water. Tank-held adults feed on fish, squid, molluscs, crustaceans, worms, and beef liver; pond-held adults do not receive supplementation (Conte et al., 1988).

Bacterial agents that cause diseases in sturgeon include *Aeromonas hydrophilia*, *A. sobria, Pseudomonas* spp., *Edwardsiella tarda*, *Yersinia ruckeri*, *Streptococcus* spp., and, rarely, *Flavobacterium columnariae*. Factors that may predispose cultured sturgeon to bacterial diseases include stress factors, such as handling, and water quality problems, such as low DO levels, traces of hydrogen sulfide, and accumulation of organic loads on the bottom of holding tanks. *Streptococcus* spp. can be treated with erythromycin (100 milligrams/kilogram body weight daily for 10 days), and *Edwardsiella tarda* can be treated with daily oxytetracycline baths (Francis-Floyd, 2000).

4.3.6.7 Sunfish Family

Sunfish are produced for sport and foodfish, forage fish for predators including bass, and stocker fingerlings for recreational ponds. The sunfish family *(Centrarchidae)* is exclusive to North America and includes 30 species, the most popular of which are the bream (*Lepomis* spp.) and crappie (*Pomoxis* spp.).

Species from the genus *Lepomis* are commonly referred to as bream, sunfish, sun perch, or panfish. Only 4 out of the 11 *Lepomis* species are extensively cultured as sport fish. They are the bluegill (*Lepomis macrochirus*), redear sunfish (*Lepomis microlophus*), warmouth (*Lepomis gulosus*), and green sunfish (*Lepomis cyanellus*). The bluegill is probably the most well known of all sunfish species and has been stocked throughout North America as a game fish. It is most abundant in shallow, eutrophic lakes and ponds but can also be found in streams. The redear, also known as “shellcracker” and “chinquapin,” has also been stocked throughout North America as a game fish or used as a companion to bluegill in controlled systems, but it prefers sluggish waters. The warmouth, or “goggle-eye,” occupies sluggish waters and is not usually used to stock recreational waters. Its main use is for the production of hybrids with other primary *Lepomis* species. Green sunfish are also known to hybridize with other *Lepomis* species. They are found in a wide range of habitats (from ponds and lakes to river systems) and are perhaps the most adaptable and abundant of all the sunfish species.

The most popular size for stocking of bluegills, redears, and sunfish hybrids is 50 millimeters. Bluegills and redears are stocked as forage species for largemouth bass and also for sportfishing. There has also been newfound interest in using bluegills, redears, and some sunfish hybrids in nontraditional markets such as foodfish for human consumption and use in fee-fishing operations.

The two *Pomoxis* species, the black crappie (*Pomoxis nigromaculatus*) and white crappie (*Pomoxis annularis*), are cultured for stocking ponds, lakes, and reservoirs. Black crappies are common in Quebec and Manitoba provinces in Canada, the northern and eastern portions of the United States, and as far south as Florida and Texas. White crappies are common in southern Ontario, Canada, in Minnesota and states eastward, and as far south as the Gulf of Mexico.

Production Systems

Most culture of sunfish occurs in ponds. Spawning ponds should be less than 3 acres and 2 to 5 feet deep, with a smooth, evenly sloped bottom. It is recommended that the ponds be filled at least 2 to 4 weeks before spawning activity commences and that the ponds be completely free of any other fish species. A plankton bloom should also be established before the spawning activity begins. This can be accomplished through the use of organic or inorganic fertilizers. Groundwater is the preferred water source for production ponds, and the water level can be manipulated by drainpipes (Brunson and Robinette, 2000).

Culture Practices

It is critical to properly identify broodfish used for sunfish culture to ensure that the desired offspring are produced because *Lepomis* species have a tendency to hybridize. *Lepomis* broodfish spawn very soon after optimum temperatures have been reached. A powder or mash is usually the first food given, and then feed particles matched to the size of the fish are given as the fish grow. The fish are grown out to at least 2 inches before harvesting because smaller-sized fish stress easily (Brunson and Robinette, 2000).

Both *Pomoxis* species are cultured similarly. Usually, 2-year-old crappies are put into ponds to spawn, and they are given fathead minnows, threadfin, or gizzard shad as forage. Spawning and egg incubation proceed naturally in the open ponds. After crappie eggs hatch, the fish can be transferred to small raceways to be trained to accept prepared rations or pelleted feeds. They are then harvested as fingerlings.

Care needs to be taken during harvesting because handling stress can increase the incidence of columnaris disease. It has been found that harvesting fingerlings during winter can reduce handling stress, and that black and hybrid crappies endure handling stress better than white crappies (Brunson and Robinette, 2000).

4.3.6.8 Walleye

Walleye *(Stizostedion vitreum)* are raised as foodfish and for stocking purposes. Most commercial harvest of wild walleye in North America occurs on the Canadian shore of Lake Erie and in isolated lakes of western Ontario and the Canadian Prairie Provinces. In the United States, some tribes harvest a small amount of walleye on the Great Lakes for subsistence and also commercial purposes.

Production Systems

Several types of culture systems are used, including pond culture, tandem pond-to-tank culture, pond-to-tank-to-pond culture, cage culture, and intensive culture. In any of the culture systems, walleye eat diatoms, rotifers, and copepod nauplii, cyclopoid copepods, or small soft-bodied cladocerans when the fish are young. As they grow, their diet switches to larger cladocerans and then to immature aquatic insects.

Fingerling walleye can be produced in drainable ponds, with levees on all four sides, or undrainable ponds, which include farm and ranch ponds, shallow natural lakes, marshes, borrow pit ponds, and dug ponds. Drainable ponds are prepared by seeding pond bottoms with an annual rye grass, if there is adequate time between pond drainage and the next production cycle, or drying and disking the ponds if seeding is not possible. Additions of agricultural lime ($CaCO_3$) may be necessary to increase alkalinity, and additions of caustic (hydrated) lime ($Ca(OH)_2$) may be necessary to kill parasites after pond drainage. Ponds may be filled with groundwater or surface water, but surface water must be filtered so that unwanted organisms are not introduced to the pond. There is little information on pond culture in undrainable ponds. It is known that stocking densities in undrainable ponds are much less than in drainable ponds; however, there is a wide range of stocking densities in both types of ponds.

Tandem pond-tank culture is used to grow phase II fingerlings because it is hard to raise a large number of walleye to sizes over 4 inches in ponds (unless forage fish are added). Fingerlings are transferred from ponds to indoor culture tanks after they are accustomed to formulated feed diets and are raised to a size of 5 to 8 inches

In pond-to-tank-to-pond culture, phase II fingerlings are pond-raised and overwintered. In early spring they are transferred to cages in small ponds (0.16 acres), where they are put on formulated feed diets. Feed-trained fingerlings are then returned to ponds, where they remain on the manufactured feed diet, and raised for a few years to produce food-size fish. This culture method is uneconomical because of high mortality rates in all stages of the culture process (between fry stocking and fingerling harvest, during overwintering in ponds, during transfer of fingerlings from ponds to cages and to formulated feed diets, and during transfer back to ponds and another overwintering).

Walleye can also be raised in cages tethered to piers, docks, or rafts. This culture method has been used in water-filled gravel and rock quarries, natural and artificial lakes, and farm ponds. It has been used to raise fry to fingerlings, phase I pond-raised fingerlings to phase II fingerlings for enhancement stocking, and food-size fish. The survival rate of fingerlings from summer to fall is higher if feed-trained fingerlings are used instead of trying to train pond-raised fingerlings to take commercial feed in the cages.

Intensive culture refers to raising finfish in flowing water systems, such as flow-through systems, at a high density, and it encompasses single-pass (one-use), serial-reuse (stair-step raceway), and recirculating systems. These systems use high exchange rates of water, which allows for a good supply of oxygen in the culture tank and removal of dissolved wastes such as ammonia. Intensive culture is most often used to adjust phase I fingerlings to formulated feed and then to grow them to fall fingerlings. The feed-trained fingerlings can reach a marketable food-size when raised in intensive culture. Advantages of intensive culture include raising the fish indoors under optimum conditions, raising the fish where space or water supply is limited, and acclimating fingerlings or fry to formulated feed rations under controlled conditions (Summerfelt, 2000).

Culture Practices

In drainable and undrainable ponds, fingerlings can be partially harvested by trapping or seining; however, in drainable ponds, they are most often harvested all at once by being drained into a catch basin. A distinctive characteristic of walleye fingerlings is their attraction to light, allowing for easy capture in light traps to monitor populations. After the fish cease to have an attraction to light (when they are around 1.6 inches in size), sampling can be done through nighttime seining (Summerfelt, 2000).

Culture practices for raising walleye to food-size include combinations of the above-mentioned systems. Phase I fingerlings can be raised in ponds until the fish are about 1.25 to 2.5 inches in length, at which time they must be harvested so that fish density can be determined. The phase I fingerlings need to be trained to accept formulated feed, and this can be initiated in intensive culture systems or in ponds. Ponds must be restocked at densities suitable for growth of the fish to a larger size, and then fingerlings can be raised through the end of the growing season to an average size of 5 to 8 inches, when they are known as phase II fingerlings (Summerfelt, 1996). Phase II fingerlings must be overwintered in adequately aerated ponds and can reach sizes of 12 to 14 inches by the end of the second summer in southern Iowa and the middle to end of the third summer in more northern locations (Summerfelt, 1996). Disadvantages to pond culture of food-size walleye include the length of time for the fish to grow out to market size, potential winterkill, and potential summerkill in instances of prolonged high temperatures (Summerfelt, 1996).

Walleye can be raised to food-size in flow-through systems, such as raceways or circular tanks, as long as the tanks are covered to reduce intense sunlight, to which walleye are sensitive. The greatest limitation of these flow-through culture systems is the necessity for available water sources with desirable water temperature. Flow-through systems must have a plentiful supply of water in the 66 to 77 °F range because growth rates diminish to nearly zero at temperatures lower than 60 °F. Intensive culture has a much higher survival rate than ponds for growout of walleye to food-size (Summerfelt, 1996). Fry can be cultured intensively by feeding them brine shrimp or formulated feed, and then grown out to food-size. Another option is to transfer phase I fingerlings raised in ponds into intensive culture systems to be habituated to formulated feed (Summerfelt, 1996).

Recirculating systems are another choice for raising walleye to food-size. The systems can be used throughout North America, with new water use minimized to around 5% or less of the total system volume per day and fish stocked at high densities and raised on pelleted feeds (Summerfelt, 1996). Recirculating systems are advantageous because the controlled water temperature allows for a 12-month growing season. These systems also have low water requirements relative to production capabilities, produce a small volume of concentrated waste, and offer the opportunity to locate facilities near major markets.

4.3.6.9 Yellow Perch

Yellow perch *(Perca flavescens)* is a popular food fish with high market demand. It is a coolwater species found in the Great Lakes region and Canada. Yellow perch harvests from the Great Lakes surpassed 33 million pounds/year in the 1950s and 1960s, and market demand kept up with the large supply. In the 1980s and 1990s, harvests fell to between 11 million and 17.6 million pounds/year. Commercially cultured yellow perch now add to the supply, and the market demand is high for them because of their freshness and because of concerns regarding microcontaminants in wild-caught fish (Manci, 2000).

Production Systems

Most commercial yellow perch production is conducted in ponds, but there is also potential for cage culture (KSUAP, n.d.). Ponds are prepared by adding organic fertilizer to stimulate the growth of zooplankton, which acts as a food source for newly hatched fry (Wallat and Tiu, 1999).

Culture Practices

It takes about 18 months to grow yellow perch to a harvest size of 0.25 pounds. Some research has indicated that stocking yellow perch at high densities could be advantageous to production because the high densities stimulate feeding activity and allow for maximum growth (KSUAP, n.d.).

4.3.7 Baitfish

Baitfish is the term used to describe live fish sold as fishing bait or as "feeders," which are fish fed to ornamental fish and to invertebrates with piscivorous food habits (Stone, 2000). More than 20 species are caught in the wild and used for bait, but fewer species are raised on farms. Farmers face strong price competition from wild-caught bait, which has negatively affected the profitability of baitfish farming. If farm-raised fish cannot be supplied at a competitive price, the result is increased harvest pressures on wild stocks to meet market demands (Stone et al., n.d.). The common farm-raised species are the golden shiner (*Notemigonus crysoleucas*), the fathead minnow (*Pimephales promelas*), and the goldfish (*Carassius auratus*) (Stone, 2000). The baitfish industry is one of two non-food production sectors in U.S. AAP. (The other sector is ornamental fish production.) According to the 1998 Census of Aquaculture, the baitfish industry generated $37.5 million in total sales with 275 growers throughout the country (USDA, 2000).

According to the Census of Aquaculture, Arkansas leads the industry in production of baitfish in the United States, with 62 growers and $23 million in total sales; however, it is believed that the number of farms and the value of the industry are higher than the Census figures indicate. For example, Collins and Stone (1999) estimated the 1998 value of Arkansas baitfish production at $37.9 million. Compared to foodfish culture, baitfish culture is unique in the vast number of individual fish produced and the variety of sizes required by the market. In addition, the impact of competitive market forces plays a critical role in the baitfish industry. Demand for bait is seasonal, driven by regional customer preferences, and sensitive to weather conditions (Stone, 2000). Farmers monitor weekend weather forecasts for regions where their fish are sold to determine how many fish to harvest, grade, and harden in vats in anticipation of sales orders. For example, a warm winter means fewer days of ice fishing and a reduced market for minnows (Stone et al., n.d.)

Sources of baitfish include wild capture, extensive culture, and intensive culture. In the past, most baitfish were captured in the wild. In some areas, collecting small fish for bait is still legal, and commercial fishermen use seines or traps to harvest the fish. Farming fish for bait grew in response to shortages of wild-caught minnows in the 1930s and 1940s, as well as concerns over the possible depletion of wild stocks. Extensive culture, more common in northern states, is the practice of raising seasonal crops of fathead minnows or white suckers in shallow lakes. Fry are stocked in the spring and allowed to grow. The fish are raised on natural food alone. With this form of culture, the production yields are lower than those in intensive culture, which has a higher biomass within the production unit, but the costs incurred by the operator are also lower.

In 1934 the Michigan Department of Conservation began experimenting with minnow propagation (Stone, 2000). In the late 1940s through the 1970s, baitfish farms grew rapidly (Stone et al., 1997). Today about half of all baitfish are farm-raised (Stone, 2000). The first baitfish farms in Arkansas began in the late 1940s. In 1997 Arkansas had an estimated 27,800 acres under cultivation for baitfish (USDA, 2000). Most baitfish farm acreage produces golden shiners and fathead minnows (Stone et al., 1997). Golden shiners are the predominant species raised in Arkansas (Collins and Stone, 1999). Fathead minnows are the most common species raised in the North Central Region, which includes Illinois, Michigan, Minnesota, Ohio, South Dakota, and Wisconsin (Meronek et al., 1997). Goldfish are primarily raised in Arkansas and the southern part of the North Central Region (Gunderson and Tucker, 2000). Golden shiners are both wild-harvested and cultured in the North Central Region, while goldfish are only cultured (Gunderson and Tucker, 2000).

The production of farm-raised baitfish can help to minimize environmental impacts by reducing the demand for wild-caught baitfish. These fish are an integral part of the food chain for freshwater systems. Their decline could impact the entire ecosystem by reducing the number of forage fish. Also, the transfer of wild-caught baitfish from their native populations to other sites across the country raises concern about possible infiltration of non-native species.

4.3.7.1 Production Systems

Although culture practices vary with species and from farm to farm, most baitfish are raised in earthen ponds. Ponds used for golden shiners range in size from 5 to 20 acres, while ponds for fathead minnows are usually up to 10 acres (Stone, 2000). Ponds for goldfish are even smaller, with an average pond size of 2 acres. Water depth is relatively shallow, ranging from 2.5 to 6 feet to help farmers harvest fish without draining the ponds. Groundwater is used most often to fill ponds for baitfish culture. If surface water is used, farmers use fine-mesh, self-cleaning filters to prevent the introduction of wild fish into baitfish ponds. Golden shiners and fathead minnows are partially harvested from ponds during the year. Fish are baited into a corner and harvested by surrounding the fish with a seine. By the time the pond is emptied, the standing crop has been reduced to 25 to 50 pounds/acre.

4.3.7.2 Culture Practices

Golden shiners and goldfish have traditionally been propagated using either the wild-spawn method or the egg-transfer method (Stone, 2000). With the wild-spawn method, broodfish are stocked into newly filled ponds with aquatic vegetation in shallow water. Fish spawn freely on the vegetation, and then juveniles are either raised with their parents or transferred to another pond. In the wild-spawn method, fry are often vulnerable to predation by older generations of fish. Although fathead minnow growers generally use the wild-spawn method, most golden shiner and goldfish farmers use the egg-transfer method. In the latter method, spawning mats are used to collect eggs, and then the eggs are transferred to a rearing pond filled with a shallow layer of fresh well water (not filled to capacity) for incubation and hatching. Eggs hatch in 3 to 7 days.

Usually, eggs or fry are stocked into prepared ponds at higher densities, and when the juvenile fish are large enough, they are spread out into other ponds at lower densities. Juvenile fish can be stocked into ponds with adult fish once they are large enough to avoid being eaten. The growing season in the North Central Region is shorter (120 to 150 days) than that in Arkansas (180 days); therefore, the size attained by golden shiners and goldfish over a single growing season in the North Central Region is smaller (Gunderson and Tucker, 2000).

In preparation for stocking fry, ponds are fertilized to encourage the development of natural food. Golden shiners feed on zooplankton, but they also eat a wide variety of other animal and plant materials (Stone, 2000). Fathead minnows are primarily algae eaters, but they also eat zooplankton and insect larvae. Young goldfish feed primarily on zooplankton; as they age, they also feed on algae and detritus.

Feeding practices vary greatly among producers. Unlike in foodfish culture, the primary goal in baitfish production is not to grow the fish to market size as fast as possible; instead, producers manipulate the stocking density and feeding rate to produce a variety of sizes (Stone et al., 1997). Feeding rates for baitfish are determined by the market demand for various fish sizes. In Arkansas many farmers start feeding at 5 pounds/acre/day, then gradually increase to 10 or 15 pounds/acre/day. Most of the feed input to ponds is thought to contribute to the natural production of food organisms (Stone and Park, 2001, personal communication). Many baitfish farmers feed in one area of a pond, where aerators are placed, to attract fish for ease of harvest with seines. In the northern North Central Region, golden shiners are usually not fed prepared feed (Gunderson and Tucker, 2000).

Farmers also apply fertilizer to promote the growth of natural food. As a general rule, a single application of inorganic fertilizer for baitfish ponds should contain 3 to 4 pounds/acre of phosphorus (Stone et al., 1997). Organic fertilizers, such as vegetable meals, hay, and poultry litter, are normally used only for fry nursery ponds in combination with inorganic fertilizer. Fertilizer use has declined as farmers have switched to using prepared feeds, but natural food is still an important part of the baitfish diet (Stone et al., n.d.).

The biomass for baitfish in pond culture is low. Average yields for baitfish production are 350 pounds/acre for golden shiners and fathead minnows, and 790 pounds/acre for goldfish (Collins and Stone, 1999). In contrast, foodfish raised in ponds are stocked at approximately 6,000 pounds/acre.

4.3.7.3 Water Quality Management Practices

A common practice in Arkansas is to drain and pump water from pond to pond. The most common type of drain used in baitfish production ponds is the inside swivel drain (Stone et al., 1997). The swivel drain allows baitfish farmers to drain the pond from the top of the pond (the surface of the water) and minimize the release of solids during draining.

Water is transferred when ponds are drained to conserve water and reduce pumping costs. Drains are installed to transfer water between ponds, or diesel pumps are used to pump water from pond to pond. Boyd (1990) describes a method developed to reuse water on a large minnow pond in Arkansas. The farm installed pipes at the water level of adjacent ponds. When the pond is emptied, water is pumped into adjacent ponds and stored. After the pond is harvested, the cross pipes are opened, and the pond is refilled. Baitfish farmers in Arkansas routinely capture rainwater and prevent overflow from the ponds by maintaining pond water levels at least 6 inches below the overflow pipe. When a pond is emptied, water is often captured in a ditch and then transferred to another pond for reuse.

During the spring spawning season, a number of baitfish ponds are drained to make room for the new crop of fish (Stone et al., n.d.). A pond being prepared for fry is drained to adjacent ponds. After drying to ensure that organisms that could eat the small fry will not be present, the pond is filled to a depth of about 1 foot with well water. The well water is fertilized, and as the fry grow larger, water from the adjacent ponds is transferred back. Ponds are drained sequentially, so that old water from one pond can be used to top off ponds with new fry. (During incubation and hatching, these ponds contain only a shallow layer of fresh well water.) The old water has the advantage of containing natural foods for the young fry. Generally, this is the only time of year at which any discharge reaches receiving streams (Stone et al., n.d.). The volume of discharge is typically less than the volume of the pond because of water management practices that support the transfer and reuse of pond water.

Few data are available on water quality in commercial baitfish ponds or on effluents from these ponds; however, the impact is likely to be minimal. Baitfish production uses low biomass stocking densities. Also, current management practices within the industry reduce potential impacts of effluent discharges. Farmers seine by hand to prevent stirring up sediments because small baitfish are sensitive to muddy conditions. Farmers begin with a low biomass and lower the biomass density even further with partial harvests throughout the year. The combination of low biomass and reduced feed input prior to draining makes it likely that baitfish effluents will have lower solids concentrations than effluents from catfish ponds (Stone et al., n.d.). Also, it is likely that farmers' efforts to conserve water have also reduced effluent quantities.

4.3.8 Ornamental Fish

The culture of ornamental, or tropical, fish is primarily to supply animals for the home aquarium where fish are kept as a hobby or as pets. The ornamental fish industry is one of the two major non-food production sectors in the AAP industry; the baitfish industry is the other sector. Although many freshwater ornamental species are cultured, some examples are guppies *(Lebistes reticulates*), mollies (*Mollienesia* sp.), swordtails (*Xiphophorus* sp.), tetras (*Hemigrammus* sp.), gouramis (*Osphroneums, Sphaerichthys, Trichogaser* sp.), and goldfish (*Carassius auratus auratus*). More than 1,000 freshwater species in about 100 families are represented in the ornamental fish trade at any one time; however, only about 150 species are in great demand and account for the largest volume of trade (Chapman, 2000). Most ornamental fish currently produced in the United States are freshwater fish. Nearly 80% of the freshwater ornamental fish sold in the United States are raised in confinement, and the majority of those are raised in pond operations (Stoskopf, 1993). The production of marine ornamental fish is an emerging industry with few species regularly reproduced in captivity. Some of the most common species available from marine culture facilities include the clownfish (*Amphiprion* spp. and

Premnas biaculeatus), the neon goby (*Gobiosoma oceanops*), and the dottyback (*Psuedochromis* spp. and *Ogilbyina novaehollandiae*).

According to the 1998 Census of Aquaculture, there are 345 ornamental fish farm operations in the United States, which produce roughly $68 million in total sales (USDA, 2000). Florida, with 171 growers, dominates the domestic ornamental fish industry, with approximately $56 million in total sales, or 81% of the total sales in ornamental fish species in 1998. California, Arkansas, Indiana, and Hawaii also produce ornamental fish.

Ornamental fish culture may benefit wild ornamental populations by preventing destructive collection practices, which deplete wild populations and degrade natural habitat. The Asia Pacific region is the global center of marine diversity; it supports more species of coral and fish than any other region in the world (Holt, 2000). This region is home to 4,000 species of reef fish and more than one-third of the world’s coral reefs. In this region and throughout the tropics, natural populations of coral reef fish, which make up the majority of marine ornamental species, are increasingly threatened by development, dredging, coral collecting, and the live foodfish and aquarium fish trade (Holt, 2000). Many common collection methods, which include the use of dynamite and sodium cyanide, are destructive and cause damage to coral reef habitats. Loss of habitat reduces the area available for the settlement of new fish recruits.

4.3.8.1 Production Systems

Ornamental fish farming is characterized as an extensive culture (very low biomass densities) and often has two phases of production: a hatchery phase and a growout phase. Most breeding for ornamental fish takes place in recirculating systems, while the growout phase usually occurs in ponds. In many cases, ornamental fish farms are small businesses owned and operated by a family (Chapman, 2000). Farms often use a combination of both indoor and outdoor facilities for production. Indoor areas, usually built from modified greenhouses and wooden and steel sheds, are used primarily for breeding, hatching eggs, and raising larvae or fry. The remaining indoor area is used for holding, sorting, and shipping fish. The most common outdoor facilities are earthen ponds and concrete tanks. In Hawaii, broodstock of live-bearing ornamental fish are typically held in net cages in ponds, and then the juveniles are transferred to ponds for growout (JSA, 2000b). Net cages are small floating structures that allow water to flow through while retaining the confined animals (Stickney, 2000d). They are used for raising early life stages of various species, or sometimes to hold fish in advance of spawning. In Florida, live-bearer production is typically done in ponds.

Pond Systems

Although some ornamental fish are raised in recirculating systems, most are produced in outdoor earthen ponds (Watson and Shireman, n.d.). In Florida these ponds are almost all water-table ponds in sandy loams. Because the water table is so close to the surface, ponds can be created by digging out the appropriate area and letting the pond fill with water from the water table. The water level in the pond is dependent on the existing hydrology. In many areas, during dry seasons, well water is used to supplement the water table source. A typical outdoor pond in Florida is approximately 65 to 82 feet in length, 20 to 30 feet wide, and 5 to 6 feet deep. Farmers often cover outdoor ponds and tanks with nets to protect the fish from predators, or they use plastic to provide shade and maintain water temperatures, depending on the time of year (Chapman, 2000).

A typical growout pond (approximately 2,152 square feet) may be stocked with 10,000 to 80,000 fish from egg-laying parents or with around 200 live-bearing broodfish (Chapman, 2000). After 2 months, the live-bearing population in the pond can reach 30,000 fish. Growout ponds for juvenile freshwater ornamental fish are prepared for stocking by draining the pond after each production cycle and washing and preparing the bottom. After washing, the ponds are disinfected with hydrated lime to ensure that all predators are eliminated from the system. Although specific to individual species, some ponds remain in production unwashed for 1 to 2 years (Chapman, 2000). The ponds are fertilized to stimulate the growth of phytoplankton in the water. Organic fertilizers are used to sustain a release of nutrients over a longer period of time. Cottonseed meal is a common organic fertilizer used by ornamental fish producers. Inorganic fertilizers provide a short-term nutrient release and are often used to initialize phytoplankton growth. After the ponds are fertilized, they are filled with water and an algae bloom is allowed to develop to encourage the creation of a natural food source.

Recirculating Systems

Although recirculating systems are used primarily for the hatchery phase of ornamental production, producers are exploring opportunities to expand growout production, using technologies from recirculating systems (JSA, 2000b). Stocking densities are higher in recirculating systems than in ponds, approaching 15 fish/gallon without oxygen injection and 58 fish/gallon with oxygen injection. Water for facilities using recirculating systems is often treated internally with mechanical and biological filters (Chapman, 2000). Internal processes within recirculating systems include settling basins, baffles, screens, and upflow solids contact clarifiers to remove suspended and settleable solids. To break down organic wastes, some culturists use microbes in trickling filters and modified upflow clarifiers. To disinfect treated water, some culturists use ozone and/or ultraviolet light.

Ozone is also used to oxidize organic compounds. Fine suspended solids and other dissolved organics are stripped with dissolved air flotation or foam fractionation technology.

4.3.8.2 Culture Practices

With the exception of a few species like koi and goldfish, most ornamental fish are native to tropical regions of the world and cannot tolerate temperatures below 64 °F. Ornamental fish are relatively small in size, with a market weight between 0.1 and 1.4 ounces and length between 0.8 and 6 inches. Aquarium fish usually live from 6 to 10 years; however, some koi have been recorded as living as long as 70 to 80 years (Chapman, 2000).

Based on their reproductive cycles, freshwater ornamental fish are divided into egg layers and live-bearers. Egg-laying fish deposit their eggs on spawning mats or broadcast them for external fertilization. Live-bearing fish, such as guppies, release fully developed young that are ready to feed on their own.

Most egg layers are artificially bred in indoor hatcheries. Broodfish are paired in tanks or spawned together in large groups (Chapman, 2000). Fish are stimulated into breeding by using spawning mats and by manipulating the temperature, flow, pH, and hardness of the water. Culturists sometimes use hormones like human chorionic gonadotropin or carp pituitary extract, which are injected into individual fish to induce spawning. After spawning the eggs either are allowed to hatch where they are laid or are collected and placed in incubators. The larvae that hatch are pooled and transferred to rearing tanks or outdoor ponds. The fertilization and spawning of live-bearing fish is allowed to occur naturally in breeding ponds or tanks. In production, live-bearing parents are usually separated from their offspring to prevent cannibalism.

Many of the more popular and expensive marine ornamental fish, such as butterfly fish, angelfish, and wrasses, are difficult to raise in captivity. Clownfish, neon gobies, and dottybacks are easier to raise because they can change sex; therefore, a spawning pair is not needed. Clownfish have eggs that take several days to hatch, and they produce larvae that are large enough to feed on rotifers when they hatch. In captivity, gobies spawn regularly every 2 to 3 weeks and produce large eggs. Young can be raised on rotifers and zooplankton and, later, brine shrimp nauplii. Like gobies, dottybacks produce large larvae that grow quickly.

In general, it takes 3 to 6 months to produce market-ready fish. The typical survival rate for freshwater ornamental fish in a pond is 40% to 70%. Most losses in outdoor culture systems are due to predation, deterioration of water quality, and disease. Fish are harvested with fine seine nets, dip nets, and traps (Chapman, 2000). The process for harvesting ornamental fish differs from that for foodfish because the fish are individually selected and must be kept alive. Ornamental fish are sorted by hand, based on color and size. Mechanical graders are not yet available for the ornamental industry.

Feed Management

There is very little published information on the nutrition and feeding of ornamental fish. Most dietary knowledge has evolved from trial-and-error tests by individual farmers and a few studies in research laboratories (Chapman, 2000). Although most producers rely on a natural food sources for fish in outdoor ponds, these sources are sometimes supplemented with formulated feed. Fish raised in indoor tanks are fed commercial feed mixtures. Feed is delivered by hand or automatic feeder. Because of the small particle size of the feed and the low volume of feed used for the growout phase, feed is often allotted at a constant rate of 3% to 10% of fish biomass per day for freshwater ornamental fish (Chapman, 2000). Because the biomass for ornamental fish production is small, the feed input is also small.

Health Management

Parasites and bacteria are the two most common causes of infectious diseases in ornamental fish. The most common external parasites are ciliated protozoans, primarily *Ichthyophthirius multifiliis*, or “ich,” and *Trichodina.* Common treatments for external parasites include salt, formalin, copper sulfate, and potassium permanganate. The most prevalent infectious bacteria are in the aeromonad and columnaris groups. Common drugs used to treat bacterial infections are tetracycline, erythromycin, mitrofurazones, nalidixic acid, potassium permanganate, and copper sulfate (Chapman, 2000). Drugs are not often used in pond systems because of the high cost to treat a large volume of water. Drugs applied for ornamental culture are more commonly used in tanks for indoor recirculating systems (Watson, 2002, personal communication).

4.3.8.3 Water Management Practices

While there are few data in the literature on ornamental fish farm effluent characteristics, the impact from water discharged from ornamental fish production facilities is likely to be minimal. Assuming the average size of a growout pond is 2,152 square feet, with approximately 80,000 gallons of water, ornamental culture facilities typically discharge the volume of one pond, or less, per year (Watson, 2002, personal communication). Also, ornamental fish are extremely sensitive to water quality; therefore, water quality in the production system is constantly monitored by producers. Many producers are already implementing BMPs to reduce the impacts of effluents. For example, when ponds are drained, some facilities discharge water into settling basins, while others discharge into channels and ditches that run into surface waters. In Florida, ornamental fish farm effluents are regulated by the Florida Department of Agriculture and Consumer Services. The producer agrees to adhere to a set of BMPs, most of which deal with treatment of effluent prior to discharge (JSA, 2000b). When in compliance with Florida's BMP program, ornamental fish producers are issued an aquaculture certificate to verify their compliance. This program has a high compliance rate, estimated at 95% of the ornamental fish producers in Florida (Watson, 2002, personal communication). Because consumers and distributors often choose to buy fish only from certified aquaculture facilities, the demands of the market reinforce compliance with the BMP program.

4.3.9 Shrimp

Most commercial shrimp farms in the United States produce Pacific white shrimp (*Penaeus vannamei*), which were introduced from the Pacific coast of Central and South America, for a single annual crop (Iversen et al., 1993). According to the 1998 Census of Aquaculture (USDA, 2000), Texas is the leading producer of cultured shrimp in the United States, producing 3.7 million pounds/year with a value of $9.3 million. Hawaii, with 12 farms, produced 197,000 pounds with a value of $1.7 million. South Carolina, with six farms, produced approximately 43,000 pounds of shrimp annually. Overall, there are 42 shrimp farms in the United States that produce a total of 4.2 million pounds/year and generate sales of $11.6 million. Blue shrimp *(P. stylirostris*) from the Pacific coast of Central and South America and giant tiger prawn (*P. monodon*) from the western Pacific have also been introduced into the United States for shrimp farming.

4.3.9.1 Production Systems

Although shrimp can be raised in tanks, raceways, or ponds, most commercial facilities raise shrimp in levee ponds. Penaeid shrimp ponds rely on access to supplies of seawater. In general, shrimp farming in the United States takes place in coastal areas, primarily along estuary systems or waterways, such as tidal rivers or canals. A facility must be able to obtain seawater from the ocean, adjacent estuaries, or a reservoir. Pumping systems are used to transfer water to the ponds, and some facilities maintain reservoirs with supplemental supplies of seawater. Shrimp ponds usually have water gate inlets and outlets to fill and drain the pond. The gates are covered with screens to keep out unwanted predators and to prevent the escape of non-native cultured species to the receiving waters.

4.3.9.2 Culture Practices

In the wild, shrimp mate in the ocean. A single female can spawn 100,000 eggs or more at a time (Boyd and Clay, 1998). Within 24 hours of fertilization, the eggs hatch into larvae and begin feeding on plankton. The nauplius is the first larval stage. After approximately 12 days, the larval period ends and the young shrimp, now postlarvae, are carried on currents from the open ocean into nutrient-rich bays and estuaries. There they transform from organisms suspended in the water column into bottom-dwelling animals. Maturation from postlarvae to juveniles generally takes 4 to 5 months (Treece, 2000). In the late juvenile or early adult stage, the shrimp return to the ocean to mature and mate.

Culture for marine shrimp has three phrases—hatchery, nursery, and pond growout. Many shrimp producers rely on hatcheries that specialize in the production of postlarvae or juveniles for supplies of animals to stock their growout ponds. Shrimp hatcheries require relatively small tracts of land compared to growout facilities (Treece, 2000). Broodstock shrimp are harvested from the ocean and brought to the hatchery for sexual maturation and reproduction. Mated females harvested from the wild are allowed to spawn in a nauplii production facility. Some hatcheries prefer to control all production inputs; therefore, they harvest both males and females from wild stocks and quarantine them to ensure they are free of disease and other pathogens. The most important parameters for successful maturation of penaeid shrimp are constant temperature and acceptable levels of salinity, pH, light, and nutrition. Hatcheries rely on a readily available supply of high-quality seawater for successful shrimp maturation.

Shrimp are stocked in hatchery tanks at densities of 5 to 7 shrimp/10 square feet. The tanks are about 13 feet in diameter and are supplied with water through a flow-through system or a recirculating system (Treece, 2000). Most hatcheries now recirculate roughly 80% of the water to maintain better control over water quality (Treece, 2000). Once hatched, the young larvae (nauplii) are disinfected and evaluated for physical attributes. Nauplii with suitable physical characteristics are transferred to larval rearing tanks and stocked at densities ranging from 379 to 568 nauplii/gallon (Treece, 2000). At the postlarvae stage, shrimp are transferred from the larval rearing tank to a postlarvae-rearing/holding tank. Once the postlarvae have reached the PL8-18 stage (8 to 18 days old), they are usually sold to production farms for growout. Nursery ponds are smaller ponds used for an intermediate growout phase and to eliminate substandard juveniles. Not all farms use the nursery phase. Many farms stock postlarvae, either from the wild or from the hatchery, directly into growout ponds.

Climate plays an important role in shrimp production in the United States. Compared to tropical locations, the cooler climate in the continental United States limits outdoor shrimp culture to 9 months in southern regions of the country. Growout ponds are stocked in the early spring. Based on the characteristics of a typical facility from a 1998 report prepared for EPA, growout ponds are usually stocked at densities of 50,000 to 75,000 postlarvae/acre. Adult shrimp are harvested in the fall (September through

November) approximately 140 to 170 days after stocking (SAIC, 1998). Shrimp are usually harvested by draining the pond and collecting the shrimp in bags or containers on the outside of the pond at the end of the drainpipe. Shrimp can also be harvested by pumps that draw the shrimp out of the pond with a vacuum suction. Growout ponds remain dry throughout the winter. Most shrimp farmers manage bottom sediments by allowing the ponds to dry naturally, then mechanically tilling the pond bottoms.

Feed Management

In early spring growout ponds are filled with water from a nearby estuary. Inorganic fertilizer is added to the ponds to promote plankton growth. Postlarval shrimp feed on plankton and a commercial feed supplement for several weeks after stocking. Four to six weeks after stocking, the shrimp are large enough to receive pelleted feed. The shrimp are fed by broadcasting feed into the ponds with mechanical feeders. To prevent overfeeding, most marine shrimp farmers feed at least twice a day and use feeding trays to monitor consumption. Feed placed on the feeding trays is visually inspected ½ to 1 hour after being placed in the ponds to evaluate feed use. Feeding rates and quantity are determined by visual water quality, feeding tray assessments, and percent body weight increase (SAIC, 1998).

Health Management

Viruses frequently cause high mortalities in shrimp crops and limit shrimp farming production. More than 20 known viruses are associated with penaeid shrimp culture; however, only 4 of these pose a serious threat to the shrimp culture industry (Treece, 2000). The four disease-causing viruses that affect marine shrimp culture are infectious hypodermal and hematopoietic necrosis (IHHN) virus, taura syndrome virus (TSV), white spot syndrome virus (WSSV), and yellow head virus (YHV).

There are several theories on possible sources for shrimp viruses. These include entry to the facility through contaminated feed, infected broodstock or seed, and bird or animal transport. Two other potential sources are carrier organisms in ship ballast water and frozen seafood products (Browdy and Holland, 1998). Current treatment options for shrimp diseases are similar to traditional livestock disease treatment methods. Shrimp diseases are not harmful to humans due to the freezing and cooking processes typically conducted prior to consumption (Iversen et al., 1993). Facilities that do have an outbreak of disease dispose of the contaminated stock and water, and then sanitize the pond facilities. Ponds are chlorinated, dechlorinated, quarantined, and inspected before reuse (SAIC, 1998). Many shrimp facilities buy specific pathogen-free (SPF) or specific pathogen-resistant (SPR) shrimp to reduce disease outbreaks. Shrimp hatcheries are developing a new strain of *P. stylirostris* that is resistant to TSV and WSSV.

In addition to concern for the health of cultured species, there is concern for wild native populations, which can be infected by viruses carried out of an AAP facility through the discharge of pond effluent, processing plant wastewater, pond flooding, the escapement of cultured species, and the use of infected bait shrimp (SAIC, 1998). The spread of shrimp viruses is one of the most important problems limiting shrimp culture production worldwide (Browdy and Bratvold, 1998). Control of disease will depend on the development of biosecure production systems, which prevent pathogen transfer and establishment. Researchers at the Waddell Mariculture Center in South Carolina are exploring ways to create biosecure systems by identifying paths of pathogen transfer and evaluating existing technologies.

4.3.9.3 Water Quality Management

Shrimp farmers use aeration, water exchange, management of stocking densities, and feed management to improve water quality and support healthy stocks of shrimp. Shrimp production ponds are aerated to maintain sufficient levels of DO and to keep the water column well mixed. Shrimp farmers typically use more aeration per acre than finfish farmers because shrimp farmers must maintain sufficient oxygen levels on the pond bottom where the shrimp live. Good pond aeration also encourages natural processes within the pond to assimilate nutrients and wastes and to reduce total pollutant loads to receiving waters when pond water is discharged (Boyd, 2000).

After the shrimp are harvested in the fall, the ponds are drained and left to dry. This oxidizes the organic matter and reduces the likelihood of disease problems from growing season to growing season. Most shrimp facilities use surface water as a source and screen the inlets to prevent predators from entering the ponds. Because many of the shrimp grown in the United States are non-native species, escapement and disease are concerns for regulatory agencies. Outlets are screened to prevent escapement. Water is often reused by draining it into closed ditches, allowing sediments to settle, and then moving the water back into the ponds from the ditches.

In the past, water use in shrimp pond production was high, with average water exchange rates ranging from 8% to 23% of the pond volume per day to flush the pond system (Hopkins et al., 1993).

In a 1991 study, Hopkins et al. compared the effect of a typical exchange rate of 14% of the pond volume per day to the effect of a lower exchange rate of 4% on the growth and survival of *P. vannamei* stocked at 76 animals/square meter and found no difference in productivity (Table 4.3–9) (Hopkins et al., 1991). Hopkins et al. (1993) studied the effects of high water exchange at 25% and low water exchange at 2.5% on ponds stocked with *P. setiferus* at 4.1 postlarvae/square foot. Nutrient concentrations were higher in the pond with the lower exchange rate, but the total mass of pollutants discharged was lower. Growth and survival were good under both exchange conditions, with a higher production in the pond with the reduced exchange.

Table 4.3–9. Water Quality of Inlet Water and Various Water Exchanges (Mean Values) of Shrimp Stocked at a Density of 4.1/Square Foot

Water Exchange Treatment	*Mean Size (lb)*	*Survival (%)*	*TSS (mg/L)*	*BOD (mg/L)*	*DO (mg/L)*	*Organic Solids (mg/L)*
Inlet water	N/A	N/A	178.9	1.5	N/A	132.2
Normal exchange (25% per day)	0.035	81.9	183.3	8.5	5.4	122.5
Reduced exchange (2.5% per day)	0.040	79.5	196.2	14.7	5.0	115.4
No exchange (0% per day)	0.041	0.2	157.3	18.8	4.8	85.3

Source: Hopkins et al., 1991.

Currently, shrimp farmers rely on lower water exchange rates. Aeration is preferred over water exchange to enhance DO levels (Browdy et al., 1996). In the early 1990s Texas shrimp farms, under the requirements of more strict water quality and discharge regulations, initiated a shift in water use practices. Using semiclosed systems, farmers began reusing and recirculating water within the facility. In 1998, one Texas farm, Arroyo Aquaculture Association (AAA), produced more than 1.4 million pounds of shrimp on 345 acres, or approximately 4,000 pounds/acre, in a semiclosed system (Treece, 2000). The farm decreased its water use from 4,500 gallons/pound of shrimp produced in 1994 to 300 gallons/pound of shrimp produced in 1998 through 2000. Most of the water added is used to fill ponds and offset evaporation.

Shrimp farmers like AAA have also decreased their stocking densities and increased aeration to promote optimum conditions for shrimp production. AAA decreased its stocking density from 4.7 to 3.3 shrimp/square foot and increased its aeration from 8 to10 horsepower/acre (Fish Farming News, 2000). Research and industry practices have demonstrated that water exchange rates can be reduced without affecting shrimp production as long as DO levels are maintained.

4.3.9.4 Effluent Characteristics and Treatment Practices

The composition of pond effluents during water exchange, overflow after heavy rains, and initial stages of pond draining is similar to that of catfish pond water (Boyd and Tucker, 1998). Marine shrimp AAP facilities have two types of discharges: routine water exchange and water drained during harvest.

Shrimp pond effluents can have high concentrations of nutrients and suspended solids, high BOD, and low levels of DO. When discharged into receiving waters, effluents with high levels of suspended solids can cause turbidity, which can reduce light available for photosynthesis. Low DO levels can affect estuarine organisms in the receiving waters, and excessive nutrients can accelerate plankton growth, resulting in die-offs and increased BOD in receiving waters.

There is some evidence to suggest that effluent characteristics for marine shrimp ponds are similar to effluent characteristics for catfish farms (Boyd and Tucker, 1998). For example, as stocking densities increase, the quality of effluents deteriorates. In a study by Dierberg and Kiattisimkul (1996), data presented (Table 4.3−10) show average concentrations of water quality variables in effluent from shrimp (*P. monodon*) stocked at different rates. The quality of effluent declines for stocking densities above 3.7 shrimp/foot.

When shrimp ponds are drained, the effluent is almost identical in composition to pond water until about 80% of the pond volume has been released (Boyd, 2000). During the draining of the final 20% of the pond volume, concentrations of (BOD_5), TSS, and other substances increase because of sediment resuspension caused by harvest activities, crowding of agitated shrimp, and shallow and rapidly flowing water. The average BOD_5 and TSS concentrations often are about 50 milligrams/liter and 1,000 milligrams/liter, respectively (Boyd, 2000). The draining effluent contributes more to potential pollution than water exchange at 2%. Settling basins offer a treatment method for effluent released during shrimp harvest, especially for the highly concentrated final 20%. Settling basins or ponds remove coarse solids and the BOD_5 associated with them. Studies have shown that 60% to 80% of TSS and 15% to 30% of BOD_5 can be removed in a settling basin with only 6 to 8 hours of holding time (Teichert-Coddington et al., 1999). Settling basins also reduce TSS levels.

Table 4.3–10. Composition of Discharge Waters from Ponds Stocked at Different Densities of *Penaeus Monodon*

Variable	***Stocking Density (shrimp/ft^2)***				
	2.8	***3.7***	***4.6***	***5.7***	***6.5***
Nitrite-nitrogen (mg/L)	0.02	0.01	0.06	0.08	0.08
Nitrate-nitrogen (mg/L)	0.07	0.06	0.15	0.15	0.15
Total ammonia nitrogen (mg/L)	0.98	0.98	6.36	7.87	6.50
Total nitrogen (mg/L)	3.55	4.04	14.9	20.9	17.1
Total phosphorus (mg/L)	0.18	0.25	0.53	0.49	0.32
Biochemical oxygen demand (mg/L)	10.0	11.4	28.9	33.9	28.8
Total suspended solids (mg/L)	92	114	461	797	498
Chlorophyll a (µg/L)	70	110	350	460	350

Source: Dierberg and Kiattisimkul, 1996.

Based on the 1998 report for EPA, settling ponds are the method of water treatment most commonly used by shrimp facilities discharging effluent (SAIC, 1998). Based on the facilities monitored, some commercial farms discharge as much as 600 million gallons/year (MGY), while others report zero discharges. One facility has a 20-acre settling area where discharged pond water remains for 2 days before being discharged into receiving waters. Another facility uses weirs to allow discharged water to drop 10 feet before entering a drainage ditch. Many drainage ditches are designed as settling basins to trap solids from effluent discharged from ponds.

In addition to reusing water during production in a closed ditch system, AAA uses drainage ditches equipped with aerators to serve as settling basins for water discharged during harvest. This facility also uses weirs so that the water discharged drops 10 feet into the drainage ditch, helping to promote natural aeration and mixing. Drainage ditches are periodically monitored to ensure that the BOD levels are in compliance with the state standard of 6 milligrams/liter. Arroyo also uses screens on its effluent pipes to capture foam and prevent its transfer to receiving waters. Also, water drained from the ponds during the yearly harvest is collected and allowed to settle in empty ponds for 15 days before being released into the drainage ditches.

The Southern Star facility (Texas) has a constructed wetland area that is used to treat effluent from shrimp ponds. The wetland was constructed by building a dike around 100 acres of previously unused land adjacent to the facility. The wetland is designed to treat discharged wastewater and then filter recirculated water back to the ponds for reuse (SAIC, 1998).

Harlingen Shrimp Farms, located in Texas, is one of the largest shrimp farming operations in the United States. Pond effluent is usually discharged through water exchanges that begin 30 days after stocking the ponds, and all growout ponds are drained for harvesting 140 to 170 days after stocking. Routine water exchange rates of 10% to 20% occur until DO level fluctuations stabilize. Each pond is equipped with six to fifteen 8-inch pipes and one 35-inch gate for draining water during harvest (SAIC, 1998).

4.3.9.5 Freshwater Prawn

The Malaysian prawn (*Macrobrachium rosenbergii*), a freshwater prawn, has been cultured on a limited scale in the United States (KSU, 2002). The primary economic challenges associated with culturing shrimp in the United States are the availability of low-cost, high-quality feed; shorter growing seasons, with only one crop per year in some areas due to temperatures; the high cost of land and labor; high operating costs; foreign competition; and price fluctuations (Treece, 2000).

The Malaysian prawn spends part of its natural life cycle in saltwater. Adult shrimp migrate down rivers to estuaries to have their young. The prawns spend their early larval lives in brackish water, migrating to freshwater as juveniles and remaining there as adults (Iversen et al., 1993). The larvae feed by sight on zooplankton, worms, and larval stages of other aquatic invertebrates. Larvae undergo 11 molts before transforming into postlarvae. Transformation from newly hatched larvae to postlarvae requires 15 to 40 days, depending on food quality and quantity and temperature. After their metamorphosis to postlarvae, prawns change from living suspended in the water column to dwelling principally near the bottom (D'Abramo and Brunson, 1996a). Postlarvae can tolerate a range of salinities. They migrate to freshwater upon transformation, where they take on a bluish to brownish color as they change to the juvenile stage. Postlarvae are juveniles, but the common usage for the term *juvenile* is to describe freshwater prawns between postlarva and adult (D'Abramo and Brunson, 1996a).

Production Systems

As in penaeid shrimp production, most freshwater shrimp culture facilities use earthen ponds to produce shrimp. Ponds used for raising freshwater prawns have many of the same features as ponds used for the culture of channel catfish. Surface areas for growout ponds range from 1 to 5 acres, but some producers use larger ponds. Ponds are usually rectangular with a minimum depth of 2 to 3 feet at the shallow end and a maximum depth of 3.5 to 5 feet at the deep end (D'Abramo and Brunson, 1996b).

Culture Practices

As in penaeid shrimp culture, there are three phases of culture for freshwater prawns—hatchery, nursery, and pond growout. Many prawn producers purchase juveniles for the pond growout phase. Commercial hatcheries in Texas, California, and Mexico produce postlarvae and juveniles (D'Abramo and Brunson, 1996b).

Ponds are filled and then fertilized to provide natural food for the prawns and to create a phytoplankton bloom to shade out unwanted bottom plants. Juveniles are usually stocked at densities of 12,000 to 16,000 per acre. The length of the growout period depends on the water temperature of the ponds, but it is generally 120 to 180 days in the southern United States (D'Abramo and Brunson, 1996b). At the end of the growout season, prawns are harvested by seine or by draining the pond. For seining, the water volume is decreased by one-half before seining. During drain-down harvests, prawns are usually collected outside the pond levee as they travel through a drainpipe to a collecting device (D'Abramo and Brunson, 1996b). Some producers selectively harvest large prawns 4 to 6 weeks before the final harvest. After the harvest prawns are chilled and then marketed fresh on ice. They may be processed and frozen, or frozen whole for storage and shipment.

Feed Management

Juveniles stocked in growout ponds initially feed on natural pond organisms. As the juveniles grow to a weight of 0.011 pounds or greater, prawns are fed a manufactured feed. Channel catfish feed with 28% to 32% crude protein can be used for prawns. The feeding rate is determined by the mean weight of the population.

Health Management

Diseases do not appear to be a significant problem in freshwater prawn culture; however, as densities are increased, diseases are likely to be more prevalent (D'Abramo and Brunson, 1996b). Blackspot disease, also called shell disease, could affect freshwater shrimp. This disease is caused by bacteria that break down the outer skeleton.

Water Characteristics and Effluent Treatment Practices

Like catfish ponds, freshwater prawn ponds use aerators to maintain adequate DO levels and prevent thermal stratification. Farmers monitor dissolved levels in the bottom 1 foot of the pond water to make sure that DO concentrations do not fall below 3 parts per million. A common method in freshwater prawn culture is the use of full-time or nightly aeration. Farmers typically use 1 horsepower/acre (D'Abramo and Brunson, 1996b). Because standing crops rarely exceed 1,000 pounds/acre, this level of aeration is usually sufficient to prevent oxygen depletion. Some farmers use only emergency aeration as needed. Unlike marine shrimp production, there is no water exchange for freshwater prawn production. Nutrients in the pond are partially assimilated by pond processes (Boyd and Tucker, 1998).

There are very few data available in the literature describing the characteristics of effluent from freshwater shrimp ponds or effluent management practices associated with these ponds.

4.3.10 Crawfish

Red swamp crawfish (*Procambarus clarkii*) and white river crawfish (*Procambarus acutus acutus*) account for about 90% of all crawfish cultured in the United States (Davis, n.d.). Currently, crawfish represent the only crustacean species cultured on a large-scale basis in the United States (USDA, 1995). As a commercially available food source, crawfish can be traced back to New Orleans French Market records from the 1800s (LSU, 1999). A commercial fishery for wild crawfish was developed in the 1940s in the Atchafalaya River swamp in Louisiana, where crawfish are still harvested today. Because catches from the wild were unpredictable and driven by seasonal changes, an increase in consumer demand for a year-round supply eventually led to the development of a crawfish AAP industry in Louisiana (de la Bretonne and Romaire, 1990b).

In 1993 more than 59.5 million pounds of crawfish with a value of $26.7 million were produced in Louisiana on more than 143,000 acres of ponds operated by 1,618 producers (USDA, 1995). Production in Louisiana represents over 90% of the total U.S. farmed production for crawfish, 70% of which is consumed locally (de la Bretonne and Romaire, 1990a). In addition to Louisiana, some 21,000 acres of ponds are used for culturing crawfish in Texas; Mississippi, Maryland, South Carolina, North Carolina, Florida, Georgia, and California also have commercial crawfish farms. There are also some smaller producers in the midwestern and northeastern United States that culture crawfish for fish bait (Eversole and McClain, 2000).

4.3.10.1 Production Systems

Culture methods used to grow crawfish complement farm management plans by using marginal agricultural land, permanent farm labor, and farm equipment in the off-peak agricultural farming periods (de la Bretonne and Romaire, 1990b). There are two types of crawfish ponds: permanent ponds and rotational ponds (LSU, 1999). Permanent ponds are ponds that remain in the same location and have a continuous management plan applied year after year. Rotational ponds describe the practice of rotating the annual sequence of crops grown in a pond or rotating the physical location of the field in which crawfish are grown.

Permanent Ponds

Approximately half of the ponds in Louisiana are classified as permanent ponds (LSU, 1999). The three primary types of permanent ponds are single-crop crawfish ponds, naturally vegetated ponds, and wooded ponds. The typical culture cycle for permanent ponds is as follows (LSU, 1999):

Time	Procedure
April–May	Stock 50 to 60 pounds of adult crawfish per acre (new ponds only)
May–June	Drain pond over a 2- to 4-week period

June–August	Plant crawfish forage or manage natural vegetation
October	Reflood pond
November–May/June	Harvest crawfish
May/June	Drain pond and repeat cycle without restocking crawfish

Single-crop crawfish ponds are managed solely for the purpose of cultivating crawfish. Crawfish can be harvested 1 or 2 months longer because there is no overlap with planting, draining, or harvesting schedules for other crops. Naturally vegetated ponds usually refer to marsh impoundments and agricultural lands that are managed to encourage the growth of naturally occurring vegetation as a forage base for crawfish. High amounts of organic matter in the soil often lower the water quality, which decreases production. Though marsh ponds exist in Louisiana, they are not usually recommended for commercial production because of inconsistent yields. The last type of permanent pond is a wooded pond. Wooded ponds are built on heavy clay soils in forested areas (cypress-tupelo swamps) near drainage canals. Leaf litter provides the bulk of forage, but water quality is difficult to manage. While wooded ponds may provide advantages such as potential for waterfowl hunting, low initial start-up costs, and selective removal of unwanted vegetation, overall production per acre is usually lower than that for other management regimes (LSU, 1999).

Rotational Ponds

The most common crawfish-agronomic crop rotations are rice-crawfish-rice; rice-crawfish-soybeans; rice-crawfish-fallow; and field rotation. In rice-crawfish-rice rotations, rice and crawfish are double-cropped annually. A rice farmer can use the same land, equipment, pumps, and farm labor that are already in place. Farmers plant rice in a drained field (a shallow pond with a depth of roughly 18 inches) and then flood the field 6 to 8 weeks later. After the field has been flooded, crawfish are stocked to grow and reproduce. When the fields are drained in August to harvest the rice, crawfish burrow underground. Crawfish burrow when water temperatures become too warm and when oxygen levels are low. They can survive as long as their gills stay moist. After the grain is harvested, the remaining stubble is fertilized, flooded, and allowed to regrow (ratoon) (LSU, 1999). The ratoon crop is used as a forage base for crawfish. Crawfish are harvested between November and April; however, the harvest season is shortened in rotational ponds because ponds are usually drained in March or April to prepare fields to replant rice in the spring. Crawfish are harvested using baited traps. Harvesting crawfish is labor-intensive and accounts for nearly two-thirds of the production costs (LSU, 1999).

The following is a typical rotation schedule for rice-crawfish-rice rotations (LSU, 1999):

Time	Procedure
March–April	Plant rice
June	At permanent flood (rice 8 to 10 inches high), stock 50 to 60 pounds of adult crawfish per acre
August	Drain pond and harvest rice (later in northern Louisiana)
October	Reflood rice fields
November–April	Harvest crawfish
March–April	Drain pond and replant rice

In rice-crawfish-soybeans rotations, three crops are produced in 2 years. This rotation has the advantage of allowing for a longer crawfish harvest season than the rice-crawfish-rice rotation. The rice-crawfish-fallow rotation allows the farmer to leave the land fallow for a certain period of time to break the natural cycle of certain weeds and prevent overpopulation of crawfish. This is a common practice in southwest Louisiana. After several years in production, rotational ponds may develop stunted crawfish as a result of overpopulation in the pond. Some farmers relocate crawfish in stunted ponds by moving mature crawfish from the affected pond to stock a new pond that will be used in a crawfish-agronomic rotation. The affected pond is left dry during the part of the cycle during which crawfish would be harvested (LSU, 1999). When crawfish are produced with other crops through the rotational crop system, producers use the same amount of water they would need if they were raising only crawfish.

Health Management

Crawfish are sensitive to most chemicals. Four herbicides are approved for use in rice or soybean fields intended for use as crawfish ponds: Stam, Basagran, 2,4-D and Rodeo (LSU, 1999). The use of herbicides to control weeds is a common management tool for rice and soybean crop production. Farmers use broad-spectrum herbicides like 2,4-D as a pre-emergent treatment prior to planting rice to kill any native vegetation (weeds). Narrow spectrum herbicides like Rodeo are used to spot-treat post-emergent weeds. The mixture of herbicides, both broad-spectrum and narrow-spectrum, used to support rice and soybean growth is independent from crawfish production.

Of all insecticides available, only Malathion and Bt are labeled for use in crawfish ponds. Malathion is commonly used to control mosquitoes. There are no plant fungicides labeled for use in crawfish ponds or in fields intended for use as crawfish ponds. The frequency with which herbicides are used is unknown. Considering the potential to eliminate the crawfish crop plus the added expense of the chemicals, it is not likely that herbicides are used often; therefore, the impact on water quality would be negligible. If herbicides are used, farmers use them in association with their agricultural crops and use them sparingly to avoid building up chemical toxicities that could adversely affect crawfish.

Primary disease pathogens of crawfish include bacteria, fungi, protozoans, and parasitic worms; however, disease problems associated with current crawfish culture practices have been minor (LSU, 1999). In estimating variable costs of crawfish production for a 40-acre pond in southwestern Louisiana, herbicides are listed as a potential expense, but drugs to treat diseases are not included in the report (de la Bretonne and Romaire, 1990a). Using drugs to treat crawfish ponds for disease is not likely to be a common practice; however, if a disease outbreak does occur, this might result in a reduced crawfish crop for the season.

4.3.10.2 Effluent Characteristics

In a study conducted by the Southern Regional Aquaculture Center (Tucker, 1998) to characterize the quality of effluents from commercial crawfish ponds, samples were collected from 17 commercial ponds in south-central and southwest Louisiana. Three types of culture systems were selected: crawfish-rice field (rotational), single-crop crawfish (permanent), and wooded (permanent). Rice-field ponds included rice-crawfish double-cropping systems. Permanent crawfish ponds selected either were planted with rice or sorghum-sudan grass in early to late summer or were not planted with cultivated forages and had native aquatic and terrestrial plants.

DO concentrations in crawfish pond effluents ranged from 0.4 to 12.6 milligrams/liter. The concentration in effluent in fall (mean = 6.5 milligrams/liter) was higher than the concentration in winter (mean = 4.7 milligrams/liter), spring (mean = 4.9 milligrams/liter), and summer (mean = 4.3 milligrams/liter). Ponds with native vegetation had the lowest concentration of DO in effluents (mean = less than 3.5 milligrams/liter) because relatively high quantities of vegetative biomass depleted oxygen in the ponds.

Total solids concentration in the spring and summer ranged from 143 to 2,431 milligrams/liter (mean = 522 milligrams/liter), and total volatile solids ranged from 0 to 432 milligrams/liter (mean = 96 milligrams/liter). Effluents from ponds with native vegetation had significantly lower concentrations of total solids and total volatile solids in spring and summer (mean = 286 and 69 milligrams/liter, respectively) than in rice ponds (mean = 646 and 113 milligrams/liter) and sorgham-sudan grass ponds (mean = 578 and 92 milligrams/liter). Soluble reactive phosphorus concentrations ranged from 0.002 to 0.653 milligrams/liter (mean + 0.116 milligrams/liter), and total phosphorus concentrations ranged from 0.039 to 1.126 milligrams/liter (mean = 0.329 milligrams/liter).

Results from the study showed that concentrations of nutrients and solids in effluents in crawfish ponds were generally higher in the spring and summer. Effluent quality was poorest during the summer drainage period. The type and quantity of summer vegetation had a significant influence on the quality of water discharged from crawfish ponds. Ponds with native vegetation generally had lower concentrations of nutrients and solids than ponds with rice or sorghum-sudan grass. The presence of aquatic macrophytes in spring and summer in ponds with native vegetation increased nutrient uptake and reduced the level of suspended sediments. This study suggests that ponds with native vegetation are more likely to have better water quality.

4.3.10.3 Current Effluent Treatment Practices Within the Industry

As in other pond systems, the most important water quality concern in crawfish ponds is the level of DO. DO should be maintained above 3 milligrams/liter for optimal crawfish production (LSU, 1999). Problems with DO in crawfish AAP are compounded by the presence of large amounts of decomposing vegetation, which make typical remedies like emergency aerators ineffective (Eversole and McClain, 2000). Instead, crawfish farmers rely on preventive management measures such as the choice of forage type, the timing of flooding dates, the close monitoring of water quality conditions, and pond designs that divert flow to all areas of the pond. To improve levels of DO, some crawfish farmers use paddlewheel aerators coupled with diversion levees in the pond to improve circulation and maintain adequate DO levels (Eversole and McClain, 2000). Whereas feed management and the impacts of adding pelleted feed to the system are usually important water quality considerations for the culture of other species, feeding is not a regular practice in crawfish culture (Eversole and McClain, 2000). Instead, current production practices rely on a forage-based system. There are no feed management practices to recommend for this subcategory because the feed input is low and additional feed management practices would not likely have a significant impact.

Because farmers rely on soils to grow multiple crops like rice and soybeans in addition to crawfish, farmers using rotational crop systems in Louisiana, the region that accounts for 90% of the crawfish production in the United States, drain ponds slowly to prevent loss of soil. Ponds are also drained slowly to encourage crawfish to burrow into the pond bottom to start their reproductive cycle. There are some examples of crawfish farmers discharging water from crawfish ponds into siltation ditches and ponds prior to discharging the effluent into receiving surface waters like streams and rivers (Tetra Tech, 2002d). There is also cooperation with the NRCS to implement BMPs to minimize erosion and reduce the amount of nutrients and pesticides in effluent discharges (LSU, 1999). Examples of these practices include channel vegetation to improve turbidity problems, filter strips to reduce sediment in inflow and discharge water and help reduce soil erosion, and irrigation water management with planned flooding and draining to manage forage and crawfish.

BMP guidelines from NRCS also describe the positive environmental impacts of well-managed crawfish ponds (LSU, 1999). In many cases, flooded crawfish ponds benefit and improve the quality of the water entering and exiting fields by developing or restoring wetlands. Crawfish ponds provide more than 115,000 acres of man-made wildlife wetland habitat, benefiting waterfowl, wading birds, shorebirds, furbearers, reptiles, amphibians, and other invertebrate animals.

Although there is limited information about the quality of water discharged from either rotational ponds or permanent ponds, the impact of the volume of water discharged and the quality of the water discharged is likely to be minimal. First, crawfish production relies on the forage-based system for feeding, so feed management practices would not significantly impact water quality because the feed input is so low. Also, although DO levels are a concern, particularly as vegetation decays, crawfish farmers routinely check levels and use BMPs and technologies like mechanical aeration to maintain appropriate DO levels. Crawfish farmers also use siltation ditches to minimize the impact of discharge from crawfish ponds. Finally, when water is discharged from ponds, farmers release the water slowly to prevent the loss of valuable topsoil needed for productive agricultural crops and to encourage crawfish to burrow.

4.3.11 Lobster

The impoundment or pounding of the American lobster (*Homarus americanus*) in tidal lobster pounds is an important part of the lobster industry in Maine. Pounds are man-made tidal pools or impounded coves (Loughlin et al., 2000). They are flushed daily at high tide, replacing the holding area with fresh seawater. Pounds help lobster fishers and pound operators control the supply of lobsters to meet the market demand in the off-season when fishers are not harvesting wild catches. Although pounding is an important practice in Canada, according to the Maine Lobster Pound Association, Maine is the only state in the United States using this cultivation practice (Hodgkins, 2002, personal communication). There are 65 lobster pounds in Maine owned by 50 operators (Hodgkins, 2002, personal communication; Tetra Tech, 2002e).

In 2000, 57 million pounds of American lobster with a commercial value of more than $187 million were landed in Maine (Maine, 2002). Most wild-caught lobster harvests are shipped immediately to market, but some are held in pounds to extend their growth cycle. Tidal pounds in Maine hold about 5 million pounds, or approximately 10% of the total lobster landed in the state (Hodgkins, 2002, personal communication). In the colonial period, lobsters were considered poverty food, served daily to children, prisoners, and indentured servants (Gulf of Maine Aquarium, 2000). In today's market, the increased demand for lobster and the decline in wild lobster harvests has transformed lobster into a high-priced commodity, thereby encouraging the development of pounding.

4.3.11.1 Production Systems

For fall pounding, lobster fishers sell their catches of newly shed lobsters from September through November to pound keepers, who hold the shellfish in pounds (AII, 1989). Without aeration, lobsters are typically stocked 1 pound per square foot of bottom area. The average size of a lobster pound is 70,000 ft^2 (Hodgkins, 2002, personal communication; Tetra Tech, 2002e). From early September through April, the lobsters fill in their new larger shells with meat while the pound operators wait for a favorable market price. There are also shorter spring and summer pounding seasons with fewer lobsters. Spring pounding starts in May when the Canadian season opens, and spring-pounded lobsters are sold before they molt in July and August. From July to August soft shell lobsters are placed in pounds, where they harden and are sold. Summer pounding caters to the airfreight market (Tetra Tech, 2002e).

Lobsters are harvested using one of three methods: pumpers, dragging, or divers (Loughlin et al., 2000). Because of their speed and efficiency, airlift or hydraulic pumpers are considered the most cost-effective means of harvesting lobsters from a pound. With diver-operated pumpers, a diver works on the bottom, collecting lobsters and placing them into the end of the suction tube. Water flowing through the tube carries the lobsters to the surface. Dragging or seining is another common harvest method; however, lobsters are sometimes crushed or damaged when the work crew hauls the drag over the edge of the platform. Divers are also used to remove lobsters from pounds. They use a mesh bag to collect the lobsters. The bag is attached to a line that extends to the workstation. When the bag is full, the diver signals the crew to haul up the bag. Some pound owners drag their pounds until they recover about 80% of their lobsters; then they use divers to collect another 15%. The remaining lobsters are harvested when the pound is drained (Loughlin et al., 2000).

4.3.11.2 Culture Practices

Feed Management

Pound operators feed lobsters while they are in the pound. Most lobster pound facilities feed lobster freshly killed fish such as sculpin, pickled and smoked herring, and menhaden (Hodgkins, 2002, personal communication). Fresh or salted fish racks can also be used as a food source for lobsters. Operators generally use manufactured feed only when they need to apply medicated feed. The average feeding rate for Maine lobster pounds is approximately 70 pounds of fish per day per 5,000 lobsters (Hodgkins, 2002, personal communication; Tetra Tech, 2002e). Winter is the primary pounding season in

Maine. On average, lobsters are fed for 40 days within the winter pounding season. Feeding rates drop off when water temperatures drop below 40 °F. When water temperatures approach 32 °F, lobsters begin hibernating and do not consume food during this period. The summer and spring pounding seasons are shorter (1 to 2 months), with fewer lobsters and very few feeding days (Hodgkins, 2002, personal communication; Tetra Tech, 2002e).

Health Management

The three main diseases that affect lobsters are red tail, vibrio, and ciliated protozoan disease. Red tail (caused by *Gaffkemia*) is a fatal, infectious bacterial disease of lobsters that passes from one lobster to another through a break in the tail (Loughlin et al., 2000). Symptoms of red tail include inactive, weak, and lethargic lobsters; red tint under the tail; and a tendency in lobsters to remain near the shore (Loughlin et al., 2000). Red tail disease is present in an average of 5% to 7% of wild lobsters (Lobster Institute, 1995). If infected lobsters are placed in a pound and die, the live bacteria cells spread to other lobsters (Gulf of Maine Aquarium, 2000). Gram negative rod bacteria, such as *Vibrio*, are hard to detect and difficult to treat. To stop the spread of the bacteria to healthy lobsters, pounds prevent overfeeding and remove weak lobsters (Loughlin et al., 2000). Ciliated protozoan disease is fatal to lobsters, with mortality usually occurring in 1 to 2 months (Loughlin et al., 2000). As in red tail, the protozoan enters the lobster through a break or wound in the tail. The disease has no approved treatment and has shown up in more than a dozen pounds over the past 10 years (Loughlin et al., 2000).

Pound operators conduct an initial health screening of the lobsters before they are stocked into the pound to remove weak and sick animals. This practice reduces the frequency of disease in the pound. Pound operators also conduct periodic inspections using divers or a small hand drag to sample the pond and to screen out sick and dead lobsters (Loughlin et al., 2000).

When needed, pound keepers use medicated feed containing oxytetracycline (brand name Terramycin) to treat bacterial diseases like red tail. The frequency of use varies from facility to facility. On average, about half of the pound facilities use oxytetracycline in a pound season. Treatments with medicated feeds usually last 5 days before pound keepers switch back to regular feed, and pound keepers commonly use the drug for two cycles, or 10 days, in a pound season. Oxytetracycline is administered through medicated feed at approximately 6 to 8 pounds of feed per 1,000 pounds of lobster. As temperatures drop, feeding rates also decline to 3 to 5 pounds of feed per 1,000 pounds of lobster. Assuming an average facility holds 70,000 pounds of lobster, a facility would use roughly 3,850 pounds of medicated feed in a year. (For the entire industry, this would be approximately 127,050 pounds of medicated feed per year.)

The FDA regulates the use of medicated feed and requires lobster growers to apply a 30-day withdrawal period. Facilities must wait at least 30 days after feeding lobsters medicated feed before they remove lobsters from the pound to ensure that residues from the medication are flushed from the lobster before human consumption. Currently, oxytetracycline is the only FDA-approved medication for lobsters (Bayer, 2002, personal communication). Generally, this is the only drug used by lobster pound facilities.

4.3.11.3 Water Quality Management Practices

Mechanical aeration enhances DO levels in lobster pounds. Approximately two-thirds of lobster pound facilities in Maine use mechanical aeration, especially in months with warmwater temperatures (Hodgkins, 2002, personal communication). Dams for the impoundment are built to the height of the mean low water mark with a notch at the mean low water mark. As incoming water flows through this notch at high tide, the increase in water velocity promotes water mixing inside the impoundment (Tetra Tech, 2002e). Pounds rely on tidal flushing to maintain the water quality in the impoundment. Currently, there are no existing control technologies in the industry to reduce discharge.

Although there is little information about the quality of water discharged from lobster pounds, the impact of the effluent is likely to be minimal. Currently, lobster pounds are found only in Maine, and they are not likely to expand to other states. This is a small industry subcategory that is site-specific to Maine. Based on a relatively low input of food and a limited number of feeding days, feed management BMPs are not likely to improve water quality in the system. Regular tidal flushing for all pounds and supplemental aeration for many pounds in Maine also help maintain water quality and DO levels. Finally, the industry is regulated by the FDA 30-day withdrawal requirement limiting the number of days that pound keepers can use medicated feed, so the impact from inputs of medicated feed into the system is likely to be minimal and is already regulated by another agency.

4.3.12 Molluscan Shellfish

Molluscan shellfish AAP systems are used to raise oysters, clams, mussels, and scallops. These animals are bivalves; that is, they have a soft body enclosed by two hard shells or valves. The valves are attached at a hinge and are held shut by a strong muscle. Most cultured molluscan shellfish are filter feeders that rely on phytoplankton and particulate detritus delivered by water currents as their food source (JSA, 2000c).

Oyster farming is practiced on the Atlantic, Gulf of Mexico, and Pacific coasts of the United States. In the United States, two species currently dominate the oyster culture industry: the Pacific or Japanese oyster (*Crassostrea gigas*) and the American oyster (*Crassostrea virginicia*). Oysters usually inhabit areas from low intertidal zone to approximately 45 feet deep, forming a reef-like mass on firm bottom. Depending on the geographic location, oysters take from 18 to 48 months to reach market size (JSA, 2000c).

Clam farming is widespread throughout the United States, particularly on the east coast. Two species dominate commercial production. The hard clam (*Mercenaria mercenaria*), also known as the quahog, hard-shelled clam, cherrystone clam, or little neck clam, is indigenous to the Atlantic and Gulf of Mexico coasts, with smaller populations present on the west coast. The hard clam prefers relatively protected areas that have stable sandy to muddy bottoms with small amounts of shell. Populations exist from the low intertidal zone to nearly 60 feet in depth. The second species of clam most often cultured in the United States is the Manila clam (*Tapes philippinarum*) on the Pacific coast. Manila clams are typically found in habitats similar to those of the hard clam, but they generally exist slightly higher in the intertidal zone in areas with a coarser substrate like gravel. As with the hard clam, Manila clams have short siphons (necks), and this limits the depth to which they can burrow. Like oysters, clams typically take from 18 to 48 months to reach market size. Two additional species may be produced commercially in the near future: the geoduck (*Panope abrupta*) on the west coast and the surf clam (*Spisula solidissima*) on the east coast.

Mussel farming is a relatively new sector in the United States. Three principal mussel species are cultivated: *Mytilus edulis* on the east coast and *M. galloprovincialis* and *M. trossulus* on the west coast. Mussels usually form dense aggregations, like reefs, from the low intertidal zone to 30 feet deep. These aggregations may be on hard substrate or stabilized muds or sands. Both species typically reach commercial size in 19 to 24 months.

Scallop farming, like mussel farming, is also a relatively new sector in the United States. Scallop culture is limited, and most commercial efforts have been confined to the bay scallop (*Argopecten irradians*). This species lives in shallow bays from Massachusetts through Florida and is often associated with beds of eelgrass (JSA, 2000c). Cultured scallops reach commercial size in 10 to 24 months There is also a growing interest in the northeastern United States in the sea scallop (*Placopecten magellanicus*), but currently these efforts are experimental. In Washington there is a project exploring the possibility of culturing the rock scallop (*Hinnites giganteus*).

Harvest data related specifically to the molluscan shellfish industry are very limited and inconsistent (Kraeuter et al., 2000). Shellfish production as reported by most states is not divided based on whether the shellfish are cultured or from a wild harvest fishery, and there is no consistency among states regarding reporting units. For example, some states report oysters by live weight and some in shucked meat weight. Based on data from the 1998 Census of Aquaculture (USDA, 2000), there are 535 molluscan shellfish farms in the United States—268 in the Southern Region, 150 in the Northeastern Region, 108 in the Western Region, 5 in the Tropical/Subtropical Region, and 4 in the North Central Region. Though it has fewer facilities, the Northeastern Region leads the country in revenue with approximately $26.7 million in total sales, followed by the Southern Region with $24.7 million in total sales.

4.3.12.1 Production Systems

Shellfish AAP activities vary widely throughout the United States. Different species are cultured in different regions and use a variety of culture systems. Determining what is actually AAP is a challenge (Kraeuter et al., 2000). On one end of the spectrum are managed wild fisheries, which rely on natural recruitment to reseed public beds. At the other end of the spectrum is intensive culture on privately owned tidelands. Beds are seeded with juveniles that began as larvae in a hatchery, raised in an upland nursery on cultured algae, transferred to a land-based nursery that relies on natural algae present in the water, and finally planted in some sort of growout system. Between these two ends of the spectrum are a range of other options with varying levels of control over the product being cultured.

Intertidal culture, or shallow-depth culture (less than 3 feet), is the most common bottom culture in the United States. Intertidal techniques vary and are dependent on the species being cultured. Clams, oysters, and mussels may be placed directly on the bottom in beds. Clams dig in, whereas oysters and mussels remain on the bottom surface. In clam culture, mesh is usually placed over the clams or they are placed in mesh bags to prevent predators from consuming the crop. Oysters and mussels are usually planted without protective devices; in Washington's Puget Sound, however, farmers sometimes use plastic mesh bags, which are attached to the bottom on a longline. Intertidal plantings of oysters and mussels can also be suspended above the bottom on racks, trays, longlines, or bags strung on lines or wrapped on pilings. These techniques usually suspend the crop 1 or 2 feet off the bottom and rely on tidal action to feed the animals and remove wastes.

Subtidal water column culture is used where tidal amplitudes are not sufficient to support intertidal beds or where the organisms do not require sediment. Scallops, mussels, and oysters are cultured in subtidal water column systems. Water column culture in deeper waters, or floating culture, uses either rafts or longlines attached to floats, or a tray or rack system. Tray systems require specialized diving or lifting gear for maintenance in deeper waters. Subtidal water column culture is less common in the United States because these systems require floats or rafts on the surface that create conflicts with competing recreation or commercial uses of the water surface or column, as well as concerns from upland owners regarding visual impact.

4.3.12.2 Culture Practices

The intensive culture of molluscan shellfish has five phases: food production, broodstock maintenance/conditioning, hatchery, nursery, and growout.

Bivalve hatcheries are used to condition (i.e., prepare for spawning) broodstock, spawn animals, and raise larvae. Food for conditioning broodstock, larval, and post-set bivalves consists of various forms of unicellular algae that are grown and added to the water for the bivalve to filter (Kraeuter et al., 2000). The production of algae is one of the most time-consuming and expensive parts of bivalve culture. There are two methods for producing phytoplankton for use as food for molluscan shellfish. The Wells-Glancey method involves filtering raw seawater to remove large diatoms and algae consumers, such as copepods, and enriching the filtrate to promote the growth of small diatoms and flagellates. This method is inexpensive, but it provides little control over the species cultured. The Milford method uses a single species of phytoplankton in bacteria-free or clean, but not contaminant-free, cultures. This method provides more control over algal growth, but the need to maintain cultures and sterile conditions increases the expense.

Broodstock are used to produce the gametes for the next generation. Most broodstock are maintained in field sites until they are to be conditioned, the process of gonadal maturation for spawning. The animals are brought into a hatchery where water temperatures can be controlled to manipulate spawning. Animals to be spawned are placed in tanks and slowly warmed, and then cultured algal food is added to the water. Tanks range in size from 150 to 500 gallons. This process is a batch culture, and water is typically exchanged every 2 days (Kraeuter et al., 2000). The conditioning phase takes approximately 6 to 8 weeks. A small hatchery may condition 50 to 100 animals, and a larger hatchery may condition up to 2,000 animals. Only algal food is added to the water during this phase.

With strip spawning, eggs or sperm are removed from the animal, and the eggs are fertilized and placed in a tank of filtered seawater. Mass or individual spawning is achieved by placing the animals in a seawater bath. In most instances, volumes of water used are small (usually less than 100 gallons) because hatcheries minimize the amount of water for which they need to control the water temperature. Once spawning begins, the eggs are retained in a dish, or a container for single spawning individuals. In mass spawning, fertilization takes place in a tray with animals. After the eggs hatch the larvae are fed algae beginning on the second day. Water is exchanged every 1 or 2 days. Some hatcheries use flow-through systems with screens to prevent the escape of larvae. The number of days in larval culture varies, but typically ranges from 14 to 20 days.

Setting is the process by which a bivalve grows a shell and changes from a planktonic, pelagic animal to a benthic animal. Though procedures vary from species to species, usually the animals are set and maintained with food inputs of cultured algae. Oysters may be set at the hatchery or moved to a remote site, where they are added to tanks that have been filled with bags of shell and filtered seawater and some unicellular algal food. The tanks are aerated. Setting can take 1 to 3 days, and individuals may remain in the tank for 1 to 3 weeks before they are placed in a field nursery. Clams, scallops, and mussels are all set by attaching to a substrate by their byssal threads. These animals are removed from the larval culture tanks and placed in downwellers (cylinders with a mesh bottom through which water is passed by pumping it in through the top), in bags of setting material, or in trays. Many of these methods continue to feed with unicellular algae for 1 to 2 weeks and then transition to a nursery culture.

Nurseries hold animals until they are ready to be planted in the substrate. The longer the larvae, or seed, can be raised in protected nursery systems, the higher the survival rate will be when they are planted in the final growout phase. As with the hatchery phase, the number of animals being cultured in a nursery is large, but the biomass is very small when compared to fish or crustacean culture (Kraeuter et al., 2000). Nurseries use two different culture methods: induced circulation and natural circulation. Induced circulation uses pumps, paddlewheels, or airlifts to move large volumes of water to create a flow so the bivalves can filter feed. For natural circulation, animals are placed in bags suspended in the water or on trays on the bottom, and natural circulation moves water over the animals to bring them food and remove waste. Animals are usually kept in a nursery until they are large enough to be planted. For most bivalve nurseries, the individuals increase in size from 1 to 10–20 millimeters (Kraeuter et al., 2000). The only significant addition to the production water in this phase is the freshwater used to wash the seed and flush the trays, upwellers, raceways, or sieves. Some nursery facilities also add cultured algae to the system, but costs limit this practice.

The growout phase is the last phase in bivalve culture. Some producers buy seed and focus only on growout. All growout techniques for bottom culture rely on naturally occurring food sources at the site. There are no feed management practices because there is no feed input.

Feed Management

Bivalve (molluscan) AAP is substantially different from other forms of AAP in that no food is added to the culture water during the growout phase (Kraeuter et al., 2000). Shellfish are grown out in the open, protected coastal waters. They feed by filtering large volumes of seawater through their gills and extracting natural phytoplankton present in estuaries. Depending on the species, size, water temperature, and other variables, volumes of water filtered can range from 20 to 80 gallons/day, per animal (Kraeuter et al., 2000). This demand at the growout stage for high volumes of water and physical space generally requires that molluscan shellfish are produced in the natural environment.

Although hatcheries and some nurseries add cultured algae to the water as a food source, the impact from this addition is not significant. The risk of nonindigenous microalgae grown for shellfish feed disrupting natural phytoplankton ecology is very low (Wikfors, 1999, personal communication). Cultured algae strains have been sheltered in artificial culture conditions. If they were to escape, they would most likely have lost most of their ability to compete with indigenous phytoplankton. Furthermore, there have been no examples of nonindigenous algal strains from shellfish hatcheries creating a bloom or even a low-level introduction in receiving waters.

Health Management

Drug and pesticide use in molluscan shellfish culture is very limited. The common industry practice is to maintain bacteria at low levels in the early stages of culture by sterilizing the water (Kraeuter et al., 2000). It is not economically feasible to use drugs to control disease in bivalves. If hatcheries use chlorine to clean tanks or sterilize seawater, these facilities are required to dechlorinate prior to discharge (Tetra Tech, 2001). Abalone culture is the only culture activity that uses spawning aids like hydrogen peroxide and L-Dopa to enhance settlement (Tetra Tech, 2001).

4.3.12.3 Water Quality Management Practices

The importance of bivalve filtration, or lack of filtration, in natural systems has been used as an argument for restoring the abundance of oysters in the Chesapeake Bay and the New York harbor through either AAP or natural reef restoration (Revkin, 1999; Zimmerman, 1998). Restoring this filter feeding population would increase DO and water clarity and remove nitrogen and phosphorus from the system through direct harvest (Newell and Ott, 1999). The ecological consequence of this current lack of filter feeders is significant. Rice et al. (1999) have estimated that the northern quahog (hard clam) could remove up to 167,000 pounds of nitrogen from the water column and that sustainable harvest of the population would completely remove 17,000 pounds of organic nitrogen annually. Another study found that an intensive mussel culture raft system increased the rate of energy flow, as well as nitrogen and phosphorus deposition and regeneration; but unlike fish farming, the mussel culture did not cause eutrophication by nutrient input (Rodhouse and Roden, 1987). Still another study proposes the use of mussels as a means to clean up eutrophied fjord systems in Sweden (Haamer, 1996).

Fertilizers used in hatcheries are not likely to affect receiving waters. The fertilizer mix used in shellfish hatcheries is designed to be deficient in nitrogen, the nutrient of most concern in coastal eutrophication (Wikfors, 1999, personal communication). Nitrogen is the limiting factor for phytoplankton growth. The standard hatchery operation involves growing algae to a density at which all nitrogen is assimilated by the microalgae and the algae stop growing.

The growout phase of molluscan shellfish production does not add food to the system. The bivalves rely on natural food found in coastal waters. In terms of a mass balance, materials are extracted from the estuary as they are converted into bivalve flesh and shell, or used for respiration (Kraeuter et al., 2000). Because there is no feed input, there are no feed management practices. Bivalve culture can actually result in the net removal of nitrogen, phosphorus, and other pollutants when crops are harvested and removed from the system (Kraeuter et al., 2000). Because bivalves filter nutrients out of the water, they do not pose a threat to water quality. EPA believes there is little, if any, impact on water quality; therefore, no current technologies or BMPs are being used by this industry subcategory.

4.3.13 Other Aquatic Animal Production (Alligators)

American alligators (*Alligator mississippiensis*) are raised in captivity primarily for their hides and meat. The leather is used to make luxury apparel items such as belts, wallets, purses, briefcases, and shoes. In the past the high value of these leather products led to extensive hunting of alligators in the wild. By the 1960s this exploitation, plus loss of habitat, had depleted many wild populations (Masser, 2000). Research into the life history, reproduction, nutrition, and environmental requirements of the American alligator, along with the rapid recovery of wild populations, led to the establishment of commercial farms in the United States in the 1980s. In 1996 wild harvest and farm-raised alligators from the United States supplied more than 240,000 hides to world markets. Approximately 83% of these hides were from alligator farms (Masser, 2000). States with licensed alligator farms are Alabama, Florida, Georgia, Idaho, Louisiana, Mississippi, and Texas.

The American alligator was once native to the coastal plain and lowland river bottoms from North Carolina to Mexico (Masser, 2000). The only other species of alligator (*A. sinensis*) is found in China and is endangered. Hunting alligators for their hides began in the 19th century. At the turn of the 20th century, the annual alligator harvest in the United States was around 150,000 animals. Overharvesting and habitat destruction depleted the wild population, and by the 1960s, most states had stopped allowing alligator hunting. To protect alligators from further exploitation, they were designated under the Endangered Species Act as endangered or threatened throughout most of their range, with the exception of Louisiana. Alligator populations recovered quickly, particularly in Louisiana, which had stopped legal harvesting in 1962 (Masser, 2000). Louisiana reopened limited harvesting of wild alligators in 1972, but the population continued to increase even with sustained harvesting. Most other southern states also experienced population increases after federal protection.

In 1983, under the CITES, the USFWS changed the designation for the American alligator to “threatened for reasons of similarity in appearance” (Masser, 2000). This classification means that the alligator is not threatened or endangered in its native range; however, the sale of its products must be strictly regulated so that the products of other crocodilian species that are endangered are not sold illegally as those of American alligators. Today, in addition to alligator farming, nuisance control is allowed in several southern states, and limited harvests from the wild are permitted in Louisiana, Texas, and Florida (Masser, 2000).

Alligators inhabit all types of fresh to slightly brackish aquatic habitats. Males grow larger than females, and growth and sexual maturity are dependent on climate and the availability of food (Masser, 2000). Along the Gulf coast, females usually reach sexual maturity at a length of 6.5 feet and an age of 9 to 10 years. As in other cold-blooded animals, maturation age is affected by temperature. Optimum growth occurs at temperatures between 85 and 91 °F (Masser, 2000). No apparent growth takes place below 70 °F, and temperatures above 93 °F cause stress and sometimes death.

In the wild, young alligators usually consume invertebrates such as crawfish and insects. As they grow, fish become a part of their diet. Adults consume mammals such as muskrats and nutria. Large adult alligators even consume birds and other reptiles, including smaller alligators (Masser, 2000). Females do not move or migrate over long distances once they have reached breeding age, and they prefer heavily vegetated marsh habitat. Males move extensively but prefer to establish territories in areas of open water.

4.3.13.1 Production Systems

Alligator farming uses a unique production system that is not easily categorized as either a pond system or a flow-through system. Alligator systems use more water than typical pond systems and less water than typical flow-through systems. Available literature suggests that pond-like systems, in the form of outdoor ponds and lagoons, are most often used for raising and maintaining breeding alligators for a source of eggs. Young alligators are typically raised for growout in indoor pens with shallow pools that use concrete tanks to hold the animals. Within the concrete tanks, water is usually pumped from a well and then heated before it is pumped into the pools of each pen. At some facilities, water in the indoor pools is completely drained and replaced daily or every other day to maintain good water quality (Coulson et al., 1995). Some facilities drain less frequently. Maintaining water temperature and minimizing heating costs are often major concerns of alligator farmers. Based on daily drainings, the production system could be described as a batch-like flow-through system with a daily exchange of water. When facilities drain less often, the system could be described as a pond with frequent drainings.

In an effort to reduce costs, some producers are using outside growout facilities (Masser, 2000). In this system, alligators are raised in indoor facilities for the first year of growth and then moved to outdoor fenced ponds. The alligators are fed a commercial diet during warm weather and are allowed to hibernate during cooler seasons. After about 2 years, the ponds are drained, usually during the winter, to facilitate handling and harvesting of the animals.

4.3.13.2 Culture Practices

The commercial production of alligators can be divided into three phases: management of adult alligators for breeding; egg collection, incubation, and hatching; and growout of juvenile alligators to market size. Alligator farmers must either purchase eggs or hatchlings from other producers or produce their own eggs. In Louisiana, Florida, and Texas, eggs and/or hatchlings can be taken from the wild under special permit regulations. Today, the primary source for eggs is wild populations; however, Louisiana law does not allow the sale of alligator eggs outside Louisiana.

Some farmers have completely integrated operations with their own breeding stocks, hatching facilities, nursery facilities, and growout houses, but most alligator farmers focus on only growout operations (Jensen, 2000, personal communication). This approach is also called ranching, an open-cycle system that does not maintain adult breeders or produce its own stock, similar to a cattle feedlot operation (Lane and King, 1996). With growout operations, hatchlings are purchased from a farm or ranch specializing in the production of hatchlings, usually from eggs collected from wild stocks. Most of the eggs used to produce hatchlings are collected on private lands, which provide a source of income to marsh landowners who, in turn, maintain and manage wetland habitat for the benefit of the alligator population. Egg collection from wild populations is regulated by state agencies that set site-specific quotas for the number of nests that may be harvested (Heykoop and Freschette, 1999). Hatchlings may also be available from state agencies that regulate wild populations. The wild population is a source of young stock for domestic populations, and in Louisiana, where a percentage of hatchlings is returned to the wild, the domestic population is a source of juveniles for the wild populations (Heykoop and Freschette, 1999).

The first phase, maintaining adult alligators and achieving successful and consistent reproduction, is extremely difficult and expensive (Masser, 1993a). Adult alligators that have been raised entirely in captivity or confinement behave differently from wild stock. Farm-raised alligators accept confinement and crowding as adults better than wild alligators. Also, adult alligators raised together tend to develop a social structure, adapt quicker, and breed more successfully than animals without an established social structure.

For the few farms that maintain breeding stocks or specialize in producing eggs, pens for adult alligators are built approximately 1 to 2 acres in size (Masser, 1993a). Pens must be carefully fenced to prevent the escape of the adult alligators. Breeding pen design, particularly the water ratio and configuration, is very important. The land area-to-water area ratio in the pen is approximately 3:1, and the shape of the pond maximizes the shoreline area with an ‘S’ or ‘Z’ shape. The depth of the breeding pond is at least 6 feet. Breeding ponds have dense vegetation around the pond to provide cover, shade, and nesting material. Alligators burrow into the pond banks if adequate shade is not provided. Stocking densities for adult alligators are approximately 10 to 20 animals per acre. The female-to-male ratio should be approximately 3:1.

Adult breeders should be disturbed as little as possible from February through August, during egg maturation, courting, and nesting. Nesting success in captive alligators has been highly variable. Wild versus farm-raised origin, pen design, density, the development of social structure within the group, and diet all affect nesting (Masser, 2000). Nesting rates for adult females in the wild averages around 60% to 70% with the most favorable habitat and environmental conditions. Nesting rates in captivity are usually much lower (Masser, 2000). Clutch sizes vary with the age and condition of the female, with larger and older females usually laying more eggs. Clutch size averages around 35 to 40 eggs and egg fertility varies from 70% to 95%. Survival of the embryo also varies from 70% to 95% and the hatching rate from 50% to 90%. Land costs, long-term care of adults, and low egg production contribute significantly to the cost of maintaining breeding stocks (Masser, 2000).

The method and timing of egg collection are very important; alligator embryos are very sensitive to handling from 7 to 28 days after the eggs are laid (Masser, 2000). Eggs should be collected in the first week or after the fourth week of natural incubation. When eggs are collected, they must be kept in the same position and not turned or rotated during handling. Compared with wild nesting, artificial incubation improves hatching rates because of the elimination of predation and weather-related mortality (Masser, 2000). The best hatching rates for eggs left in the wild are less than 70%, while hatching rates for eggs taken from the wild and incubated artificially average 90% or higher (Masser, 2000).

Eggs should be transferred into incubation baskets and placed in an incubator within 3 or 4 hours after collection. Eggs are completely surrounded with nesting material like grasses and other vegetation. The natural decomposition of the nesting materials helps with the breakdown of the eggshell. The incubation temperature is critical for the survival and development of the hatchlings. Temperature also determines the sex of the alligator. Temperatures of 86 °F or below produce females, and temperatures of 91 °F or above produce males. Temperatures much above or below these ranges result in high mortalities (Masser, 2000). After the alligators hatch, the hatchling are kept in the incubation baskets for the first 24 hours and then moved to small tanks heated to 86 to 89 °F. Maintaining 89 °F helps hatchlings absorb the yolk. Usually, hatchlings will begin to feed within 3 days. Once hatchlings are actively feeding, they can be moved into growout facilities.

A variety of growout facilities are used for raising alligators. Growout buildings are usually heavily insulated concrete block, wood, or metal buildings with heated foundations. They usually do not have windows. Most animals are kept in near or total darkness except during feeding and cleaning times. The concrete slab is lined with hot water piping or, sometimes, electric heating coils. A constant temperature is maintained by pumping hot water through the pipes. Covering about two-thirds of each pen is a pool of water about 1 foot deep at the drain. The bottom of the pool is sloped down toward the drain to facilitate cleaning. The remaining one-third of the pen area is above water and is used as a feeding area and basking deck (Masser, 1993b). Pens vary in size. In general, smaller pens are used for smaller alligators and larger pens are used as alligators grow. Usually, farmers construct several sizes of growout pens and reduce the density by moving the animals as they grow. Common stocking densities include 1 square foot/animal until the animal reaches 2 feet in length; 3 square feet/animal until the animal reaches 4 feet in length; and 6 square feet/animal until the animal reaches 6 feet in length.

A common construction plan uses a 5,000-square foot building with an aisle down the middle and pens on either side. A 4-foot aisle creates pens that are approximately 14 feet wide. Pens are usually 13 feet long with a 3-foot concrete block separating individual pens from the aisle. Another popular building design is the single round house, a structure about 15 to 25 feet in diameter constructed as a single pen (Masser, 2000). Round houses have also been built from concrete blocks, or from a single section and roof of a prefabricated metal silo used for storing grain. The round concrete slab on which the house sits is sloped from the outer edge toward a drain in the center of the structure. The round house is filled with water so that approximately one-third of the floor is above the water level. Because they are single-pen units, round houses have the advantage of not disturbing alligators in other pens during feeding, cleaning, and handling operations.

The heating system, which consists of water heaters and pumps, is an important part of the growout facility. Warmwater is needed to heat the building, fill the pools, and clean the pens. Some heating systems have industrial-size water heaters, while other systems have flash-type heaters to heat water for cleaning and standard heaters to circulate warmwater through the slab. Thermostats regulate the temperature and circulation pumps. The temperature in growout pens must be between 86 and 88 °F for optimal growth (Masser, 2000).

Written approval and hide tags must be obtained from the appropriate state regulatory agency before any alligators may be harvested. Some states also require a minimum length of 6 feet at harvest. Alligators may be skinned only at approved sites. Skinning, scraping, and curing must be done carefully to protect the quality of the skin; hides that are cut, scratched, or stretched have a reduced value. Most hides are sold to brokers, who purchase and hold large numbers of hides and then sell them to tanneries for processing. A few larger farms sell directly to the tanneries, the best of which are in Asia and Europe.

Feed Management

In general, alligators in the wild consume a diet high in protein and low in fat. Early alligator producers manufactured their own feed using inexpensive sources of meat like nutria, beef cattle, horse, chicken, muskrat, fish, and beaver. Today, several feed mills are manufacturing pelleted alligator feed. Most farmers feed their animals only commercially available feed; however, some continue to feed the animals a combination of raw meats and commercial diets.

Feed is spread out on the deck in small piles to reduce competition. Typically, farmers feed alligators 5 times per week, although some may feed 6 or 7 days/week. The feeding rate is roughly 25% of the animal's body weight per week the first year; then the rate is gradually reduced to 18% of body weight as the animal approaches 3 years old or a length of 6 feet. Feed conversion efficiencies decrease as alligators grow larger. The food conversion ratio is between 2:1 and 3:1 (Masser, 2000). Monthly growth rates in alligators can be as high as 3 inches when the temperature is held at a constant 86 to 89 °F and they are fed a quality diet with minimal stress. Many producers grow hatchlings to 4 feet in 14 months, and some producers have grown alligators to 6 feet in 24 months (Masser, 2000).

Health Management

There is very little information available in the literature to characterize drug and pesticide use for alligator farming. No antibiotics are approved for use on alligators; therefore, any antibiotics needed must be obtained through a prescription from a veterinarian (Masser, 2000). Two antibiotics, oxytetracycline and virginiamycin, have been used by alligator producers and added to feed to fight bacterial infections (Masser, 1993b).

Alligators need clean water to maintain the quality of their skins. Poor water management can lead to brown spot disease, which scars the skin and reduces its value (Masser, 1993b). After pools are drained, veterinarians suggest that the refill water contain 1 to 2 milligrams/liter of chlorine to reduce bacteria and fungi (Schaeffer, 1990).

4.3.13.3 Water Quality Management and Effluent Treatment Practices

Raw wastewater from alligator production facilities closely resembles domestic wastewater. The major difference is that alligators tend to excrete approximately twice the amount of ammonia per body mass when compared to humans (Pardue et al., 1994). The concentrations of various alligator raw wastewater constituents are presented in Table 4.3–11.

Effluent treatment practices vary significantly from facility to facility. Most facilities use oxidation ponds or lagoons to treat effluent from the raising operations. In some cases, facilities have begun to experiment with the use of "package plants" to treat raw wastewater before it is recycled for cleaning purposes. These "plants" are small filtration units designed for the needs of individual facilities.

Table 4.3–11. Pollutant Concentrations in Alligator Raw Wastewater

Parameter	*Concentrations (mg/L)*
Ammonia	77.5
Nitrate	4.6
TKN	153.4
Total phosphorus	10.9
Soluble phosphorus	7.6
BOD_5	452
pH	6.9
Calcium	13.4
Magnesium	5
Sodium	14.8
Conductivity	650
Total solids	379
Volatile solids	219

Source: Pardue et al., 1994.

4.4 TRENDS IN THE INDUSTRY

Based on an estimated increase in population in the United States from 270 million in 1998 to 310 million in 2015, it is likely that the U.S. demand for AAP products will continue to increase (Tomasso, 2002). The dependency of the United States on imported seafood might also be a factor in the future growth of AAP in this country. As world capture fisheries continue to decline and collapse, it is likely that AAP products will provide a source to meet the growing demand for fish products. Recently, American consumers have demanded more fresh seafood rather than canned or cured. If the trend toward fresh seafood continues, AAP will provide an important supply (Tomasso, 2002).

Despite an anticipated increase in demand for AAP products, the opportunities for expansion within the industry are limited by the demands of production systems. For pond systems, there are limited sites available with suitable land and water supplies for additional pond facilities. Increased profitability for production in pond systems will depend on improving efficiencies in farm management.

The expansion of flow-through systems is also limited by the availability of appropriate sites with suitable water sources. Development of this sector will depend on increased demand and its impact on profitability based on price. It is likely that conventional flow-through systems will be modified to some form of recirculating system or partitioned AAP system (Chen et al., 2002; Losordo and Timmons, 1994).

Recirculating systems have potential for expansion with continued research and technology development. There is a great deal of interest in recirculating systems because of their ability to reuse and recycle water. Although they are too expensive to use for the production of most species at this time, recirculating systems have the potential to expand in the future because they rely on smaller spaces for their facilities and use less water.

For net pen systems, limited nearshore sites are available for AAP, and net pens are not permitted in the Great Lakes. There are potentially an unlimited number of offshore sites, but the technology to support these offshore sites is expensive and not fully developed. This option is not likely to be developed in the near future while it is still less expensive to import salmon from other countries.

4.5 AQUATIC ANIMAL PRODUCTION SIZE CATEGORIES

In evaluating the detailed industry survey data related to facility annual production, EPA identified several variables distinguishing various types of facilities. CAAP facilities varied by type of facility operation (species and production system) and type of wastewater management (e.g., direct discharger, indirect discharger, no discharge/wastes applied to land on site). EPA identified annual production levels (by mass) at facilities based on the responses provided by individual facilities to the detailed industry survey. For the final regulation, EPA grouped facilities into two size categories:

- <100,000 pounds annual production
- ≥100,000 pounds annual production

For the purposes of estimating costs, loads, economic impacts, and non-water quality impacts (NWQIs), EPA used facility-level production and revenue data to project facilities that would meet the definition of a CAAP facility as defined in 40 CFR 122.24 and Appendix C to Part 122. The Small Business Administration's (SBA's) standard to determine a "small business" in the AAP industry is $750,000 annual revenues at the company level.

EPA used the results of the production rate thresholds to exclude facilities annually producing less than 100,000 pounds from the scope of the rule because the Agency anticipates that the technologies on which the options are based would not be economically achievable (and in some cases would be cost-prohibitive) for the facilities with the lowest production threshold (the smallest facilities).

4.6 INDUSTRY DEFINITION

The AAP industry includes sites that fall within the North American Industry Classification System (NAICS) codes 112511 (finfish farming and fish hatcheries), 112512 (shellfish farming), 112519 (other animal aquaculture), and part of 712130 (aquariums, part of zoos and botanical gardens). The first three groups (NAICS 112511, 112512, and 112519) have SBA size standards of $750,000, while the SBA size standard for NAICS 712l30 is $5.0 million. SBA sets up standards to define whether an entity is small and eligible for Government programs and preferences reserved for “small business” concerns. Size standards have been established for types of economic activity, or industry, generally under the NAICS. Refer to 13 CFR Part 121 for more detailed information. EPA uses these SBA size standards to conduct preliminary analyses to determine the number of small businesses in an industrial category and whether the proposed rule would have a significant impact on a substantial number of small entities.

4.7 REFERENCES

AII (Aeration Industries International, Inc.). 1989. *Lobster Pounds Add Weight and Profits*. Aeration Industries International, Inc., Minneapolis, MN.

Avault, J. 1996a. *Fundamentals of Aquaculture*, pp 188–192. AVA Publishing, Baton Rouge, LA.

Avault, J. 1996b. *Fundamentals of Aquaculture*, pp 75–77. AVA Publishing, Baton Rouge, LA.

Bayer, B. 2002. Lobster Institute, Orono, ME. Personal communication, January 23, 2002.

Bengtson, D.A., and G. Nardi. 2000. Summer Flounder Culture. In *Encyclopedia of Aquaculture*, ed. R.R. Stickney, pp. 907–913. John Wiley and Sons, Inc., NY.

Bergheim, A., J.P. Aabel, and E.A. Seymour. 1991. Past and Present Approaches to Aquaculture Waste Management in Norwegian Net Pen Culture Practices. In *Nutritional Strategies and Aquaculture Waste*, *Proceedings of the 1st International Symposium on Nutritional Strategies and Management of Aquaculture Waste, University of Guelph, Ontario, Canada*, ed. C.B. Cowey and C.Y. Cho, pp. 117–136.

Billard, R., and J.O.T. Jensen. 1996. Gamete Removal, Fertilization, and Incubation. In *Principles of Salmonid Culture*, ed. W. Pennell and B.A. Burton, pp. 291–364. Elsevier Science, Amsterdam, The Netherlands.

Boardman, G.D., V. Maillard, J. Nyland, G.J. Flick, and G.S. Libey. 1998. *The Characterization, Treatment, and Improvement of Aquacultural Effluents.* Departments of Civil and Environmental Engineering, Food Science and Technology, and Fisheries and Wildlife Sciences, Virginia Polytechnic Institute and State University, Blacksburg, VA.

Boyd, C.E. 1990. *Water Quality in Ponds for Aquaculture*. Alabama Agricultural Experiment Station, Auburn University, Auburn, AL.

Boyd, C.E. 2000. Farm Effluent During Draining for Harvest. *The Global Aquaculture Advocate* (August):26–27.

Boyd, C.E., and J.W. Clay. 1998. Shrimp Aquaculture and the Environment. *Scientific American* (June):42–49.

Boyd, C.E., J. Queiroz, J. –Y. Lee, M. Rowan, G. Whitis, and A. Gross. 2000. Environmental Assessment of Channel Catfish (*Ictalurus punctatus*) Farming in Alabama. *Journal of the World Aquaculture Society* 31:511–544.

Boyd, C.E., and C.S. Tucker. 1995. Sustainability of Channel Catfish Farming. *World Aquaculture* 26(3):45–53.

Boyd, C.E., and C.S. Tucker. 1998. *Pond Aquaculture Water Quality Management*. pp. 541–575. Kluwer Academic Publishers, Norwell, MA.

Browdy, C.L., and D. Bratvold. 1998. *Preliminary Development of Biosecure Shrimp Production System*. U.S. Marine Shrimp Farming Program Biosecurity Workshop, February 1998.

Browdy, C.L., and A.F. Holland. 1998. Shrimp Virus Risk Management: A South Carolina Case Study. *Aquatic Nuisance Species Digest* 2(3, February).

Browdy, C.L., A.D. Stokes, and P.A. Sandifer. 1996. *Shrimp Farm Management: Meeting the Challenges*. Waddell Mariculture Center, Proceedings of the International Forum of Shrimp Culture, August 1, 1996.

Brunson, M.W., and H.R. Robinette. 2000. Sunfish Culture. In *Encyclopedia of Aquaculture*, ed. R.R. Stickney, pp. 913–917. John Wiley and Sons, Inc., NY.

Bury, D., and J.S. Graves. 2000. Status of Knowledge of Atlantic Sturgeon (*Acipenser oxyrinchus oxyrinchus*), Gulf of Mexico Sturgeon (*A. oxyrinchus desotoi*), Shortnose Sturgeon (*A. brevirostrum*), White Sturgeon (*A.transmontanus*), and Bester Hybrid Sturgeon (*Huso huso* x *A. ruthenus*) as It Relates to Risks for Their Culture in the State of Florida. Revised August 2000. In *Proceedings of the Florida Sturgeon Culture Risk Assessment Workshop,* April 6–7, 2000, Sarasota, FL.

Cain, K., and D. Garling. 1993. *Trout Culture in the North Central Region*. North Central Regional Aquaculture Center. Lansing, MI.

Chapman, F.A. 2000. Ornamental Fish Culture, *Freshwater*. In *Encyclopedia of Aquaculture*, ed. R.R. Stickney, pp. 602–610. John Wiley and Sons, NY.

Chen, S., S. Summerfelt, T. Losordo, and R. Malone. 2002. Recirculating Systems, Effluents, and Treatments. In *Aquaculture and the Environment in the United States*, ed. J. Tomasso, pp. 119–140. U.S. Aquaculture Society, A Chapter of the World Aquaculture Society, Baton Rouge, LA.

Clarke, W.C., R.L. Saunders, and S.D. McCormick. 1996. Smolt Production. In *Principles of Salmonid Culture*, ed. W. Pennell and B.A. Burton, pp. 517–567. Elsevier Science, Amsterdam, The Netherlands.

Clarke, W.C. 2000. Smolting. In *Encyclopedia of Aquaculture*, ed. R.R. Stickney, pp. 879–884. John Wiley and Sons, NY.

Collins, C., and N. Stone. 1999. Arkansas Aquaculture Production 1998. *Aquaculture Magazine* 25(4):64–66.

Conte, F.S., S.I. Doroshov, P.B. Lutes, and E.M. Strange. 1988. *Hatchery Manual for the White Sturgeon,* Acipenser transmontanus *Richardson, With Application to Other North American Acipenseridae*. Publication 3322. Cooperative Extension, University of California Division of Agriculture and Natural Resources, Berkeley, CA.

Coulson, R.A., T.D. Coulson, and J.D. Herbert. 1995, December. Some Comments on Growing Alligators. Supplement to *Gator Tales*, Louisiana Alligator Farmers and Ranchers Newsletter.

D'Abramo, L.R., and M.W. Brunson. 1996a, July. *Biology and Life History of Freshwater Prawns*. SRAC publication no. 483. Southern Regional Aquaculture Center, Stoneville, MS.

D'Abramo, L.R., and M.W. Brunson. 1996b, July. *Production of Freshwater Prawns in Ponds*. SRAC publication no. 484. Southern Regional Aquaculture Center, Stoneville, MS.

Davis, J.T. n.d. *Crawfish Production*. Special project no. 87–EXCA–3–0836. U.S. Department of Agriculture, Cooperative State Research Service and Extension Service, Washington, DC.

Davis, J.T., and J.T. Lock. 1997, October. *Culture of Largemouth Bass Fingerlings*. SRAC publication no. 201. Southern Regional Aquaculture Center, Stoneville, MS.

de la Bretonne, L.W., Jr., and R.P. Romaire. 1990a. *Crawfish Production: Harvesting, Marketing, and Economics*. SRAC publication no. 240. Southern Regional Aquaculture Center, Stoneville, MS.

de la Bretonne, L.W., Jr., and R. P. Romaire. 1990b. *Crawfish Culture: Site Selection, Pond Construction and Water Quality*. SRAC publication no. 240. Southern Regional Aquaculture Center, Stoneville, MS.

Deniseger, J., and L.J. Erickson. 1998. *Salmon Aquaculture in Broughton Archipelago: the Results of a Sediment Sampling Program — 1996/97, A Data Report*. Ministry of Environment, Lands, and Parks, Pollution Prevention and Pesticides Management, Environmental Section, Nanaimo, British Columbia.

Dierberg, F.E., and W. Kiattisimkul. 1996. Issues, Impacts, and Implications of Shrimp Aquaculture in Thailand. *Environmental Management* 20(5):649–666.

Doroshov, S.I. 2000. The Escape of Cultured Sturgeon and the Interbreeding with Wild Stock. In *Proceedings of the Florida Sturgeon Culture Risk Assessment Workshop*. April 6–7, 2000, Sarasota, FL.

Dunning, R., and D. Sloan. n.d. *Aquaculture in North Carolina, Rainbow Trout: Inputs, Outputs, and Economics*. North Carolina Department of Agriculture and Consumer Services, Division of Aquaculture and Natural Resources, Franklin, NC.

Erickson, L.J. 1999. Personal communication with Mike Clipper (USEPA), April 20, 1999.

Eversole, A.G., and W. R. McClain. 2000. Crawfish Culture. In *Encyclopedia of Aquaculture*, ed. R.R. Stickney, pp. 185–198. John Wiley and Sons, NY.

FDA (Food and Drug Administration). 2002. *Drugs Approved for Use in Aquaculture (Poikilothermic Food Species).* <http://www.fda.gov/cvm/index/aquaculture/appendixa6.htm>. Accessed March 2002.

Fish Farming News. 2000. Water Reuse Key to Future of Texas Shrimp Farming. *Fish Farming News* (January/February).

Fitzpatrick, M.S, W.M. Contreras-Sanchez, and C.B. Schreck. 2000. *Fate of Methyltestosterone in the Pond Environment: Detection of MT in Soil After Treatment with MT Food*. Pond Dynamics, Aquaculture CRSP, Corvallis, OR.

Francis-Floyd, R. 2000. Disease History of Cultured Sturgeon in Florida, 1990–1999. In *Proceedings of the Florida Sturgeon Culture Risk Assessment Workshop*, April 6–7, 2000, Sarasota, FL.

Friedland, K. 2000. *Status of Fisheries Resources off Northeastern United States—Atlantic and Shortnose Sturgeons*. <http://www.nefsc.nmfs.gov/sos/spsyn/af/sturgeon/>. Last revised January 2000. Accessed March 2002.

Gatlin, D.M., III. 2001. *Guidelines for Nutrition and Feeding of Striped Bass and Hybrids*. <http://www.aquafeed.com/sp_stbass1.html>. Accessed December 2002.

Government of British Columbia. n.d. *Conservation Fish Culture for White Sturgeon*. <http://www.bcfisheries.gov.bc.ca/fishhabitats/Sturgeon/Culture.htm>. Accessed March 2002.

Grace, G.R., and R.H. Piedrahita. 1994. Carbon Dioxide Control. In *Aquaculture Water Reuse Systems: Engineering Design and Management,* ed. M. B. Timmons and T. M. Losordo, pp. 209–234. Elsevier Science Publishing Company. Amsterdam, The Netherlands.

Gulf of Maine Aquarium. 2000. *All About Lobsters: Lobstering History*. Lobster History—Gulf of Maine Aquarium. <http://www.octupus.gma.org/lobsters/allaboutlobsters/lobsterhistory.html>. Accessed January 2002.

Gunderson J.L., and P. Tucker. 2000. *A White Paper on the Status of Needs of Baitfish Aquaculture in the North Central Region*. Draft document. North Central Regional Aquaculture Center, Lansing, MI.

Haamer, J. 1996. Improving Water Quality in a Eutrophied Fjord System with Mussel Farming. *Ambio* 25(5):356–362.

Hardy, R.W., G.C.G. Fornshell, and E.L. Brannon. 2000. Rainbow Trout Culture. In *Encyclopedia of Aquaculture*, ed. R.R. Stickney, pp. 716–722. John Wiley and Sons, Inc., NY.

Harrell, R.M. 1997. *Morone* Pond Production. In *Striped Bass and Other Morone Culture: Developments in Aquaculture and Fisheries Science,* ed. R.M. Harell, vol. 30. pp. 75–97. Elsevier Science, Amsterdam, The Netherlands.

Harrell, R.M., and D.W. Webster. 1997. An Overview of *Morone* Culture. In *Striped Bass and Other Morone Culture: Developments in Aquaculture and Fisheries Science,* ed. R.M. Harell, vol. 30. pp. 1–10. Elsevier Science, Amsterdam, The Netherlands.

Hartman, K.J., and B. Preston. 2001. Stocking. pp 661–686. In *Fish Hatchery Management*, 2d ed. G.A. Wedemeyer, ed. American Fisheries Society, Bethesda, MD.

Heidinger, R. 2000. Black Bass/Largemouth Bass Culture. In *Encyclopedia of Aquaculture*, ed. R.R. Stickney, pp. 108–117. John Wiley and Sons, Inc. NY.

Heykoop, J., and D. Freschette. 1999. A Dynamic Model of the U.S. Alligator Industry: Lessons for Sustainable Use and Farm Management. In *Proceedings from the American Agricultural Economics Association Annual Meeting*, Nashville, TN, August 8–11, 1999.

Hinshaw, J.M. 2000. *Trout Farming: Carrying Capacity and Inventory Management*. SRAC publication no. 222. Southern Regional Aquaculture Center, Stoneville, MS.

Hochheimer, J.N., and F.W. Wheaton. 1997. Intensive Culture of Striped Bass. In *Striped Bass and Other Morone Culture: Developments in Aquaculture and Fisheries Science,* ed. R.M. Harell, vol. 30. pp. 127–168. Elsevier Science, Amsterdam, The Netherlands.

Hodgkins, H. 2002. Executive Secretary Maine Lobster Pound Association, Orono, ME. Personal communication, January 23, 2002.

Holt, G.J. 2000. Ornamental Fish Culture, Marine. In *Encyclopedia of Aquaculture*, ed. R.R. Stickney, pp. 610–614. John Wiley and Sons, Inc., NY.

Hopkins, J.S., R.D. Hamilton, P.A. Sandifer, C.L. Browdy, and A.D. Stokes. 1993. Effect of Water Exchange Rate on Production, Water Quality, Effluent Characteristics and Nitrogen Budgets of Intensive Shrimp Ponds. *Journal of the World Aquaculture Society* 24(3):304–320.

Hopkins, J.S., A.D. Stokes, C.L. Browdy, and P.A. Sandifer. 1991. The Relationship Between Feeding Rate, Paddlewheel Aeration Rate and Expected Dawn DO in Intensive Shrimp Ponds. *Aquacultural Engineering* 10(1991):281–290.

Howell, W.H., and M. K. Litvak. 2000. Winter Flounder Culture. In *Encyclopedia of Aquaculture*, ed. R.R. Stickney, pp. 998–1005. John Wiley and Sons, Inc., NY.

IDEQ (Idaho Department of Environmental Quality). n.d. *Idaho Waste Management Guidelines for Aquaculture Operations*. Idaho Department of Environmental Quality. <http://www2.state.id.us/deq/water/gw/Aquaculture_Guidelines.pdf>. Accessed August 2002.

Illinois-Indiana Sea Grant. n.d. *Sea Grant Tip Sheet Series AS-508.* <http://www.aquanic.org/publicat/state/il-in/as-508.htm>. Accessed March 2002.

Imperial Irrigation District. 1998. *Aquatic Weed Control.* <http://www.iid.com/aboutiid/env-weed.html>. Accessed March 2002.

Iversen, E.S., D.M. Allen, and J.B. Higman. 1993. *Shrimp Capture and Culture Fisheries in the United States*. Halsted Press, an Imprint of John Wiley and Sons, Inc., NY.

Jensen, G. 2000. U.S. Department of Agriculture, Washington DC. Personal communication with Marta Jordan (USEPA), December 12, 2000.

Jensen, J. 1989. *Watershed Fish Production Ponds: Site Selection and Construction*, SRAC publication no. 102. Southern Regional Aquaculture Center, Stoneville, MS.

JSA (Joint Subcommittee on Aquaculture). 2000a. *Effluents from Catfish Aquaculture Ponds*. Prepared by the Technical Subgroup for Catfish Production in Ponds, Joint Subcommittee on Aquaculture, Washington, DC.

JSA (Joint Subcommittee on Aquaculture). 2000b. *Comments submitted to EPA in response to Industry Profile Draft: Ornamental Fish,* Washington, DC.

JSA (Joint Subcommittee on Aquaculture). 2000c. *Comments submitted to EPA in response to Industry Profile Draft: Molluscan Shellfish*, Washington, DC.

Karlsen, L. 1993. Chapter 3: Developments in Salmon Aquaculture Technology. pp. 59–82. In *Salmon Aquaculture*. eds. K. Heen, R.L. Monahan, and F. Utter. Halsted Press, New York, NY.

Kendra, W. 1991. Quality of Salmon Hatchery Effluents During a Summer Low-Flow Season. *Transactions of the American Fisheries Society* 120:43-51.

KSU (Kentucky State University). 2002. *Shrimp Manual*. Kentucky State University Aquaculture Program. <http://www.ksuaquaculture.org/ShrimpManual%20text%20Apr.pdf>. Accessed May 2002.

Kohler, C.C. 2000a. Striped Bass and Hybrid Striped Bass Culture. In *Encyclopedia of Aquaculture*, ed. R.R. Stickney, pp. 898–907. John Wiley and Sons, Inc., NY.

Kohler, C.C. 2000b. *A White Paper on the Status and Needs of Tilapia Aquaculture in the North Central Region*. North Central Regional Aquaculture Center, Lansing, MI.

Kraeuter, J., B. Dewey, and M. Rice. 2000. *Preliminary Response to EPA's Aquaculture Industry Regulatory Development Data Needs*. Joint Subcommittee on Aquaculture, Molluscan Shellfish Aquaculture Technical Subgroup, Washington, DC.

KSUAP (Kentucky State University Aquaculture Program.) n.d. Perca flavescens: *Yellow Perch*. <http://www.ksuaquaculture.org/fish.yperch.htm>. Accessed March 2002.

Lane, T.J., and F.W. King. 1996. *Alligator Production in Florida*. VM-52. Department of Large Animal Clinical Services, College of Veterinary Medicine, Florida Cooperative Extension Service, University of Florida, Gainesville, FL.

Lawson, T. 1995a. Raceway Culture Systems. pp. 176–179. In *Fundamentals of Aquacultural Engineering*. Chapman & Hall, New York, NY.

Lawson, T. 1995b. Water Supply. pp. 48–57. In *Fundamentals of Aquacultural Engineering*. Chapman & Hall, New York, NY.

Lawson, T. 1995c. Oxygen and Aeration. pp. 248–310. In *Fundamentals of Aquacultural Engineering*. Chapman & Hall, New York, NY.

Lobster Institute. 1995, October 18. *Lobster Pound*. Lobster Pound—University of Maine. <http://www.lobster.um.maine.edu/lobster/library/publications/lobsterpound/lobsterpound.html>. Accessed November 2001.

Losordo, T.M. and M.B. Timmons. 1994. An Introduction to Water Reuse Systems. In *Aquaculture Water Reuse Systems: Engineering Design and Management*, ed. M.B. Timmons and T.M. Losordo, pp1–7. Elsevier, Amsterdam, The Netherlands.

Loughlin, M., R. Bayer, D. Prince, T. Axford, and H. Hodgkins. 2000, October. *Lobster Pound Manual*. The Lobster Institute and the Maine Agricultural and Forest Experiment Station at the University of Maine, Orono, ME.

Loyless, J.C., and R.F. Malone. 1997. A Sodium Bicarbonate Dosing Methodology for pH Management in Freshwater-Recirculating Aquaculture Systems. *The Progressive Fish-Culturist* 59:198–205.

Loyless J.C., and R.F. Malone. 1998. Evaluation of Airlift Pump Capabilities for Water Delivery, Aeration, and Degasification for Application to Recirculating Aquaculture Systems. *Aquacultural Engineering* 18:117–133.

LSU (Louisiana State University). 1999. *Crawfish Production Manual*. Publication no. 2637. Louisiana State University, Agricultural Center, Louisiana Cooperative Extension Service, Baton Rouge, LA.

Maine. 2002. *Maine Landing Statistics for 2000*. Maine Department of Marine Resources. <http://www.state.me.us/dmr/Comfish/maine_2000_landing_statistics.htm>. Accessed January 2002.

Malone, R.F., and L.E. Beecher. 2000. Use of Floating Bead Filters to Recondition Recirculating Waters in Warmwater Aquaculture Production Systems. *Aquacultural Engineering* 22(1–2):33–56.

Manci, B. 2000. Prospects for Yellow Perch Aquaculture. *Global Aquaculture Advocate* 3(6):62–63.

Masser, M.P. 1993a. *Alligator Production: Breeding and Egg Incubation*. SRAC publication no. 231. Southern Regional Aquaculture Center, Stoneville, MS.

Masser, M.P. 1993b. *Alligator Production: Growout and Harvest*. SRAC publication no. 232. Southern Regional Aquaculture Center, Stoneville, MS.

Masser, M.P. 2000. Alligator Aquaculture. In *Encyclopedia of Aquaculture*, ed. R.R. Stickney, pp. 27–33. John Wiley and Sons, Inc., NY.

McGee, M.V., and C. E. Boyd. 1983. Evaluation of the Influence of Water Exchange in Channel Catfish Ponds. *Transactions of the American Fisheries Society* 112:557–560.

Meronek, T.G., F.A. Copes, and D.W. Coble. 1997. *The Bait Industry in Illinois, Michigan, Minnesota, Ohio, South Dakota, and Wisconsin*. Technical bulletin series 105. North Central Regional Aquaculture Center, Lansing, MI.

Mims, S.D., W.L. Shelton, F.S. Wynne, and R.J. Onders. 1999, November. *Production of Paddlefish*. SRAC publication no. 437. Southern Regional Aquaculture Center, Stoneville, MS.

Mosso, D., J. Harcum, and J. Hochheimer. 2003. *Water Quality and Sediment/Benthic Impacts and Modeling Tools used in Assessment at Net Pen Facilities*. Tetra Tech, Inc., Fairfax, Virginia.

Nash, C., ed. 2001, September. *The Net-Pen Salmon Farming Industry in the Pacific Northwest.* NOAA Technical Memorandum NMFS-NWFS-49.

Newell, R.I.E., and J.A. Ott. 1999. Macrobenthic Communities and Eutrophication. pp. 265–293. In Ecosystems at the Land-Sea Margin: Drainage Basin to Coastal Sea. eds. T. Malone, A. Malej, L. Harding, N. Smodlaka, and R. Turner, *Coastal and Estuarine Studies* 55, American Geophysical Union.

Novotny, A.J., and W. Pennell. 1996. Rearing Salmonids to Market Size in Marine Waters. In *Principles of Salmonid Culture*, ed. W. Pennell and B.A. Burton, pp. 569–611. Elsevier Science, Amsterdam, The Netherlands.

Papoutsoglou, S.E., and G. Tziha. 1996. Blue Tilapia (Oreochromis aureus) Growth Rate in Relation to DO Concentrations Under Recirculating Water Conditions. *Aquacultural Engineering* 15. 181–192.

Pardue, J.H., R.D. DeLaune, W.H. Patrick, Jr., and J.A. Nyman. 1994. Treatment of Alligator Farm Wastewater Using Land Application*.* *Aquacultural Engineering* 13:129–145.

Parker, N.C. 1981. An Air-Operated Fish Culture System with Water-Reuse and Subsurface Silo*,* pp. 131–137. In *Proceedings of the Bioengineering Symposium for Fish Culture*, Vol. 1.

Parker, N.C., and M.A. Suttle. 1987. Design of Airlift Pumps for Water Circulation and Aeration in Aquaculture*.* *Aquacultural Engineering* 6:97–110.

Pennell, W., and W.E. McLean. 1996. Early Rearing. In *Principles of Salmonid Culture*, ed. W. Pennell and B. A. Burton, pp. 365–465. Elsevier Science, Amsterdam, The Netherlands.

Pennell, W., E.D. Lane, and F. Dalziel. 2001. Open Systems: The Culture of Fish for Release into Natural Systems. pp. 187–239. In *Fish Hatchery Management*, 2d ed. G.A. Wedemeyer, ed. American Fisheries Society, Bethesda, MD.

Pepper, V.A., and L.W. Crim. 1996. Broodstock Management. In *Principles of Salmonid Culture*, ed. W. Pennell and B.A. Burton, pp. 231–289. Elsevier Science, Amsterdam, The Netherlands.

Piper, R., I. McElwain, L. Orme, J. McCraren, L. Fowler, and J. Leonard. 1982. *Fish Hatchery Management*. U.S. Fish Department of the Interior, Fish and Wildlife Service, Washington, DC.

Plumb, J.A. 1994a. *Health Maintenance of Cultured Fishes: Principal Microbial Diseases*. pp. 49–55. CRC Press, Boca Raton, FL.

Plumb, J.A. 1994b. *Health Maintenance of Cultured Fishes: Principal Microbial Diseases*. pp. 135–160. CRC Press, Boca Raton, FL.

Plumb, J.A. 1994c. *Health Maintenance of Cultured Fishes: Principal Microbial Diseases*. pp. 177–222. CRC Press, Boca Raton, FL.

Plumb, J.A. 1997. Infectious Diseases of Striped Bass. In *Striped Bass and Other Morone Culture: Developments in Aquaculture and Fisheries Science*, ed. R.M. Harell, vol. 30. pp. 271–313. Elsevier Science, Amsterdam, The Netherlands.

Popma, T., and M. Masser. 1999. *Tilapia: Life History and Biolog*y. SRAC publication no. 283. Southern Regional Aquaculture Center, Stoneville, MS.

PSMFC (Pacific States Marine Fisheries Commission). 1996. *White Sturgeon. (Fact sheet)*. Revised December 12, 1996. <http://www.psmfc.org/habitat/edu_wsturg_fact.html>. Accessed March 2002.

Radonski, G.C., and R.G. Martin. 1986. Fish Culture is a too, not a Panacea. pp. 7–13. In *Fish Culture in Fisheries Management*. R.H. Stroud, ed. American Fisheries Society, Bethesda, MD.

Rakocy, J.E. 1989. *Tank Culture of Tilapia*. SRAC publication no. 282. Southern Regional Aquaculture Center, Stoneville, MS.

Rakocy, J.E., and A.S McGinty. 1989. *Cage Culture of Tilapia*. SRAC publication no. 281. Southern Regional Aquaculture Center, Stoneville, MS.

Reinemann, D.J., and M.B. Timmons. 1989. Prediction of Oxygen Transfer and Total Dissolved Gas Pressure in Airlift Pumping. *Aquaculture Engineering* 8:29–46.

Revkin, A.C. 1999, June 24. Making Up Their Beds and Hoping the Oysters Will Move In. *New York Times*.

Rice, M.A., A. Valliere, M. Gibson, and A. Ganz. 1999. Eutrophication Control by Bivalves: Population Filtration, Sedimentation and Nutrient Removal Through Secondary Production. (Abstract). *Journal of Shellfish Research* 18:333.

Roberts, R.J., and R.W. Hardy. 2000. Salmon Culture. In *Encyclopedia of Aquaculture*, ed. R.R. Stickney, pp. 773–778. John Wiley and Sons, Inc., NY.

Rodhouse, P.G., and C.M. Roden. 1987. Carbon Budget for Coastal Inlet in Relation to Intensive Cultivation of Suspension-Feeding Bivalve Molluscs. *Marine Ecology Progress Series* 36:225–236.

SAIC (Science Applications International Corporation). 1998. *Final Report: Best Conventional Pollutant Control Technology and/or Best Available Technology to Support Economically Achievable Effluent Limitations for Shrimp Farm Operations*. Submitted to U.S. Environmental Protection Agency by Science Applications International Corporation, San Diego, CA.

Sastry B.N., A.A. Delos Reyes, Jr., K.A. Rusch, and R.F. Malone. 1999. Nitrification Performance of a Bubble-Washed Bead Filter for Combined Solids Removal and Biological Filtration in a Recirculating Aquaculture System. *Aquacultural Engineering* 19:105–117.

Schaeffer, D.O. 1990. Preventing Bacteria Disease Problems in Farm Raised Alligators. *Gator Tales* 1(1):5–6. (Louisiana Alligator Farmers and Ranchers newsletter).

Schramm, H.L., Jr., and R.G. Piper, eds. 1995. Uses and Effects of Cultured Fishes in Aquatic Ecosystems. American Fisheries Society Symposium 15, Bethesda, MD.

Schwartz, M.F., and C.E. Boyd. 1994a. Effluent Quality During Harvest of Channel Catfish from Watershed Ponds. *Progressive-Fish Culturist* 56:25–32.

Schwartz, M.F., and C.E. Boyd. 1994b. Channel Catfish Pond Effluents. *Progressive Fish-Culturist* 56:273–281.

Seok, K., S. Leonard, C.E. Boyd, and M.F. Schwartz. 1995. Water Quality in Annually Drained and Undrained Channel Catfish Ponds over a Three-Year Period. *Progressive Fish-Culturist* 57:52–58.

Smith, B.W., and W.C. Reeves. 1986. Stocking Warmwater Species to Restore and Enhance Fisheries. pp 17–29. In *Fish Culture in Fisheries Management*. R.H. Stroud, ed. American Fisheries, Bethesda, MD.

SRAC (Southern Regional Aquaculture Center). 1998. *Improving Feeds for Hybrid Striped Bass*. SRAC publication no. 304. Southern Regional Aquaculture Center, Stoneville, MS.

Stickney, R.R. 2000a. Carp Culture. In *Encyclopedia of Aquaculture*, ed. R.R. Stickney, pp. 147–153. John Wiley and Sons, Inc., NY.

Stickney, R.R. 2000b. History of Aquaculture. In *Encyclopedia of Aquaculture*. ed. R.R. Stickney, pp. 436–446. John Wiley and Sons, Inc., NY.

Stickney, R.R. 2000c. Tilapia Culture. In *Encyclopedia of Aquaculture*, ed., R.R. Stickney, pp. 934–941. John Wiley and Sons, Inc., NY.

Stickney, R.R. 2000d. Cage Culture. In *Encyclopedia of Aquaculture*, ed., R.R. Stickney, pp. 139–141. John Wiley and Sons, Inc., NY.

Stone, N. 2000. Baitfish Culture. In *Encyclopedia of Aquaculture*, ed. R.R. Stickney, pp. 97–104. John Wiley and Sons, Inc., NY.

Stone, N., and E. Park. 2001. University of Arkansas and Arkansas Baitfish Association. Personal communication, August, 1, 2001.

Stone, N., H. Thomforde, and E. Park. n.d. *Baitfish Production in Ponds*. Prepared for U.S. Environmental Protection Agency by the Technical Subgroup of the Joint Subcommittee on Aquaculture, Aquaculture Effluent Task Force, Washington, DC.

Stone, N., E. Park, L. Dorman, and H. Thomforde. 1997. *Baitfish Culture in Arkansas: Golden Shiners, Goldfish, and Fathead Minnows*. MP 386. Cooperative Extension Program, University of Arkansas at Pine Bluff.

Stoskopf, M.K. 1993. *Fish Medicine*. W.B. Saunders Company, Philadelphia, PA.

Sullivan, C.V., D.L. Berlinsky, and R.G. Hodson. 1997. Reproduction. In *Striped Bass and Other Morone Culture: Developments in Aquaculture and Fisheries Science,* ed. R.M. Harell, vol. 30. pp. 11–73. Elsevier Science, Amsterdam, The Netherlands.

Summerfelt, R.C. 2000. Walleye culture. In *Encyclopedia of Aquaculture*, ed. R.R. Stickney, pp. 970–985. John Wiley and Sons, Inc., NY.

Summerfelt, R.C., ed. 1996. *Walleye Culture Manual*, NCARC Culture Series 101. North Central Regional Aquaculture Center Publications Office, Iowa State University, Ames, IA.

Teichert-Coddington, D.R., D.B. Rouse, A. Potts, and C.E. Boyd. 1999. Treatment of Harvest Discharge from Intensive Shrimp Ponds by Settling. *Aquacultural Engineering* 19:147–161.

Tetra Tech, Inc. 2001. Technical Memorandum: Summary of Molluscan Shellfish Conference Call and Follow-Up. Tetra Tech, Inc., Fairfax, VA.

Tetra Tech, Inc. 2002a, August. Site visit report for Cantrell Creek Trout Farm, Brevard, NC.

Tetra Tech, Inc. 2002b, August. Site visit report for Clear Springs Foods, Inc., Box Canyon Facility, Buhl, ID.

Tetra Tech, Inc. 2002c, August. Site visit report for Lake Wheeler Road Agricultural Facility, Raleigh, NC.

Tetra Tech, Inc. 2002d, August. Site visit report for Aubrey Onley Aquaculture, Hertford, NC.

Tetra Tech, Inc. 2002e, August. Site visit report for DB Rice Fisheries, Birch Harbor, ME.

Tidwell, J.H., S.D. Coyle, and T.A. Woods. 2000. *Species Profile: Largemouth Bass*. SRAC publication no. 722. Southern Regional Aquaculture Center, Stoneville, MS.

Tomasso, J.R. 2002. Global Aquaculture Production with an Emphasis on the United States. In *Aquaculture and the Environment in the United States*, ed. J. Tomasso, pp. 1–7. U.S. Aquaculture Society, A Chapter of the World Aquaculture Society, Baton Rouge, LA.

Treece, G. 2000. Shrimp Culture. In *Encyclopedia of Aquaculture*, ed. R.R. Stickney, pp. 798–868. John Wiley and Sons, Inc., NY.

Tucker, C.S. 1996. The Ecology of Channel Catfish Ponds in Northwest Mississippi. *Reviews in Fisheries Science* 49(1):1–55.

Tucker, C.S., ed. 1998. *Characterization and Management of Effluents from Aquaculture Ponds in the Southern United States*. SRAC final report no. 600. Southern Regional Aquaculture Center, Stoneville, MS.

Tucker, C.S. 2000. Channel Catfish Culture. In *the Encyclopedia of Aquaculture*, ed. R.R. Stickney, pp.153–170. John Wiley and Sons, Inc., NY.

Tucker, C.S., and J.A. Hargreaves. 1998. *Effluents from Channel Catfish Aquaculture Ponds*. Thad Cochran National Warmwater Aquaculture Center, Mississippi State University, Stoneville, MS.

Tucker, C.S., S.W. Kingsbury, J.W. Pote, and C.W. Wax. 1996. Effects of Water Management Practices on Discharge of Nutrients and Organic Matter from Channel Catfish Ponds. *Aquaculture* 147:57–69.

Tucker, C.S., and E.H. Robinson. 1990. *Channel Catfish Farming Handbook*. Van Nostrand, Reinhold, NY.

Tucker, C.S., and M. van der Ploeg. 1993. Seasonal Changes in Water Quality in Commercial Channel Catfish Ponds in Mississippi. *Journal of the World Aquaculture Society* 24(4): 473–481.

Tucker, C.S., C.E. Boyd, and J.A. Hargreaves. 2002. Characterization and Management of Effluents from Warmwater Aquaculture Ponds. In *Aquaculture and the Environment in the United States*, ed. J. Tomasso, pp. 35–76. U.S. Aquaculture Society, A Chapter of the World Aquaculture Society, Baton Rouge, LA.

USDA (U.S. Department of Agriculture). 1995. *Overview of Aquaculture in the United States*. U.S. Department of Agriculture, Animal and Plant Health Information Services, Centers for Epidemiology and Animal Health, Fort Collins, CO.

USDA (U.S. Department of Agriculture). 1997. *Catfish NAHMS '97, Part II: Reference of 1996 U.S. Catfish Management Practices*. Centers for Epidemiology and Animal Health, USDA/APHIS, Fort Collins, CO.

USDA (U.S. Department of Agriculture). 2000. *The 1998 Census of Aquaculture*. U.S. Department of Agriculture, National Agriculture Statistics Service, Washington, DC.

USDA (U.S. Department of Agriculture). 2001. *Aquaculture Outloo*k. U.S. Department of Agriculture, Economic Research Service, Washington, DC.

USDA (U.S. Department of Agriculture). 2002, February 7. *Catfish Production.* U.S. Department of Agriculture, National Agricultural Statistics Service, Washington, DC.

USEPA (U.S. Environmental Protection Agency). 2002. National Pollutant Discharge Elimination System Permit no. ME0036234, issued to Acadia Aquaculture, Inc. Signed February 21, 2002.

Wallat, G., and L. Tiu. 1999. *Production and Feed Training of Yellow Perch Fingerlings*. The Ohio State University, Centers at Piketon, Piketon, OH.

Watson, C.A. 2002. University of Florida. Personal communication, February 2, 2002.

Watson, C.A., and J.V. Shireman. n.d. *Production of Ornamental Aquarium Fish*. Fact Sheet FA-35. University of Florida, Cooperative Extension Service, Gainesville, FL.

WDF (Washington Department of Fisheries). 1990. *Final Programmatic Environmental Impact Statement, Fish Culture in Floating Net-Pens*. Washington Department of Fisheries, Olympia, WA.

Weber, M.L. 1997. *Farming Salmon: Briefing Book*. The Consultative Group on Biological Diversity, San Francisco, CA.

Wellborn, T., and M. Brunson. 1997. *Construction of Levee-type Ponds for Fish Production,* SRAC publication no. 101. Southern Regional Aquaculture Center, Stoneville, MS.

Westers, H. 2001. Production. In *Fishery Hatchery Management,* 2d ed., ed. G.A. Wedemeyer, pp. 31–90. American Fisheries Society, Bethesda, MD.

Weston, D. 1986, August. *The Environmental Effects of Floating Mariculture in Puget Sound*. Prepared for the Washington State Department of Fisheries and the Washington State Department of Ecology, Seattle, WA.

Weston, D. 1992. Status of Waste Management Practices in Marine Net-Pen Systems in Washington State. In *National Livestock, Poultry, and Aquaculture Waste Management*, ed. J. Blake, J. Donald, and W. Magette, pp. 211–214. publication no. 03-92. American Society of Agricultural Engineers, St. Joseph, MI.

Wikfors, G.H. 1999. Personal communication to Tim Motte, Coastal Resources Management Council. Cited in JSA (Joint Subcommittee on Aquaculture), Comments Submitted to EPA in Response to Draft Industry Profile: Molluscan Shellfish.

Winton, J.R. 2001. Fish Health Management. In *Fish Hatchery Management,* 2d ed., ed. G. Wedemeyer. American Fisheries Society, Bethesda, MD.

Zimmerman, T. 1998. How to Revive the Chesapeake Bay: Filter it with Billions and Billions of Oysters. *U.S. News & World Report*, December 29, 1997– January 5, 1998.