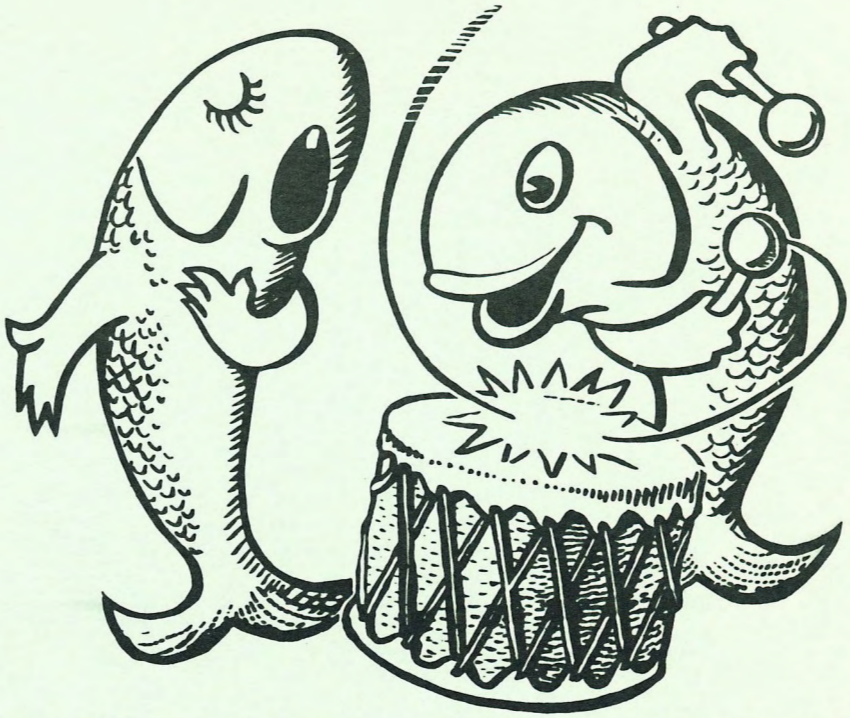

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John H. Prescott
Executive Director
New England Aquarium

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ILLUSTRATIONS

NOMEUS GRONOVII AS A COMMENSAL OF PHYSALIA PHYSALIA

Robert L. Jenkins, Curator

Marineland Research Laboratory
Marineland, Florida

The Portuguese Man-of-War, Physalia physali, is a common siphonophore in the Gulf Stream along the east coast of Florida and is capable of inflicting painful stings with their nematocyst laden tentacles. (1) The basic morphology of Physalia is that of a cup-shaped pneumatophore, which floats on the surface, with the gastrozooids and gonozooids suspended just beneath the "cup" and the dactylozooids hanging further down to form the drift net. (2) It is the dactylozooid that is responsible for the stinging and capturing of prey and its transport by contraction to the gastrozooids. These have two basic shapes: long forms that have large "fishing" tentacles and shorter forms with small tentacles. (3) The float of Physalia may be 30 cm. long with the drift net extending for "many meters". (4)

Although very venomous, there are records of fish living commensalistically with Physalia, among which are the Yellow Jack (Caranx bartholomaei), the Pilotfish (Naucrates ductor), the Spotted Ruff (Mupus maculatus), and the Longspine Snipefish (Macrohamphosus scolopax), but are usually only found in juvenile stages. (5) It also should be noted that such fish normally are only in association with the compound "bunch" just under the float near the surface and not with the long drift net of the dactylozooids. (6)

The Portugese Man-of-War fish, Nomeus gronovii (Gmelin), however, has long been accepted as a symbiont of Physalia. (7) This relationship differs from the fishes previously mentioned, as Nomeus not only inhabits the area just under the pneumatophore but also swims among the dactylozooids with presumably little or no harm. (8,9) However, it should be noted that pieces of Nomeus have been isolated from the gastrozooids of Physalia, and the Nomeus has been observed feeding on the zooids and tentacles of its host, with tissue and discharge nematocysts of Physalia later being found in its gastro-intestinal tract. (10,11,12) This phenomena is unlike that of the anemonefish and the anemone where the fish usually exhibits total immunity to the anemone's sting. Here, the fish may swim directly into the tentacles of its host and has little to fear, providing its mucus remains intact. This immunity also can be renewed, even after separation has taken place for some time. (14, 15) This method of immunity sometimes

has been assigned to the relationship of Physalia and Nomeus. With these facts in mind, it was interesting to make behavioral observations during recent filming of Physalia and Nomeus together at the research laboratory.

Both Physalia and Nomeus were obtained from John Noyes of Fort Lauderdale, Florida on August 21, 1977, which had been captured some time beforehand and were shipped separately. The Nomeus were kept separate from any Physalia for nearly 15 days during which they were maintained in natural seawater and fed daily. The original Physalia were used in the initial filming and lasted in time similar to previous experience, the average being about four days. (16) On the eighth of August, 1977, freshly caught Physalia were obtained and placed in a large 475 gallon filming tank, over four feet deep. After preliminary filming of the one Physalia, a single, seven centimeter Nomeus was acclimated to the system in a solid container from which the fish could not see its intended host.

Upon introduction into the tank, the Nomeus immediately started swimming in a large circular pattern around the Physalia in both directions. The fish appeared to be rather leary of its natural host and stayed approximately 10 to 20 cm. from the main body of tentacles while making these circular patterns. This possibly was due to the fact that the Physalia remained rather stationary, as this behavior was noted later on when observations were made while moving the Physalia in the tank. Approximatley 15 minutes after introduction, the Nomeus began to swim closer to the Physalia and started to "nip" at the edges of the gonozooids after several times having swam past and inspecting them. At this point, the fish was avoiding very intentionally the larger dactylozooids. During one of the sorties of "nipping", the fish actually was stung and held by several of the smaller dactylozooids on the tip of its mouth but appeared to disengage itself easily. For filming purposes, a fresher Physalia was then exchanged for the one currently in use and was approximately 1.5 times larger than the first with an equally greater mass of "drift net". The larger dactylozooids with the "fishing" tentacles were very conspicuous on the Physalia.

About 15 minutes after introduction of the new Man-of-War, the Nomeus again started its slow, close swimming and periodic nipping of the smaller gonozooids. After five minutes the fish moved in and tore off about 5 mm of one of the small dactylozooids and ingested it. The fish then returned to its circular swimming pattern and did not approach the Physalia for about 15 minutes, whereupon it moved in and tore off and ingested an

entire 2.5 cm tentacle. For the next 15 minutes, it tore off small tentacles around 10 mm in length at about 2 minute intervals. At times when the fish would go to bite, some of the larger tentacles would move. Whenever this happened, the Nomeus would veer away and resume its circular swimming for a while without attempting to feed and then move in again when the Man-of-War had stopped.

A little more than an hour after being introduced, the Nomeus deliberately swam in and grabbed a larger tentacle than before and tore it loose violently enough to shake the entire Physalia. Ten minutes after this the fish came into direct contact with the smaller tentacles which did not appear to sting it too well. This was the first direct contact with any of the dactylozooids noted with either Physalia since the original introduction of the fish. At slightly less than two hours from the time of introduction, the fish started to swim in and around the larger tentacles but was careful not to come into direct contact with them. At no time did the fish engage in the "rubbing" acclimating behavior as noted above for the anemonefish/anemone commensalism. The fish did come into contact with the smaller tentacles with its "nose" and apparently was not stung. However, a larger tentacle did again sting and hold the fish by the caudal fin for an instant, but it was capable of shaking free.

A dead Nomeus (45 minutes old) from the same group as the experimental specimen was then offered to the Physalia's "feeding" tentacles. The carcass was stung immediately and held, and the tentacle contracted up to the gastrozooids. The live Nomeus at this time obviously became distressed, swam more erratically, and moved in very close to the carcass as if to inspect it. The tentacle holding the carcass then relaxed, and the carcass dropped from the gastrozooids but still was held by the dactylozooids. This lasted for two minutes, whereupon the tentacle contracted and redelivered the carcass to the gastrozooids. The gastrozooids then formed their characteristic "bag" and began to digest the fish, and it was approximately 25% digested at the termination of the experiment two hours later.

Six minutes after the Physalia started to digest the dead Nomeus, the live Nomeus, who had been swimming closer than ever to the Physalia, was caught severely by one of the largest "fishing" tentacles across the left side of its body in a dorsoventral direction just caudal of the primary dorsal fin. As the fish was stung and held strongly, intervention by the observer was necessary in order for the fish to remove itself. After separation, it swam very erratically and at a distance from the Physalia, favoring its right side. Although apparently severely

stung, the Nomeus resumed its previous swimming that was being used just before the large sting. From the time of feeding the dead Nomeus to the Physalia, the live fish did not feed on the tentacles until after adjusting to the severe sting as noted above. The erratic behavior following the sting was identical to that of previous observers. Forty minutes after recovering from the large sting, the Nomeus again started to feed upon the gastrozooids and the smaller dactylozooids but only in those areas away from where the dead Nomeus was being digested. Feeding was observed to take place at five to ten minute intervals for the next hour and ten minutes. During the final period of the experiment, the Physalia was allowed to remain stationary for about ten minutes and then slowly moved about for approximately five minutes. This moving was done five times, and in each instance, the Nomeus displayed the same behavior.

When stationary, the Nomeus would swim around the periphery of the drift net and only rarely swim between the tentacles, as noted previously. Upon moving, however, the fish would dart into the center mass of the large dactylozooids and swim among the tentacles, again being careful not to come into direct contact with them. The fish displayed a relative ease at maintaining a safe distance from the dactylozooids which appeared to be due to its swimming style. Utilizing its pectoral fins much like a wrasse for propulsion and its pelvic fins spread like a fan on which it would "float", it apparently only uses its caudal fin for short, fast darts. This swimming "style", or behavior, appears to be well adapted for its commensal existence with Physalia. Observations were then terminated at this point, due to filming consideration, which gave a total observation time of just over four hours.

Discussion

Nomeus is capable of withstanding injections ten times the normal strength of Physalia stings that would kill another fish. (18, 19) However, it does not appear from the observations made that Nomeus gronovii adapts to Physalia in the same manner as does the anemone fish to anemones. These damselfish become immune to the anemone sting by absorbing onto its skin and mucus the anemone's substance that it secretes onto its tentacles to keep them from stinging each other. (20) The Man-of-War fish clearly can be stung if it comes into contact with the large dactylozooids and does not engage in the "anemone fish-acclimitization" behavior mentioned earlier. The fish also can be stung in the wild if it comes into contact with the tentacles which usually happens in a capture situation. (21) The fish does indeed ingest portions of its host and, therefore, would require some immunity to the discharged nematocysts that

often are found in its stomach. Therefore, it is felt by the author at this time that Nomeus gronovii does not generate a direct physiological immunity to the Physalia sting, as has been thought in the past. Rather, it utilizes avoidance behavior instead of physiological adaptation as its primary means of "immunity" while living among the drift net of Physalia.

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NOTES ON THE TREATMENT OF *CRYPTOCARYON*

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The following discussion centers around a review of drugs and chemicals that have been used and reported in the literature as attempts to control or cure the disease of marine fishes caused by the ciliated protozoan parasite, *Cryptocaryon irritans* (Protozoa: Ciliata: Hymenostomatida: Ophryoglenidae). A description of the disease and the signs by which it may be recognized are to be found in a number of sources and will not be dealt with here (Blasiola, 1976; Spotte, 1973). However, in order to better understand why the parasite is difficult at best to eradicate, a brief review of the characteristics of three primary stages in its development will be given along with an analysis of treatments in general at each particular stage in its life cycle.

The parasite first attaches itself to the integument of the fish and then burrows just underneath the skin where it feeds on epidermal cells from the fish's body. Here it is more or less protected from harm by the body of its host while it grows, and any attempt to kill the trophant (as it is called at this stage) must involve either highly protozoan specific drugs or ones that will act faster on parasite cell structures than it does on fish cells in order to kill the parasite before it kills or otherwise adversely stresses the fish you are trying to save. Drugs placed in the water are rarely absorbed through the skin of a fish in sufficient quantity to affect parasites embedded there. Freshwater as a treatment is a notable exception as it is drawn into the fish's body by osmotic forces.

Upon maturing, the parasite forms a cyst and drops off of the fish onto the aquarium substrate and undergoes a series of divisions within the cyst to form a large number of daughter cells. At this stage it is virtually impervious to treatment as any known medication that would destroy the organism within the cyst also would kill the fish you are trying to treat long before the encysted parasite is affected. This stage is extremely resistant and may last for 5-10 days or even longer at low water temperatures. For all practical purposes it is impossible to treat at this stage, and most treatment failures probably are due to the fact that effective drugs are not maintained at a lethal concentration for a sufficiently long period of time to allow for all of the existing cysts to fully develop and burst open so that the next stage, the tomite, can be killed.

The daughter cells, or tomites, which issue from the cyst, are

free-swimming ciliates and have only a brief life span of a day or two in which to find a host. It is at this point that the disease is most vulnerable. There are a number of good medications which will kill the tomite stage. The problem is that the medication must be present in sufficient strength to kill the parasite before it can find a host and become safely embedded. Quite often a good medication may be present but not in sufficient strength to be effective. For instance, drugs that are rapidly absorbed by the substrate or inactivated over a short period of time may have been present initially but will have diluted their strength until they are no longer present at an adequate concentration.

To repeat, an effective drug must be kept in the tank at a sufficient concentration to kill the parasite for at least 5-10 days, since there is no way to determine exactly when a cyst is going to discharge its cargo of pathogens. Quite often, all the free-swimming tomites will be killed, and the disease will seemingly be cured for a few days. The medication level will be allowed to drop as the fish are looking much better (all of the embedded trophonts will have matured and dropped off as cysts). Then suddenly, a week or so later, a new swarm of tomites is released and the problem seemingly begins anew. Actually, it never went away, it was only incubating. While the outright killing power of a drug is certainly important, the length of time it will remain stable in a salt water solution, the amount of medication present, and the duration of treatment are all equally or more important.

Among the drugs that have been discovered to be effective against one stage or another are a group of quinine derivatives that have been used for years in humans as anti-malaria drugs. They all have similar actions but differ slightly in toxicity to the fish, solubility, and speed of action in combating the parasite. Their usage will now be described.

Atebrine hydrochloride, also known as atebrine, chinacrine, mepacrine, or quinacrine hydrochloride may be used at 8-12 mg./gallon in saltwater tanks as 4-6 mg./gallon treatments, two days apart. Keep the tank dark as strong light will inactivate this drug. It is effective in established aquaria even though it might damage, but probably will not destroy the biological filter bed. Although it may tint fish yellow, this will disappear after a short time. It is the treatment of choice in my book (van Duijn, 1973; Kingsford, 1975).

Chloroquine phosphate, also known as truxquine, roquine or aralen phosphate may be used at a dosage of 40 mg./gallon. (Kingsford, 1975).

Primaquine phosphate available as 26.3 mg. tablets from Winthrop Laboratories, may be used at a dosage of 15 mg./gallon. Prolonged treatment may result in dark pigmentation of the fish, but this resolves itself upon discontinuance of treatment (Kingsford, 1975).

Quinine hydrochloride and quinine sulfate may be considered together and may be used interchangeably with atebrine at a rate of 8-12 mg./gallon. The only difference being that quinine hydrochloride is more readily soluble in saltwater than quinine sulfate and is slightly more effective over a wider pH range. If a choice is available, then quinine hydrochloride is most effective, followed by atebrine and then quinine sulfate. Atebrine is by far the least expensive to use. Toxic reactions may occasionally be encountered and is evidenced in the fish by loss of equilibrium, lying on the bottom, rapid breathing, and loss of appetite. The solution to this dilemma, should it occur, is to change water as soon as possible or administer a mild potassium permanganate solution (0.2% stock solution at 2-4 drops per gallon). Removal by carbon filtration will clear up the water, but too slowly to save the fish.

Another class of chemical compounds, which originally were developed not as fish medicants but as dyes or biological stains, are among the most widely used drugs available in freshwater for "Ich". Therefore, when "saltwater Ich" came along, it only seemed natural to utilize these same drugs in saltwater. Their effectiveness does not seem to be quite so great against Cryptocaryon as it is against Ich, however.

Acriflavine, neutral, may be used at a dosage of 3-10 ppm in water as a prolonged bath. However, it may damage the slime coating of the skin and create vulnerability to bacterial disease in marine fish (Kingsford, 1975). Therefore, it is not advisable as a treatment although it may be effective against the tomite stage. Mostly though, it is just inhibitory in its effects.

Malachite green (zinc free oxalate) and variations such an aniline green, bright green, evergreen B, light green N, victoria green B or WB, and malachite green G sulfate are reported to be effective against Cryptocaryon at a dosage of 2 ppm. However, it also may be toxic to small marine fishes as well. It probably is best not to use malachite green by itself because of its potential toxicity. When used in combination with formalin, their combined effects become multiplied, or synergistic, and a much smaller dose does a lot more good. Malachite green does break down fairly rapidly, so this combination should be administered three separate times on alternate days. Malachite

green at 0.1 ppm added to formalin at 50 ppm is more effective than either drug used alone. Malachite green may produce sterility in breeding fishes, so do not use on any breeding stock.

Methylene blue, or methylthionine chloride, added to the water until you can just vaguely see the back wall of the aquarium may be effective. Keep the solution a deep, dark, rich blue for 5-10 days, adding more dye if the water begins to clear only slightly. In freshwater "Ich", this concentration of medication has been shown to be absorbed through the fish's skin and to kill the embedded parasite. Presumably, it would do the same thing in saltwater fish, only more effectively because of osmotic pressures. Corals and other carbonate structures will absorb this dye more or less permanently, so treatment should be in a bare tank (Kingsford, 1975).

The remaining group of drugs and chemicals contains an assortment of inorganic compounds and antibiotics which have been found effective or which are suspected of being so. In addition to these, if you wish to experiment, any proven anti-protozoan drug, or more specifically any known cure, for freshwater "Ich" might be tried. Who knows, maybe you will discover something different about the similarities between freshwater "Ich" and Cryptocaryon which will make its control more positive than it is at present.

Copper sulfate at 0.15-0.25 ppm as a long duration bath (3-10 days) is inhibitory. It is a very slow cure, but it might work if treatment is prolonged (30 days). Treat in bare tanks as copper is affected adversely by calcium carbonate levels in water (Nigrelli & Ruggieri, 1966; Dempster, 1955; Braker, 1961; Kingsford, 1975; Reichenbach-Klinke & Landolt, 1973; Dulin, 1976). There is no scientific evidence to support the contention that copper will kill a biological filter bed, nor is there anything but empirical knowledge and experience to support the contention that it will not. The question is still at issue, but the effectiveness of its use as a treatment certainly becomes more questionable in the presence of calcium carbonate buffered sea water. Citric acid or glacial acetic acid will chelate the copper and make it stay in solution for a longer period of time before precipitating out as $CuCO_3Cu(OH)_2H_2O$, but chelated copper is much less effective as a treatment, simply because the chelating agent is too effective. It keeps the copper in solution so well it does not act against the parasite either.

A formulation of cupric acetate, $Cu(CH_3COO)_2$, or verdigris, with formalin and Tris buffer also has been tried against

Cryptocaryon. Use it in the proportions of cupric acetate (0.42 ppm) + formalin (5.26 ppm) + Tris buffer (4.8 ppm) (Nigrelli & Ruggieri, 1966).

Daraprim, or pyrimethamine, has been reported by Kingsford, 1975, to be useful but dangerous at 4-8 mg./gallon. Different bottles have different strengths. Toxic reactions, should they occur, are typified by a marked darkening of the fish's colors, and death may occur with an open gaping mouth indicating respiratory distress or obstruction. It is manufactured by Burroughs Welcome & Company, Research Triangle Park, North Carolina.

Flagyl, or metronidazole, at 250 mg./10 gallons will kill tomites. It is manufactured by Searle Laboratories. This is a very safe drug to use as considerable overdose is possible with no harm to your fish.

Formalin (37-40%) at 1 cc./5 gallons in a bare tank is effective against tomites. Formalin mixes very slowly in sea water, so dilute before adding or stir well while adding. Otherwise, you can get a lens of almost pure formalin which can be lethal to a fish swimming through it. A white precipitate occasionally found in formalin is paraformaldehyde. It is toxic to fish and should not be allowed to get into the water.

Formalin probably works best when used in conjunction with a freshwater dip at 1 cc./gallon of freshwater for 5-15 minutes. Freshwater alone also is effective as it can kill the embedded organism on the fish's body as well as the tomites. Water enters through the cell wall of the parasite rapidly, expanding the cell by osmotic pressure until it ruptures. It is a very effective treatment. Repeat every 5 days to kill deeply embedded parasites and newly infective stages. Treatment for less than 3 minutes will not kill the parasite and is a waste of time at such short duration. The only drawback is that marine fishes can go into pH shock unless precautions are taken to buffer the freshwater with an agent such as sodium bicarbonate to a pH of 8.0-8.3.

Hydrogen peroxide may be added to freshwater dips at a rate of 17.5 ml./liter of the 3% standard solution instead of, but not in addition to, formalin.

Potassium permanganate has been used in two fashions. One, as a dip of 10-45 seconds in a 1,000 ppm solution, or two, as a 0.2% solution added to the aquarium water at a rate of one cc./5 gallons. This produces a temporary immediate effect and is unworkable as a long term treatment. Therefore, I would not recommend its use. In addition, potassium permanganate is a

powerful oxidizing agent, and it either can burn a fish's gills or skin or can produce a manganese precipitate that will clog the gills. Its effectiveness also may be altered by the prevailing water chemistry or by high organic content in the water.

Sodium chlorite also is reportedly effective against the tomite stage of Cryptocaryon, but there may be some question as to its efficacy (Dempster, 1970; Garibaldi, 1971; Kingsford, 1975).

Sulfathiazole and its more soluble salt, sulfathiazole sodium, can be used at a rate of 250 mg./gallon to kill tomites (any sodium or hydrochloride form of any drug will be more soluble and hence more effective at the higher pH range of saltwater). This is one of the early drugs developed to combat Cryptocaryon and Oodinium in place of copper and may be used safely with invertebrates in the tank. It is no longer as widely used as previously, but it is still one of the better drugs available.

While all of the above mentioned drugs are effective to a greater or lesser degree, usually lesser, I would make no definitive assertions that any known treatment is "the" answer to curing Cryptocaryon outbreaks and would not fully endorse any of them. The inclusion of a drug or manufacturer in this review is not an endorsement, nor is the exclusion of a drug or manufacturer a denial of that product or producer. Cryptocaryon is a very troublesome parasite, and I do not believe the final chapter has been written on its eradication in the aquarium. However, in my own experience, I have found that the following sequence of treatments works best for me in my area. I would caution readers that different strains of the parasite, pH, water hardness, temperature, or differing mineral ions contained naturally in the water supplies in your area may modify the results of this procedure for you and your fish.

Upon the discovery of a Cryptocaryon outbreak (or as a routine precautionary measure in suspect new arrivals), a 5-15 minute bath in freshwater, followed by quarantine in a bare 10 gallon tank containing 4-6 mg./gallon of atebrine (usually simply one-half of a 100 mg. tablet per 10 gallons) in saltwater obtained from the tank in which the fish were being kept. Filtration and aeration is provided by means of a sponge filter (inert substance). The tank is kept darkened by black plastic panels on all sides (a plastic covered trash can also will work). This is to keep the drug from being inactivated by light and also to reduce the amount of stress on the fish.

The second day the fish is given another freshwater dip followed by the addition of the other half of the atebrine tablet to the quarantine tank, making a total dosage of 8-12 mg./gallon.

On the third day, fourth day, and fifth day, a freshwater dip is given and the fish returned to the medicated quarantine tank. Usually by this time the disease is gone, and the water is changed in the quarantine tank. The fish is then watched closely for an additional 10 days. At the first sign of a reoccurrence, the entire procedure is started over. No food is offered during the time the fish is in the atebrine solution.

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JAWS TOO??

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Hull, Massachusetts

Contrast defines the extremes, giving perspective, which enables the middle to be seen more clearly. Case in point, imagine Caribbean diving with its optimum conditions, sunny blue skies, warm temperatures (air 80-85 F, water 75), excellent visibility, tropical breezes, and a host of other ideal feelings. With that image in your mind, now imagine diving in the icy North Atlantic where one has to contend with frigid temperatures, both water and air, murky visibility, strong tidal currents, etc. Now compare the native fauna of these areas. Beauty and color abound in the tropics, while in the North Atlantic, many "beasts" of grotesque size and shape inhabit the oft murky waters.

Probably one of the most beast-like fish to be encountered anywhere in the world, Lophius americanus, or the American Goosefish, lives along the Atlantic coast of the United States. The goosefish often captures sea birds as its vernacular name suggests. It is so unlike all other fish, that there is no danger of mistaking it for any other once it is seen. Most goosefish are seen washed up on the beach after a storm, when they resemble possibly something from another planet. They are seldom displayed in a public aquarium since they are difficult to obtain in good condition.

The goosefish is a member of the anglers, or family Lophidae. The soft, scaleless body is flattened with the head rounded and as broad as it is long. The most noticeable characteristic is its enormous mouth, which is directed upward. The lower jaw protrudes well beyond the upper jaw, exposing most of the lower teeth even when the mouth is closed. Both jaws are armed with long, slender, curved teeth, all alike in form, but of various sizes and all very sharp. They point inward toward the gullet. Some of the teeth may be as long as one inch in larger fish.

The top of the head bears three stiff, slender spines which represent the anterior part of the spiny dorsal fin. The moveable first spine, about 3-5 inches long, near the tip of the snout, bears an irregular, leaflike flap of skin on the end. This "fishing" mechanism is characteristic of most of the anglers and is important in the daily life of the goosefish as a lure for its prey.

Goosefish are chocolate brown above, often mottled with pale and dark. The ventral surface is dirty white. The boundary

line between dorsal and ventral is ringed with fleshy flaps which add to its bizarreness as well as aiding in camouflage.

Capture of a live goosefish for a public aquarium usually necessitates donning a wet suit and scuba equipment, as any other method (gill nets, trawls, etc.) seems to disturb the protective mucous layer, especially in the caudal area, leading to the rapid demise of the specimen. Antibiotics appear ineffective once this bacterial infection sets in.

Goosefish up to five feet and weighing up to 60 pounds have been reported, but the larger they are, the harder to handle and acclimate. Specimens from 10-20 pounds are ideal. Small specimens, less than three pounds, are reported from deep water. Goosefish have a wide tolerance for depth and temperature, but the best time to observe them is during the summer when they move into shallow water (10-50').

Goosefish usually lie motionless on the bottom, often partially buried under material that is tossed up while using their large pectoral fins and tail to dig a shallow hole. They have the ability to lighten or darken depending on the color of the bottom. Since they cannot swim too fast, it's relatively simple to catch them. The only difficulty is discovering them, since they are masters of camouflage. The best technique is to use a large hand net, allowing the fish to swim head first into the net. This technique avoids irritation to the caudal area, which is most sensitive. Once the fish is in the net, it should be carefully carried back to the transport container, preferably just large enough to hold the animal, thereby preventing movement and swimming, which again minimizes abrasion.

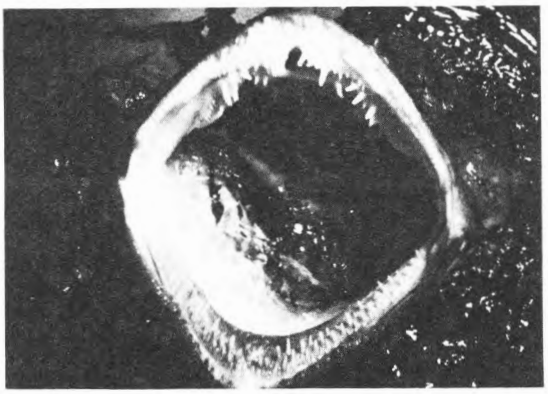
Once at the aquarium, old Mr. Goosefish should be placed alone in a shallow container with a large volume of water. Much mucous will accumulate in the tank during the first 24 hours. The following day, an attempt must be made to force feed the new acquisition, as goosefish will not thrive long in captivity on voluntary feeding. Besides, with its enormous mouth, capable of swallowing a fish almost its own size, it might easily devour other prize specimens, should it become too hungry.

Force feeding is quite simple, once the technique is mastered. A multiple vitamin supplement should be added to the food. Here briefly is the technique I have employed to keep goosefish for periods of more than a year in captivity (possibly a world's record). The use of gloves is recommended as a psychological assurance until the technique is perfected. Remember, the dentition is awesome, and one word of caution, goosefish are accustomed to lunging at their prey. However, a warning to the lunge is given when they erect the three dorsal spines on the top of the head. The trick is to slide a 10-12 inch fish headfirst directly

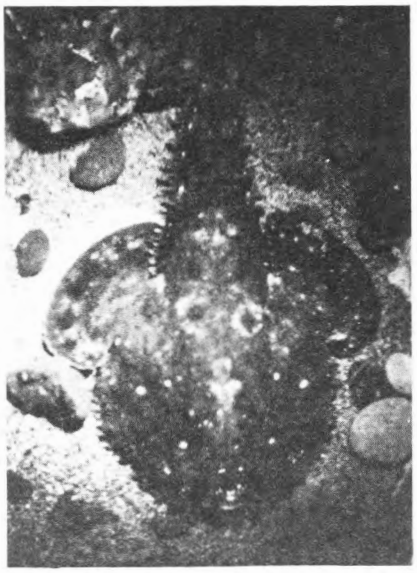
into the middle of the mouth. Once the head touches the triangular set of bones on the bottom of the mouth in front of the gullet, ingestion begins automatically and the remainder of the fish disappears, much like a child sucking in that last piece of spaghetti (mmm..good). The use of a feeding stick is recommended, but is sometimes more tedious.

The goosefish I have maintained usually swim for only two purposes, excretion or hunger. It's quite easy to slide a fish into the huge mouth, as the fish swims by with most of its mouth above water. They need only to be fed one or two fish every other day or so, since their activity is minimal.

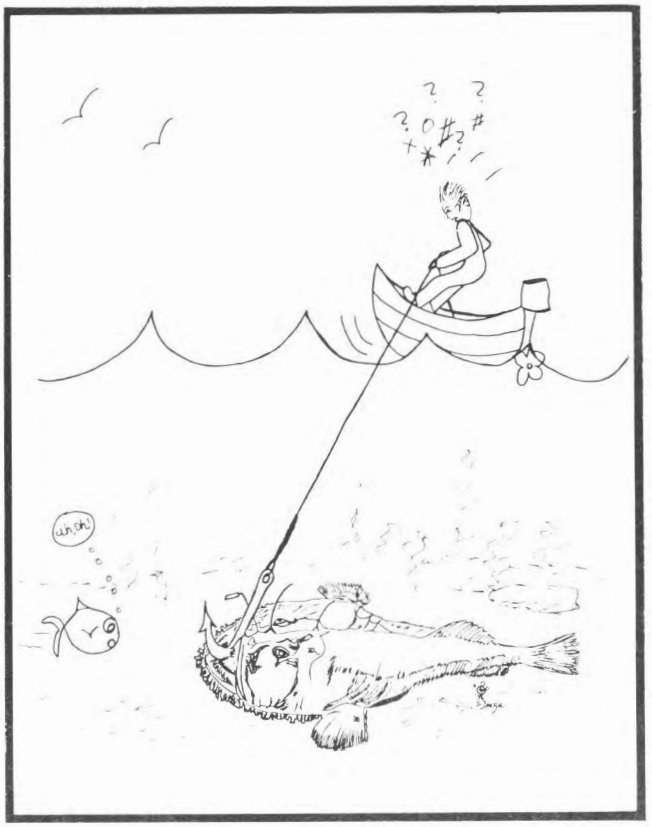
When people discover a live goosefish in the tank, the usual utterance is "Is that found around here?" or "Is that a fish?" What I believe to be one of the most beast-like creatures of the sea is indeed a fish, the sea bird eating goosefish.



25 lb. goosefish. Note mackeral in mouth. Photo by John Dinga and John Crowley.



15 lb. goosefish. Photo by John Crowley and John Dinga



CHLORAMPHENICOL PALMITATE IN THE TREATMENT OF BACTERIAL DISEASES OF TROPICAL FISHES

A. Baczewski and F. Kozlowski

Sea Fisheries Institute
Oceanographical Museum and Sea Aquarium
Gdynia, Poland

One of the most frequently occurring disease of salt and fresh-water fishes in the Aquarium of the Sea Fishing Institute in Gdynia are bacterial infections. The infections affect the following fish species: Puntius arulius (Jerdon), Puntius filamentosus (Cuvier and Valenciennes), Symphysodon aequifasciata axelrodi (Schultz), Symphysodon discus (Heckel).

Affected fishes display dropsical conditions with a distended abdomen, protruding scales, exophthalmos, and they develop skin ulcers. Postmortem examination reveals accumulation of serous fluid in the peritoneal cavity, edema of the kidney and a varied degree of catarrhal enteritis.

Gram-negative organisms such as Pseudomonas, Achromobacter, enteric bacteria, and Vibrio were isolated most frequently from the kidney and liver.

Treatments such as intensive water exchange and restriction in feeding were ineffective. As the disease progressed, fishes refused food and died.

During the treatment with sulfonamides and antibiotics, very good therapeutic effects were obtained when Detreopal "Polfa"* was added to the food. The compound contains detreomycin palmitynicum 3.74 g, sirupus and corrigentia to 60 ml. The syrup with detreomycin (chloramphenicol) as an active substance is quickly and almost entirely absorbed from the alimentary tract.

For larger fish, Detreopal is administered with shrimps (Crangon crangon) or herring (Clupea harengus) meat injected previously with the antibiotic. Smaller fish are fed with herring meat that has been minced and saturated for a half hour with Detreopal. Before preparing the food with medication, it should be washed to remove small pieces of meat which would contaminate the aquarium water.

* Detreopal "Polfa" is the designation for chloramphenicol palmitate, made in Poland by Krakowskie Zaklady Farmaceutyczne "Polfa", Krakow, POLAND. It is equivalent to Parke Davis chloramphenicol, chloromycetin palmitate.

Treatment is administered 5 to 14 days. The antibiotic is given once a day in a dose of 25 mg per 100 g of fish. A teaspoon contains approximately 250 mg of pure detreomycin. There is virtually no danger of an overdose of the drug. A marked improvement is noted very early in the course of treatment and continues after the period of medication is over. Seven days after therapy has ceased, recovery is complete.

This treatment was administered to Parascyllium collare (Ramsey and Ogilby) and Scatophagus argus (Gmelin). The fishes displayed characteristic septicemic ulcers on the skin with red centers and pale necrotic tissue on the periphery. The fish were lively, willingly took food, but the ulcers increased in size. After treatment with Detreopal for 14 days, recovery was complete.

It was observed that septicemic diseases seem to affect mainly voracious fishes in which overfeeding leads to the accumulation of adipose tissue and to metabolic disturbances and also delicate fishes which are not supplied with food to which they are accustomed in their natural environment.

SPECIAL REQUEST

Anyone having any information, or experience, in regards to the breeding and rearing of Octopus briareus (Common Reef, or Grass Octopus), Octopus joubani (Jouban's Octopus), or Octopus maya (Yucatan Octopus), is asked to contact the following individual.

Robert L. Jenkins, Curator
Marineland Research Laboratory
Route #1, Box 122
St. Augustine, Florida 32084

GOOD NEWS FOR AQUACULTURE CONNOISSEURS

Colin E. Nash
The Oceanic Institute
Makapuu Point
Waimanalo, Hawaii 96795

Reports from recent visitors to Cuba have revealed some preliminary and brief information of an extensive and valuable culture practice which has been developed there. Along the flat coastal areas of the island there are many hectares where the traditional activity of farming Lepisosteus corona c. (Raleigh) has been developed into an industrial occupation producing a select export commodity.

Known locally as the sea gar, this familiar species, when in short supply, can fetch export prices as high as $50/kg. The Cuban species is considered a great delicacy which maintains its high value, and individuals are consumed singly in smoked form. Before smoking, the head is cut off, but traditionally the head should be bitten off and spat into the fire.

The biology is not all that well documented. A herbivore throughout all its life stages (there is one major metamorphosis from a flat to a round form), the sea gar appears to be very easy to culture in Cuba. Unlike other Lepisosteidae found in the tropical regions, the Cuban specialty is brown in color with a distinct gold band across the pectoral region. It is generally a retiring individual, and its appendages are poorly developed. Maturity is reached within one year, and the preferred market size is about 15 cm or 10 g (dry weight) after curing.

Production is seasonal and is very dependent on the local weather conditions. There are indications that the pre-metamorphosed stage is susceptible to heteroecious fungus disease and mosaic virus infection, but effective treatments have been developed.

There are no external morphological differences between the sexes, and individuals can be selected for breeding only by sampling, using traditional techniques for taste and flavor.

It is hoped that more visitors to Cuba will bring back boxes of these fine sea gars. At present the samples enter the United States through European markets or are smuggled into Florida. There is some talk about an international trading agreement to share the results of successful culture techniques, which is exciting the aquaculture world. A herbivore with a high market value, it will sooth the nerves of many frustrated aquaculturists and, of course, the entrepreneurs in their search for that perfect bottom line.

MAINTAINING A "HANDS-ON" EXHIBIT

Judy Sullivan
Children's Museum of Hartford
West Hartford, Connecticut

The Aquarium of the Children's Museum of Hartford, Conn. has the fortune (or misfortune depending on whether you are the curator or educator) of owning a 250 gallon cold water, invertebrate "touch tank". The tank was designed by the maker as a unit for lobster culture but has been converted by the Aquarium staff to hold a number of different kinds of indigenous marine invertebrates allowed to be touched by visitors (table 1). The system contains its own hidden refrigeration unit, and filtration is achieved by continual water flow through the filter bed, thereby eliminating the use of airstones (fig. 1). The filter bed is made up of approximately 5 inches of dolomite, on top of which are placed a number of large stones, rocks, and seashells that simulate the natural environment and provide shelter for the residents.

There are many curatorial problems that are unique to a tank such as this, the most obvious being the problem of public hands which introduce bacteria and other pollutants of unknown character and origin to the water. I have seen atrocities such as an overly excited child falling into the tank, an overly excited adult dropping her chewed bubble gum into the water, not to mention the inky hands, the drooling toddler or the person with the bad cough and horrible sneeze who has never heard of antihistamines.

The handling of the animals exerts a tremendous stress on them. Coping with the introduction of external pollutants, as mentioned above, is done through a rigorous and strict curatorial schedule. The animals themselves also are on a schedule. They are "rotated", that is, animals on exhibit periodically get fresh replacements to give them time for R & R in our reserve tank. Rotation helps increase the lifespan of the animals but even more important to this end is the presence of an aquarium teacher stationed at the Hands-on tank at all times. (The teachers also are rotated for R & R.)

The presence of natural "enemies" and predator-prey relations are dealt with in a number of ways:

1. Designation of certain species to specific areas of the tank, thereby keeping the prey separated from the predator. This is

done by using a plastic grate (mesh size = 1.5mm), cut to size, in order to block off certain corners to provide protected areas for those delicate, tasty or otherwise vulnerable individuals (fig. 2). Although the fence is not foolproof, it does lessen the number of animals lost to attack by others (i.e. sea anemones, starfish, ailing crabs, etc.).

2. Eliminating the predator's weapon, i.e. the lobster's claws are kept from doing harm with the use of heavy duty rubber bands. This not only saves crabs but human fingers as well.

3. Allowing the predator-prey relationship to exist. The whelks, for instance, are not discouraged from eating clams since clams are replaced easily throughout the year.

The highest level of water quality is maintained at all times. A 20% water change is done every other week; bucket by bucket, I might add, since there is no built-in drain. Temperature, pH, specific gravity, NH_3, NO_2, and NO_3 determinations are done weekly. We have had only a few situations where a diatom filter had to be put into use to counteract poor water quality. One of these instances occurred during the summer when all too generous beach-goers brought in their souvenirs from the seashore; giant surf clams, hermit crabs by the dozen, and half-dead horseshoe crabs were all accepted, under protest, by the Aquarium staff and deposited into the hands-on tank. We soon had an overloaded system on our hands and the DE was put into action. We consider this a drastic measure, and we've had to resort to the use of the DE only three or four times in the past year. As with the rest of the Aquarium, tanks are kept biologically balanced so the use of external filter units are not needed.

Although there are 12 different species and 38 individuals living together, the Hands-on tank is a relatively tranquil settlement (at least until feeding time when all hell breaks loose). As a precaution to overloading an already "filled" system (table 1) with organics, each animal is fed individually. The type of food, of course, depends on the animal's nutritional requirements. The omnivores are fed a gelatin diet consisting of spinach, carrots, smelt, squid, shrimp and vitamins. Sea urchins thrive on spinach while the small sea anemones savor brine shrimp. All uneaten food is removed immediately.

Despite the problems and the amount of time involved for maintenance, the "Hands-on" tank is an indispensable exhibit. It is the focal point for many of our educational programs and used extensively to teach both children and adults about seashore inhabitants and their ecology. To many inner-city

residents, this hands-on experience is the first introduction to animals of the seashore. Watching a child's reaction when the crabs do not pinch or when the sea "flower" withdraws at touch or when a seemingly empty shell is filled with a curious inhabitant is a truly delightful experience which makes it all worth the extra effort of maintaining it.

	Number	Total wet weight (g)
MOLLUSCA *		
Busycon canaliculatum	6	787.2
Mytilus edulis	5	120.0
Venus mercenaria	3	101.2
ARTHROPODA		
Homarus americanus	1	337.5
Carcinus maenas	5	39.5
Cancer borealis	2	354.4
Pagurus longicarpus	5	1.0
Libinia	4	120.0
Limulus polyphemus	2	502.3
ECHINODERMATA		
Asterias forbesi	1	311.5
CNIDARIA		
Metridium	4	183.5
TOTAL	38	2858.3

Transient members: Littorina, Arbacia, Crossostrea, Nassarius, Spisula

Table 1: Number and weight of inhabitants.

* = weight without shell

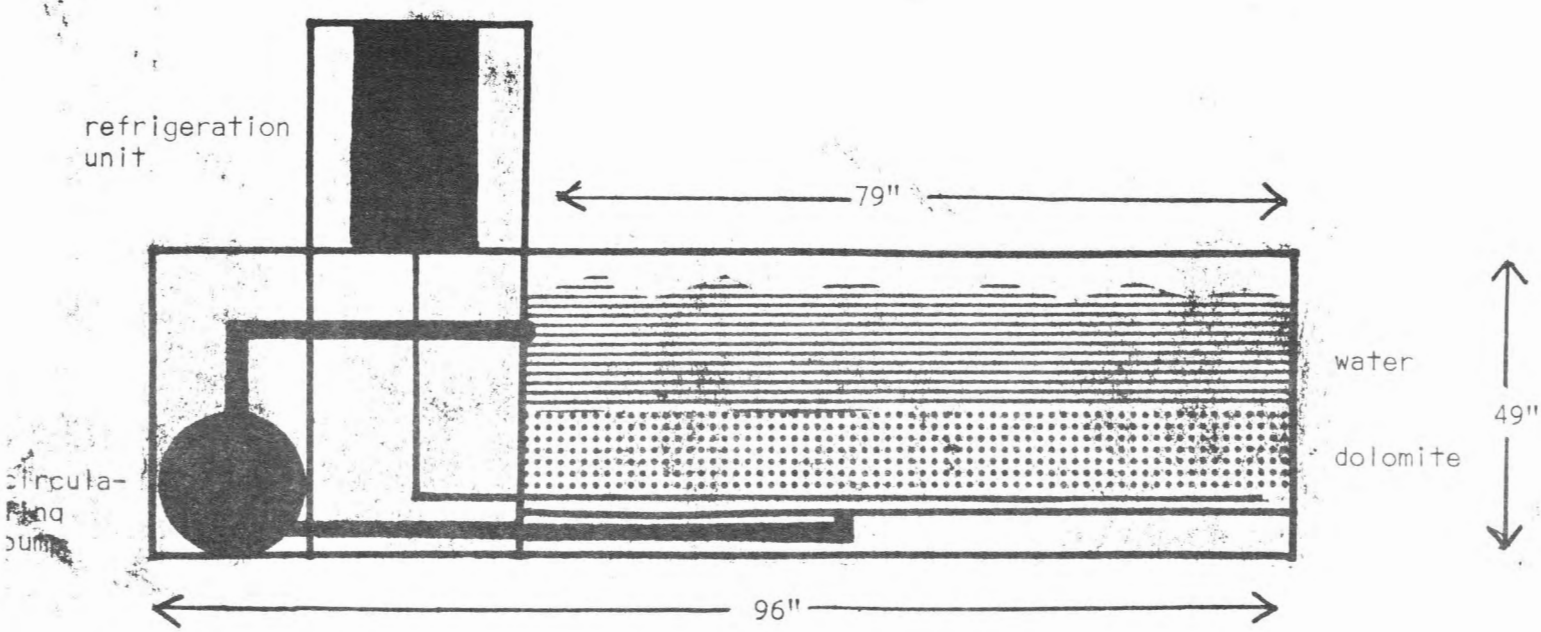


Fig. 1: Diagram of Hands-on Tank showing flow of water and dimensions.

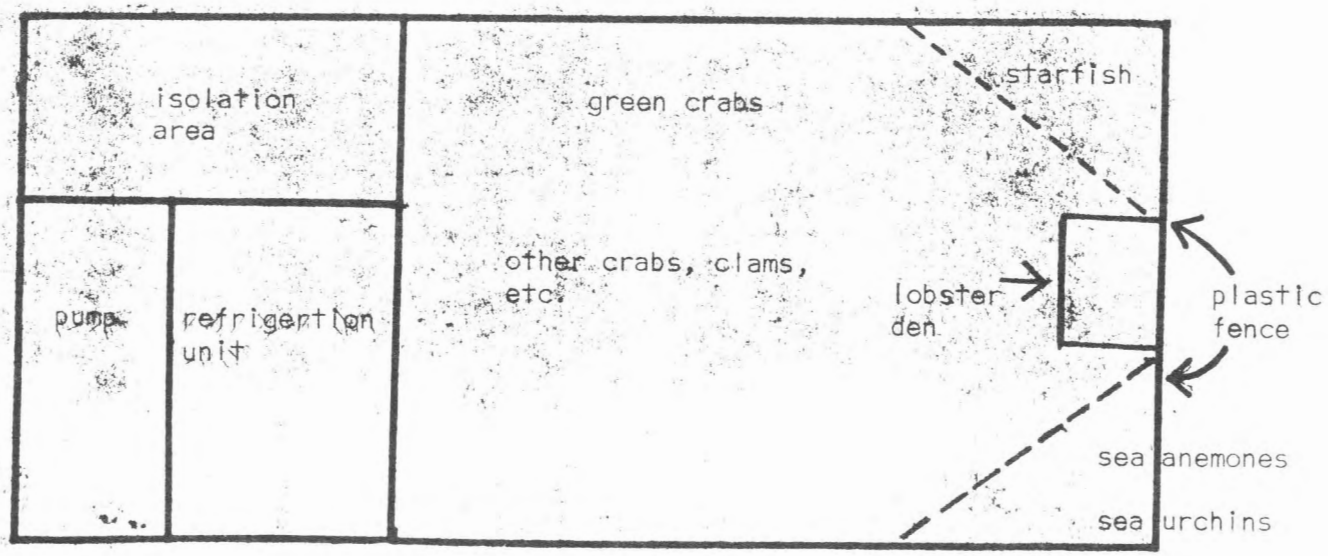


Fig. 2: Location of inhabitants and fenced regions.

A SYSTEM FOR REARING LARVAL MARINE FISH

Stephen D. Walker
San Antonio Zoological Gardens & Aquarium

Many factors are involved in successful reproduction of marine fish in captivity, and one of the most important of these is supplying a suitable rearing aquarium for the larvae. The design of such an aquarium becomes difficult, however, since it must try to simulate a pelagic environment within the confining restrictions of the aquarium itself. Most marine fish spend their larval period as part of the plankton drifting in the upper strata of the ocean. They thus are not inhibited by any physical barriers and are bathed in very pure seawater, rich in oxygen. Aquarium walls and closed system metabolic waste build-ups subject the larvae to radically different circumstances.

In order to relieve some of the problems of tank rearing, the aquarist must utilize a system that minimizes damage to the larvae. Before giving particulars about the set-up used at the San Antonio Zoo Aquarium for this purpose, I first would like to explain some of the factors that were considered in its design.

Most importantly, water quality must be kept at high standards. An open system can solve water quality problems, and indeed, many researchers whose facilities are close to the sea have made use of this. On the other hand, most aquarists (including us at the S.A. Zoo) are too far from a coastline for using an open system and, therefore, must rely on recirculated water. In this latter case, efficient biological filtration along with frequent water changes can be a reasonable substitute for an open system and in some ways is superior to it, since it is easier to monitor and control such things as temperature and salinity in closed systems.

When using recirculated water, one advantage can be had by making the volume large, thereby reducing rapid temperature fluctuation and slowing the rate of self-pollution brought on by the culture animals. Unfortunately, a large tank requires more food organisms to keep the concentration high enough to feed fish larvae, and if these food organisms are cultured rather than wild-caught, it may be very difficult to supply a sufficient amount. One workable compromise to this dilemma is to keep the water volume relatively large while restricting the larvae and their food to a smaller area. Biological filtration of a larvae tank is complicated by the fact that the fry do

much better in a bare tank. Gravel, it seems, presents a threat to the young, as they may sink into it and be unable to get out. The biological filter, therefore, should be outside the rearing tank.

Some authors (Houde and Ramsey 1971, Marliane 1976) support the use of cylindrical larvae culturing tanks on the premise that some of the larvae may become trapped in the corners of a rectangular aquarium. Indeed, in the final analysis, a cylindrical design may produce higher survival rates, but rectangular tanks have been used in numerous successful rearings (Richards and Palko 1969, Saksena et al. 1972, Arnold et al. 1976). This information, coupled with the fact that round tanks are difficult to construct and work with, led us to use readily available rectangular aquariums.

Most marine species are photopositive at hatching and will swim toward a light source. To keep the larvae from being attracted toward an outside light and thus contacting the glass, the tank should have black walls. Larvae tend to avoid a black wall, and in addition, it has been suggested by Blaxter (1962) that black makes a good contrasting background against which the fry may distinguish their food.

With these facts in mind, construction was begun on a rearing system that, hopefully, would prove suitable. After a number of refinements (and the loss of many batches of eggs and larvae), a workable solution was achieved. The set-up described below has been employed successfully to rear captive-spawned marine fish from eggs through metamorphosis to the juvenile stage. It is simple to construct with generally available materials, yet it has proven quite versatile and relatively maintenance-free in use.

Briefly, the system consists of two standard all glass ten gallon aquariums arranged one in front of the other with the long sides butted together. The front tank is divided into two sections by a fine mesh (100 micron) screen a quarter of the way from one end. The larger of these two sections serves as the actual rearing area. The rear tank contains an undergravel filter covered with dolomite chips to a depth of about three inches. The two tanks are joined together by a pump and a series of siphon tubes which allows water to flow continuously between them. Water is drawn from the smaller section of the front tank by siphons into the rear tank. There it undergoes biological filtration, removing ammonia and nitrite by the action of a well established nitrifying filter bed. Next, the water flows into an outside power filter which pumps it back into the rearing area, thus completing the circuit. The rear

tank then may be considered a large outside filter for the forward one.

The construction of this system is fairly simple, but some components warrant more details. First, the screen in the forward tank is made by joining strips of 1 1/2" by 1/4" clear acrylic plastic into a rectangular frame the height of the tank by 1/4" less than its inside width, using acrylic cement or acetone. This framework can be strengthened by attaching narrow braces on one side over each joint. The braces should be kept toward the inside edge to leave at least a 1/2" border of single plastic thickness along the outside of the frame. This is to allow the frame to slip easily into a holding bracket which is formed by two pieces of flexible 1/2" vinyl tubing with a single slit along the length. The slit is cut by inserting a 3/8" dowel rod with a straight line drawn longitudinally on it through the tubing and then carefully slicing down the tubing, following the line. For the bottom of the bracket, a strip of styrofoam is glued to the tank floor using silicone sealer. The two pieces of tubing then are fastened with silicone onto the side of the tank with the slits facing inward. To keep the tubing from curling while the sealer dries, a dowel rod is left in each tube until they are secure. The screening for the frame is a synthetic cloth used for making silk screen prints and generally is available at art supply stores. The screen is attached to the plastic by placing a piece of the cloth on the side of the frame without braces. Acrylic cement is applied where the screen contacts the plastic and pressure placed on this area until dry (about 30 seconds). When completed, the frame with screen slides into the vinyl tubing bracket until it seats against the styrofoam on the bottom. A small part of the plastic top rim of the tank may need to be cut away to allow the divider screen access to the holding bracket.

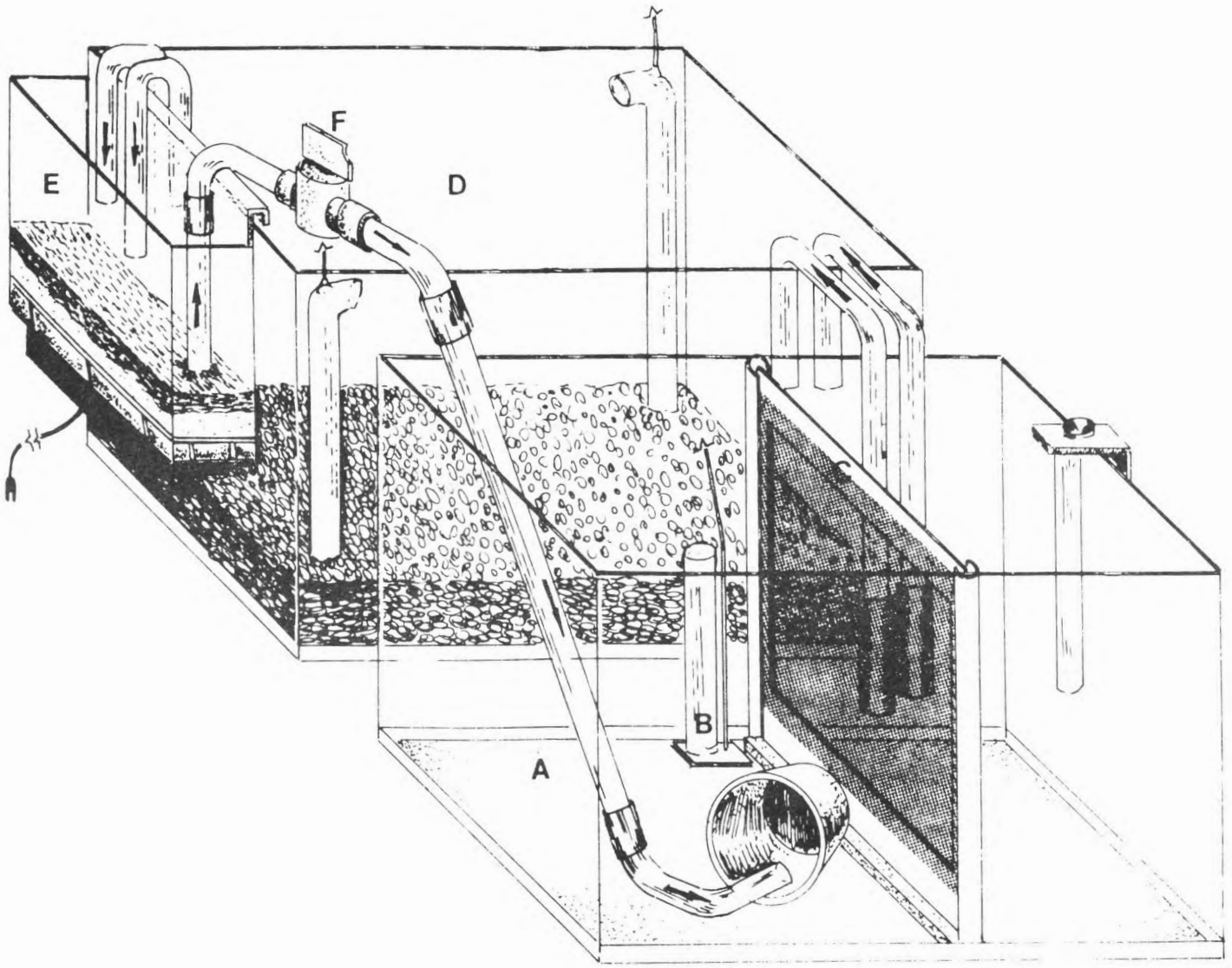
In operation, the relatively large surface area of the screen allows a rapid water flow through the system without any one point having a suction strong enough to draw in the fragile larvae. Having a partitioned area also makes it quite simple to change water, add a U.V. sterilizer, D.E. filter or any other device that requires the removal of water from the tank. When the screen becomes clogged with food, debris, algae, etc., a thin sheet of rigid plastic is placed in the bracket along side the screen which then can be removed and cleaned. The water flow should be turned off while this procedure is completed.

Another important component of the rearing system is the power filter and the tubing connecting it with the larvae tank. In our case, a Metaframe Dynaflow model 410 (the older version with

a larger return tube) is used, but any reliable type should do the job. By joining several pieces of curved and straight rigid plastic tubing together with flexible tubing as joints, the return flow of the filter can be routed to the bottom of the fry tank and directed to wash over the eggs, keeping them oxygenated and moving. This service usually is performed by a brooding parent in the wild but must be substituted for if the eggs are to be hatched away from parental care (Allen, 1972). A flow of air bubbles on the eggs also will work, but the advantage of this system is that a jet of water can be directed deep under a rock or other inaccessable areas where bubbles could form an air pocket on the egg mass. Usually during incubation the full force of the power filter can be used on the eggs. After hatching, however, the flow should be slowed down to keep from pushing the fry around the aquarium too rapidly. A valve inserted in the return tube can regulate the flow and thus give the aquarist more control over the water circulation speed. After spending considerable time searching for a suitable non-corrosive valve, an Eheim product model number 400-451 was found to work admirably well. To assist in circulation and to help break up surface tension, a light flow of large air bubbles is provided to the fry rearing area through a lift tube.

Lighting of the system is achieved by standard fixtures. Each tank is illuminated with a 15 watt DuroTest Vita-Lite fluorescent bulb positioned immediately above the water surface. The rear tank is lit constantly to promote algae growth with its resultant advantages (de Graff 1973). While the eggs are being incubated, the light cycle of the front tank is regulated by a timer to correspond to the photoperiod of the spawning tank. After hatching, constant illumination is provided which stimulates continuous feeding by the larvae and produces faster growth (Marliane 1976).

Despite remarkable advances recently in the field of captive marine fish breeding, a great deal more work must be done before aquarium reared fishes can be produced in sufficient numbers and species variety to provide stock for the growing marine aquarium trade. Hopefully, the system described here may prove useful to others involved in efforts to achieve this worthy goal.



Culture system in use at San Antonio Zoo Aquarium for rearing larval marine fish. A-rearing area, B-aerator, C-divider screen, D-filtration tank, E-outside power filter, F-flow control valve.

Arrows indicate water flow direction.

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HER UNDERWATER YARD HAS PLENTY OF ROOM FOR MARINE RESEARCH

(An article by Barry Schatz, reprinted from the Miami Herald, December 18, 1977.

Mary Lou Klay's underwater back yard stretches for miles north of her secluded home nestled in a hardwood hammock in Grassy Key. Part of the house doubles as a dormitory. Her back yard is a living marine laboratory.

Klay recently opened up the natural educational resources around her and calls it the Grassy Key Marine Study Center. It is the newest addition to a number of low-profile scientific centers in the Keys, including Key West's Florida Keys Marine Institute, Big Pine Key's Newfound Harbor Marine Institute, the University of Miami's Pigeon Key Environmental Research Station and Grassy Key's Institute for Delphinid Research.

While Klay operates her facilities as a private business, many of the others receive government funding and cater to such diverse clientele as children and doctorate candidates.

Her two-month-old idea takes its roots from 10 years of involvement in marine projects with her former husband Jerry Klay, a widely-respected marine science consultant and shark capturing expert. Since the property was once used to hold sharks and prepare them for live shipment to universities, aquariums, and oceanariums the world over, Klay says the new study center is naturally well-suited to shark research.

Two large above-ground cement aquariums contain nurse sharks tagged with iridescent orange letters. New College (Sarasota) experimental psychology student Dave Kramer monitors the sharks for psychological stress. He has conducted three weeks of independent study experiments at the center in preparation for writing his senior thesis.

Several offshore wire holding pens temporarily house larger sharks. A nearby building houses a basic laboratory setup where baby sharks, including one nurse shark born in captivity, are more closely monitored.

There Kramer and his two undergraduate assistants, Mark Martindale and Jeff Ayres, conduct blood tests and take tissue, organ, and gland samples for analysis. Live habitat observation is done in the open water with snorkle and fins.

"I'm using sharks as a model system that is representative of the lower vertebrates," says Kramer. He studies the sharks' less complicated psychological makeup to achieve more accurate results because, he says, "it doesn't have the confounding psychological implications that it does in humans."

Klay says her idea for the center is simply to make available the wealth of information in her underwater back yard and provide the bare facilities on land to conduct laboratory experiments and house a small group of researchers.

"It's for interested university students who are in marine biology and have a certain field project they want to work on. And they are on their own to produce." she says. "I don't monitor their work or give any evaluation to the university."

(Submitted by Daniel H. Moreno, Director
The Cleveland Aquarium

DEATHS OF AQUARIUM-HELD FISHES CAUSED BY MONOGENETIC TREMATODES. I. *ASPINATRIUM POGONIAE* (MacCallum, 1913) ON *POGONIAS CROMIS* (LINNAEUS).

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This is the first in a series of papers we plan concerning deaths of aquarium-held fishes caused by monogenetic trematodes. It is appropriate that this paper should cover the black drum (apropos this journal's name). Thus far we have had seven species of monogenetic trematodes contributing to, or causing, deaths of five species of fish in aquaria.

On August 31, 1977, the senior author examined a male black drum, *Pogonias cromis* (Linnaeus), which had died the previous day during an oyster predation study conducted by the second author at the GCRL oyster hatchery in Biloxi. The fish (93 cm total length; 42 pounds = 19 kg) was refrigerated overnight. It was caught in a trammel net on July 28, 1977 near Chevertte Point (30° 05' N; 89° 14' W) in the Louisiana marshes about 14 miles south of Pass Christian, Mississippi.

Although some fins were frayed, no external lesions were apparent. A section from the middle of the first right gill arch about 3 cm long was excised for examination under a dissecting microscope. From this small part of the complete branchial basket, 141 monogenetic trematodes over 1 mm long were recovered and mounted, and several hundred (estimated) worms less than 1 mm long were not saved. Some of the worms still were alive. Scrappings from the first and second dorsal fins revealed no worms. Scrappings around the isthmus and the left pectoral fin revealed four worms which apparently had come from the gills.

The worms were identified as *Aspinatrium pogoniae* (MacCallum, 1913) Yamaguti, 1963. Only two previous records for this parasite occur in the literature. MacCallum (1913) reported it from the gills of black drum obtained from either the N.Y. Aquarium, Woods Hole, Massachusetts, or a N.Y. fish market (no precise locality was given in his original report). Hargis (1956) reported it from the gills of black drum from Alligator Harbor, Florida. Our report from the Mississippi Gulf coast represents a new locality record.

The fish was held alone for experiments on oyster predation, being used primarily for obtaining daily and weekly feeding limits. It also was used in a clear water tank for behavioral observations

and photography of feeding habits. Experiments also were run to determine oyster size preference. The fish was held for daily tests and photography (usually 2-3 hours) in a 500 gallon recirculating-filtered tank system (filtered sea water, 19 ppt; tank sterilized with Chlorox and new water added prior to introduction of the present fish); longer tests were held in either a 500 or 1000 gallon flow-through system using Mississippi Sound water of varying salinity and no filtration.

Until the time of illness, the fish was consuming an average (based on seven days of tests) of 42 oysters (2-3 inches in length = 5-7.5 cm) per 24 hour period. It was fed only oysters (singles) during captivity. The fish's symptoms, in approximate order of occurrence, were unusual passiveness when transferred from tank to tank, picking up and rejecting oysters, no feeding on August 28, 1977 after a gradual reduction in feeding, and on the day before death a choking reaction, as if something was caught in the throat, was noticed.

When abnormal behavior first was noticed, the fish began to reject more oysters than usual; for example, usually it rejected one out of 12 oysters picked up, but when it started acting ill, it rejected 10 out of 12 oysters. At the time of death, the fish was involved in a weekly feeding test which began on August 26, 1977. Its oyster consumption was as follows: day 1, 28; day 2, 8; day 3, 0; day 4, 0; day 5, death.

The fish was held only about a month; however, either it had a sizeable monogenetic trematode burden when caught or there was a very rapid build-up of worms in one or more of the holding tanks. The small section of gill arch we examined had several hundred worms less than 1 mm long, which would indicate a recent massive reinfection of the fish. The maximum size of the worms recovered was 12.2 mm; one tangled egg mass found among the gill filaments contained 38 eggs, presumably from one worm. Unfortunately, the life cycle of this species of worm has not been described, therefore we cannot pinpoint the time that the latest massive infection occurred.

Hargis (1956) noted that the species needs to be redescribed; we agree, because a comparison of our specimens with the original description of MacCallum (1913) shows various details unclear or lacking.

MacCallum (1913) was aware of the possibility of this parasite being harmful to black drum. He said (p. 391), "The worm occurs...sometimes in such great numbers as to menace the life of the host." In the present case, where we estimate that the fish had many thousands of worms on its gills, we believe the

worms were the cause of death. Another major killer of fish in closed systems, the parasitic dinoflagellate Amyloodinium ocellatum (see Lawler, 1977), was not present on the gills of this fish.

At present we are holding different sizes of black drum in various sized closed system tanks in order to ascertain the approximate lengths of times it will take the worms to increase in a known volume of water to sufficient numbers to kill the different sizes of fish.

Although we have not tried to control this monogenetic trematode, one of the standard treatments, a 1:4000 formalin bath for 15 minutes to one hour, probably would work.

Acknowledgment

David E. Zwerner of the Virginia Institute of Marine Science kindly checked his files in order to verify all references to this parasite and supplied a copy of the original description. Steven L. Shepard provided some technical assistance.

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A PARTIALLY ALBINO BLUE CRAB

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This is our second report on blue crab abnormalities. Lawler and Van Engel (1973) reported on the triple regeneration of the fifth pereiopod of a blue crab from Virginia.

On June 13, 1977, the second author caught a blue crab, Callinectes sapidus Rathbun, in a trawl near Round Island, Mississippi Sound, Mississippi that exhibited partial albinism. It was a male measuring 116 mm in width with an almost totally white left cheliped. Dull red splotches were on the outside edges of the tips of the propodus and dactylus, and the most distal spine on the merus had a red tip. The right cheliped and the first right walking leg were missing. The coloration of the rest of the crab appeared normal.

The crab was placed in a 10-gallon aquarium and fed fish and shrimp scraps and artificial shrimp-fish food; it is still alive as of this date. On July 11, 1977, it molted and regenerated its two missing appendages, which were normal in color. After the molt the crab measured 140 mm in width. The left cheliped remained almost all white, with the following pigmented areas appearing with the molt:

(1) The reddish areas on the propodus and the dactylus were larger, and the red was more uniform at the tips.
(2) The proximal inside area of the carpus had bluish pigmentation.
(3) A bluish pigmented area appeared on top near the joint of the merus with the basis-ischium.
(4) Violet, red, and blue pigmentation appeared around the base of the most distal spine on the merus, primarily on the ventral side of the merus.

Since the molt, a pale blue pigmented area has developed on the top of the merus just opposite the distal spine on the merus, and the red areas on the propodus and the dactylus have enlarged. Thus, the chromatophores are expanding in area with time. Figure 1 illustrates the crab five months after the last molt.

We found four previous references on albinism in the blue crab. Rathbun (1930) reported a male from Texas with a white left claw, a wholly white crab from Maryland, and a female with a white right claw from Virginia. Newcombe (1945) mentioned an albino

crab from Virginia. Sims and Joyce (1965) reported a partially albino crab from Florida, and a partially albino crab was reported from near Deer Island, Mississippi (Marine Briefs, 1977). Our crab was caught prior to the latter record and was being held for observation.

In none of the previous references did we find any mention of the partially albino crabs being held alive through molts. Ours has molted once thus far, retaining most of its albinism. Unfortunately, the crab appears almost mature, so it may not molt again. If the crab had been small, we could have broken off the left cheliped at various joints, starting at the most distal, to see if regenerated parts came back as albino through several molts.

One of us (ARL) has been working in marine biology, plus crabbing and shrimping, since 1962, primarily in Virginia and Mississippi, and has seen only two crabs exhibiting albinism. The other (SLS), working on local waters since 1965, saw an additional crab with a white fifth pereiopod (paddle) in Gautier, Mississippi in 1965. Albinism in the blue crab is apparently rare, as the both of us have seen only a total of three in 15 years out of the many thousands caught.

Acknowledgment

Dr. R.M. Overstreet of GCRL took the photograph.

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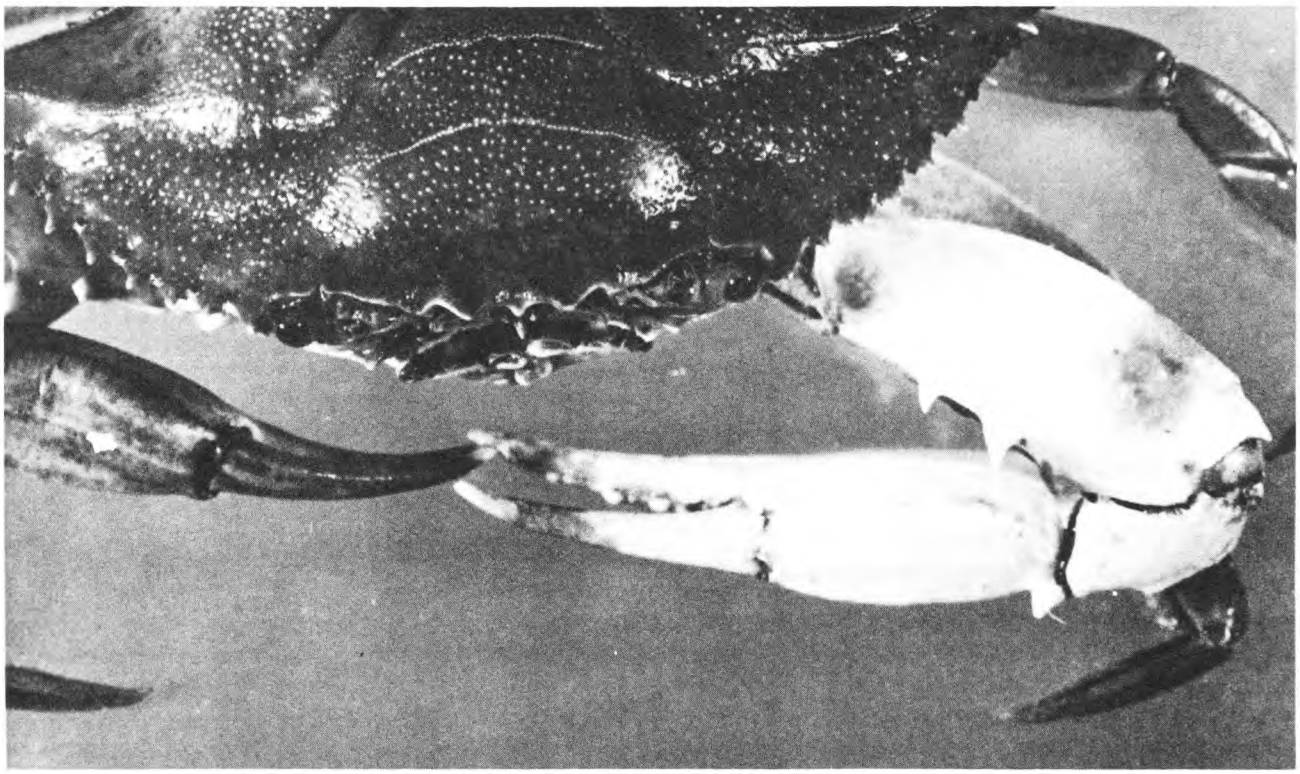

Figure 1. Partially albino blue crab. Photograph taken five months after the last molt.

NEEDLEFISH

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The needlefish or silver gar, Tylosurus marinus, always has interested me with its shape, color, and actions. As anyone who's ever collected this fish can tell you, they are very fragile and present some problems to those who would like to display them. This article will describe briefly some of those problems and how, from my point of view, they can be solved.

Collection is perhaps the most important stage in obtaining good, healthy specimens that will survive in the aquarium. This fish commonly is found along the Gulf Coast and South Atlantic seaboard of the United States, running into the mouths of rivers and sometimes past the tide mark.[1] They do appear along the southern shores and rivers of Cape Cod during the summer, sometimes abundantly. The summer of 1977 was such with great numbers being found in the Grand Cove area of Bass River. In the past we had not seen this many and had problems with those few that were gotten. We used a fifty foot beach seine, and by the time weeds and mummichogs were gotten through, the two or three needlefish on the bottom were badly bruised, scraped, and consequently died. This was not the road to success.

So at the risk of repeating the obvious, I will go into some detail about how we managed not to massacre the fish before they were in holding. They seemed to be the most plentiful and easiest to handle about two hours before full high tide. At this time the water was neither too shallow nor deep to prevent working on the inside of the seine. Previously we'd pulled the whole seine out of the water after making the tow. Now we'd make a tow of about fifty yards, work over to a bank in about two feet of water, and have one person get inside while the other stayed outside and behind. He'd pull the seine around the person inside. When the bag was reached the person inside pulled the bottom up, trapping the fish in a small, easily accessible area. Many of them could be removed by using a bucket while the rest who had gotten their beaks thoroughly tangled in the net could be removed by holding only the beak.

As I said earlier, this is a fragile fish. If the mucus layer is disturbed by netting them out of the water or handling, they last about twenty-four hours. Once they were in holding, our

1. Fishes of the Gulf of Maine, Bigelow and Schroeder, 1954.

success rate was about seventy percent. Feeding is no problem. They start on live silversides and progress rapidly to bits of fish or gelatin fish food skipped over the surface. They appear to have a high rate of metabolism and are fed on a daily basis.

These slender silvery fish look great on display; they stay out in the open and are quite active, zooming from here to there. Visitors seem to thoroughly enjoy the display and are surprised to know they appear locally. They are in a twelve hundred gallon tank with small black bass (Centropritis striata), tautog (Tautoga onitis), and cunner (Tautogolabrus adspersus) - a harmonious arrangement. However, small herring or silversides disappear rapidly, with the needlefish looking like the snake that swallowed the pig. One word of caution should be added here. These fish are jumpers, so some appropriate action should be taken to keep them in their tank and off the floor.

After being as careful as possible with collection, acclimation, and re-doing a tank just for this display, I still managed a massacre. Many of the black bass and cunner seemed to have gill flukes so the tank was dyloxed. A dose of .8 ppm was used in water with a pH of 7.7. This sounds like a lot of dylox but has been used in the past on both local and tropical fish with no problems. There was no problem this time with the black bass and cunner. All forty-eight needlefish, however, rolled over and died in very short order. The dylox was added in the afternoon, and the next morning about half the needlefish were dead and the others were on the way. Those still alive were removed to another tank and also died.

This happened in late September, so I was unable to collect anymore; they had left about two weeks before. Also, those in holding were running into trouble with leaking tanks and insufficient space. Suffice it to say, I now have six of the little critters who appear to be healthy and doing well.

DETERMINATION OF pH OF SEAWATER AND FRESHWATER WITH A SPECTROMETER

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Hydrogen ion concentration of water, usually referred to as pH, is commonly determined by a colorimetric method without instrumentation of any kind. This method involves a comparison of color intensities after a water sample has been treated with a specific dye. The color intensity of the treated sample will vary in direct proportion to the pH of the sample. The intensity of color that develops after the addition of the dye is then compared against a set of standards that represent carefully calculated pH values.

It is difficult to determine the intensity of color accurately with the unaided eye; consequently, this method of pH determination often gives unreliable results. However, variation in color intensities may be quite accurately determined electronically with the aid of a spectrometer.

The pH of seawater may range from about 7.4 to 8.3, and the dye - usually referred to as the indicator - that is used for the spectrometric determination of pH values within this range is cresol red. A cresol red indicator solution is made up by adding 100 mg of cresol red powder to 26.2 ml of a 0.01N sodium hydroxide solution. After the indicator has dissolved completely, the solution is diluted to 250 ml with boiled deionized water. The color of a water sample with a pH of 7.2, after a specified amount of cresol red indicator has been added to it, will be yellow with a pinkish tinge, whereas a water sample with a pH of 8.3 becomes red after the indicator has been added.

The pH of freshwater inhabited by fishes may range from 5.8 to 7.6. Bromothymol blue is a satisfactory indicator for producing color intensities for the spectrometric determination of water with pH values within this range. Bromothymol blue indicator solution is prepared by dissolving 100 mg of bromothymol crystals in 16 ml of 0.01N sodium hydroxide and adding 234 ml of boiled deionized water to produce a total volume of 250 ml. Both bromothymol blue and cresol red indicator solutions should be stored in dark bottles away from light. Colors produced after the addition of bromothymol blue to freshwater samples with pH values within this range vary from yellow to

dark blue.

In order to determine the pH of a water sample with an unknown pH, a small portion of it is treated with a specified amount of indicator solution, the color intensity is read in the spectrometer, and its pH value is determined from a graph. The following procedure to produce pH values of a number of saltwater samples for the construction of a seawater reference graph for this study is as follows: Various predetermined amounts of 0.1 molar sodium hydroxide (NaOH), depending on the pH value required, are added to a number of 50 ml portions of 0.1 molar potassium dihydrogen phosphate (KH_2PO_4) and 3 grams of sodium chloride (NaCl) are added to each of the prepared samples. Sodium chloride is added so as to approximate the amount of sodium chloride contained in normal seawater. The sodium hydroxide and potassium dihydrogen phosphate mixtures produce pH values from 7.2 to 7.8 (table 1). Values from 8.0 to 8.6 are produced by the addition of varying amounts of 0.1 molar hydrochloric acid (HCL) to 50 ml portions of 0.025 molar sodium borate ($Na_2B_4O_7 \cdot 10\ H_2O$) solutions (table 2). After the addition of the various solutions to produce the required pH values, each sample is checked with a pH meter, and more acid or base is added as needed to produce the desired pH values for the points on the graph. The samples are all increased to 100 ml with boiled deionized water after the pH adjustment has been made. After the pH of each prepared saltwater sample is carefully checked by the pH meter, 20 ml of each sample is transferred to a small vial, and exactly 0.3 ml of cresol red indicator solution is added to it. A little of the colored sample is then transferred to a spectrometer tube and read in the spectrometer at a wave length of 565 millimicrons. The reading is recorded from the absorbance scale, and the value is plotted on a graph. Eight saltwater samples with pH values ranging from 7.2 to 8.6 were treated with the indicator, read in a Bausch and Lomb spectrometer 20, and their pH values were recorded on the graph for this study. This provided the required number of points for the seawater reference graph (fig. 1).

The procedure for the construction of the freshwater graph was the same as that for the seawater graph except that 0.5 ml of bromothymol blue indicator was added to 10 ml water samples to obtain the various spectrometer readings, and a wave length of 575 instead of 565 millimicrons on the spectrometer absorbance scale was used to detect the various color intensities of the treated water samples. No sodium chloride was added to these samples. This wave length proved quite satisfactory for the detection of color intensities representing pH values from 5.8 to 7.6 (table 3 and fig. 2).

The amount of indicator added to each water sample in preparation for the spectrometer test is very important. A slight variation in the amount will produce a substantial error in the pH reading. Unfortunately all eye droppers do not produce the same amount of liquid per drop. If eye droppers instead of small graduated pipets are used to dispense the indicators for routine pH tests, it is imperative that they dispense the amount of indicator equivalent to the amount used to determine the points on the graphs included in this study.

This method is convenient, fast, and accurate for pH testing of both seawater and freshwater. I have used it at Steinhart Aquarium for several years and have found it to be a very satisfactory method for performing pH tests. Testing the pH of seawater with a pH meter may become quite time consuming and often very exasperating unless an especially good pH meter with a sodium electrode is available.

Table 1

Various amounts of sodium hydroxide (NaOH 0.1 molar) combined with 50ml portions of potassium dihydrogen phosphate (KH_2PO_4 0.1 molar) to produce pH values of 7.2 to 7.8 in saltwater. Three grams of sodium chloride (NaCl) is added to each 100ml sample, and cresol red is used as the indicator to produce the spectrometer readings.

pH	KH_2PO_4 0.1M	NaOH 0.1M	H_2O	NaCl	Spectrometer reading
7.2	50ml	34.7ml	15.3ml	3g	0.18
7.4	50ml	39.1ml	10.9ml	3g	0.27
7.6	50ml	42.8ml	7.2ml	3g	0.39
7.8	50ml	45.3ml	4.7ml	3g	0.45

Table 2

Various amounts of hydrochloric acid (HCL 0.1 molar) combined with 50ml portions of sodium borate ($Na_2B_4O_7 \cdot 10\ H_2O$ 0.025 molar) to produce pH values of 8.0 to 8.6 in saltwater. Three grams of sodium chloride (NaCl) is added to each 100ml of sample. Cresol red is used as the indicator to produce the spectrometer readings.

pH	$Na_2B_4O_7.10\ H_2O$ 0.025 M	HCl 0.1M	H_2O	NaCl	Spectrometer reading
8.0	50ml	20.5ml	29.5ml	3g	0.52
8.2	50ml	18.8ml	31.2ml	3g	0.60
8.4	50ml	16.6ml	33.4ml	3g	0.64
8.6	50ml	13.5ml	36.5ml	3g	0.70

Table 3

Various amounts of sodium hydroxide (NaOH 0.1 molar) combined with 50ml portions of potassium dihydrogen phosphate (KH_2PO_4 0.1 molar) to produce pH values of 5.8 to 7.6 in fresh water. Bromothymol blue is used as the indicator to produce the spectrometer readings.

pH	KH_2PO_4 0.1M	NaOH 0.1M	H_2O	Spectrometer reading
5.8	50ml	3.6ml	46.4ml	0.03
6.00	50ml	5.6ml	44.4ml	0.06
6.40	50ml	11.6ml	38.4ml	0.13
6.80	50ml	22.4ml	27.6ml	0.24
7.00	50ml	29.1ml	20.9ml	0.29
7.40	50ml	39.1ml	10.9ml	0.45
7.60	50ml	42.8ml	7.2ml	0.50

Figure 1. Graph for determining pH of sea water, using a 0.04% solution of cresol red as the indicator and a wave length of 565 millimicrons on the spectrometer absorbance scale.

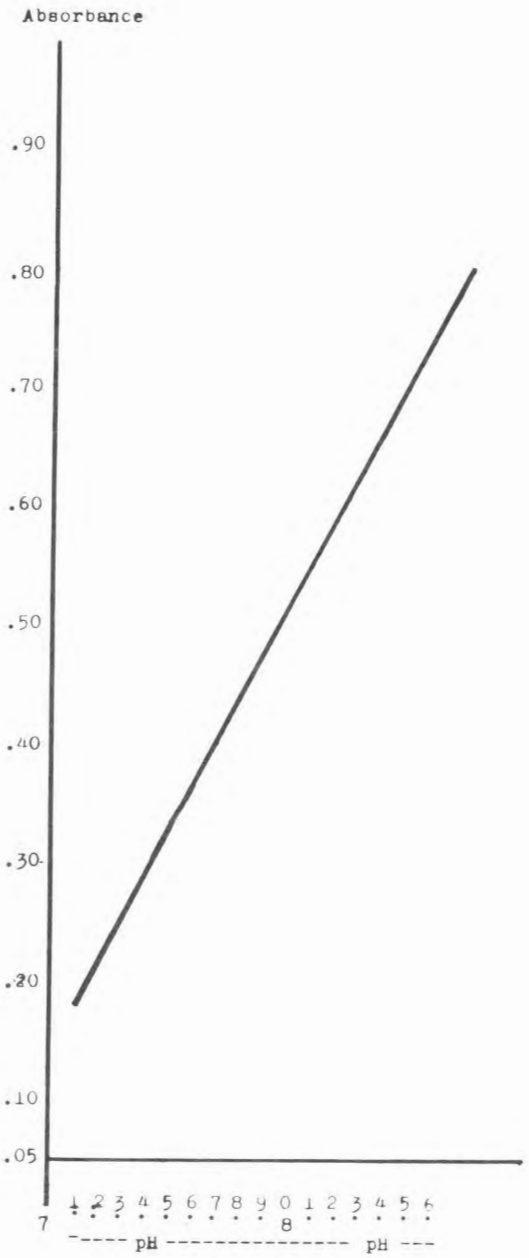
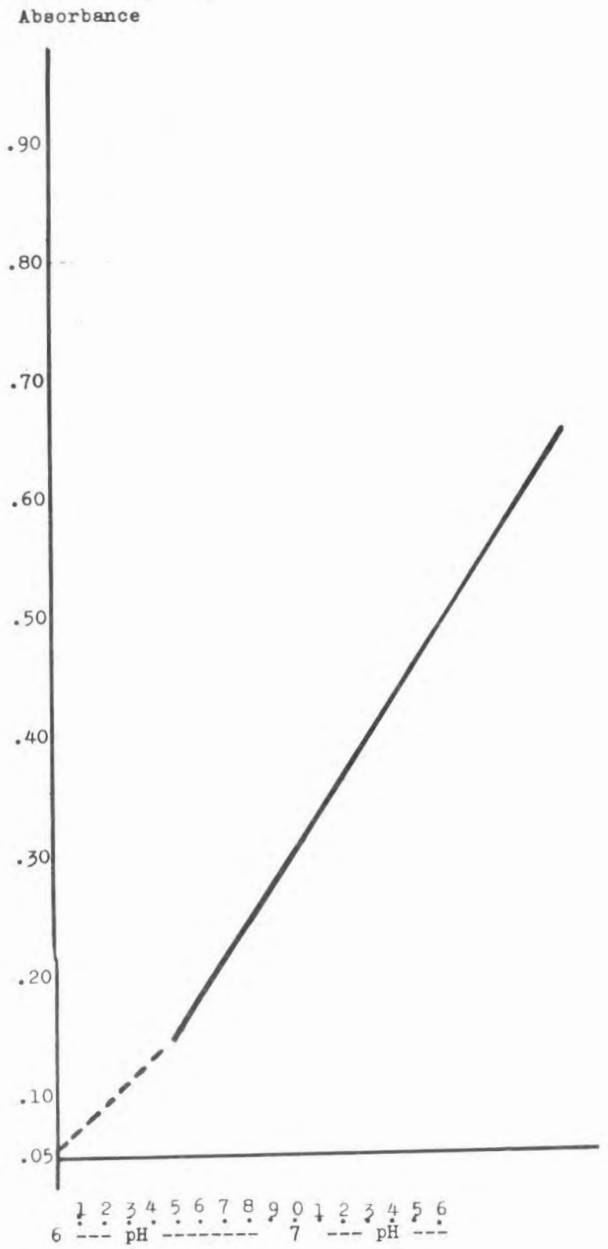


Figure 2. Graph for determining pH of freshwater, using a 0.04% solution of bromothymol blue as the indicator and a wave length of 575 millimicrons on the spectrometer absorbance scale.



JAPANESE AQUARIUM TOUR

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The number of Japanese aquaria has increased since the idea of a Japanese aquarium tour was proposed by Earl Herald[1] seven years ago. The 1974-75 edition of the "Directory of the Public Aquaria of the World" lists over 80 aquaria, many of them built since 1970.

The large number of aquaria presents the problem of how to see as much as possible in a small period of time. This tour would include 14 aquaria in eighteen days. In addition, some free time would be available throughout the tour.

The tour would leave Los Angeles, arriving in Tokyo. Travel between cities on the islands of Honshu, Shikoku and Kyushu will be primarily by train, between islands by Inland Sea Liner with first class accommodations.

The tentative list of aquaria is: Ueno Zoo Aquarium, Yomiuri-land Marine Aquarium, Aburatsubo Marine Park Aquarium, Tokai University Sea Science Museum, Shima Marineland, Kushimoto Sea Park Center, Misaki Park Aquarium, Yashima Mt. Aquarium, Ashizuri Submarine Palace, Sukumo Sea Museum, Oita Ecological Aquarium.

The price for this tour will be approximately $2,000 which includes airfare, ground transportation, lodging & a guide-interpreter. Two nights lodging will be at Japanese inns, and meals will be provided for these two days.

This price may seem high, but every effort is being made to keep the costs down. For example, utilizing a charter airlines specializing in the Orient will save $230 over the group air-fare of a scheduled airlines. At this point in time, negotia-tions are still in progress in an effort to obtain lower rates for the tour.

The dates for the trip will be from October 1-18, 1978.

For specific information, please write to: Charles Farwell, Scripps Aquarium A-007, La Jolla, California 92093.

1 "Drum and Croaker", Vol. 12(71); No. 1, Jan. 71.

ATTENTION!

A Reminder - Members of the AAZPA get ready to submit your Edward H. Bean Award Nominations.

The Edward H. Bean Awards are the highest awards in the zoological profession and cite individual zoological efforts in the captive management and husbandry of mammals, birds, reptiles, amphibeans, fish and invertebrates.

The significance of these awards and the accomplishments they represent should not be held lightly, for they are physical proof that the receiving institutuions are conservation and education oriented.

The Honors and Awards Committee must rely on the membership of the American Association of Zoological Parks and Aquariums to submit nominations, and it is the Committee's collective expertise that serves as a clearinghouse for these nominations. The Committee feels that many institutions do not submit their significant births or hatchings for consideration because they judge their accomplishments not significant enough. The Committee wants to receive all noteworthy nominations and, therefore, invites and urges any AAZPA member to submit entries. We also would like to stress that the award is for the "most significant birth" and is not necessarily a first breeding award.

Nominations received for consideration will be scrutinized by the various committee members in their specialty areas. During this review, every attempt is made to keep the nominees anonymous. The forms do not have any institutional identification at the time of review by the specialist committee members. The membership of the Committee has been increased so that nominations in each animal category will be reviewed by three specialists in that area.

Nomination forms, which soon will be distributed in the mail, are to be filled out and returned no later than June 1, 1978. The Committee solicits any additional information from the nominating member in the form of graphs, charts, breeding histories, behavioral observations and nutritional needs of the species nominated. All supplementary information provided will greatly assist the specialists in their deliberations.

Please submit a separate form for each nomination. Additional forms may be requested from the Committee Chairman or may be reproduced locally.

Nominations received with incomplete information will be returned to the submitting member for clarification.

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