DRUM *and* CROAKER

A Highly Irregular Journal for the Public Aquarist

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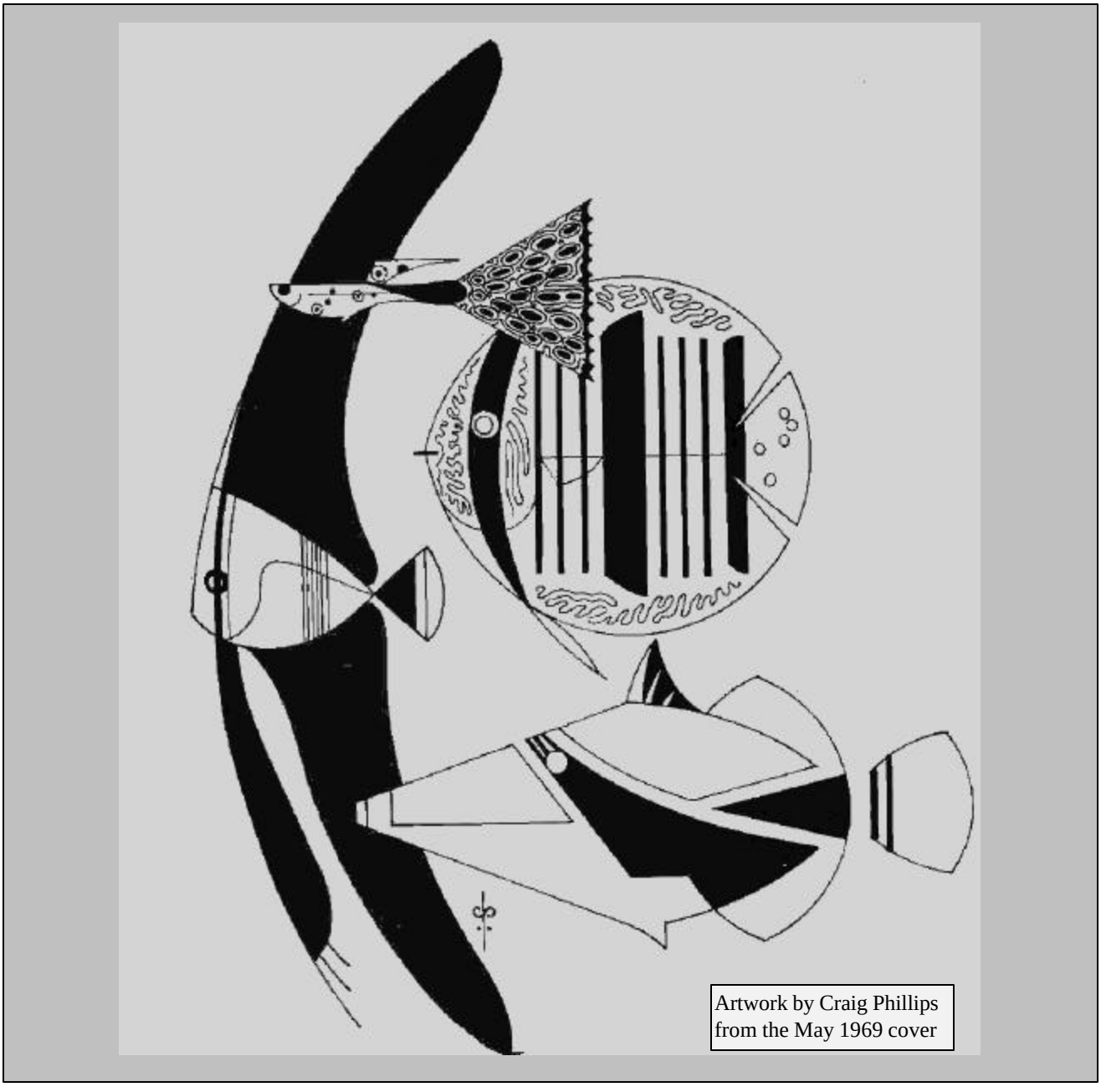

Artwork by Craig Phillips
from the May 1969 cover

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DRUM AND CROAKER 30 YEARS AGO

Richard M. Segedi

From the May and September 1970 issues, published and edited at the National Fisheries Center and Aquarium, Washington, D.C.

Washington, D. C.: *Professional Aquarium Symposium, Ichs and Herps Meeting - New Orleans* by Lou Garibaldi

The 16th Annual Aquarium Symposium at the fiftieth Annual Meeting of the American Society of Ichthyologists and Herpetologists, was held in New Orleans, Louisiana, March 26-30. The discussion dwelt mainly on the future of the aquarium symposium. Should professional aquarists continue to meet in conjunction with the ASIH or should they meet with the American Association of Zoological Parks and Aquariums, or the Ecological Society, or should they form an association of their own and meet whenever they felt it would be convenient? Many felt that there was need of some change — that the poor stepdaughter role should end and we should have more to say as to how, when and where we meet and the scheduling of events.

Quebec City, Quebec, Canada: *The Quebec Aquarium* by Karl-Heinz Krumke

The Aquarium has expanded considerably in the last ten years, presenting more species and exhibits, and improving upon our technical installations. We began with ten tanks of 70 gallons each and ten of 10 gallons. These were all placed, together with their equipment, in specially built floor displays. All of these aquaria were made of plexiglass in our own workshop. This material eliminates the troubles one has with saltwater exhibits in metal—framed aquaria. Many of these have been in use for more than seven years. A 3/16' replaceable plastic sheet protects the plexiglass from scratches by visitors.

Vancouver B. C., Canada: *Biology Laboratory at the Vancouver Public Aquarium* by Sharon Proctor

The Vancouver Public Aquarium, in addition to a guided tour program, presents a laboratory session for grade 11 biology students. The laboratory has bench space for 30 students, and along one wall are large pans, filled with constantly flowing seawater. The students are provided with live marine animals, instruments, and two Spencer A-O binocular dissecting microscopes. There is also a sink with tap water.

San Francisco, California: *Sodium Chlorite for Water Clarity in the Marine Dolphin System* by Robert P. Dempster

At the Steinhart Aquarium sodium chlorite is currently being employed as an aid to maintaining clear water in the marine dolphin system. The chlorite is maintained at concentrations of from 0.5 to 2.5 ppm. A simplified test for the presence of this chemical in sea water has been developed by Anderson and Dempster through a modification of the Black and Whittle procedure for determining the presence of total chlorine in fresh water. Maintaining concentrations of 0.5 to 2.5 ppm has had no apparent adverse effect on the dolphins, seal, and two cormorants in this water system. The chlorite maintains a clarity in the dolphin tank that has never before been equaled at Steinhart Aquarium.

FISHING AT THE SPEED OF LIGHT

Scott DePalma, Aquarist

Belle Isle Aquarium

During the war with Iraq, Coalition air forces used laser guided bombs to destroy targets with pinpoint accuracy. By doing so, they avoided dropping hundreds of bombs on target areas before destroying their objective. This was often done with pairs of aircraft. One would designate a target with a laser beam, keeping the target in sight at all times. Another aircraft would swoop in and drop a bomb, which homes in on the laser light, and quickly and efficiently destroy the target. What in the world does this have to do with the nation's oldest municipal aquarium? Well, we are currently using this cutting edge technology for a similar mission at the Belle Isle Aquarium.

One of our projects at BIA is the Lake Victoria Species Survival Plan. We have numerous holding and rearing tanks behind the scenes, as well as several 350 gallon exhibit tanks. Since space is at a premium in most aquariums, we attempt to use display tanks as breeding tanks if at all possible. To do this successfully, we must be sure that all of the fish in the display tank are of one species and one generation, and we must remove cichlid offspring while still being held my the female cichlids.

If you are familiar with Lake Victoria cichlids, you probably realize that the females of most haplochromines look almost identical, regardless of the species. This is especially true when they are viewed from above. When trying to single out a brooding cichlid from a tank full of virtually identical non-brooding females, an aquarist soon becomes overwhelmed with small flashing silver fish, any one of which could be the target. Since you can't see the female's distended throat from above, it is all the more difficult. We could catch all of the fish, but this is very time consuming and stress inducing (for both fish and aquarist). Also, a female holding young will often eject them if she feels too threatened, with subsequent loss of most or all of the fry. We could anesthetize the tank, but this can sometimes cause unacceptable losses for the fry. We have come up with a novel way to eliminate these problems.

Our solution involves a two person team, a designator and a "bomber". One aquarist stands on the public floor with a small laser pointer. Using the laser pointer, the aquarist designates the mouthbrooding female with the bright laser light. Another aquarist is behind the scenes, standing over the tank with a net. What was formerly an indistinguishable silver fish is now carrying a bright red spot of light on it's flank, easily visible from above. As long as the designator keeps the laser on the right fish, the "bomber" simply has to home in on the target with the net. This process usually takes about five minutes for the average catch. Previously, this was a major undertaking involving thirty to sixty minutes of fish chasing, often with no result. In addition to being loads of fun, this method also greatly reduces stress on the fish population, which in turn encourages more reproduction. The old method meant that the tank would take a day or two to return to normal activity. Now, it is back to normal in minutes.

Using this technique, we have been able to successfully remove fry from several of our LVSSP species. Previous to the implementation of this method, all fry production resulted from smaller breeding groups in small holding tanks. A larger tank with more individuals produces far better results.

As with all new techniques, there are positive and negative aspects to this method. As previously mentioned, the pluses include reduced catch-induced stress, higher fry production, increased breeding activity, and the capability to use exhibit tanks as breeding space. There are some negatives

which may apply to other aquariums. First, our LVSSP display tanks are manageably small for catching fish by net (they can only run so far!). Second, our tanks housing Victorians are rather sparsely decorated, mostly with small granite fieldstones or similar sized rocks in small piles. This makes it difficult for large numbers of fish to hide in massive rockwork. Clearly, if your tanks are five times this size, filled with plants, wood, and massive stone outcrops, the fish will be much more likely to find refuge before you catch them. Also, we try to keep only one, or two vastly different species in a tank, to avoid hybridizing. For instance, one of our tanks features *Yssichromis argens* and *Oreochromis variabilis*. We have never observed hybridizing between these two genus', so we are able to strip young from both species. Also, it is very important to maintain a single generation in the tanks. Luckily, due to the piscivorous nature of the fishes involved, as well the relative lack of fry refugia, any fry that we don't strip are quickly eaten by the tank inhabitants. Finally, try to keep from shining the laser into the eye of the fish, as extended exposure could cause eye damage.

If you have similar tanks for your mouth-brooders, you might want to give this method a try. We may be the oldest aquarium, but we still manage to use cutting-edge technology for innovative new techniques!

AZA 2001 EASTERN WORKSHOP
TENNESSEE AQUARIUM, CHATTANOOGA, APRIL 4-7, 2001

Formerly referred to as a "regional conference", this meeting will use an interactive workshop format rather than paper presentations, and is not intended simply for eastern AZA members. A variety of special meetings will be held, as will interesting post-conference trips to local natural areas and the new Ripley's Aquarium of the Smokies. Three workshops to address husbandry and conservation needs at Public Aquariums are being prepared:

- **Management of metazoan parasites of fish.**

  Covers life history, drug therapy, treatment protocols, etc. This will be an intensive full day ordeal led by Dr. George Benz and others.

- **Creating Beneficial Conservation efforts for native aquatic fauna.**

  Chris Coco (Chattanooga) and professionals from other facilities will report on partnerships, refuge population issues, and case histories of various projects.

- **Managing *Mycobacterium* in closed systems**.

  Mycobacteria infections are common in Victorian cichlids, pupfish, and other captive populations of endangered fishes. This workshop will describe the disease, discuss management techniques, and provide hands-on training of diagnostic techniques.

Information: Visit <http://www.tnaqua.org/aza.html>. The November and January issues of AZA's Communiqué will include a registration form and related Workshop information. Contact Chris Coco, Curator of Fishes, directly at: 423-785-4069 (Phone), or 423-267-3561 (Fax). E-mail: e-mail: csc@tennis.org <mailto:csc@tennis.org>

THE TRANSPORTATION OF LIVE SILKY SHARKS (*Carcharhinus falciformis*) ON STANDARD PALLETIZED AIRCRAFT CARGO POSITIONS FOR LONG DISTANCE AND TIME PERIOD TRANSPORTS

Forrest A. Young, Director, Dynasty Marine Associates, Inc.

David C. Powell, Curator, Emeritus, Monterey Bay Aquarium

Richard Lerner, Curator of Fishes, Ocean Journey, Denver CO.

The silky shark, *Carcharhinus falciformis*, is a species that has only recently been displayed in public aquariums, and to date, little information has been compiled on its behavior and husbandry requirements. As far as the authors are aware, this species has only been collected and displayed by Steve Kaiser, Dave Wert and Glenn Kelly at Atlantis Resorts, Paradise Island, Bahamas, Jerry Crow at Sealife Park, Hawaii and Kiyonori Nishida at the Osaka Aquarium Kaiyukan. Atlantis' and Sealife Park's proximity to the open sea obviously minimizes the transportation difficulties that most aquariums face in acquisition and acclimation of display animals. The transport to Osaka was by sea and truck and was relatively short in its time period. Nonetheless, the work done at the three institutions with pelagic sharks has been quite impressive.

The silky shark is quite similar to the blacknose shark, *C. acronotus'* capability to survive brief capture stress and brief transportation times to the quarantine system. In total, about 60 to 90 minutes represents the maximum time from capture (by rod and reel) to introduction into a 7m by 20m by 1.5m holding tank where mostly all animals that are captured, will survive. The author suspects that longline capture is even more stressful as it greatly increases the "in water stress" time period. Once the silky shark is acclimated to captivity it seems to be a hardy and visually impressive husbandry candidate.

Until the present time, they have not ever been transported over any significant distance or transported in an aircraft (David C. Powell, pers. com). It was the hypothesis of the authors, that the methods that we have developed at Dynasty Marine Associates, Inc., that considers the basic spatial and dynamic requirements of similar species such as *C. acronotus* and the more challenging blacktip shark, *C. limbatus*, would also work for *C. falciformis*.

This approach was developed during experimentation that was initiated in the desire to provide captive specimens of small pelagic sharks, especially *Carcharhinus limbatus*, *C. acronotus* and bonnethead shark, *Sphyrna tiburo*, and the scalloped hammerhead, *Sphyrna lewini* to public aquariums. Small Caribbean carcharhinids, specifically *C. limbatus* are extremely sensitive, obligate ram-ventilators and are difficult to collect and transport. On board the collecting vessel, maintenance of totally pelagic, obligate ram-ventilators requires highly specialized methods and holding containers.

All totally pelagic shark species have a great degree of difficulty recognizing and negotiating barriers in their swim path and the resultant stress levels caused by repeated container contact is an extreme major obstacle to their adaptation to immediate captivity. The energy budget required to negotiate these tight turns in rectangular boxes very quickly depletes the energy reserves of the shark,

especially when the animal is already arriving on board the boat in a highly stressed condition from being caught on a baited hook, fought on the tackle and landed at the boat. The staff at Dynasty Marine Associates has for some time been developing and modifying a large, round holding tank system (hammer paper in press) that takes into account the initial difficulties for these species. The primary obstacle is that the size of the container is limited to the size and carrying capacity of the boat.

The results that have been achieved bear out the merit of the hypothesis. Collections of two 1m *C. falciformis* were made on two separate collecting trips and they were acclimated in the holding system for about three weeks. During this time, the sharks were fed daily and ate ravenously on squid and cut mackerel at about 2% of total body weight/ day.

Once the sharks were adequately acclimated, food was withheld for four days and the authors shipped them via truck to Orlando and from there by cargo aircraft to Denver. The transport containers that were previously developed for transporting hammerheads were used (hammer paper in press) and one 1m shark was shipped in each of two 2.5 m diameter containers. Total transportation time from holding tank to aquarium was about 26 hours. Both sharks swam in the containers during the entire trip and ate the day after the transport.

One shark developed a slightly cloudy eye, presumably from contact with the transport container in route. An oral Baytril regimen was administered and this cleared up the injured eye within 10 days. Slight rostral damages, also presumably from perhaps either capture injury and or container contact were also observed and these healed completely within 30 days from the time of transport. This is not unusual in small pelagic carcharhinids or sphyrnids.

The ideal temperature for the species seems to be above 25°C. and below 28 °C. There were several low pressure weather systems that occurred during the holding and acclimation period and the resultant cool water in the semi-open system with temperatures below 23° significantly slowed swimming speeds and overall activity levels. Feeding was also sporadic below 23°C. and did not occur much at all below 21°C. The senior author has since observed feeding in *C. falciformis* twice in about ten observations at about 20°C. It is interesting to also note that the senior author has noted several mortalities in *C. acronotus* below 18 and 19°C. that for lack of any other factor that could be found have been attributable to the cold temperatures. In the sea, these species are never found in water below about 22 or 23°C.

The short term conclusions indicate that medium length transports of duration's of 24 to 36 hours present slight risk of mortality the principal risk is of equipment failure, transport mishandling by the airlines or flight cancellations. It is also the authors' conclusions that transport durations up to as much as 48 hours are possible with only slightly elevated risk. Also, the transport of two sharks per container is quite likely to represent a minimal risk on shorter duration transports of 24 hours or so transports.

The senior author recently supervised a 38 hour transport of two, one meter sized *C. acronotus* with no mortalities, a thirty hour transport of three *C. acronotus,* where one mortality was

observed out of six animals transported and up to six, 50cm to 60cm *C. limbatus*, for transports up to 56 hours with a high degree of success. The risk of stress and mortality is always minimized if fewer animals are transported in the same container and is always recommended.

It is the authors' conclusion that many other species of small and medium size, totally pelagic sharks can be collected and transported by these methods and that their husbandry will be researched in detail during the next few years. Up until this time, very few species of visually impressive sharks were available for aquariums to display. These have been primarily the brown shark, *Carcharhinus plumbeus* and the sand tiger shark, *Odontaspis taurus*. With the increase of availability and interest in other new and sometimes smaller, pelagic species, new displays are sure to follow, especially those that more easily accommodate the requirements of the smaller species.

2001 REGIONAL AQUATICS WORKSHOP (RAW)
JUNE 4-7*, ATLANTIS RESORTS, NASSAU, BAHAMAS

What is RAW? RAW is an independent, loosely organized group of public aquarium professionals that meets annually to discuss issues important to our field. It is not affiliated with AZA or any other organization and has no formal structure beyond what is provided by the host facility. Many long-time RAW participants also happen to be active in AZA. Therefore AZA aquatic TAG, SSP and CAP meetings are usually conveniently held in conjunction with RAW.

The "region" encompassed by this annual workshop has expanded once again and this coming June we will participate in the first internationally-hosted RAW. The host facility, **Atlantis Resorts** in the Bahamas, is just a short shuttle flight from Florida. Atlantis has one of the largest Aquarium complexes in the world, has routinely displayed tiger sharks, and continues to break new ground with the husbandry of unusual elasmobranchs such as manta rays and great hammerhead sharks. Field trips to nearby reefs are planned. Accommodations are being provided at a significant discount over the normal rates and costs should be similar to what might be encountered in a large US city.

***RAW presentations and workshop meetings will be held on June 4-7, but arrival on the 3rd is suggested. AZA TAGs will meet on the 2nd and 3rd (arrival on the 1st suggested), while post-conference field trips are being organized for the 8th.**

***NOTE:* Participants must carry *either* a passport *or both* an official copy of their birth certificate *and* a photo ID (drivers license) in order to enter the Bahamas.**

For more information contact Atlantis' curator, Michelle Liu at (242) 363-2000 ext.65482# , or michelle.liu@sunint.com

OCTOPUS ENRICHMENT TECHNIQUES

Mark J Rehling, Aquarist.

Cleveland Metroparks Zoo.

The application of animal enrichment has proven itself to be beneficial to captive husbandry for many years. So much so that certain techniques have become standard practice for many species. Recent trends towards realistic habitats combined with other practices allow the captive animals to show their "true colors", often-exhibiting behaviors that make many unique (Shepherdson, Mellen, & Hutchins, 1998). The application of enrichment becomes exceedingly more important when the exhibit is limited due to husbandry needs. Intelligent animals such as primates, pachyderms, or cetaceans easily fall into this category. Training serves to provide much stimulation but a host of other techniques are also applied. For most of the non–mammalian aquatic animals enrichment is often limited to forms of aquascaping or the presentation of live foods. Of these aquatic forms, the octopus expresses an obvious need for varied forms of enrichment.

The octopus in the wild is a reclusive but proficient nocturnal hunter using its unique make up to its full means in various techniques of prey acquisition. It is an animal of surprising intelligence, capable of acts of amazing dexterity (Hanlon & Messenger, 1996). The wild octopus is presented with a multitude of challenges while hunting. Crustaceans, the primary prey of the octopus, can dart into small holes and crevices to avoid capture. Food, at times, is captured by touch alone (Hanlon & Messenger, 1996). Unfortunately, the ideal setting for both species and husbandry can result in a rather dim and Spartan exhibit. This can cause an interesting but cryptic Octopus to easily be overlooked or passed by. It also has to be fairly devoid of stimulation for an intelligent predator. Similar situations with vertebrate animals have been addressed with different forms of enrichment, but it seems surprisingly little is done with many Octopuses.

A recent revamping of the Octopus exhibit at the Cleveland Metroparks Zoo seemed to magnify this point. Great pains were taken (figurative and literal) to recreate an accurate image of the Northern Pacific coast for a specimen of *Octopus dolfleini*. A specimen was acquired and the exhibit was officially opened. The levels of interest in the exhibit were limited to feedings and occasional glimpses of activity. Having seen the activity levels of terrestrial animals increased with some kind of stimulation, enrichment seemed the logical solution. However, with the little information available, it seemed that we would need to create our own forms. This proved challenging.

The focus of our efforts was centered on investigative hunting documented in the wild. With exposure to the marine exhibit, the items had to be durable and easy to retrieve. The devices had to be challenging but solvable and expense was to be kept at a minimum. Ease of construction and ease of preparation were also strong considerations.

The result was the construction of various devices later called “prey puzzles”. Each had size, shape and challenge to provide as wide a range of stimulation as possible. They were often constructed of spare filter parts; pieces of plumbing and other assorted odds and ends that seem to congregate around aquarists. These puzzles were named for ease of recognition on daily keeper reports. With repeated presentations notes were made on acceptance levels, construction, and ease of use. The following is a compilation of those notes and other observations.

The Jar

The first item presented was a large plastic pretzel jar. The jar was introduced open and empty. The new object was observed but not touched. After a 15 minute period, the jar was removed. Two hours later the jar was reintroduced with a crab placed inside. This time the jar was investigated, and the Octopus removed the prey. After the prey was accepted, the jar was promptly removed. Food was offered in this fashion, and accepted, for two more feedings. A lid was added to increase the difficulty. Placed loosely at first, the lid was slowly screwed down with every introduction until the octopus was able to open the closed jar. The lid was later attached with monofilament for ease of recovery.

Intended Challenge:

Introduction of Foreign Object - Manipulation of Obstacle

Notes: (Poor …1…2…3…4…5… Good)

- Acceptance level. 4
- Ease of Construction. 5
- Cost. 5
- Durability. 4
- Ease of Retrieval. 4
- Ease of Preparation. 5
- Capable of presenting multiple sizes of food. . . 5

Observations:

The jar was the simplest of the devices and always the first to be introduced. This introduction seemed to establish that new objects could contain rewards, but needed to be manipulated some way. Later holes were drilled in the body of the jar to aid in recovery. This puzzle was introduced to a newly arrived specimen that was substantially larger and surmised older than the previous and required no such introduction. Approximate time for solution by the elder, 20 seconds.

The Hamster Ball

The “hamster ball” was a hollow plastic sphere with a locking lid that was adapted to push in with pressure. Elastic bands held the lid in place and replaced it when the puzzle was solved. Food could be accessed by locating the weak point on the surface of the sphere. Once the opening was

found, the lid had to be held open while the food was obtained. For introduction, the sphere was weighted down, but later the weight was removed and the sphere was allowed to float
freely throughout the exhibit. Food placed inside counteracted the sphere's positive buoyancy allowing it to be neutrally buoyant or "hover" in the water column. The normal currents present carried the sphere repeatedly passed the octopus's den requiring it to give chase. Once the food was removed and the sphere released, it could be easily retrieved as it floated to the surface.

Intended Challenge:

Free Floating - Mobile - Manipulation of Obstacle

Notes: (Poor …1…2…3…4…5… Good)

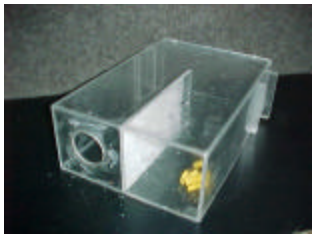
- Acceptance level. 4
- Ease of Construction. 4
- Cost. 2
- Durability. 3
- Ease of Retrieval. 5
- Ease of Preparation. 5
- Capable of presenting multiple sizes of food. . . 5

Observations:

Initial introductions with the "twist-to-lock" lid proved to be successful for the Octopus but not as intended. The Octopus gained access by pulling apart the glued hemispheres of the ball and ignoring the door. Changes in the design of the door and four nylon cable ties overcame the structural flaws of the ball. The elastic bands that hold the door in place were replaced with elastic cord. Later a "Ferretball" was tried and found to be more durable (hemispheres held together with screws) and have a larger capacity. Nice colors too.

The Blind Box

The "blind box" was a clear acrylic box partially divided by a center wall. A hole at one side of the divided end provided the only access to the interior. The box was constructed from the external skimmer box of trickle filter. The open end was covered with a scrap piece of acrylic held in place with nylon cable ties. The drain hole was left open as an access point. Closed off in this way, the box formed a "U" shaped corridor whose end could only be seen from the exterior. Prey items were placed in the far end and could only be gained by reaching blindly around the center-dividing wall. The food items had to be obtained tactilely.



Intended Challenge:

Tactile - Remote View - Variable Manipulation of Obstacles

Notes: (Poor …1…2…3…4…5… Good)

- Acceptance level. 2
- Ease of Construction. 5
- Cost. 5
- Durability. 4
- Ease of Retrieval. 4
- Ease of Preparation. 5
- Capable of presenting multiple sizes of food. . . 4

Observations:

The Octopuses met the blind box with limited enthusiasm. The puzzle seemed to present a limited challenge as it was readily solved. The back panel was removable and various objects were often placed in the box to provide additional obstacles. These seemed to only provide limited difficulty. The acceptance rate was approximately 70 – 75%. It is noted that this is the only hunting puzzle that is square or rectangular in shape.

The Tube

The tube consisted of a 3"diameter acrylic cylinder salvaged from a broken protein skimmer. Left over PVC disks from hole saw cuts were used to make doors. Two smaller disks were notched at opposite ends to allow for a small piece of 3/8" rigid tubing. The small pieces of tubing formed a shaft that the disk could rotate on. These smaller disks were mounted inside the cylinder to form a 10" long chamber with a door at each end. Additional disks were added to both ends of the tube with single cable ties to form flaps. Access to the center of "the tube" required lifting a flap on the end and rotating one of the inside doors. Pieces of rigid tubing were placed as door stops allowing the doors to be rotated in one direction. These "door stops" could be added or removed on either side of the rotating doors. This proved to make the tube puzzle capable of varying degrees of difficulty.

Intended Challenge:

Tactile – Remote View – Manipulation of Obstacle

Notes: (Poor …1…2…3…4…5… Good)

- Acceptance level. 4
- Ease of Construction. 3
- Cost. 3
- Durability. 3
- Ease of Retrieval. 3
- Ease of Preparation. 4
- Capable of presenting multiple sizes of food. . . 2

Observations:

The doorstops were added after the puzzle was solved with regularity. This exposed a flaw in the design. The shafts that held the internal doors proved too weak as the doors were removed rather than manipulated. A later model used half-inch thick PVC disks that were tapped at either end allowing the shafts to be glued in place. Small sections of vinyl tubing were added to the shafts of the doors. These prevented the doors from swinging freely and held the food in the center section. A larger diameter and length of tube was used which increase the variety of food that could be presented. This length of the tube allowed for improved visibility with larger specimens.

The Roto-Cylinder

The "roto-cylinder" was a miniature revolving door in a plastic can. It was constructed from the main cylinder of a Magnum canister filter. The filter's original design allowed for a PVC pipe to be mounted along the cylinder's axis. This formed a shaft for the revolving door. The "door" was made of a slightly larger diameter pipe, rectangles of clear acrylic, and PVC disks from hole saw cuts, put together to resemble a small paddle wheel. When placed on the shaft in the cylinder, the rectangles and the cylinder walls, formed small compartments that could be rotated to an opening cut on the side of the cylinder. A top and bottom were made from the two halves of an empty wire spool held in place with nylon cable ties. The motor housing on the bottom of the Magnum cylinder was filled with gravel and capped with the wire spool. This bottom weight made the puzzle sit up right when presented.

Intended Challenge:

Remote View –Complex Manipulation of Obstacle

Notes: (Poor …1…2…3…4…5… Good)

- Acceptance level. 3
- Ease of Construction. 2
- Cost. 4
- Durability. 2
- Ease of Retrieval. 4
- Ease of Preparation. 4
- Capable of presenting multiple sizes of food. . . 3

Observations:

The durability of the device decreased as the Octopuses grew. Larger specimens were strong enough to loosen the internal panels from their settings and impair the movement of the revolving section. No design improvements have as yet been made. This device proved to be the most challenging, sometimes taking several minutes for the Octopus to solve.

The Travel Mug

The Travel Mug was constructed from a 9-inch section of 3-inch clear PVC pipe. A 3 x 5-inch rectangle of 1/4inch sheet PVC was attached to close off one end of the pipe. The handle was made from a 7 ¾-inch piece of ½-inch diameter PVC pipe. Two pieces of 3/8-inch PVC rod were heated and bent, then threaded through holes drilled in the handle. One rod was shaped like an omega and formed the attachment point to the rectangular base. The other rod was "Z" shaped with one end attaching to the handle and the other to a 3-inch diameter "hole saw" disk that formed a door. A slot was cut in the clear pipe to allow the rod and disk to slide up and out through the pipe. The "S" shape of the rod stopped the disk/door from being pulled clear of the pipe. Lifting up or pushing down on the handle effectively opened or closed off the open end of the clear pipe. Access to the center of the pipe could only be accomplished by pulling up on the external handle.

Intended Challenge:

Remote View – Remote Complex Manipulation

Notes: (Poor …1…2…3…4…5… Good)

- Acceptance level. 4
- Ease of Construction. 2
- Cost. 4
- Durability. 5
- Ease of Retrieval. 5
- Ease of Preparation. 5
- Capable of presenting multiple sizes of food. . . 5

Observations:

The PVC rods were heated using a small blowtorch and water-cooled. The ends of the rods that passed through the base and the disk door were later heated and flattened to prevent them from being pulled off. This design proved to be the most durable of the devices. The small size caused the manipulation to be concealed at times, but was easy to visualize. The first introduction was met with much interest and took an adult Octopus approximately 16 seconds to solve. It took four hours to construct.

Summation:

The development of the 'prey puzzles" significantly improved the activity level of the housed specimens. While there was no apparent increase in longevity, the visitor interest in the exhibit rose dramatically. A progression of complexity seemed to occur as each device was made. This was mainly

due to the relative ease with which the puzzles were solved. It quickly became a challenge to stump the Octopus. This was rarely accomplished for any duration.

It must also be noted that the puzzles listed are but a few of those presented. Most were of simpler designs that either proved too ungainly or had failing marks on ease of use or retrieval. A few mass-produced enrichment devices designed for primates were presented as well. These proved too simplistic as they relied on the movement limitations of a rigid skeleton. One of note, an acrylic peanut maze, was solved in less than 6 seconds. It became apparent that if the interest was there the solution was quick to follow.

Behavioral trends were also observed. The readiness of the individuals to release the devices varied. Younger/ smaller specimens tended to acquire the prey and quickly discard the device. As they grew larger, the puzzles were held and inspected for longer periods even after the prey was acquired. Overall the individual's proficiency of solving the given puzzle increased with the number of presentations. However, the ease of finding the solution for each puzzle varied per individual. Some puzzles seemed to offer little challenge to some, and great difficulty to others. The proficiency level for the more complex puzzles, specifically the roto-cylinder, would drop if the device had not been presented for several feedings. The simpler puzzles seemed to suffer little loss in proficiency if not presented to the octopus for some time.

In conclusion, the introduction of the devices had an overall positive effect. The octopus displayed a marked increase in it's investigation of the exhibit and the items found within. The public showed keen interest in this hunting, and the aquarists routinely had their egos kept in check by the animal's prowess. If any negatives could be noted it would be that any objects introduced into the exhibit were often met with increased interest including those regularly used in tank maintenance, which made routine cleaning more of a challenge.

These devices are intended as a first installment of an enrichment notebook for Octopuses. Anyone looking to contribute to the notebook can contact the aquatics department of the Cleveland Metroparks Zoo at (216)-661-6500 Ex.4485 or Mark Rehling at MLR@clevelandmetroparks.com. Interested parties may also receive a copy of the notebook upon request.

Editor's Note: Additional photos of the above devices are included in the version of the article that is available directly from Mark.

References:

Hanlon, Roger T. & Messenger, John B. (1996) *Cephalopod Behavior*. Cambridge University Press, New York, NY

Nesis, Kir N. (1987) *Cephalopods of the World*. T.F.H. Publications, Inc., Neptune City, NJ

Shepherdson, David J., Mellen, Jill D., and Hutchins, Michael. (1998) *Second Nature: Environmental Enrichment for Captive Animals*. Smithsonian Institution Press, Washington, D.C.

FIRST REARING OF THE HAWAIIAN SEAHORSE, *Hippocampus fisheri* AT THE WAIKIKI AQUARIUM

Karen Brittain, Aquarium Biologist

Waikiki Aquarium

Since December of 1996 the Waikiki Aquarium has had success in raising seahorses thought to be *Hippocampus erectus*. We have been able to rear *H. erectus* in small glass aquariums with minimal effort and are able to get them to eat non-living foods by the time they are one month old. Some of our juveniles even consume flake food at this young age. Encouraged by this success, we decided to attempt the captive culture of the "Hawaiian" seahorse collected off the leeward coast of Oahu. Surprisingly, we found this to be much more of a challenge than we had anticipated!

The Hawaiian seahorse has been tentatively identified as *Hippocampus fisheri*. The life history and conservation status of this unique seahorse are unknown (Lourie, 1999). University students at the Waikiki Aquarium are conducting research projects to investigate the genetic identification of the species and its pair bonding behaviors.

Most seahorses spend their lives in shallow seagrass beds or coral reef habitats. Not so with the Hawaiian seahorse. The Hawaiian seahorses on exhibit at the Waikiki Aquarium were originally collected by fishermen who spend their nights at sea off the Waianae (western) coast of Oahu. The seahorses are attracted to the fishermen's lights and are collected using scoop nets. These relatively small animals are found quite far offshore in very deep water. Fishermen report finding the bodies of seahorses in the stomachs of tuna and other pelagic fishes. Sometimes these seahorses can be obtained from local pet stores, but they are not at all common in the aquarium trade. Newly collected animals have a beautiful red to orange coloration, and after a few months in captivity they take on more of a yellow hue. Adults reach a length of only 8 cm and are most often found in small groups by the fishermen.

The Hawaiian seahorse is a prolific spawner, producing up to 250 hatchlings every 15 days. The 6 mm hatchlings are smaller than those released by *H. erectus* which average 9.8 mm. We started our larval rearing trials using the same techniques as for *H. erectus* and ran into problems almost immediately. While the *H. erectus* had exceptional survival rates, few of the Hawaiian seahorses survived more than four days. Most mortalities were attributable to small gas bubbles within the esophagus of the hatchlings. Many hatchlings also become "stuck" at the surface of the water, seemingly trapped within the surface tension. We tried various techniques to correct these problems, but to no avail. Then we had a breakthrough!

One of the male Hawaiian seahorses involved in a mate selection study looked ready to release young, so we transferred it to one of our large outdoor larval rearing tanks. We hoped that this 4,000 liter tank with its larger volume and currents would more closely resemble the open ocean habitat of these small seahorses. A gentle current is created by releasing large bubbles of air at the bottom center

of the tank. As water is pulled up with the rising bubbles, it flows outward to the walls of the tank and then down to the bottom to once again rise with the bubbles to the surface. The larval tank is constructed of fiberglass and is 2.4 meters in diameter and 1 meter deep. The tank sides and bottom are painted a medium blue. We chose this color because it is light enough to allow us to see the animals and it is dark enough to allow the young fish to see the light colored food items they are hunting. This tank is located outdoors under a fiberglass roof and receives indirect sunlight. The tank is also partially covered with 50% shade cloth.

Once the young seahorses were released from the male's pouch, we added rotifers, *Brachionus plicatilis*, as well as copepods, *Euterpina acutifrons*, to the tank as food for the new seahorses. A microalga, *Tetraselmis chuii*, was added as a food for the rotifers and copepods. Because the young had hatched out directly into the rearing tank we could not get an accurate total count of the hatchlings. It appeared that this was not a very large batch of hatchlings, but because they looked healthy, we decided to continue. Rearing runs like this one involving a large rearing tank demand large amounts of algae and zooplankton to keep the food density high. This represents a significant investment of effort and it is important to make sure that the hatch is good in terms of number and quality of hatchlings. Otherwise the amount of effort put into the plankton production is not worth the expected outcome.

For the first three days, food densities were maintained at 1-2 rotifers / ml and 0.5 copepods / ml. During this time water was neither added to nor removed from the tank. On the third day, the surface of the water became scummy, so we started a slow water exchange of 0.3 liters / min. We are very fortunate to have an excellent source of water from a saltwater well at the Waikiki Aquarium. A 100 micron mesh screen was placed over the center stand pipe so that water could drain, but the seahorses and most of the food items would not be flushed out. Once we initiated the flowthrough system, we had to add 30-50 liters of algae every day. Rotifers and copepods reproduced within the tank so there was no need to add additional zooplankton. Temperature in the rearing tank ranged from 77.5^{o} to 79.0^{o} F during this phase. Although many species of young fish will usually be evenly distributed in rearing tanks, these young Hawaiian seahorses swam together in loose "herds" of ten to twenty individuals.

At the end of the second week, copepods were the dominant food organism in the tank. Rotifer numbers has declined because they are small enough to slowly escape through the 100 micron screen, and because the seahorses had been consuming them. Copepods grow well when they are fed the diatom *Chaetocerus gracilis*, so we began to add this alga to the rearing tank and discontinued the use of the alga *T. chuii*. The copepods were attracted to the sunny areas of the tank and we could see the seahorses (which at this age are a lovely golden color with black bands) snapping up the copepods with gusto.

On day 24, we took measurements of the young seahorses, and they averaged 1.6 cm total length. We decided to provide larger foods for them, by adding three day old *Artemia* enriched on Algamac 2000® for 24 hours. The seahorses soon had pink bellies full of the brine shrimp. We increased the incoming water flow and changed the standpipe screen to a 500 micron mesh to allow

uneaten *Artemia* to flush out of the tank. This may sound a bit wasteful, especially considering the price of *Artemia*, but we wanted to make sure that the seahorses were eating only freshly enriched *Artemia*.

When the seahorses reached 8 weeks old we saw a change in their behavior. They began using their tails to hang onto the air lines and each other as well as to filamentous algae growing on the sides and bottom of the tank. They would also move slowly across the bottom dragging their tails behind them. This was quite disconcerting at first, it looked like there were dead seahorse bodies sprawled across the tank floor. On closer inspection we could see that they were moving ever so slowly, seemingly studying the bottom of the tank. We immediately started adding mashed up frozen mysids in the hopes that the seahorses would come across them during their surveys of the bottom. We are not sure if they actually consumed the frozen mysis at this point.

On December 10, 1999, we celebrated completion of the life cycle of the Hawaiian seahorse in captivity. At the tender age of less than 4 months, our tank bred seahorses produced hatchlings of their own. Adults and hatchlings were measured from the tip of the snout to the tip of the tail with the head tilted up and the tail stretched out straight. The adult females averaged 3.8 cm, and adult males 4.2 cm. Although these adults were half the size of those collected from the wild, the average size of the hatchlings was the same as that of the hatchlings from the wild caught adults. The number of hatchlings produced in the first batches was quite small, but we expect that the number will increase as the new parents attain their full size.

We are pleased that our investment was rewarded. Three generations of the Hawaiian seahorses are now on display in the Miniature Marine Life exhibit at the Waikiki Aquarium.

References:

Lourie, S.A., A.C.J.Vincent and H.J. Hall (1999) Seahorses: An Identification guide to the world's species and their conservation. Project Seahorse. London, UK

INCREASED VISITOR RESIDENCY TIME AT A RENOVATED AQUARIUM EXHIBIT
or:
"Damn it Jim! I'm a marine biologist - not a statistician!"
or:
"Cripes, isn't there any topic that Jay won't try to make into a paper?"

Jay Hemdal

The Toledo Zoo

The "Pollution Exhibit" at the Toledo Zoo was constructed around 15 years ago. This 550-gallon exhibit originally had no fish in it, just various "artifacts": cans, bottles, an old tire and the requisite boot. The backdrop is a brick wall with a simulated sewer outfall. Overhead backlit graphics discuss local water pollution issues. The whole exhibit is allowed to grow algae and the build up of detritus adds to the overall effect. With the introduction of zebra mussels and round gobies to local waters in the early 1990's, these species were exhibited in this display as examples of exotic introductions or "living pollution". The exhibit remained moderately popular with our visitors.

In order to "punch it up" a bit we decided to add some fresh accoutrements to the exhibit; a lost tackle box, more bottles and fishing lures were all discussed. One of the aquarists mounted a pair of rubber boots to the top of the brick wall. The result looked like somebody was sitting on the wall, dangling their boots in the water. We immediately noticed an increased response from our visitors – pointing and laughing, but also stopping to read the important messages about pollution. Another aquarist dangled a fishing lure and bobber in the center of the tank in front of the boots. More laughter and finger pointing, "Hey somebody is fishing in that tank!" We had a stepped down motor gear assembly from a peristaltic pump (left over from a failed experiment). By attaching a cog to the motor and tying that to the fishing line above the exhibit we were able to simulate the bobber being jigged up and down. The effect was now complete – and we had people crowding in front of the exhibit and trying to look up through the surface of the water to "see who was fishing". Most adults understood it was just a gimmick, but children often were heard to wonder what was going on. The adults would then explain it to them, often reading them the graphics at the same time.

The aquarium staff knew that these changes were popular, but just how much better was the exhibit? Its only natural for aquarists to interpret visitor's reactions to new exhibits more favorably than they should – nobody wants their efforts at new displays to be in vain. We devised a plan to measure visitor's "residency time" at the exhibit, and then "de-constructed" the exhibit's new components piece by piece in order to measure the change in the time spent in front of the exhibit by the visitors.

The survey method used was to simply observe visitors as they passed by the tank. The observer was stationed far enough away so as to be as unobtrusive as possible. Since people almost always visit aquariums in cohesive groups, it was these that were measured as opposed to individuals themselves. The time a group spent in front of the exhibit was averaged to the nearest second. In some cases part of a group moved on before the remainder was finished looking. In these cases, the time was counted up to the point where 50% of the group had moved on. A time of zero was given to any "walk-bys". This term was coined to describe those visitors who walked by the exhibit, glanced at it, but did not stop. Groups who were just traveling through the building and not looking at the exhibits

were not counted. Twenty groups were timed as they passed by the exhibit in four different scenarios. A second recently renovated exhibit was also timed for comparison:

Pollution exhibit with boots and moving bobber. The twenty groups timed while passing the completed exhibit comprised 56 individuals. Four groups were "walk-bys" and the time spent in front of the exhibit ranged from 0 to 150 seconds with an average of 37 seconds per group.

Pollution exhibit with just the moving bobber. The twenty groups timed while passing the exhibit with only the moving bobber installed comprised 52 individuals. Five groups were "walk-bys" and the time spent in front of the exhibit ranged from 0 to 130 seconds with an average of 29 seconds per group.

Pollution exhibit with boots alone. The twenty groups timed while passing the exhibit with only the pair of boots installed comprised 47 individuals. Five groups were "walk-bys" and the time spent in front of the exhibit ranged from 0 to 55 seconds with an average of 22 seconds per group.

Pollution exhibit as it was prior to the renovation. The twenty groups timed while passing the exhibit while it was returned to its original state (no new items) comprised 50 individuals. Nine groups were "walk-bys" and the time spent in front of the exhibit ranged from 0 to 50 seconds with an average of 13 seconds per group.

New "Fishes of Madagascar" exhibit. The twenty groups timed while passing this recently renovated exhibit comprised 47 individuals. Eleven groups were "walk-bys" and the time spent in front of the exhibit ranged from 0 to 31 seconds with an average of 6 seconds per group.

There seemed to be a direct correlation between the additions of various exhibit components to the length of time visitors spent viewing the exhibit. With the boots and moving bobber in place, visitors spent more than 2 ½ times longer in front of the tank. The low residency time at the Madagascar exhibit was disheartening. This exhibit contained a number of highly endangered fishes, had recently been completely aquascaped, and arguably has the best quality graphics of any of our regular display aquariums. We suspected before construction even started that our typical visitor wouldn't find this a very exciting exhibit, but proving that point didn't make us feel any better! It can't all be about "attraction" – serious conservation messages need to be presented despite the chance that visitors may pan them.

Overall, this method seemed to accurately measure the average time visitors spent viewing a particular exhibit, and this is probably in direct correlation with the visitor's interest. By extrapolation, it is hoped that the longer visitors spend at an exhibit, the more enjoyment they receive, and the more they learn. With practice, it isn't very time consuming to observe a group of 20 visitors. On a busy day, the groups tend to run together making analysis more difficult, while on slow days you might spend too much time waiting for the next group to approach the exhibit. By picking a time when attendance was between these two extremes, it took less than an hour and a half to gather all the data used here.

SIZE DISTRIBUTION OF A POPULATION OF GIANT FLOATER MUSSELS (*PYGANODON GRANDIS*) IN BOWMAN LAKE, ALLEN COUNTY, INDIANA

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Introduction:

The native freshwater mussels of the United States are in peril. The US Fish and Wildlife Service currently lists sixty-one species of American mussels as endangered and eight as threatened (USFWS 2000). Treats to their survival include river impoundment, water pollution, poaching and competition from exotic zebra mussels (Neves 1997). McMahon (1991) gives an excellent review of the biology and ecology of the group.

Fortunately, some diverse and abundant mussel beds persist, and several are located in the rivers and lakes near Fort Wayne, Indiana. Since 1994, I have been conducting a field survey of the mussels of Allen County, and have documented the presence of twenty-four species. Most of the survey has been done on the rivers, and will be reported elsewhere.

This paper reports on a related study, which focused on a lake-dwelling population of one of the more common and hardy species of Midwestern unionids, the giant floater, *Pyganodon (=Anodonta) grandis* (Cummings & Mayer 1992). I was originally invited by the staff of Fox Island County Park, to SCUBA dive the site in 1999, in order to identify the mussels which they knew resided in their lake. That year, my assistant and I dived Bowman Lake twice, on 1 July and 20 August 1999. The outcomes of those dives yielded the following facts about the lake's freshwater mussel fauna.

- The vast majority of mussels are located in a single bed, positioned just west of the swimming beach.
- The mussels were of one species, the giant floater, *Pyganodon grandis* (Figure 1).
- The average mussel density was about 0.654 mussels per m^2.
- There were an estimated number of 365 mussels in the population.

Those results were considered to be preliminary, due to the small number of mussels which we were able to measure using the Point-Quarter Method.

The objective in 2000 was to measure enough mussels to generate a meaningful size frequency histogram. The approach was simple. We set out to find and measure as many mussels as possible in the limited time available.

Figure 1. The Giant Floater Mussel, *Pyganodon grandis*.

Methods:

We SCUBA dived the mussel bed in Bowman Lake twice on 24 July 2000, from 10 AM to 12:30 PM, and again from 2:00 PM to 3:30 PM. The total man-hour bottom-time for the day was eight hours. The search began just beyond the swimming area, in about 1 m of water, and moved through the mussel bed until we reached its western end. There, we turned to follow the bed's northern edge to a point approximately half way back to the starting point.

When a mussel was encountered, one diver marked its location, and removed it for examination and measurement of its length, height and width. It was immediately returned to its original location. The other diver recorded the caliper readings in a waterproof Nalgene® notebook.

This work was conducted under permit from the State of Indiana viz., Indiana Department of Natural Resources, Division of Fish and Wildlife, Scientific Collectors License # 2080, issued on 28 January 2000.

Results:

A total of 172 *P. grandis* were examined and measured, which was approximately half of the total population estimated in 1999. Although the shell of a *P. grandis* is not a perfect geometrical shape, it is approximately that of an ellipsoid, the formula of which is given by:

$(4/3)\pi$ abc = V where: a = ½ length, b = ½ width, c = ½ height, v = volume.

A modification of this formula to suit the data collected from the *P. grandis* in this study is:

$$\frac{0.523(SL)(SH)(SW)}{1000} = V$$

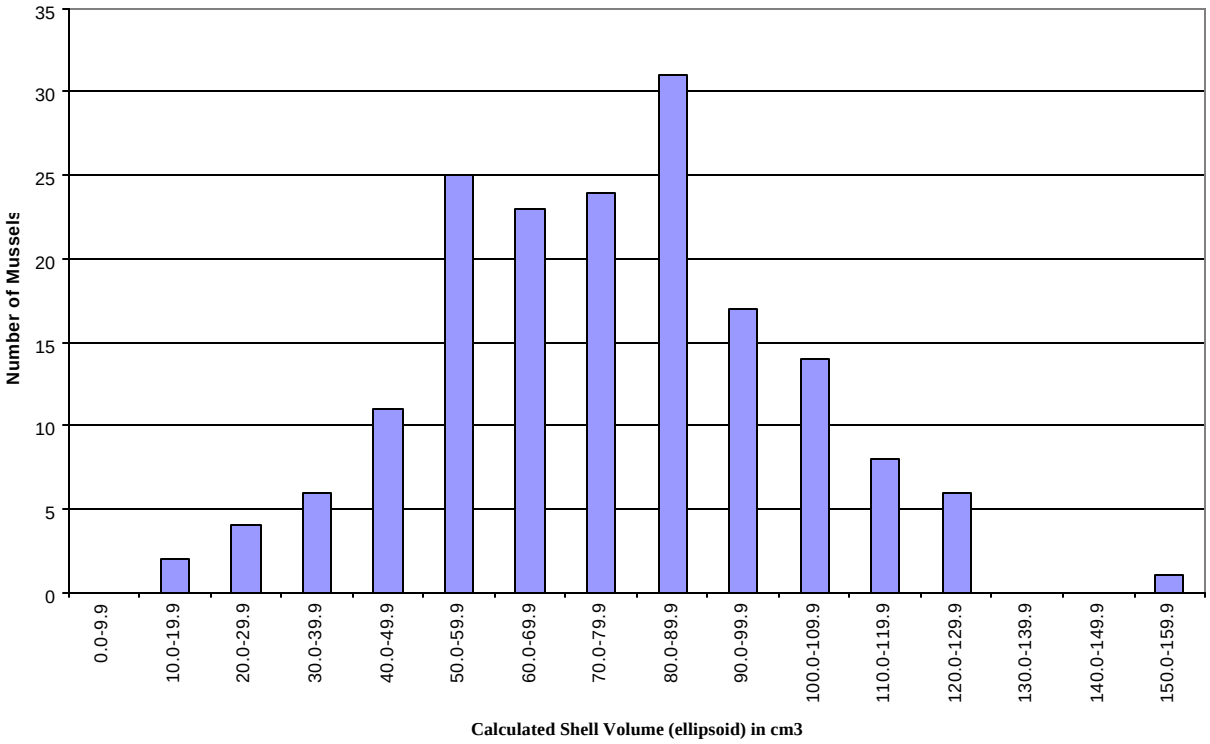
where: SL = shell length (mm), SH = shell height (mm)
SW = shell width (mm), V = ellipsoid volume (cm^3)

This latter equation was used to combine the length, height and width of each shell into a single index of shell size, V (ellipsoid volume). The size frequency distribution for *P. grandis* is shown in Figure 2. The pattern of size distribution is reasonably normal, with the exception of a rather unexpectedly high number of mussels in the 50.0-59.9 cm^3 size class, and no mussels in the size classes 130.0-139.9 and 140.0-149.9 cm^3.

Conclusions:

The pattern of shell size frequency distribution suggests that a stable and reproductively active population of *P. grandis* currently occupies Bowman Lake. Since this artificial lake is supplied entirely by ground water input, is protected as part of a county park, and has no significant motor boat traffic, it is likely that this population of freshwater mussels will remain healthy at least for the near future. In addition, since this species of unionid is apparently thriving at the site, there is good reason to consider the possible use of Fox Island County Park as a future refuge for other more endangered species of freshwater mussels which are found in Allen County, Indiana. These species include the federally endangered clubshell (*Pleurobema clava*) and the catspaw (*Epioblasma obliquata*). In the event that zebra mussels or pollution put pressure on the endangered mussels in the county, a small culture facility could be constructed

Figure 2: Size Frequency Distribution of *Pyganodon* at Bowman Lake, 24 July 2000



near the lake which could draw in lake water to feed open system aquariums in which the endangered species would be maintained and propagated.

Future research on this population of *P. grandis* is warranted. The same methods as were used in the present study should be repeated in subsequent years, to monitor growth, reproduction and other changes in the population structure. The mussel bed should be accurately mapped. A statistically significant number of individuals should be marked, measured and re-captured to measure growth rates and mortality rates in the population.

The positive balance between the modest resources and time which were spent, and the useful results which were achieved, demonstrate the potential contributions which even small public aquariums and zoos can make to local unionid conservation. The skills, equipment and expertise which are part of the daily operation of a public aquarium are easily applied to such efforts.

Acknowledgements:

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FRESHWATER R&D AT MONTEREY BAY AQUARIUM

Mark Faulkner, Aquarist (mfaulkner@mbayaq.org)

Monterey Bay Aquarium

Introduction:

In February 2000 I began a project behind the scenes at the Monterey Bay Aquarium to setup a 500-gallon R&D aquatic plant display. My goal was to recreate an Amazonian flooded forest, using plants and animals native to the Amazon River Basin. The tank was designed to promote luxuriant plant growth, using carbon dioxide injection, metal halide lighting, and nutrient supplementation.

Over time, this aquarium went through some interesting changes. The first four months of setup the tank showed moderate growth by the large swords, while some plants, mainly low foreground plants, slowly withered and died. The next two months the tank was choked with filamentous algae. During the last three months, however, the display showed incredible plant growth, with zero filamentous algae growth. Over the last nine months I changed many variables in order to obtain a stable, healthy system. This article describes the plants and animals in this aquarium, approaches I took to obtain a lush display, and mistakes I made along the way.

Plants and Animals:

Plants:
5 *Echinodorus paniculatus*
6 *E. cordifolius*
5 *E. rigidifolius*
60 *E. tennellus*
50 *E. quadrocostatus*
1 *E. apart*
3 *E.* 'Rubin'
1 *E.* 'Oriental'
2 *E.* 'Rose'
7 *Eleocharis montevidensis*
5 Hydrocotyle leucocephala
100 + *Sagittaria subulata*
Lemna major

Animals:
11 *Symphysodon aequifasciata* - discus
50 *Hemigrammus bleheri* – rummy-nose tetra
60 *Otocinclus affinis* – otocinclus
4 *Farlowella acus* – twig catfish
3 *Crossocheilus siamensis* – Siamese algae eater
130 *Cardina japonica* – Yamato shrimp

Sword plants made up the majority of the vegetative life in this display. *Echinodorus tennellus, E. quadrocostatus* and *Sagittaria subulata* were in the foreground. All of these plants produced runners and eventually filled in the foreground to make a "lawn" at the front of the tank. The back was filled in by a variety of large swords such as *Echinodorus* 'Rubin', *E. cordifolius,* and *E. rigidifolius.* The rest of the plants were used as accent plants because of leaf color and or shape.

The display contained eleven wild discus measuring 4 – 6 in. and 50 rummy-nose tetras. Four twig catfish (Amazon natives) were placed in the tank because of their interesting morphology as well as for algal control. The rest of the animals are not native to the Amazon, but were used solely for their roles in algae control as well.

Plumbing:

The aquarium dimensions were 75"x 24"x 25". Water left the tank via a weir box mounted on the outside of the display tank, where it passed through a prefilter and continued into an 80-gallon sump

located beneath the display. The sump contained all heaters, a UV sterilizer and power-head used for CO2 injection. Water was pumped back to the display using a 1/8th horsepower pump. Originally the majority of the return water went to a spray bar located in the display and the rest was diverted to a slow-flow reverse under gravel filter (RUGF). [Aquatic plant hobbyists have commonly used under gravel heating cables to keep substrate temperatures the same or slightly warmer than the water column. These cables also supplied plant roots with nutrients through convection currents. The slow-flow RUGF was intended to provide the same functions as the heating cables.] It was later determined that the RUGF was not allowing anoxic or anaerobic conditions to occur in the substrate. A process that makes certain nutrients, like iron, more available to plant roots. Flows to the RUGF were shut off and all of the water going to the display went through the spray-bar.

Driftwood And Substrate:

The tank was started with one large driftwood stump as a centerpiece. The driftwood was collected off a local beach and was soaked in fresh water for six weeks to remove salts, prior to being placed in the display tank. This piece of wood was cable-tied to the RUGF plate to keep it submerged. Approximately 300 pounds of Profile's Turface was added as substrate. Turface is clay-based gravel that is often used as baseball infield material. It has qualities that make it suitable for an aquatic plant display. It is relatively small grained (2-3mm), has a natural look to it, and it has a high cation exchange capacity. The main drawback to the Turface is that it is not as dense as other substrate materials. This makes it "light" under water and doesn't allow sloping gravel beds and initially plants are easily uprooted. In retrospect I would have used a mixture of Seachem Flourite and 2-3mm river gravel. Another mistake I made with substrate was not adding a few handfuls of peat to the lower half of the substrate when initially setting up the display. The acidic nature of peat makes certain nutrients and dissolved organics available to plant roots.

Carbon Dioxide:

The water column was supplied with carbon dioxide via a compressed gas cylinder. A needle valve was used to limit flow and was set at four to five bubbles per second. During the first three months a Pinpoint pH controller controlled CO2 injection. The pH controller was set to keep the pH between 6.9 and 7.0 (CO2 levels around 20ppm). During these three months the controller failed two times. Once, because of a bad electrode and the second time because the main system completely shut down. Due to the lack of reliability of this controller I decided to bubble the CO2 directly into the system and remove the Pinpoint controller altogether. CO2 flow was set at 3 bubbles per second, which maintained pH near 7.05 when tank lights were on and around 6.9 when the lights were out. Since the water used in the aquarium had a relatively high carbonate hardness value, pH plummet was of little concern.

Ideal CO2 levels in a highly lit, planted aquarium should be between 10 and 20ppm. CO2 levels are easy to determine if pH and Carbonate Hardness are known, since there is a direct relationship between the three. An easy to read CO2 chart and explanations are available on the Internet at www.thekrib.com/Plants/CO2/kh-ph-co2-chart.html.

Lighting:

Lighting of the display was accomplished using metal halide fixtures. Originally two 250-watt, 5500K lights were hung 10 inches above the aquarium. As the plants began to grow, the larger ones began to shade to the smaller, foreground growth. Another 250-watt fixture was added. This fixture contained a 6500K Iwasaki bulb and was placed in the center of the tank between the two existing fixtures. The 6500K bulb produced a much "whiter" light that really brought out the colors of the fish and plants. As the plants continued to grow and cover the surface a fourth fixture was added. This contained a 175-watt 5500K bulb.

Nutrients:

Determining nutrient levels proved to be one of the greatest challenges in creating a stable, algae-free environment. I made several mistakes along the way, which resulted in a tank choked by epiphitic green and red algae.

As stated earlier, I began this experiment using a slow-flow RUGF. Even after nutrient dosing, extreme at times, plants still showed signs of deficiencies. Iron levels would drop from 1 ppm to 0 within a few hours. When the RUGF was finally shut off, the need for dosing decreased, nuisance algae disappeared, and the plants began to literally grow out of the display. I believe a couple of things were going on with the use of the RUGF. Potentially, the water flowing up through the substrate from the RUGF was causing nutrients to be oxidized (this is only a theory). Also, the RUGF was not allowing for any anoxic conditions to occur in the substrate. Anoxic conditions make nutrients trapped in the substrate more available to plant roots.

When the plant growth increased dramatically, naturally so did plant density. The density of healthy growing plants is also a factor in creating an algae free environment. Higher plant density leads to a more stable environment.

Measuring nutrient levels proved to be a valuable tool in fine tuning water chemistry specifically for the plants. A mistake I made early on was not measuring for potassium, even though I knew it was a macro nutrient essential for plant growth. It wasn't until several of my swords showed signs of serious deficiency and growth throughout the tank had stalled, that I sought advice from Steve Dixon, a knowledgeable planted aquarium enthusiast. Steve advised me to order a potassium test kit and begin dosing more potassium nitrate (KNO3). When I raised potassium levels in the display to around 25-ppm, growth picked back up.

Nitrate tended to be deficient in my system as well, even after the heavy feeding the discus required. The KNO3 used to raise potassium levels also raised nitrate. Ideal nitrate levels were between 3-5 ppm.

Phosphate was also measured regularly. Ideally PO4 levels should have remained as close to 0 as possible. At the point when algae be came a problem, phosphate levels reached 1 ppm. Once the plants began thriving (after potassium addition and shut off of RUGF) phosphate levels dropped off to nearly undetectable levels (<0.1 ppm). The plants were utilizing the phosphates quicker than they were

introduced to the water column. With a steady decline in PO4, the nuisence algae disappeared from the system.

The fourth nutrient measured was iron. Iron levels were used to gauge the need for iron as well as trace element addition. Iron was measured accurately in our water quality lab on a weekly basis, which allowed me to maintain only a trace in the water column. Throughout the experiment iron levels never rose above 0.08ppm and regularly stayed below 0.02ppm. On a daily basis I used a quick test to determine whether or not iron was present. If the test showed iron in the water column I would hold off dosing until the test read 0. When iron did read 0, I would dose both Seachem Flourish Iron and Seachem Flourish (a trace element solution) following amounts recommended on the bottles. This technique proved to be effective in supplying plants both iron and trace elements in the water column. Once I shut off the RUGF and anaerobic conditions developed in the soil, iron became available to the roots as well, and the water column required less frequent additions of the Seachem fertilizers.

Substrate fertilizers were placed directly under sword plants every month to 2 months. Under most plants I would use ½ of a "Jobe's fertilizer stick for lush palm and ferns". This stick was chosen because of its low phosphate content.

Summary:

The main mistake I made was using the RUGF. When this was shut down, dramatic improvements in display quality ensued. It is theorized the anaerobic conditions that subsequently occurred in the substrate made nutrients more available to the plant roots. This in turn gave the plants what they needed to thrive, increasing their density, and utilizing water column nutrients more efficiently. This left little for nuisance algae to utilize, since they rely on water column nutrients.

Planted aquariums can make dramatic displays and create natural environments for freshwater fishes. Each aquarium is different due to the number of variables inherent in such a system. If sufficient lighting is supplied, CO2 is made available, the right substrate is provided, and nutrient levels are managed, an awe-inspiring aquarium can be created.

MORE ON PLANTED AQUARIUMS - EDITOR'S NOTES

It is always refreshing when someone highlights subjects that have been bypassed by public aquariums. Mark Faulkner's article on planted aquariums should be a wake-up call to aquarists. In the early days of our "industry", nicely planted aquariums were somewhat common at public aquariums. In the rush to expand marine collections in the 60s and 70s, most of our older facilities seem to have lost their plant cultivation skills. New facilities have been light on freshwater displays in general, with only a couple of notable exceptions (Chattanooga and Duluth).

The high-tech methods discussed by Mark are reminiscent of the techniques used for coral cultivation, and anyone who has mastered the latter is also capable of creating lushly planted aquariums. My own interest in plants was re-ignited in 1994 by Karen Randall's "Sunken Gardens" column, then simply a monthly contribution to the Boston Aquarium Society's Xeroxed club newsletter, *The Daphnian*. This column now appears nationally in *Aquarium Fish Magazine*. Karen has been very generous with her time, and has been one of the leaders in bringing this aspect of the Freshwater hobby back into the public eye.

Planted tank enthusiasts have been well organized on the WWW for many years. A key site is http://www.aquatic-gardeners.org/, the homepage of the Aquatic Gardeners Association. Look for links to the *Aquatic Plant Digest* (APD), an excellent listserver. "Lurking" here, and asking a few questions of the experts, is a profitable use of time. You will also discover links to *The Krib*, (URL in Mark's paper) an excellent FW site with lots of planted tank information. **Look at these sites for more information on the subjects I'll mention below.** The beautiful "coffee-table" books by the Japanese aquarist and photographer, Amano, are a wonderful source of ideas for planting schemes. Most catalogs and large shops carry them.

So-called "low-tech" (plant-tank jargon) plant tanks do not require CO_2 supplementation, and typically are conservative in their need for fertilizers. "High-tech" systems include CO_2 injection, daily fertilization with phosphate-free mixes, and the use of metal halide lighting. You will find that planted tank enthusiasts have as many theories about the "right" way to set up a plant system as coral keepers. My comments target just one successful method, which is essentially just a variation of the techniques Mark is using.

Driftwood: If you want to avoid battles with algae, fungus, and the innate buoyancy of dry wood, avoid using materials found in the woods. Aquarium Driftwood (800-600-4132) is a business that specializes in locating submerged stumps in southeastern swamps and wrestling them away from the local 'gators. They harvest aged cypress and cedar wood that is dense, saturated with water (it is shipped wet) and unlikely to stimulate the growth of unwanted fuzz of any kind.

Substrate: This is one of the great debates being fought by the "experts". I have chosen to ignore the expensive brand-name substrate amendments. Most plants appear to like fine, silty material around their roots, and the best source I have found to date is "red art clay", a dry clay mix available for a few cents a pound through any ceramics/pottery supply house (check the yellow pages). It is thought by many to be a good replacement for laterites. I have even used untreated, clay kitty litter (caterite?) with some success at home. Mix the clay with half the gravel and use the remaining gravel to layer over it. Don't gravel-wash below the plain gravel layer. Pea gravel should be #1 grade (smaller than most commercial aquarium gravels). Avoid the use of undergravel filters if you plan to use clay or other fine

materials in your substrate. Placing styrofoam insulation under the aquarium before you fill it eliminates the need for substrate heating in cool back areas.

Fertilizers and Micronutrients: Access the recipes for PMDD (poor man's dosing drops) through *The Krib* and APD FAQs. The standard recipe can be manufactured for far less than the cost of commercial products. Perhaps because, like Mark, we have our tanks heavily planted with swords, we also had to increase the proportion of nitrate-rich compounds in the mix. If CO_2 and metal halide lighting is used, daily of additions of up to 3-5 cc per 100 gallons may be needed. As Mark notes, Jobe's Plant Sticks for ferns are a cheap way to add additional fertilizer to the substrate around certain plants. If you want to avoid "green water" episodes, this is the only way that supplemental phosphate should enter the tank.

Carbon Dioxide: We've injected through the special "CO_2 reactors" available through the big catalog houses, but have also found that injecting gas into the intake of a pump is convenient. As we have all trained ourselves never to entrain air into the suction side of a pump (to prevent supersaturation), this almost requires breaking a taboo! Inexpensive CO_2 regulators can be picked up at any brewing supply store. These can be fitted with needle valves (see FAQs) to control the flow of gas. I've never owned a pH controller and have found that small daily adjustments are usually adequate to keep $CO2_2$ levels where I want them. Small CO_2 indicators are available which contain a fluid that varies from blue to chartreuse depending on the CO_2 level and pH of the tank. These are cheap and reliable devices that allow fine tuning without the use of bench tests.

Pests: Panacure is useful to control hydra, if you can't use pearl gouramis for this purpose. Snails are not usually a problem in lush planted aquariums, but clown loaches are dependable snail predators if you really need them. Planktonic algae blooms (green water) are best controlled using the "towel method". This involves covering the aquarium for 5-7 days (no room light should enter the tank) and is usually effective. Recently we had to darken a tank for 10 days and then reduce the light period to 6 hours for another two weeks in order to get a particularly nasty bloom under control. Green water is most common in unstable aquariums, typically newly set up plant tanks, but occasionally in older systems that have somehow been disturbed through a major change in water quality. Suspend fertilizer additions and add an airstone while the tank is dark. Turn off the carbon dioxide if pH control is not an issue.

Our Amazon River tank draws visitors like a magnet. It is bright, colorful and unusual. Hopefully more planted displays will appear in public aquariums in the next few years. Mark, thanks for reminding us about this "lost art", and for sharing your experiences.

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ADVANCES IN CORAL HUSBANDRY: PROPAGATION FOR THE FUTURE

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Coral husbandry has greatly improved since the first reported long-term success of its maintenance at the Noumea Aquarium, New Caledonia in 1956 (Catala, 1964). Advances in life support system technology, ease of exporting corals from developing countries, and a growth in the hobbyist interest across North America and Europe have all contributed to a dramatic increase in the live coral trade (Green and Shirley, 1999). Prior to the early 1990's, dead corals accounted for more than 90% of the trade for use as ornamentals. Since then, the live coral trade has increased dramatically to the point where it comprises approximately 50% of all coral trade (Green and Shirley, 1999). This increase is primarily due to advances in mini-reef technology and subsequent growth in the home aquarium industry.

In 1989, the Waikiki Aquarium published a list of the most popular corals maintained in captivity which included large-polyped corals that inhabit back reef or lagoon environments (*Euphyllia* spp., *Fungia* spp., *Goniopora* spp., *Physogyra* spp., *Plerogyra* spp., and *Sarcophyton* spp.) (Carlson, 1989). At that time, *Acropora* species and other small polyped stony corals were "proven to be much more difficult to maintain in captivity, with 'table' *Acropora* species remaining a challenge" (Carlson, 1989). In the past decade, it is apparent that there remains a strong interest in colorful, large-polyped corals. A recent study based on CITES records from 1985-1997 revealed that the most common genera of living, scleractinian coral traded were those that are usually more colorful or have large polyps including *Euphyllia* spp., *Goniopora* spp., *Catalaphyllia* spp., *Trachyphyllia* spp., *Heliofungia* spp., and *Plerogyra* spp. (Green and Shirley, 1999).

At the present time, aquarists can maintain a much greater variety of scleractinian corals than a decade ago, including many species of small-polyped stony corals. The technology associated with such advancements has come full circle from being very technical and complex to the bare minimum. At the Long Beach Aquarium of the Pacific, we prefer the minimum approach using only a protein skimmer, calcium reactor, live rock, metal halide lighting, and a powerful jet system to provide water motion. One of my favorite sayings is "more flow" and I have no doubt that proper water flow is essential to a successful reef aquarium.

The Long Beach Aquarium of the Pacific began its Live Coral Exhibit in March 1997. Most of our original corals were procured as fragments from the Waikiki Aquarium or from US Fish and Wildlife confiscations. Due to the latter resource, we have been fortunate enough to experiment with many different stony and soft coral species (Table 1). We currently maintain 38 scleractinian coral genera, including some of the less common species such as *Montipora foliosa*, and several table *Acropora* species. We are also maintaining 17 soft coral genera including *Dendronephthya* spp.,

Scleronephthya sp., and *Nepthogorgia* sp., and two species of the hydrozoans, *Distichopora* and *Stylaster*. Although we have not yet formally established our coral growth research, many of our colonies, especially *Acropora* spp. and *Montipora* spp. have exhibited tremendous development. Estimates at this time range from 18-20cm in length per year.

Table 1. Coral genera maintained at the Long Beach Aquarium of the Pacific (58 genera).

Class Anthozoa	Montastrea	**Subclass Octocorallia**
Subclass Hexacorallia	Montipora	
	Nemenzophyllia	Alcyonium
Acrehelia	Pachyseris	Anthelia
Acropora	Pavona	Chironephthya
Blastomussa	Pectinia	Cladiella
Catalaphyllia	Platygyra	Clavularia
Caulastrea	Plerogyra	Dendronephthya
Cynarina	Pocillopora	Heliopora
Dendrophyllia	Polyphyllia	Lobophytum
Echinophyllia	Porites	Nephthea
Echinopora	Psammocora	Nephthyigorgia
Euphyllia	Seriatorpora	Pachyclavularia
Favia	Stylophora	Sarcophyton
Favites	Symphyllia	Scleronephthya
Fungia	Trachyphyllia	Sinularia
Galaxea	Tubastrea	Studeriotes
Goniastrea	Turbinaria	Tubipora
Goniopora		Xenia
Heliofungia	**Subclass Corallimorpharia**	
Hydnophora	Cirripathes	**Class Hydrozoa**
Lobophyllia		Millepora
Merulina		Distichopora

Nonphotosynthetic soft corals (e.g., family Nephtheidae) remain to be a challenge for aquarists. At the Aquarium of the Pacific, we have discovered some of the basics and are always striving to learn the intricacies of maintaining these species. Here are some of the lessons we have learned:

1. Water flow is essential. It appears that nonphotosynthetic soft coral species are more sensitive to particular types of flow than other soft and stony corals, much like the crinoids we maintain. Most species seem to prefer a moderate to strong, diffuse, laminar flow.
2. Food is another critical component in that both the proper size and composition is important. We use a variety of phytoplankton including *Chlorella* sp., *Spirulina* sp., *Isochrysis* sp., and *Nanochloropsis* sp. To this, we add rotifers and supplemented baby brine shrimp (*Artemia* sp.). We then drip approximately 1 liter of "soup" per 100 gallons into each system very slowly over the course of the night.
3. Nonphotosynthetic soft corals do not seem to need specific levels of lower light. It appears however, that they prefer shaded areas.

The hydrozoan, *Distichopora* sp. is also a challenging species. Unlike nonphotosynthetic soft corals, these animals unquestionably require low light levels. If left in even moderate light, fouling organisms quickly adhere to their delicate tissues and result in mortality. *Distichopora* sp. prefers a water flow and feeding regime similar to nonphotosynthetic soft corals.

In addition to the increased capabilities of maintaining a wide variety of soft and stony corals in captivity, there have been great advances in the captive propagation of coral. Surprisingly, it is those species that were previously considered difficult to maintain that are currently the easiest to propagate. In 1992 there were approximately 21 genera of scleractinian corals that had good or excellent captive propagation potential (Yates and Carlson, 1992) (Table 2). Recently, a short survey of several US public aquariums showed that 25 species of stony corals, 13 species of soft corals, and 5 species of corallimorphs are currently being cultured at those facilities (Table 3). It is obvious that the interest in coral propagation has increased dramatically over the past 10 years. However, important issues remain to be researched including the best use of cultured corals.

Currently, 58% of the world's reefs are threatened by human activity primarily through destructive fishing practices, coastal development, pollution, sedimentation, and eutrophication (Bryant et. al., 1998) (Table 4). The effects of the international coral trade are miniscule compared with these factors. However, this should not deter those associated with the hobbyist and public aquarium industries from finding alternative sources of living corals. As conservation facilities, public aquariums are obligated to assist in the reduction of coral reef degradation.

A survey of Los Angeles wholesalers revealed that very few facilities offer cultured corals (Table 5). Most of the coral supplied to this industry is collected from the wild (96%) with only a minor amount (0.03%) produced from aquaculture (Green and Shirley, 1999). Only a fraction of facilities sell cultured corals, and those that do, usually carry less than 1% of their total coral inventory. Some of the reasons given for the decreased stock of cultured corals were price (cultured corals cost more), quality (cultured corals lack color and large size), survivability (cultured corals do not ship well), and lack of demand by retailers. When asked if they would be interested in selling cultured corals were these problems to be resolved, all wholesalers interviewed suggested they would be supportive of such efforts.

At the opposite end of the spectrum, hobbyists are interested in procuring cultured corals but report a lack of availability. A survey of 683 home aquarists presented by the World Conservation Monitoring Centre showed that when given the choice between cultured and wild collected corals, an overwhelming majority preferred cultured (Green and Shirley, 1999). Most in fact, reported a willingness to pay more for cultured corals and accept pieces that may not be as colorful as wild-caught corals. However, the survey also suggested that the majority of home aquarists do not have regular access to cultured corals. This seems contrary to what the wholesalers described as a “lack of demand” for cultured corals. Could it be not so much a lack

Table 2. Coral species propagated in 1992 (Yates and Carlson, 1992) and 1999.

1992	1999	
Subclass Hexacorallia	**Subclass Hexacorallia**	**Subclass Octocorallia**
Acropora spp. (13 species)	*Acrhelia horrescens*	*Alcyonium* sp.
Anacropora sp.	*Acropora* spp.	*Capnella* sp.
Catalaphyllia jardinei	*Anacropora forbesi*	*Cladiella* sp.
Caulastrea furcata	*Caulastrea furcata*	*Clavularia* sp.
Cycloseris sp.	*Cycloseris* sp.	"Seamats" (*Pachyclavularia*?)
Diaseris fragilis	*Echinopora lamellina*	*Eunicea* sp.
Echinopora lamellina	*Euphyllia* spp. (3 spp.)	*Gorgonia ventalina*
Euphyllia spp.	*Fungia* sp. ("Mushrooms?")	*Heliopora coerulea*
Goniopora sp. (2 species)	*Galaxea* spp. (2 spp.)	*Heteroxenia* sp.
Hydnophora rigida	*Goniopora* sp.	*Nephthea* sp.
Lobophyllia hemprichii	*Hydnophora* sp.	*Rumphella* sp.
Montipora spp. (3 species)	*Lobophyllia hemprichii*	*Sarcophyton* spp. (2 spp.)
Pavona cactus	*Merulina scabricula*	*Sinularia* spp. (2 spp.)
Plerogyra sinuosa	*Montipora* spp.	TOTAL: 13 genera
Pocillopora damicornis	*Mycedium elephantotus*	
Polyphyllia talpina	*Oxypora* sp.	**Subclass Corallimorpharia**
Porites compressa	*Pachyseris speciosa*	"Corallimorphs"
Seriatopora hystrix	*Pavona* spp.	*Antipathes* spp. (2 spp.)
Stylophora pistillata	*Pectinia* spp. (2 spp.)	*Cirrhipathes anguina*
Turbinaria sp.	*Platygyra lamellina*	*Discosoma* sp.
Zoopilus echinatus	*Plerogyra sinuosa*	*Ricordea yuma*
TOTAL: 21 genera	*Pocillopora* sp.	TOTAL: 5 genera
	Porites spp. (5 spp.)	
	Seriatopora hystrix	
	Stylophora pistillata	
	TOTAL: 25 genera	
1992 GRAND TOTAL: 21 genera	**1999 GRAND TOTAL: 43 genera**	

of demand from home aquarists, but rather a lack of information regarding the option of obtaining cultured corals?

Obviously, it is easier for collectors to obtain corals from the wild and therefore only a handful of collectors actually culture corals. Time and energy must be invested in such a project, and ultimately it has to be profitable for the collector. How can we provide the option of cultured corals to home aquarists, yet make it advantageous to the collectors? These questions remain to be answered. However, we in the public aquarium industry must explore ways to support this important process.

Education is of course paramount to this endeavor. By procuring cultured corals, public aquariums can promote such programs elsewhere. Even better, the establishment of in-house

Table 3. Public aquariums surveyed**.**

Institution	**# yrs. propagating**	**# frags produced/yr.**	**# frags donated/yr.**
Aquarium of Pacific	1	50	30
Florida Aquarium	2	15	0
Living Seas	2	10	0
New England Aquarium	4	100	40
Waikiki Aquarium	15	lots	lots
Birch Aquarium	3	60	50
Steinhart Aquarium	1.5	50	25

Table 4. Some sources of coral reef degradation (Green and Shirley, 1999)

Reason for coral extraction	**Location**	**Amount per year (tonnes)**
Dead and Live coral trade (Green and Shirley, 1999)	All areas	1000
Lime (Cesar, 1996)	West Lombok	1600
Coral mining - 1998 (Bentley, 1998)	Indonesia?	5,000
Coral mining - 1988 (Brown and Dunne, 1988)	Maldives	25,000
Coral mining - 1983 (Polunin, 1983)	Indonesia	15,000-37,500
Dynamite fishing (Cesar, 1996) (Spalding and Grenfill, 1998)	Indonesia	52,450,000

Table 5. Average stock of cultured corals at selected wholesalers.

Wholesaler	**Average % of cultured corals in stock**	**Reasons for reduced stock**
1	0	Lack of retail demand; lack of quality (too small, no color)
2	<1	Cost too high; lack of quality (not lg. or colorful enough)
3	0	Cost too high; high mortality upon receiving shipment
4	0	Cost too high; lack of retail demand
5	1	Lack of retail demand; lack of quality (too small, no color)

coral propagation programs can provide cultured corals to additional institutions. Presently, the Aquarium of the Pacific has distributed corals to other aquariums in the hopes of decreasing the demand for wild caught corals. Questions arise however regarding the future of such programs. Can public aquariums do more than simply supplying cultured corals to other aquariums? What about providing wholesalers and marine aquarium societies with cultured corals? Can public aquariums combine efforts to create coral banks so that each facility can focus on culturing a few specific species in order to reduce repetition and increase production? What about assisting foreign collectors in establishing culture facilities in order to help local economies and promote coral reef conservation?

Much remains to be done in these areas. With the increasing probability of a ban on wild-caught coral exports, the time to focus on propagation endeavors is now. Public aquariums must collaborate with hobbyists, wholesalers, and themselves in order to coordinate and maximize the efficiency of coral propagation. The husbandry of corals begins with such efforts for the present and future.

References:

Bentley, N. 1998. An overview of the exploitation, trade and management of corals in Indonesia. *TRAFFIC Bulletin*, 17 (2): 67-78.

Brown, B. E. and R. P. Dunne. 1988. The impact of coral mining on corals reefs in the Maldives. Environmental Conservation, 15: 159-165.

Bryant, D., L. Burke, J. McManus, and M. Spalding. 1998. *Reefs at Risk, A Map-Based Indicator of Threats to the World's Coral Reefs.* World Resources Institute, Washington D. C. 56pp.

Carlson, B. A. 1989. Living Coral in the Aquarium. AAZPA 1989 Annual Conference Proceedings: 237-241.

Catala, R. L. A. (1964). *Carnival sous la mer*. Paris: Les Editions R. Sicard.

Cesar, H. 1996. Economic analysis of Indonesian coral reefs. The World Bank, Washington D. C., USA. p. 97.

Green, E. P. and F. Shirley. 1999. *The Global Trade in Coral*. World Conservation Monitoring Centre. World Conservation Press, Cambridge, UK. 70pp.

Polunin, N. V. 1983. The marine resources of Indonesia. Oceanography and Marine Biology Review 21: 455-531.

Spalding, M. and A. Grenfill. 1998. New estimates of global and regional coral reef areas. Coral Reefs 16: 225-230.

Yates, K. R. and B. A. Carlson. 1992. Coral in Aquariums: How to Use Selective Collecting and Innovative Husbandry to Promote Reef Conservation. Proceedings of the Seventh International Coral Reef Symposium, Guam, 1992: 1091-1095.

BRINE SHRIMP HOLDING

Mark Schick

John G. Shedd Aquarium

The maintenance of large stocks of adult brine can be problematic. Although *Artemia* sp. are quite forgiving with respect to water quality, the goal should be to keep live food as healthy as possible; thus limiting the potential problems that can be passed on to the animals being fed. This is usually accomplished by massive water changes or daily cleanings of filtration systems. Both methods have drawbacks in the form of costly salts or time-consuming upkeep.

The opening of the Seahorse Symphony exhibit at the John G. Shedd Aquarium required large amounts of live brine to use as a bulk food item. In an attempt to sustain the brine in a small amount of space a new holding area was engineered. This new holding unit maintains 10 boxes (10 wet pints) of live brine in an area that takes a little more than 40 square feet. The unit employs a biological filter, UV sterilizer and foam fractionator.

The holding area consists of a preformed kidney-bean shaped fiberglass pond that can hold approximately 180gallons. The preformed rounded sides allow for good water circulation and unobstructed movement of the brine. The holding pool feeds into an oversized sump via a 1 ½" bulkhead installed in the approximate center of the pond. Threaded into the top portion of the bulkhead is a 1 ½" male PVC adapter. The adapter is used to hold an 8" standpipe on the female side and an acrylic box with a hole cut into the center. More on the acrylic box in a moment.

The bottom of the pond is approximately 2' off the floor, supported by a 4'x 8' sheet of plywood on top of cinder blocks. Due to space and material restraints the bulkhead from the holding pool is plumbed directly to an oversized sump, which is placed to the left of the holding pool. If the sump could have been placed under the pool the entire system would take only 32 square feet of space. Inside the sump there are two pumps that run the filtration set-up. The main system pump runs a 3' venturi foam fractionator, flow through twin tube 30watt UV sterilizer, and a bypass line that feeds directly to the main holding pool. The bypass line to the holding pool remains closed except to prime the pump. The second pump runs the biological filter, which is a five-gallon bucket filled with bio-media.

As mentioned earlier, the PVC fitting holds an acrylic box in place. This is what makes the system work. The box is made of ¼" acrylic and forms a rectangle that is 17" tall and 15" wide on each side. The box is left open on top. The center of the bottom piece has a hole cut in it so the 1 ½" PVC adapter male side can fit through, but the female side holds it in place against the bulkhead. Each of the four sides of the box have two horizontal rectangular pieces cut out, each of these cut outs measures 9"x 3". The bottom of the lower cut out is 3" off of the bottom piece of acrylic and the second cut out is 2" above the top of the first cut out (see figure 1). These cut outs are centered on the acrylic walls and sized to fit Whisper® 1000 sponges. The 4 sides and bottom piece are glued together to form the open toped box and allowed to dry. Small triangular pieces of acrylic can be glued into the top corners for added support (see figure 2).

The acrylic box is now screwed into the bulkhead and a Whisper® 1000 sponge is inserted into each of the eight cutouts. Water will now be able to pass through the box without trapping the *Artemia*. The water level is maintained at mid-level of the top sponge. This allows for surface extraction along with diffused water flow that keeps animals off of the sponges.

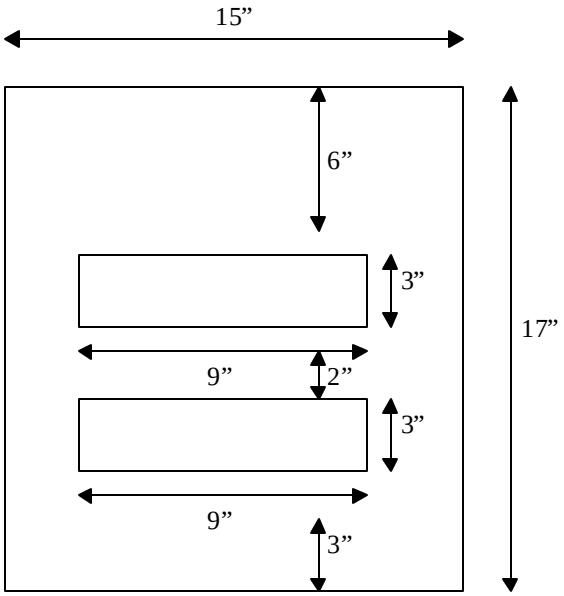

Figure 1. Side view

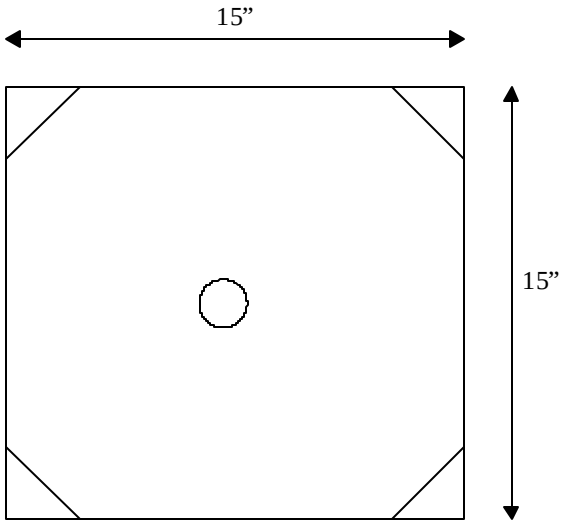

Figure 2. Top view

Maintenance on the system consist of a once a week cleaning that takes less than 30 minutes. After catching any remaining brine, the entire system is drained of water. The acrylic box is removed and the sides of the holding pool are scrubbed lightly to remove any loose debris then rinsed with freshwater. The sponges are removed, rinsed clean in the sink and then placed in a mild bleach solution (10mL/4gallons of fresh water) to soak for a day or two. It is important to mix the bleach and water before putting the sponges in, or the bleach can melt the sponges. A second set of clean sponges is put in place in the acrylic box and the system is refilled.

In all, the system runs on approximately 175 gallons of water at a salinity of 20 ppt. The water for this system is taken out of a saltwater system and diluted with tap water, thus recycling water that would otherwise be dumped. The system has two large air stones for movement and a flow-through rate of 2.0-2.5 gpm. This flow-through rate could be increased, but may affect the success of the UV sterilizer in reducing bacteria levels. (Shedd currently is maintaining seven boxes per shipment at this flow rate to reduce bacteria levels. Higher *Artemia* concentrations were not tested.).

Since the adult brine are from an external source, there was concern about what outside pathogens could potentially spread to the collection. To get a baseline of what bacteria are present, water from multiple shipments was tested. All of the samples registered bacterial levels too numerous to count. Once the samples were diluted, plated out and identified, at least six different species of *Vibrio* were seen (including *V. alginolyticus* and *V. parahemaliticus*) along with a variety of other bacteria. When a UV sterilizer is running effluent from the unit shows no *Vibrio* and the holding water shows a

reduced load of bacteria (see table 1). Currently the flow rate is being increased in hopes of eradicating all *Vibrio* from the water column.

Table 1. Concentration of *Vibrio sp*. per 100 ml. BSH = water sample taken from holding water, UV = water sample taken from UV effluent.

Date	**BSH**	**UV**
1/15/2000	85,000	0
1/18/2000	0	0
1/20/2000	70,000	0
1/25/2000	5,000	0
1/29/2000	200,000	0
2/5/2000	135,000	0
2/12/2000	10,000	0
2/15/2000	0	0

Current experiments to extend the usage time of the water have met with varying success. The system can be successfully maintained for two weeks without a water change until the sponges begin to block. The two-week running time has not been tested for bacterial levels, but mortality levels increase during the second week.

The only problem with this system occurs when the brine shrimp make it past the sponges. This will happen if care is not taken when putting clean sponges in place, or if they are moved out of position while capturing the *Artemia*. When this occurs the pumps slow and bacteria levels rise considerably. Other than the once a week cleaning the system will need little to no maintenance if the sponges are properly installed.

This method of upkeep has allowed us to reduce our loss of brine and decrease our time and effort in the maintenance of live brine. The system runs cleaner and more smoothly than others we have tried. In fact, Shedd currently is working with enrichment of the brine in this system so they can be fed out right from the holding basin.

CULTURING THE UMBRELLA JELLY, *EUTONINA INDICANS* (ROMANES, 1876)

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Introduction:

So you or your institution has been successfully culturing the moon jelly, *Aurelia aurita.* The visitors love the jellies and you are thinking that it might be nice to display another type of jellyfish. One species you might want to consider is *Eutonina indicans* (Romanes, 1876), commonly known as the umbrella jelly (Figure 1). While they don't get that big (25–35mm), they are one of the easiest jellies to culture, are relatively hardy, and make a beautiful display animal. Umbrella jellies are from the class Hydrozoa, subclass Hydromedusae, order Leptomedusae, and family Eirenidae. Hydromedusae tend to be small clear jellies. This species is found nearshore in the eastern Pacific from at least Santa Barbara to Vancouver Island, Aleutian Islands, Bering Sea, Kamchatka, and Hokkaido (Wrobel and Mills, 1998). However, the umbrella jelly is probably a circumboreal species having been described in the North Sea, Japan, and Mexico (Reese, 1975).

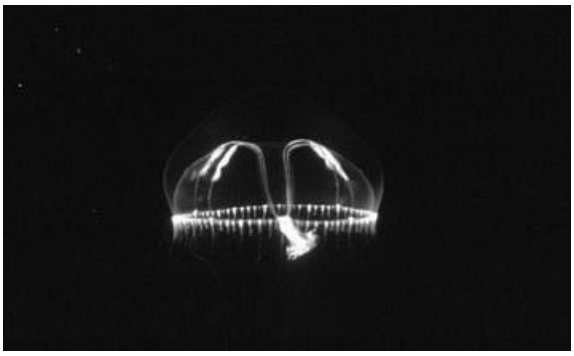

Figure 1: The umbrella jelly, *Eutonina indicans.* Photo by Dave Wrobel ©

While the visual impact of exhibiting a single umbrella jelly may not be great, you can create a nice effect with numerous individuals in a large or small display tank. Due to the ease of culturing these jellies, they are readily available to exhibit. In addition, if you already have a culture of moon jellies, there are educational benefits from culturing *Eutonina*. Maintaining cultures of both species allows you to show visitors and students examples of Scyphozoan and Hydrozoan polyps.

The life history of umbrella jellies and other Leptomedusae includes an alternation of generations between an asexual polyp generation and a sexual medusa generation. The sessile, polyp stage of *Eutonina* has two types of polyps: gastrozooids, which are feeding polyps, and gonozooids, which are reproductive polyps (Figure 2). Tiny medusae escape from the distal, open end of the gonozooid. These medusae are produced asexually but contain the gonads for sexual reproduction and are either male or female (gonochoristic). Mature medusae free-spawn either eggs or sperm into the water where fertilization occurs. Ciliated planulae develop from the fertilized eggs. The planulae swim around for a short time, then settle out to initiate the new polyp generation. One of the reproductive advantages of

Figure 2: Eutonina hydranths. A gonozooid (reproductive polyp) is on the left and a gastrozooid (feeding polyp) is on the right.

Eutonina (as well as other Leptomedusae and certain Scyphozoans) is that there is an indefinite asexual reproduction of both polyps and medusae by budding (Nybakken, 1996; Pearse et al., 1987). Once you solidly establish the hydroid phase you can obtain the umbrella jelly continuously and in great numbers.

Materials:

- running seawater (~15° C)
- plastic aquariums (like inexpensive hamster cages)
  large (15″x8.5″x9.75″)
  medium (12″x7.5″x8″)
  small (9″x6″x6.75″ or 7″x4″x5.5″)
- finger bowls
- glass microscrope slides (optional)
- eyedropper
- in-line pool filter
- 5 um filter cartridge
- Nalgene tubing®
- 200 um Nitex® Mesh
- 1 mm Nitex® Mesh
- Popsicle® sticks
- drill with quarter inch bit
- silicone sealant (must be appropriate for long immersion in seawater like Dow 999 clear)
- rigid airline tubing or glass pipet
- flexible airline tubing
- pvc pipe
- water table

Methods:

"Let'em steep"

To start a culture of *Eutonina indicans* it is best to get 5–20 wild-caught animals. You can either collect this species yourself or have specimens shipped to you. Put about 6–10 mature umbrella jellies in a large finger bowl filled with seawater and let them sit overnight in a water bath. If the adult *Eutonina* have either sat for a long time after collection or have been shipped to you, it is also a good idea to set aside the collection or shipping seawater for a day to see if the adults have already spawned during transport.

After 24 hours, remove the medusae. Place the finger bowl under a dissecting scope and look for planulae or fertilized eggs. You should see a number of white planulae moving on the bottom of the finger bowl. If you do not see planulae but do see eggs, you might want to let the water steep another day to see if you get a late hatch. If there is a high density of planulae (greater than 50), move them into other finger bowls with fresh seawater using an eyedropper. If desired you can also put in glass slides for the planulae to settle on. Let these finger bowls sit for several days. The planulae do not need to be fed or the water changed until they settle out (usually within 7 days). Instead of using mature medusae to produce your polyp generation, you can also have a colony of hydroids shipped to you or start new cultures of *Eutonina* by moving a sample of the hydroid colony to another tank.

Building the factory:

To make the hydroid polyp container, drill a quarter-inch hole about 2 inches below the lip in the center on one side of a medium or large plastic aquarium. Insert about an inch of Nalgene® tubing to make an exit spout for newly produced medusae to drain through. On another side of the tank drill 2–4 holes about an inch below the lip of the tank to accommodate emergency drainage.

Next prepare a catch basin for the newly released medusae. In order to culture delicate jellies such as these, tanks need to be modified to prevent medusae from getting drawn into or against the drain. Many holding and culture tanks are made from modifying square tanks (Sommer, 1995). Take a small plastic aquarium and drill a few drainage holes about half an inch below the lip of the tank. On the same side attach a piece of 200 µm Nitex® mesh with silicone across the drain end. As has been described in previous papers, this is a design modification that is important in keeping jellies alive. Installing mesh across one side of a plastic aquarium maximizes the drain area and insures that the pull from the drainage is spread across a large area. This feature ensures that the jellies don't get pancaked against the screen (Sommer, 1993)!

Installing the screen:

This next section is a rather long aside on the best way to silicone Nitex® mesh or a similar type of screen across one side of the tank in front of the drain. I have found that this system decreases the amount that the silicone fights back.

1) First, measure the Nitex® mesh so that it covers the entire drain end of the tank plus about 2 inches for glue flaps. Looking down on the tank, the mesh should be wide enough to curve slightly out toward the drain.
2) Figure out where you want your screen to be placed. The edge of the screen should not be touching the drain. Use a sharpie to draw vertical lines where the edge of the mesh will be siliconed.
3) Using the drawn lines as a guide, put a vertical bead of silicone on each side of the tank.
4) Carefully, place the vertical sides of the mesh on top of the silicone bead so that the mesh fits inside the tank with the bottom part of the screen touching the bottom of the tank. Be careful not to pick up some of the silicone onto the center of the Nitex® mesh. This will dry and clog the screen.
5) Using a Popsicle® stick, start at the bottom and gently press the mesh into the silicone and move the Popsicle stick up to the lip of the tank. Repeat this process several times until the side of the mesh is firmly embedded in the silicone and the silicone is as smooth as possible. It may also help if the stick is wetted with soapy water.
6) Use the same technique to imbed the other side of the Nitex® mesh into the silicone.
7) Let this dry overnight. (If you are extremely adept, you can skip this step and attach the bottom at the same time you are attaching the sides. But it is at this point that I start to get silicone on my fingers, the mesh, the plastic tank, and especially in my hair. So I let the two sides dry first.)
8) Squeeze another bead of silicone between the bottom of the mesh and the bottom of the tank.
9) Again, using a Popsicle® stick, gently press in the silicone so that the bottom of the screen adheres to the bottom of the tank using the same technique as in step 5.
10) Let the tank dry overnight.

<u>Back to live organisms:</u>

As mentioned previously it usually takes about a week for *Eutonina* planulae to settle and develop into hydroids. However, after the planulae have developed from eggs, you should check daily

for primary polyp settlement. Once you see that most of the planulae have metamorphosed into at least a primary polyp, transfer the finger bowls or glass slides into the hydroid polyp container that you prepared earlier and place in a water table for drainage. Fill the hydroid container with seawater and add a trickle of ~ 15° C make-up seawater. The flow rate of the seawater should be high enough to overflow through the tubing in the primary hole. However, be sure the flow rate is not high enough to drain out the emergency drain holes on the other side or you will lose medusae. Since *Eutonina* hydroids are easily fouled by other hydroids and diatoms, use extra filtration for your seawater source. It is best to hook up an in-line pool filter with a 5 µm filter cartridge to the inlet into the hydroid polyp tank. You now have the primary production site for *Eutonina* medusae. Feed a small amount of enriched brine shrimp nauplii or rotifers to the hydroid tank daily.

After two months, add the catch basin to the running water setup (Figure 3). It takes 40 days after a planula settles to see the release of the first medusa (Reese, 1975). *Eutonina* hydroids gradually grow and spread out along the finger bowls or slides producing both types of polyps (feeding and reproductive). We have found that it takes a couple of months before sufficient numbers of medusae are produced. But once the medusae are budding off in sufficient numbers, the culture setup is designed to allow new medusae to wash out of the hydroid polyp tank and into the small catch basin tank. Here the medusae are safe from predation by the hydroid generation (Sommer, 1993).

Figure 3: Hydroid polyp culture setup for *Eutonina indicans.* A seawater inlet with a 5 µm pre-filter trickles into the hydroid polyp tank. Medusae pop off from the hydroid colony and are washed into the catch basin.

Designing the grow-out system:

Now you will want to set up grow-out tanks for the growing medusae. It is easiest and most productive to move batches of medusae from the catch basin to grow-out tanks and then to the display tank. Depending on how much room you have for your culture, prepare one or two plastic grow-out tanks by drilling drainage holes and attaching a screen with silicone across the entire drain end (see above). Choose the mesh size according to the size of the medusa. Newly produced *Eutonina* medusae require 10–14 days of daily brine shrimp nauplii feedings before they are large enough not to slip through 1 mm mesh and are easily visible (especially after feeding) to the naked eye. Add a water inlet (using Nalgene tubing) and a rigid airline to each tank. Place the rigid airline next to the screen to keep jellies away from it and add seawater on the opposite side. Set up the flow such that the jellies gently circulate without grouping in any part of the tank. The use of small pvc rings are a great device to keep the airline tubing and water inlet where you want them. The PVC diameter does not matter, the length should be as small as can be cut, and the ring should be open at one end. Place the ring around the inlet or airline tube and onto the side of tank to keep the tubing in place.

At the Monterey Bay Aquarium, we have several cultures of *Eutonina indicans*. One culture utilizes a three-step grow-out system to produces adult umbrella jellies for a "bubble" tank in the Tiny Drifters gallery. A hydroid polyp tank pops off medusae into a small catch basin. When the medusae are at least 5 mm in diameter they are transferred individually to a medium size grow-out tank. Here they grow until they are roughly 20 mm. Then the medusae are transferred individually to the display tank. The entire culture covers a 2 ft by 1.5 ft area and fits inside a plastic tray, plumbed to serve as a wet table.

The system that I favor is a five-step grow-out system used to supply umbrella jellies for our Crystal/Umbrella jelly display. The 84-gallon kreisel serves as an interesting interactive that exhibits how some gelatinous animals camouflage themselves by having a clear body. When visitors press a button the side lighting turns off and the displayed hydromedusae seem to disappear. The main species for this tank is *Aequorea victoria* and the backup species are *Eutonina indicans* or *Mitrocoma cellularia*. We have displayed *Eutonina indicans* since the spring of 2000 because this species has been the easiest to culture and has supplied a high enough density to fill the 6-ft diameter kreisel.

The 5-step grow-out system easily supplies the needs of the display and leaves extra medusae as backup, to feed other jellies, or as possible trade animals. There are two hydroid polyp cultures. Every 7–14 days, the entire contents of either one or both of the culture catch basins are moved into a medium grow-out tank. This grow-out tank is fitted with 200 µm to ensure that the medusae are large enough not to get sucked through the screen. Once the medusae are at least 5 mm and easy to see, they are moved to a large grow-out tank with 1 mm mesh (about a week). After another week they are moved into a pseudokreisel. The display kreisel is re-stocked using the jellies from the pseudokreisel. The jelly culture setup is like a batch production line. Different-sized umbrella jellies are constantly being moved to larger grow-out tanks until they are big enough to display.

The captive lifespan of the umbrella jelly is approximately 2-4 months depending on how carefully they are handled while on display. One thing that has eluded us so far is to produce a second (F2) generation of *Eutonina indicans* from wild-caught animals.

Maintenance of cultures

As described in Sommer, 1992, the most important aspects of jelly culture are the "seawater system, tank designs, feeding, and tank cleaning." Each different species of jelly culture has different primary care concerns. Not every species of polyps requires addition of a 5 µm pre-filter. But *Eutonina* tends to be extremely sensitive to contamination so it is highly recommended. Each culture and grow-out tank of *Eutonina* needs a daily feeding of enriched brine shrimp nauplii. The hydroid polyp cultures need to be cleaned on a regular basis. Removal of diatoms and other polyps is very important. Cleaning can be done using a turkey baster. The hydroid colonies can be removed from water for a short time and gently basted to remove diatoms and other debris. To remove other types of polyps (such as *Aurelia* or the fouling hydroid, *Bougainvillia*) use a small scraping tool such as plastic tweezers or a fingernail. As you move the *Eutonina* to larger grow-out tanks, be sure to clean and rinse out each tank before adding new jellies.

After a certain amount of time (3 months to a year), depending on how well the hydroid polyp tank is maintained and the strength of the hydroid colony, you may notice a drop-off in medusae production. For instance, the MBA's 1999 *Eutonina* hydroid culture produces a very small fraction of what the April 2000 hydroid culture can produce. Furthermore, when the culture from April 2000 was 7 months old, it produced about half as much as it did when it was 3 months old. Although you can experiment with abiotic induction of medusae production (factors such as temperature, light, and water chemistry can be manipulated), when an *Eutonina* culture stops producing enough medusae it is best to start a completely new culture.

Conclusions:

While jelly culture and exhibitry may seem like a relatively new field, scientists and aquarists have been working with gelatinous animals for a long time. Culture work on *Eutonina* was first begun in 1897, although reproductive colonies were not established until 1968 (Reese, 1975). The Enoshima Aquarium in Japan has been displaying jellies for over 35 years (Powell, 2000). In order to open our first temporary jellyfish exhibit in 1992, the Monterey Bay Aquarium first dedicated significant effort to the display and culture of jelly species in the late 1980s. However, the popularity of displaying Cnidarians and Ctenophores has exploded in the last decade. Numerous aquariums have either recently opened a jelly gallery or will be opening one in the near future. In fact, in addition to our permanent Drifters gallery which opened in 1996, the Monterey Bay Aquarium plans to open a temporary exhibition of jellies in May 2002. Jellyfish displays, especially *Aurelia*, have even become popular for some dedicated hobbyists.

The most interesting thing about culture work with any type of jelly is figuring out what works and doesn't work. The system described above to culture *Eutonina indicans* has enabled us at MBA to supply the needs of our display with ease and a minimum of maintenance. However, the umbrella jelly can also be cultured by setting up a regular system of water changes instead of using flow-through systems (see Reese, 1975). Just because one system works for one species or institution doesn't mean it will work for another. Sometimes medusae production will be a "piece of cake" for a certain species, other times you will need to start a culture again and again. Even the cultures themselves are constantly evolving as they age. Katrina Cross, the present head of the Monterey Bay Aquarium Drifters gallery recently sent us jellyheads an e-mail offering the following insights:

<u>Things I have learned:</u>

- If the water flow is up too high anything will go through the screen.
- When in doubt use smaller screening.
- When in doubt raise in a watchglass.
- Polyps will re-settle out if given time and fresh water every now and again (and some food).
- Humming Barry White to sperm and eggs seems to get them "in the mood."
  (Katrina Cross, 2000, personal e-mail)

Jellyfish culture is a very dynamic process so the more methods you know the better chance that you will get the results that you want. Even if you have never worked with gelatinous zooplankton, you

can rear jellies with a little time, patience, attention to detail, and maybe humming a few bars of Barry White!

Acknowledgments:

I would like to thank Dave Wrobel and Katrina Cross at the Monterey Bay Aquarium. While I may have written this paper, almost all the methods I have described come from their extensive knowledge of and experience with gelatinous zooplankton culture.

References:

Nybakken, J.W., 1996. Diversity of the Invertebrates, a Laboratory Manual, Pacific Coast Version. California State University, Hayward and the Moss Landing Marine Laboratories, Wm. C. Brown Publishers, pp. 28-30.

Pearse, V., Pearse, J., Buchsbaum, M., and Buchsbaum, R., 1987. Living Invertebrates. Boxwood Press and Blackwell Scientific Publications, Inc., pp. 133-141.

Powell, D.A., 2000. Jellies…A sense of Wonder. Communique, American Zoo and Aquarium Association, Nov 2000.

Rees, J. T., 1975. Studies on Hydrozoa of the Central California Coast: Aspects of Systematics and Ecology. PhD thesis, Zoology, University of California, Berkley.

Sommer, F. A., 1992. Husbandry aspects of a jellyfish exhibit at the Monterey Bay Aquarium. American/Canadian Association of Zoological Parks and Aquariums Annual Conference Proceedings, pp. 362-369.

Sommer, F. A., 1995. Jellyfish and Beyond: Husbandry of Gelatinous Zooplankton at the Monterey Bay Aquarium. Proceedings of the Third International Aquarium Congress, April 25-29 1993, pp.249-261. New England Aquarium.

Wrobel, D., and Mills, C., 1998. Pacific Coast Invertebrates A guide to the Common Gelatinous Animals. Sea Challengers and Monterey Bay Aquarium.

BAT STAR (*ASTERINA MINIATA*) WASTING DISEASE

Nancy Lightowler Caruso, Aquarist

Long Beach Aquarium of the Pacific

The Bat star, *Asterina miniata*, is very common in the Pacific Ocean from Alaska to Baja California. It is also very common in public aquariums where they are usually on display in touch tanks. At the Aquarium of the Pacific, Bat stars are displayed in temperate (15°C) as well as cold water (10°C) exhibits. These exhibits range in size and include touch tank systems. Over the past year, without an apparent cause, a strange Bat star "meltdown" has resulted in several mortalities. The onset of this problem is sudden and appears as a necrotic area usually on one of the arms of the star. It spreads quickly and usually within 48 hours, the animal expires.

After doing some quick research, I have found that a similar problem is commonly associated with the occurrence of El Nino in the Pacific Ocean. It has been reported that during these periods of warmer water temperatures, Bat stars either migrate into colder, deeper waters or "succumb to a wasting disease" (National Park Service, 1997). Additionally, the disease was noted to occur in *Pisaster giganteus* (Giant Spined sea star) and in *Pycnopodia helianthoides* (Sunflower star). The cause of the "wasting disease" is thought to be a new species of *Vibrio* but this has not yet been confirmed through DNA analysis (J.Dixon, pers. comm.).

After talking with several other aquariums, I have concluded that the "wasting disease" seems to be widespread in public aquaria. In both flow-through and closed systems and in cold water and temperate systems, the disease was noted to have occurred. In all cases, it seems the trigger for the onset of the meltdown is stress. All of the aquariums kept Bat stars in touch tank systems, which are inherently stressful due to people touching the animals. A few of the aquariums kept bat stars in flow through systems, which are subject to stress from seasonal water temperature changes. Bat stars seem to prefer temperatures in the low to mid 50's (F) and tend to be vulnerable to the disease at temperatures above 57 F. Other aquariums noted the disease to also occur in *Pisaster giganteous*, *Pycnopodia helianthoides,* and *Mediaster aequalis.*

Keeping the animals at lower temperatures and out of touch tanks seems to be the answer to the disease problem. However, at the Aquarium of the Pacific, bat star mortalities (due to the wasting disease) have been curbed by implementing a Spectrogram treatment. At first sign of the disease, the affected animals begin a daily, 2-hour, 132g/L, Spectrogram bath and their progress noted. The results have been positive. The rapid progression of the disease has been slowed and over a period of 1–4weeks, the animal's appearance actually improves enough to be put back on exhibit. This procedure has been successful on all infected batstars as long as the treatment is started at first sign of the disease.

References

National Parks Service Annual Report, *Inventory and Monitoring Program: Kelp Forests in Channel Islands National Park, California.* 1997

Dixon, John Ph.D., California Coastal Commission, Personal Communication. 10/2000

NAME CHANGE OF GIANT PACIFIC OCTOPUS

Roland C. Anderson

The Seattle Aquarium

The scientific name of the giant Pacific octopus has been changed. It formerly was *Octopus dofleini* (Wülker, 1910), but an eminent octopus taxonomist, Dr. F. G. Hochberg, has changed it to *Enteroctopus dofleini* (Wülker, 1910). Note that the part in parenthesis refers to the original describer of the species. This practice is used in scientific literature. The describer's name is put into parentheses if the animal has been redescribed (had its name changed); otherwise it is in normal letters with a comma after the name.

The name was changed by Dr, Hochberg for several reasons. There are three known species of giant octopuses, *E. magnificus* (Villanueva and Sanchez, 1992) found off South Africa, *E. megalocyathus* (Gould, 1852) found off southern South America, and *E. dofleini*, found in the north Pacific. These species are now placed in the same genus because of their size. The term *Enteroctopus* has precedence (was used first) for any of the giant species and hence should be used. In addition, the number of gill lamellae differ significantly from those in *Octopus*. In lieu of DNA analysis, which has yet to be done, most cephalopod workers are now using the new name, as is the Seattle Aquarium.

It has been suggested that the common name of *E. dofleini* should be North Pacific Giant Octopus (Hochberg, 1998; Turgeon, et al., 1998), to distinguish it from the South American and South African species. I believe most aquariums and zoos continue to use "giant Pacific octopus," with some basis for doing so. *E. dofleini* is still the largest species of octopus in the world. It can grow to over 200 kg (High, 1976; Newman, 1994). I believe such a distinction as the largest species allows us to use the term "giant Pacific octopus" with impunity. The other reason for maintaining use of this name is that it is the common name now used. If there were any doubt over which giant octopus was which, for example, if an aquarium was lucky enough to display several species of giant octopuses, the distinction could be made with their scientific names. I believe the term "giant Pacific octopus" will continue to be used.

References:

High, W.L. 1976. The giant Pacific octopus. Marine Fisheries Review. 38:17-22.

Hochberg, F.G. 1998. Class Cephalopoda. pp 175-236. In: Scott, P.V. and J.A. Blake. Taxonomic atlas of the benthic fauna of the Santa Maria Basin and the western Santa Barbara Channel, volume 8. Santa Barbara Museum of Natural History. 250 pp.

Newman, M. 1994. My life in a fishbowl. Douglas and McIntyre, Vancouver. 262 pp.

Turgeon, D.D., J.F. Quinn, Jr., A.E. Bogan, E.V. Coan, F.G. Hochberg, W.G. Lyons, P.M. Mikkelsen, R.J. Neves, C.F.E. Roper, G. Rosenberg, B. Roth, A. Scheltema, F.G. Thompson, M. Vecchione, and J.D. Williams. 1998. Common and scientific names of aquatic invertebrates from the United States and Canada: molluscs, 2nd edition. American Fisheries Society, special publication 26 (Bethesda, MD). 526 pp.

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A BRIEF GUIDE TO AUTHORS

As always, Drum & Croaker articles are not peer reviewed and content will not be edited. Other types of contributions may be edited to meet space limitations. **The approximate deadline for submissions is November 1st.**

Computer files are preferred. All files or disks (now including Apple files) can be sent directly to: Pete Mohan, SeaWorld Cleveland (formerly known as Sea World of Ohio), 1100 Sea World Drive, Aurora, OH, USA 44202. **Please save all documents as Word 7.0 for Windows 95 files** where possible. I can open most other word processing files of **similar or older** vintage. **My E-mail address is pete.mohan@anheuser-busch.com.** Please send a 3.5" floppy diskette if e-mail isn't available. As a final resort you can send typed manuscripts in Times 12pt font.

1. "Regular" style articles: should normally follow the following basic format:
   TITLE (boldface, capitals & centered)
   one and one half space
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   double space
   Text: single spacing with 1" margins. Please indent using a .5 inch tab stop at the beginning of each paragraph and double space between paragraphs. Section headings should be in bold (but not all caps) at the left margin.
   Figures: we can print black & white photographs.

2. Optional "Journal" style articles: (guidelines provided by George Benz, Tennessee Aquarium) [also see his article, "Morphology of the Fish Louse" in Volume 27]. This can be used to give your article a more formal appearance suitable for reprints. For a copy of the directions for this format please contact me at the above street or E-mail addresses.

3. Short contributions ("Ichthyological notes") are any articles, observations, or point of interest that are less than 1⅛ pages in length. A brief bold faced and capitalized title should be centered, text should be single spaced, and author and affiliation should be placed at the end of the piece with the left end of each line at the center of the page. Reformatting to meet guidelines for margins, etc. may reduce a shorter "main" article to a note.

4. Reviews, abstracts, translations and bibliographies are welcome.

DRUM AND CROAKER ARCHIVE PROJECT

Progress on, and problems with, the archiving process continue. Most issues from the 50s and early 60s are now in electronic format. Barbara Chauhan, our Zoology Administrative Assistant here at SeaWorld Cleveland, has helped retype many of the old issues. These came to me as old faded Xeroxes of old faded mimeographs, and were unrecognizable by OCR software. Not surprisingly, a good typist works faster than the OCR software installed on my old Pentium 1 at home.

ACKNOWLEDGEMENTS

Barbara Chauhan helped prepare hard copies of this issue for mailing and coordinated other mailings. My family also helped assemble any hard copies of this volume. Leigh Woodall formated the pdf version. Once again, I'd like to extend my special thanks to all those who contributed articles to this issue.

"NOT RESPONSIBLE"

The content of original manuscripts submitted to *Drum and Croaker* is not edited (except by express request of an author). I will fix obvious typographical errors if I find them. Articles not sent on disk or by E-mail are retyped which may lead to transcription errors, so please send all documents as PC files when possible.

Figures may be reduced to save space, and photos, tables, and figures not referred to in the text may be omitted for the same reason.

Announcements may be edited where they are needed as filler at the ends of articles.

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