JELLYFISH
(CNIDARIA/CTENOPHORA)
CARE MANUAL

CREATED BY THE
AZA AQUATIC INVERTEBRATE TAXON ADVISORY GROUP
IN ASSOCIATION WITH THE
AZA ANIMAL WELFARE COMMITTEE
Jellyfish Care Manual
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Disclaimer: This manual presents a compilation of knowledge provided by recognized animal experts based on the current science, practice, and technology of animal management. The manual assembles basic requirements, best practices, and animal care recommendations to maximize capacity for excellence in animal care and welfare. The manual should be considered a work in progress, since practices continue to evolve through advances in scientific knowledge. The use of information within this manual should be in accordance with all local, state, and federal laws and regulations concerning the care of animals. While some government laws and regulations may be referenced in this manual, these are not all-inclusive nor is this manual intended to serve as an evaluation tool for those agencies. The recommendations included are not meant to be exclusive management approaches, diets, medical treatments, or procedures, and may require adaptation to meet the specific needs of individual animals and particular circumstances in each institution. Commercial entities and media identified are not necessarily endorsed by AZA. The statements presented throughout the body of the manual do not represent AZA standards of care unless specifically identified as such in clearly marked sidebar boxes.
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</table>
**Introduction**

**Preamble**
AZA accreditation standards, relevant to the topics discussed in this manual, are highlighted in boxes such as this throughout the document (Appendix A).

AZA accreditation standards are continuously being raised or added. Staff from AZA-accredited institutions are required to know and comply with all AZA accreditation standards, including those most recently listed on the AZA website (http://www.aza.org) which might not be included in this manual.

**Taxonomic Classification**

Table 1. Taxonomic classification for jellyfish

<table>
<thead>
<tr>
<th>Classification</th>
<th>Taxonomy</th>
<th>Additional information</th>
</tr>
</thead>
<tbody>
<tr>
<td>Kingdom</td>
<td>Animalia</td>
<td></td>
</tr>
<tr>
<td>Phylum</td>
<td>Cnidaria</td>
<td></td>
</tr>
<tr>
<td></td>
<td>Ctenophora</td>
<td>The term “jellyfish” applies to the species found in these two phyla.</td>
</tr>
<tr>
<td>Class</td>
<td>Various</td>
<td></td>
</tr>
<tr>
<td>Order</td>
<td>Various</td>
<td></td>
</tr>
<tr>
<td>Suborder</td>
<td>Various</td>
<td></td>
</tr>
<tr>
<td>Family</td>
<td>Various</td>
<td></td>
</tr>
</tbody>
</table>

**Genus, Species, and Status**

Table 2. Phylum, species, and status information for jellyfish

<table>
<thead>
<tr>
<th>Phylum</th>
<th>Species</th>
<th>Common Name</th>
<th>USA Status</th>
<th>IUCN Status</th>
<th>AZA Status</th>
</tr>
</thead>
<tbody>
<tr>
<td>Cnidaria</td>
<td>Various</td>
<td>Various</td>
<td>Not specified</td>
<td>None</td>
<td>-</td>
</tr>
<tr>
<td>Ctenophora</td>
<td>Various</td>
<td>Various</td>
<td>Not specified</td>
<td>None</td>
<td>-</td>
</tr>
</tbody>
</table>

**General Information**

The information contained within this Animal Care Manual (ACM) provides a compilation of animal care and management knowledge that has been gained from recognized species experts, including AZA Taxon Advisory Groups (TAGs), Species Survival Plan® Programs (SSPs), Studbook Programs, biologists, veterinarians, nutritionists, reproduction physiologists, behaviorists and researchers. They are based on the most current science, practices, and technologies used in animal care and management and are valuable resources that enhance animal welfare by providing information about the basic requirements needed and best practices known for caring for *ex situ* jellyfish populations. This ACM is considered a living document that is updated as new information becomes available and at a minimum of every five years.

Information presented is intended solely for the education and training of zoo and aquarium personnel at AZA-accredited institutions. Recommendations included in the ACM are not exclusive management approaches, diets, medical treatments, or procedures, and may require adaptation to meet the specific needs of individual animals and particular circumstances in each institution. Statements presented throughout the body of the manuals do not represent specific AZA accreditation standards of care unless specifically identified as such in clearly marked sidebar boxes. AZA-accredited institutions which care for jellyfish must comply with all relevant local, state, and federal wildlife laws and regulations; AZA accreditation standards that are more stringent than these laws and regulations must be met (AZA Accreditation Standard 1.1.1).

The ultimate goal of this ACM is to facilitate excellent jellyfish management and care, which will ensure superior jellyfish welfare at AZA-accredited institutions. Ultimately, success in our jellyfish...
management and care will allow AZA-accredited institutions to contribute to jellyfish conservation, and ensure that jellyfish are in our future for generations to come.

**Basic Taxonomy:** The higher-level phylogeny of commonly maintained gelatinous plankton was adapted from works of Cairns et al. (2002), Cornelius (1997), and Wrobel & Mills (1998). We have added some of the commonly kept genera for each taxonomic group to help you recognize the groupings. The array of diversity of gelatinous plankton is astounding with a myriad of sizes, shapes, and colors to make Dr. Seuss feel right at home.

Gelatinous plankton is the name used to describe a group of actively swimming and drifting organisms. Their tissues consist of 95% or more of water. The tissue is soft, gelatinous-like, and easily damaged when handled. The bell typically is used for propulsion and most species (not in the ctenophores) have stinging cells to assist in prey capture.

**Table 3. Physical description of the species within the phylum Cnidaria**

<table>
<thead>
<tr>
<th>Class</th>
<th>Order</th>
<th>Description</th>
</tr>
</thead>
<tbody>
<tr>
<td>Hydrozoa</td>
<td>Anthomedusae (Anthoathecatae)</td>
<td>Jellies with a bell typically as tall or taller than wide. The gonads are located on the stomach or manubrium. All form a hydroid stage that is not enclosed in a cup (e.g., <em>Polyorchis, Sarsia</em>).</td>
</tr>
<tr>
<td></td>
<td>Limnomedusae</td>
<td>Jellies that are usually dome shaped with gonads either on the stomach or on the radial canals. All form a hydroid stage that is not enclosed in a cup (e.g., <em>Craspedacusta, Maeotias, and Olindias</em>).</td>
</tr>
<tr>
<td></td>
<td>Leptomedusae (Leptothecatae)</td>
<td>Jellies that usually have a bell as wide as or wider than tall. Gonads are located on the radial canals. They typically form colony hydroids and have a well-developed cup that produces medusae (e.g., <em>Aequorea, Clytia, Eutonina, and Timia</em>).</td>
</tr>
<tr>
<td></td>
<td>Siphonophorae</td>
<td>Polymorphic jellies that may have well developed chain-like-appearance or floats (e.g., <em>Physalia</em>).</td>
</tr>
<tr>
<td>Cubozoa</td>
<td></td>
<td>Jellies with marginal tentacles consisting of four, or in four groups of 2–15+ (e.g., <em>Chironex, Carybdea, Tripedalia</em>).</td>
</tr>
<tr>
<td>Scyphozoa</td>
<td>Rhizostomeae</td>
<td>Jellies lacking marginal tentacles and mouth arms fused somewhere along their length, bearing mouth like openings (e.g., <em>Mastigias, Catostylus, Cassiopea, Stomolophus</em>).</td>
</tr>
<tr>
<td></td>
<td>Semaeostomeae</td>
<td>Jellies with tentacles around margin of bell. More than four tentacles present and with well developed mouth arms (e.g., <em>Aurelia, Chrysaora, Cyanea, Pelagia, and Sanderia</em>).</td>
</tr>
</tbody>
</table>

**Table 4. Physical description of the species within the phylum Ctenophora**

<table>
<thead>
<tr>
<th>Class</th>
<th>Order</th>
<th>Description</th>
</tr>
</thead>
<tbody>
<tr>
<td>Tentaculata</td>
<td>Cydippida</td>
<td>Pelagic, globular or ovoid body, with long retractable tentacles arising from sheaths (e.g., <em>Pleurobrachia, Hormiphora</em>).</td>
</tr>
<tr>
<td></td>
<td>Lobata</td>
<td>Pelagic; body compressed with a pair of oral lobes; tentacles generally small (e.g., <em>Bolinopsis, Leucothea, Mnemiopsis</em>).</td>
</tr>
<tr>
<td>Nuda</td>
<td>Beroida</td>
<td>Pelagic; body cylindrical, thimble shaped or compressed; tentacles and sheaths absent (e.g., <em>Beroe</em>).</td>
</tr>
</tbody>
</table>

**Species Identification Using General Morphology:** The basic genera of gelatinous plankton can usually be identified. However, species identification can be problematic due to very brief descriptions and a general lack of worldwide material to examine. As pointed out in Gershwin & Collins (2002), with jellyfish, there is a general lack of standardization of morphological characters and many species are insufficiently described. Using general morphological characters to identify species can cause confusion.
because not all members of a genus are typically examined to ensure that the described characters are diagnostic. As an example, *Cassiopea medusa* was described from Culion Bay, Philippines as being a distinct species based on a single specimen found in a group of *Cassiopea polypoides* (Light, 1914). It is extremely important to examine the population variation in characters before creating additional species. In Hawaii, Holland et al. (2004) found large variation in color and basic morphology within a population of Cassiopea. To further confound species identification descriptions are often incomplete and type specimens are often in very poor shape. Species are commonly described from isolated remote geographic locations or from unusual color morph specimens. This makes it difficult to get specimens from the original description site to compare with a current specimen (Dawson & Jacobs, 2001) and specimens can be polymorphic (Bolton & Graham, 2004).

Gershwin & Collins (2002) reexamined a commonly accepted species *Pelagia colorata* and found morphological characters that were better suited to the genus *Chrysaora*. In addition, work by Freya Sommer at Monterey Bay Aquarium found this species also had a scyphistoma stage that is also more indicative of *Chrysaora* than *Pelagia* (Sommer, 1988). The combination of morphological and life history studies is essential to place species in correct taxonomic groups. However, using morphological characters alone may not provide true taxonomic relationships.

**Species Identification Using Molecular Genetics:** When inspected carefully there may be many cryptic species within a genus. Using *Aurelia* as an example, what seemed to be a worldwide species, *Aurelia aurita* rapidly became a cryptic species complex containing at least nine species (Dawson & Jacobs, 2001; Dawson et al., 2005). The molecular differences in *Aurelia* species complex revealed strong reproductive isolation. Combining genetic sequencing and geographic location information provides a clearer picture for species identification. Using this technique and then applying morphological comparison even cryptic species can be separated (Dawson, 2003). Molecular data can therefore assist in the evaluation of a given morphological character(s) (Knowlton, 2000).

Species identification can also be used to define the geographic origin of an introduced or invasive species. The genus *Cassiopea* was not reported in Hawaii during a 1902 survey and was first documented between 1941 and 1945 (Doty, 1961). It was thought that military boat traffic transported the species to Hawaii. In Hawaii, using general gross morphology *Cassiopea* were examined and found to be highly polymorphic within a given location (Holland et al., 2004). Based on the high degree of polymorphism a single species was assigned *Cassiopea andromeda*. Molecular investigation revealed two cryptic species being present that were 20.3% genetically divergent (Holland et al., 2004). In this case two introduced species entered Hawaii from two different ocean basins one from Indo-Pacific and one from the western Atlantic/Red Sea. Genetics not only provide species identity but also provide information on the species’ original distribution in the case of an invasive species.

**List of Jellies Exhibited in Public Aquariums and Zoos:** The following list of jellies currently or previously on exhibit is from public aquarium and zoo professionals who are on the Jelly Directory kept by Mike Schaadt at Cabrillo Marine Aquarium. Note: spelling of scientific names is based upon: Kramp, P.L. (1952). Synopsis of the medusae of the world. *Journal of Marine Biology Association of the U.K.*, 40, 1–469.
<table>
<thead>
<tr>
<th>Class</th>
<th>Subclass</th>
<th>Order</th>
<th>Species</th>
<th>Common Name</th>
</tr>
</thead>
<tbody>
<tr>
<td>Hydrozoa</td>
<td>Anthomedusae</td>
<td>Bougainvillia sp.</td>
<td></td>
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<tr>
<td></td>
<td></td>
<td>Catalema sp.</td>
<td></td>
<td></td>
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<tr>
<td></td>
<td></td>
<td>Halimeda typus</td>
<td></td>
<td></td>
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<tr>
<td></td>
<td></td>
<td>Leuckartiara sp.</td>
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<tr>
<td></td>
<td></td>
<td>Polypeorchis penicillatus</td>
<td></td>
<td>Mudflat jelly</td>
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<td></td>
<td></td>
<td>P. halus</td>
<td></td>
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<tr>
<td></td>
<td></td>
<td>Sarsi sp.</td>
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<td></td>
<td></td>
<td>Spirocodon saltator</td>
<td></td>
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<tr>
<td></td>
<td></td>
<td>Turrutopsis nutricula</td>
<td></td>
<td></td>
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<tr>
<td></td>
<td>Leptomedusae</td>
<td>Aequorea victoria</td>
<td>Crystal jelly</td>
<td></td>
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<tr>
<td></td>
<td></td>
<td>Clytia gregaria</td>
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<td></td>
<td></td>
<td>Earleria purpurea</td>
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<tr>
<td></td>
<td></td>
<td>Eirene vindula</td>
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<tr>
<td></td>
<td></td>
<td>Eucheilota sp.</td>
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<td></td>
<td></td>
<td>Eutonina indicans</td>
<td></td>
<td>Bell jelly</td>
</tr>
<tr>
<td></td>
<td></td>
<td>Microcoma cellularia</td>
<td></td>
<td>Cross jelly</td>
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<tr>
<td></td>
<td></td>
<td>Ptychogena spp.</td>
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<tr>
<td></td>
<td></td>
<td>Timo formosa</td>
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<td></td>
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<tr>
<td></td>
<td>Limnomedusae</td>
<td>Craspedacusta sowerbi</td>
<td>Freshwater jelly</td>
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<tr>
<td></td>
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<td>Maeotias inexpectata</td>
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<td>Flower-hat jelly</td>
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<tr>
<td></td>
<td></td>
<td>Olindias formosa</td>
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<tr>
<td></td>
<td></td>
<td>Physalia physalis</td>
<td>Portuguese Man ‘o War</td>
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</tr>
<tr>
<td></td>
<td>Siphonophorae</td>
<td>Physalia physalis</td>
<td>Portuguese Man ‘o War</td>
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<tr>
<td></td>
<td>Cubozoa</td>
<td>Carukia barnesi</td>
<td>Box Jelly</td>
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<td></td>
<td></td>
<td>Carybdea alata</td>
<td>Box Jelly</td>
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<td></td>
<td>C. marsupialis</td>
<td>Box Jelly</td>
<td></td>
</tr>
<tr>
<td></td>
<td></td>
<td>Chironex fleckeri</td>
<td></td>
<td>Sea Wasp</td>
</tr>
<tr>
<td></td>
<td></td>
<td>Tripedalia cystophora</td>
<td></td>
<td>Box Jelly</td>
</tr>
<tr>
<td></td>
<td>Scyphozoa</td>
<td>Cassiopea xamachana</td>
<td>Upside-down Jelly</td>
<td></td>
</tr>
<tr>
<td></td>
<td>Rhizostomae</td>
<td>C. andromeda</td>
<td>Upside-down Jelly</td>
<td></td>
</tr>
<tr>
<td></td>
<td></td>
<td>C. frondosa</td>
<td>Upside-down Jelly</td>
<td></td>
</tr>
<tr>
<td></td>
<td></td>
<td>Canostylus mosaicus</td>
<td>Blubber Jelly</td>
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<tr>
<td></td>
<td></td>
<td>Drymonema dalmatium</td>
<td></td>
<td></td>
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<tr>
<td></td>
<td></td>
<td>Mastigias papua</td>
<td></td>
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<td></td>
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<td>Netrostoma setouchianum</td>
<td>Spotted lagoon jelly</td>
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<tr>
<td></td>
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<td>Phylorrhiza punctata</td>
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<td>White-spotted Jelly</td>
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<tr>
<td></td>
<td></td>
<td>Pseudorhiza haeckeli</td>
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<tr>
<td></td>
<td></td>
<td>Rhizostoma pulmo</td>
<td></td>
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<td></td>
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<td>Stomolophus meleagris</td>
<td>Cannonball jelly</td>
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<td>Cephea cephea</td>
<td>Crown jelly</td>
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<tr>
<td></td>
<td>Semaeostomae</td>
<td>Aurelia aurita</td>
<td>Warm water moon jelly</td>
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<td></td>
<td></td>
<td>A. labiata</td>
<td>Cold water moon jelly</td>
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<tr>
<td></td>
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<td>Chrysaora fuscescens</td>
<td>West coast nettle</td>
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<td></td>
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<td>C. quinquecirra</td>
<td>East coast nettle</td>
<td></td>
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<tr>
<td></td>
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<td>C. colorata</td>
<td>Purple-striped jelly</td>
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<tr>
<td></td>
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<td>C. melanaster</td>
<td>Black star northern sea nettle</td>
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<tr>
<td></td>
<td></td>
<td>C. achlyos</td>
<td>Black sea nettle</td>
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<td>Cyanea capillata</td>
<td>Lion’s mane jelly</td>
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<td>Pelagia noctiluca</td>
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<td>Phacellophora camtschatica</td>
<td>Egg yolk jelly</td>
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</tr>
<tr>
<td></td>
<td></td>
<td>Sanderia malayensis</td>
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</tr>
</tbody>
</table>
### Table 6. Species exhibited in zoos and aquariums found within the phylum Ctenophora

<table>
<thead>
<tr>
<th>Class</th>
<th>Order</th>
<th>Species</th>
<th>Common name</th>
</tr>
</thead>
<tbody>
<tr>
<td>Tentaculata</td>
<td>Cydippida</td>
<td>Hormiphora</td>
<td>Sea gooseberry</td>
</tr>
<tr>
<td></td>
<td></td>
<td>Pleurobrachia bachei</td>
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<tr>
<td>Lobata</td>
<td></td>
<td>Bolinopsis mikado</td>
<td>Sea walnut</td>
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<tr>
<td></td>
<td></td>
<td>B. infundibulum</td>
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<tr>
<td></td>
<td></td>
<td>Leucothea pulchra</td>
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<tr>
<td></td>
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<td>Mnemiopsis leidy</td>
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<td>M. mccradyi</td>
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<tr>
<td>Nuda</td>
<td>Beroida</td>
<td>Beroe cucumis</td>
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<td>Beroe forskali</td>
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<td></td>
<td></td>
<td>B. ovata</td>
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</tr>
</tbody>
</table>

**Jellyfish, Sea Jellies, and Gelatinous Zooplankton:** Individual aquariums and zoos differ in their terminology when referring to jellyfish that they display to their visitors. Some call them sea jellies while others stick to the traditional common name of jellyfish. Those that insist that sea jellies are not fish. It is the prerogative of the individual aquarium or zoo to choose the common name that best suits their educational mission. Also, since this care manual includes ctenophores, which are not even in the same phylum as jellyfish, a more scientifically precise name for this group would be “gelatinous zooplankton” which could include many other groups like pteropod and heteropod mollusks and salps (chordates). When we use the words jellyfish, jellies, or sea jellies in this manual, we are referring to medusae belonging to the phylum Cnidaria. When we include members of the phyla Mollusca, Ctenophora, and Chordata, along with Cnidaria, we use the term “gelatinous zooplankton.”

**Cnidaria:** Most common cnidarian jellies, and almost all that are displayed by public aquariums, exhibit two morphologic stages. The pelagic medusa stage is most familiar. The sessile polyp stage is inconspicuous, but it is this life stage that makes possible and practical the culture and propagation of jellies for aquarium exhibits.

The sessile stage is typically small (1–2 mm tall) and usually colonial. There is no sexual reproduction in this stage. Sessile polyps can spread asexually to generate other polyps—or they can give rise to the pelagic (medusa) stage by another asexual process called strobilation. The medusae are free-swimming and solitary. Most medusae become conspicuously large, although some hydrozoan species (Hydromedusae) can be quite small (1–2 mm). With few exceptions, jellies are dioecious—each medusa is either male or female. In most cases, the medusa stage can only reproduce sexually, and in most species the fertilized egg eventually gives rise to a new sessile polyp. An exception to this, for example, would be *Aequorea* sp. with medusae that can asexually reproduce through fission. There are species in which the fertilized egg develops directly into another medusa, either exclusively (*Pelagia noctiluca*) or rarely (*Aurelia aurita*) (Årai, 1997).

**Medusoid Stage** (see Chapter 7, Figure 4 for illustration) *(Hydromedusa and Scyphomedusa—orders Anthomedusae, Leptomedusae, Semaeostomeae, and Rhizostomeae):* Most medusae are reminiscent of a bell or an umbrella shaped structure. The convex top of the bell is referred to as the *exumbrella* and is the aboral side, as it is away from the mouth. The concave underside of the bell is known as the *subumbrella* and is the oral side, where the mouth is centrally located. The bell consists primarily of *mesogloea* (a jelly-like matrix with or without cells), muscle and nerve fibers, and the subumbrella, which serves as an attachment site for digestive and reproductive structures. The bell plays an important role in locomotion and generally represents a high percentage of the total volume of the animal (Årai, 1997). A *velum* is a thin muscular tissue, arising from the inner margin of the bell and is present in hydrozoans only, acting as a key distinguishing feature of the two classes (Wrobel & Mills, 1998). Cubozoans have an analogous (but not homologous) structure called a velarium.

Additional structures can be found at the margins of the bell. Tentacles (marginal, stinging, or fishing) are present (except in Rhizostomeae). The tentacles serve primarily in prey capture and defense. Generally they house the highest proportions of stinging cells (i.e., nematocysts) utilizing a variety of toxins (Lotan et al., 1995). The stinging cells fire upon mechanical stimulus of the cnidocil, or from a variety of chemical cues (Burnett & Clayton, 1986; Brusca, 1990; Årai, 1997). *Rhopalia* (marginal sense...
organs only found in the scyphozoans) occur at regular intervals along the margin of the bell. The rhopalia house concentrated levels of sensory receptors. They are complex structures that can show sensitivities to light, gravity, touch, and chemicals (Arai, 1997). The intervals between rhopalia are known as lappets.

Scyphozoan medusae capture food by using the marginal tentacles and oral arms, the large frilly tissues originating from the mouth on the oral side of the bell. Once captured, food is transferred to stomach pouches (typically four are present). In some species, digestion begins within the oral arms, but the bulk of the process occurs in the stomach pouches (Arai, 1997).

Hydrozoans lack oral arms and separate stomach pouches. Instead, the mouth is located at the end of an often highly maneuverable structure called a manubrium, which functions as a gastric cavity. It remains very active during feeding, picking off captured prey items from the marginal tentacles. Scyphozoans also have manubria. Many scyphozoans and hydrozoans also have radial canals associated with the gastrovascular system and some have an attached marginal ring canal. Gonads are present in mature specimens. Gonad tissues arise from gastrodermis in scyphozoans. They are usually located on the floor of the gastrovascular cavity, peripheral to the gastric cirri (Arai, 1997). Gonad tissues in hydrozoans are either adjacent to the manubrium or associated with the radial canals. Most scyphozoans and hydrozoans are dioecious (i.e., separate sexes).

**Polypoid Stage** (see Chapter 7, Figure 2 for illustration).

*(Hydromedusa—orders Anthomedusae, Leptomedusae):* Hydrozoan polyps, or hydroids, attach to a substratum. The anchoring structure is termed a **hydrorhiza**.

The stem or stalk-like structure is termed a **hydrocaulus**. From the hydrocaulus, several types of polyps can occur, including **hydranths** or **gastrozooids** (for feeding), gonozooids (for reproduction), and **dactylozooids** (for defense). Dactylozooids are heavily armed with cnidae and often surround gastrozooids as protection and to assist in food capture (Brusca, 1990). According to Richard Brusca, the gonozooids form medusa buds called gonophores that can either be retained or released.

*(Scyphomedusa—orders Semaeostomeae, Rhizostomeae):* Scyphozoan polyps, **scyphistomae**, generally resemble their cousins, the sea anemones, from the class Anthozoa. They attach to suitable substrate with a base called a **pedal disc**. The polyp extends out from the substrate, forming a stalk that concludes with a **calyx**. The calyx includes an oral disc with a central mouth and a ring of up to 24 tentacles (Arai, 1997). The stalk can be cylindrical, as in most Chrysaora spp., or fluted as in many rhizostomes. Runners known as **stolons** may be present that extend longitudinally from the stalk to give rise to new polyps or aid in movement. Scyphistomae of some species produce podocysts, from which new scyphistomae emerge. In such cases, the parent polyp encapsulates material in chitin that remains dormant until triggered to “bloom” into an entirely new polyp.

**Ctenophora:** Ctenophores have clear, gelatinous bodies characterized by having eight rows of ciliated plates (comb rows) radiating from aboral to oral surfaces that they use for locomotion. (They are the largest animal to use cilia for locomotion.) Their bodies are round, oblong, or even compressed with two lobes.

**Lobate ctenophore:** Lobate ctenophores have small tentacles and compressed bodies with two large lobes at the oral end (e.g., *Mnemiopsis, Bolinopsis*).

**Cydippid ctenophore:** Cydippid ctenophores are solid spherical or ovoid bodies with a pair of long retractable tentacles arising within sheaths on opposite sides of the body. Tentacles have smaller branches called **tentilla**. The mouth is on one end. Prey are captured on the tentilla by the sticky cells called **colloblasts**, and eventually transferred to the mouth as the ctenophore tumbles through the water. The tumbling is called “Veronica’s Display” since it resembles a matador swishing his cape in a bullring (e.g., *Pleurobrachia, Hormiphora*).

**Beroid ctenophore:** Beroe ctenophores are oblong in shape and laterally flattened. There are no tentacles. The large mouth is on one end. Prey is exclusively tentaculate ctenophores (e.g., *Pleurobrachia*).

**Jellyfish Terminology**

- **Bell:** The umbrella shaped body of jellies.
• Colloblasts: Adhesive cells found only on the tentacles and tentilla of ctenophores.
• Cnidaria: Phylum that contains organisms (diploblastic) that possess nematocysts (stinging cells) situated around a central mouth.
• Ctenophora: Phylum that contains organisms possessing colloblasts and comb rows. Also known as comb jellies or sea gooseberries.
• Cubozoaa: A class of Cnidaria where the jellies’ bells are roughly box-shaped.
• Dactylozooids: A specialized polyp in a hydroid colony that provides defense for the colony.
• Dioecious: Each medusa has either male or female sex organs (also known as gonochoristic).
• Ephyra: Small, first stage, free-swimming jelly destined to grow up to mature and become the adult medusa.
• Exumbrellar: The aboral surface of the bell of a jelly.
• Gastrozooids: A specialized feeding polyp in a hydroid colony that shares its food with the rest of the colony (also known as a hydranth).
• Gonozooids: The medusa-producing polyp in a hydroid colony (also known as gonangium).
• Hartford loop: A system of piping that acts as a safety device to prevent water from draining out of an aquarium if a leak develops in the return piping. Also maintains water level in remote tanks.
• Hydrocaulus: Branched upright stem of a hydroid colony.
• Hydromedusae: A class of cnidaria that differs from the scyphozoa in that the medusae tend to be smaller and possess a velum on the rim of the bell. The velum is important in locomotion. Medusae belonging to the class Hydrozoa.
• Hydorhiza: Root-like structure of a hydroid colony.
• Hydrozoa: A class of cnidaria in which most members have polyps arranged in colonies that are made up of different polyps with different functions.
• Manubrium: An extension of the mouth in jellies that often is the place where the oral arms originate.
• Medusa: The planktonic stage of most jellies, which is responsible for sexual reproduction.
• Mesoglea: Translucent jelly-like substance found between the two epithelial cell layers in the bodies of cnidarians. Mesoglea makes up the bell of a jelly and is mostly water. In scyphozoan there are living cells that can move throughout the mesoglea. There are no living cells in the mesoglea of hydrozoans.
• Monoecious: Jellies that contain both male and female sex organs in the same individual.
• Nematocysts/cnidocysts/cnidae/stinging cells: Specialized cells that when mechanically stimulated shoot a tiny harpoon-like dart into the tissue of prey or predators.
• Ocelli: Light sensing organs embedded within the rhopalia.
• Oral arms: Also known as mouth arms. Extension of the manubrium (mouth) in jellies.
• Phytoplankton: Photosynthesizing plankton (mostly unicellular) at the mercy of ocean currents.
• Planktonkreisel: A name coined by the German scientist W. Greve in the 1960s, and further refined by Bill Hamner of UCLA in the 1990s, to describe a type of aquarium with gentle circular currents designed to simulate a water column for planktonic organisms. Many adaptations to the original design have resulted in pseudokreisels (those without laminar flow for water entering) and stretch kreisels (those wider than tall and usually two gentle gyres of currents).
• Planula larva: What the fertilized egg develops into. Cnidarian larval form.
• Podocysts: A tiny round, flat structure that some scyphozoan polyps grow at their base. They are capable of growing into a polyp when the conditions are right. Podocysts can remain dormant for months and even years.
• Polyp: The common name for the asexual sessile stage of hydrozoan, scyphozoan, and cubozoan.
- Rhopalia: Found embedded in the lappets of the ephyrae and in the rim of the bell in adult medusa. Contains two sense organs each (ocelli for light reception and statocyst for gravity reception).

- Scyphistoma: The scientific name of the polyp stage of most jellies.

- Scyphomedusae: A class of cnidaria that tend to have medusa much larger than hydrozoan medusae and lack a velum at the rim of the bell.

- Scyphozoa: A class of cnidaria in which the polyps tend to be solitary, medusae tend to have mouth arms and gonads arranged in a cloverleaf pattern just under the bell.

- Statocyst: Gravity sensing structure embedded within the rhopalia.

- Strobilation: A form of asexual reproduction found in the scyphozoa beginning with segmentation of the polyp and ending with an ephyra.

- Subumbrellar: The oral surface of the bell of a jelly.

- Tentilla: Short extensions arising from the tentacles of ctenophores that have the adhesive cells (colloblasts) attached.

- Vellum: A structure found in the hydrozoan jellies that looks much like a shelf attached to the rim of the bell.

- Zooplankton: Animals that are at the mercy of ocean currents. Most have the ability to move on their own in a small space, but are swept along with ocean currents.
Chapter 1. Ambient Environment

1.1 Temperature and Humidity

Animal collections within AZA-accredited institutions must be protected from weather detrimental to their health (AZA Accreditation Standard 1.5.7). Animals not normally exposed to cold weather/water temperatures should be provided heated enclosures/pool water. Likewise, protection from excessive cold weather/water temperatures should be provided to those animals normally living in warmer climates/water temperatures.

As different systems are discussed in this chapter some of the important variables associated with raising jellies will be briefly addressed. Not every species or every water quality issue can be covered. This chapter is intended to help an experienced fish aquarist understand some of the unique problems encountered when a jelly display is set up.

Variation in species and display requirements allow for a huge range of acceptable temperatures to be used. Widmer (2008) gives 21 °C (69.8 °F) as the fastest growth rate for Aurelia aurita ephyrae. This temperature may be required to raise enough moon jellies for a large exhibit or as food for other jellies. Lower temperatures result in healthier adult jellies that can live for a much longer time. For Aurelia labiata, 13–16 °C (55.4–60.8 °F) will allow the jellies to last up to a year or more with much fewer deformities due to heat stress or rapid growth. Temperature for temperate water jellies can run from 10–20 °C (50–68 °F) with some species being able to tolerate an even larger range, but given the delicate nature of jellies, cooler water normally results in better results. Tropical jellies, on the other hand, tend to have more trouble dealing with water too cold than with water too warm. Most tropical species (Mastigias, Catostylus) do well between 22–28 °C (71.6–82.4 °F). Cold water jellies (e.g., Chrysaora colorata, Phacellophora camtschatica) are normally much harder to keep (e.g., most are medusasivorous and colder temperature water requires chillers), so they are not recommended for people trying jelly husbandry for the first time.

AZA institutions with exhibits which rely on climate control must have critical life-support systems for the animal collection and emergency backup systems available, while all mechanical equipment should be included in a documented preventative maintenance program. Special equipment should be maintained under a maintenance agreement or records should indicate that staff members are trained to conduct specified maintenance (AZA Accreditation Standard 10.2.1).

For coldwater jellies (about 15 °C [59 °F] or colder) a chiller is required. For tropical jellies, a heater may be required to reliably keep the water within about 2–3 degrees Celsius of the desired temperature. In both cases, electrical power is required and there should be a backup generator to make sure that chillers and heaters can continue to maintain water temperature. There should be an alarm system that notifies a staff member with experience to respond if temperature changes more than about 2–3 degrees Celsius.

1.2 Light

Careful consideration should be given to the spectral, intensity, and duration of light needs for all animals in the care of AZA-accredited zoos and aquariums.

Lighting is an easy way to enhance a display’s appearance and allow visitors a better view of an almost transparent animal. The choices for jelly lighting can include standard outside floodlights, track lights (both standard bulbs and high intensity), aquarium lights such as compact fluorescent, metal halide, fiber optic, or LED lights. Each of these has pros and cons, except in the case of tropical jellies with commensal, photosynthetic zooxanthellae in their tissue. The choice would be based more on costs and aesthetic preference.
Standard white lighting is fine for moon jellies or darker jellies, but experimenting with colored lights and gels should result in the right choice for each application. Blue lighting on moon jellies gives people more of a feeling of being out in the ocean with them. Red light gives an ominous appearance. With animals like ctenophores, narrow intense lights show off the comb rows. If the tank is long and narrow, try changing from one color lighting to another. Use your imagination!

Lighting can also be helpful in hiding the screens, walls, and tops of tanks so that a visitor’s attention is drawn and remains on the jellies. This can be done with spotlights, “mail slots,” and “barn doors” that redirect light. Such devices will also eliminate glare on windows.

With the few jellies that harbor zooxanthellae in their tissues, lighting becomes a husbandry issue. Even though the jellies are moving in the tank, think of them like a coral. The spectrum of light used should contain significant amounts of light in the wavelengths between 400–500 µm. A photoperiod between 10–14 hours is also important. If mixed lighting is used, the most intense lighting (similar to lighting for corals) should be a shorter duration toward the middle of the photoperiod. If staff is more familiar with bulb “color temperatures,” expressed in degrees Kelvin (K), a mix of daylight bulbs in the 5,000–6,500° K range, and at least one in the blue range of 10,000–20,000° K, will keep the zooxanthellae happy and the jellies healthy while giving the tank an attractive hue.

Although tropical jellies can deal with very warm waters, strong lighting can rapidly heat up water to dangerous levels especially in small volume closed systems. Running the lights before you put jellies in the tank will help keepers gauge how much heat is added to the tank. Remember, if the water system stops, turn off any high intensity lights to prevent overheating the jellies.

1.3 Water and Air Quality

AZA-accredited institutions must have a regular program of monitoring water quality for collections of aquatic animals and a written record must document long-term water quality results and chemical additions (AZA Accreditation Standard 1.5.9). Monitoring selected water quality parameters provides confirmation of the correct operation of filtration and disinfection of the water supply available for the collection. Additionally, high quality water enhances animal health programs instituted for aquatic collections.

Jellies’ needs vary from fish partly because they are plankton. Flow is of utmost importance with most species of jellies, so specially designed tanks are required to house them. Without enough flow, the jellies may get pulled into the drain screens or settle to the bottom. The soft body of a jelly might be torn (or crushed against a wall) if a pump sprays directly on a jelly. To prevent this from happening, create what is known as “laminar flow.” This spreads the flow into a thin line across an entire plane of the tank. Laminar flow can be accomplished by using a spray bar or slots cut through a divider in the tank. By creating this laminar flow over a drain screen, jellies are pushed away from the screen and a directional current is created. Species that sit on the bottom most of the time can be maintained in conventional tanks, but incoming water flow should enter the tank in such a way as to push animals away from the drain screen. It is hard to quantify how strong a flow rate should be until the tank has water in it. Flow rates will vary with tank design and size but it should always be such that it pushes the jelly along with the flow and not spin the jelly in the flow. Both eversion and balling up can be a side effect of too high a flow rate. Malnutrition can also occur when the flow is too strong. Learning to judge flow is one of the harder jelly husbandry skills to master and has to be ascertained for each individual species. Achieving a balanced flow depends on individual tank design, species, age and relative fitness of the species. For example, once staff have created suitable flow for moon jellies, the whole process has to be redone to learn how to judge correct flow again for a lion’s mane trailing a foot of oral arms and three feet of tentacles. It is best to introduce jellies into a new system with no flow at all and then incrementally turn the flow up as needed to give the particular species you are working with its adequate flow rate. Observation of wild specimens will assist in learning what is adequate. Different size jellies will require different flow rates. This again is the reason to keep similar size jellies together both on exhibit and in the off-exhibit holding area.

Open Systems Versus Closed Systems: When discussing life support systems most aquariums define “open systems” as systems that water is continually replaced with new water from a constant source such
as the ocean. Open systems normally contain all the needed nutrients found in nature, but may contain unwanted contamination found in an ecosystem not suited for the animals being housed. No true “closed system” exists since water will always need to be replaced due to evaporation or to maintain water quality. For these reasons, closed systems can be defined as aquariums that recycle water and receive a limited supply of new water as needed. Both open and closed systems can be used for jellies, but both require an understanding of each system’s benefits and limitations.

Open systems provide many advantages for most aquariums because water quality normally is very consistent in large bodies of water, but one should be aware of possible problems that can arise from the use of variable natural seawater. Since all but a couple of species of jellies are found in seawater, making sure the incoming water salinity matches the tank water salinity can be accomplished with a reservoir where salinity can be checked, and salt or fresh water can be added as needed. Jellies can handle small, slow changes in salinity, but because the density in the jelly depends on temperature and salinity of the water to which they are exposed, rapid changes can cause them problems. It is best if the temperature and salinity of the area in which each species is found is replicated as closely as possible.

When using water from the ocean, the flow will often bring in animals such as hydroids, copepods, or amphipods. Also the jellies themselves sometimes harbor these creatures making it nearly impossible to have a system free of these fouling animals.

Most aquarists can attest to how hard removing hydroids can be. To help minimize the pests from getting out of control, filtering the water and using UV sterilization can help control unwanted populations (although planulae are reported to swim right through UV sterilization; the contact time is not long enough to destroy them). However, nothing is 100% effective, so oftentimes breaking the tanks down and bleaching them is the only option.

Typical water quality considerations include how water quality affects medusa, but polyps will be mentioned in this manual when issues arise that might affect them. Most people wanting to maintain systems with jellies have usually already learned how to keep tropical fish. If fish are safely maintained, then water quality for jellies should not be an issue with most jellies commonly housed in public aquariums. Jellies can be hardy when it comes to most water quality issues, but one should be aware that things proven harmful in reef tanks (with other invertebrates) also affect jellies. Some of these issues may be bad for an adult medusa, but desired to induce strobilation in the polyps. Unlike fish, jellies can tolerate very high levels of ammonia, but can be killed by very low quantities of copper and zinc. While copper levels are required to be reported on EPA Consumer Confidence Reports (CCR) for an aquarium’s city, zinc is not. It is commonly added to mitigate pipe corrosion. Pre-treating city water with both activated carbon and reverse osmosis may be necessary to bring metal levels to non-lethal levels. There are a number of parameters to consider. The following chart shows ideal conditions for some of the factors that affect the health of jelly systems. These are suggested values for a healthy stable system, not the maximum or minimum levels jellies can tolerate.

<table>
<thead>
<tr>
<th>Parameter</th>
<th>Ideal Conditions</th>
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<tbody>
<tr>
<td>Ammonia (unionized)</td>
<td>&lt;2 ppm</td>
</tr>
<tr>
<td>Nitrites</td>
<td>&lt;2 ppm</td>
</tr>
<tr>
<td>Nitrates</td>
<td>&lt;20 ppm</td>
</tr>
<tr>
<td>Copper</td>
<td>&lt;0.01 ppm</td>
</tr>
<tr>
<td>Salinity (most medusa)</td>
<td>32–35 ppt</td>
</tr>
<tr>
<td>pH</td>
<td>8.0–8.4</td>
</tr>
<tr>
<td>Dissolved Oxygen</td>
<td>75–99%</td>
</tr>
</tbody>
</table>

Jellies can have problems with metals that may be dissolved in the water. Copper is very harmful even in very low quantities. If at all possible make sure copper levels are as close to zero as possible. When testing for copper, a colorimeter will give a high enough degree of accuracy where a test kit made for home aquariums might not read low enough levels to ensure safety. Mechanical parts of pumps, pipes, wiring fixtures and metal sensors could all be sources of metal leakages. It is important to verify and test potential leakages in the system. If the water source being used is such that it gets large amounts of seasonal run off or is from a lagoon that might be dredged periodically (this might suspend metals or pesticides in the water column), running the tanks as a closed system should be considered. Carbon filters can be added to either open or closed systems, but carbon dust can be an issue in jelly tanks. The dust particles could be picked up by the jellies in place of the food they would need. Always rinse new carbon filters with running tap water until water runs clear of any carbon dust just prior to use. A filter pad that has carbon imbedded into it is one possible way to help insure no copper
gets into the system and reduces the possibility of added carbon dust. There are also other copper absorbers on the market, which seem to work well. Be aware that copper might not be the only thing present in the water and carbon is known to adsorb many harmful items very effectively. It should be noted that carbon does not effectively remove ionic copper, only copper that is bound to organic compounds.

Closed systems require more maintenance in most cases, but allow the aquarist direct control over the water supply. Rapid changes stress almost all animals and jellies are no exception. When changing system water, matching the new water to the system’s parameters will help reduce problems. Reverse osmosis or de-ionized (RO/DI) waters are best to use to make seawater for closed systems. Aerate the make-up water for 24 hours prior to use for best results. It is not recommended to use tap water to make seawater for jelly systems. Although most commercial chlorine neutralizers claim to be safe for invertebrates, some tend to cause pitting and eventual breakdown of the jelly’s tissue.

Most of the same rules apply when setting up a closed system for jellies that apply for fish, but with a few exceptions. The major one being the drain and water return lines. Unlike fish, jellies can’t swim away from drains so they have to be pushed away. This is where the laminar flow is put to the test. Laminar flow pushes water over large areas so drains can be made larger and screens or a perforated wall in front can help to reduce excessive suction, which can capture the jelly as it floats around the tank.

Return lines that normally provide flow also have to be a bit different. Unlike fish, jelly systems can’t have waterfalls or air bubblers or return lines pushing air into the tank. Bubbles that become trapped in a jelly bell will work their way through it, causing serious damage. Bubbles can also cause the jelly to float, which inhibits feeding. In some cases, floating jellies can also clog the screens or overflows. Return lines can be set below the surface and flushed of air before adding jellies. A well-designed system will take this into account so that when water changes are done, or if the power goes out, air won’t flush into the tank and into the jellies. Low flow rates, standpipes, Hartford loops, and check valves can save the aquarist a world of headache. Almost every person who raises jellies has spent a few hours trying to remove air bubbles from the underside of a jelly’s bell. It may seem easy to tip a jelly over to let the air float up and out but it is not when the jelly is a Fried Egg Jelly with long frilly oral arms and tentacles that stick to everything, including the aquarist. It is soon discovered that air loves mucus and “mild stings” are not so mild when the tentacles remain on a tool that an aquarist accidentally lets rest against the back of their arm. So before an institution ends up with floating jellies and a staff covered in arm rashes, make sure the system is prepared for a power outage or that water changes won’t fill the tank(s) with bubbles.

Another way jelly systems might end up with bubbles is supersaturation. Jellies can tolerate much lower oxygen levels, but if those levels go too high and air starts diffusing out of the water and sticks to the tank walls, it is only a matter of time before a jelly will get some in its bell. Super saturation is often caused when cool, saturated water is added to a system, and then a pump increases the pressure and warms it by a couple of degrees. Once the pressure is removed (the water enters your tank) the excess gas starts to work its way out of solution. Trickle towers or gravity feed systems can help prevent this from becoming an issue.

1.4 Sound and Vibration
Consideration should be given to controlling sounds and vibrations that can be heard by animals in the care of AZA-accredited zoos and aquariums. Jellyfish do not have auditory or vibration sense organs.
Chapter 2. Habitat Design and Containment

2.1 Space and Complexity

Careful consideration should be given to exhibit design so that all areas meet the physical, social, behavioral, and psychological needs of the species. Animals should be displayed, whenever possible, in exhibits replicating their wild habitat and in numbers sufficient to meet their social and behavioral needs (AZA Accreditation Standard 1.5.2).

Setting up the System: Almost all jelly tanks will be free of sharp corners or obstructing objects including substrate. Whether the tank design is a kreisel type (i.e., a circular or semi circular design), it will have a return line providing laminar flow and some type of drain screen where water exits the tank. The simplest return is a spray bar that is aimed so the incoming water pushes the jellies away from the area of exiting water. The shape of the tank will determine if this flow will be enough to keep your jellies happily moving in the water column or if additional spray bars or other flow solutions are needed. Once the water leaves the tank, a Hartford loop can insure the tank does not drain below the spray bar (if it is near the top).

If the system is closed, this line should lead to a sump of some kind. Jelly tanks tend to have large amounts of excess food getting flushed out the drain. To help the system stay clean, a simple 200 micron polypropylene felt filter bag that can be rinsed and replaced every couple of days is highly recommended. If the sump is designed with a trickle plate, simply tipping it slightly and covering the plate with a filter pad will work in place of a bag. The sump should be big enough to hold enough bio-media to ensure proper bio filtration for the system. Since almost all jelly tanks will get invasive hydroids, adding disconnect couplings will save time when cleaning. This is also a good idea in case of pump failures or system changes. Cartridge filters will work fine, but because of the large amounts of brine shrimp, nauplii, and chopped foods needed to maintain healthy jellies, the cartridges tend to clog quickly reducing flow and leading to troublesome water quality issues.

Although a jelly can withstand the higher ammonia levels associated with cycling a tank, adding the jellies during the cycling is not recommended. Jellies under stressful conditions produce copious amounts of slime, release gametes into the water, and develop problems, such as pits on their bells. These conditions are rarely fatal, but all of them can cause problems to a system’s balance and appearance.

Both the slime and gametes in the water can be removed with protein skimming. Unfortunately, protein skimmers require large quantities of fine air bubbles, so degassing may be needed. If possible, run the effluent line from the protein skimmer into the filter bag and over bio balls in the wet/dry area of the sump. This can help to eliminate bubble problems. If planula larvae are released (which will happen with adult moon jellies at some point) not all larvae will be removed by protein skimming, but it will help to control their numbers.

Ozone is becoming more common in aquaria because it promotes water clarity and parasite control, but unless it can be fully broken down before the water returns to the tank it should not be used. If added to the tank in a contact chamber or protein skimmer, it should pass through an activated carbon filter before remixing with the tank water. Other options such as UV sterilization will help control bacterial blooms and kill some pathogens, but planula larvae tend to pass through UV filters unharmed. This is good for when one is trying to raise polyps of a jelly, but unwanted hydroids or very prolific jellies such as moon jellies (Aurelia sp.) or upside down jellies (Cassiopeia sp.) will continue to spread.

Once the water has passed through your filter, the only challenge left is returning it to the tank. Since bubbles are a major problem for jellies, pump intakes should be toward the very bottom of the sump. Float switch shut offs can be added to prevent the sump from getting too low and air being pulled into the intake. A sump can be drained too low from evaporation, leaks, or most commonly an aquarist or volunteer who forgets to keep an eye on the level while changing water. Ball valves and check valves should be installed so water can be maintained in the pipes in case of such an event.

Water flow back to the tanks can often be regulated simply by adding a ball valve but in some cases where a large pump is required, water may have to be diverted either back to the sump or used to drive filter mechanisms. If large flows for circulation in a big tank are required, multiple spray bars or a trickle plate with an acrylic deflecting piece might allow strong directional currents without the direct pressure that occurs at the outlets on a single spray bar.

AZA Accreditation Standard

(1.5.2) Animals should be displayed, whenever possible, in exhibits replicating their wild habitat and in numbers sufficient to meet their social and behavioral needs. Display of single specimens should be avoided unless biologically correct for the species involved.
The same careful consideration regarding exhibit size and complexity and its relationship to the jellyfish’s overall well-being must be given to the design and size of all enclosures, including those used in exhibits, holding areas, hospital, and quarantine/isolation (AZA Accreditation Standard 10.3.3). Jellyfish do not exhibit social behaviors. Off-exhibit enclosures require the same basic design as exhibits in public displays.

2.2 Safety andContainment

Animals housed in free-ranging environments should be carefully selected, monitored, and treated humanely so that the safety of these animals and persons viewing them is ensured (AZA Accreditation Standard 11.3.3).

Species Selection: Species are usually chosen on the basis of:
1. Difficulty of species husbandry;
2. Available food sources;
3. Staff expertise;
4. Regional focus;
5. Availability of suitable aquarium tanks.

The following safety and containment considerations need to be taken into account when choosing to exhibit jellies:

- Jellies exhibit aquaria should have the normal alarms that alert the aquarist in charge. These parameters include: temperature and water flow.
- Jelly exhibits in public displays should have designs that keep visitors from having access to reaching into the displays.
- Jellyfish are best kept in unspecies aquarium tanks both for their safety (they can be very delicate) and for the safety of other animals (jellyfish will sting when touched, and will consume some other jellies).
- Jellies release planulae that can easily flow through the outflow and can colonize underwater surfaces. Planulae from one species of jelly can be accidently introduced into the culturing container of another. To reduce this risk, aquarists should keep separate tools (e.g., basters, cleaning devices, glass jars, etc.). Also, care needs to be taken when food is being introduced so that water from one aquaria doesn’t get introduced from another aquaria filled with a different species of jellies that might have been releasing planulae.
- It is also important to take measures not to release water from jelly systems into waterways where the jellies can be an invasive species. For this reason, water leaving coastal institutions should go to a water treatment facility. Always follow local laws governing the proper treatment of waste water.
- Jellies should be kept in appropriate aquaria designed to meet their planktonic lifestyle.
- Jellies can be harmed by anything jutting into the aquaria. Care needs to be taken that jellies are kept away from overflow or drain screens attached to the aquaria. Plastic burrs or plastic cement slivers or shavings can be extremely harmful if undetected. New and renovated aquariums should be inspected by hand for such defects.
- Jelly aquaria should have simple lids when their display is in public spaces. This will prevent people touching the jellies and keep people from putting foreign objects into the aquarium.

Animal exhibits and holding areas in all AZA-accredited institutions must be secured to prevent unintentional animal egress (AZA Accreditation Standard 11.3.1). Exhibit design must

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AZA Accreditation Standard
(10.3.3) All animal enclosures (exhibits, holding areas, hospital, and quarantine/isolation) must be of a size and complexity sufficient to provide for the animal’s physical, social, and psychological well-being; and exhibit enclosures must include provisions for the behavioral enrichment of the animals.

AZA Accreditation Standard
(11.3.3) Special attention must be given to free-ranging animals so that no undue threat is posed to the animal collection, free-ranging animals, or the visiting public. Animals maintained where they will be in contact with the visiting public must be carefully selected, monitored, and treated humanely at all times.

AZA Accreditation Standard
(11.3.1) All animal exhibits and holding areas must be secured to prevent unintentional animal egress.
be considered carefully to ensure that all areas are secure and particular attention must be given to shift
doors, gates, keeper access doors, locking mechanisms and exhibit barrier dimensions and construction.
In an emergency where the aquariums may become compromised jellies can be moved into a container for
transport (a plastic bag lined trash can filled with water is sufficient) to a safe holding facility.

Exhibits in which the visiting public may have contact with animals must have a guardrail/barrier that separates the two (AZA
Accreditation Standard 11.3.6).

All emergency safety procedures must be clearly written, provided to appropriate staff and volunteers, and readily available
for reference in the event of an actual emergency (AZA Accreditation Standard 11.2.3).

The most dangerous emergency would probably be someone being stung by the jelly. The severity of jelly stings varies greatly
amongst species. For instance, moon jellies rarely cause any sensation at all when touched with fingers. However, if someone
went swimming in a “smack” (swarm) of moon jellies and bare skin comes into contact with their tentacles, an itchy rash
can develop. For jellies that can cause a painful sting, like purple-striped jellies (Chrysaora colorata) or black sea nettles
(Chrysaora achlyos) one needs to be careful to reduce the chance of being stung. If a person is exposed to a jelly that stings
strongly, the procedure is to remove the tentacle (if there is one on the skin) with a pair of tweezers. The person removing the tentacle should wear latex gloves to reduce the danger of being stung themselves. Once the tentacle is removed, vinegar can be applied to the affected area to help keep remaining
tentacles from firing. The person should be monitored for the next 12–24 hours looking for symptoms that might develop. If symptoms do develop, the person should be sent to the doctor with a note of which species of jelly was responsible for the sting. It is important to remember that individuals’ reactions to jellyfish stings may be different. The typically benign moon jelly may elicit a strong allergic reaction or painful sting to a particularly sensitive individual.

Staff or volunteers feeding jellies should wear gloves to reduce the danger of being stung. There is no
danger of jellies stinging people outside their aquarium tanks. Security personnel may be called in to help
secure an area where a jelly aquarium burst and jellies are on the
ground.

Staff training for emergencies must be undertaken and records of such training maintained. Security personnel must be
trained to handle all emergencies in full accordance with the policies and procedures of the institution and in some cases, may
be in charge of the respective emergency (AZA Accreditation Standard 11.6.2).

Emergency drills should be conducted at least once annually for each basic type of emergency to ensure all staff are aware of
emergency procedures and to identify potential problematic areas that may require adjustment. These drills should be recorded and
evaluated to ensure that procedures are being followed, that staff training is effective and that what is learned is used to correct
and/or improve the emergency procedures. Records of these drills should be maintained and improvements in the procedures
duly noted whenever such are identified. AZA-accredited institutions must have a communication system that can be
quickly accessed in case of an emergency (AZA Accreditation Standard 11.2.4).

Staff should know who to call in an emergency and this procedure should be in the institutional safety manual, included in
new staff orientation and in ongoing safety meetings.

If someone shows an adverse reaction to being stung by a
jellyfish, they should go to the emergency room and make sure they tell medical personnel which species of jelly was responsible for the sting.

In general, jellyfish stings are not life threatening situations (except box jellies like *Chironex fleckeri*). A small minority of people may show an allergic reaction and should be treated by medical personnel.

AZA-accredited institutions must also ensure that written protocols define how and when local police or other emergency agencies are contacted and specify response times to emergencies (AZA Accreditation Standard 11.2.5).

**First Aid for Jellyfish Stings**

Severity of stings depends on the species of jellyfish, penetrating power of nematocysts, thickness of the exposed skin of the victim, and sensitivity of the victim to the venom. Penetration of a nematocyst into a victim is limited by the length of the tubule. Some nematocysts are unable to penetrate through human skin, so handlers will not feel a stinging sensation when handling these jellyfish. Areas of the body such as the palm of the hands and soles of the feet, where the skin is thicker, are not as affected by the nematocysts as compared to other sensitive areas of the body such as the eyes and wrist. Some people are more sensitive to the venom and may have allergic reactions (e.g., shortness of breath, slurred speech, disorientation, or unconsciousness). Medical attention should be sought immediately in these situations.

When stung by a jellyfish, carefully remove tentacles that are still on skin with seawater. Treat the affected areas with vinegar (acetic acid), alcohol, 1/4 strength household ammonia or baking soda, which deactivates attached nematocysts and prevents further envenomation. Apply liberally onto the skin. Use cold packs to reduce swelling. **Do not** use fresh water to remove tentacles; it will cause nematocysts to fire.

For more detailed information visit the website of the International Consortium of Jellyfish Stings (ICJS): [http://www.medschool.umaryland.edu/dermatology/jellyfish.asp](http://www.medschool.umaryland.edu/dermatology/jellyfish.asp)

In case of a jellyfish sting, make sure any tentacle or bell material is removed with forceps or other device to preclude the rescuer from additional stings. First aid should be administered as appropriate. If the person stung shows signs of an adverse reaction, emergency medical assistance should be summoned as soon as possible. First responders and medical personnel need to be told the kind of species of jelly involved. **Jellyfish do not attack**, but someone could be stung accidentally if they put parts of their bodies in the jellyfish aquarium tank.

Animal attack emergency response procedures must be defined and personnel must be trained for these protocols (AZA Accreditation Standard 11.5.3).

Animal attack emergency drills should be conducted at least once annually to ensure that the institution’s staff know their duties and responsibilities and know how to handle emergencies properly when they occur. All drills need to be recorded and evaluated to ensure that procedures are being followed, that staff training is effective, and that what is learned is used to correct and/or improve the emergency procedures. Records of these drills must be maintained and improvements in the procedures duly noted whenever such are identified (AZA Accreditation Standard 11.5.3).

If an animal attack occurs and injuries result from the incident, a written account outlining the cause of the incident, how the injury was handled, and a description of any resulting changes to either the safety procedures or the physical facility must be prepared and maintained for five years from the date of the incident (AZA Accreditation Standard 11.5.3).
Chapter 3. Transport

3.1 Preparations

Animal transportation must be conducted in a manner that adheres to all laws, is safe, and minimizes risk to the animal(s), employees, and general public (AZA Accreditation Standard 1.5.11). Safe animal transport requires the use of appropriate conveyance and equipment that is in good working order.

Equipment Inspection Routines and Protocols: Jellyfish are often shared among institutions; therefore, requiring transportation. Jellies should not be fed the day of transport in order to reduce the waste products which foul the water in the transport bag. The jellies are carefully dipped using a plastic beaker/jug into plastic bags filled with seawater from their aquarium tank. The bags are sealed tightly with rubber bands and without air. Some aquarists have found that saturating the water with oxygen has been a good way to increase survivability of jellies during shipping. If this is done, it is important to make sure all bubbles are gone before adding jellies (bubbles in the bags are sometimes entrained under the bell or ingested by the jelly adversely affecting normal belling). Double bagging helps prevent breakage. The size of the bag should be chosen according to the size of the jellies. For most species, there should be no more than four jellies per bag.

Standard fish bags 23 x 38 cm (9 x 15 in.), 3 mil bag holds 3 L (1 gal) of saltwater which will safely ship 1–4 juvenile 7.5 cm (3 in.) moon jellies, for example. Medusavores with a >7.5 cm (>3 in.) bell should be bagged individually.

- Each bag should be labeled with species, institution of origin, and date. The filled bags should be put into a Styrofoam box with packing material to serve as a shock absorbent. It is recommended that a large box bag be put into the Styrofoam box to confine water that might leak from the bags with jellies. This precaution will keep the corrugated box from losing integrity. To help maintain temperature, either an ice pack or warm pack should be put into the Styrofoam box to help maintain the temperature required by the jelly. The Styrofoam box should have a lid that is sealed with tape. The Styrofoam box should then be sealed into a corrugated box. The box should be labeled with the receiving institution’s address and the sender’s address. The sending institution should inform the receiving institution that the box is in transit. Most jellies need to be sent overnight express (2 day transport maximum). When packing the jellies, be sure to fill up any voids with packing material to secure the jellies from tumbling around inside the box. Keep the boxes under 18 kg (40 lb). Use plenty of tape to secure both the Styrofoam container and the outside cardboard box.

- Temperature control is the only parameter to try to maintain. For jellies, they can live for about 48 hours before they start showing adverse effects from lack of oxygen food, immobility, and temperature issues.

- Jellies are sent via overnight express and do not require support staff to travel with them. It is common for the boxes to be dealt with rather harshly by a shipping company despite warning labels to “handle with care.” Two suggestions to make the shipping successful are: Use smaller Styrofoam containers (inside of corrugated boxes) like the traditional 40.64 cm (16 in.) square boxes, or to use Igloo coolers and arrange to get them back later.

The equipment should provide for the adequate containment, life support, comfort, temperature control, food/water, and safety of the animal(s). Safe transport also requires the assignment of an adequate number of appropriately trained personnel (by institution or contractor) who are equipped and prepared to handle contingencies and/or emergencies that may occur in the course of transport. Planning and coordination for animal transport requires good communication among all affected parties, plans for a variety of emergencies and contingencies that may arise, and timely execution of the transport. At no time should the animal(s) or people be subjected to unnecessary risk or danger.
3.2 Protocols

Transport protocols should be well defined and clear to all animal care staff. Jellies that are taken off institutional grounds for education purposes should be kept in jars that are filled to the top to prevent bubbles. The temperature needs to be maintained with either ice packs or heat packs. Jellies should never be taken out of the water. Dipping them out with plastic bowls is the best way to move them from one container to another.

Acclimating the jellies once they arrive at the receiving institution is mandatory. As soon as the water temperature has equalized by floating the newly arrived and bagged jellies, conduct small 20% water changes inside the bags over 30 minutes to 1 hour. This will ensure salinity and pH have also equalized. Since jellies are comprised mostly of water and water quality parameters vary from institution to institution, allowing the jellies to adjust to their new environment takes time, patience, and observation. The jellies will demonstrate when they are ready to be released into their new environment—they will begin belling nice and evenly. One of the most obvious reactions of jellies to new aquarium water is that they will float or sink according to the specific gravity (salinity) of the seawater. If the salinity is higher than the salinity from which the jellies originated then they will float and if the salinity is lower, then the jellies will sink. Acclimation timeframes vary from species to species. Some acclimate very quickly while others do not. It is up to the jelly aquarist to observe and respond as needed.
Chapter 4. Social Environment

4.1 Group Structure and Size

Careful consideration should be given to ensure that animal group structures and sizes meet the social, physical, and psychological well-being of those animals and facilitate species-appropriate behaviors. Jellyfish are not social animals. With few exceptions, notably members of the Rhizostomae, only jellyfish of the same species are kept in an aquarium tank. It is also prudent to keep the sizes of the jellyfish about the same to reduce the likelihood of cannibalism (as seen in Aequorea sp) and because their flow requirements are equal.

4.2 Influence of Others and Conspecifics

Animals cared for by AZA-accredited institutions are often found residing with other animals of their species but may also be found residing with conspecifics. For the most part, jellyfish are displayed in single species groups. There has been some mixing of species of jellies in the same aquaria tanks with good results. If an aquarist does try a multispecies exhibit, close attention should be made to any deleterious effects and if detected, the jellies should be put back into single species tanks. Deleterious effects may include bell flattening, bell everting, flaccid oral arms, pitted bells, holes in the bell, or sloughing off of exumbrellar surface.

Jellies should not be kept with other types of animals (e.g., fish) or plants due to the danger of being caught on, torn or otherwise damaged. A notable exception is the upside down jelly (Cassiopea sp.), which does well with other shallow water tropical invertebrates and fish.

4.3 Introductions and Reintroductions

Managed care for and reproduction of animals housed in AZA-accredited institutions are dynamic processes. Animals born in or moved between and within institutions require introduction and sometimes reintroductions to other animals. It is important that all introductions are conducted in a manner that is safe for all animals and humans involved.

In general, medusoid jellies are moved from smaller tanks when young to larger tanks as they grow. There is no social structure in jellyfish. Medusoid jellies should be put with others of their own species and roughly the same size. Some medusoid jellyfish have the ability to eat younger stages.

Every attempt should be taken to keep the polyp stage in single species enclosures. Some polyps can overgrow and kill other species.

Jellies should be acclimated slowly when moving them to a new aquarium tank. They can usually adjust if water from the tank they will be put into is slowly added over a couple hours. One of the most obvious reactions of jellies to new aquarium water is that they will float or sink according to the specific gravity (salinity) between the jelly and the aquarium water.
5.1 Nutritional Requirements

A formal nutrition program is recommended to meet the nutritional and behavioral needs of all jellyfish (AZA Accreditation Standard 2.6.2). Diets should be developed using the recommendations of nutritionists, the Nutrition Scientific Advisory Group (NAG) feeding guidelines (http://www.aza.org/nutrition-advisory-group/), and veterinarians as well as AZA Taxon Advisory Groups (TAGs), and Species Survival Plan® (SSP) Programs. Diet formulation criteria should address the animal’s nutritional needs, feeding ecology, as well as individual and natural histories to ensure that species-specific feeding patterns and behaviors are stimulated.

In their planktonic environment, sea jellies are opportunistic in their feeding habits. They can and do feed 24 hours a day in the wild. Most sea jellies feed on smaller zooplankton. They can grow very fast in the wild. Some sea jellies get most of their nutrition from symbiotic phytoplankton that inhabits their epidermal tissues called zooxanthellae (Hofmann & Kremer, 1981; Rahat & Adan, 1980). For those jellies, lights with proper wavelengths are critical to keeping them healthy in public displays (see Chapter 1.2). For one of the best overviews on sea jelly nutrition in aquarium settings, please refer to How to Keep Jellyfish in Aquariums by Chad Widmer. Chad includes recommended food for many species of sea jellies commonly displayed in public aquariums and zoos.

Although virtually all sea jellies are carnivorous, there are a variety of foods that have been found to work well for culture and display depending upon species. Also, the various life stages of sea jelly development need to be fed food of appropriate size and type.

The scientific literature includes little information on species specific nutrient requirements, but there are some scientific papers that have investigated nutritional composition of enrichments/supplements and potential prey items (Sullivan et al., 1994; Sullivan et al., 1997; Yamamoto, 1996; Bamstedt et al., 2001), metabolic rates which can dictate feeding requirements (Larson, 1987; Schneider, 1989; Bailey et al., 1995), stomach contents (Graham & Kroutil, 2001; Ishii & Tanaka, 2001) and digestion times (Heeger & Miller, 1987; Titleman & Hansson, 2006).

Aquarists have come up with a variety of live and frozen food that have allowed the culture of the many species of jellies commonly exhibited in aquariums. The basic food types include:

- Rotifers
- Artemia (brine shrimp) nauplii
- Mysids
- Krill (euphausiids)
- Blood worms
- Blended fish
- Fish eggs
- Larval fish
- Wild plankton
- Other jellies (commonly moon jellies, chopped to appropriate size.)

All carnivorous sea jellies are planktivores (i.e., they eat plankton) and some are also medusavores (i.e., they eat other jellies). Some medusavores will not grow unless they are fed other jellies. The most commonly used jelly to feed medusavore jellies held in public aquariums is moon jellies (Aurelia sp.). There are a few studies that have investigated the nutritional value of moon jellies (Rackmil et al., 2009; Fukuda & Naganuma, 2001; Martinussen & Bamstedt, 1999; Hansson, 1997). For this reason, aquarists that want to display medusavore jellies should have a ready supply of moon jellies to use as food. Medusavore jellies include the genera: Chrysaora, Cyanea and Phacellophora.

The hydrozoan sea jellies are almost all small (1-5mm) and planktivorous. They are fed enriched rotifers, enriched Artemia nauplii, and wild plankton. One common hydrozoan sea jelly in public displays is the crystal jelly Aequorea victoria. In order for the crystal jelly to display its bioluminescence it needs to be fed food that has the light emitting compound luciferin (Haddock et al., 2001).
The polyp stages of sea jellies are very small (0.2–1.0 cm tall) and they have very small mouths. The preferred foods for polyps are rotifers with occasional enriched Artemia nauplii. Many of the medusavore sea jelly polyps will not grow without being fed finely diced pieces of moon jellies (Hiromi et al., 1995).

The idea behind enrichment is to gut load rotifers and Artemia nauplii so the metabolized and stored food (mostly lipids or fats) can be consumed by the jelly polyp, ephyra or medusa. Without enrichment there is very little nutritional value in rotifers and Artemia nauplii. The most common enrichment media include live phytoplankton (various species like Isochrysis and Nannochloropsis) and commercially available media (e.g., Algamac 3050 from Aquafauna or Selco from INVE).

When hatching Artemia nauplii, it can be difficult to separate them from the waste cyst material. The cyst capsule pieces can be eaten by jellies, but have no nutritional value. The separation process can be eliminated by subjecting the cysts to a decapsulation process. By decapsulating the cysts, the hard outside covering of the cyst is dissolved. The decapsulated nauplii can be refrigerated for about a month or more and still hatch. Once hatched, the nauplii are put into seawater spiked with the enrichment media. The nauplii are allowed to feed for about 24-hours. They are then rinsed and fed out. The nutritional value of the nauplii diminishes the longer they are out of the enrichment media, as they use the stored food for their own growth and development.

5.2 Diets

The formulation, preparation, and delivery of all diets must be of a quality and quantity suitable to meet the animal’s psychological and behavioral needs (AZA Accreditation Standard 2.6.3). Food should be purchased from reliable, sustainable and well-managed sources. The nutritional analysis of the food should be regularly tested and recorded.

Many institutions have fine-tuned their nutritional requirements based upon the response seen in the jellies they keep. Jellies are opportunistic feeders. They catch small prey items on their tentacles either by stinging the prey or the prey being caught up in the discharged nematocyst. In general, jellies need to be fed at least every other day, but most aquariums feed them every day since in the wild they are attempting to feed 24-hours a day. Some feed their jellies twice a day or more.

Jellies are fed multiple food sources depending upon the species. Moon jellies can be kept by just feeding them Artemia nauplii. For best results most institutions enrich the Artemia nauplii with various media. The most commonly used is Super Selco or algal paste (Nannochloropsis). Some jellies are medusavores in that they eat other jellies. Medusavores are often fed smaller, whole or cut up pieces of moon jellies (Aurelia sp.).

Regulations on storage and thawing of seafood also apply to organisms used for jelly diets.

Food preparation must be performed in accordance with all relevant federal, state, or local regulations (AZA Accreditation Standard 2.6.1). Meat processed on site must be processed following all USDA standards. The appropriate hazard analysis and critical control points (HACCP) food safety protocols for the diet ingredients, diet preparation, and diet administration should be established for the taxa or species specified. Diet preparation staff should remain current on food recalls, updates, and regulations per USDA/FDA. Remove food within a maximum of 24 hours of being offered unless state or federal regulations specify otherwise and dispose of per USDA guidelines.

If browse plants are used within the animal’s diet or for enrichment, all plants must be identified and assessed for safety. The responsibility for approval of plants and oversight of the program should be assigned to at least one qualified individual (AZA Accreditation Standard 2.6.4). The program should identify if the plants have been treated with any chemicals or near any point sources of pollution and if the plants are safe for the jellyfish. If animals have access to plants in and around their exhibits, there should be a staff member responsible for ensuring that toxic plants are not available.
5.3 Nutritional Evaluations

The health of jellies is determined by observations compared to the condition seen in the wild. Some of the abnormalities to look out for are bell flattening, or bell deformities such as curling of the margin, bell evert ing, flaccid oral arms, pitted bells, holes through the bell, belling abnormally, or sloughing off of exumbrellar surface.
6.1 Veterinary Services

Veterinary services are a vital component of excellent animal care practices. A full-time staff veterinarian is recommended, however, in cases where this is not practical, a consulting/part-time veterinarian must be under contract to make at least twice monthly inspections of the animal collection and to any emergencies (AZA Accreditation Standard 2.1.1). Veterinary coverage must also be available at all times so that any indications of disease, injury, or stress may be responded to in a timely manner (AZA Accreditation Standard 2.1.2). All AZA-accredited institutions should adopt the guidelines for medical programs developed by the American Association of Zoo Veterinarians (AAZV) www.aazv.org/associations/6442files/zoo_aquarium_vet_med_guidelines.pdf.

Protocols for the use and security of drugs used for veterinary purposes must be formally written and available to animal care staff (AZA Accreditation Standard 2.2.1).

Procedures should include, but are not limited to: a list of persons authorized to administer animal drugs, situations in which they are to be utilized, location of animal drugs and those persons with access to them, and emergency procedures in the event of accidental human exposure.

Few veterinarians have aquatic invertebrates as their specialty much less know a lot about jelly health. The condition of jellies should be monitored daily and a veterinarian should be consulted for advice if jellies appear differently from their wild condition.

Drugs are not commonly used with jellies. However, there has been some work done on removing parasites with the antibiotic milbemycin oxime and attempts to cure bell rot with tetracycline baths have been attempted with mixed results (see Chapter 6.7).

Jellies do not have accepted health-related factors at this time, nor does any agency regulate their care.

Animal recordkeeping is an important element of animal care and ensures that information about individual animals and their treatment is always available. A designated staff member should be responsible for maintaining an animal record keeping system and for conveying relevant laws and regulations to the animal care staff (AZA Accreditation Standard 1.4.6). Recordkeeping must be accurate and documented on a daily basis (AZA Accreditation Standard 1.4.7). Complete and up-to-date animal records must be retained in a fireproof container within the institution (AZA Accreditation Standard 1.4.5) as well as be duplicated and stored at a separate location (AZA Accreditation Standard 1.4.4).

6.2 Identification Methods

Ensuring that jellyfish are identifiable through various means increases the ability to care for individuals more effectively. Animals must be identifiable and have corresponding ID numbers whenever practical, or a means for accurately maintaining animal records must be identified if individual identifications are not
practical (AZA Accreditation Standard 1.4.3).

The only animal record for jellies is the number of the different species and an approximate number of each species. Polyps should be recorded as separate colonies. Entire enclosures or even species could typically be reflected as groups in ISIS/ZIMS.

Inventory of jellyfish should follow institutional procedures on acquisitions and dispositions, which are done at least annually. Medusoid jellyfish are sometimes in such large numbers that their numbers are approximated or listed as "too numerous to count (TNTC)." Polyps are virtually impossible to monitor due to their great capacity to bud new polyps asexually and are usually referred to as a colony.

AZA member institutions must inventory their jellyfish population at least annually and document all jellyfish acquisitions and dispositions (AZA Accreditation Standard 1.4.1). Transaction forms help document that potential recipients or providers of the animals should adhere to the AZA Code of Professional Ethics, the AZA Acquisition/Disposition Policy (see Appendix B), and all relevant AZA and member policies, procedures and guidelines. In addition, transaction forms must insist on compliance with the applicable laws and regulations of local, state, federal and international authorities. All AZA-accredited institutions must abide by the AZA Acquisition and Disposition policy (Appendix B) and the long-term welfare of animals should be considered in all acquisition and disposition decisions. All species owned by an AZA institution must be listed on the inventory, including those animals on loan to and from the institution (AZA Accreditation Standard 1.4.2).

6.3 Transfer Examination and Diagnostic Testing Recommendations

The transfer of animals between AZA-accredited institutions or certified related facilities due to AZA Animal Program recommendations occurs often as part of a concerted effort to preserve these species. These transfers should be done as altruistically as possible and the costs associated with specific examination and diagnostic testing for determining the health of these animals should be considered.

Aquariums and zoos commonly share jellies. A simple review of morphological characteristics such as bell, oral arm, and tentacle morphology is commonly employed to determine if the animals are acceptable for transfer.

6.4 Quarantine

AZA institutions must have holding facilities or procedures for the quarantine of newly arrived animals and isolation facilities or procedures for the treatment of sick/injured animals (AZA Accreditation Standard 2.7.1). All quarantine, hospital, and isolation areas should be in compliance with AZA standards/guidelines (AZA Accreditation Standard 2.7.3; Appendix C). All quarantine procedures should be supervised by a veterinarian, formally written and available to staff working with quarantined animals (AZA Accreditation Standard 2.7.2). If a specific quarantine facility is not present, then newly acquired animals should be kept separate from the established collection to prohibit physical contact, prevent disease transmission, and avoid aerosol and drainage contamination. If the receiving institution lacks appropriate facilities for quarantine, pre-shipment quarantine at an AZA or American Association for Laboratory Animal Science (AALAS) accredited institution may be applicable.
Local, state, or federal regulations that are more stringent than AZA Standards and recommendation have precedence.

Newly acquired jellies should be put into quarantine aquarium tanks off exhibit, if possible, and observed for a few days to assure they are in good health. Their health can be determined by observing normal morphology, movement, and feeding for the species. Initial observations determine the presence of damage from collecting or shipping, and the presence of any possible pathogens or parasites.

Feeding equipment, such as basters, containers, and nets, should be rinsed well with freshwater and allowed to dry between uses. This practice limits the possibility of introducing infectious diseases or planulae from other jelly species being unintentionally introduced into a system. Other methods to prevent cross-contamination of planulae, parasites, or diseases include bleaching all tools and utensils once a week. Color coding tools and utensils to use with respective jelly culture systems further reduces cross-contamination of cultures and populations.

Jellies should be in quarantine until they eat regularly, which could be a couple of days to a week. Jellies in quarantine should be observed for a few days to assure they are in good health. There are no recommended diagnostic tests for determining the health of jellies in quarantine. Minor bell damage often begins to heal noticeably in a few days, allowing microbial causes to be eliminated from consideration.

The most common ectoparasites of jellies are copepods or hyperiid amphipods. There are no generally accepted methods for removing ectoparasites. Suggested methods include freshwater dips to cause the ectoparasites to fall off the jelly or physically removing them one by one with a pair of forceps. Crossley et al. (2009) describe a treatment with diflubenzuron that shows promise as a way to eradicate hyperiid amphipods from jellies. When jellies exhibit normal morphology, movement, and feeding for the species they can be moved into exhibit tanks.

There are no local or federal laws concerning necropsy procedures for jellies. Neither are there published or accepted procedures for necropsy procedures for jellies. Medusoid stages of jellies are relatively short-lived animals (most less than one year). Senescence is common in a grouping of jellies of mixed ages and may also be seen in jellies in quarantine. Examination for parasites and possible culture for bacteria are a couple of suggestions if a condition of the entire group suggests an issue.

AZA institutions must have zoonotic disease prevention procedures and training protocols established to minimize the risk of transferable diseases (AZA Accreditation Standard 11.1.2) with all animals, including those newly acquired in quarantine. Keepers should be designated to care only for quarantined animals if possible. If keepers must care for both quarantined and resident animals of the same class, they should care for the quarantined animals only after caring for the resident animals. Equipment used to feed, care for, and enrich animals in quarantine should be used only with these animals. If this is not possible, then all items must be appropriately disinfected, as designated by the veterinarian supervising quarantine before use with resident animals.

If possible with tanks appropriate for jellies available quarantine durations should span of a minimum of 30 days (unless otherwise directed by the staff veterinarian). If additional mammals, birds, reptiles, amphibians or fish of the same order are introduced into their corresponding quarantine areas, the minimum quarantine period must begin over again. However, the addition of mammals of a different order to those already in quarantine will not require the re-initiation of the quarantine period.

During the quarantine period, specific diagnostic tests should be conducted with each animal if possible or from a representative sample of a larger population (e.g., birds in an aviary or frogs in a terrarium) (see Appendix C). A complete physical, including a dental examination if applicable, should be performed. Animals should be evaluated for ectoparasites and treated accordingly. Blood should be collected, analyzed and the sera banked in either a -70 °C (-94 °F) freezer or a frost-free -20 °C (-4 °F) freezer for retrospective evaluation. Fecal samples should be collected and analyzed for gastrointestinal parasites and the animals should be treated accordingly. Vaccinations should be updated as appropriate, and if the vaccination history is not known, the animal should be treated as immunologically naive and given the appropriate series of vaccinations.

A tuberculin testing and surveillance program must be established for animal care staff as appropriate to protect both the health of both staff and animals (AZA Accreditation Standard 11.1.3).

AZA Accreditation Standard (11.1.2) Training and procedures must be in place regarding zoonotic diseases.

AZA Accreditation Standard (11.1.3) A tuberculin testing and surveillance program must be established for appropriate staff in order to ensure the health of both the employees and the animal collection.
Depending on the disease and history of the animals, testing protocols for animals may vary from an initial quarantine test to yearly repetitions of diagnostic tests as determined by the veterinarian. Animals should be permanently identified by their natural markings or, if necessary, marked when anesthetized or restrained (e.g., tattoo, ear notch, ear tag, etc.). Release from quarantine should be contingent upon normal results from diagnostic testing and two negative fecal tests that are spaced a minimum of two weeks apart. Medical records for each animal should be accurately maintained and easily available during the quarantine period.

If a jellyfish should die in quarantine, a necropsy should be performed on it and the subsequent disposal of the body must be done in accordance with any local or federal laws (AZA Accreditation Standard 2.5.1). Necropsies should include a detailed external and internal gross morphological examination and representative tissue samples from the body organs should be submitted for histopathological examination (see Chapter 6.7).

### 6.5 Preventive Medicine

AZA-accredited institutions should have an extensive veterinary program that must emphasize disease prevention (AZA Accreditation Standard 2.4.1). The American Association of Zoo Veterinarians (AAZV) has developed an outline of an effective preventative veterinary medicine program that should be implemented to ensure proactive veterinary care for all animals (www.aazv.org/associations/6442/files/zoo_aquarium_vet_med_guidelines.pdf).

Few veterinarians specialize in aquatic invertebrates, much less know a lot about jelly health, which is still an emerging science. Unlike many invertebrates studied as aquaculture subjects, few studies of disease in medusae exist. The condition of jellies should be monitored daily and a veterinarian should be consulted for advice if jellies exhibit behavior abnormal from the wild condition. Water quality maintenance/general tank and life support system maintenance is always a good way to maintain healthy jellies. It has been said, “cleanliness is close to jelliness.”

As stated in the Chapter 6.4, AZA institutions must have zoonotic disease prevention procedures and training protocols established to minimize the risk of transferable diseases (AZA Accreditation Standard 11.1.2) with all animals. Keepers should be designated to care for only healthy resident animals, however if they need to care for both quarantined and resident animals of the same class, they should care for the resident animals before caring for the quarantined animals. Care should be taken to ensure that these keepers are “decontaminated” before caring for the healthy resident animals again. Equipment used to feed, care for, and enrich the healthy resident animals should only be used with those animals.

Zoonotic diseases have not been identified that are specific to jellies. General precautions are the same as for any aquarium. Vigilance is needed regarding aquatic water borne zoonotic sources like *Mycobacterium marinum* (e.g., wash hands, avoid contact if immunocompromised, and wear gloves).

Animals that are taken off zoo/aquarium grounds for any purpose have the potential to be exposed to infectious agents that could spread to the rest of the institution’s healthy population. AZA-accredited institutions must have adequate protocols in place to avoid this (AZA Accreditation Standard 1.5.5).

Jellies are not often used as program animals and if they are, they should not be taken out of the seawater they are kept in from their original exhibit. No human or environmental infectious agents are known to be transmittable to or from jellies.

Also stated in Chapter 6.4, a tuberculin testing and surveillance program must be established for animal care staff, as appropriate, to protect the health of both staff and animals (AZA Accreditation Standard 11.1.3). Depending on the disease and history of the animals, testing protocols for animals may vary from
an initial quarantine test, to annual repetitions of diagnostic tests as determined by the veterinarian. To prevent specific disease transmission, vaccinations should be updated as appropriate for the species.

Tuberculin testing is not recommended or required for animal care staff working with jellies. While TB testing is not needed for working with fishes and aquatic invertebrates, testing sometimes becomes an issue at institutions that also house tetrapods. Misleading TB test positives have been reported in individuals that have been previously infected with *Mycobacterium marinum* (i.e., fish tank granuloma). *M. marinum* has low pathogenicity in humans, and typically is limited to skin lesions.

### 6.6 Capture, Restraint, and Immobilization

The need for capturing, restraining and/or immobilizing an animal for normal or emergency husbandry procedures may be required. All capture equipment must be in good working order and available to authorized and trained animal care staff at all times (AZA Accreditation Standard 2.3.1).

Jellies can be harmed if picked up by hand or net. Their bodies need to be in water at all times. Most aquarists move them in bowls or plastic bags. Maneuver the animal to the surface using a swirling or vortex motion with your hand or extension "wand" (e.g., PVC pipe) in a clock-wise motion if it is not circumnavigating the tank normally and arriving at the surface on its own. Once at the top, use gentle but constant action drawing water slowly into the container or bag, pulling the jelly with it.

### 6.7 Management of Diseases, Disorders, Injuries and/or Isolation

AZA-accredited institutions should have an extensive veterinary program that manages animal diseases, disorders, or injuries and has the ability to isolate these animals in a hospital setting for treatment if necessary. Jellyfish keepers should be trained for meeting the animal’s dietary, husbandry, and enrichment needs, as well as in restraint techniques, and recognizing behavioral indicators animals may display if their health becomes compromised (AZA Accreditation Standard 2.4.2). Protocols should be established for reporting these observations to the veterinary department. Jellyfish hospital facilities should contain appropriate equipment and supplies on hand for treatment of diseases, disorders or injuries, and have staff available that are trained to address health issues, manage short and long term medical treatments and control for zoonotic disease transmission.

Jellies can exhibit a variety of abnormal morphology or behaviors that can be indicators of poor health. Jellies showing abnormal morphology for the species (e.g., tattered or shrinking bells, oral arms too long or too short or hanging abnormally, everted bells, loss of tentacles) or abnormal behavior (ineffective bell movement, general lethargy or lack of feeding) should be considered for removal to quarantine areas if available. Those jellies that show no improvement should be considered for euthanizing.

Very little is known about jellyfish diseases. Most of what has been observed and published has been about parasites of jellies. Often organisms are found in some type of close relationship with a jelly where it is not known if the organism is truly a parasite or simply a commensal; or if the observation is a random chance encounter that is rarely found in nature. Several reviews of diseases and parasites of jellyfish have been published and are cited in the reference section. Chapter 6 (*Diseases of Cnidaria*) from *Diseases of Marine Animals* Volume 1 (Kinne, 1980) has been used extensively for this chapter. The paper by Laval (1980) on hyperiid amphipods as parasitoids to gelatinous zooplankton was also used to a large degree. Due to a general scarcity of published material, personal communication has also been used and sources are cited throughout the text.

Although bacterial diseases have been discovered infecting coral reefs and incidents of infections of corals in aquariums are not uncommon, there is very little evidence of viral, bacterial, or fungal disease of marine cnidarians in the wild. Occasionally medusae, especially larger and/or older medusae, develop lesions or irregularities on the surface or at the margins of the bell. This is not uncommon, and is often
referred to as “bell rot.” It could be a dimpling of the surface or small “craters,” which can eventually descend into the viscera. Sometime there is an erosion of the margins and loss of marginal tentacles. One aquarium reported the development of a white caseous material forming on the surface along with the lesions, but no positive identification was made (S. Crossley, personal communication). Bacterial involvement in these bell problems is the subject of much discussions and speculation. At this point, there is no proven cause and effect relationship between bacteria and lesions on the bells of jellies, although it cannot be ruled out. There is also a possibility of secondary bacterial infection after the occurrence of some insult to the integument. It is difficult to obtain accurate isolated bacterial cultures from jellyfish.

A treatment that has been reported to have some success is a 5-day course of 2-hour tetracycline baths, using the drug at 20 ppm (Raskoff et al., 2003). In the reported case where the caseous material developed, bacterial cultures were collected from the material. Two species of *Pseudomonas* and a *Streptococcus* were isolated. The cultured bacterial isolates were found to be sensitive to tetracycline. The jellies seemed to respond initially to the treatment described above, but ultimately died (S. Crossley, personal communication). At another aquarium, some success treating bacterial infections with the tetracycline treatment was observed, but after treatments the jellies slowly senesced (B. Upton, personal communication). An additional aquarium reports success when treating *Cyanea* and *Catostylus* for bacterial infections using oxytetracycline and enrofloxacin (2 mg/L). The treatments were 6-hour baths every other day for a total of 3 treatments. At this time, there is much research still to be done to find reliable antibiotic treatments for treating bell erosion problems found in jellies in aquariums.

Although protozoans cause many common and well-known health problems in fishes and some other aquatic taxa, there is little evidence of protozoans causing health problems in scyphozoan or marine hydrozoans. Ciliates and other protozoans may be found secondarily in lesions, but there is no evidence that they are the cause of such lesions.

Intermediate stages of digenetic trematodes have been reported in several species of scyphozoan jellies, including *Chrysaora quinquecirrha*, *Pelagia noctiluca*, and *Cotylorhiza tuberculata*. There have also been reports of digenes in a number of hydrozoan jellies. Fishes are the definitive host of these reported digene species, which ultimately take up residence in the host’s gastrointestinal system. There are numerous species of fishes that associate with and/or prey upon jellies (Arai, 1988) and jellies may be important intermediate hosts for a number of trematode species.

The cabbagehead (or cannonball) jelly (*Stomolophus meleagris*) is sometimes infected by cestode larva (Phillips & Levin, 1973). The larva burrows in mesogleal tissue; large bacterial populations are associated with some of these burrows. It is assumed that fishes serve as the definitive host for the cestode, as a large variety of fishes associate with *Stomolophus* and/or utilize it as a food source. Cestodes have also been reported to infect Ctenophores, including *Beroe* and *Pleurobrachia* (Theodorides, 1989).

Although it is of interest to aquarists collecting and displaying certain species of jellies from the wild, it is unlikely that trematodes or cestodes would be a major problem for aquarists keeping jellies in closed systems using cultured animals. The burrowing sea anemone (*Peachia quinquecapitata*) can be found as an ectoparasite on hydrozoan jellyfish. Spaulding (1972) reviewed this anemone’s life cycle and concluded that it is an obligate parasite during its larval development. The *Peachia* planula is ingested by the host and spends a short period of time in the radial canals before moving and attaching to the outside. There it feeds on gonads and other body tissue of the medusa host. In the laboratory, *Peachia* were able to move from one jelly host to another if given the opportunity.

On the Pacific coast of the U.S., larval *Peachia quinquecapitata* can be found on several species of hydromedusae (Spaulding, 1972; D. Wrobel, personal communication). Other species of *Peachia* have been observed attached to jellies in European and Australian waters (Lauckner, 1980). *Peachia* adults are free-living. They burrow in sand or gravel after leaving their host medusa by falling off or after the death of the host.

The larva of another burrowing sea anemone, *Edwardsiella lineata*, (until recently, known as *Edwardsia lineata*) is a common parasite of the lobate ctenophore *Mnemiopsis leidyi* along the east coast of the US. It burrows into the mesoglea and inserts its mouth into the aboral end of the ctenophore’s pharynx. There it feeds on food previously ingested by the ctenophore. This anemone parasite exhibits a high degree of host specificity, and as many as 30 individuals per host have been found (Bumann & Puls, 1996). When parasitized by *Edwardsia*, growth in *Mnemiopsis* is insignificant or negative (Bumann & Puls, 1996). In a laboratory observation, Crowell (1976) observed liberated *Edwardsia* enter uninfected *Mnemiopsis*. This has significance to the jelly culture aquarist. It is recommended that infected
Mnemiopsis be removed from a population of uninfected animals if possible. It is often not possible to do this, as the percentage of infected Mnemiopsis in the wild is very high when this parasite appears. Due to the high host specificity, it is unlikely that Edwardsia will spread to other jellies in a collection. At one aquarium, they have never been observed in any jelly other than Mnemiopsis in over a period of roughly 14 years. This despite the frequent holding and exhibition of infected Mnemiopsis in systems linked with other tanks holding scyphozoan and hydrozoan medusae and other species of ctenophores.

The most commonly encountered parasites of jellyfish and the ones that are of most concern for purposes of husbandry are crustaceans, especially the hyperiid amphipods. Other crustaceans that parasitize or associate with jellies are the pedunculate (or stalked) barnacles, most commonly Alepas pacifica, and several species of decapod crustaceans which “hitchhike” on medusae, especially during early stages of their lives. In addition, one case of a sea spider (pycnogonid) has been reported as a parasite on the mesopelagic scyphomedusae Periphylla periphylla (Child & Harbison, 1986).

Pages (2000) reviewed the literature on barnacles attached to jellies in the wild. Most reports were of the pedunculate barnacle Alepas pacifica attached to at least nine species of jellies, including Pelagia noctiluca and Phacellophora camtschatica. A. pacifica may also be found on Chrysaora colorata (C. Widmer, personal communication). Geographical location is widespread, encompassing the Atlantic, Pacific, and Indian Oceans. This stalked barnacle has highly reduced calcified plates, apparently to decrease its weight as a modification for attachment to swimming jellies. Pages proposed that the barnacle feeds upon the gonads of the jellies. Observations of the barnacle manipulating and removing captured food from the tentacles of medusae have been observed (C. Widmer, personal communication).

Decapod crustaceans commonly “hitchhike” upon jelly medusae, especially when they are in a juvenile stage. On the Pacific coast of the U.S., larval and juvenile slender crabs (Cancer gracilis) can often be found on larger scyphozoans like Chrysaora colorata and Phacellophora camtschatica before assuming their normal free-living benthic lifestyle (D. Wrobel, personal communication). On the Atlantic Coast of the U.S., larval and juvenile spider crabs (Libinia dubia) take up residence on Aurelia aurita and Stomolophus meleagris in much the same fashion as C. gracilis does. (Jachowski, 1963; Tunberg & Reed, 2004) Adult L. dubia are free-living. These crabs will eat food gathered by the host jelly, but may also feed on host tissue. There may be several individuals residing on a single medusa.

When found on a collected jelly, these decapod crabs can usually be removed quite easily with some blunt forceps. They do not attach to the jelly and are easily manipulated into a position where removal can be done without injury to the jelly. Since these crabs are only found on jellies during their immature stages, there is no danger of larvae being left behind.

Amphipods of the suborder Hyperidae are entirely marine and pelagic, and not normally found inshore; as compared to the more commonly encountered amphipods of the suborder Gammaridae, which are almost entirely benthic and are common inshore. The hyperiids also differ from the gammarids in that they have large heads made up almost entirely of a pair of eyes. As stated earlier, the hyperiid amphipods are the most common and the most serious parasite encountered by aquarists collecting or exhibiting wild-collected jellies. There is ample documentation of infestation of both scyphozoan and hydrozoan jellies by hyperiids. Hyperiids are also known to infect ctenophores, siphonophores, and salps. Especially common on jellyfish are those of the genus Hyperia. Hyperia galba is one of the most well-known species, and has a cosmopolitan distribution. It has been reported in association with Aurelia aurita, Cyanea capillata, Pelagia noctiluca and several other scyphozoan species (Kinne, 1980). Hyperia medusarum can often be found attached to jellies of the Pacific coast of the U.S. Hyperiid amphipods are commonly found on Chrysaora fuscescens, C. colorata, Aequorea sp., Phacellophora camtschatica, Aurelia aurita, and A. labiata. There may very well be other species that harbor these amphipods, especially if collecting is being done offshore. Members of the genus Hyperoche have also been associated with jellies, especially H. medusarum that parasitizes medusae and occasionally ctenophores. At one aquarium C. gracilis was allowed to be on C. fuscescens on display to eat hyperiid amphipods that might come in with the jellyfish. The crabs were removed when they reached a size that their legs might damage the soft jelly bells (C. Widmer, personal communication).

It has been proposed by Laval (1980) that nearly all hyperiids have evolved to utilize gelatinous zooplankton as a “pelagic substratum allowing the continuation of a benthic-like existence” similar to their gammarid relatives. The gelatinous zooplankton they associate with can be thought of as “islands in the ocean, providing sites of attachment, food, and shelter.” This implies that nearly all hyperiids are parasites on gelatinous animals, for at least part, if not all of their lives. Some may be thought of as commensals, but it is likely that there is always some detrimental effect upon the jellies that they associate with.
Hyperiids may eat prey items captured by the jelly, thus robbing the jelly of food; or, if food supply is not adequate, it may feed directly upon the jelly itself. Larvae and juveniles are often found embedded in the mesoglea, or in natural cavities of their host jelly. It is difficult to imagine any benefit to a jellyfish by harboring these amphipods.

When hyperiid amphipods are found on jellies, they can usually be physically removed rather easily with some blunt forceps, care being taken to not damage or injure the host jelly. Adults are often moving about freely upon the bell or on the sub-umbrellar surface, and are not attached to the jelly. They should be removed from the tank immediately. The host jelly should be re-examined frequently for several weeks, as sub-adult stages may have been missed upon the initial examinations and removal of the amphipods. Often more amphipods are noticed several weeks after the removal of all conspicuous adults. Jellies can usually recover from any damage caused by the amphipods if the infestation is not too heavy and if the situation is remedied immediately upon discovery.

Crossley et al. (2010) found that the use of diflubenzuron to eradicate hyperiid parasites from scyphomedusae to be a safe and useful option when properly applied in a controlled environment.

Little is known about host specificity. Since some species such as Hyperia galba have been reported residing on a number of jelly hosts, it is assumed that these amphipods could infect species of jellies other than the ones initially found on. Hyperiids may be found free-living in tanks that had previously held parasitized jellies after the removal or death of those jellies. It is advised that tanks be well disinfected with chlorine after housing jellies known to be hosting hyperiid amphipods.

There are no social or behavioral problems associated with jellies, as they do not possess a central nervous system. Jellyfish welfare is determined by observing locomotion and eating habits. If abnormal locomotion or eating habits are observed, steps should be taken to determine the cause. Once the cause is determined and fixed, staff should share their findings appropriately in their institution and to the larger community of jelly aquarists. Welfare concerns should be communicated to the Institutional Welfare Committee.

AZA-accredited institutions must have a clear process for identifying and addressing jellyfish animal welfare concerns within the institution (AZA Accreditation Standard 1.5.8) and should have an established Institutional Animal Welfare Committee. This process should identify the protocols needed for animal care staff members to communicate animal welfare questions or concerns to their supervisors, their Institutional Animal Welfare Committee or if necessary, the AZA Animal Welfare Committee. Protocols should be in place to document the training of staff about animal welfare issues, identification of any animal welfare issues, coordination and implementation of appropriate responses to these issues, evaluation of the outcome of these responses (and adjustment of these responses if necessary), and the dissemination of the knowledge gained from these issues.

AZA-accredited zoos and aquariums provide superior daily care and husbandry routines, high quality diets, and regular veterinary care, to support jellyfish longevity. In the occurrence of death however, information obtained from necropsies is added to a database of information that assists researchers and veterinarians in zoos and aquariums to enhance the lives of jellyfish both in their care and in the wild. As stated in Chapter 6.4, necropsies should be conducted on deceased jellyfish to determine their cause of death, and the subsequent disposal of the body must be done in accordance with local, state, or federal laws (AZA Accreditation Standard 2.5.1). Necropsies should include a detailed external and internal gross morphological examination and representative tissue samples from the body organs should be submitted for histopathological examination. Many institutions utilize private labs, partner with Universities or have their own in-house pathology department to analyze these samples. The AZA and American Association of Zoo Veterinarians (AAZV) website should be checked for any AZA Jellyfish Program approved active research requests that could be filled from a necropsy.

The most humane euthanasia protocol for jellies is for them to be placed in a bath of MS-222. Once deceased, the jellies should be considered for educational purposes in an exhibit or education program. The most common cause of death in jellies in aquariums is senescence. Necropsy procedures are not defined for jellies. It is recommended that a general review of the morphology should be recorded.
7.1 Reproductive Physiology and Behavior

It is important to have a comprehensive understanding of the reproductive physiology and behaviors of the animals in our care. This knowledge facilitates all aspects of reproduction, artificial insemination, birthing, rearing, and even contraception efforts that AZA-accredited zoos and aquariums strive to achieve.

Most common cnidarian jellies, and almost all that are displayed by public aquariums, exhibit two morphologic stages. The pelagic medusa stage is most familiar. The sessile polyp stage is inconspicuous, but it is this life stage that makes possible and practical the culture and propagation of jellies for aquarium exhibits.

The sessile stage is typically small (1–3 mm tall depending on the species) and colonial. Sexual reproduction does not occur in the benthic life history stages of jellyfish. Sessile polyps can spread asexually to generate other polyps—or they can give rise to the pelagic (medusa) stage by undergoing the asexual process of strobilation. The medusae are free-swimming and solitary. Most medusae become conspicuously large, although some hydrozoan species (Hydromedusae) can be quite small (1–2 mm tall). With few exceptions, jellies are dioecious—they possess either male or female reproductive organs. The medusa stage can only reproduce sexually, and in most species the fertilized egg eventually gives rise to a planula larva that settles and metamorphoses into a new sessile polyp. There are species in which the fertilized egg develops directly into another medusa, either exclusively (e.g., *Pelagia noctiluca*) or rarely (e.g., *Aurelia aurita*) (Arai, 1997).

**Life Cycle of the Scyphozoa:** The following section will describe the life cycle of typical Semaeostomae and rhizostome scyphozoan jellies (see Figure 1). Coronate jellies and stauromedusae are not typically exhibited by public aquariums and for the most part will not be considered. A *Functional Biology of Scyphozoa* by Mary N. Arai (1997) was used extensively as a guide to the descriptions of the scyphozoan life cycle.
Polyp/Scyphistoma: The jelly polyp, or scyphistoma, resembles a small sea anemone (see Figure 2). They are typically 1–2 mm in diameter and 2–3 mm tall, although some rhizostome polyps are smaller than 2 mm. The polyp is attached to a hard surface by a pedal disk. A tubular stalk leads to a ring of tentacles surrounding a central mouth. This oral disc containing the mouth and ring of tentacles is called the calyx. In the known polyps of jellies from the order Coronatae (e.g., Linuche, Nausithoe), there is a chitinous tube surrounding the stalk (Arai, 1997; Ortiz-Corp’s, Cutress & Cutress, 1987); this is absent in polyps of the other orders.

The mouth leads to a blind pouch where food is digested; waste is expelled back out through the mouth. During early development of the polyp, there are just two to four tentacles. This number increases as the polyp grows, and can increase to as many as 24 in some species. Uchida & Sugiura (1978) found as many as 40 tentacles in polyps of Sanderia malayensis. In nature, most polyps prefer to reside in a position where the tentacles can be suspended downward or sideways. Brewer (1976) reports, “the scyphistoma is usually found suspended upside-down in a shaded location.” This allows better extension of the tentacles and more efficiency in the capture of prey items, usually consisting of small zooplankton. It would also aid in preventing the polyp from being covered by silt or other debris (Cargo & Schultz, 1966), and provide an advantage when strobilation occurs and ephyrae are released (Brewer, 1976).

Polyps can spread asexually to form small colonies; one way this is done is by budding. There are various means of budding, but in all cases tissue generated by a parent polyp becomes detached from the parent and gives rise to a new polyp. Usually these polyps are in very close proximity to the parent, but in several species of Cassiopea and in Mastigias papua, a ciliated planula-like bud, called a planuloid,
is produced which can swim away and settle remotely, away from the parent (Van Lieshout & Martin, 1992; Sugiura, 1963). *Cotylorhiza tuberculata* are also capable of producing a planuloid bud (Arai, 1997).

In many, but not all species, polyps can also spread through the formation of podocysts. Arai (1997) defines podocysts as “cysts which form beneath the pedal disk of scyphistomae. They are surrounded by chitin.” These cysts are more or less dormant and can survive periods of unfavorable conditions. They are small, usually less than .5 mm in diameter and normally have a pale brown or yellowish color. Podocysts can remain viable for some time. Black et al. (1976) documented the survival of *Chrysaora quinquecirrha* podocysts for up to 25 months. Chapman (1968) mentions the ability of podocysts to survive for over 3 years. Some species commonly kept in aquarium cultures that can form podocysts are *Cyanea capillata*, *Aurelia aurita*, *A. labiata*, *Mastigias papua*, and most (if not all) species of *Chrysaora*.

Lesh-Laurie & Corriel (1973) reported that severed tentacles of *Aurelia aurita* polyps were able to regenerate entire polyps. This may not be a common means of asexual reproduction, but should be noted as another potential reproductive factor.
Figure 2: Polyp of scyphozoan jelly
Illustration courtesy of C. Schaadt
**Strobilation:** Strobilation is the process by which free-swimming ephyrae are liberated from polyps. According to Spangenberg (1968), it is actually two processes: segmentation and metamorphosis. At the onset of strobilation, the polyp often undergoes a color change. A constriction develops at the terminal end of a polyp to form a segment. As the constriction continues to develop, morphological changes occur. The tentacles of the polyp are resorbed. The outer margin of the polyp begins to convolute into bifurcated lobes. Each paired branch of the lobe is the marginal lappet of the developing ephyra. The constriction continues to develop until the terminal end is completely pinched off. At this time, the marginal lobes with a rhopalium at the junction between paired lappets are completely developed. The rhopalium is a sensory receptor used by ephyrae to determine orientation. This entire structure, which is now nearly segregated from the parent polyp, is called an ephyra. It begins to pulsate and ultimately breaks free from the polyp. The entire process of strobilation, from initiation of segmentation to release of the first ephyra, usually takes 3 to 5 days for some species of *Aurelia* and is longer for *Chrysaora* sp. Strobilation can continue to occur for several days to several weeks.

Strobilation can be monodisc, in which one ephyra at a time is generated, or polydisc, in which multiple ephyrae are in the process of developing at the terminus of the polyp. Most Rhizostome jelly polyps, such as *Mastigias*, and *Cassiopeia*, exhibit monodisc strobilation or polydisc strobulation with a small number of developing ephyrae. Most of the Semaeostomae jellies commonly kept in culture (e.g., *Aurelia*, *Chrysaora*, and *Cyanea*) exhibit polydisc strobilation. There are typically 4–20 segments formed during polydisc strobilation, although there may be more; Gershwin & Collins (2002) observed 56 developing ephyrae on a strobilating polyp of *Chrysaora colorata*. The nutritive condition of the polyp (i.e. well-fed vs. starved) seems to play a role in the number of segments formed (Spangenberg, 1965a).

**Ephyrae and Juvenile Medusae:** Ephyrae are the initial stage of the pelagic phase of a jelly’s life, but they do not look like adult medusa (see Figure 3). They are normally 1–2 mm in diameter just after strobilation, although some can be slightly larger. Most ephyrae have 8 marginal lobes, each with a pair of lappets. Some species (e.g., *Sanderia malayensis*) have 16 marginal lobes, and often individuals with an irregular number of lobes will be found. A single gravity-sensing rhopalium lies between the lappets. Ephyrae can be clear, brown, or even red or maroon (e.g., *Chrysaora colorata*). As the ephyra grow (between 6–10 mm), the lobes begin to disappear, and they become more disc-like. Later, oral appendages begin to grow, the center thickens with mesoglea and the ephyra begins to “bell up,” or take on more of the bell-shaped adult form. Eventually oral arms lengthen and tentacles appear in species that have adult tentacles. At this point, the jelly can be considered a juvenile medusa. It will look like a miniature version of an adult, although often the coloration and external markings are not the same as in the adult. The swimming behavior also will be the much the same as an adult of the species.
Figure 3: Ephyra of scyphozoan jelly
Illustration courtesy of C. Schaadt
**Adult Medusa:** Sexual reproduction occurs in the jellies' medusa stage (see Figure 4). Most adult medusae are dioecious—that is, each medusa is either male or female. It is very difficult to tell males from females, although it may be possible in some species to distinguish between the sexes when gonads are mature and samples of gonad tissue are examined under a dissecting microscope. Size is often the determining factor in maturation in the wild (Arai, 1997), although some species (e.g., *Cyanea*) will reach maturity even if they do not reach the large size usually associated with maturity (Brewer, 1989). In aquariums, jellies that would be considered too small in the wild to be mature will often become gravid.

Gonads in the Scyphozoa reside in the gastric pouches. A typical Semaeostomae jelly will have four gastric pouches, each containing a horseshoe-shaped gonad. Mature sperm and eggs are released into the gastrovascular cavity and leave via the mouth into the seawater, where fertilization and embryo development take place. In *Aurelia*, sperm heads are embedded in thin strings that travel down channels in the oral arms and are released at the tip of each oral arm. Sperm may fertilize the eggs externally in seawater. In some jellies (e.g., *Aurelia aurita, A. labiata, Cotylorhiza tuberculata, Cyanea capillata*), the sperm is ingested. The eggs are fertilized within the gonad or genital sinus. These species brood the developing embryo and planula larvae in specialized brood pouches and release fully developed swimming planulae.
Figure 4: Adult medusa of scyphozoan jelly
Illustration courtesy of C. Schaadt
Planula: A fertilized egg begins to divide and develop into a ciliated planula larva within hours. The planula is described as “elongated and radially symmetrical, but with distinct anterior and posterior ends …there is neither a gastrovascular cavity nor a mouth” (Barnes, 1974). The cilia allow the planula to swim freely. Planulae are usually oval or pear-shaped, 100–400 microns in length. They remain free-swimming, in search of a suitable substrate for attachment, for several hours up to 10 days (Arai, 1997). As previously noted, in Aurelia, the planula larvae develop and are held in specialized brood pouches, on the oral arms in A. aurita, or the manubrium in A. labiata (Gershwin, 2001) for a period of time before being released. In Cyanea, the planulae are carried on the numerous oral folds found in this species.

In Pelagia noctiluca, the planula develops directly into an ephyra, rather than attaching and developing into a scyphistoma. Direct development of planula to ephyra has also been known to happen under rare circumstances in Aurelia aurita (Arai, 1997).

Planula Settlement: As stated above, planulae remain free swimming for several hours to several days. In some species, such as Linuche unguiculata, the planula may remain free-swimming for 3 to 4 weeks (Ortiz-Corp’s, Cutress and Cutress, 1987). During the free-swimming stage, the planulae search for a suitable substrate for attachment, known as settlement. The surface texture of the substrate can be a factor. Planulae from a number of common coastal species (Cyanea capillata, and to a lesser extent, Aurelia aurita and Chrysaora quinquecirrha) exhibit a preference for rough surfaces or surfaces covered by organic and bacterial films vs. smooth surfaces (Brewer 1984; Cargo, 1979). Orientation in the water column is a factor. Polyps are often found in nature with their tentacles facing downward, so planulae seem to be selecting the sides or undersides of an object for settlement. Light levels may also be a factor, as many polyps found in the wild are found in shaded conditions (Dolmer & Svane, 1993).

Once settled, the planula begins to develop into a polyp, or scyphistoma. The stalk begins to take form and the terminal end begins to thicken. The mouth and tentacles begin to develop. Under normal circumstances, and with the role of temperature kept in mind, within one week after settling a recognizable polyp with at least two tentacles will be present. It will usually be weeks or months before the polyps begin to strobilate. However, Calder (1982) reported that polyps of Stomolophus meleagris began to strobilate nine days after settling and developing from planula larvae.

7.2 Artificial Insemination

The practical use of artificial insemination (AI) with animals was developed during the early 1900s to replicate desirable livestock characteristics to more progeny. Over the last decade or so, AZA-accredited zoos and aquariums have begun using AI processes more often with many of the animals residing in their care. AZA Studbooks are designed to help manage animal populations by providing detailed genetic and demographic analyses to promote genetic diversity with breeding pair decisions within and between our institutions. While these decisions are based upon sound biological reasoning, the efforts needed to ensure that transports and introductions are done properly to facilitate breeding between the animals are often quite complex, exhaustive, and expensive, and conception is not guaranteed.

AI has become an increasingly popular technology that is being used to meet the needs identified in the AZA Studbooks without having to re-locate animals. Males are trained to voluntarily produce semen samples and females are being trained for voluntary insemination and pregnancy monitoring procedures such as blood and urine hormone measurements and ultrasound evaluations. Techniques used to preserve and freeze semen have been achieved with a variety, but not all, taxa and should be investigated further.

In-vitro fertilization of jellies is regularly accomplished at institutions. Eggs are removed from ovaries and sperm from testes of dioecious species. Eggs and sperm are put into a Petri dish filled with clean seawater in temperature appropriate for the species of jelly. In about 24 to 72 hours, free-swimming planula should be observable under a microscope. The planulae can be removed to a Petri dish with clean seawater using a pipette. Over the next 24 to 72 hours many of the planula settle on the sides of the Petri dish and grow into scyphistomae (polyps). Some of the scyphistomae settle onto the surface of the seawater, hanging down into the water. These can be removed by placing a small plastic or glass plate into the surface where the scyphistomae are hanging. Most of the scyphistomae will attach themselves to the plate, which is then suspended in a small container of seawater. The developing scyphistomae are fed food appropriate for their size (e.g., rotifers or newly hatched enriched Artemia).
There have been attempts at cryopreservation of gametes of jellies with varying results. More work on this would be beneficial. Jellies are not reliably available from natural sources so robust in-vitro and culturing efforts would ensure jellies to be available to aquariums and zoos for exhibits.

7.3 Pregnancy and Parturition

It is extremely important to understand the physiological and behavioral changes that occur throughout an animal's pregnancy. Jellies do not reproduce in a way that is relevant to parturition approaches.

7.4 Birthing Facilities

As parturition approaches, animal care staff should ensure that the mother is comfortable in the area where the birth will take place, and that this area is “baby-proofed.” Jellies do not reproduce in a way that is relevant to parturition approaches.

7.5 Assisted Rearing

Although mothers may successfully give birth, there are times when they are not able to properly care for their offspring, both in the wild and in ex-situ populations. Fortunately, animal care staff in AZA-accredited institutions are able to assist with the rearing of these offspring if necessary. Jelly females do not rear their young.

7.6 Contraception

Many animals cared for in AZA-accredited institutions breed so successfully that contraception techniques are implemented to ensure that the population remains at a healthy size.

Most jellies produce massive numbers of both scyphistomae (polyps) and ephyrae (larval medusae). Those scyphistomae not used in culturing are typically considered fouling organisms and are cleaned from containers or aquaria. Successfully raising ephyrae to adult medusae is very difficult so efforts are usually focused on a relatively few of the ephyrae released. Surplus ephyrae are considered for sharing with other institutions or for food for medusavore jellies.
Chapter 8. Behavior Management

8.1 Animal Training

Classical and operant conditioning techniques have been used to train animals for over a century. Classical conditioning is a form of associative learning demonstrated by Ivan Pavlov. Classical conditioning involves the presentation of a neutral stimulus that will be conditioned (CS) along with an unconditioned stimulus that evokes an innate, often reflexive, response (US). If the CS and the US are repeatedly paired, eventually the two stimuli become associated and the animal will begin to produce a conditioned behavioral response to the CS.

Operant conditioning uses the consequences of a behavior to modify the occurrence and form of that behavior. Reinforcement and punishment are the core tools of operant conditioning. Positive reinforcement occurs when a behavior is followed by a favorable stimulus to increase the frequency of that behavior. Negative reinforcement occurs when a behavior is followed by the removal of an aversive stimulus to also increase the frequency of that behavior. Positive punishment occurs when a behavior is followed by an aversive stimulus to decrease the frequency of that behavior. Negative punishment occurs when a behavior is followed by the removal of a favorable stimulus also to decrease the frequency of that behavior.

AZA-accredited institutions are expected to utilize reinforcing conditioning techniques to facilitate husbandry procedures and behavioral research investigations.

Animal training is not applicable for jellies.

8.2 Environmental Enrichment

Environmental enrichment, also called behavioral enrichment, refers to the practice of providing a variety of stimuli to the animal’s environment, or changing the environment itself to increase physical activity, stimulates cognition, and promote natural behaviors. Stimuli, including natural and artificial objects, scents, and sounds are presented in a safe way for the animals to interact with. Some suggestions include providing food in a variety of ways (i.e., frozen in ice or in a manner that requires an animal to solve simple puzzles to obtain it), using the presence or scent/sounds of other animals of the same or different species, and incorporating an animal training (husbandry or behavioral research) regime in the daily schedule.

Enrichment programs for animals should take into account the natural history of the species, individual needs of the animals, and facility constraints. The animal’s enrichment plan should include the following elements: goal-setting, planning and approval process, implementation, documentation/record-keeping, evaluation, and subsequent program refinement. The animal enrichment program should ensure that all environmental enrichment devices (EEDs) are “animal” safe and are presented on a variable schedule to prevent habituation AZA-accredited institutions must have a formal written enrichment program that promotes jellyfish-appropriate behavioral opportunities (AZA Accreditation Standard 1.6.1).

Jellyfish enrichment programs should be integrated with veterinary care, nutrition, and animal training programs to maximize the effectiveness and quality of animal care provided. AZA-accredited institutions must have specific staff members assigned to oversee, implement, train, and coordinate interdepartmental enrichment programs (AZA Accreditation Standard 1.6.2).

Jellies are at the mercy of ocean, or aquaria, currents. This planktonic lifestyle means that they are opportunistic, constant feeders. Appropriate behaviors include belling (locomotion), feeding, and reproduction. They lack a brain. Training or environmental enrichment are not appropriate for jellies.

8.3 Staff and Animal Interactions

Animal training and environmental enrichment protocols and techniques should be based on interactions that promote safety for all involved.
Training and environmental enrichment are not applicable for jellies.

8.4 Staff Skills and Training

Jellyfish staff members should be trained in all areas of jellyfish behavior management. Funding should be provided for AZA continuing education courses, related meetings, conference participation, and other professional opportunities. A reference library appropriate to the size and complexity of the institution should be available to all staff and volunteers to provide them with accurate information on the behavioral needs of the animals with which they work.

Training staff in all areas of jellyfish behavior and management is not necessary since behavioral training is not applicable for jellies.
9.1 Program Animal Policy

AZA recognizes many public education and, ultimately, conservation benefits from program animal presentations. AZA’s Conservation Education Committee’s Program Animal Position Statement (Appendix D) summarizes the value of program animal presentations. For the purpose of this policy, a program animal is described as an animal presented either within or outside of its normal exhibit or holding area that is intended to have regular proximity to or physical contact with trainers, handlers, the public, or will be part of an ongoing conservation education/outreach program.

Program animal presentations bring a host of responsibilities, including the welfare of the animals involved, the safety of the animal handler and public, and accountability for the take-home, educational messages received by the audience. Therefore, AZA requires all accredited institutions that give program animal presentations to develop an institutional program animal policy that clearly identifies and justifies those species and individuals approved as program animals and details their long-term management plan and educational program objectives.

AZA’s accreditation standards require that the conditions and treatment of animals in education programs must meet standards set for the remainder of the animal collection, including species-appropriate shelter, exercise, sound and environmental enrichment, access to veterinary care, nutrition, and other related standards (AZA Accreditation Standard 1.5.4). In addition, providing program animals with options to choose among a variety of conditions within their environment is essential to ensuring effective care, welfare, and management. Some of these requirements can be met outside of the primary exhibit enclosure while the animal is involved in a program or is being transported. For example, housing may be reduced in size compared to a primary enclosure as long as the animal’s physical and psychological needs are being met during the program; upon return to the facility the animal should be returned to its species-appropriate housing as described above.

Jellies can be program animals by putting them into a closed, clear container that can be used to show the animals to visitors at on-site or off-site programs. When jellies are used as program animals, they are kept in clear containers with lids (preferably plastic rather than glass to reduce the chance of breakage). In all cases, bubbles should not be allowed in the water with the jellies. It is not recommended to allow visitors to touch jellies (for the safety of the visitor and the jelly). People may not know that they are allergic to the sting of jellies. Temperature should be kept within 5 °C (10 °F) of optimum temperature and be rotated frequently. A good schedule of rotating program jellies would be to change the jellies every 30 minutes in order to minimize any deleterious effects of handling and thermal stress.

9.2 Institutional Program Animal Plans

AZA’s policy on the presentation of animals is as follows: AZA is dedicated to excellence in animal care and welfare, conservation, education, research, and the presentation of animals in ways that inspire respect for wildlife and nature. AZA’s position is that animals should always be presented in adherence to the following core principles:

- Animal and human health, safety, and welfare are never compromised.
- Education and a meaningful conservation message are integral components of the presentation.
- The individual animals involved are consistently maintained in a manner that meets their social, physical, behavioral, and nutritional needs.

AZA-accredited institutions that have designated program animals are required to develop their own Institutional Program Animal Policy that articulates and evaluates the program benefits (see Appendix E for recommendations). Program animals should be consistently maintained in a manner that meets their social, physical, behavioral, and nutritional needs. Education and conservation messaging must be an integral component of any program animal demonstration (AZA Accreditation Standard 1.5.3).
Jellies used in program situations should not be touched by visitors. Jellies should be kept in containers that preclude bubbles since they can be ingested or captured under the bell and cause the bell to float and expose the delicate epidermis of the bell to air resulting in injury. Jellies can be in a program for up to 8 hours without food. Jellies do not have social needs.

Some of the conservation stories that can be shared are the invasive nature of some jellies and the phenomenon of jellyfish blooms, which may be enabled by global climate change.

Animal care and education staff should be trained in program animal-specific handling protocols, conservation, and education messaging techniques, and public interaction procedures. These staff members should be competent in recognizing stress or discomfort behaviors exhibited by the program animals and be able to address any safety issues that arise.

Program staff should be trained in the needs of the jellies kept in the transparent plastic containers. They should know the jellies cannot be touched and the temperature regimes to be maintained. Jellies should always be kept in their containers reducing the chance of them contracting diseases. The size of the transparent plastic container should be sufficient to allow the jelly to bell freely, but not so large that the weight of the container with water is too heavy for the program presenters to move about.

Program animals that are taken off zoo or aquarium grounds for any purpose have the potential to be exposed to infectious agents that could spread to the rest of the institution’s healthy population. AZA-accredited institutions must have adequate protocols in place to avoid this (AZA Accreditation Standard 1.5.5).

Careful consideration must be given to the design and size of all program animal enclosures, including exhibit, off-exhibit holding, hospital, quarantine, and isolation areas, such that the physical, social, behavioral, and psychological needs of the species are met and species-appropriate behaviors are facilitated (AZA Accreditation Standard 10.3.3; AZA Accreditation Standard 1.5.2).

Similar consideration needs to be given to the means in which an animal will be transported both within the Institution’s grounds, and to/from an off-grounds program. Animal transportation must be conducted in a manner that is lawful, safe, well planned, and coordinated, and minimizes risk to the animal(s), employees, and general public (AZA Accreditation Standard 1.5.11).

Time limitations should be imposed so as not to have the water in the container raise or lower more than about 3 °C (5 °F) or more than about 2 hours. Jellies should be re-acclimated to their regular tank after participation in a program.

9.3 Program Evaluation

AZA-accredited institutions which have Institutional Program Animal Plan are required to evaluate the efficacy of the plan routinely (see Appendix E for recommendations). Education and conservation messaging content retention, animal health and well-being, guest responses, policy effectiveness, and accountability and ramifications of policy violations should be assessed and revised as needed.

Program Animal Plans should be reviewed at least every five years or as needed.
10.1 Known Methodologies

AZA believes that contemporary jellyfish management, husbandry, veterinary care and conservation practices should be based in science, and that a commitment to scientific research, both basic and applied, is a trademark of the modern zoological park and aquarium. AZA-accredited institutions have the invaluable opportunity, and are expected, to conduct or facilitate research both in *in situ* and *ex situ* settings to advance scientific knowledge of the animals in our care and enhance the conservation of wild populations. This knowledge might be achieved by participating in AZA Taxon Advisory Group (TAG) or Species Survival Plan® (SSP) Program sponsored research, conducting original research projects, affiliating with local universities, and/or employing staff with scientific credentials (AZA Accreditation Standard 5.3).

Research and conservation initiatives for jellies are varied and certainly change from different regions of the US and the world. Some examples in Southern California for research include: neuroscience (UCLA), biomechanics, and applications to engineering (California Institute of Technology), and taxonomy and biogeographics (UC Berkeley). The most important conservation research issues are jellyfish blooms and jellies as invasive species. Institutions that have opportunities to conduct or otherwise support research on jellies should do so.

Research investigations, whether observational, behavioral, physiological, or genetically based, should have a clear scientific purpose with the reasonable expectation that they will increase our understanding of the species being investigated and may provide results which benefit the health or welfare of animals in wild populations. Many AZA-accredited institutions incorporate superior positive reinforcement training programs into their routine schedules to facilitate sensory, cognitive, and physiological research investigations and these types of programs are strongly encouraged by the AZA.

AZA-accredited institutions are required to have a clearly written research policy that identifies the types of research being conducted, methods used, staff involved, evaluations of the projects, the animals included, and guidelines for the reporting or publication of any findings (AZA Accreditation Standard 5.2). Institutions must designate a qualified individual to oversee and direct its research program (AZA Accreditation Standard 5.1). If institutions are not able to conduct in-house research investigations, they are strongly encouraged to provide financial, personnel, logistical, and other support for priority research and conservation initiatives identified by Taxon Advisory Groups (TAGs) or Species Survival Plan® (SSP) Programs.

10.2 Future Research Needs

This Animal Care Manual is a dynamic document that will need to be updated as new information is acquired. Knowledge gaps have been identified throughout the Manual and are included in this section to promote future research investigations. Knowledge gained from areas will maximize AZA-accredited institutions’ capacity for excellence in animal care and welfare as well as enhance conservation initiatives for the species.

The husbandry and public display of jellies is still a relatively young discipline having begun on a large scale in Japan in the 1960s and not in a serious way until the 1990s in the United States. Nutrition, culturing, husbandry, and display issues are regularly being improved upon.

Chapter 6. Veterinary Care
6.7 Management of Diseases, Disorders, Injuries and/or Isolation
There is still much to learn about diseases of jellyfish. There will likely be more discoveries of parasitic associations between jellyfish and other invertebrates. The subject of jellyfish eversions has not even been mentioned. This is a common occurrence in jelly culture and husbandry, yet very little is known about the cause. Freeman et al. (2009) studied this syndrome and concluded that it is a complex phenomenon associated with degenerative changes of the bell matrix.

Often the aquarist will feel helpless when observing the deteriorating condition of specimens in his/her care. At the present time, little is known to be of clear value in the treatment of most jelly diseases or problems. Providing the best nutrition, water quality, cleanliness, tank flow conditions, etc. is the best guarantee for preventing diseases or undesirable jellyfish conditions such as inversions or bell rot. Jellies in the wild do not normally live very long; most survive 1 year or less. Perhaps there is little that can be done once a jelly begins to senesce. Without more laboratory research, we won’t know the answers.

**Jellyfish Conservation**

AZA institutions should continue to support research and conservation efforts as their resources allow. One major conservation concern is the phenomenon of jellyfish blooms in the wild. Many researchers all around the world are working on this issue (Mills, 2001; Purcell et al., 2001; Purcell et al., 2007). AZA institutions should share information about jellyfish blooms in their jellyfish exhibits and programs and take advantage of collaborating with scientists working on trying to understand this issue. Institutions need to continue to enhance sustainability of collections by improving culturing efforts and increasing support for sharing of surplus jellies.
Chapter 11. History

11.1 History of Jellyfish Displays in Japan, United States, and Europe

A story is told that back in the early days (1950s and 60s) an aquarium wanted to display adult medusa of the purple striped jellyfish. No tank was found that would enable the 40cm wide purple stripe jellies to swim let alone survive for more than a couple days. So aquarists threaded a needle with a monofilament line that terminated in a large button up through the manubrium which suspended the jellies in a tank. As the jellies belled and eventually relaxed, they would stay suspended for people to see. Attempts at feeding the jellies were unsuccessful and the technique resulted in the disintegration of the bell over just a few days.

Many years later, Monterey Bay Aquarium (MBA) had a small exhibit of live moon jellies that seemed to catch the imagination of visitors Japan (Powell, 2001). MBA already had a good relationship with aquariums in Japan and Yoshitaka Abe from the Ueno Aquarium and Kazuko Shimura from the Enoshima Aquarium worked with MBA staffers Dave Powell and Freya Summer to share successful jellyfish husbandry and display techniques (Powell, 2001). Mr. Abe had extensive experience raising moon jellyfish (Abe, 1969) and Ms. Shimura had experience raising many other jellies. Freya became singularly successful and raising jellyfish and gave a paper at the Western Regional Conference of the American Association of Zoological Parks and Aquariums (AAZPA now Association of Zoos and Aquariums or AZA) in Tacoma, WA (Sommer, 1986) which is still one of the best overviews of culturing moon jellies. Freya expanded her interest to include other species and was soon culturing all sorts of jellies at MBA. One of her projects turned out to be a significant contribution to science as she discovered that the purple striped jelly had a scyphistoma stage, something the literature of the day said it did not. This eventually resulted in a publication describing the reproduction in the purple striped jelly and assigning it to the genus *Chrysaora* (from *Pelagia*) (Gershwin & Collins, 2002).

As Freya’s work continued to expand the husbandry of heretofore undisplayed jellies, there was a push to make an exhibit that would allow visitors to see these little known creatures. Despite some reservations that jellies were not interesting enough to visitors, the MBA opened the “Planet of the Jellies” in 1992. This exhibit became the most successful temporary exhibit MBA had ever presented and was critically acclaimed by all. Other MBA staff that worked closely with this project included Bruce Upton, Mark Ferguson and Dave Wrobel (creator of www.jellieszone.com). In 2003, MBA opened another very successful special exhibition featuring jellyfish called “Jellies: Living Art.”

One of the advances in the successful display of jellies was the development of tank designs that simulated their planktonic environment. In Germany in the 1960’s, Wolf Greve was studying ctenophores and chaetognaths. He developed a tank he called a planktonkreisel referring to the tank’s circular design that helped keep plankton suspended so he could study them (Greve, 1968). In 1975, Greve further modified his planktonkreisel to improve viewing for behavioral studies and called his new design the meteor planktonkuvette (Greve, 1975). Hamner (1990) working with the staff of MBA, further refined Greve’s design to come up with a shipboard version he also called a planktonkreisel for studying plankton freshly caught from a ship at sea. Hamner’s design also made for a great way to see planktonic organisms swimming in gentle currents. Many planktonkreisels were incorporated into the “Planet of the Jellies” exhibit. Planktonkreisels and the similar pseudokreisels became the basic design used many aquariums and zoos to display jellyfish and other gelatinous zooplankton (e.g., ctenophores).

In Europe, Paul Van Den Sande at the Antwerp Zoo Aquarium in Belgium and Jurgen Lange at the Berlin Zoo in Germany led the way in culturing jellies (Lange, 1995). In the early 1990s a few aquariums opened special exhibitions of jellies. The reaction from the public led to many of these exhibitions traveling to other aquariums in the country.

As more and more aquariums and zoos wanted to either acquire jelly exhibits or expand their species list, attempts were made to gather people together to share information. In 1998, Mike Schaadt organized a jelly husbandry workshop at the Western Regional Meeting of the AZA in Monterey, CA. The Jelly Directory was the result of that meeting where staff working on jellyfish at aquariums and zoos would supply their contact information, the species of jellies they culture and display and the types of tanks they use. Anyone on the list can get an updated list by contacting Mike (mike.schaadt@lacity.org). In 2003, the Jellyfish listserv was started under the website of AZA. There was also a session given at the 2005
Annual Conference of AZA in Chicago, IL, which was the beginning of this version of the Jellyfish Husbandry Manual.

**The Role of the Aquarist in Collecting Data**

As aquarists we have a unique opportunity to document species in known geographic locations. Coastal aquarists and aquarists collecting their own specimens can observe species in the wild and document newly introduced species. It is important to describe seasonal occurrence, new geographic locations, and new records for collected specimens.

It is the responsibility of the aquarist to ensure that no form of the jelly life cycle (planula, polyp, or medusa) is released into non-native waters. Many species can rapidly proliferate into a new environment. The cases of the plant Caulerpa in the Mediterranean, the scyphozoan Phyllorhiza in Southern California with its spread into the Gulf of Mexico and the ctenophore Mnemiopsis in the Black Sea serve as good lessons for both an accidental aquarium release and invasive species spread into new geographic locations.

It is clear that gelatinous plankton speciation is much more complex than our current 200 species of Scyphozoa (Mianzan & Cornelius, 1999) and perhaps over 3,000 species of Hydrozoa (Schuchert, 1998) reveal. Molecular genetics can provide taxonomic verification, differentiate cryptogenic species, identify source populations, and assess the extent and impact of invasive species (Dawson et al., 2005). Genetics coupled with detailed morphological assessment is the best method to access the speciation of gelatinous plankton.
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Nancy Sowinski, Sunset Marine Labs, CA
Chad Widmer, Monterey Bay Aquarium, CA
References


Additional Resources


**Personal Communication**

Sharyl Crossley, Tennessee Aquarium, TN  
Chad Widmer, Monterey Bay Aquarium, CA  
David Wrobel, New England Aquarium, CA  
Bruce Upton, Monterey Bay Aquarium, CA

**Websites**

[www.jellieszone.com](http://www.jellieszone.com) – Excellent overview of sea jellies and other gelatinous zooplankton.  
[http://dockwatch.disl.org/glossary.htm](http://dockwatch.disl.org/glossary.htm) – Jellies in the Gulf of Mexico.  
Appendix A: Accreditation Standards by Chapter

The following specific standards of care relevant to jellyfish are taken from the AZA Accreditation Standards and Related Policies (AZA, 2011) and are referenced fully within the chapters of this animal care manual:

General Information
(1.1.1) The institution must comply with all relevant local, state, and federal wildlife laws and regulations. It is understood that, in some cases, AZA accreditation standards are more stringent than existing laws and regulations. In these cases the AZA standard must be met.

Chapter 1
(1.5.7) The animal collection must be protected from weather detrimental to their health.
(10.2.1) Critical life-support systems for the animal collection, including but not limited to plumbing, heating, cooling, aeration, and filtration, must be equipped with a warning mechanism, and emergency backup systems must be available. All mechanical equipment should be under a preventative maintenance program as evidenced through a record-keeping system. Special equipment should be maintained under a maintenance agreement, or a training record should show that staff members are trained for specified maintenance of special equipment.
(1.5.9) The institution must have a regular program of monitoring water quality for collections of fish, pinnipeds, cetaceans, and other aquatic animals. A written record must be maintained to document long-term water quality results and chemical additions.

Chapter 2
(1.5.2) Animals should be displayed, whenever possible, in exhibits replicating their wild habitat and in numbers sufficient to meet their social and behavioral needs. Display of single specimens should be avoided unless biologically correct for the species involved.
(10.3.3) All animal enclosures (exhibits, holding areas, hospital, and quarantine/isolation) must be of a size and complexity sufficient to provide for the animal's physical, social, and psychological well-being; and exhibit enclosures must include provisions for the behavioral enrichment of the animals.
(11.3.3) Special attention must be given to free-ranging animals so that no undue threat is posed to the animal collection, free-ranging animals, or the visiting public. Animals maintained where they will be in contact with the visiting public must be carefully selected, monitored, and treated humanely at all times.
(11.3.1) All animal exhibits and holding areas must be secured to prevent unintentional animal egress.
(11.3.6) Guardrails/barriers must be constructed in all areas where the visiting public could have contact with other than handleable animals.
(11.2.3) All emergency procedures must be written and provided to staff and, where appropriate, to volunteers. Appropriate emergency procedures must be readily available for reference in the event of an actual emergency. These procedures should deal with four basic types of emergencies: fire, weather/environment; injury to staff or a visitor; animal escape.
(11.6.2) Security personnel, whether staff of the institution, or a provided and/or contracted service, must be trained to handle all emergencies in full accordance with the policies and procedures of the institution. In some cases, it is recognized that Security personnel may be in charge of the respective emergency (i.e., shooting teams).
(11.2.4) The institution must have a communication system that can be quickly accessed in case of an emergency.
(11.2.5) A written protocol should be developed involving local police or other emergency agencies and include response times to emergencies.
(11.5.3) Institutions maintaining potentially dangerous animals (sharks, whales, tigers, bears, etc.) must have appropriate safety procedures in place to prevent attacks and injuries by these animals. Appropriate response procedures must also be in place to deal with an attack resulting in an injury. These procedures must be practiced routinely per the emergency drill requirements contained in these standards. Whenever injuries result from these incidents, a written account outlining the cause of the incident, how the injury was handled, and a description of any resulting changes to either the safety procedures or the physical facility must be prepared and maintained for five years from the date of the incident.
Chapter 3

(1.5.11) Animal transportation must be conducted in a manner that is safe, well-planned and coordinated, and minimizes risk to the animal(s), employees, and general public. All applicable local, state, and federal laws must be adhered to.

Chapter 5

(2.6.2) A formal nutrition program is recommended to meet the behavioral and nutritional needs of all species and specimens within the collection.

(2.6.3) Animal diets must be of a quality and quantity suitable for each animal’s nutritional and psychological needs. Diet formulations and records of analysis of appropriate feed items should be maintained and may be examined by the Visiting Committee. Animal food, especially seafood products, should be purchased from reliable sources that are sustainable and/or well managed.

(2.6.1) Animal food preparations must meet all local, state/provincial, and federal regulations.

Chapter 6

(2.1.1) A full-time staff veterinarian is recommended. However, the Commission realizes that in some cases such is not practical. In those cases, a consulting/part-time veterinarian must be under contract to make at least twice monthly inspections of the animal collection and respond as soon as possible to any emergencies. The Commission also recognizes that certain collections, because of their size and/or nature, may require different considerations in veterinary care.

(2.1.2) So that indications of disease, injury, or stress may be dealt with promptly, veterinary coverage must be available to the animal collection 24 hours a day, 7 days a week.

(2.2.1) Written, formal procedures must be available to the animal care staff for the use of animal drugs for veterinary purposes and appropriate security of the drugs must be provided.

(2.6.4) The institution should assign at least one person to oversee appropriate browse material for the collection.

(2.1.3) Written, formal procedures must be available to the animal care staff for the use of animal drugs for veterinary purposes and appropriate security of the drugs must be provided.

Chapter 7

(1.4.6) A staff member must be designated as being responsible for the institution's animal record-keeping system. That person must be charged with establishing and maintaining the institution's animal records, as well as with keeping all animal care staff members apprised of relevant laws and regulations regarding the institution's animal collection.

(1.4.7) Animal records must be kept current, and data must be logged daily.

(2.7.1) The institution must have holding facilities or procedures for the quarantine of newly arrived animals and isolation facilities or procedures for the treatment of sick/injured animals.

(2.7.3) Quarantine, hospital, and isolation areas should be in compliance with standards or guidelines adopted by the AZA.

(2.7.2) Written, formal procedures for quarantine must be available and familiar to all staff working with quarantined animals.

(11.1.2) Training and procedures must be in place regarding zoonotic diseases.

(11.1.3) A tuberculin testing and surveillance program must be established for appropriate staff in order to ensure the health of both the employees and the animal collection.

(2.5.1) Deceased animals should be necropsied to determine the cause of death. Disposal after necropsy must be done in accordance with local/federal laws.

(2.4.1) The veterinary care program must emphasize disease prevention.

(1.5.5) For animals used in offsite programs and for educational purposes, the institution must have adequate protocols in place to protect the rest of the collection from exposure to infectious agents.
(2.3.1) Capture equipment must be in good working order and available to authorized, trained personnel at all times.

(2.4.2) Keepers should be trained to recognize abnormal behavior and clinical symptoms of illness and have knowledge of the diets, husbandry (including enrichment items and strategies), and restraint procedures required for the animals under their care. However, keepers should not evaluate illnesses nor prescribe treatment.

(2.3.2) Hospital facilities should have x-ray equipment or have access to x-ray services.

(1.5.8) The institution must develop a clear process for identifying and addressing animal welfare concerns within the institution.

**Chapter 8**

(1.6.1) The institution must have a formal written enrichment program that promotes species-appropriate behavioral opportunities.

(1.6.2) The institution must have a specific staff member(s) or committee assigned for enrichment program oversight, implementation, training, and interdepartmental coordination of enrichment efforts.

**Chapter 9**

(1.5.4) A written policy on the use of live animals in programs should be on file. Animals in education programs must be maintained and cared for by trained staff, and housing conditions must meet standards set for the remainder of the animal collection, including species-appropriate shelter, exercise, social and environmental enrichment, access to veterinary care, nutrition, etc. Since some of these requirements can be met outside of the primary enclosure, for example, enclosures may be reduced in size provided that the animal’s physical and psychological needs are being met.

(1.5.3) If animal demonstrations are a part of the institution’s programs, an education and conservation message must be an integral component.

(1.5.5) For animals used in offsite programs and for educational purposes, the institution must have adequate protocols in place to protect the rest of the collection from exposure to infectious agents.

(10.3.3) All animal enclosures (exhibits, holding areas, hospital, and quarantine/isolation) must be of a size and complexity sufficient to provide for the animal’s physical, social, and psychological well-being; and exhibit enclosures must include provisions for the behavioral enrichment of the animals.

(1.5.2) Animals should be displayed in exhibits replicating their wild habitat and in numbers sufficient to meet their social and behavioral needs. Display of single animals should be avoided unless biologically correct for the species involved.

(1.5.11) Animal transportation must be conducted in a manner that is safe, well planned, and coordinated, and minimizes risk to the animal(s), employees, and general public. All applicable local, state, and federal laws must be adhered to. Planning and coordination for animal transport requires good communication among all involved parties, plans for a variety of emergencies and contingencies that may arise, and timely execution of the transport. At no time should the animal(s) or people be subjected to unnecessary risk or danger.

**Chapter 10**

(5.3) Institutions should maximize the generation of scientific knowledge gained from the animal collection. This might be achieved by participating in AZA TAG/SSP sponsored research when applicable, conducting original research projects, affiliating with local universities, and/or employing staff with scientific credentials.

(5.2) Institutions must have a written policy that outlines the type of research that it conducts, methods, staff involvement, evaluations, animals to be involved, and guidelines for publication of findings.

(5.1) Research activities must be under the direction of a person qualified to make informed decisions regarding research.
Appendix B: Acquisition/Disposition Policy

I. Introduction: The Association of Zoos and Aquariums (AZA) was established, among other reasons, to foster continued improvement in the zoological park and aquarium profession. One of its most important roles is to provide a forum for debate and consensus building among its members, the intent of which is to attain high ethical standards, especially those related to animal care and professional conduct. The stringent requirements for AZA accreditation and high standards of professional conduct are unmatched by similar organizations and also far surpass the United States Department of Agriculture's Animal and Plant Health Inspection Service's requirements for licensed animal exhibitors. AZA member facilities must abide by a Code of Professional Ethics — a set of standards that guide all aspects of animal management and welfare. As a matter of priority, AZA institutions should acquire animals from other AZA institutions and dispose of animals to other AZA institutions.

AZA-accredited zoological parks and aquariums cannot fulfill their important missions of conservation, and science without living animals. Responsible management of living animal populations necessitates that some individuals be acquired and that others be removed from the collection at certain times. Acquisition of animals can occur through propagation, trade, donation, loan, purchase, capture, or rescue. Animals used as animal feed are not accessioned into the collection.

Disposition occurs when an animal leaves the collection for any reason. Reasons for disposition vary widely, but include cooperative population management (genetic or demographic management), reintroduction, behavioral incompatibility, sexual maturation, animal health concerns, loan or transfer, or death.

The AZA Acquisition/Disposition Policy (A/D) was created to help (1) guide and support member institutions in their animal acquisition and disposition decisions, and (2) ensure that all additions and removals are compatible with the Association's stated commitment to “save and protect the wonders of the living natural world.” More specifically, the AZA A/D Policy is intended to:

- Ensure that the welfare of individual animals and conservation of populations, species and ecosystems are carefully considered during acquisition and disposition activities;
- Maintain a proper standard of conduct for AZA members during acquisition and disposition activities;
- Ensure that animals from AZA member institutions are not transferred to individuals or organizations that lack the appropriate expertise or facilities to care for them;
- Support the goal of AZA’s cooperatively managed populations and associated programs, including Species Survival Plans (SSPs), Population Management Plans (PMPs), and Taxon Advisory Groups (TAGs).

The AZA Acquisition/Disposition Policy will serve as the default policy for AZA member institutions. Institutions may develop their own A/D Policy in order to address specific local concerns. Any institutional policy must incorporate and not conflict with the AZA acquisition and disposition standards.

Violations of the AZA Acquisition/Disposition Policy will be dealt with in accordance with the AZA Code of Professional Ethics. Violations can result in an institution's or individual's expulsion from membership in the AZA.

II. Group or Colony-based Identification: For some colonial, group-living, or prolific species, such as certain insects, aquatic invertebrates, schooling fish, rodents, and bats, it is often impossible or highly impractical to identify individual specimens. These species are therefore maintained, accessioned, and disposed of as a group or colony. Therefore, when this A/D Policy refers to animals or specimens, it is in reference to both individuals and groups/colonies.

III. Germplasm: Acquisition and disposition of germplasm should follow the same guidelines outlined in this document if its intended use is to create live animal(s). Ownership of germplasm and any resulting animals should be clearly defined. Institutions acquiring or dispositioning germplasm or any animal parts or samples should consider not only its current use, but also future possible uses as new technologies become available.
IV(a). General Acquisitions: Animals are to be acquisitioned into an AZA member institution’s collection if the following conditions are met:

1. Acquisitions must meet the requirements of all applicable local, state, federal and international regulations and laws.
2. The Director or Chief Executive Officer of the institution is charged with the final authority and responsibility for the monitoring and implementation of all acquisitions.
3. Acquisitions must be consistent with the mission of the institution, as reflected in its Institutional Collection Plan, by addressing its exhibition/education, conservation, and/or scientific goals.
4. Animals that are acquired for the collection, permanently or temporarily, must be listed on institutional records. All records should follow the Standards for Data Entry and Maintenance of North American Zoo and Aquarium Animal Records Databases®.
5. Animals may be acquired temporarily for reasons such as, holding for governmental agencies, rescue and/or rehabilitation, or special exhibits. Animals should only be accepted if they will not jeopardize the health, care or maintenance of the animals in the permanent collection or the animal being acquired.
6. The institution must have the necessary resources to support and provide for the professional care and management of a species, so that the physical and social needs of both specimen and species are met.
7. Attempts by members to circumvent AZA conservation programs in the acquisition of SSP animals are detrimental to the Association and its conservation programs. Such action may be detrimental to the species involved and is a violation of the Association's Code of Professional Ethics. All AZA members must work through the SSP program in efforts to acquire SSP species and adhere to the AZA Full Participation policy.
8. Animals are only to be acquired from sources that are known to operate legally and conduct their business in a manner that reflects and/or supports the spirit and intent of the AZA Code of Professional Ethics as well as this policy. Any convictions of state, federal, or international wildlife laws should be reviewed, as well as any previous dealings with other AZA-accredited institutions.
9. When acquiring specimens managed by a PMP, institutions should consult with the PMP manager.
10. Institutions should consult AZA Wildlife Conservation and Management Committee (WCMC)-approved Regional Collection Plans (RCPs) when making acquisition decisions.

IV(b). Acquisitions from the Wild: The maintenance of wild animal populations for education and wildlife conservation purposes is a unique responsibility of AZA member zoos and aquariums. To accomplish these goals, it may be necessary to acquire wild-caught specimens. Before acquiring animals from the wild, institutions are encouraged to examine sources including other AZA institutions or regional zoological associations.

When acquiring animals from the wild, careful consideration must be taken to evaluate the long-term impacts on the wild population. Any capture of free-ranging animals should be done in accordance with all local, state, federal, and international wildlife laws and regulations and not be detrimental to the long-term viability of the species or the wild or captive population(s). In crisis situations, when the survival of a population is at risk, rescue decisions are to be made on a case-by-case basis.

V(a). Disposition Requirements – living animals: Successful conservation and animal management efforts rely on the cooperation of many entities, both within and outside of AZA. While preference is given to placing animals within AZA member institutions, it is important to foster a cooperative culture among those who share the primary mission of AZA-accredited facilities. The AZA draws a strong distinction between the mission, stated or otherwise, of non-AZA member organizations and the mission of professionally managed zoological parks and aquariums accredited by the AZA.

An accredited AZA member balances public display, recreation, and entertainment with demonstrated efforts in education, conservation, and science. While some non-AZA member organizations may meet minimum daily standards of animal care for wildlife, the AZA recognizes that this, by itself, is insufficient to warrant either AZA membership or participation in AZA's cooperative animal management programs. When an animal is sent to a non-member of AZA, it is imperative that the member be confident that the animal will be cared for properly.
Animals may only be disposed of from an AZA member institution’s collection if the following conditions are met:

1. Dispositions must meet the requirements of all applicable local, state, federal and international regulations and laws.
2. The Director or Chief Executive Officer of the institution is charged with the final authority and responsibility for the monitoring and implementation of all dispositions.
3. Any disposition must abide by the Mandatory Standards and General Advisories of the AZA Code of Professional Ethics. Specifically, "a member shall make every effort to assure that all animals in his/her collection and under his/her care are disposed of in a manner which meets the current disposition standards of the Association and do not find their way into the hands of those not qualified to care for them properly."
4. Non-domesticated animals shall not be disposed of at animal auctions. Additionally, animals shall not be disposed of to any organization or individual that may use or sell the animal at an animal auction. In transactions with AZA non-members, the recipient must ensure in writing that neither the animal nor its offspring will be disposed of at a wild animal auction or to an individual or organization that allows the hunting of the animal.
5. Animals shall not be disposed of to organizations or individuals that allow the hunting of these animals or their offspring. This does not apply to individuals or organizations which allow the hunting of only free-ranging game species (indigenous to North America) and established long-introduced species such as, but not limited to, white-tailed deer, quail, rabbit, waterfowl, boar, ring-necked pheasant, chukar, partridge, and trout. AZA distinguishes hunting/fishing for sport from culling for sustainable population management and wildlife conservation purposes.
6. Attempts by members to circumvent AZA conservation programs in the disposition of SSP animals are detrimental to the Association and its conservation programs. Such action may be detrimental to the species involved and is a violation of the Association’s Code of Professional Ethics. All AZA members must work through the SSP program in efforts to deacquisition SSP species and adhere to the AZA Full Participation policy.
7. Domesticated animals are to be disposed of in a manner consistent with acceptable farm practices and subject to all relevant laws and regulations.
8. Live specimens may be released within native ranges, subject to all relevant laws and regulations. Releases may be a part of a recovery program and any release must be compatible with the AZA Guidelines for Reintroduction of Animals Born or Held in Captivity, dated June 3, 1992.
9. Detailed disposition records of all living or dead specimens must be maintained. Where applicable, proper animal identification techniques should be utilized.
10. It is the obligation of every loaning institution to monitor, at least annually, the conditions of any loaned specimens and the ability of the recipient to provide proper care. If the conditions and care of animals are in violation of the loan agreement, it is the obligation of the loaning institution to recall the animal. Furthermore, an institution's loaning policy must not be in conflict with this A/D Policy.
11. If live specimens are euthanized, it must be done in accordance with the established policy of the institution and the Report of the American Veterinary Medical Association Panel on Euthanasia (Journal of the American Veterinary Medical Association 218 (5): 669-696, 2001).
12. In dispositions to non-AZA members, the non-AZA member's mission (stated or implied) must not be in conflict with the mission of AZA, or with this A/D Policy.
13. In dispositions to non-AZA member facilities that are open to the public, the non-AZA member must balance public display, recreation, and entertainment with demonstrated efforts in conservation, education, and science.
14. In dispositions to non-AZA members, the AZA members must be convinced that the recipient has the expertise, records management practices, financial stability, facilities, and resources required to properly care for and maintain the animals and their offspring. It is recommended that this documentation be kept in the permanent record of the animals at the AZA member institution.
15. If living animals are sent to a non-AZA member research institution, the institution must be registered under the Animal Welfare Act by the U.S. Department of Agriculture Animal and Plant
Health Inspection Service. For international transactions, the receiving facility should be registered by that country's equivalent body with enforcement over animal welfare.

16. No animal disposition should occur if it would create a health or safety risk (to the animal or humans) or have a negative impact on the conservation of the species.

17. Inherently dangerous wild animals or invasive species should not be dispositioned to the pet trade or those unqualified to care for them.

18. Under no circumstances should any primates be dispositioned to a private individual or to the pet trade.

19. Fish and aquatic invertebrate species that meet ANY of the following are inappropriate to be disposed of to private individuals or the pet trade:
   a. species that grow too large to be housed in a 72-inch long, 180 gallon aquarium (the largest tank commonly sold in retail stores)
   b. species that require extraordinary life support equipment to maintain an appropriate captive environment (e.g., cold water fish and invertebrates)
   c. species deemed invasive (e.g., snakeheads)
   d. species capable of inflicting a serious bite or venomous sting (e.g., piranha, lion fish, blue-ringed octopus)
   e. species of wildlife conservation concern

21. When dispositioning specimens managed by a PMP, institutions should consult with the PMP manager.

22. Institutions should consult WCMC-approved RCPs when making disposition decisions.

V(b). Disposition Requirements – dead specimens: Dead specimens (including animal parts and samples) are only to be disposed of from an AZA member institution's collection if the following conditions are met:

1. Dispositions of dead specimens must meet the requirements of all applicable local, state, federal and international regulations and laws.
2. Maximum utilization is to be made of the remains, which could include use in educational programs or exhibits.
3. Consideration is given to scientific projects that provide data for species management and/or conservation.
4. Records (including ownership information) are to be kept on all dispositions, including animal body parts, when possible.
5. SSP and TAG necropsy protocols are to be accommodated insofar as possible.

VI. Transaction Forms: AZA member institutions will develop transaction forms to record animal acquisitions and dispositions. These forms will require the potential recipient or provider to adhere to the AZA Code of Professional Ethics, the AZA Acquisition/Disposition Policy, and all relevant AZA and member policies, procedures and guidelines. In addition, transaction forms must insist on compliance with the applicable laws and regulations of local, state, federal and international authorities.
Appendix C: Recommended Quarantine Procedures

Quarantine facility: A separate quarantine facility, with the ability to accommodate mammals, birds, reptiles, amphibians, and fish should exist. If a specific quarantine facility is not present, then newly acquired animals should be isolated from the established collection in such a manner as to prohibit physical contact, to prevent disease transmission, and to avoid aerosol and drainage contamination.

Such separation should be obligatory for primates, small mammals, birds, and reptiles, and attempted wherever possible with larger mammals such as large ungulates and carnivores, marine mammals, and cetaceans. If the receiving institution lacks appropriate facilities for isolation of large primates, pre-shipment quarantine at an AZA or American Association for Laboratory Animal Science (AALAS) accredited institution may be applied to the receiving institution’s protocol. In such a case, shipment must take place in isolation from other primates. More stringent local, state, or federal regulations take precedence over these recommendations.

Quarantine length: Quarantine for all species should be under the supervision of a veterinarian and consist of a minimum of 30 days (unless otherwise directed by the staff veterinarian). Mammals: If during the 30-day quarantine period, additional mammals of the same order are introduced into a designated quarantine area, the 30-day period must begin over again. However, the addition of mammals of a different order to those already in quarantine will not have an adverse impact on the originally quarantined mammals. Birds, Reptiles, Amphibians, or Fish: The 30-day quarantine period must be closed for each of the above Classes. Therefore, the addition of any new birds into a bird quarantine area requires that the 30-day quarantine period begin again on the date of the addition of the new birds. The same applies for reptiles, amphibians, or fish.

Quarantine personnel: A keeper should be designated to care only for quarantined animals or a keeper should attend quarantined animals only after fulfilling responsibilities for resident species. Equipment used to feed and clean animals in quarantine should be used only with these animals. If this is not possible, then equipment must be cleaned with an appropriate disinfectant (as designated by the veterinarian supervising quarantine) before use with post-quarantine animals.

Institutions must take precautions to minimize the risk of exposure of animal care personnel to zoonotic diseases that may be present in newly acquired animals. These precautions should include the use of disinfectant foot baths, wearing of appropriate protective clothing and masks in some cases, and minimizing physical exposure in some species; e.g., primates, by the use of chemical rather than physical restraint. A tuberculin testing/surveillance program must be established for zoo/aquarium employees in order to ensure the health of both the employees and the animal collection.

Quarantine protocol: During this period, certain prophylactic measures should be instituted. Individual fecal samples or representative samples from large numbers of individuals housed in a limited area (e.g., birds of the same species in an aviary or frogs in a terrarium) should be collected at least twice and examined for gastrointestinal parasites. Treatment should be prescribed by the attending veterinarian. Ideally, release from quarantine should be dependent on obtaining two negative fecal results spaced a minimum of two weeks apart either initially or after parasiticide treatment. In addition, all animals should be evaluated for ectoparasites and treated accordingly.

Vaccinations should be updated as appropriate for each species. If the animal arrives without a vaccination history, it should be treated as an immunologically naive animal and given an appropriate series of vaccinations. Whenever possible, blood should be collected and sera banked. Either a -70°C (-94°F) frost-free freezer or a -20°C (-4°F) freezer that is not frost-free should be available to save sera. Such sera could provide an important resource for retrospective disease evaluation.

The quarantine period also represents an opportunity to, where possible, permanently identify all unmarked animals when anesthetized or restrained (e.g., tattoo, ear notch, ear tag, etc.). Also, whenever animals are restrained or immobilized, a complete physical, including a dental examination, should be performed. Complete medical records should be maintained and available for all animals during the quarantine period. Animals that die during quarantine should have a necropsy performed under the supervision of a veterinarian and representative tissues submitted for histopathologic examination.

Quarantine procedures: The following are recommendations and suggestions for appropriate quarantine procedures for jellyfish:
Required:
1. Direct and floatation fecals
2. Vaccinate as appropriate

Strongly Recommended:
1. CBC/sera profile
2. Urinalysis
3. Appropriate serology (FIP, FeLV, FIV)
4. Heartworm testing in appropriate species
Appendix D: Program Animal Policy & Position Statement

Program Animal Policy

Originally approved by the AZA Board of Directors – 2003
Updated and approved by the Board – July 2008 & June 2011

The Association of Zoos & Aquariums (AZA) recognizes many benefits for public education and, ultimately, for conservation in program animal presentations. AZA’s Conservation Education Committee’s Program Animal Position Statement summarizes the value of program animal presentations (see pages 42-44).

For the purpose of this policy, a Program Animal is defined as “an animal whose role includes handling and/or training by staff or volunteers for interaction with the public and in support of institutional education and conservation goals”. Some animals are designated as Program Animals on a full-time basis, while others are designated as such only occasionally. Program Animal-related Accreditation Standards are applicable to all animals during the times that they are designated as Program Animals.

There are three main categories of Program Animal interactions:

1. On Grounds with the Program Animal Inside the Exhibit/Enclosure:
   i. Public access outside the exhibit/enclosure. Public may interact with animals from outside the exhibit/enclosure (e.g., giraffe feeding, touch tanks).
   ii. Public access inside the exhibit/enclosure. Public may interact with animals from inside the exhibit/enclosure (e.g., lorikeet feedings, ‘swim with’ programs, camel/pony rides).

2. On Grounds with the Program Animal Outside the Exhibit/Enclosure:
   i. Minimal handling and training techniques are used to present Program Animals to the public. Public has minimal or no opportunity to directly interact with Program Animals when they are outside the exhibit/enclosure (e.g., raptors on the glove, reptiles held “presentation style”).
   ii. Moderate handling and training techniques are used to present Program Animals to the public. Public may be in close proximity to, or have direct contact with, Program Animals when they’re outside the exhibit/enclosure (e.g., media, fund raising, photo, and/or touch opportunities).
   iii. Significant handling and training techniques are used to present Program Animals to the public. Public may have direct contact with Program Animals or simply observe the in-depth presentations when they’re outside the exhibit/enclosure (e.g., wildlife education shows).

3. Off Grounds:
   i. Handling and training techniques are used to present Program Animals to the public outside of the zoo/aquarium grounds. Public may have minimal contact or be in close proximity to and have direct contact with Program Animals (e.g., animals transported to schools, media, fund raising events).

These categories assist staff and accreditation inspectors in determining when animals are designated as Program Animals and the periods during which the Program Animal-related Accreditation Standards are applicable. In addition, these Program Animal categories establish a framework for understanding increasing degrees of an animal’s involvement in Program Animal activities.

Program animal presentations bring a host of responsibilities, including the safety and welfare of the animals involved, the safety of the animal handler and public, and accountability for the take-home, educational messages received by the audience. Therefore, AZA requires all accredited institutions that make program animal presentations to develop an institutional program animal policy that clearly identifies and justifies those species and individuals approved as program animals and details their long-term management plan and educational program objectives.

AZA’s accreditation standards require that education and conservation messages must be an integral component of all program animal presentations. In addition, the accreditation standards require that the conditions and treatment of animals in education programs must meet standards set for the remainder of the animal collection, including species-appropriate shelter, exercise, appropriate environmental enrichment, access to veterinary care, nutrition, and other related standards. In addition, providing program animals with options to choose among a variety of conditions within their environment is
essential to ensuring effective care, welfare, and management. Some of these requirements can be met outside of the primary exhibit enclosure while the animal is involved in a program or is being transported. For example, free-flight birds may receive appropriate exercise during regular programs, reducing the need for additional exercise. However, the institution must ensure that in such cases, the animals participate in programs on a basis sufficient to meet these needs or provide for their needs in their home enclosures; upon return to the facility the animal should be returned to its species-appropriate housing as described above.

**Program Animal Position Statement**

*Last revision 1/28/03*
*Re-authorized by the Board June 2011*

The Conservation Education Committee (CEC) of the Association of Zoos and Aquariums supports the appropriate use of program animals as an important and powerful educational tool that provides a variety of benefits to zoo and aquarium educators seeking to convey cognitive and affective (emotional) messages about conservation, wildlife and animal welfare. Utilizing these animals allows educators to strongly engage audiences. As discussed below, the use of program animals has been demonstrated to result in lengthened learning periods, increased knowledge acquisition and retention, enhanced environmental attitudes, and the creation of positive perceptions concerning zoo and aquarium animals.

**Audience Engagement**

Zoos and aquariums are ideal venues for developing emotional ties to wildlife and fostering an appreciation for the natural world. However, developing and delivering effective educational messages in the free-choice learning environments of zoos and aquariums is a difficult task. Zoo and aquarium educators are constantly challenged to develop methods for engaging and teaching visitors who often view a trip to the zoo as a social or recreational experience (Morgan and Hodgkinson, 1999). The use of program animals can provide the compelling experience necessary to attract and maintain personal connections with visitors of all motivations, thus preparing them for learning and reflection on their own relationships with nature.

Program animals are powerful catalysts for learning for a variety of reasons. They are generally active, easily viewed, and usually presented in close proximity to the public. These factors have proven to contribute to increasing the length of time that people spend watching animals in zoo exhibits (Bitgood, Patterson and Benefield, 1986, 1988; Wolf and Tymitz, 1981).

In addition, the provocative nature of a handled animal likely plays an important role in captivating a visitor. In two studies (Povey, 2002; Povey and Rios, 2001), visitors viewed animals three and four times longer while they were being presented in demonstrations outside of their enclosure with an educator than while they were on exhibit. Clearly, the use of program animals in shows or informal presentations can be effective in lengthening the potential time period for learning and overall impact.

Program animals also provide the opportunity to personalize the learning experience, tailoring the teaching session to what interests the visitors. Traditional graphics offer little opportunity for this level of personalization of information delivery and are frequently not read by visitors (Churchman, 1985; Johnston, 1998). For example, Povey (2001) found that only 25% of visitors to an animal exhibit read the accompanying graphic; whereas, 45% of visitors watching the same animal handled in an educational presentation asked at least one question and some asked as many as seven questions. Having an animal accompany the educator allowed the visitors to make specific inquiries about topics in which they were interested.

**Knowledge Acquisition**

Improving our visitors' knowledge and understanding regarding wildlife and wildlife conservation is a fundamental goal for many zoo educators using program animals. A growing body of evidence supports the validity of using program animals to enhance delivery of these cognitive messages as well.
MacMillen (1994) found that the use of live animals in a zoomobile outreach program significantly enhanced cognitive learning in a vertebrate classification unit for sixth grade students. Sherwood and his colleagues (1989) compared the use of live horseshoe crabs and sea stars to the use of dried specimens in an aquarium education program and demonstrated that students made the greatest cognitive gains when exposed to programs utilizing the live animals. Povey and Rios (2002) noted that in response to an open-ended survey question (“Before I saw this animal, I never realized that . . .”), visitors watching a presentation utilizing a program animal provided 69% cognitive responses (i.e., something they learned) versus 9% made by visitors viewing the same animal in its exhibit (who primarily responded with observations). Povey (2002) recorded a marked difference in learning between visitors observing animals on exhibit versus being handled during informal presentations. Visitors to demonstrations utilizing a raven and radiated tortoises were able to answer questions correctly at a rate as much as eleven times higher than visitors to the exhibits.

Enhanced Environmental Attitudes

Program animals have been clearly demonstrated to increase affective learning and attitudinal change. Studies by Yerke and Burns (1991) and Davison and her colleagues (1993) evaluated the effect live animal shows had on visitor attitudes. Both found their shows successfully influenced attitudes about conservation and stewardship. Yerke and Burns (1993) also evaluated a live bird outreach program presented to Oregon fifth-graders and recorded a significant increase in students’ environmental attitudes after the presentations. Sherwood and his colleagues (1989) found that students who handled live invertebrates in an education program demonstrated both short and long-term attitudinal changes as compared to those who only had exposure to dried specimens. Povey and Rios (2002) examined the role program animals play in helping visitors develop positive feelings about the care and well-being of zoo animals. As observed by Wolf and Tymitz (1981), zoo visitors are deeply concerned with the welfare of zoo animals and desire evidence that they receive personalized care.

Conclusion

Creating positive impressions of aquarium and zoo animals, and wildlife in general, is crucial to the fundamental mission of zoological institutions. Although additional research will help us delve further into this area, the existing research supports the conclusion that program animals are an important tool for conveying both cognitive and affective messages regarding animals and the need to conserve wildlife and wild places.

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References


Appendix E: Developing an Institutional Program Animal Policy

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Rationale

Membership in AZA requires that an institution meet the AZA Accreditation Standards collectively developed by our professional colleagues. Standards guide all aspects of an institution's operations; however, the accreditation commission has asserted that ensuring that member institutions demonstrate the highest standards of animal care is a top priority. Another fundamental AZA criterion for membership is that education be affirmed as core to an institution's mission. All accredited public institutions are expected to develop a written education plan and to regularly evaluate program effectiveness.

The inclusion of animals (native, exotic and domestic) in educational presentations, when done correctly, is a powerful tool. CEC's **Program Animal Position Statement** describes the research underpinning the appropriate use of program animals as an important and powerful educational tool that provides a variety of benefits to zoo and aquarium educators seeking to convey cognitive and affective messages about conservation and wildlife.

Ongoing research, such as AZA's Multi-Institutional Research Project (MIRP) and research conducted by individual AZA institutions will help zoo educators to determine whether the use of program animals conveys intended and/or conflicting messages and to modify and improve programs accordingly and to ensure that all program animals have the best possible welfare.

When utilizing program animals our responsibility is to meet both our high standards of animal care and our educational goals. Additionally, as animal management professionals, we must critically address both the species' conservation needs and the welfare of the individual animal. Because "wild creatures differ endlessly," in their forms, needs, behavior, limitations and abilities (Conway, 1995), AZA, through its Animal Welfare Committee, has recently given the responsibility to develop taxon- and species-specific animal welfare standards and guidelines to the Taxon Advisory Groups (TAG) and Species Survival Plan® Program (SSP). Experts within each TAG or SSP, along with their education advisors, are charged with assessing all aspects of the taxons' and/or species' biological and social needs and developing Animal Care Manuals (ACMs) that include specifications concerning their use as program animals.

However, even the most exacting standards cannot address the individual choices faced by each AZA institution. Therefore, each institution is required to develop a program animal policy that articulates and evaluates program benefits. The following recommendations are offered to assist each institution in formulating its own Institutional Program Animal Policy, which incorporates the AZA Program Animal Policy and addresses the following matters.

The Policy Development Process

Within each institution, key stakeholders should be included in the development of that institution's policy, including, but not limited to representatives from:

- the Education Department
- the Animal Husbandry Department
- the Veterinary and Animal Health Department
- the Conservation & Science Department
- the Behavioral Husbandry Department
- any animal show staff (if in a separate department)
- departments that frequently request special program animal situations (e.g., special events, development, marketing, zoo or aquarium society, administration)
Additionally, staff from all levels of the organization should be involved in this development (e.g., curators, keepers, education managers, interpreters, volunteer coordinators).

To develop a comprehensive Program Animal Policy, we recommend that the following components be included:

I. Philosophy
In general, the position of the AZA is that the use of animals in up close and personal settings, including animal contact, can be extremely positive and powerful, as long as:

1. The use and setting is appropriate.
2. Animal and human welfare is considered at all times.
3. The animal is used in a respectful, safe manner and in a manner that does not misrepresent or degrade the animal.
4. A meaningful conservation message is an integral component. Read the AZA Board-approved Conservation Messages.
5. Suitable species and individual specimens are used.

Institutional program animal policies should include a philosophical statement addressing the above, and should relate the use of program animals to the institution's overall mission statement.

II. Appropriate Settings
The Program Animal Policy should include a listing of all settings both on and off site, where program animal use is permitted. This will clearly vary among institutions. Each institution's policy should include a comprehensive list of settings specific to that institution. Some institutions may have separate policies for each setting; others may address the various settings within the same policy. Examples of settings include:

I. On-site programming
   A. Informal and non-registrants:
      1. On-grounds programming with animals being brought out (demonstrations, lectures, parties, special events, and media)
      2. Children's zoos and contact yards
      3. Behind-the-scenes open houses
      4. Shows
      5. Touch pools
   B. Formal (registration involved) and controlled settings
      1. School group programs
      2. Summer Camps
      3. Overnights
      4. Birthday Parties
      5. Animal rides
      6. Public animal feeding programs

II. Offsite and Outreach
   1. PR events (TV, radio)
   2. Fundraising events
   3. Field programs involving the public
   4. School visits
   5. Library visits
   6. Nursing Home visits (therapy)
   7. Hospital visits
   8. Senior Centers
   9. Civic Group events

In some cases, policies will differ from setting to setting (e.g., on-site and off-site use with media). These settings should be addressed separately, and should reflect specific animal health issues, assessment of distress in these situations, limitations, and restrictions.
III. Compliance with Regulations

All AZA institutions housing mammals are regulated by the USDA's Animal Welfare Act. Other federal regulations, such as the Marine Mammal Protection Act, may apply. Additionally, many states, and some cities, have regulations that apply to animal contact situations. Similarly, all accredited institutions are bound by the AZA Code of Professional Ethics. It is expected that the Institution Program Animal Policy address compliance with appropriate regulations and AZA Accreditation Standards.

IV. Collection Planning

All AZA accredited institutions should have a collection planning process in place. Program animals are part of an institution's overall collection and must be included in the overall collection planning process. The AZA Guide to Accreditation contains specific requirements for the institution collection plan. For more information about collection planning in general, please see the Collection Management pages in the Members Only section.

The following recommendations apply to program animals:

1. Listing of approved program animals (to be periodically amended as collection changes). Justification of each species should be based upon criteria such as:
   - Temperament and suitability for program use
   - Husbandry requirements
   - Husbandry expertise
   - Veterinary issues and concerns
   - Ease and means of acquisition / disposition according to the AZA code of ethics
   - Educational value and intended conservation message
   - Conservation Status
   - Compliance with TAG and SSP guidelines and policies

2. General guidelines as to how each species (and, where necessary, for each individual) will be presented to the public, and in what settings

3. The collection planning section should reference the institution's acquisition and disposition policies.

V. Conservation Education Message

As noted in the AZA Accreditation Standards, if animal demonstrations are part of an institution's programs, an educational and conservation message must be an integral component. The Program Animal Policy should address the specific messages related to the use of program animals, as well as the need to be cautious about hidden or conflicting messages (e.g., "petting" an animal while stating verbally that it makes a poor pet). This section may include or reference the AZA Conservation Messages.

Although education value and messages should be part of the general collection planning process, this aspect is so critical to the use of program animals that it deserves additional attention. In addition, it is highly recommended to encourage the use of biofacts in addition to or in place of the live animals. Whenever possible, evaluation of the effectiveness of presenting program animals should be built into education programs.

VI. Human Health and Safety

The safety of our staff and the public is one of the greatest concerns in working with program animals. Although extremely valuable as educational and affective experiences, contact with animals poses certain risks to the handler and the public. Therefore, the human health and safety section of the policy should address:

1. Minimization of the possibility of disease transfer from non-human animals to humans, and vice-versa (e.g., handwashing stations, no touch policies, use of hand sanitizer)
2. Safety issues related to handlers' personal attire and behavior (e.g., discourage or prohibit use of long earrings, perfume and cologne, not eating or drinking around animals, smoking etc.)

AZA's Animal Contact Policy provides guidelines in this area; these guidelines were incorporated into accreditation standards in 1998.
VII. Animal Health and Welfare

Animal health and welfare are the highest priority of AZA accredited institutions. As a result, the Institutional Program Animal Policy should make a strong statement on the importance of animal welfare. The policy should address:

1. General housing, husbandry, and animal health concerns (e.g. that the housing and husbandry for program animals meets or exceeds general AZA standards and that the physical, social and psychological needs of the individual animal, such as adequate rest periods, provision of enrichment, visual cover, contact with conspecifics as appropriate, etc., are accommodated).
2. Where ever possible provide a choice for animal program participation, e.g., retreat areas for touch tanks or contact yards, evaluation of willingness/readiness to participate by handler, etc.)
3. The empowerment of handlers to make decisions related to animal health and welfare; such as withdrawing animals from a situation if safety or health is in danger of being compromised.
4. Requirements for supervision of contact areas and touch tanks by trained staff and volunteers.
5. Frequent evaluation of human / animal interactions to assess safety, health, welfare, etc.
6. Ensure that the level of health care for the program animals is consistent with that of other animals in the collection.
7. Whenever possible have a “cradle to grave” plan for each program animal to ensure that the animal can be taken care of properly when not used as a program animal anymore.
8. If lengthy “down” times in program animal use occur, staff should ensure that animals accustomed to regular human interactions can still maintain such contact and receive the same level of care when not used in programs.

VIII. Taxon Specific Protocols

We encourage institutions to provide taxonomically specific protocols, either at the genus or species level, or the specimen, or individual, level. Some taxon-specific guidelines may affect the use of program animals. To develop these, institutions refer to the Conservation Programs Database.

Taxon and species-specific protocols should address:

1. How to remove the individual animal from and return it to its permanent enclosure, including suggestions for operant conditioning training.
2. How to crate and transport animals.
4. Situation specific handling protocols (e.g., whether or not animal is allowed to be touched by the public, and how to handle in such situations)
5. Guidelines for disinfecting surfaces, transport carriers, enclosures, etc. using environmentally safe chemicals and cleaners where possible.
7. Limitations and restrictions regarding ambient temperatures and or weather conditions.
8. Time limitations (including animal rotation and rest periods, as appropriate, duration of time each animal can participate, and restrictions on travel distances).
9. The numbers of trained personnel required to ensure the health and welfare of the animals, handlers and public.
10. The level of training and experience required for handling this species.
12. The use of hand lotions by program participants that might touch the animals.

IX. Logistics: Managing the Program

The Institutional Policy should address a number of logistical issues related to program animals, including:

1. Where and how the program animal collection will be housed, including any quarantine and separation for animals used off-site.
2. Procedures for requesting animals, including the approval process and decision making process.
3. Accurate documentation and availability of records, including procedures for documenting animal usage, animal behavior, and any other concerns that arise.

X. Staff Training
Thorough training for all handling staff (keepers, educators, and volunteers, and docents) is clearly critical. Staff training is such a large issue that many institutions may have separate training protocols and procedures. Specific training protocols can be included in the Institutional Program Animal Policy or reference can be made that a separate training protocol exists.

It is recommended that the training section of the policy address:

1. Personnel authorized to handle and present animals.
2. Handling protocol during quarantine.
3. The process for training, qualifying and assessing handlers including who is authorized to train handlers.
4. The frequency of required re-training sessions for handlers.
5. Personnel authorized to train animals and training protocols.
6. The process for addressing substandard performance and noncompliance with established procedures.
7. Medical testing and vaccinations required for handlers (e.g., TB testing, tetanus shots, rabies vaccinations, routine fecal cultures, physical exams, etc.).
8. Training content (e.g., taxonomically specific protocols, natural history, relevant conservation education messages, presentation techniques, interpretive techniques, etc.).
9. Protocols to reduce disease transmission (e.g., zoonotic disease transmission, proper hygiene and hand washing requirements, as noted in AZA's Animal Contact Policy).
10. Procedures for reporting injuries to the animals, handling personnel or public.
11. Visitor management (e.g., ensuring visitors interact appropriately with animals, do not eat or drink around the animal, etc.).

XI. Review of Institutional Policies

All policies should be reviewed regularly. Accountability and ramifications of policy violations should be addressed as well (e.g., retraining, revocation of handling privileges, etc.). Institutional policies should address how frequently the Program Animal Policy will be reviewed and revised, and how accountability will be maintained.

XII. TAG and SSP Recommendations

Following development of taxon-specific recommendations from each TAG and SSP, the institution policy should include a statement regarding compliance with these recommendations. If the institution chooses not to follow these specific recommendations, a brief statement providing rationale is recommended.