SEABIRDS
(*Alcidae*)
CARE MANUAL

CREATED BY THE
AZA Charadriiformes Taxon Advisory Group
IN ASSOCIATION WITH THE
AZA Animal Welfare Committee

Seabirds (*Alcidae*) Care Manual

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Authors and Significant Contributors:

Aimee Greenebaum, Charadriiformes TAG Vice-Chair, Monterey Bay Aquarium, USA
Alex Waier, Sea World of Florida, USA
Cindy Pinger, Charadriiformes TAG Program Leader, Birmingham Zoo, USA
Debbie Zombeck, North Carolina Zoo, USA
Elizabeth Chittick Nolan, DVM, SeaWorld Orlando, USA
Ellen Dierenfeld, PhD, Ellen S. Dierenfeld, LLC, Saint Louis, USA
Eric Miller, Monterey Bay Aquarium, USA
Heidi Cline, Alaska SeaLife Center, USA
Jennifer Miller, SeaWorld San Diego, USA
Justin Brackett, SeaWorld San Diego, USA
Kim Peterson, SeaWorld San Diego, USA
Kim Wanders, Monterey Bay Aquarium, USA
Kristin Pelo, Alaska SeaLife Center, USA
Liane Berlepsch, SeaWorld Orlando, USA
Linda Henry, SeaWorld San Diego, USA
Lori Smith, National Aquarium, USA
Mary Carlson, Charadriiformes Program Advisor, Seattle Aquarium, USA
Mike Macek, Piciformes Tag Chair, Curator of Birds, St. Louis Zoo, USA
Sara Perry, Seattle Aquarium, USA
Sherry Branch, SeaWorld Orlando, USA
Stephanie McCain, DVM, Charadriiformes Veterinarian Advisor, Birmingham Zoo, USA
Tasha DiMarzio, Alaska SeaLife Center, USA
Todd Schmitt, DVM, SeaWorld San Diego, USA
Wendy Turner, Curator of Birds, SeaWorld San Diego, USA

Reviewers:

Tasha DiMarzio, Avian Curator, Alaska SeaLife Center, USA
Jo Gregson, Curator of Birds, Paignton Zoo Environmental Park, UK

AZA Staff Editors:

Emily Wagner, Animal Care Manual Editor Consultant
Felicia Spector, MA, Animal Care Manual Editor Consultant
Rebecca Greenberg, Animal Programs Coordinator
Candice Dorsey, PhD, Senior Vice President, Conservation, Management & Welfare Sciences
Hana Johnstone, Conservation, Management & Welfare Sciences Intern
Raven Spencer, Conservation, Management & Welfare Sciences Intern

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Jeff Pribble

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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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 Alcidae

Classification	Taxonomy
Kingdom	Animalia
Phylum	Chordata
Class	Aves
Order	Charadriiformes
Suborder	Alcae
Family	Alcidae

Genus, Species, and Status

Table 2. Genus, species, and status information for Alcidae

Genus	Species	Common Name	USA Status	IUCN Status	AZA Status
Uria	*aalge*	Common Murre	Not Listed	Least Concern	Green SSP
Uria	*lomvia*	Thick-billed Murre	Not Listed	Least Concern	
Alca	*torda*	Razorbill	Not Listed	Near Threatened	
Alle	*alle*	Dovekie	Not Listed	Least Concern	
Cepphus	*columba*	Pigeon Guillemot	Not Listed	Least Concern	
Cepphus	*grylle*	Black Guilllemot	Not Listed	Least Concern	
Cepphus	*carbo*	Spectacled Guillemot	Not Listed	Least Concern	
Synthliboramphus	*hypoleucus*	Guadalupe murrelet	Not listed	Endangered	
Synthliboramphus	*scrippsi*	Scripps' murrelet	Not listed	Vulnerable	
Synthliboramphus	*craveri*	Craveri's Murrelet	Not Listed	Vulnerable	
Synthliboramphus	*antiquus*	Ancient Murrelet	Not Listed	Least Concern	
Synthliboramphus	*wumizusume*	Japanese murrelet	Not Listed	Vulnerable	
Brachyramphus	*brevirostris*	Kittlitz's Murrelet	Not Listed	Near Threatened	
Brachyramphus	*marmoratus*	Marbled Murrelet	Threatened	Endangered	
Brachyramphus	*perdix*	Long-billed Murrelet	Not Listed	Near Threatened	
Ptychoramphus	*aleuticus*	Cassin's Auklet	Not Listed	Near Threatened	
Aethia	*psittacula*	Parakeet Auklet	Not Listed	Least Concern	
Aethia	*pusilla*	Least Auklet	Not Listed	Least Concern	
Aethia	*cristatella*	Crested Auklet	Not Listed	Least Concern	
Aethia	*pygmaea*	Whiskered Auklet	Not Listed	Least Concern	
Cerorhinca	*monocerata*	Rhinoceros Auklet	Not Listed	Least Concern	
Fratercula	*cirrhata*	Tufted Puffin	Under Review	Least Concern	Green SSP
Fratercula	*corniculata*	Horned Puffin	Not Listed	Least Concern	Yellow SSP
Fratercula	*arctica*	Atlantic Puffin	Not Listed	Vulnerable	Green SSP

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), biologists, veterinarians, nutritionists, reproduction physiologists, behaviorists, and researchers (visit the AZA Animal Program page to contact these individuals). It is based on the most current science, practices, and technologies used in animal care and management, and is a valuable resource that enhances animal welfare by providing information about the basic requirements needed and best practices known for caring for *ex situ* alcid 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 alcids must comply with all relevant local, state/provincial, and federal wildlife laws and/or regulations; AZA accreditation standards that are more stringent than these laws and/or regulations must be met (AZA Accreditation Standard 1.1.1).

AZA Accreditation Standard

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

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

Natural history: Alcids, also known as auks, are a diverse group of seabirds that spend most of their year in open water. They are the black and white counterpart to the penguin of the Southern Hemisphere. There are 23 species in the Alcidae family. Despite intensive surveys and censuses, our knowledge of alcid populations is very fragmented. Population numbers are well known in certain parts of their range, but they are poorly known in others. This makes it hard to know total world populations very accurately. Three species stand out as vulnerable to extinction in the short-term: Japanese, Xantus', and Craveri's murrelets. All have global populations estimated to be less than 10,000 breeding pairs (Gaston & Jones, 1998).

Habitat preferences, flight, and anatomy: Little is known about alcids' time out at sea. Most information about alcids is based on the small amount of time spent on land for breeding. They are only found in the Northern Hemisphere. They forage entirely in the ocean and only come to land for breeding. They are relatively clumsy on land and have difficulty taking off in flight. This can make alcids vulnerable to land-based predators and humans hunting them. Because of the demands of wing-propelled swimming, they have small wings relative to their body weight. Therefore, when in flight, they are entirely dependent on flapping to remain aloft and this puts heavy demands on their pectoral muscles, which are supported by an elongated attachment to the sternum (Gaston & Jones, 1998). See Figure 1 below for a visual of alcid anatomy.

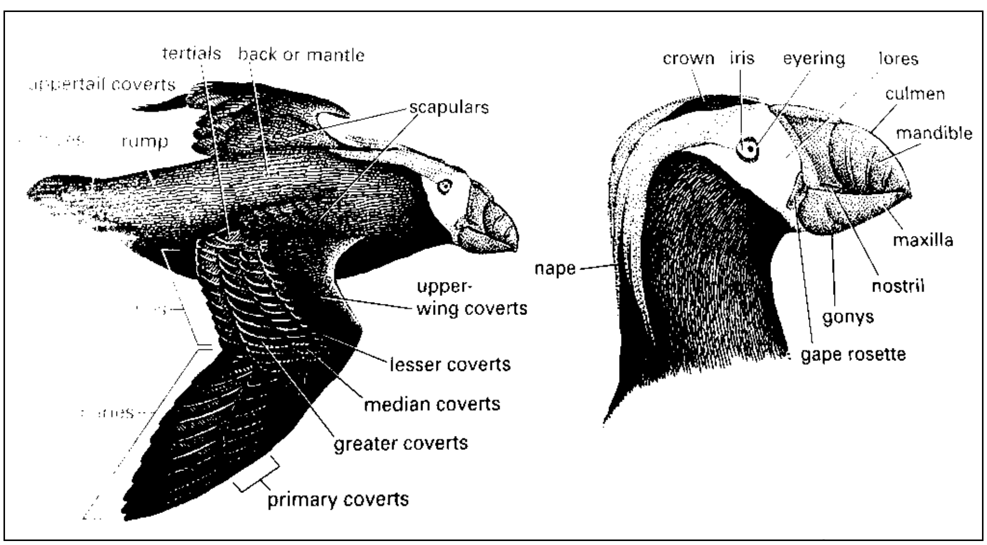

Figure 1. Alcid anatomy (Gaston & Jones, 1998)

Feeding habits: Alcids can be, for the most part, categorized as piscivore or planktivore. The smaller alcids (i.e., dovekie, murrelets, guillemots, and auklets) tend to eat more plankton, crustaceans, and invertebrates. Larger species including the murre, puffin, and razorbill eat more fish. The diets are not exclusive to one group, and there is overlap of the two groups.

For most species of this group, food destined for the chicks is carried externally, dangling from the bill (external transporters). In some species of auklets and in the dovekie, breeding adults develop a pouch in the throat in which the food is carried for their chicks (internal transporters). The chicks of the external transporters are fed almost exclusively on fish, while the internal transporters will also feed planktonic crustaceans and/or larval fish (Gaston & Jones, 1998).

Alcids are capable of diving to impressive depths. Dovekies (one of the smallest auks) can dive up to 30 m (98.43 ft), while murres can dive to over 150 m (492.13 ft) (Gaston & Jones, 1998). The vast majority of dives are much shallower than the maximum depths recorded for various species. For example, thick-billed murres mainly forage between 20–40 m (65.62–131.23 ft), while auklets usually concentrate between 10–30 m (32.81–98.43 ft) (Gaston & Jones, 1998).

Migration: Alcids do not exhibit the typical “leapfrog” migration, in which northern populations pass over southern populations to winter further south, that many species of land birds and waders exhibit. In general, alcid species and populations that breed furthest north also winter furthest north. Few alcids engage in any systematic long-distance movements; most migrations consist of a gradual southerly drift. The main exceptions to this general rule are the thick-billed murres breeding in the Barents Sea and Baffin Bay, many of which migrate several thousand kilometers to winter off southwest Greenland and Newfoundland, and those species breeding in Alaska that winter in the cold-current waters off California (Gaston & Jones, 1998).

History, habitat, and vulnerability: Although alcids’ main role in the ecosystem tends to be either as predators or prey, they can affect the ecosystems in a variety of ways including nutrient enrichment through defecation. Even though they only visit the land for short periods of time, the dense concentrations of birds have appreciable effects on local soils and vegetation. For example, in the Arctic, manuring by thick-billed murres, auklets, and dovekies creates local pockets of vegetation in otherwise barren areas (Gaston & Jones, 1998).

Alcids have been harvested for their meat as well as their eggs. In addition to their use as food, certain alcids were historically valued for their body parts that have been used for clothing and decoration. Substantial harvesting still occurs in several areas. For example, traditional harvesting occurs in Iceland, the Faeroes, West Greenland, and Newfoundland and Labrador. They also suffer from predators like rats, cats, and foxes introduced to their nesting areas. Furthermore, they are often affected by oil spills and caught in fishing nests when diving for food.

Breeding and lifecycle: All species are socially monogamous, and both sexes play equal roles in the incubation and rearing of chicks. Most species show strong site fidelity, in that they come back to breed in the location where they were raised (Gaston & Jones, 1998). Most species lay a single egg and have two lateral brood patches. The murres and the dovekies have a central brood patch (see Figure 2). Most alcids are colonial breeders and will have a high density of birds in one area.

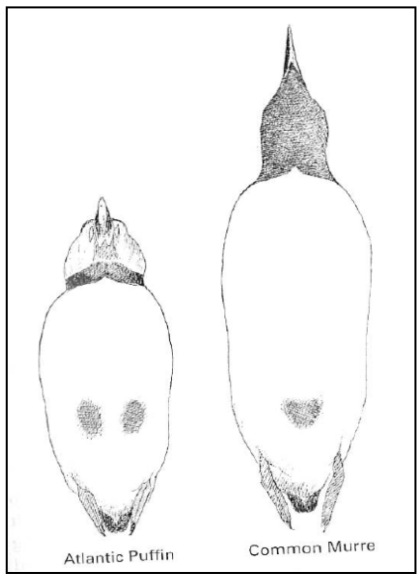

Figure 2. Examples of brood patches (Gaston and Jones, 1998)

Regulations: Alcids are managed by the United States Fish and Wildlife Service (USFWS) under the Migratory Bird Treaty Act. Most non-releasable alcids are acquired from local wildlife rehabilitation centers, especially after oil spills. Please consult with your regional USFWS Migratory Bird Office, as well as check for any specific state laws, when acquiring or transferring alcids that came from the wild.

Relevant alcid terminology:

- Pelagic: Relating to or living in the sea far from the shore
- Auks: English equivalent to Alcidae, to include all members of the family

Chapter 1. Ambient Environment

1.1 Temperature and Humidity

The animals must be protected or provided accommodation from weather, and any adverse conditions. (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.

> **AZA Accreditation Standard**
>
> **(1.5.7)** The animals must be protected or provided accommodation from weather or other conditions clearly known to be detrimental to their health or welfare.

The air temperature of indoor alcid exhibits should range between 4.44–15.56 °C (40–60 °F), with an average temperature of 10 °C (50 °F). This is similar to the temperatures that alcids face in their natural range during the summer months, which is what most exhibits aim to replicate. The temperature can be decreased slightly during winter months as the photoperiod is changed, but this is not considered essential. Excessively high temperatures or temperature fluctuations can encourage fungal growth and sporulation which is potentially detrimental to flock health.

Alcids in outdoor displays have been kept successfully in air temperatures ranging from -2.22–29.44 °C (28–85 °F) with the high ranges only occurring for short periods during the day and not usually sustained overnight (K. Anderson, personal communication, 2005). Provisions should be made for birds to have both shaded and sunny areas of the exhibit to swim in and haul out at all times. Having a variety of areas with differing ranges of light and temperature gives the birds many choices depending on their need and/or desire. Sunny locations provide areas for both basking and loafing or for warming during inclement weather while shaded areas provide protection from extensive heat and/or sun. Alcids have been observed basking in sunny spots in outdoor exhibits on cool days (Zombeck & Carlson, 2004), and pigeon guillemots in the field have been observed utilizing shade during hot and sunny days (Konyukhov, 2000). In addition, making use of shade might be a less energetically expensive method of cooling as opposed to using physiological methods (Schreiber & Burger, 2001), and, therefore, less stressful to the bird. Light-filled and shaded areas also assist the collection in dealing with humidity levels.

Exhibit orientation and the strategic placement of vegetation, canopies, and rockwork (caves, crevices, various heights of cliffs, and outcroppings) will create sunny and shady areas for the collection to utilize. Canopies are useful in that they can create shade, protect the collection from excessive snow or wind, and reduce the amount of fecal material that drops into the exhibit. A canopy cover needs to be able to withstand wind, ripping, snow load, and freezing conditions. Care in its placement is required so it does not restrict too much airflow into the exhibit.

Cold weather: More research is needed about how the birds do in freezing temperatures. In the wild, they are usually out to sea during this time and are not typically exposed to accumulating snow and ice. Recurrent freezing for long periods of time in the exhibit may have detrimental effects on the birds, such as frostbite on their feet. If severe freezing conditions are encountered, extra care should be taken to prevent ice buildup on the rocks, for keeper safety and for ease of movement for the birds, especially as a result of cleaning/hosing. Freshwater freezes at higher temperatures than saltwater, so waterfalls in the exhibit may need to be drained and/or winterized to avoid large icicle build up and rockwork damage. Snowfall within the exhibit should be limited because alcids normally do not encounter snow on land during breeding season. If snow accumulates on the rockwork where the birds can access it, it should be rinsed with saltwater to get rid of it. Since snow can also pile up on the exhibit netting, this netting must either be strong enough to withstand the additional weight or it must be accessible so snow can easily be knocked off.

Important considerations should be made for ambient daily and seasonal temperature changes, and also for humidity. Exhibits need to avoid extremes of temperatures that could affect the health of the collection.

Care should be given to older birds or very young chicks and additional heat sources may be needed.

AZA institutions with exhibits which rely on climate control must have critical life-support systems for the animal collection and emergency backup systems available. Warning mechanisms and

> **AZA Accreditation Standard**
>
> **(10.2.1)** Critical life-support systems for the animals, 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. Warning mechanisms and emergency backup systems must be tested periodically.

backup systems must be tested periodically (AZA Accreditation Standard 10.2.1).

There are a number of facilities that successfully house alcids in climate-controlled, indoor habitats using regular HVAC air systems with air filters. A regular maintenance schedule should be determined and adhered to by trained life support staff. An air testing protocol and schedule should also be put in place; testing should be done at least once a year. Heat and lighting sources should also have a maintenance schedule and be checked by trained personnel on an annual basis.

For those facilities with climate-controlled habitats for their alcids, backup systems for keeping the temperature and humidity within normal parameters should always be in place in the event of equipment breakdown. Mechanisms should also be in place for detecting any changes in conditions from the normal range. Appropriately trained personnel should maintain life support systems and keep pertinent records (hardcopy and electronic) so as to find and repair failures and maintain regular checks.

1.2 Light

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

Alcids are diving seabirds found on the open ocean, seacoasts, and islands at latitudes greater than 25^{o}N. Within this range, solar radiation varies with latitude and season but includes electromagnetic (EM) bands measured in nanometers (nm) from ultraviolet (UV, 280–400 nm) through the human visual EM spectrum (VIS, 400–700 nm) to infrared (IR, >700 nm).

In general, alcids spend winters at sea and summers at breeding sites on remote islands and rocky shores (Gaston & Jones, 1998; Schreiber & Burger, 2001). In these habitats, alcids experience a variety of light environments on land and at sea. On land, nesting strategies vary from sunlit cliffs to shadowy rock crevices or dark earthen burrows (Gaston & Jones, 1998; Ydenberg, 1989). While most species are diurnal, some species are crepuscular or nocturnal (Gaston & Jones, 1998); a few species are socially active near their nests at night (Zubakin et al., 2010). Higher latitude species experience greater seasonal changes in the range of both daily light intensity and day length. At sea, reflected sunlight from the ocean surface intensifies light levels. But when diving, alcids transition from air to water, into a light environment that is immediately reduced in intensity and altered in spectrum (narrowing to ~450 nm) with increasing depth (Talley et al., 2011; Denton, 1990). As visual predators, alcids forage at ocean depths that range from 50 m to more than 170 m during the day; murre have been recorded diving to at least 80 m at night (Regular et al., 2010; Hedd et al., 2009) during very limited light levels (Regular, Hedd, & Montevecchi, 2011). Alcids, therefore, face several visual challenges that include winter low-light flight, flight at night, the need for visual accommodation when diving from a high-light ocean surface to low-light conditions at depth, foraging at depth in low-light or near-darkness, and visual detection and capture of prey in low light conditions.

The alcid eye may have specialized visual adaptations that balance both low- and high-light conditions. Unfortunately, limited species-specific data are available on alcid vision; however, an understanding of general avian visual systems, paired with studies on the visual adaptations of similar species, can inform light provision for alcids. Birds, including alcids, possess a complex visual system (Hart & Hunt, 2007). With a few exceptions, most birds have large eyes relative to their body size which, given the costs of flight, suggests the significance of this structure (Hall & Ross, 2007; Garamszegi, Møller, & Erritzøe, 2002). Diurnal birds discern light along a broad range of the electromagnetic (EM) spectrum (Hart, 2001), and have oil droplets that serve to fine-tune spectral delivery to cones (Vorobyev et al., 1998). In addition to a single type of rod (low-light vision), bird retinae have up to four-types of single cones (color vison), and a single type of double cone (non-color vision: luminance, texture, and motion detection) (Hart, 2001). As well as taking in light information via the eyes, birds have deep brain photoreceptors (extra-retinal light sensors involved in photoperiod recognition) which perceive light directly through the skull (Halford et al., 2009; Nakane et al., 2010; Menaker & Underwood, 1976). Birds also possess a capacity for rapid vision that allows images to be processed quickly as an adaptation for flight (Boström et al., 2016) and may be important for underwater foraging. Finally, visual fields in birds often favor lateral rather than binocular vision and are nearly panoramic (Martin, 2007). It is not surprising, then, that birds are highly visually dependent; mate choice, sexual signaling, prey detection, predator avoidance, flight, migration, geographic orientation, egg recognition, and chick-rearing are all visually driven (Martin, 2012; Shealer, 2001; Nelson & Baird, 2001). This multiplicity of visual characteristics underscores how differently a bird eye perceives the environment than that of the human eye (Martin, 2012). For polar seabirds, light plays an important role in behavior and reproduction (Ostaszewska et al., 2017).

Spectrum: In birds, color vision is based on four cone types: short-wavelength sensitive 1 and 2 (SWS1, SWS2), long-wavelength sensitive (LWS), and medium-wavelength sensitive (MWS). Birds also have one of two types of color vision—violet sensitive (VS) or ultraviolet sensitive (UVS)—based on a bias of the SWS1 cone. Several studies contend that seabirds, including alcids, have VS vision; peak sensitivities have been measured for three species of VS marine birds at 402–406 nm (Hart, 2004). Despite the "violet" description, birds with VS vision still perceive light into the near-UV end of the spectrum due to the bell-shape of the cone's response curve. Furthermore, Ross et al. (2013) found that birds, including alcids, from Polar Regions showed a preference for UVAdded light environments and this preference suggests that their welfare is enhanced by the presence of UV light in the habitat.

It is important, when assessing light from the avian perspective, to think of light as a series of wavelengths delivered as photons that are captured by photopigments (within photoreceptors) that in turn excite neural pathways; the brain processes these signals such that the sum of the photoreceptor signals creates the perception of "white light" (possibly including intensity), and the contrast of opponent signal input creates the perception of "color" (Kelber, Vorobyev, & Osorio, 2003). Color perception is important in mate recognition and choice (Olsson, Wilby, & Kelber, 2017). Auks employ a variety of visual displays (Bakhturina & Klenova, 2016) but few have been evaluated for their spectral components. Many alcids display colorful seasonal ornaments. Wails et al. (2017) found that the rictal plates of all crested auklets fluoresced when exposed to blue light (440–460 nm). While the definitive function of this is unknown, the growth of this structure prior to the breeding season suggests a possible role in mate selection. Kelly (2015) found early and late breeding season variances in the coloration and brightness of bills and feet in Atlantic puffins and Doutrelant et al. (2013) measured the spectral characteristics of Atlantic puffins facial and bill ornaments and found a mutual social or sexual signaling potential. Despite the lack of clarity as to the purpose of colorful ornaments in alcids, it seems clear that lighting of sufficient intensity along broad wavelengths should be provided in order for birds to properly perceive color signals.

While spectrum predominantly affects visual perception, light in the UVB range of the EM spectrum (280–315 nm) is important for Vitamin D synthesis in some bird species. Alcids have not been evaluated for their ability or need to synthesize Vitamin D and there are no reports of metabolic bone disease in the literature. For piscivorous birds fed a high quality diet of whole fishes, the diet should provide sufficient levels of Vitamin D (McWilliams, 2008). However, Adkesson and Langan (2007) reported metabolic bone disease in juvenile Humboldt penguins (*Spheniscus humboldti*) and Hoopes (2016) reported on a possible Vitamin D deficiency in an African penguin (*Spheniscus demersus*). Puffins have been held successfully with little or no access to UVB light with no apparent negative outcomes.

Intensity: The perception of intensity in birds is difficult to measure. Lux meters measure light intensity based on human visual parameters and are inadequate to assess intensity from the avian perspective (Nuboer, Coemans, & Vos, 1992). Double cones may play a role in intensity perception, along with the presence of a broad light spectrum and the peak sensitivities of photoreceptors. Rods may also play a role in the sensation of intensity. In assessing artificial light sources, Nuboer et al. (1992) proposed matching the Spectral Power Distribution (SPD) curve for the lamp with the spectral sensitivity curve for the species (in this case chickens). Regrettably, spectral sensitivity in alcids has not been measured. But, for the Humboldt penguin, a seabird with a similar life history, spectral sensitivities were measured at 504 nm (rods) and at 403, 450, 500 and 543 nm (cones) with sensitivity below 400 nm suggesting Humboldt penguin vision extends into the UV (Bowmaker & Martin, 1985). This species may serve as a reasonable proxy for assessing intensity for alcids.

Duration: Birds use photoperiod to control the timing of seasonal activities such as breeding and molt (Dawson et al., 2001). See Figure 3 for an example of a normal alcid molt cycle. Alcids, like other mid- and high-latitude species, benefit from a consistent Light/Dark (L/D) cycle. Most alcid managers understand the importance of a range-specific photoperiod to ensure the maintenance of circannual rhythms. However, diel spectral changes may also influence photoperiod detection (Kumar et al., 2007) so a gradual transition in light intensity and spectrum at dawn and dusk may enhance outcomes (Kumar et al., 2017; Malik, Rani, & Kumar, 2004; Kumar & Rani, 1999; Pohl, 1999). Variable by latitude and time of year, twilight is characterized by low intensity short wavelengths (below 450 nm), sunrise and sunset by longer wavelengths (~565–645 nm), followed by an increase in intensity and broadening of spectrum during the morning hours until the sun approaches its zenith (Henry, 2017; Malik et al., 2004). The phasing of different light sources with appropriate spectrum at low intensity, timed to come on and go off in the morning and evening hours, can achieve this effect.

Relevant to photoperiod, sufficient darkness (in this context referring to the absence of artificial light) may be as necessary to maintaining avian health as is the appropriate provision of light. Managers should take care in the design and implementation of exhibits to avoid extraneous light inputs from entering the habitat. Light bleed can occur from non-exhibit areas into bird habitats, e.g., from holiday lights, pathway lighting, animal support areas lighting (including windows, doors), guest areas lighting, reflective guest area surfaces, graphics displays, exit signs, etc., and urban sky glow where birds are housed outdoors. Such light inputs can serve to confuse L/D cycles and, at a minimum, may suppress melatonin expression with subsequent potential adverse effects on immune function (Dominoni et al., 2013; Falchi et al., 2011).

Photoperiodic induction occurs in birds at wavelengths between approximately 450–550 nm with a peak at 492 nm. Therefore, if a night light is to be used, it should be of very low intensity and in a spectral range that does not adversely affect photoperiod. Red light (~650 nm or greater) will penetrate the skull of birds and will induce photoperiodic response with long exposure. Light spectra in the 550–600 nm range, at very low intensity (20 lux or less, human vision), can provide sufficient light. Alcid eyes are likely, similarly to penguins, highly adapted for low light conditions (Henry, 2017).

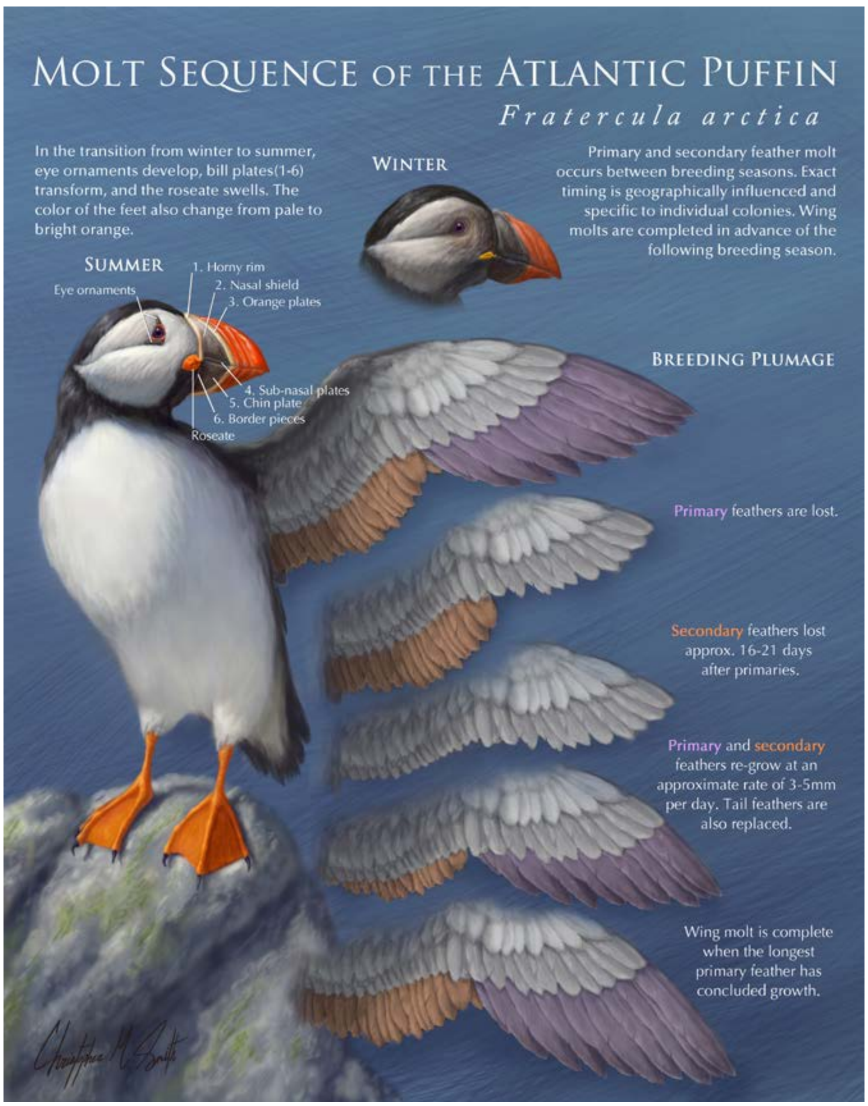

Figure 3. Molt sequence of the Atlantic puffin (Dial, 2015)

Source: The best source of illumination is sunlight; however, when artificial sources are supplemented or the sole basis of illumination, it may be necessary to combine light sources to meet alcid visual needs. Given the scope of light environments experienced by alcids, combined with visual-behavioral data, alcid light provision should include visible and UVA light spectra ranging from at least 350 nm to 650 nm. This is best achieved by combining natural and artificial light sources to maximize spectral range within the habitat. It should be noted that due to the spectral output peaks and valleys of metal halide (MH) and solid state lighting (SSL, e.g., light emitting diode, LED) avian color rendering (true color perception from the avian perspective) may be affected if these types of lights are used as a single-source light. (Color Rendering Indices (CRI) and other measures listed by light bulb manufacturers are not relevant to bird visual perception.) For all light installations, it is important to assure that the glass or plastics used between the light source and the subject are UV-transmissible to prevent the loss of desired spectrum as it is delivered into the space. In all cases, light sources should be evaluated using objective spectral measurement via the manufacturer-provided SPD Graphs and/or independent measurements of spectrum within the habitat using a spectrometer (e.g., StellarNet, Inc.). See Table 3 for a list of commonly used light sources and their characteristics.

Table 3: Natural and artificial light sources characteristics. It is important to assess the Spectral Power Distribution (SPD) graph for all lights under consideration to assure the presence of desired wavelengths. Overlaying SPD graphs can help determine if all desired wavelengths will be represented and in sufficient quantity. Follow-up spectrometer measurements are beneficial.

Source	Examples	Characteristics
Outdoor habitat/sunlight	• Habitats with daily outdoor access • Netted habitats	• Provides natural light spectrum from 280–700 nm • Requires increased awareness of extraneous light sources (light bleed) from adjacent areas • Has been used successfully for alcid exhibits • Supplement as needed for photoperiod or exhibitry
Daylighting	• Windows • Skylights • Reflected tube lighting (Solatube®)	• Skylights have been used successfully in alcid habitats • Provides natural light spectrum when used with UV-transmissible products • Natural light may only need to be supplemented with artificial light for exhibitry or photoperiod • Access for cleaning glass/plastic is necessary for maintaining light transmission quality
High Intensity Discharge (HID) lamps	• Metal halide (MH)	• Energy-efficient • Emit spectrum below 400 nm • Short-wavelength-shifted with discrete spikes along the EM spectrum • MH lamps have been used successfully in alcid habitats • Require annual relamping due to spectral shift with prolonged hours of use • High heat load

Solid State Lighting	• LED (Cree™)	• Point-source light; requires diffusers for light spread
		• Energy efficient
		• Low heat load; better function and lifespan in cooler environments
		• Little or no output below 400 nm and a dip in spectrum in the cyan range (~490–550 nm)
		• High flicker is associated with this technology
		    o There are reports of health consequences associated with flicker in humans (Wilkins, Veitch, & Lehman, 2010)
		    o Birds' process images more quickly and can discern the modulation of the light output of a lamp (the flicker)

		o Studies assessing the effects and perception of flicker in birds are conflicting (Inger et al., 2014; Rubene et al., 2010; Rubene, 2009; Evans, Cuthill, & Bennett, 2006; Greenwood et al., 2004) ▪ LEDs should be used judiciously ▪ Dimming of LED lights increases flicker ▪ High speed drivers may mitigate effects o UV-added environments may increase avian sensitivity to flicker (Rubene et al., 2010; Rubene, 2009) • RGB LED are not recommended for use in bird habitats
Light Emitting Plasma	• Chameleon® • Luxim® (high bay lights)	• Can be used in high-bay (large space) applications • SPD curve mirrors natural sunlight, including the UV range • Cost per unit for this technology is still quite high, but the spectral output may be worth the cost to introduce UV spectrum into an exhibit • No flicker associated with this type of light source • Relamping is recommended every 2-3 years
UVA light	• Ultraviolet light cannons or bird lights • Wildfire®, • Zoo Med® • Exo-Terra® (Solar Glo) • Osram Sylvania® (Ulltra Vitalux; 350BL Fluorescent tube blacklights)	• Short-wave length light is easily attenuated • Products vary regarding effective distance for sufficient UV light levels • Many include UVB
Incandescent	• Tungsten halogen	• Long wavelength shifted • Dimmable • Can be used in combination with MH to balance light spectrum • Can be dimmed up and down for sunrise/sunset effect to replicate diel cycles of light spectrum • Less energy efficient • No need to replace bulbs until failure
Fluorescent	• T8 "full spectrum"	• Full spectrum specifications are always based on human visual needs • Lower intensity than other sources • Possible association with cataract formation • Have been used successfully in alcid exhibits • Annual relamping is required due to spectral shift with use • Emit spectrum below 400 nm depending on bulb and filter coating

In penguins, fluorescent lighting was identified as a potential risk factor for cataract formation in macaroni penguins (Woodhouse, Peterson, & Schmitt, 2016) and may have a similar affect in alcids. By contrast, MH lighting was associated with decreased odds of cataracts (Woodhouse et al., 2016). Metal halide lamps have been successfully used in puffin habitats for many years. Light emitting plasma (LEP) is a type of lighting gaining interest among aquarists and herpetologists due to its spectral output that spans UVB to IR. These bulbs should be considered as a light source supplement due to their broad spectral output; the SPD looks very similar to that of natural light. Proper maintenance of light fixtures is essential to good quality light. Institutions should make provision for easy access to luminaires for maintenance. Annual replacement of some light bulbs may be necessary because some types of lamps experience a shift in the lamp spectral output with use, including “white” LED (Fan et al., 2017).

The importance of light provision for alcid neonates has not been investigated. Specialized lighting may not be required, because many alcid nestlings spend their first few months of life in a nest burrow without significant light exposure. The provision of an appropriate photoperiodic day length and spectrum during the nestling period, consistent with the parental habitat, may be beneficial as chicks develop and fledge.

Areas of future research should include investigating correlations between the various light provision schemes and alcid health and reproductive outcomes, as well as finding the most effective light sources for broad-spectrum light provision that includes UVA. Facilities in the North American Region have a variety of habitats with and without access to natural light and it would be worthwhile to survey light sources and photoperiods relative to health and reproductive outcomes to inform future best practices in lighting provision for alcids. As LED and LEP technologies become more widespread, especially when used as the only light source, such data will help determine if they are appropriate for alcid light provision.

Alcids are an excellent flagship species for highlighting conservation and light pollution effects on seabirds. Seabirds are among the species most affected by night time light pollution in coastal areas (Rodríguez et al., 2017a; Rodríguez et al., 2017b; Troy, Holmes, & Green, 2011; Montevecchi, 2006). Seabirds are also the most endangered group of birds (Croxall et al., 2012). As such, information gleaned from light provision, responses and outcomes for alcids in human care may benefit and inform conservation actions for free-living alcids and other seabirds. Light pollution is one of the fastest growing forms of pollution but often the least recognized (Hölker et al., 2010). Artificial lighting schemes developed for birds in human care can serve to highlight the importance of light, and dark, in the lives of all animals, and especially the balance required between the physiological need for darkness and the human fascination to light-up environments at night (Cabrera-Cruz, Smolinsky, & Buler, 2018; Davies et al., 2013; Kenney, 2016; Longcore & Rich, 2004).

1.3 Water and Air Quality

AZA-accredited institutions must have a regular program of monitoring water quality for 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.

AZA Accreditation Standard

(1.5.9) The institution must have a regular program of monitoring water quality for fish, marine mammals, and other aquatic animals. A written record must be maintained to document long-term water quality results and chemical additions.

Water Quality: Alcids are uniquely adapted for life on the water. They only come ashore in the spring and summer months to lay eggs and rear their young. It is important to design a water space that has a large enough surface area and depth to encourage natural behaviors such as swimming, diving, skimming, porpoising, roosting, preening, and exploration. It is thought that the more time the birds spend active in the water, the better overall condition they will be in; this will also mimic their natural pelagic behavior.

Either fresh or salt water (natural and artificial) is used in alcid exhibits pools. To date, there have been no documented problems associated with either. Many facilities with saltwater pools have sources of fresh water in the exhibit as well, such as a waterfall and/or small pools. It is possible that problems could occur if birds housed in a freshwater exhibit were sent to a saltwater exhibit, because pelagic birds kept in human care for long periods of time without a regular intake of salt can lose the ability to utilize salt water (Holmes & Phillips, 1985). More research is needed in this area.

The exhibit should also have efficient skimming to prevent materials (e.g., feathers, oils, etc.) from gathering at the surface. Good skimming will help maintain the good quality of alcids’ feathers. Skimmer numbers should correspond to pool size and configuration. Their entrances should also be safely covered to prevent birds from entering or getting caught in them, while still being easily accessible for ease of cleaning by staff. The skimmer cleaning schedule should be increased in accordance with seasonal events, such as molting. In 2004, an Alcid Exhibit Design Survey was sent out to 17 U.S. zoos and aquariums with alcid exhibits. Based on information returned by 14 of these facilities, the following table (Table 4) provides a summary of water quality parameters, ranges, and averages.

Table 4. Water quality parameters based on 2004 Alcid Exhibit Design Survey sent to AZA institutions, and recommendations.

Water quality	Range	Average	Recommended range
Water temperature (indoor & outdoor)	0.56–15.56 °C (33–60 °F)	10 °C (50 °F)	10–15.56 °C (50–60 °F)
Water temperature (open systems)	0.56–13.89 °C (33–57 °F)	-	-
Salinity (open systems)	26–35 ppt	-	30–34 ppt
Water turnover rate (once every…)	20–300 mins	1.5 hours	-
pH	-	-	7.2–8.2
Oxidants (mg/L)	-	-	0.0
Coliforms (100/ml)	-	-	<1000
Nitrates/NH_3 (mg/L)	-	-	<0.1

Facilities drawing water from the ocean tend to have lower water temperatures, at least during the winter months. Outdoor alcid exhibits have used either semi-open (with 75% re-circulation) or open systems utilizing salt water from natural sources (Zombeck & Carlson, 2004). Generally, the higher the turnover rate of water within pools, the more beneficial it is to the system. The minimum recommended pool depth is 2.13 m (7 ft) because this depth will allow the birds to dive and forage for food, explore the pool, and get some exercise. Numerous and varying water currents should be used within the pools to avoid pockets of slow moving or stagnant water and for bird enrichment and exercise purposes.

Water quality can be assessed for bacteria levels, and chemical analysis should be conducted for nitrogen levels. Pertinent water parameters include testing for pH and coliforms. Records should be kept according to the standards of AZA and institutions' internal Operations or Facilities departments. Where fecal load could be an issue, water quality should be tested once a week and the results of the tests should be recorded. Trends and changes should be analyzed and life support crews should be informed if there are any concerns or anticipated needs for changes.

A sterilized container should be used for water quality testing and the sample refrigerated immediately afterwards. While more research is needed in the maximum coliform count, it is recommended that there should not be a coliform count over 1,000 MPN (most probable number) per 100 mL (3.38 fl oz) of water. If a pool fails to meet water quality recommendations, appropriate life support staff should be informed immediately, and measures to drain and clean the pool should be taken and/or filtration, cleaning, and water turnover should be increased. If water from the pools is recirculated through filters, sand and gravel filters with ozone and/or UV sterilization treatments are recommended. Water exchange rates should be frequent enough so that water quality test results are within acceptable standards, yet there is a need for further research to be conducted on water exchange rates for alcids.

Ozone content, pH, coliforms, free and combined chlorine, alkalinity, total chlorides, salinity, lead, and ammonia are all tests that have been conducted at various alcid exhibits throughout the U.S. It is recommended that periodic testing be done at least for pH and coliforms. In those alcid exhibits where fish are also displayed, the fish may serve as sentinels for poor water quality or facility failures (e.g., power outage, pump failure, etc.) before the birds are affected.

Air Quality: In indoor exhibits, air handling units and filters should be properly maintained to provide adequate ventilation as well as minimize or prevent fungal sporulation within the exhibit and the air handling system itself. Alcids are highly susceptible to air-borne fungal infections, such as aspergillosis. *Aspergillus fumigatus* spores range in size from 2.5–3 microns, with other *Aspergillus sp.* spores as large as 10 microns. In order to remove them from the air, a filter should remove particles of the smallest size. If possible sources of *Aspergillus* are external to the exhibit, then consideration should be given to reducing fresh air intake and providing a high quality filter on the incoming airline as well as on the re-circulation line (Beall et al., 2005).

Air should be tested for fungal spores about every six months. Air quality can be tested by means volumetric air sampling.. All air quality test results and measures should be recorded. See Appendix I for a sample air testing protocol.

If fungal problems arise, a good quality HEPA filter system may reduce infection rates. Indoor air spore numbers should be lower than outdoor or outdoor-exposed areas. Environmentally controlled spaces are expected to have a greater than 90% reduction in air spores. Any *Aspergillus sp.* should be noted and addressed if need be, and any air quality result over 60 cfu/m^3 should be taken very seriously (S. Mansergh, personal communication, 2011). If high counts are found, notify veterinary staff immediately. They may choose to put the birds on an anti-fungal medication. In addition, consider removing the birds from that area, try to filter the air to help remove the *Aspergillus sp.*, and/or modify the exhibit to decrease the counts.

Air turnover rates range from 1–14.6 air changes per hour, with an average turnover rate of 8 changes per hour, based on data from four alcid facilities. Air turnovers in the range of 15 air changes per hour have been recommended for laboratory animals (Lane-Petter, 1976). These parameters may be acceptable for alcids; however, the specific design of an air-system needs to balance the tradeoffs between filter efficiency, air flow, fresh air exchange, and temperature regulation capacity (Beall et al., 2005). More research is needed in this area. Air turnover rates should be maximized in indoor facilities to maintain air quality while still maintaining recommended air temperatures.

Many alcids exhibits are under positive air pressure, which allows air to be forced out, instead of into, the exhibit when a door is open. Doors should be well sealed to prevent air exchanges with outside areas. Pre-filters (like Purolater® pleated filters) can be used to remove the larger airborne particles, and high-efficiency particulate air HEPA filters have been used successfully to remove small spores. Manufacturer recommendations for filter replacements should be closely followed to achieve optimum levels of operation.

In outdoor exhibits, there are no standards for air quality, but quality can be improved by natural air flows/currents either through exhibit design, care in arranging exhibit furniture, or by the use of screens and/or air vents.

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. Alcids can become habituated to the day-to-day sounds of zoo and aquarium operations like leaf blowers, trucks, and large crowds of visitors. Many different species of alcids are often seen resting comfortably in close proximity to large groups of visitors. Sounds that are not routine, such as unscheduled maintenance work or construction projects, may be stressful to the birds. In situations where there are unscheduled noises for an extended period of time over several days, animal keepers should discuss what course of action would cause the birds the least amount of stress—either moving the birds or allowing them to become habituated to the work. Another option is to educate maintenance and construction workers on behaviors that indicate that the birds are stressed, such as fleeing, flying/crashing into things, porpoising, etc., so that the workers can alert keepers if they see these behaviors. Keepers can monitor food intake, vocalizations, and activity patterns to determine the stress levels of the birds.

Specific pieces of equipment such as water circulation pumps tend to emit noise at all times and may need to be housed in a soundproofed box. Events that utilize loud music and/or PA systems in close proximity to alcid habitats may not be appropriate, particularly if the music has high/low intermittent noise levels and/or takes place during times when birds normally have a quiet period to rest and/or forage. In general, it is thought that alcids seem to do fine with the intermittent nature of visitors to a habitat. While it is clear that sounds and vibrations have the potential to cause stress, further knowledge is needed to ascertain exactly how birds respond to large noise sources and if this may negatively affect other aspects of their lives.

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 must be well cared for and presented in a manner reflecting modern zoological practices in exhibit design (AZA Accreditation Standard 1.5.1). All animals must be housed in safe enclosures that meet their physical and psychological needs, as well as their social needs. (AZA Accreditation Standards 1.5.2, 1.5.2.1, 1.5.2.2).

AZA Accreditation Standard

(1.5.1) All animals must be well cared for and presented in a manner reflecting modern zoological practices in exhibit design, balancing animals' welfare requirements with aesthetic and educational considerations.

AZA Accreditation Standard

(1.5.2) All animals must be housed in enclosures which are safe for the animals and meet their physical and psychological needs.

AZA Accreditation Standard

(1.5.2.1) All animals must be kept in appropriate groupings which meet their social and welfare needs.

AZA Accreditation Standard

(1.5.2.2) All animals should be provided the opportunity to choose among a variety of conditions within their environment.

Species-appropriate behaviors: Swimming, diving, resting, feeding/foraging, walking, climbing, nesting, copulating, vocalizing, exploring, porpoising, rafting, sunning, bathing, and preening are all species-appropriate behaviors that should be encouraged. There are a variety of ways to encourage those behaviors when it comes to exhibit design.

Enclosure design: If the birds are normally flighted, they need to be flight restrained in an open environment. Care should be taken to design the exhibit space so that all birds can access the exhibit and be able to get out of areas within the exhibit should they feel trapped or threatened by conspecifics. As with most avian species, a double door entry for both the public and keeper entries is recommended.

Horizontal spaces have various purposes for both birds and keepers. These areas are used by the birds for feeding, breeding activities such as courtship and copulation, nest material collection, roosting, preening, exploration, enrichment, and other behaviors such as fighting and chasing. Keepers use these areas to move around the exhibit for the purpose of cleaning and maintenance, feeding, bird observations, and capturing. Consideration should be given to training needs when designing the exhibit. It is recommended to leave enough space for training sessions that may include scales, kennels, or other large objects. Also, it would be helpful to include a space designed such that birds can be herded and their access to the water blocked for ease of capture.

Alcids are colonial birds. They are seen in high densities in the wild. For example, common murres can be seen in the wild with 28–34 individuals per square meter (Gatson & Jones, 1998). However, all of the exhibit birds should be able to comfortably fit on the land surface at one time, without aggression. The birds also need to have additional space to retreat away from aggressive birds or from keepers as they work in the exhibit. The exhibit should be cleaned daily, but cleaning should not disrupt nesting birds. If the land and the water are being cleaned simultaneously, it is recommended to clean these areas on the same side of the exhibit at the same time to give the birds space away from keepers. The provision of neutral roosting areas where birds can congregate away from the breeding sites during the breeding season is also recommended. There isn't a specific formula for space needs; it is dependent on the dynamic of the flock. Common murres are less aggressive with each other and can be kept at higher densities compared to pigeon guillemots and horned puffins, who tend to be more aggressive. At a minimum, six individuals per species are recommended for population size. If building a new exhibit, it is recommended to start with a conservative number and add in more birds slowly, watching carefully for their behaviors, territory, and aggression.

Vertical spaces, which include cliffs and ledges, are used by the birds for courtship behaviors, as an entrance into nest tunnels, for guarding of nest tunnels, nest material collection, roosting, preening, exploration, and other behaviors such as fighting and chasing. Table 5 shows that the rock cliffs in current alcid exhibits vary in height from 1.52–8.53 m (5–28 ft), with an average height of 4.27 m (14 ft). Flight distance required to the water surface (depth of the exhibit) is dependent on the height of the rockwork. The higher the rockwork, the faster the birds will fly/glide down toward the water. The larger alcids are heavy bodied and are not able to quickly turn once in flight. Birds will have a difficult time avoiding the exhibit glass if the exhibit does not have enough depth to meet their needs. The cliffs need to have a gradual slope to the back of the exhibit. This will make the rockwork more accessible (easier to climb) for birds and keepers. For example, a rocky cliff 6.10 m (20 ft) high with a slope of 1.22 m (4 ft) would be a high cliff with a very steep slope, and thus be difficult for keepers to safely access to clean, and may be difficult for the birds to climb. In contrast, a 3.05 m (10 ft) high cliff with a slope of 3.05 m (10 ft) would be easier for both staff and birds to access. The pathways and ledges need to be wide enough for a bird to comfortably pass another bird that is roosting in front of its nest tunnel and for staff to safely use to access all areas of the rockwork to clean it. Hiding spots, however, should be kept to a minimum to prevent seclusion of young or ill birds.

Caves built into exhibit rockwork have been used as burrows by burrow-dwelling alcids; due to the inherent drainage problems and health risks associated with moisture build up in the caves, this is a less desirable alternative. For non-burrowing alcids, such as murres, exhibits will need to have adequate ledge space for breeding. Ledges should be level or slope slightly back towards rockwork to prevent eggs from falling off the ledge, and they should drain properly to avoid water collecting. The minimum ledge depth should be 30.48 cm (12 in.). Murres will nest on communal ledges in close proximity to each other. During courtship and incubation, it is sometimes difficult to differentiate pairs, thus making parentage of abandoned eggs difficult to determine. With some species, such as puffins, adequate numbers (1.5 nests for every pair of birds) of nesting sites are recommended for successful reproduction. It is also important to consider the angle of the nest openings. If a bird is guarding the front of the nesting entrance, it should not be able to see directly in another nest because that can cause aggression. It is recommended to place nest openings in a way that they are not angled at another nest opening. See Chapter 8 for more information on alcid reproduction.

Table 5. Exhibit design parameters based on 2004 Alcid Exhibit Design Survey sent to AZA facilities

Exhibit parameter	Range	Average
Total size	73.15–2393.90 m^2 (240–7854 ft^2)	412.70 m^2 (1354 ft^2)
Exhibit length	5.03–3.05 m (16.5–10 ft)	12.50 m (41 ft)
Exhibit width/depth	2.74–15.24 m (9–50 ft)	6.71 m (22 ft)
Exhibit height	3.51–15.24 m (11.5–50 ft)	6.71 m (22 ft)
Exhibit land area	21.95–228.60 m^2 (72–750 ft^2)	88.09 m^2 (289 ft^2)
Rock cliff height	1.52–8.53 m (5–28 ft)	4.27 m (14 ft)
Pool volume	10410–397468 L (2750–105,000 gallons)	94635 L (25,000 gallons)
Pool depth	0.79–6.40 m (2.6–21 ft)	2.29 m (7.5 ft)
Pool surface area	41.91–438.91 m^2 (137.5–1440 ft^2)	170.08 m^2 (558 ft^2)
Pool length	2.44–21.95 m (8–72 ft)	10.06 m (33 ft)
Pool width	1.68–12.19 m (5.5–40 ft)	5.49 m (18 ft)

Pool size and complexity: These pelagic birds spend a considerable percentage of their time on the water, and, if the water surface area is perceived to be too small or cramped, the birds may not feel comfortable on the water. This discomfort could result in alcids spending an unnatural amount of time on land, which could ultimately lead to long term behavioral and health problems (e.g., bumblefoot). Ideally, a minimum of 75% of the colony should be able to occupy the water at one time. It is important for these birds to maintain a healthy level of exercise which can be achieved by offering variety in water flow and interesting underwater topography to create a diverse aquatic environment. Features that could add interest to the underwater environment include underwater plants and invertebrates, rock outcroppings, feeding chutes, water motion devices, and bubble curtains. More information about how to encourage birds into the water by using behavioral management can be found in Chapter 9.

The minimum recommended pool depth is 2.13 m (7 ft), as this depth will allow the birds to dive and forage for food, explore the pool, and allow the birds to get some exercise. The length and width of the pool are important for the birds to be able to exhibit skimming and porpoising behaviors, which are good exercise for the birds. The width of the pool is also important for giving the birds' adequate surface area to land on when flying/gliding down from the cliffs. A proportion of 1/3 land surface area to 2/3 water surface area has been used successfully at AZA institutions to get the birds to exhibit species-appropriate behaviors.

Ample ramps and/or sloped exit areas are recommended so that the birds can exit the water easily. The ramps should extend down into the water in the event of a drop in water level. Birds normally prefer to take the shortest route to their destination, especially compromised, older, or juvenile birds. In light of this, as many ramp areas as possible should be provided within the exhibit. A minimum of three wide exits from the water spread evenly throughout the length of the exhibit are recommended. Rockwork overhanging the water should not be able to trap diving birds under the water. Inexperienced birds may encounter problems with this and get trapped without air.

Enclosure substrates: Alcid exhibits can be constructed of artificial rockwork made from gunite, resin and cement, fiberglass, or a gunite/shotcrete combination. The rockwork should have an uneven surface because this is a more natural substrate for the birds, and it will help prevent foot problems. The rockwork should also be able to drain well in order to create a dry surface for the birds to stand or roost on. Highly abrasive surfaces can be detrimental to the birds' feet, but both the birds and the keepers need a non-slip surface. It is difficult to describe how much abrasion is too much. It is advisable to visit several alcid exhibits whose birds have little to no problem with bumblefoot to inspect the texture of their rockwork. Additional substrates that have been used in exhibits to give the birds a softer, dry surface are: Nomad™ matting, Solutia™ matting, and/or gravel over a resin/cement surface. It is important to give the birds a variety of different substrates throughout the exhibit so the bird can choose where to stand to help discourage foot problems, like bumblefoot, from forming.

Live plants growing in soil (and other organic material) are not highly recommended for alcid exhibits because they increase the risk of fungal spore proliferation. However, this is less of a concern in outdoor exhibits than it is in indoor exhibits because of increased air circulation outside. If live plants are included in the exhibit, planters with their own separate drainage should be incorporated so that soil drainage does not spread throughout the exhibit. The type of soil used should not include vermiculite or other lightweight particles that could foul the water surface if they should fall in. Planter location should be based on natural vegetation in a wild alcid habitat, so most vegetation should be concentrated on the top of the rock cliffs. As an alternative to built-in planters, plants in containers can be strategically placed during the growing season, and these can be moved out of the exhibit during the winter. Birds should not have access to the soil if at all possible. Larger vegetation, like trees or bushes, could be used as a source of shade or as a windbreak, if needed.

Some nesting material should be provided in the exhibit for the birds to bring into the burrows, as this stimulates nesting behaviors. Materials that can be provided include pine needles that are at least 7.62 cm (3 in.) in length, sheet moss, sphagnum moss, plastic grass, feathers collected during molt, shells, cobblestone, and/or dried grass. It is also recommended to provide a substrate in the bottom of the nest boxes and burrows to prevent eggs from rolling around and becoming damaged. The types of substrates that have been used include cat litter, aquarium sand, sheet moss, or Nomad™ matting.

It is important to disinfect and thoroughly dry all nesting materials before providing them to the birds. With the exception of fresh cat litter, which is already considered clean, all items used as substrates should be disinfected prior to use. Aquarium sand can be disinfected by immersing it in an iodine based disinfectant solution, then rinsing and drying it thoroughly. The sheet moss can be disinfected by freezing it for a minimum of 24 hours. Cat litter should be a "dust free" non-clumping, clay variety, if available, without deodorants or additives.

The same careful consideration regarding exhibit size and complexity and its relationship to the alcids' 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). Sufficient shade must be provided by natural or artificial means when sunlight is likely to cause overheating or discomfort to the animals (AZA Accreditation Standard 10.3.4).

For animals in off-exhibit holding, appropriate substrate should be available in case the birds have an extended stay. Products that have been used successfully with alcids include Nomad™ matting, Solutia™ matting, Dri-dek® tiles, Turtle Tiles™, and indoor/outdoor carpeting. Many facilities put the Nomad™ matting on top of the Dri-dek® or Turtle Tiles™ to give the birds a softer surface. This also allows the water to drain through the Nomad™ matting. There should be enough land space for the birds to roost comfortably and tray feed. If the room

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. AZA housing guidelines outlined in the Animal Care Manuals should be followed.

AZA Accreditation Standard

(10.3.4) When sunlight is likely to cause overheating of or discomfort to the animals, sufficient shade (in addition to shelter structures) must be provided by natural or artificial means to allow all animals kept outdoors to protect themselves from direct sunlight.

is used for hand-rearing small chicks, enough land space should be provided to set up kennels or other rearing structures for the chicks that are too young to have access to the pool. Artificial rockwork is not typically used in holding rooms. However, it is advisable to create some vertical space for the birds to climb as far away from the entrance door as possible. Shelves can be placed on the walls, with ramps leading to the various levels, to give the room a vertical component. Shelves can also be placed over the pool along the walls to give the birds' added space and easy access to the pool.

A pool is an important part of any off-exhibit holding room, and it should be large enough for all birds housed in the room at any given time to enter the water all at once. It should have a sufficient surface area and minimum depth of 0.61 m (2 ft) to allow the birds to be able to swim, dive, bathe, and roost on the water. It should have good surface skimming and appropriate routes into and out of the water. The pool should be located as far away from the entrance door as possible because the birds usually go to the pool when a keeper enters the room. The birds should be able to exit the pool from multiple locations. It is recommended that the land height is the same level to the entrance to the water, but a ramp has been used going up to the water successfully. The holding area should be designed in a way to encourage the birds to swim. It is recommended to try to make the water area as stimulating as possible. Some suggestions are a live fish or invertebrate component, currents, feeding ports, kelp, or other underwater plants. The area should be designed with features that can be changed up easily to keep it fresh and interesting for the birds.

The door to the holding room should have a window so that keepers can observe the birds without entering the room. The room should also be easy to clean and disinfect. Drains should be strategically placed on the floor to reduce runoff into the pool when cleaning the land surface area. The air temperature should be within 5 °F of the exhibit holding as to not make a dramatic change for the birds. The air and water quality should be similar to exhibit standards.

Isolation can be a very stressful situation for any bird, and it is important to make isolated birds as comfortable as possible during this period. Providing the security of a burrow (e.g., a kennel, cardboard box, or hollow rock) is recommended, and offering favorite food items can also be considered. The bottom part of a Sky Kennel® turned upside down works well as a burrow for most species. If the need for isolation is not related to an infectious agent, adding another bird for company should be considered as this might minimize stress to the compromised bird. However, this may also cause undue stress to the healthy bird and therefore should be considered carefully. The utilization of mirrors has also been successful as an alternative to a live companion bird.

The ability to isolate a bird temporarily in a dry area can be useful. Birds can be isolated in a large kennel out of sight of zoo visitors, but still within the main exhibit. This makes the bird easily accessible to the aviculturist so that food intake and behavior can be closely monitored. The bird should be made as comfortable as possible, with a mat on the kennel floor and fresh food offered throughout the day. Some birds may become too stressed in the kennel, as indicated by increased pacing or jumping. If this is the case, other arrangements should be made.

The placement of the holding/isolation room should be as close to the exhibit as possible. It is possible that if isolated birds can hear the sounds of the birds on exhibit, that this may reduce the stress of separation from the colony. The risk of disease transmission, however, should be weighed against the social and psychological benefit of cooperative housing.

2.2 Safety and Containment

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).

During the design of an alcid exhibit, consideration should be given to future bird round ups. It would be easiest if the pool area could be surrounded and the birds herded towards an area of rockwork where they can climb out. If this is not the case, it may be necessary to train the birds to target and/or kennel voluntarily.

In walk-through exhibits, care should be given to provide an area out of public view/contact for the birds. Public should be closely monitored to make sure no bird harassment or throwing of foreign objects into the water is occurring. For walk-through alcid aviaries where birds can leave the water and access the public walkways, all entries and exits should have a

AZA Accreditation Standard

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

double door system to prevent collection birds from escaping. During the day, it may be beneficial to have staff monitor the aviary for birds that require assistance returning to the water. After hours, a ramp or alternative way for birds to return to the water portions of the exhibit might need to be offered.

Outdoor alcid exhibits can be very vulnerable to rodent, avian, insect, and miscellaneous mammal pests. These pests are potential disease vectors and can spread a variety of pathogens. In general, preventative measures used by many zoological facilities for outdoor aviaries (Powell, 1992) are also useful for outdoor alcid exhibits. Wild birds, such as feral pigeons, are a potential infectious disease hazard if they come into contact with any part of the exhibit, such as the netting, or if they can fly over and defecate into the exhibit. Some species, such as corvids, are naturally aggressive and may attempt to get into the exhibit, causing undue stress on the birds inside. Gulls have been observed tearing some types of exhibit netting. Methods of reducing the threat from avian pests include:

- Netting with small mesh size to keep small birds from entering the exhibit
- Solid barriers extend from the ground 6 feet high above all perimeter surfaces to help keep rodents from climbing up.
- Strong netting to prevent tearing resulting from birds pecking at or walking across the net
- Hanging netting far above the highest exhibit area possible that collection birds can reach, to keep some distance between aggressive wild birds and collection animals
- Regular checking of netting for tears/rips that might allow a wild bird into the collection
- Avoiding or reducing areas near the exhibit that might attract wild birds to take shelter in or roost
- Canopies to shelter the exhibit from fecal waste of wild birds

Rodents present a threat to bird collections by being disease vectors, killing collection birds, or destroying eggs. The following are some techniques used by outdoor alcid aviaries to reduce the incidence of rodents (Zombeck & Carlson, 2004):

- Use of stainless steel mesh or solid barriers (e.g., glass or walls) along the lower areas of the aviary and for at least 1.83 m (6 ft) above the water level to prevent easy entry to the exhibit
- Bait boxes containing either snap traps or anticoagulants, placed around the perimeter of the exhibit
- Trapping
- Food placement in exhibit that prevents rodents from being able to access it

A variety of mammals have the potential of being predatory hazards. These include, but are not limited to, ermine, raccoons, rats, river otters, mink, large ground squirrels, and cats. Trapping and removal is the usual technique employed to prevent these predators from accessing the exhibit (Zombeck & Carlson, 2004), but local and state regulations should be followed where relevant.

Mosquitoes and other biting insects are potential disease vectors that constitute a risk in outdoor exhibits. A variety of viral pathogens can be transmitted from outside sources through the netting to the birds in the exhibit and within the colony. Examples are West Nile Virus, Avian Malaria, and Avian Pox. Mosquitoes can be kept under control by:

- Eliminating standing water in all areas in and around the exhibit
- Using mosquito dunks (sustained-release mosquito larvicides) in non-exhibit fresh water
- Orientating the exhibit to take advantage of any natural wind currents and/or providing fans
- Encouraging the prevalence of natural predators of flying insects
- Utilizing mosquito netting over the exhibit during mosquito season (C. King, personal communication, 2004)

Animal exhibits and holding areas in all AZA-accredited institutions must be secured to prevent unintentional animal egress (AZA Accreditation Standard 11.3.1). All animal exhibit and holding area air and water inflows and outflows must also be securely protected to prevent animal injury or egress (AZA Accreditation standard 1.5.15). Pest control methods must be administered so there is no threat to the animals, staff, public, and wildlife (AZA Accreditation Standard 2.8.1). Exhibit design must

AZA Accreditation Standard

(11.3.1) All animal exhibits and holding areas must be secured to prevent unintentional animal egress.

AZA Accreditation Standard

(1.5.15) All animal exhibit and holding area air and water inflows and outflows must be securely protected to prevent animal injury or egress.

AZA Accreditation Standard

(2.8.1) Pest control management programs must be administered in such a manner that the animals, paid and unpaid staff, the public, and wildlife are not threatened by the pests, contamination from pests, or the control methods used.

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.

As most alcids are fully flighted, the aviary should be covered with a double door entry system if it is a walk-through aviary that the public can access. As the birds may get out of the water portions of the exhibit and onto the public pathway in this kind of aviary, there should be staff/volunteers monitoring the birds during business hours to provide assistance should birds exit the water and enter the public walkway. Outside of business hours, a ramp or other system may need to be in place to allow birds access back into the non-public sections of the exhibit. Even if the birds are flighted, the public walkways might not provide a long enough runway for them to be able to fly back into the non-public sections.

The exhibit containment materials used may be a risk for alcids and should be evaluated for any possibility of entrapment, entanglement, wing injuries, etc. Glass-fronted exhibits can cause issues with the birds flying/gliding into them from the top of the rockwork. The rockwork should be designed to give the birds plenty of room to land into the water. Netting enclosures could allow for predators and/or their feces to enter the exhibit which could cause disease or other issues. Piano wire could cause beak or wing entrapment or other injuries. Concerning keeper safety, walkways within an exhibit can frequently be slippery and wet, especially after cleaning. Therefore, proper footwear should be worn and careful stepping is advised to help prevent keepers from slipping and falling.

Exhibits in which the visiting public is not intended to have contact with animals must have a barrier of sufficient strength and/or design to deter such contact (AZA Accreditation Standard 11.3.6).

Most alcid exhibits have glass and/or netting to contain birds. In facilities with walk-through aviaries, birds are able to come in close contact with visitors. If this is the case, staff/volunteers should always be at the exhibit during public hours to monitor the public's interaction with the birds.

Care should also be taken when walking in the exhibit. Birds move fast and may walk under foot (especially young, hand-reared birds), so it is important for staff/volunteers to keep an eye on the ground to prevent accidentally injuring birds. When keepers are working within the exhibit, they should be aware of the location of birds as they move fast and may fly directly in front of, over, or into keepers while trying to escape. Also, moving slow allows birds the opportunity to get away from keepers if they so choose. This prevents injuries to birds while they are trying distance themselves from keepers.

If staff will be diving in the exhibit to clean the water, your state's Environmental Protection Agency's legal limits for bacteria counts for human safety must be implemented.

All emergency safety procedures must be clearly written, provided to appropriate paid and unpaid staff, and readily available for reference in the event of an actual emergency (AZA Accreditation Standard 11.2.4). Each institution should have an emergency evacuation protocol in place that addresses evacuation and alternate holding in the case of emergencies. Within that protocol, alternate holding sites should be considered, including other AZA institutions that are close by that could potentially hold birds. There should be enough crates on site to transport all of the animals in the exhibit. In addition, nets should be kept on hand. In situations where life support system fail, air and water sampling should occur to help monitor conditions until transport can be arranged.

AZA Accreditation Standard

(11.3.6) There must be barriers in place (for example, guardrails, fences, walls, etc.) of sufficient strength and/or design to deter public entry into animal exhibits or holding areas, and to deter public contact with animals in all areas where such contact is not intended.

AZA Accreditation Standard

(11.2.4) 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.

AZA Accreditation Standard

(11.6.2) Security personnel, whether employed by 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).

AZA Accreditation Standard

(11.2.5) Live-action emergency drills (functional exercises) must be conducted at least once annually for each of the four basic types of emergency (fire; weather or other environmental emergency appropriate to the region; injury to visitor or paid/unpaid staff; and animal escape). Four separate drills are required. These drills must be recorded and results evaluated for compliance with emergency procedures, efficacy of paid/unpaid staff training, aspects of the emergency response that are deemed adequate are reinforced, and those requiring improvement are identified and modified. (See 11.5.2 and 11.7.4 for other required drills).

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).

An emergency response procedure should be in place, and staff should be trained for bird escapes. Practice drills should be done annually in case of a bird escape.

Emergency drills must be conducted at least once annually for each basic type of emergency to ensure all staff is aware of emergency procedures and to identify potential problematic areas that may require adjustment. These drills must be recorded and results evaluated for compliance with emergency procedures, efficacy of paid/unpaid staff training, aspects of the emergency response that are deemed adequate are reinforced, and those requiring improvement are identified and modified (AZA Accreditation Standard 11.2.5). AZA-accredited institutions must have a communication system that can be quickly accessed in case of an emergency (AZA Accreditation Standard 11.2.6). A paid staff member or a committee must be designated as responsible for ensuring that all required emergency drills are conducted, recorded, and evaluated in accordance with AZA accreditation standards (AZA Accreditation Standard 11.2.0).

Security personnel should be trained to call the appropriate staff members via an established phone tree should an emergency arise.

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 11.2.7).

AZA-accredited institutions that care for potentially dangerous animals must have appropriate safety procedures in place to prevent attacks and injuries by these animals (AZA Accreditation Standards 11.5.2 and 11.5.3).

Care should be taken during nest inspections as incubating birds may bite in an effort to protect their eggs. Alcids that are heavily imprinted could be dangerous to the public in walk-through aviaries because they will approach strangers. Then, the general tendency is to reach out to the approaching bird which makes the bite risk high. These birds will bite, so keepers need to be careful when hand feeding them and when restraining them. Sometimes gloves are recommended to be worn when handling alcids. When sticking a hand in to retrieve an egg from an incubating parent, some birds may become aggressive and try to bite.

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

AZA Accreditation Standard

(11.2.6) The institution must have a communication system that can be quickly accessed in case of an emergency.

AZA Accreditation Standard

(11.2.0) A paid staff member or a committee must be designated as responsible for ensuring that all required emergency drills are conducted, recorded, and evaluated in accordance with AZA accreditation standards (see 11.2.5, 11.5.2, and 11.7.4).

AZA Accreditation Standard

(11.2.7) A written protocol should be developed involving local police or other emergency agencies and include response times to emergencies.

AZA Accreditation Standard

(11.5.2) All areas housing venomous animals must be equipped with appropriate alarm systems, and/or have protocols and procedures in place which will notify paid and unpaid staff in the event of a bite injury, attack, or escape from the enclosure. These systems and/or protocols and procedures must be routinely checked to insure proper functionality, and periodic drills (at minimum annually) must be conducted to insure that appropriate paid and unpaid staff are notified (See 11.2.5 and 11.7.4 for other required drills).

AZA Accreditation Standard

(11.5.3) Institutions maintaining potentially dangerous animals 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.

duly noted whenever such are identified (AZA Accreditation Standards 11.5.3 and 11.5.2).

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). Antivenin must be readily available and all paid and unpaid staff working in areas containing venomous animals should know the antivenin's location (AZA Accreditation Standard 11.5.1).

AZA Accreditation Standard

(11.5.1) Institutions maintaining venomous animals must have appropriate antivenin readily available, and its location must be known by all paid and unpaid staff working in those areas. An individual must be responsible for inventory, disposal/replacement, and storage of antivenin.

Chapter 3. Records

3.1 Definitions

In the zoo and aquarium world, animal records are defined as "data, regardless of physical form or medium, providing information about individual animals, samples or parts thereof, or groups of animals" (AZA Accreditation Standard 1.4.4). Most animals in zoo and aquarium collections are recorded as (referred to as) individuals, though some types of animals are recorded as (referred to as) groups or colonies of animals, particularly with invertebrates and in aquariums (see Appendix B for definitions and Recordkeeping Guidelines for Group Accessions). The decision about how to record its animals usually resides with each institution, but in certain cases, the AZA Animal Program Leader (i.e., TAG Chair, SSP Coordinator, or Studbook Keeper) may request that animals be recorded in a certain manner, whether as individuals or as groups. For alcids, it is preferred if they are recorded as individuals.
Species are typically recorded as individuals and tracked that way in SSP programs.

3.2 Types of Records

There are many types of records kept for the animals in our care, including but not limited to, veterinary, husbandry, behavior, enrichment, nutrition and collection management. These types of records may be kept as separate records as logs in separate locations or as part of the collection records and some may be required by regulatory agencies (e.g., primate enrichment records) or per AZA Accreditation Standards (e.g., emergency drill records).

Recordkeeping is an important element of animal care and ensures that information about individual animals or groups of animals is always available. The institution must show evidence of having a zoological records management program for managing animal records, veterinary records, and other relevant information (AZA Accreditation Standard 1.4.0). These records contain important information about an individual animal or group of animals, including but not limited to taxonomic name, transaction history, parentage, identifiers, sex, weights, enclosure locations and moves, and reproductive status (see Appendix C for Guidelines for Creating and Sharing Animal and Collection Records).

Animal transaction confirmation documents (and breeding loan agreements, where applicable) should be maintained for all animals entering the collection by means other than birth or leaving the collection by means other than death. This applies not only to live animals, but to living and non-living biomaterials (other than samples for health testing) derived from those animals. These documents contain the terms and conditions of transactions, should be signed by both parties to the transaction (except invoices), and kept at the institutions as proof of legal possession or ownership and compliance with applicable laws.

Each bird is typically individually identified with bands. Each bird should have its own medical record. Many institutions use ZIMS or Tracks although other systems are also used. See 3.4 Identification for more information. Institutions should keep continuous records of animal diet, housing, reproductive history, weight, behavior, and medical history. Data from any veterinary exam should be entered into the institutional record keeping system. This should include history, anesthesia if used, physical exam findings, blood work results and interpretations, radiographic findings, fecal parasite check results, and any other diagnostics performed. Any medical problems and medications and treatments prescribed should also be recorded. In the event of death, necropsy findings should also be included. A designated paid staff member must be responsible for maintaining the 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 current (AZA

AZA Accreditation Standard

(1.4.0) The institution must show evidence of having a zoological records management program for managing animal records, veterinary records, and other relevant information.

AZA Accreditation Standard

(1.4.6) A paid 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 paid and unpaid animal care staff members apprised of relevant laws and regulations regarding the institution's animals.

AZA Accreditation Standard

(1.4.4) Animal records, whether in electronic or paper form, must be duplicated and stored in a separate location. Animal records are defined as data, regardless of physical form or medium, providing information about individual animals, or samples or parts thereof, or groups of animals.

AZA Accreditation Standard

(1.4.7) Animal and veterinary records must be kept current.

Accreditation Standard 1.4.7). Complete and up-to-date animal records must be duplicated and stored at a separate location (AZA Accreditation Standard 1.4.4) and at least one set of historical records safely stored and protected (AZA Accreditation Standard 1.4.5).

AZA member institutions must inventory their alcid population at least annually and document all alcid acquisitions, transfers, euthanasias, releases, and reintroductions (AZA Accreditation Standard 1.4.1). All alcids 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). All AZA-accredited institutions must abide by the AZA Policy on Responsible Population Management (Appendix D) and the long-term welfare of animals should be considered in all acquisition, transfer, and transition decisions.

3.3 Permit Considerations

Alcids are regulated by federal and/or state governments. Therefore, possession and/or specific activities involving these species usually require a permit(s) issued by the regulating agency, granting permission for possession and/or the specific activities. Depending on the agency involved, the application and approval process may take a few days to many months. These permits must be received by the applicant before the proposed possession or activity can occur.

These birds can fall under the Federal Migratory Bird Act. Most species fall under the 50 CFR 21.12. If the birds are from the wild, special permits may be required. It is recommended to always check with your local government representative as well as institution's regional USFWS Migratory Bird Office.

AZA Accreditation Standard

(1.4.5) At least one set of the institution's historical animal and veterinary records must be stored and protected. Those records should include permits, titles, declaration forms, and other pertinent information.

AZA Accreditation Standard

(1.4.1) An animal inventory must be compiled at least once a year and include data regarding acquisition, transfer, euthanasia, release, and reintroduction.

AZA Accreditation Standard

(1.4.2) All species owned by the institution must be listed on the inventory, including those animals on loan to and from the institution.

AZA Accreditation Standard

(1.4.3) Animals must be identifiable, whenever practical, and have corresponding ID numbers. For animals maintained in colonies/groups or other animals not considered readily identifiable, the institution must provide a statement explaining how record keeping is maintained.

3.4 Identification

Ensuring that alcids are identifiable through various means increases the ability to care for individuals more effectively. All animals held at AZA facilities must be individually identifiable whenever practical, and have corresponding identification (ID) numbers. For animals maintained in colonies or groups, or other animals not considered readily identifiable, institutions must have a procedure for identification of and recording information about these groups or colonies. (AZA Accreditation Standard 1.4.3). These IDs should be included in all documents in which a bird is referenced. Types of identifiers include:

<u>Physical identifier</u>: These include, but are not limited to leg bands and microchips/transponder. Permanent physical identifiers are often required when a species is regulated by a government agency and to distinguish separate animals in studbooks. Wing bands are not considered appropriate.

<u>Intangible identifiers: T</u>hese include, but are not limited to, institutional accession number, house name, public name, studbook number, and ZIMS Global Accession Number.

For most zoos and aquariums, the easiest way to identify individual birds is with leg bands. Microchips are also an option that some institutions are now using. For alcids, two different types of bands are primarily used: plastic (bands and cable ties) or metal. Plastic or nylon bands and cable ties come in a variety of sizes and colors. The most popular is the plastic cable tie because of its ease of use. Numeric values can be ascribed to each color so each bird can have a number to facilitate record keeping. Cable ties with different color variations can be affixed around a single tie, which is then zipped around a bird's leg. Because plastic can become brittle when exposed to water, cable ties have a tendency to break and require periodic replacement. The locking mechanism for the cable tie should be secured with super glue or filed down to avoid cinching up and cutting off the flow of blood to the foot.

Metal bands have durability that plastic bands lack. The disadvantage of the metallic bands is that the bird should be restrained in order for the band to be read for identification. Great care should be taken when attaching metal bands. Extreme caution is recommended when using pliers to clamp a split metal band around delicate bones. Because some alcids walk using their whole foot, aluminum bands can have their numbers wear and become unreadable.

Bands are attached above the toes, around the tarsometatarsus. Bands should be secure enough so they do not slip over the toes or above the ankle. Allowing the band the ability to slide up and down the tarsometatarsus avoids constriction of blood vessels and pressure necrosis of the tissue. Bands that are not properly closed can get caught on foliage or other projections, and may cause injury to the leg.

Bands should be placed on the birds when they are fully grown, a few days away from typical fledge age for that species. Many institutions band males on the right leg and females on the left to differentiate between the sexes. In order to identify a bird in the event of band loss, many zoos and aquariums will band with cable ties on one leg and a metal band on the other. See Figure 4 for sample band placements.

Figure 4. Sample band placements
Photos courtesy of Randy Wilder

Chapter 4. Transport

4.1 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). All temporary, seasonal, and traveling live animal exhibits must meet the same accreditation standards as the institution's permanent resident animals, with foremost attention to animal welfare considerations (AZA Accreditation Standard 1.5.10). Safe animal transport requires the use of appropriate conveyance and equipment that is in good working order. Include copies of appropriate permits and authorizations in transport documentation. If the animal is not owned by the shipping institution, permission is to be obtained from the owner well in advance of the move.

One must check with the following for specific regulations: the State Veterinarian of the destination state (and if shipped by ground, the states through which the bird travels); the regional US Fish and Wildlife Service office(s) of the institutions involved; and the state wildlife agencies of the sender, the recipient, and states through which the bird travels. Birds imported from other countries may be subject to federal quarantine."'

AZA Accreditation Standard

(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 laws and/or regulations must be adhered to.

AZA Accreditation Standard

(1.5.10) Temporary, seasonal and traveling live animal exhibits, programs, or presentations (regardless of ownership or contractual arrangements) must be maintained at the same level of care as the institution's permanent resident animals, with foremost attention to animal welfare considerations, both onsite and at the location where the animals are permanently housed.

The equipment should provide for the adequate containment, life support, comfort, temperature control, food/water, and safety of the animal(s). There are two recommended shipping methods for alcids that are dependent on the species and the distance the alcid needs to be transported: airlines and refrigerated trucks. When shipping alcids by air, all International Air Transport Association (IATA) guidelines for alcids should be met. IATA regulations can be found at www.iata.org.

IATA regulations require that the transport container allows the bird being transported to stand fully erect without touching the roof and sides of the container. As with the kori bustard, close confinement seems to calm most birds and reduce struggling (Hallager, Maslanka & Baily, 2009). There are different methods that can be used to transport alcids that are dependent on the individual species, the animals' health, and their overall temperament.

Large, sturdy peg-board shipping crates (made in-house) have been successful for the transport of alcids. For example, for transport of smaller species such as auklets or horned puffins, each crate can contain ten compartments (20.32 cm x 25.40 cm x 30.48 cm/8 in. x 10 in. x 12 in.) to house individual birds, with divisions made by two sheets of pegboard creating a hollow wall wide enough to put in a frozen ice pack if needed (see Figure 5).

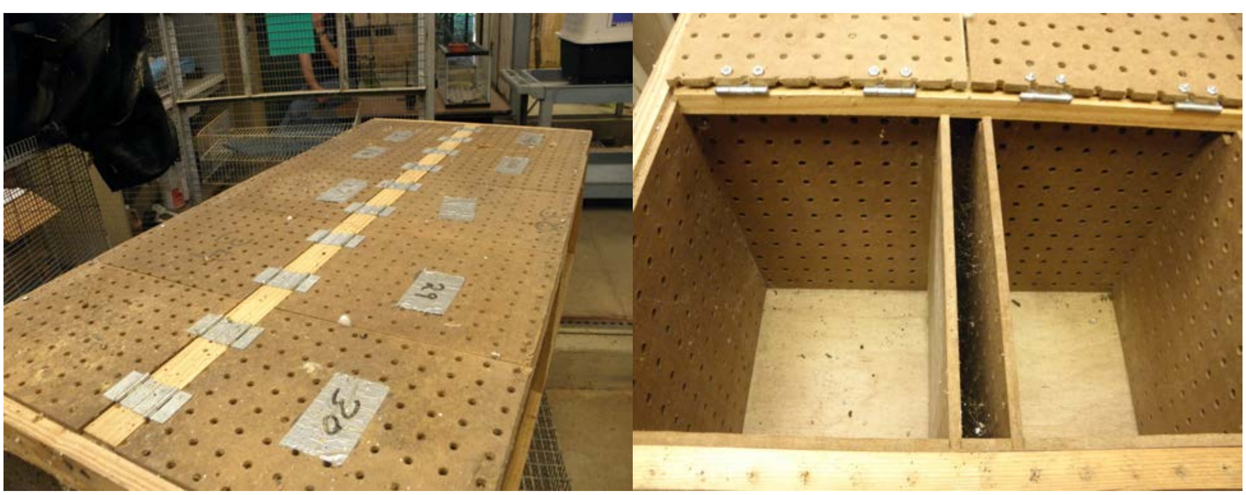

Figure 5. Example of a shipping crate and compartments
Photos courtesy of Debbie Zombeck

Plastic ‘Vari Kennels’ can also be used to transport alcids. . Depending on the species, small (53 cm x 41 cm x 38 cm/21 in. x 16 in. x 15 in.) to medium (71 cm x 52 cm x 54.5 cm/28 in. x 20.5 in. x 21.5 in.) sizes of ‘Vari Kennels’ have been used. See Figure 6 for an example.

Figure 6. Example of a plastic transport kennel
Photos courtesy of CJ McCarty

Another institution uses 10-inch diameter irrigation tubes, with perforations for air circulation. Up to 5 of these tubes will fit inside a medium sized Sky Kennel® (see Figure 7). Small pieces of duct tape affixed to the outside of each tube will prevent them from rotating during shipment. The tubes are cut to fit snugly within the kennel and covered on each end with shade cloth for privacy. These tubes can be kept cool, if necessary, with the addition of ice packs firmly fixed in place outside of the tub. However, this addition will affect tube arrangement, eliminating stacking. These tubes should have substrate for fecal absorption. These tubes provide a snug, dark environment for the birds’ security. These tubes can only be used for smaller species such as the puffins as they must be able to stand up without touching per the IATA recommendations. Small ramekin food containers can be taped to the entrance of the tubes before sealing, and filled with water and fish, to meet IATA requirements.

Figures 7. Irrigation tubes inside of plastic transport kennel
Photos courtesy of Alex Weier.

In general, soft, absorbent, and non-slippery substrate materials should be used. Examples of suitable options are Nomad™ matting or a trampoline made of suspended soft mesh that has been constructed to cover the base of the kennel. The mesh should be a material that allows for feces to fall through so that the bird does not get soiled feathers. Under the trampoline, an absorbent pad should be placed to absorb any feces and spilled drinking water (see Figure 8). This pad should be changed every eight hours, or as flight restraints allow. As previously discussed, some AZA institutions have also used pine needles as substrate successfully.

Figure 8. Example of mesh transport kennel base
Photos courtesy of CJ McCarty

The standard ventilation openings on the kennel should be covered by a dark shade cloth that has mesh with holes approximately 1–2 mm (0.04–0.08 in.) in diameter to prevent entry of biting insects and to partially block the bird from being exposed to too much activity/movement around their transport container. The kennel door should be secured closed with at least two plastic tie wraps on each side of the door, one set to secure the door shut and one set to secure the door to the crate. Small food and water bowls should be fastened to the inside of the kennel so they do not tip over to comply with IATA regulations. Food should be chilled and on ice and should be changed every 12 hours or as flight restraints allow. Table 6 contains all items that should accompany an alcid transport depending on transport length.

Table 6. Necessary equipment needed for alcid transport, either short or long in duration.

Short Trips (less than 12 hours)	**Long trips (more than 12 hours)***
Kennel	Hydration syringe and fluids
Appropriate bottom substrate	Spare tie wraps for crates
Food bowls	Cutters to remove tie wraps
Food supply	Disposable gloves to handle the birds
Clean water	Paper towels
Copies of permits and health certificate	Towel for restraint, if needed
“This way up” and “Live bird” labels	Medical supplies, such as quick stop
	Spare absorbent pads, trash bags
	Gavage Tube
	Cooler and ice packs
	Fish (kept in cooler)

*This equipment is in addition to all the supplies listed for “short trips”

Alcids need cooler temperatures. Shipping alcids in the winter is optimum, but every precaution should be taken to avoid snow delays. A refrigerated truck is the preferred method of alcid shipment for short drivable trips; this method also allows for closer monitoring of the birds. Alcids have been successfully transported this way at a temperature of 7 °C (45 °F). Providing icepacks within shipping crates and kennels is recommended for air transportation in order to maintain cool temperatures in the shipping containers. Multiple large, clear temperature recommendation labels should be affixed to the kennel so that airport personnel have guidelines within which to work. The recommendations should allow for some flexibility: 4 °C (40 ˚F) to 16 °C (60 ˚F). Multiple large, clear “this way up” and “live bird” labels should be taped or stuck onto the kennel on all sides so that airport personnel know how to handle the kennel.

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 (AZA Accreditation Standard 1.5.11).

On short trips (less than 12 hours) inspection of the kennel and bird is not necessary, and it is not essential for the bird to be accompanied by a keeper. On extended trips (more than 12 hours and/or including multiple flights/overseas transport) it is recommended that a keeper—one is sufficient—accompany the bird so that absorbent pads and food can be changed and the condition of the bird can be evaluated at opportune times between flights. The trained staff member accompanying the bird on the trip should be familiar with that bird’s behavior, signs of stress, and handling protocol.

Bird and kennel inspections would need to be done in an enclosed room at an airport cargo warehouse by a qualified keeper. Since most alcids are fully flighted, access to a closed suitable room for this procedure should be obtained. Necessary precautions should be taken to prevent escape during kennel inspection. These may include placing a small towel over the kennel door before opening it to provide a secondary containment method. On extended trips, fish should be injected with water to help bird stay hydrated. If the bird is refusing fish and needs to be hydrated, the bird can be tube fed fluids by keeper or vet staff. Severe dehydration can result in lack of ability to digest food initially and it can take a few days for the alcids to recover from this, resulting in weight loss.

It is a good idea to design an access door into all shipping containers in case an animal problem or transport delay occurs, or if animals need to be fed. Shipping birds via airlines is a common method for travel because it is the fastest. Direct flights via the shortest route should be scheduled whenever possible. Since alcids are very susceptible to *Aspergillus*, it is recommended to start the birds on a preventative medication, such as Itraconazole, a week before transport as well as a week after transport.

Problems that could arise include delayed flights and/or missed flight connections. Flights should be carefully monitored, and the animal’s location should be tracked. Contact the airline at once if any problems arise. Alcids can also overheat, so care should be given to transport in the correct temperature ranges. In order to reduce the risk to alcids and staff handling birds during transport, the following recommendations are useful:

- Using conveyance and equipment that is in good working order
- Obtaining the most direct flight
- Performing transports when temperatures are the best for species (seasonally and/or daily) in the area where they are coming from, as well as where they are going
- Avoiding transports during breeding season (especially if female could be carrying eggs) or when a bird is molting
- Having an in-house protocol established for shipping
- Confirming previously made flight arrangements 48 hours prior to shipping

4.2 Protocols

Transport protocols should be well defined and clear to all animal care staff. When catching birds for transport, it is best to have the birds in a confined area. Birds should be caught using a net, towel, or catch cage by trained staff only.

It is best to have the birds well fed and watered a maximum of 12 hours before transport to allow the birds time to digest their food and decrease the chance of regurgitation. During transport, the water and food should be provided in spill-proof dishes. This is a cautionary measure in case of flight delays. If transport time is expected to exceed 12 hours, the crate should be fitted with access ports to provide fresh food and water without opening the door of the crate. Soft, absorbent substrate can be used such as Nomad™ matting or pine needles. Optimal temperatures for shipping most alcids are between 4–16 °C (40–60 °F), but care should be given to not put the birds in a dramatic change of temperature from where the birds were being held.

IATA standards require holes for sufficient ventilation. These holes can be covered with burlap or shade cloth to reduce the light, noise, and stress on the birds. This covering should not restrict airflow. Alcids typically will not feed in the dark; therefore some light should be provided during long journeys by ground so that the birds can see and eat their food. Ensuring that refrigerated trucks have internal lights for the birds in an important consideration. Shipping early in the day or at night helps avoid high temperatures during transport and gives the birds enough time to reach the destination. Group size during transport is typically one bird per crate, but can be dependent on species/individual bird. Again, if a transport lasts longer than 12 hours, a trained staff person should accompany the birds to monitor and add food/water as needed. If a staff person is accompanying birds and can safely access the birds in a secure area to provide food/water, the birds may be kept in their kennels for up to 24–48 hours, but this can vary on the species/individual temperament.

Once the kennel has reached its final destination, but before releasing the birds, it is important to make sure that the birds have not been injured in shipment. Once that has been determined, the weight of the crate (still with a bird inside) should be taken and recorded. Once each bird has been released, weigh the crate again to get a baseline weight on the bird without having to restrain it. If a blood draw is scheduled for health screening, this should not be conducted on the first day of quarantine. Always allow the birds to recover from the stress of the travel to avoid accumulative stress and skewed results. If the birds were collected from the wild, they will likely be more stressed than usual. Extra care should be given to minimize human contact as well as maximize group size with other birds or mirrors until they are eating well and appear calm. Desensitization to humans should proceed slowly. For release, the crate door should be opened by trained staff. It is less stressful if each bird can be allowed to exit the crate on its own timetable. Each institution should have its own protocol for post-transport release.

Chapter 5. Social Environment

5.1 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.

The minimum recommended number of individuals for each species of alcid held in a managed group is six, but larger groups are more appropriate. The majority of alcid species are gregarious in the wild, and it is optimal to keep 20 individuals or more (Holland, 2007). The larger the intraspecific group, the more opportunities there are for an even gender distribution and increased mate choice by the birds. Alcids are also stimulated to exhibit courtship behaviors by the presence of one or more birds of the same species displaying. More research is needed to determine social recommendations for alcids used in conservation and education programs, as use of these animals in programs is rather new. For information on how educational program participation may impact social behaviors of alcids, see Chapter 10.

Although realistically this can be very difficult to achieve, it is ideal to have equal numbers of both genders of all alcid species, so as to prevent nest harassment and aggression over mates. Since alcids are monomorphic, individuals will need to be DNA or surgically sexed to allow for a close-to-even male to female sex ratio in a collection. Big deviations in the sex ratio can result in aggression (Holland, 2007). An even sex ratio also discourages the formation of same sex pairs and hybridization between taxa (C. King, personal communication, 2004). It is also ideal to maintain a range of ages within the population, so as to ensure continuation of the population (H. Cline, personal communication, 2004).

Alcids in the wild are divided into three groups of chick development patterns. The most common is the semi-precocial group and this contains most of the managed alcids, including puffins, guillemots, and auklets. Semi-precocial chicks are fed by their parents at the nest site for 27–55 days, and then become independent. The second group is precocial and includes the murrelets. Precocial chicks hatch well-developed and are capable of going to sea within 1–2 days of hatching. They are accompanied by their parents at sea until they are fully grown. The third group is intermediate and includes the murres and razorbills. Parents feed their intermediate chicks for 15–23 days, and then the chick leaves the nest for the sea where the male parent cares for it for the next 4–8 weeks (del Hoyo, Elliot, & Sargatal, 1996). In general, alcids have a low reproductive rate but a high rate of hatching and fledging. It is believed that there is a low rate of recruitment of young into the population in wild populations (del Hoyo, Elliot, & Sargatal, 1996). More research is needed to determine if alcids form typical multigenerational groups in the wild.

Typically, temporary isolation of young birds is not necessary, unless there are behavioral problems like aggression towards the chicks or the chicks are not able to figure out feeding on their own. In one facility, there has been the occasional need to remove from the habitat, chicks that have fledged for a few days because they did not adjust well to feeding on their own in the large pool. Also, some mild aggression from other sub-adults towards the younger chicks was observed. These chicks were held indoors for up to a week, learned to feed well right away, and were then released back into the habitat. They were initially held in a dry tote for a day or two and then given access to swimming water (H. Cline, personal communication, 2004).

5.2 Influence of Others and Conspecifics

Animals cared for by AZA-accredited institutions are often found residing with conspecifics, but may also be found residing with animals of other species.

It is advisable to read over the Charadriiformes Regional Collection Plan for North America, which includes alcids, when deciding on which species to house in your exhibit. In general terms, it is recommended that the number of species in one exhibit is kept low, while maintaining a higher number of individuals within a species.

Most alcid facilities house a puffin species with one to two other alcid species, such as common murres or pigeon guillemots. In exhibits where tufted puffins have been housed with Atlantic puffins, the breeding success of the Atlantic puffins has been low, probably due to the competition for burrow space. Puffins in general have a reputation for behaving aggressively toward other species, as well as toward individuals of the same species, especially at the start of the breeding season. Aggression is always heightened at the beginning of, and throughout, the breeding season. If aggression becomes excessive, it may be necessary to remove the aggressive individual for a short period of time; this is preferable to removing the submissive bird. Success keeping a group of king eiders (*Somateria spectabilis*) with a group of puffins has been reported at one AZA zoo (Tieber, 2013). Hybridization of alcids in human care has been observed between horned and Atlantic puffins, and between tufted puffins and rhinoceros auklets.

Auklet species tend to do better in an exhibit without puffins, as the puffins tend to be more aggressive and harass the auklets (K. Anderson, personal communication, 2005). Also, pigeon guillemots have been reported to be very aggressive to other birds (Greenebaum, personal communication, 2015)

Typical negative interactions between conspecifics may manifest by a more dominant bird pushing a less dominant bird away from food. An additional potentially negative behavior could be an increase in activity, because one bird may be bill sparring with another bird and/or chasing another bird to excess. Indicators of negative interactions include weight loss, poor feather quality, loss of waterproofing, and scrapes on bills. Positive interactions would be acceptance of the other bird during feeding time (i.e., displacement of the bird that is less dominant does not occur). No weight loss and good feather quality and waterproofing are other indicators of positive interactions.

Housing species with different breeding requirements (e.g., burrow nesters and ledge nesters) together can reduce nest competition and aggression. It has been observed that one pair of puffins will occupy more than one burrow during the breeding season, so it is recommended to set up more nest sites than the number of active breeding pairs of birds. A good rule of thumb would be to allow at least one and a half nest sites per breeding pair. There should be multiple access points to the different nests, levels of land, etc. so no single bird can guard the whole area. All of the exhibit birds should be able to comfortably fit on the land surface at one time, without aggression. The birds also need to have additional space to retreat away from aggressive birds or from keepers as they work in the exhibit. The provision of neutral roosting areas is recommended, where birds can congregate away from the breeding sites during the breeding season.

Humans may have an impact on the social behavior of some alcids. Imprinted puffins can be very aggressive towards keepers. Care should be given to minimize imprinting by providing partial parent rearing, if available, and keeping the bird with other individuals of the same species. Imprinting murres seems not to cause the same sort of aggression that can be seen in puffins. Hand-reared murres, once introduced to rest of colony, often adapt well to the colony. For information on how educational program participation may impact social behaviors of alcids, see Chapter 10.

5.3 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.

Much care should be taken when introducing new birds into the flock. A new bird thrust into an established colony may be met with some aggression, especially during breeding season. Introductions of new birds during the height of breeding season are not recommended. It is advisable to introduce new birds into groups during a quiet time of the morning because this allows husbandry staff more time for observation. Releasing the new bird on land, so that the bird can find the easiest route to (and therefore from) the water is advisable.

There are several different methods that are used when introducing new birds into an established colony ranging from “howdy cage” methods, short intervals of contact with the colony, to complete release. No matter what method is used, close supervision is highly recommended. Evaluating aggression, feeding, swimming, and waterproofing can help determine successful integration of new birds into the colony and exhibit. It is also recommended to weigh newly released birds on a regular basis to ensure proper weight maintenance. Extra feeding stations may need to be added during introduction periods to ensure ample food supplies for all the birds and to reduce the likelihood of aggression at feeding sites. Providing additional enrichment is another good way to distract existing colony members so that the new birds have a chance to acclimate. If there are known aggressive birds in the flock, removing them from the exhibit for a few days while the new bird gets a chance to settle in should be considered.

Chapter 6. Nutrition

6.1 Nutritional Requirements

AZA Accreditation Standard

(2.6.2) The institution must follow a written nutrition program that meets the behavioral and nutritional needs of all species, individuals, and colonies/groups in the institution. Animal diets must be of a quality and quantity suitable for each animal's nutritional and psychological needs.

A formal nutrition program is required to meet the nutritional and behavioral needs of all species (AZA Accreditation Standard 2.6.2). Diets should be developed using the recommendations of nutritionists, the AZA Nutrition Scientific Advisory Group (NAG) feeding guidelines: (http://nagonline.net/guidelines-aza-institutions/feeding-guidelines/), 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.

Alcids generally have a varied diet in the wild, though fish and invertebrates are the predominant diet component during much of the year, particularly the summer (del Hoyo, Elliot, & Sargatal, 1996). Less is known about winter diets while birds are out at sea. Wild diets span all the trophic levels between plankton-feeding and near-exclusive fish-eating. Dovekies, Cassin's auklets, and Aethia auklets represent the true plankton feeders. The summer diet of the plankton-feeding species is primarily comprised of marine invertebrates including *Thysanoessa* euphausiids, mysids, hyperiids, gammarids, calenoid copepods, and some larval fish and squid. The parakeet auklet has a more specialized diet comprised of a wider variety of planktonic animals, more often including gelatinous zooplankton (Gaston & Jones, 1998). Rhinoceros auklets, razorbills, puffins, and *Cepphus* guillemots represent more generalized fish and plankton-feeders. The diet of the more generalized fish and plankton-feeding species is comprised predominately of fish, supplemented with euphausiids and other crustaceans, and squid or polychaete worms. The common murre comes closest to being exclusively fish-eating, but the diet of this species may include small quantities of crustaceans seasonally (del Hoyo, Elliot, & Sargatal, 1996).

The digestive system of alcids includes a well-developed proventriculus, a muscular stomach, and relatively short intestine. Only the auklets have a functional crop, although there is some crop-like development of the lower proventriculus in the puffins (Gaston & Jones, 1998). Additionally, auklets have a gular pouch to transport and store food for feeding chicks; the length of the pouch can more than double during breeding season and regresses after chicks fledge (Vermeer, 1981). Recent data describe an additional function: capture of copepod prey in auklets through suction-feeding, achieved through extension of the sub-lingual pouch (Enstipp et al., 2018).The bill morphology and the structure of the mouth lining and tongue of alcids correspond with their diets. Species that feed mainly on fish tend to have narrower, more pointed bills and narrower, more cornified tongues than species that feed primarily on plankton (Gaston & Jones, 1998).

Alcids acquire water and sodium through both food and free water. If the birds are held only in fresh water, salt can be added to their diet to ensure the function of the salt glands is maintained. For hatchlings in freshwater environments, supplementation may be necessary for appropriate development and function. Marine-based foods, properly thawed, should provide adequate dietary sodium.

It is thought that birds generally well-regulate their energy needs, and it is not recommended to limit energy intake unless the animal is unnaturally overweight. An estimation of energy requirements (per individual or colony) can be calculated using the energetics equations outlined in Ellis and Gabrielsen (2001). Allometric equations for seabirds (n=77 spp.) suggest that basal metabolic needs can roughly be calculated by the equations: 1) BMR = 381.8 $m^{0.721}$ where m = body weight in kg, and the calculated value equals kJ day^{-1}. Or 2) BMR = 3.201 $m^{0.719}$, with again value equivalent to kJ day^{-1}, but m = body weight in g. Thus murres (~800 to 1100 g body weight) require ~440 to 620 kJ, puffins and guillemots (~300 to 500 g) need ~300 to 335 kJ, and auklets (~85 to 250 g), 120 to 170 kJ to meet BMR needs daily. While field metabolic rates (FMR) and/or daily energy expenditure (DEE) values have rarely been determined for alcids outside the breeding season – when needs are higher -- , the energetic costs of normal activity for Charadriiformes appear to range from 2.6 to 4X BMR, determined from field studies (Ellis and Gabrielsen, 2001). Thus captive animal maintenance energy, calculated at 2X BMR, should prove adequate (see also Fort et al., 2009; Kitaysky 1999; Obst et al., 1995). Energy intake and increases are known to be associated with molting and breeding; birds will increase body mass prior to molt (Schreiber, 2002). Based on existing field data, energy provision 3 to 4X BMR may be appropriate during breeding periods.

Energy needs of these carnivorous birds will be met primarily through the digestion of fat and protein in the diet. Based on the nutrient density of primary food items (see Appendix J for values from typical food items), energy needs for most species can be met by consumption of approximately 25% of individual body weight in prey daily. On a practical feeding basis, 25–75% of total colony body weight can be fed daily to meet energy needs (Fort et al., 2009; Kitansky, 1999;), minimize competition for food, and ensure minimal wastage. Multiple food types should be offered regularly to mimic natural diet variety and minimize selection on sole ingredients to avoid potential supply issues as well as possible nutrient profile imbalances. Field studies documenting higher intake rates of alternative feedstuffs when preferred capelin was unavailable resulted in similar hatch, survival, and fledging rates of puffins in the north Atlantic (Baillie & Jones, 2003). Further, puffins and auklets show distinct physiological adaptations to reduced feed supplies, with puffins able to lower growth/metabolism by as much as 50% compared to auklets at 25-30% (Kitaysky, 1999); thus acids show multiple feeding and dietary mechanisms for coping with changing nutrient resources.

Although more research is needed into specific nutritional needs of managed alcids birds, nutrient recommendations for other avian piscivores (i.e. penguins) have been developed based on known requirements of domestic poultry and carnivore species, as well as unique needs when consuming a fish-based diet (Table 7). These basal guidelines would likewise apply to alcids until further information in compiled.

Table 7. Proposed minimum energy and nutrient concentrations (DMB) in adult alcid diets* based on requirements of domestic poultry, felids, and inferences from composition of native foodstuffs (Beall et al., 2005).

Nutrient	Minimum Concentration
Gross energy, kcal/g (kJ/g)	4.5 (18.5)
Crude protein, %	35
Fat, %	10
Calcium, %	0.8
Phosphorus, %	0.6
Magnesium, %	0.05
Potassuim, %	0.5
Sodium, %	0.2
Iron, mg/kg	80
Copper, mg/kg	5
Manganese, mg/kg	5
Zinc, mg/kg	50
Selenium, mg/kg	0.2
Vitamin A, IU/kg	3500
Vitamin D, IU/kg	500
Vitamin E, IU/kg	400**
Thiamin, mg/kg	100***

*Other nutrients, such as essential fatty acids, essential amino acids, vitamin K, and the other B-complex vitamins are probably required. Nevertheless, there is no evidence that inadequate concentrations are provided by fish and marine invertebrates. Whether vitamin C can be synthesized by alcid tissues has not been established. Freshly caught fish contain significant concentrations of this vitamin and some destruction undoubtedly occurs during storage. However, signs of vitamin C deficiency in alcids have not been described.

**Although this concentration of vitamin E may exceed the minimum requirement for other species, 400 IU vitamin E/kg of DM provided by the supplementation of 100 IU of vitamin E/kg fresh fish is recommended to compensate for losses during peroxidation of unsaturated fatty acids in fish-based diets.

***Likewise, this concentration of thiamin undoubtedly exceeds the minimum dietary requirement, but about 100–120 mg/kg DM are provided through supplementation of 25–30 mg of thiamin/kg of fresh fish to compensate for destruction by thiaminases in fish-based diets.

Whole fish and many invertebrates fed to alcids contain adequate levels of calcium, thus supplementation of this nutrient should not be necessary under normal conditions. Care should be taken to determine the actual calcium level in the diet (by analyzing the fish), however, as well as to ensure a Ca:P ratio between 1:1 and 2:1 as optimal. Adding carotenoid pigments (i.e. canthaxanthin) to the diet can improve the coloration of alcid species, although targeted research is needed in this area.

Supplements necessary in any fish-based diet, notably vitamin E (100 IU per kg fresh fish) and thiamin (25 mg per kg fresh fish) can be incorporated as pills and/or paste, hidden in fish, and hand-fed to birds to ensure appropriate and complete dosing.

6.2 Diets

The formulation, preparation, and delivery of all diets must be of a quality and quantity suitable to meet the animal's nutritional and psychological needs (AZA Accreditation Standard 2.6.2). Food should be purchased from reliable, sustainable, and well-managed sources. The nutritional analysis of the food should be regularly tested and recorded.

Food should be presented a minimum of twice daily ad libitum. There is no need to limit food intake unless obesity is a concern, and this has rarely been documented in managed alcids. Similar to other managed avian species, a good rule of thumb is to offer slightly more than is consumed. Frequency and quantity of food offered may increase seasonally, particularly pre-molt and during the breeding season. In general, food can be offered in the water to promote swimming (as long as it is consumed rapidly to not foul the water), hand fed to help with training, and/or left in dishes/ice dishes to provide ample opportunities for each individual to feed. It is recommended to use all three methods daily and/or weekly. All alcid species are solitary feeders in the wild (del Hoyo, Elliot & Sargatal, 1996). Some aggression elicited by *ex situ* feeding methods has been reported between dissimilarly sized alcids. To reduce associated aggression, food dishes should be placed in several locations around the enclosure, and should be numerous enough to prevent competition and individuals monopolizing food resources.

Alcids, like other fish eating birds, will demonstrate preferences for specific fish species, particularly small whole fish such as silversides, lake smelt, and small herring. Care should be taken to prevent the consumption of only a single kind of food item in order to maintain a nutritionally balanced diet, as well as to ensure minimal problems should availability become limited. Representative analyses of individual ingredients commonly fed to alcids can be found in Appendix J and information from an AZA survey regarding managed alcid diets can be found in Tables 8 & 9.

Table 8. Variety of food items fed to managed alcids compiled from AZA survey

Whole fish	**Cut fish**	**Invertebrates**
Silversides	Herring	Krill
Lake smelt	Capelin	Squid
Capelin	Salmon (fillets and roe)	Clams
Small herring		Mussels
Salmon smolts		Shrimp
Sand eels/lances		Brine shrimp
		Bloodworms
		Ghost shrimp
		Copepods
		Insects

Table 9. Examples of managed diets for mixed groups of larger alcid species compiled from AZA survey

	Species fed to	**Diet composition**
Alcid Diet 1	Razorbill	80% silversides, 10% capelin or eel, 5% herring, 5% krill
Alcid Diet 2	Atlantic puffin, common murre	40% silversides, 30% cut herring or capelin, 7.5% whole capelin,15% krill, 5% cut clams/squid
Alcid Diet 3	Tufted puffin	36% krill, 27% silversides, 27% capelin, 10% herring
Alcid Diet 4	Pigeon & black guillemots, tufted puffin	33% silversides, 33% herring, 33% krill
Alcid Diet 5	Horned puffin, thick-billed murre, parakeet auklet	30% silversides, 30% krill, 30% capelin or herring, 5% smelt, 3% squid, 2% eel

Feeding habits can be extremely diverse and flexible among the Alcidae, varying seasonally, regionally, and annually. In the wild, alcids obtain their food from the sea by diving from the surface and propelling themselves underwater with their feet and wings in pursuit of prey (del Hoyo, Elliot, & Sargatal, 1996). Alcids are predators and as such, whole food items are recommended for both optimal nutritional composition as well as supporting natural behaviors they may elicit. Body size is related to prey size (Vezina, 1985), and in seabirds body size seems to relate especially to the size of the organism that can be swallowed whole. In alcids, both the maximum and median sizes of prey recorded are rather closely related to body size (Gaston & Jones, 1998). Due to the inconsistency between prey size and items which are commercially available, it is often necessary to cut and/or fillet items to sizes that can be easily swallowed. When adults are feeding chicks, prey size should be appropriate for the life stage of the chick.

For ease of presentation and preparation, removing fins, spines, and tails of fish and the pens of squid can be performed prior to presentation.

Live feed is preferred by all species, but may not be practical in *ex situ* feeding. Puffins prefer whole small fish (especially the sand eels) and many only take herring if chopped. Guillemots will eat whole fish, whereas the smaller species and particularly the auklets will not consume most fish, but rather focus on aquatic invertebrates. Insects, mussels, shrimp, and bloodworms can be provided as enrichment or seasonal items rather than dietary staples. Shrimp and sand eels/lances are commonly fed before and during breeding and chick rearing seasons.

Food preparation must be performed in accordance with all relevant federal, state, or local laws and/or 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 alcids. 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.

AZA Accreditation Standard

(2.6.1) Animal food preparation and storage must meet all applicable laws and/or regulations.

The nutritional value and composition of fish is dependent upon species, stage of growth, and habitat quality, as well as proper post-harvest shipping, storage, and handling. Fish should be stored with an average product temperature range of -18 to -30 °C (-0.4 to -22 °F) (Crissey & Spencer, 1998; Crissey et al., 1998). Fish are available as bulk-frozen or individually-quick-frozen (IQF). IQF fish undergo much more rapid freezing and tend to be more expensive. The shorter the time period taken to completely freeze fish, the better the quality. The thawing process should be carefully controlled because there is great potential for nutrient loss and microbial build up if fish is thawed incorrectly. According to the Nutrition Advisory Group Handbook, the nutritional integrity of the fish is best maintained during thawing when they are placed loosely covered in a refrigerated area and thawed as close in time to the feeding as possible. The fish may be safely thawed under refrigeration at 2–3.5 °C (35.6–38.3 °F). Thawing at room temperature is not advised as it will hasten microbial growth and oxidative tissue damage, adversely affecting quality and palatability.

Fish may be considered acceptable if eyes are clear and not sunken, gills are red, flesh is firm and not broken, and if there is little to no odor. Fish should be packed in thick plastic bags and contained in thick, plastic coated boxes. Boxes should identify species and contain information regarding date of catch and vendor/distributor name and address. Delivery trucks should have correct temperature with frozen conditions maintained from origin to delivery (Crissey et al., 2002).

Freezers where food is stored should have temperature-sensitive alarms to alert staff to problems. Once thawed, food should never be refrozen and, if refrigerated, any unused fish should be discarded within 24 hours (Crissey & Spencer, 1998). If fed outside, depending on temperature, it is important to feed only the amount that will be consumed immediately or while still iced to avoid microbial build-up and nutrient loss (Beall et al., 2005). Any uneaten fish in the water should be removed within two to four hours.

Care should be taken not to include plant décor in the habitat/exhibit that could be poisonous to birds. Follow guidelines in the Zoo Horticulture poisonous plant list, which can be found at http://www.azh.org/. Any plant that is listed anywhere in any guide as being poisonous or potentially poisonous should not be placed in a bird habitat or exhibit. No plant fertilizers or pesticides should ever be used in a bird habitat or exhibit. Live plants that are placed in a habitat should be rinsed first before they go into the habitat to ensure that there are no chemical residues on the plants that could wash off into the habitat or water supply. Specifically, some facilities are known to give seaweed, moss, and pine needles as enrichment/nesting material items.

AZA Accreditation Standard

(2.6.3) The institution must assign at least one paid or unpaid staff member to oversee appropriate browse material for the animals (including aquatic animals).

6.3 Nutritional Evaluations

Neurological disorders (that respond to thiamin supplementation) and white muscle disease (associated with vitamin E deficiency) have been reported in alcids. It is known that fish-based diets necessitate additional thiamin and vitamin E supplementation to avoid such disorders. Following proper fish handling and storage protocols and dietary supplementation regimens should alleviate occurrences of these problems. Cataracts (associated with vitamin A deficiency) have also been reported in alcids, particularly auklets. Based on diet composition data, whole-fish based diets are not deficient in vitamin A. On the contrary, evidence suggests that levels may in fact be excessive. For example, Columbia River smelt fed to alcids contains very high levels of vitamin A (>200,000 IU/kg DM) (Crissey, 1998) that may antagonize the uptake of other fat-soluble vitamins. Even with vitamin E supplementation, excess vitamin A may interfere with nutrient metabolism. More research is needed in this area.

Chapter 7. Veterinary Care

7.1 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 necessary, a consulting/part-time veterinarian must be under contract to make at least twice monthly inspections of the animal collection and to respond to any emergencies (AZA Accreditation Standard 2.1.1). In some instances, because of their size or nature, exceptions may be made to the twice-monthly inspection requirement for certain institutions (e.g., insects only, etc.). 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), available at the AAZV website under "Publications", at http://www.aazv.org/displaycommon.cfm?an=1&subarticlenbr=839 (AZA Accreditation Standard 2.0.1).

Routine health inspections by a veterinarian are recommended, and should include a complete physical exam including body weight, blood collection for complete blood count and biochemical panel if size allows, and radiographs. Fecal parasite screens should be performed twice yearly. It may be appropriate to take a representative group sample where individuals are housed together. The need for annual exams should be weighed with the stress to the colony for capture.

The current Charadriiformes veterinary advisor is:
Dr. Stephanie McCain
Birmingham Zoo
smccain@birminghamzoo.com

Generally, no specific training programs are necessary to work with Charadriiformes, and general avian principles apply. Several book chapters have been written on shorebirds which may provide additional information (Ball, 2003; McCain, 2015; Pokras, 1996).

Protocols for the use and security of drugs used for veterinary purposes must be formally written and available to paid and unpaid 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.

AZA Accreditation Standard

(2.1.1) A full-time staff veterinarian is recommended. In cases where such is not necessary because of the number and/or nature of the animals residing there, a consulting/part-time veterinarian must be under written contract to make at least twice monthly inspections of the animals and to respond as soon as possible to any emergencies.

AZA Accreditation Standard

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

AZA Accreditation Standard

(2.0.1) The institution should adopt the *Guidelines for Zoo and Aquarium Veterinary Medical Programs and Veterinary Hospitals,* and policies developed or supported by the American Association of Zoo Veterinarians (AAZV). The most recent edition of the medical programs and hospitals booklet is available at the AAZV website, under "Publications", at http://www.aazv.org/displaycommon.cfm?an=1&subarticlenbr=839, and can also be obtained in PDF format by contacting AZA staff.

AZA Accreditation Standard

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

Commonly used drugs for Charadriiformes include, but are not limited to, anesthetic gases (isoflurane, sevoflurane), antibiotics (amoxicillin, cephalexin, clindamycin, enrofloxacin, metronidazole, trimethoprim-sulfamethoxazole), antifungals (amphotericin B, fluconazole, itraconazole, nystatin), and parasiticides (fenbendazole, ivermectin). As these drugs are not unique to Charadriiformes, institutions should follow their current protocols regarding storage and administration of these drugs. These drugs are generally safe at routinely prescribed dosages. Human exposure to anesthetic gases should be minimized by using appropriately sized face masks for the bird during induction. Pregnant women should avoid exposure to anesthetic (Cohen, Weldon, & Brown, 1971).

Veterinary 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 accurate animal veterinary record keeping. Health records should include all physical examination findings (including body weights), fecal examinations, blood work, radiograph interpretations, and other ancillary tests. Many institutions use ZIMS for record keeping, but other systems are also used. Institutions should keep continuous records of animal diet, housing, reproductive history, weight, behavior, molting, and medical history. Data from any veterinary exam should be entered into the institutional record keeping system. This should include history, anesthesia if used, physical exam findings, blood work results and interpretations, radiographic findings, fecal parasite check results, and any other diagnostics performed. Any medical problems and prescribed medications and treatments should also be recorded. In the event of death, necropsy findings should be included.

7.2 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.

All alcids should receive a pre-shipment examination by a veterinarian prior to transport. This exam should include a complete physical exam including body weight, confirmation or placement of individual identification method, blood collection if size allows (for complete blood count, biochemical panel, and +/- protein electrophoresis), radiographs, fecal culture, and fecal parasite screen. See Appendix K for reference values for hematological and serum biochemistry parameters of selected species.

Due to their susceptibility to aspergillosis, many institutions prophylactically treat alcids with antifungals (e.g., itraconazole 5–10 mg/kg SID to BID) beginning three to ten days prior to shipment, and continuing two to three weeks into quarantine at the receiving institution. Inappetance may be seen at higher dosages of antifungal therapy, and doses may need to be adjusted accordingly.

The following is a list of methods that have been used for clinical evaluation in alcids:

- Regular weights (see Table 11 in Chapter 7.4)
- Body condition by palpation of the pectoral muscles and keel as well as evaluation of the thoracic inlet (Thin birds will often have atrophied muscles, a prominent keel, and a cavitating thoracic inlet)
- Bill measurements
- Feather quality
- Blood draws checking CBC and biochemical profiles
- Fecal testing for parasitic ova

7.3 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. Quarantine duration should be assessed and determined by the pathogen risk and best practice for animal welfare (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 E). All quarantine procedures should be supervised by a veterinarian, formally written and available to paid and unpaid 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 recommendations have precedence.

AZA Accreditation Standard

(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. Quarantine duration should be assessed and determined by the pathogen risk and best practice for animal welfare.

AZA Accreditation Standard

(2.7.3) Quarantine, hospital, and isolation areas should be in compliance with standards/guidelines contained within the *Guidelines for Zoo and Aquarium Veterinary Medical Programs and Veterinary Hospitals* developed by the American Association of Zoo Veterinarians (AAZV), which can be obtained at: http://www.aazv.org/displaycommon.cfm?an=1&subarticlenbr=839.

In any situation, when a bird is isolated in quarantine or holding, it is important to monitor the health and behavior of the animal carefully. Many alcids may not eat well when housed alone. A mirror should be provided and may help decrease stress. Food intake should be monitored by counting or weighing food items. Birds should also be weighed often, as loss of weight is a good indicator of declining health. Detailed records of behavior and behavioral changes should be kept. Especially for highly gregarious birds, the less time spent in isolation the better; however, most places use a 30-day quarantine period at minimum. If birds are brought in from another country, they will have to go through a complete USDA 30-day quarantine period.

Any type of quarantine facility should be separate from the main exhibit and have separate air and water systems. It is advisable to have two separate facilities for quarantine and holding purposes. Holding room space is necessary for housing sick birds, problem birds (e.g., during short term timeouts), hand-rearing alcid chicks, and to house surplus birds short-term. If this is not feasible, the holding space can serve as quarantine space if the air supply and water supply are on separate systems from the alcid exhibit. In either case, lighting and temperature should be adjustable to replicate the exhibit parameters. If quarantine facilities are not available, it is preferable to physically separate new birds for the duration of quarantine. Quarantine exams should still take place.

AZA Accreditation Standard

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

Quarantine and hospitalization should take place in appropriate sized enclosures that allow for inclusion of a shallow pool. A sandy shore or soft soil is desirable and allows normal feeding behavior. The pool should be large enough for all birds housed in the room at any given time to enter the water all at once. The pool should be located as far away from the entrance door as possible, as the birds usually rush to the pool when a keeper enters the room.

Housing for animals should be easily disinfected and allow birds to be maintained in their preferred environmental parameters. For those birds with limited mobility or bandages, pool access may need to be initially restricted until the bird is more recuperated. Substrate choices should minimize trauma to the feet that could predispose a bird to pododermatitis (bumblefoot). Knotless net bottom caging has been used in some rehabilitation situations to minimize hock and keel pressure sores, reduce fecal contamination, and allow drying of wet birds.

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. 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 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.

AZA Accreditation Standard

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

Zoonotic diseases are not a major concern in Alcids, however *Salmonella* spp., *Erysipelothrix* spp., *Campylobacter* spp., and *Yersinia* spp. have been reported in charadriiformes and have zoonotic potential (Ball, 2003). *Mycobacterium avium* infection is uncommon, but it should be considered with any radiographic evidence of bony lesions, particularly when other diagnostics support an infectious cause (Ball, 2003). Good hygiene is important in limiting the spread of disease. Use of gloves when handling infected birds or their feces can help prevent disease transmission to humans.

Ideally a dedicated staff member that does not care for other birds in the collection should care for alcids that are in quarantine or sick. Alternatively, staff should change clothes and shoes (or wear shoe covers) before and after entering quarantine. There should also be a disinfectant foot bath to minimize carrying potential disease into or out of these areas. Equipment used to feed and care for sick birds or those in quarantine should be used only with these animals. If this is not possible, then all items should be appropriately disinfected as designated by the veterinarian supervising quarantine, before use with resident animals.

Standard cleaners and disinfectants can be utilized in most cases for cleaning equipment, enclosures, food containers, and enrichment items. Food containers should be cleaned daily with dish soap and warm water. General facility cleaners and disinfectant, such as ProVet Logic®, can be used for enclosures, equipment, and enrichment. Enclosures should be at least spot cleaned daily, with more thorough cleaning and disinfecting weekly while in quarantine. Bleach can be irritating to the respiratory tract of birds and should only be used in well ventilated spaces.

Quarantine should last a minimum of 30 days (unless otherwise directed by the staff veterinarian). The recommended quarantine duration for alcids is 30 days (unless otherwise directed by the staff veterinarian). If additional birds are introduced into the same quarantine area, the minimum quarantine period should begin over again.

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 E). 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.

During quarantine, weekly weights are recommended for species that are tolerant of handling. At minimum, alcids should be weighed on entry and at the end of quarantine. Routine quarantine testing generally includes physical examination, blood work (CBC, chemistry, PEP), and fecal samples (2–3 samples collected each week in individuals, or weekly group samples with larger colonies). Cloacal cultures and fecal gram stains can also be a part of the minimum avian diagnostic database. Aspergillosis serological testing may be performed, but titers can be difficult to interpret or fail to show evidence of subclinical disease. It is recommended that the use of a prophylactic antifungal drug such as itraconazole be administered throughout the duration of the quarantine period per the institution's veterinary advice. Radiography can be a useful diagnostic screening modality for quarantined birds; however, the potential risks associated with restraint or anesthesia should be weighed with the diagnostic benefit.

Aspergillosis is one of the most common causes of mortality in alcid exhibits. The most common pathogen, *Aspergillus fumigatus*, is ubiquitous within soil and many substrates found within indoor and outdoor exhibits, as well as air handlers and filters. Most immunocompetent birds have been exposed to *Aspergillus* at some time in their life without developing illness. When the immune system becomes stressed, however, birds are more susceptible to aspergillosis, particularly in environments where the fungal spore load is high. The fungus is generally acquired through the respiratory tract, causing lesions in the air sacs, lungs, and even the central nervous system and eyes. Significant changes with animal management (e.g., newly introduced birds and exhibit construction) and inherent social or physiological stressors such as breeding, molting, and interspecies competition for territory and food are major factors that enhance the birds' susceptibility to fungal disease. Poor air turnover, excessive humidity, and certain types of substrates can increase fungal growth and sporulation within the exhibit.

Early diagnosis and treatment of aspergillosis can be difficult because obvious clinical signs often do not develop until the disease is advanced. Clinical signs include dyspnea (labored/concave or open mouth breathing), lethargy, weight loss, and inappetance. In some cases, birds present acutely dyspneic with a tracheal granulomatous plug causing a life-threatening respiratory compromise. In other cases, birds gradually lose weight and develop mild chronic dyspnea characteristic of slowly-growing lung or air sac granulomas. Additional physical examination findings may include discharge from the nares, epiphora, fungal plaques on third eyelid or conjunctiva, pale mucous membranes, a prominent keel, poor plumage quality and water-proofing, and an audible pulmonic/tracheal click or decreased respiratory sounds on auscultation.

Preliminary diagnostics recommended include a CBC, chemistry, and PEP. Aspergillosis has been found to produce a characteristic hypergammaglobulinemia on PEP, which is highly suggestive of the disease in ill alcids. Elevated serologic *Aspergillus* antigen titers and galactomannan levels may increase index of suspicion of the disease, but a negative titer does not rule out fungal infection. Presumptive therapy is often started based on clinical presentation and PEP results. Further confirmation of the disease can be demonstrated with culture and cytology of *Aspergillus* from the tracheal or choanal mucous. Radiographs may be helpful in characterizing the severity and progression of the disease. Visual radiographic changes, including air sac opacities or discrete respiratory masses (granulomas), are indicative of advanced disease and a poor long-term prognosis. Endoscopy of the trachea and air sacs to obtain biopsies or swabs of lesions/fungal plaques may also be warranted. Patient medical status and risk of anesthesia should be considered before pursuing invasive diagnostics.

Therapy should be is started immediately once aspergillosis is suspected. Antifungal drugs can be administered orally, intratracheally, or by nebulization. The severity of the infection and patient condition will aid in determining what form or combination of treatments to administer. Cannulization of the abdominal air sac may be required to support the acutely dyspneic patient with a tracheal plug. Tracheal plugs are generally found at the bifurcation of the bronchi or syrinx and are extremely difficult to remove. In addition to antifungal therapy, supportive care such as nutritional support, fluid therapy, and antibiotic administration may be necessary for severely compromised individuals. Treatment for chronic fungal infections usually requires long-term antifungal therapy, for weeks to months, and in some cases, years. Quality of life in those individuals can be fairly good as long as medications are tolerated with few to no side effects. Antifungal drug toxicity is known to occur and can be monitored by testing serum transaminase levels, bile acids, serum drug levels, patient weight, body condition, and appetite. Poor response to therapy is not uncommon in alcids with advanced aspergillosis, and it can result in overwhelming disease progression to multiple organs, severe respiratory compromise, secondary bacterial infections, organ failure, and stress.

Bacterial infections in managed alcids have been known to occur via food-borne introduction or secondary to trauma. *E. coli* is considered part of the normal intestinal flora in seabirds, but it can cause illness when it is present in large amounts. In wild murres, potentially pathogenic bacterial species such as *E. coli*, *Pasteurella multocida*, *Yersinia intermedia*, *Actinobacillus sp*., *Clostridia perfringens*, *Streptococcus sp*., and *Salmonella spp*. have been found (Stoskopf, 1993; Muzaffar & Jones, 2004). *Salmonella* isolates have also been cultured from the droppings of wild razorbills (Muzaffar & Jones, 2004). *Ixodes* tick-infested alcids can also potentially serve as reservoirs for *Borrelia garinii*, one of the genomic species of *B. burgdorferi*, the causative agent for Lyme disease (Olsén et al., 1993; Muzaffar & Jones, 2004). In cases where bacterial infections result in disease in alcids, systemic antibiotics can be administered intramuscularly or orally in fish or gruel.

Adenovirus particles have been noted in two wild murres without definitive association with clinical disease (Lowenstine & Fry, 1985; Stoskopf, 1993). To date, there have been no documented cases in managed alcids involving West Nile Virus infection or Newcastle Disease (*Paramyxoviridae sp.*), but one wild common murre was found with Paramyxovirusin in the German Bight (Stoskopf, 1993). Alcids kept in outdoor exhibits in non-native ranges, even if within appropriate latitudes for the species, may potentially be exposed to viral diseases to which they are immunologically susceptible.

A wide range of parasitic species have been identified in alcids (Muzaffar & Jones 2004). The pathogenicity of many alcid parasites, however, is often poorly understood. Lice do not generally affect feather quality in auks. Occasionally, however, these parasites can cause pruritus. Treatment with oral or injectable ivermectin, or dusting with a pyrethrin-based insecticide is effective for pediculosis. Mites of the genus *Alloptes* have been identified in several alcid species including razorbills, murres, and Atlantic puffins (Muzaffar & Jones, 2004). Though feather mite infestation is generally asymptomatic in alcids, high densities of mites can cause skin irritation, leading birds to preen excessively and damage their plumage. Treatment with systemic parasiticide therapy (e.g., ivermectin) is usually effective. Many subpolar and temperate seabird species, including alcids, have been documented with *Ixodes uriae*. These ticks can serve as a reservoir for borreliosis, and heavy infestations have been reported to cause morbidity and mortality in young auklet chicks (Morbey, 1996; Muzaffar & Jones, 2004).

Eimeria fraterculae has caused renal coccidiosis resulting in renal tubular hypertrophy, tubular dilation, and mild peritubular inflammation in wild Atlantic puffins (Leighton & Gajadhar, 1986). The condition could be diagnosed by identification of oocysts in floatation of urates in droppings. *Plasmodium* and *Sarcocystis spp*. have been identified in wild common murres (Muzaffar & Jones, 2004). Intestinal protozoa resembling *Microsporidia* have been found at necropsies of wild-caught horned puffin chicks (Tocidlowski et al., 1997). Numerous digeneans, cestodes, and nematodes have been identified in wild alcid species. Tapeworm and roundworm infections have been associated with intestinal lesions, but the clinical significance is unknown (Threlfall, 1971; Muzaffar & Jones, 2004).

No vaccinations are currently recommended for routine use in alcids, but institutions may consider vaccinations based on risk assessment of disease exposure, particularly in outdoor exhibits or with emerging diseases.

Some states may require avian influenza testing prior to entry into the state, so the appropriate state agency should be contacted prior to performing a pre-shipment examination on an outgoing bird.

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 marked when anesthetized or restrained (e.g., tattoo, leg band, wing band, 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.

Additional diagnostic testing should be performed if any initial quarantine testing shows evidence of health abnormalities. Alcids should not be released from quarantine unless they appear to be in good health, based on observations, physical examination, and diagnostic findings.

Care should be taken with alcids to make sure that the birds' environment is set up to provide minimal stress. Adequate space is recommended to minimize stress and avoid compromising the health of old, young, or other potentially immunocompromised birds. Staff should offer the birds as little disturbance as possible, and the birds should be handled minimally during this period. Conversely, once quarantine is completed, a frequent human presence will help acclimate the birds to the staff and provide a calmer bird once inside the exhibit.

Adequate privacy and hiding places should be provided. The birds should be provided with some type of "safe house" to hide in during quarantine. Kennels, cardboard boxes, and large hollow rocks have all been used successfully. It is best to keep noise to a minimum. Newly arrived birds may refuse to eat. The method of feeding (water, tray, or bowl) used in the previous institution should be considered, so that the bird makes an easy transition with minimal, if any, weight loss.

If collecting alcids from the wild, the AZA Charadriiformes TAG recommends collecting pre-fledged birds or eggs, as they will acclimate to a new environment more easily than adults. Wild caught birds will take much longer to adjust to new surroundings than birds born in zoos/aquariums. While it can be tempting to try to monitor birds closely, often it is best to leave the adult birds alone for a few days as much as possible. Housing birds with at least one other individual can be beneficial. A mirror can also be added in lieu of a companion if it is necessary to house a single animal.

If an animal should die in quarantine, a necropsy should be performed on it to determine cause of death in order to strengthen the program of veterinary care and meet SSP-related requests (AZA Accreditation Standard 2.5.1). The institution should have an area dedicated to performing necropsies, and the subsequent disposal of the body must be done in accordance with any local or federal laws (AZA Accreditation Standards 2.5.2 and 2.5.3). If the animal is on loan from another facility, the loan agreement should be consulted as to the owner's wishes for disposition of the carcass; if nothing is stated, the owner should be consulted. 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 7.6).

AZA Accreditation Standard

(2.5.1) Deceased animals should be necropsied to determine the cause of death for tracking morbidity and mortality trends to strengthen the program of veterinary care and meet SSP-related requests.

AZA Accreditation Standard

(2.5.2) The institution should have an area dedicated to performing necropsies.

AZA Accreditation Standard

(2.5.3) Cadavers must be kept in a dedicated storage area before and after necropsy. Remains must be disposed of in accordance with local/federal laws.

AZA Accreditation Standard

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

7.4 Preventive Medicine

AZA-accredited institutions should have an extensive veterinary program that must emphasize disease prevention (AZA Accreditation Standard 2.0.2). AZA institutions should be aware of and prepared for periodic disease outbreaks in other animal populations that might affect the institution's animals, and should develop plans to protect the institution's animals in these situations (AZA Accreditation Standard 2.0.3). 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:

AZA Accreditation Standard

(2.0.3) Institutions should be aware of and prepared for periodic disease outbreaks in wild or other domestic or exotic animal populations that might affect the institution's animals (ex – Avian Influenza, Eastern Equine Encephalitis Virus, etc.). Plans should be developed that outline steps to be taken to protect the institution's animals in these situations.

(https://cdn.ymaws.com/www.aazv.org/resource/resmgr/files/aazvveterinaryguidelines2016.pdf).

Animal care personnel are the first line of preventative health care in institutions managing colonies of alcids. Well-trained individuals should be able to recognize changes in behavior, appetite, and clinical condition, and they should convey these to the veterinary staff.

Health screening is often performed opportunistically in managed alcid colonies. Routine health screening is recommended in alcid species tolerant of handling or when chronic health problems exist for an individual or colony. Prior to handling an alcid for physical examination, it should be observed in its regular environment. Manual restraint often masks subtle behavioral or physical changes that can be helpful in determining a diagnosis; a bird's posture, interaction with other birds, breathing, and mobility may appear differently on exhibit or in a holding area than when the bird is in hand. See Table 10 for general routine health screening components. Modalities such as electrocardiography, Doppler, pulse oximetery, capnography, and blood gas analysis have potential application in alcids.

Table 10. Routine medical examination procedures

Procedure	**Description**
Physical examination	Taking physiologic measurements including heart rate and respiratory rate
Bill or nail trimming	If needed (Some alcids experience overgrowth of their bills to the extent that they need to be trimmed. Depending on the severity of the overgrowth, either nail-clippers or a dremel tool may be used to trim the excess. In either case, a bite stick is a handy tool to use to keep the bird's mouth open.)
Band care	If needed
Diagnostic testing	Complete blood count (CBC), chemistry, protein electrophoresis (PEP), +/- fecal analysis, +/- cloacal culture, +/- fecal gram stain
Deworming	For internal or external parasites
Serum and tissue banking	Valuable when evaluating historical perspective of a disease process in an individual or colony
Ophthalmic examination	Corneal and lens evaluation (retinal examination is often difficult given the challenges of adequate pupillary dilation)
Ear examination	Evaluate for abnormalities
Oral examination	Assessment of the mucous membranes, choana, and glottis
Cardiovascular and respiratory evaluation	Should include auscultation of the heart, lungs, and thoracic inlet
Abdominal palpation	To evaluate any abdominal distention
Musculoskeletal and dermatological evaluation	Specific focus on the feet. Pododermatitis, plantar epithelial thinning, callousing, scabbing, and joint swelling are common medical problems in *ex situ* alcids.
Plumage evaluation	For poor waterproofing, oily appearance, poor molt, the presence of stress bars, and other feather abnormalities. The uropygial gland should also be checked for swelling or abnormal discharge.

Blood can be obtained from the jugular, medial metatarsal or brachial vein. If taken from the jugular vein, the head should be held so that the thumb is up and the bird's neck is outstretched. During jugular venipuncture, it is particularly important that the bird is well restrained to avoid damage to the jugular vein or adjacent structures with the needle. If blood is taken from a wing, a second person will need to assist to extend the wing outward. Radiographs can be taken without anesthesia with the use of a backboard or restraint board. This has proven to be successful and minimally invasive. When medication needs to be administered, oral medicines can be given without restraint by offering them in food items. However, if the bird refuses the food, a gavage or soft tube can be used for any liquid medication. Two people should be involved when gavaging and/or giving injections.

Each bird should be weighed opportunistically or routinely if possible (see Table 11) and weight comparisons should be established for different seasons. Handling during molting should be avoided or limited, if possible. Molting, breeding, and egg laying history should also be recorded routinely. Photographic documentation is urged to follow chronic problems and monitor therapy. Body condition can also be assessed by palpation of the pectoral muscles and keel, as well as evaluation of the thoracic inlet. Thin birds will often have atrophied muscles, a prominent keel, and a cavitating thoracic inlet.

Table 11. Weight Ranges compiled from CRC Handbook of Avian Body Masses* (Dunning, 1993)

Species	Weight Range Male	Weight Range Female
Tufted Puffin (*Fratercula cirrhata*)	743.7–867.3 g (26.23–30.60 oz.)	686.2–801.2 g (24.21–28.26 oz.)
Horned Puffin (*Fratercula corniculata*)	510.4–601.6 g (18–21.22 oz.)	472.6–561.8 g (16.67–19.82 oz.)
Atlantic Puffin (*Fratercula arctica*)	Average: 381 g (13.44 oz.), sex unknown	
Common Murre (*Uria aalge*)	927–989 g (32.70–34.89 oz.)	965–1035 g (34.04–36.51 oz.)
Pigeon Guillemot (*Cepphus columba*)	445–521 g (15.70–18.38 oz.)	467–507 g (16.47–17.88 oz.)

* Weights also vary with region and time of year in wild birds.

Neonates: Once the chick has hatched, its health can be monitored in many ways, including daily weights, observation of physiological conditions, and behavior. It is important to note that hands-on or close visual monitoring of murre chicks may not be possible because of the risk of disturbing nearby nesting birds. Chicks have been successfully fledged in exhibits, but problems can occur because of aggression from adults or unfamiliarity with the exhibit. Some institutions choose to remove chicks from nest boxes prior to fledging (Brackett, 2013). Thermoregulatory support is essential during the early neonatal period and hydration status should be assessed daily.

A preventative medicine program should be in place for all Alcids. Frequency of examinations will be dependent on a variety of factors, including previous medical history, the size of the group, and ease of access to individuals. If weights can be obtained without restraint this helps assess the health of the individual with minimal stress. Routine examinations may include physical exam, weight, blood collection for complete blood count, biochemistry panel, serum banking, and whole body radiographs. Fecal samples should be analyzed twice a year for parasite screening. Reference values for complete blood counts and biochemistry panel are listed in Appendix K.

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).

While currently there are no known alcids used for off-grounds programs, the potential does exist. Such birds should be housed separately from collection alcids and cared for by separate staff. Birds that go off grounds for medical testing (i.e., x-rays, CT scan, etc.) should not be exposed to other birds while off grounds. If this is unavoidable, when arriving back on grounds, the bird should re-enter a quarantine period.

AZA Accreditation Standard

(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 animals at the institution from exposure to infectious agents.

AZA Accreditation Standard

(11.1.3) A tuberculin (TB) testing/surveillance program must be established for appropriate paid and unpaid staff in order to assure the health of both the paid and unpaid staff and the animals.

A tuberculin testing and surveillance program must be established for paid and unpaid 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.

There is no need for animal care staff to be tested for tuberculosis to work with alcids. No specific tuberculosis surveillance is performed on alcids, but any abnormalities consistent with mycobacterial disease (i.e., severe leukocytosis, bone or pulmonary lesions on radiographs) detected on exam of a healthy or ill bird should be investigated further.

No routine vaccinations are recommended for alcids. Vaccination for West Nile virus may be considered in endemic areas (Fowler & Miller, 2008).

7.5 Capture, Restraint, and Immobilization

AZA Accreditation Standard

(2.3.1) Capture equipment must be in good working order and available to authorized, trained personnel at all times.

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).

Consideration should be given to using a net for catching a single bird. Bringing a large net into the exhibit may cause mass chaos for the entire colony. A small net may be useful if kept hidden from the birds until the last minute. When a bird is singled out, it should be approached slowly, and then grabbed swiftly by hand or with the small net when in reach. If possible, it is best to wait until the bird is in a spot where the rockwork can be utilized as a barrier. When it is necessary to capture the entire collection, it is best to catch available birds opportunistically before disturbing the whole colony. A plan of action should be devised, and as many participants as possible should be used to expedite the process. Ideally, the pool should be drained, or dropped low enough for animal caretakers to wade in the water. As the rest of the birds go into the water, nets can be used to catch the remaining birds. The use of a prophylactic anti-fungal, such as itraconazole, should be considered prior to the catch up.

Capture myopathy is not uncommon in birds that are subject to excessive handling and severe muscle exertion that often accompanies capture restraint. Birds suffer severe acidosis due to increased potassium levels leaked from damaged muscle cells, which can lead to muscle stiffness and cardiac fibrillation. Severe myopathy progresses to paralysis and secondary circulatory collapse. Treatment with vitamin E and selenium, fluids, and intravenous sodium bicarbonate can help birds with mild to moderate clinical signs only. Prevention is paramount; steps should be taken to limit excessive handling and avoid chasing individuals for handling, especially during extreme changes in temperature (Tully, Lawton, & Dorrestein, 2000).

When handling or restraining an alcid, it is recommended that clean gloves be worn to protect the bird's feathers, as well as to protect the holder against possible bites. Alcid plumage is susceptible to contamination from human oils, causing degradation of feather quality. Care should be taken to wash hands with a disinfectant that removes oils before handling alcids with bare hands.

The head of an alcid should be restrained in such a way to avoid restricting the bird's airway, and to limit the chance of being bitten by the bird. The palm of one hand should be placed at the backside of the head, and the fingers and thumb should be placed at the jawbone (with the hand in either direction). With the other hand, the wings of the bird should be contained against its body, and the whole bird should be supported next to the holder's abdomen.

Care should be taken not to constrict the bird. Simple restraint devices, such as a large 0.91 kg (32 oz.) clean plastic cup with ventilation holes at the bottom and/or a small towel to wrap around the bird's body to secure its wings, work well to reduce stress during handling. Birds are placed head first into the cup and restrained lightly by the legs. Body weights can also be collected using this technique.

A towel can be used to restrain the bird. If using a towel, one hand is used to restrain head in the way mentioned above from the outside of the towel while the feet are secured by the other hand, with one finger placed in between the legs. The towel is used to secure the wings (Figure 9). Blood samples can be collected from this method by releasing one leg and rotating bird on its side (Figure 10). Additionally, training alcids to stand on a scale is a noninvasive method to monitor health and crate training can be used to make capture easier.

Figure 9. Restraining a tufted puffin
Photo courtesy of Aimee Greenebaum

Figure 10. Restraining a tufted puffin
Photo courtesy of Aimee Greenebaum

Care should be taken to avoid repeated handling or prolonged manual restraint in order to limit the stress on an animal and prevent further disease progression, hyperthermia, or mortality. All staff should be trained on capture and restraint by observing experienced staff a number of times. Staff should then be assisted by experienced staff when restraining birds until they, and their supervisor, are comfortable with their skills.

In situations where chemical immobilization is necessary, alcids should first be captured physically using the method above. Alcids should be fasted at least 12 hours prior to anesthesia to reduce the likelihood of regurgitation and aspiration. Puffins are easily mask-induced and maintained with isoflurane or sevoflurane gas anesthesia. Intubation can be performed with a Cole tube or modified 14-gauge catheter. As with other avian patients, close physiological monitoring is important in alcids to assess depth of anesthesia, cardiac and respiratory status, and body temperature. Like other diving birds, alcids' breath-hold should be considered during mask induction of anesthesia. Additionally, respiratory depression is not uncommon at the higher anesthetic gas percentages often needed to anesthetize an alcid patient. Breath holding, respiratory depression, or apnea can be a problem with alcids maintained on mask anesthesia alone. It is generally recommended to intubate and provide assisted ventilation to any alcid undergoing general anesthesia to reduce the risk of hypoxemia or hypercapnia associated with respiratory depression. Anesthetic monitoring for alcids is similar to other birds. Temperature evaluation is important, because alcids can overheat when stressed. A towel-covered ice pack may be placed under the bird to cool its body temperature if needed. Esophageal or cloacal temperature probes can be used.

7.6 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. The owner of an animal on loan at a facility is to be consulted prior to any elective invasive procedures, including permanent contraception.

Alcid care staff should be trained in meeting the animal's dietary, husbandry, and enrichment needs, as well as in restraint techniques. Staff should also be trained to assess animal welfare and recognize behavioral indicators animals may display if their health becomes compromised, however, animal care staff should not diagnose illnesses nor prescribe treatment (AZA Accreditation Standard 2.1.3). Protocols should be established for reporting these observations to the veterinary department. Hospital facilities for alcids must have radiographic equipment or access to radiographic services (AZA Accreditation Standard 2.3.2), 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.

AZA Accreditation Standard

(2.1.3) Paid and unpaid animal care staff should be trained to assess welfare and recognize abnormal behavior and clinical signs 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, animal care staff (paid and unpaid) must not diagnose illnesses nor prescribe treatment.

AZA Accreditation Standard

(2.3.2) Institution facilities must have radiographic equipment or have access to radiographic services.

Alcids are generally healthy birds. Most birds are excellent at masking illness until it is severe. Any indicators of illness, such as poor appetite, fluffed appearance, lethargy, labored breathing, lameness, or decreased flight response, should be considered a sign of compromised health and reported to veterinary staff promptly. While awaiting veterinary care it is often helpful to separate the bird and place it in a quiet, warm environment, providing a temperature gradient using heat lamps or pads. Common signs of medical problems in alcids are listed in Table 12.

Table 12. Common signs of medical problems in alcids

Symptom	Description
Behavioral changes	Isolation from colony or social displacement, postural changes on land and in water (i.e., riding low in water), decreased preening, decreased swimming/flight activity, lethargy, depression
Thin body condition	Prominent keel, shoulders, pelvis
Weakness, unsteady balance	-
Inappetance or decrease in appetite	-
Gastrointestinal signs	Vomiting, regurgitation, abnormal feces, fecal staining around vent
Poor plumage condition	Greasy/oily feathers, loss of water-proofing
Respiratory signs	Labored or open mouth breathing, increased rate or effort of breathing, loss of vocalization, coughing, sneezing, over-inflated air sacs in neck
Neurologic signs	Seizure, obtunded, head tilt
Lameness	-
Hemorrhage	-
Signs of trauma	Wounds, fractures, etc.
Abnormal discharge	From eyes, ears, nares, oral cavity, vent, wounds
Abnormal swelling	Feet, joints, facial, etc.

Infections: Bacterial infections can be primary or secondary. *Salmonella* spp., *Erysipelothrix* spp., *Campylobacter* spp., *Chlamydophila* spp., and *Yersinia* spp. have been reported in charadriiformes and have zoonotic potential (Ball, 2003). *Mycoplasma* spp. are also occasionally encountered. *Mycobacterium avium* infection is uncommon, but it should be considered with any radiographic evidence of bony lesions, particularly when other diagnostics support an infectious cause (Ball, 2003). Wounds and pododermatitis lesions can become infected with a variety of bacteria, which may lead to osteomyelitis. *Klebsiella pneumoniae* is often implicated in cases of respiratory tract disease in charadriiformes in zoos/aquariums and was implicated in chick mortality of black stilt chicks (Reed, Sancha, & Fraser, 2000). Culture and sensitivity of suspected bacterial infections is indicated. In cases where bacterial infections result in disease in alcids, systemic antibiotics can be administered intramuscularly or orally in fish.

Avian Influenza is not uncommonly found in alcids. Typically it causes an unapparent or subclinical infection and does not require treatment. Sick birds should receive supportive care. A mortality event occurred in wild common terns in South Africa in 1961, but this is not typical (Friend & Franson, 1999). Ten of the 15 hemagglutination types and eight of the nine neuraminidase types have been identified in shorebirds, and many of the combinations are unique to alcids. The H9 and H13 types predominate. The avian influenza type H5N1 has been identified in shorebirds (Hall et al., 2013).

Newcastle disease virus and infectious bursal disease have been seen serologically in wild Charadriiformes. The significance of these diseases is unknown. A mortality event has been associated with a reovirus in American woodcocks (Docherty et al., 1994) and avian pox has been diagnosed in Sanderlings (Kreuder et al., 1999). Puffinosis is a serious viral disease of unknown cause (Pokras, 1996). Charadriiformes have been shown to be competent reservoirs for West Nile Virus, and vaccination may be considered (Travis, 2008). Supportive care should be administered to any clinically ill birds, at the discretion of each facility's veterinary staff.

Young birds particularly are at risk for candidiasis, with hand-reared birds most susceptible. Adult birds may develop candidiasis while receiving antibiotic therapy. Clinical signs include a mucoid exudate and white plaques. Improving hygiene and administering proper antifungal therapy (such as nystatin) are usually curative.

The most common reports of parasitic infections include cestodes and *Capillaria* (Ball, 2003). Cestodes are more common in wild-caught birds than those born *ex situ*, due to the lack of a suitable intermediate host. *Capillaria* has a direct life cycle and is easily transmitted between birds. Several species of ticks have been identified on wild birds and, although are not considered significant themselves, can carry hemoparasites and viruses. Lice do not generally affect feather quality. Occasionally, however, these parasites can cause pruritus. Treatment with oral or injectable ivermectin, or dusting with a pyrethrin-based insecticide is effective for pediculosis (Tully et al., 2000).

<u>Aspergillosis:</u> Aspergillosis is not uncommon in managed alcids, although it is rare in free-ranging populations. *Aspergillus fumigates* is the most common causative agent. Stress and subsequent immunosupression likely play a role in the pathogenesis of the disease *ex situ*. The fungus is generally acquired through the respiratory tract, causing lesions in the air sacs, lungs, and even the central nervous system and eyes. Significant changes with animal management (e.g., newly introduced birds and exhibit construction), and inherent social or physiological stressors such as breeding, molting, and interspecies competition for territory and food, are major factors that enhance susceptibility to fungal disease. Poor air turnover, excessive humidity, and certain types of substrates can increase fungal growth and sporulation within the exhibit (Tully et al., 2000).

Early diagnosis and treatment of aspergillosis can be difficult since obvious clinical signs often do not develop until the disease is advanced. Clinical signs include dyspnea (labored/concave or open mouth breathing), lethargy, weight loss, and inappetance. In some cases, birds present acutely dyspneic with a tracheal granulomatous plug causing life-threatening respiratory compromise. In other cases, birds gradually lose weight, and develop mild chronic dyspnea characteristic of slowly-growing lung or air sac granulomas. Additional physical examination findings may include discharge from the nares, epiphora, fungal plaques on third eyelid or conjunctiva, pale mucous membranes, a prominent keel, poor plumage quality and water-proofing, and an audible pulmonic/tracheal click or decreased respiratory sounds, on auscultation.

Diagnostics include a complete blood count, biochemistry panel, aspergillosis antigen and antibody levels, protein electrophoresis, radiography and/or endoscopy, and fungal culture. Caution should be used in interpreting serologic tests as these have not been validated in alcids. Because disease is often advanced by the time the bird exhibits signs of illness, aspergillosis may be diagnosed post-mortem. Treatment is difficult and may include systemic antifungal agents, such as itraconazole, topical application of antifungal agents to granulomas using endoscopy, or nebulization with antifungal agent such as terbinifine. Similar treatment methods can be used as with other avian species and are discussed in detail elsewhere. Prevention of aspergillosis can be attempted through adequate ventilation, removal of decaying organic debris, and monitoring risk through fungal spore surveys, including air spore sampling (Dykstra, 1997; Faucette et al., 1999).

Non-infectious health concerns: The most common non-infectious diseases in managed alcids include trauma and pododermatitis. Trauma injuries, either accidental or from inter-specific aggression, can be reduced by limiting or removing obstacles in areas where birds tend to fly or swim. Lameness often occurs in fledglings that have hard landings, and causal factors include: broken/avulsed toenails, cruciate ligament rupture, fractured legs/toes, and lacerations of webbing or footpad. Wing trauma may occur with newly introduced birds, territorial battles, incorrect restraint, or contact with obstacles during flight. Introduction of new birds should be timed during non-breeding seasons to reduce conspecific aggression, as ocular or beak injuries have been known to occur during territorial battles. Ocular treatment and beak repair are similar to those techniques used in other avian species, and acrylic splints or K-wires have been used to stabilize beak fractures in alcids.

Treatment of fractures or ligament repair should allow the bird to be functional; otherwise, its prognosis for survival in a colony setting is poor. Water access restrictions should be considered during the recuperation phase in some cases of trauma, particularly those involving wound healing or bandage placement. The length of time a bird can be removed from water varies on the severity of the injury, the health status and behavior of the individual, and facility constraints. In cases where water access is preferable, but a bird is not well flighted, exhibit modifications to allow easy ambulatory access into and out of the pool are recommended. In cases of beak trauma, it is important to restore the functionality of the beak. Birds may require nutritional support during treatment or repair.

Pododermatitis can occur in nearly all shorebird species and can be seen at any age. While a number of predisposing factors, such as stress and nutrition, may have a role in the occurrence of pododermatitis, substrate quality, character, and cleanliness seem to predominate in the development of clinical disease. Prevention is key to management. The correct substrate is strongly recommended. Routine foot checks and photographic documentation of active bumblefoot cases will aid in monitoring foot condition of affected individuals. Treatment of pododermatitis often depends on the severity of the lesion, the ease of medicating or handling a bird, and the rehabilitation environment. Mild lameness can often be treated with anti-inflammatories alone, for short or long duration. Progressive lameness may require more intervention, including diagnostics to rule out infection of soft tissue or underlying bone, anti-inflammatory therapy, padding/bandaging the foot, or even surgical debridement. In some cases, birds develop a palpable plug of granulomatous material in their foot.

Neonates: Alcid chicks are susceptible to problems such as yolk sac infection or retention, sepsis, dehydration, hypothermia, weakness, splayed leg, constipation, trauma, poor feeding reflex or appetite, and poor weight gain. Early recognition of these clinical signs, and immediate therapeutic response, is often necessary to minimize long-term complications or mortality. Thermoregulatory support is essential during the early neonatal period and hydration status should be assessed daily. Nutritional support of ill neonates is also imperative due to the high metabolic demands of growing and immunologically stressed chicks. Once the chick has hatched, its health can be monitored in many ways, including daily weights, observation of physiological conditions, and behavior. It is important to note that hands-on or close visual monitoring of murre chicks may not be possible because of the risk of disturbing nearby nesting birds. Chicks have been successfully fledged in exhibits, but problems can occur because of aggression from adults or unfamiliarity with the exhibit. Some institutions choose to remove chicks from nest boxes prior to fledging (Brackett, 2013).

<u>Angel wing:</u> Angel wing is a condition that is caused by the weight of growing flight feathers placing excess stress on the weak muscles of the carpal joint. Dietary deficiencies or massive weight gains can be a factor. This weight can cause the developing wing to hang and eventually twist outward. If caught in the early stages, feathers can be removed to reduce the weight on the wing. More aggressive treatment involves taping the drooping wing up to itself (not to the body) in a normal position for three to five days. If untreated, the wing may remain in that position and the ligaments and bones will be permanently deformed.

<u>Splayed legs:</u> Congenital and developmental abnormalities of the feet have also been noted in neonates either naturally or artificially incubated. Splay leg syndrome can often be minimized by early hobbling of the chick's legs. Bandages should be removed and replaced often, sometimes daily or every other day, to allow adjustments for growth.

<u>Angular limb deformities:</u> Angular limb deformities can be seen in hatchlings being hand-reared on inappropriate diets. Correcting diet and early intervention is crucial.

<u>Bacterial/yeast infection:</u> Overgrowth of bacteria and yeast in the gastrointestinal tract, wounds, or retained yolk sacs is a concern with neonates. Antibiotic and antifungal therapy is warranted in these cases.

Geriatrics: Arthritis has been noted in geriatric alcids. Cases of gout may be associated with age-related renal changes. Treatment is similar to other avian species. Caution is warranted with non-steroidal anti-inflammatory agents, as their use can result in adverse renal or gastrointestinal side effects. Meloxicam has been used safely in avian species, but no specific studies have been performed in alcids (Sinclair, 2012).

Hospitalization: Hospitalization should take place in appropriate sized stalls that allow for inclusion of a pool. A sandy shore or soft soil is desirable and allows normal feeding behavior. Adequate privacy and hiding places should be provided. It is best to keep noise to a minimum. A pool is an important part of any holding room and should be kept clean at all times.

Housing for ill animals should be easily disinfected, and allow birds to be maintained in their preferred environmental parameters. For those birds with limited mobility or bandages, pool access may need to be initially restricted until the bird is more recuperated. Substrate choices should minimize trauma to the feet that could predispose a bird to pododermatitis. Knotless net bottom caging has been used in some rehabilitation situations to minimize hock and keel pressure sores, reduce fecal contamination, and allow drying of wet birds.

An area separate from the exhibited population is ideal, including a separate water and air supply, in case of a transmittable disease. All holding areas should have the ability to mimic the light and temperature parameters of the main exhibit, as well as the air and water quality. Fluorescent lights can be used in the holding room if the space is used for short-term holding, but it is useful to have any lights on a timer so that the photoperiod can be controlled to match the exhibit photoperiod. A skylight can add additional lighting to the room, but it may also increase the air temperature.

Isolation can be a very stressful situation for any bird, and it is important to make the bird as comfortable as possible during this period. Providing the security of a hide (e.g., plants, driftwood, or a cardboard box) is recommended, and offering favorite food items can also be considered. If the need for isolation is not related to an infectious agent, adding another bird for company should be considered because this might minimize stress to the compromised bird. However, this may also cause undue stress to the healthy bird. The utilization of mirrors has also been successful as an alternative to a live companion bird.

The placement of the holding/isolation room should be as close to the exhibit as possible. It is possible that if isolated birds can hear the sounds of the birds on exhibit, it may reduce the stress of separation from the colony. The risk of disease transmission, however, should be weighed against the social and psychological benefit of cooperative housing.

AZA-accredited institutions must have a clear and transparent process for identifying and addressing alcid 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 (and adjustment of these responses if necessary) of the outcome of these responses, and the dissemination of the knowledge gained from these issues.

AZA Accreditation Standard

(1.5.8) The institution must develop and implement a clear and transparent process for identifying, communicating, and addressing animal welfare concerns from paid or unpaid staff within the institution in a timely manner, and without retribution.

There are no specific protocols for reporting welfare concerns for alcids and each individual institution's policies should be followed. If necessary, the appropriate SSP and/ or TAG should be contacted. Staff should be trained in normal husbandry and behaviors for all species in the collection so they can adequately assess welfare. Speaking with colleagues at other institutions can help in assessing and disseminating information regarding welfare concerns. The veterinary team may also be a resource in assessing welfare concerns and the impact on health.

AZA-accredited zoos and aquariums provide superior daily care and husbandry routines, high quality diets, and regular veterinary care, to support alcid 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 alcids both in their care and in the wild. As stated earlier, necropsies should be conducted on deceased alcids 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 Standards 2.5.1 and 2.5.3). If the animal is on loan from another facility, the loan agreement should be consulted as to the owner's wishes for disposition of the carcass; if nothing is stated, the owner should be consulted. 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 alcid SSP Program approved active research requests that could be filled from a necropsy.

Shipment and quarantine is a stressful time for avian species, and underlying disease not detected prior to shipment may result in death. Stress also suppresses the immune system, making birds more susceptible to diseases such as aspergillosis. Birds that die during quarantine should be necropsied as soon as possible. Common causes of death include chick mortality, trauma, and infectious diseases (particularly aspergillosis).

In cases where euthanasia is indicated, humane procedures should be followed. In most instances, it is recommended to anesthetize the bird via mask induction with either isoflurane or sevoflurane prior to euthanasia. Once the animal is adequately anesthetized, an injection of pentobarbital can be given intravenously or intraperitoneally. Note that pentobarbital is a controlled substance, and DEA regulations for its use should be followed. Cardiac auscultation or Doppler should be used to ensure the bird has died prior to necropsy examination. For more detailed information on euthanasia guidelines please refer to the AVMA guidelines on euthanasia: www.avma.org/issues/animal_welfare/euthanasia.pdf.

Post-mortem examination should include assessment of body weight and condition. Tissue samples should be placed in formalin for histopathology and should include the following: brain, eye, tongue, skin, muscle, bone, trachea, esophagus, thyroid, parathyroid, proventriculus, ventriculus, intestine, pancreas, heart, lung, thyroid, liver, kidney, adrenal, spleen, and gonad. In certain cases it may be advisable to obtain samples for culture or to freeze tissue for additional testing.

Chapter 8. Reproduction

8.1 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.

Alcids reach sexual maturity between three to five years of age. They exhibit slight sexual dimorphism, with some males having a marginally larger bill depth than females (Gaston & Jones, 1998). In alcid species studied to date, mating pairs remain together throughout life, returning to the same nesting area each year. Prior to the onset of breeding season, which usually occurs at the beginning of April, alcids go through seasonal plumage, feet, eye, and bill coloration changes. These are indicators of the timing of the breeding season for managed colonies. In addition to the physiological changes, alcids exhibit several distinctive breeding season behaviors. Puffins have a breeding vocalization, variously described as growling or barking, which is accompanied by head flicking (where the head is quickly jerked up and down). The male swims around the female, while calling (Nelson, 1979). It is also common to see head-bowing, billing, and wing-fluttering used by the male to persuade the female to copulate. Successful copulations in puffins are performed in the water (Freethy, 1987). Some prey species, like silversides and sand eels/lances, have been identified as popular courtship items in several managed alcid species.

In addition to the abovementioned behaviors, burrow-nesting alcids will begin to look for suitable nest locations. Ledge-nesting alcids will come on to land in large numbers and congregate on ledges. This will be an indication that it is time to open nest boxes (by removing objects blocking the entrances) and institute husbandry modifications necessary to promote breeding. Aggression can increase during breeding season, and birds should be monitored closely. If fighting escalates on a regular basis, moving some of the birds to off-exhibit holding should be considered.

The following molting timelines (Table 13) are for breeding adult birds in at least their third year of life. The molt can begin and usually ends within the time indicated. A molt cycle that overlaps with another indicates that not all birds start/end their molts at the same time. This table is to be used as a guide to molt times and not as an absolute rule regarding the beginnings and ends of molts.

Table 13. Molting time line compiled from data (Pyle, 2008)

Bird	DPB*	Breed/Raise Chicks	DPA**
Horned Puffin	January–April & August–December	May–August	March–May
Tufted Puffin	August–December	May–August	January–April
Common Murre	July–October	May–August	November–April
Pigeon Guillemot	July–October	May–August	January–April

*DPB: Definitive pre-basic molt. The molt that results in the birds Basic or non-breeding plumage.
**DPA: Definitive pre-alternate molt. The molt that ends with the bird in Alternate or breeding plumage.

At this time, natural conception is the only type of breeding that has been achieved with alcids. Hormonal and chemical changes have not been studied in alcids, and more research is needed into hormonal changes and tracking methods to better manage alcid breeding.

8.2 Assisted Reproductive Technology

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, species and should be investigated further.

Besides physical issues, AI procedures also bring issues of ownership of semen and/or the animal being inseminated. Very often, semen from multiple animals may be used. As with any natural (physical) breeding, the rights of the owners of all materials and animals involved must be considered. Appropriate transaction documents (and loan agreements, if appropriate) must be fully completed before AI is attempted.

At this time, AI has not been used with alcids in zoos and aquariums; more research is needed in this area.

8.3 Pregnancy & Egg-laying

It is extremely important to understand the physiological and behavioral changes that occur throughout an animal's pregnancy. All alcids lay one egg, except guillemots which lay two eggs. The following table (Table 14) provides a list of alcid incubation periods. Birds lay one to two clutches consisting of one to two eggs each year, and both parents participate in egg incubation and chick rearing.

Table 14. Reproductive information for some of the species of alcids, compiled from other AZA institutions and Birds of North America

Bird Group	Average clutch size	Average incubation period	Average age at fledging	Incubation temperature	Humidity (wet bulb temp)	Age of sexual maturity	Other notes
Atlantic puffin	1	36–42 days	38–44 days	36.7–36.9 °C (98.1–98.5 °F)	27.2–27.5 °C (81–82 °F)	4–6 years	
Tufted puffin	1	40–45 days	48.2 days ±4.1 SD	36.7–36.9 °C (98.1–98.5 °F)	27.2–27.5 °C (81–82 °F)	4–5 years	
Horned puffin	1	40–42 days	Wild 42 ± 0.85 days Managed 44 ±1.2 days	36.7–36.9 °C (98.1–98.5 °F)	27.2–27.5 °C (81–82 °F)	4–5 years	
Black guillemot	1–2	26–33 days	35–40days	Further research needed	Further research needed	2–3 years	
Pigeon guillemot	2	29–32 days	35–54 days	Further research needed	Further research needed	3 years	
Common murre	1	26–39 days	53–83 days	98.1° F (36.7° C)	81–82° F (27.5° C)	4–5 years	Intermediate fledge strategy, average 23 days
Thick-billed murre	1	29–35 days	45–66 days	Further research needed	Further research needed	5 years	Intermediate fledge strategy: 15–33 days
Dovekie (little auklet)	1	28–31 days	29 ±0.08 days	Further research needed	Further research needed	2–3 years	
Least auklet	1	25–39 days	30 days ±2 days	Further research needed	Further research needed	2–3 years	
Parakeet auklet	1	35 days	35 days	Further research needed	Further research needed	3–4 years	

Rhinoceros auklet	1	44 days	Average 50 days	Further research needed	Further research needed	3 years
Crested auklet	1	34 days	27–36 days	Further research needed	Further research needed	2–3 years
Whiskered auklet	1	35–36 days	39–42 days	Further research needed	Further research needed	3 years

Changes in appetite can be seen in egg laying females; sometimes there is an increase in the amount of food consumed a few days before laying. Females' calcium levels also tend to fluctuate with reproductive state.

Medical problems during reproduction are not unique to alcids and may include egg binding, thin-walled eggs, and egg yolk peritonitis. Standard diagnostic and treatment techniques used in other avian species are appropriate. Equipment used for parturition includes incubators, brooders, heat lamps, and tweezers for hatching assistance as needed.

Indicator cues for egg binding include straining without passing an egg or possibly viewing part of the egg. Treatment depends on the condition of the bird and should be evaluated by a vet. The sicker the bird at presentation, the more aggressive the treatment should be. If the bird appears stable, placing the bird in a warm environment and administering subcutaneous fluid therapy is often adequate. Some birds may require calcium and/or oxytocin treatment. This treatment may be repeated if necessary. If the egg still does not pass or the bird's condition worsens, the egg may need to be removed. Sometimes it is possible to gently manipulate the egg out with adequate lubrication. Use caution to not manually compress the egg as this will lead to sharp edges of eggshell that can cause internal damage. If this does not work, aspirate the contents of the egg using a large gauge needle and syringe. Again, it is important not to manually compress the egg and instead let the removal of the contents collapse the egg. Once this is complete, the shell should pass on its own.

In dealing with thin walled eggs, diet should be evaluated for calcium content. In addition, age of the bird could also be a factor. Older birds may have thinner eggs. If eggs are thin, artificial incubation should be considered.

Indicators for egg peritonitis could include loss of appetite, weakness, respiratory distress, lethargy, fluffed feathers, lack of vocalizations, yolk-colored droppings, and/or swollen vent and/or abdomen (the swelling feels spongy to the touch); some of these symptoms also mimic egg binding. Egg peritonitis often requires intensive treatment including systemic antibiotics, fluid therapy, and as a last resort, surgery.

It may be necessary to assist hatching if there is a problem with the chick's ability to get out of the egg. Also, as a precautionary measure, the yolk sac should be monitored for signs of infection.

8.4 Hatching 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." The area should be considered "baby-proofed" when the chicks do not have access to the water and there are no small areas that chicks can get stuck in. During the breeding season, modifications to exhibit husbandry may be necessary to ensure the health of eggs and chicks, and to minimize disturbance to nesting birds. Nesting burrows and ledges should not be made wet during exhibit cleaning. As soon as nest boxes are opened to the birds, checking for eggs should begin. Females that are suspected of getting ready to lay an egg should not be removed from the exhibit or handled as it could cause egg bound issues.

Nests should be monitored so that eggs are found and documented when they are laid. To decrease the stress on the birds, consider installing nest cams. Keepers should monitor nest activity throughout the day for the pairs to "switch off" incubating the egg. This is an indication that there is an egg in the nest box. Checking for eggs of ledge nesting alcids should be performed cautiously, to avoid flushing the birds from their nests. Nest boxes and burrows should also be checked carefully to avoid startling nesting birds. Frequency and timing of egg checking can be based on a schedule or dictated by the birds' behavior.

New eggs should be weighed and measured at the onset of incubation. Fertility can be determined by candling after one week of incubation, at which time the eggs can be weighed again. Frequency and of weighing and candling the egg during development should be based on the bird's behavior. Extra care should be taken when removing the egg from an incubating parent, using a flat sheet of plastic or wood works well to shield the hand that is removing the egg from being bitten. Placing a warmed dummy egg in the nest during the process of checking the egg will reduce the risk of nest abandonment. Dummy eggs are warmed in the keeper's hands. All weighing and candling equipment should be ready before the egg is removed so that the whole process can be completed in under a minute. The egg may be candled and weighed once more during the week that it is scheduled to hatch.

Alcids that burrow should have a nest that is dark and secluded. These can be artificially created in several ways. Wooden boxes or plastic boxes with removable lids have been fabricated in-house to meet specifications of the species housed. Plastic boxes are preferred because wood holds moisture and is harder to clean and disinfect. A ready-made alternative to wood is plastic irrigation boxes that are available at local hardware stores. Irrigation boxes are made of black plastic 33.02 cm (13 in.) tall and 25.4 cm (10 in.) wide or 30.48 cm x 30.48 cm (12 in. x 12 in.). Boxes have 8.9 cm (3.5 in.) diameter holes in the sides opposite each other with twist-on covers. They come with a slatted removable lid. Five gallon plastic buckets placed on their side, 15.24 to 20.32 cm (6 to 8 in.) diameter PVC pipes, or halves of Sky Kennels® have also been used as artificial burrows. Whatever type of nesting box is used, dimensions should allow an adult bird to stand comfortably.

For auklets, other than rhinoceros auklets, it is important to consider that they are most comfortable nesting in small, tight spaces. Smaller versions of any of the boxes described above can be used. If smaller auklets are housed with larger burrow-nesting birds, limiting the auklet burrow entrances to 7.62 cm x 7.62 cm (3 in. x 3 in.) should prevent larger birds from appropriating them. Another option is using cinder blocks. Blocks come with a variety of size openings, but openings from 12.7 cm x 12.7 cm (5 in. x 5 in.) to 7.62 cm x 12.7 cm (3 in. x 5 in.) should meet the needs of most auklet species. Cinder blocks can be laid down end-to-end to provide burrow lengths of between 0.61 to 1.52 m (2–5 ft). Blocks may also be stacked on top of each other and side-by-side to make a "condominium" style nesting area. Entrances to cinder block burrows can be partially blocked with rocks so auklets can barely squeeze inside. This promotes the privacy and security the parents need for chick rearing.

To simulate the entry tunnels of burrows used by wild nesting alcids, lengths of corrugated tubing or smooth PVC pipe can be run from the exhibit burrow entrance to a hole in the box. The length of tubing used can vary from 15.24 cm (6 in.) to 0.91 m (3 ft), and may have a bend or elbow. The nest boxes should be located behind an exhibit wall on a stable, level surface with easy access for staff to the check boxes. Entrances to the nest boxes and burrows should be blocked off when the last chick fledges until the start of the next breeding season. There should be a slight elevation leading out of the nest box such that if water gets in the tube during exhibit cleaning, it will pour back out rather than into the nest box.

It is important to provide substrate in the bottom of the nest boxes to prevent the eggs from rolling and being damaged. The types of substrates that can be used include cat litter, pine needles, aquarium sand, sheet moss, and Nomad™ matting. With the exception of the cat litter, all items used as substrates should be disinfected prior to use. Aquarium sand can be disinfected by immersion in an iodine based disinfectant solution, then rinsed and dried thoroughly. The sheet moss can be disinfected by freezing it for a minimum of 24 hours. Pine needles can be soaked for a minimum of 10 minutes in a 1% bleach solution, rinse thoroughly, and then completely dried (e.g., placed on a drying rack for two days). Cat litter should be the "dust free" non-clumping clay variety, if available, without deodorants or additives.

Some nesting material should be provided in the exhibit for the birds to bring into the burrows because this stimulates natural nesting behaviors. Materials that can be provided include: pine needles that are at least 7.62 cm (3 in.) in length, sheet moss, sphagnum moss, plastic/artificial grasses or other small plant material, collected feathers during molt, and dried grass. As with nest box substrates, it is important to disinfect all nesting materials prior to providing them to the birds.

8.5 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 members in AZA-accredited institutions are able to assist with the rearing of these offspring when deemed necessary.

Eggs that are left with the parents can be candled to determine fertility, and then any management decisions can be made at that time. Eggs can be pulled for artificial incubation because of poor parent incubation of eggs, health concerns for the incubating birds, poor nest location, or poor incubation history.

Most alcid species should be artificially incubated between 36.7–36.9 °C (98.1–98.5 °F) dry bulb and 27.2–27.5 °C (81–82 °F) wet bulb. This temperature range may need to be adjusted depending on type of incubator used, local humidity, number of eggs in the incubator, etc. Eggs should be moved to a hatcher (36.17 °C (97.1 °F) dry bulb, ~31 °C (88 °F) wet bulb) at external pip, at which time eggs should no longer be turned. If an internal pip is noticed during candling, an egg may be moved to a different rack of the incubator that is set to be non-turning. It is possible to begin checking for pips as early as four days prior to the hatch date. A pip-to-hatch interval for puffins, murres, and rhinoceros auklets is three to five days. This lengthy hatching period is not a cause for alarm. The pip-to-hatch interval for other alcids is two to three days. Once hatched, chicks should be allowed to dry in the hatcher. The yolk sac should be checked for absorption. If the yolk sac is not properly absorbed, the chick should be handled with extreme care. The umbilicus should be swabbed with a sterile povidone-iodine swab by rolling the swab gently over the area. Chicks should not be moved from the hatcher until they are dry and fluffy, approximately 12 hours after hatching.

Transporting eggs from the wild to an institution, or from institution to institution, can be completed successfully. The method used will depend on the stage of development of the egg at the time of transport and the anticipated duration of transport. Well-developed eggs (at least three quarters of the incubation period or more complete) and shorter travel times (a few hours or less) make for easier transport because temperature and humidity requirements are less critical. In this case, the eggs can maintain their own temperature for a while, so holding them in an insulated “cooler” box works well. Eggs should be cushioned during transport with disinfected towels, autoclaved recycled newspaper chips, clean bird seed, or sand. Eggs should not be in contact with one another because they can easily break during rough travel.

For eggs that are in their first or second trimester, or that require longer travel times, it is recommended to keep the temperature much closer to the ideal incubation temperatures of 37 °C (98.5 °F) by means of artificial heat. Several institutions have used “Dean’s Brooders”, which have a digital thermostat, and maintain a constant temperature. For more remote field settings, it may be necessary to use more primitive methods such as placing “hand warmer” heater packs into coolers. It is important to monitor the temperature of these coolers very frequently to prevent the eggs from getting too hot. Going above the ideal temperature can lead to embryo death. Any heat packs used should be well insulated from the nearest egg because the temperature close to the heat pack will likely be too high. If travel times are extended, the eggs should be exposed to fresh air because CO_2 is produced from the embryo, and air exchange should be allowed. If very dry conditions of travel are anticipated, it is important to maintain some humidity (75–80% relative humidity is ideal) by providing sterile, distilled water inside the incubator. A fresh pair of latex gloves or clean and dry hands should be used whenever the eggs are touched.

Fostering has been successful with burrow-nesting alcids. If eggs need to be removed from a pair of birds, they can be fostered to other pairs whose chicks’ hatch dates are within a week of their own. Chicks may need to be pulled for hand-raising if they do not gain weight at the expected rate or if the health of the parents or the chick is compromised. Indicators that the health of the chicks may be compromised include sunken, dull eyes, dehydration, dry fecals, insufficient fecal production, listlessness, trauma, and an unabsorbed yolk sac after three days. Parents can also be monitored for good rearing habits that include carrying fish to the chick, chick brooding, protecting the burrow entrance, and guarding the chick on ledges.

In many cases, it is desirable for parent-reared chicks to be introduced to keeper activity and feeding prior to fledging and integration into the general colony. This can be accomplished by removing the chicks from the nests for hand-raising just prior to their molt to juvenile plumage. Alternately, keeper activity can be introduced while chicks are still in the care of their parents. This allows for much of the acclimation process to be completed at the nest site. The following information in Table 15 represents an interaction and feeding schedule used for tufted puffins in the nest from an AZA facility:

Table 15. Partially hand-raised feeding schedule of Tufted Puffins

Age	Feeding schedule
Day 1–18	Parent fed, weigh every 7 days
Day 18–35	Feed 1–5 silversides twice daily in the nest to supplement parent feeding, weigh every 7 days
Day 35–50	Disallow parental access to the nest, feed up to 10 silversides four times daily, weigh twice weekly
Day 50+	Provide pool access, feed on the exhibit feeding schedule for adults

When hand-raising puffins, auklets, and guillemots, it is important to recreate a dark, warm, and secluded nesting area for the chicks to be raised in. This can be accomplished by artificially creating brooders. These brooders can be constructed in different ways that include Rubbermaid® tubs with holes drilled into the lid for ventilation, pet kennels, Dean's Brooders, and open-topped and open-bottom Plexiglas® frames with a small cardboard box inside to act as a "burrow". The open-topped and open-bottomed Plexiglas® frame should measure 40.64 cm (16 in.) wide x 83.82 cm (33 in.) long x 38.10 cm (15 in.) high. A small (30.48 cm x 30.48 cm (12 in. x 12 in) or less) cardboard box can act as a burrow when placed into the brooder. The box should have an opening cut in the front for a door, and a flap cut in the top for caretakers to observe the chick. If the cardboard is not waxed, the lower half of the box can be wrapped in duct tape on both the inside and outside (to aid in cleaning), but care should be taken to ensure that there are no exposed sticky areas where chicks may get their down stuck.

The brooders can be set above the ground on a stable surface like a table for easy access. Only one chick is recommended to be housed in each brooder. Heat can be provided by heat lamps placed above the brooder or heating pads placed under the brooder. In either case, it is important that the chick has the opportunity to move away from the heat source if it becomes overheated. Ice packs can be placed to one side in the brooder to offer a cooler spot for the birds to utilize if they want. The temperature in the brooder should be kept between 29.44–35 °C (85–95 °F) for the first five days. Temperature should then be lowered gradually over time and as needed to keep the chicks comfortable. Chicks that are heat stressed will gape, refuse food, hold wings and feet outstretched, and will feel hot. Chicks that are too cold will shiver, sit in a hunched position, refuse food, and be listless.

Suitable substrates for brooders include towels, Nomad™ matting, Dri-Dek®, and Solutia™ matting. The brooder and substrate should be thoroughly cleaned and disinfected on a regular basis to maintain a hygienic environment for the chick. As the chicks grow, they will need to be moved to larger brooders and then to the floor. In these large brooders, each chick should be provided with a burrow (e.g., Sky Kennels® without the door, fake hollow rocks with openings) to provide security and protection. Once the chicks have molted into their juvenile plumage, they should be given access to a pool.

Murres are ledge nesters, and they do not need hide boxes or enclosed brooders. Chicks can be raised in a suitable, well-ventilated brooder with a heat source. Unlike the burrow-nesting alcids, murre chicks can be kept together with more than one bird to a brooder. However, the chicks should have a safe place that they can retreat to, as the chicks can become aggressive toward one another.

At 15–25 days of age, parent-reared murre chicks will jump off the ledge into the water. The juvenile is unable to fly at this point. Parents will follow chicks into the water and then will guard and feed chicks for several weeks until the juvenile is independent and able to fly. If the exhibit design does not provide easy access to water, murre chicks may need to be pulled just prior to fledging for hand-rearing. When chicks are pulled from the exhibit, they will need to be placed in artificial brooders (see above). For older chicks, it may be necessary to place the food tray in the brooder and leave the area, allowing the chick to eat undisturbed.

Once they are ready, chicks can be moved to a larger holding area with more space to move around. The temperature should gradually be reduced for these older chicks, working toward an ambient temperature of 12.78–15.56 °C (55–60 °F). A proper substrate is important for all hand-rearing areas. Towels and netting should provide a soft surface during the early stages, and later on matting or Dri-Dek® (as above) should be used on flat surfaces on the floor or poolside. Appropriate substrates ensure that the chicks' feet do not develop calluses or hot/worn spots.

Chicks should not be fed until at least 12 hours post-hatch. Chicks should be fed and weighed in the brooder or bin to minimize handling. Bring fish in a petri dish and offer fish by picking up from dish and presenting to the chick (so the chick gets used to food coming from the dish). Chicks should respond by opening their bill and eagerly swallowing fish. After the first few days, the chicks should start picking up the fish on their own from the dish. The feeder may also try dropping the fish in front of the chick into the petri dish, simulating the motion from the parent's bill. Sometimes this needs to be done several times to get the chick's attention. The chick's crop should be checked prior to feeding to make sure that food is passing through properly. It is wise to check the fecals in the brooder and watch for signs of dehydration and to make sure food is processing well. On days one to four, feedings should occur every 2½–3 hours, for a total of six feedings per day. For days five through forty, feedings can be every 3 hours, for a total of five feedings per day. From around day forty until its release into exhibit, the chick can be fed every 4 hours, for a total of four feedings per day.

The amounts fed will depend on the chick's weight and the species, with typical morning weight gains between 10–15%. Newly hatched chicks should be hand-fed small, thin fish (e.g., silversides or sand lances) with the head, tail, and fins removed. Before each feeding, the chick's crop should be palpated for any undigested fish. If food is felt in the crop, no feeding should occur. Caretakers should wait 30 minutes and then check again. This protocol helps to prevent the prevalence of impacted crops. Indicators of an impacted crop include feeling a mass on the right side of a chick's neck that does not pass in a few hours, low appetite, and sour breath. In this case, it may be necessary to flush the chick's crop with water.

During the first two weeks of life, it may also be necessary to help maintain the chicks' hydration. This can be accomplished by offering fish injected with water or a 5% dextrose/lactated ringers solution for chicks with compromised health. The supplementation of chick diets with vitamins should not be started before the second day post-hatch; supplementation can begin between the third and seventh day of life.

Chicks can be allowed pool access once they have sufficient juvenile feathering as to be waterproof. See Figure 11 for more information about feather development of chicks. Pool introductions should be monitored to ensure chicks are waterproofed, buoyant, and can exit the pool safely. When handling chicks, clean hands (washed, disinfected, and dried) or gloves are important to prevent oil contamination of feathers.

Record keeping protocol should include, if available, the chick's weight gain, food intake, behavior, and medical exams. The parent's behavior should also be recorded.

When chicks are ready to be introduced into the colony, they should be monitored closely for signs of aggression, ease of getting in/out of water, ability to navigate the exhibit, food intake, and weight (if easy to weigh using scale training). There are different introduction methods that have been successful:

- Hard introduction: just putting the chick on exhibit and monitoring it closely
- Soft introduction: putting the chick on exhibit for limited amount of time and removing it each day. This method involves lengthening the amount of time the bird is on exhibit each day, leaving it on exhibit 100% of the time after several days.
- Howdy the bird on exhibit: Use a kennel or block part of the exhibit so that the chick can see the other birds on exhibit, but cannot get to them. Once the chick appears calm and not scared, there are no aggressive behaviors toward the chick from the other birds, and the chick seems eager to get to the exhibit, the chick can be released to the exhibit.
- Remove a few of the exhibit birds and introduce to the chick behind the scenes. Once they are all comfortable with each other, introduce all the birds back to the exhibit.

Regardless of the method used, careful monitoring should always be done.

Although no studies have been done with puffins, it is generally accepted that hand-reared puffins have the same success with breeding as parent-reared puffins. However, hand-reared birds tend to be less fearful of keepers, and this can cause hazards when working with them in an exhibit. For example, hand-reared birds that do not move out of the way of caretakers have a greater possibility of being accidentally stepped on. Hand-reared birds may also be aggressive in the water, and may be more likely to walk out of exhibit doors when people open them.

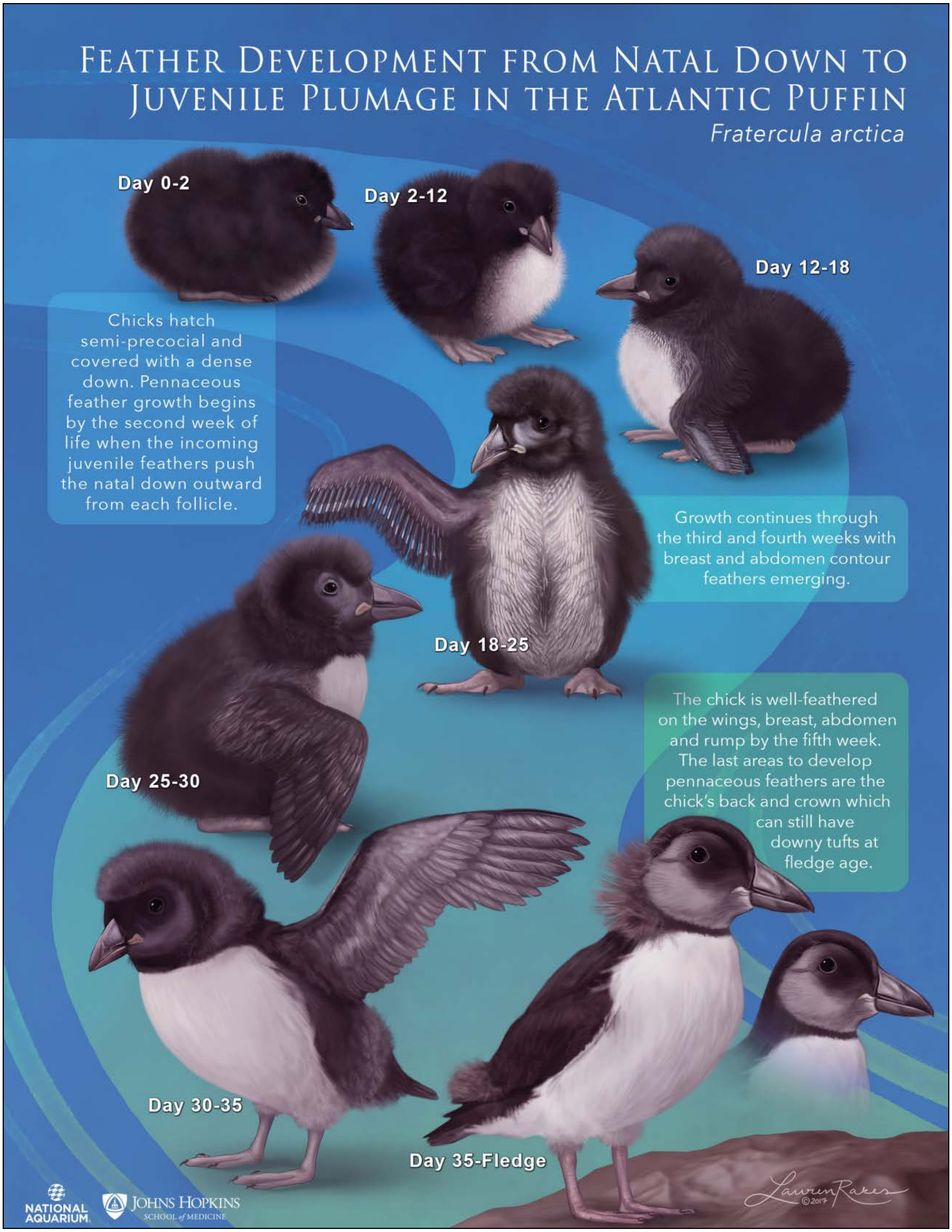

Figure 11. Feather development from natal down to juvenile plumage in the Atlantic puffin (Dial, 2017)

8.6 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. In the case of an animal on loan from another facility, consult the loan agreement or owner regarding authority to contracept. In the case of permanent contraception, prior permission of the animal's owner must be obtained.

The only current contraception method for Alcids is egg destruction. If an egg is removed, it is usually replaced with a dummy egg to discourage second clutching. The dummy egg should be warmed up in a keeper's hand before placing it underneath the bird. The dummy egg is then removed after the normal incubation period for that species. Any egg pulled should be examined to determine if it is fertile or not. In addition, the weight of the egg, the thickness of the shell, and any egg/embryo malformations should be noted. All eggs should be recorded, whether fertile or not, in support of fecundity information.

Eggs should be euthanized following the American Association of Zoo Veterinarians (AAZV) guidelines for egg euthanasia. They state that "avian embryos older than 50% gestation should be killed using decapitation, overdose of anesthetic, or other methods considered appropriate for hatched birds". Eggs that are less than 50% of gestation can be euthanized by through chilling, freezing, decapitation, overdose of anesthetic, or others method considered appropriate for hatched birds. When eggs are to be removed from nests to control reproduction, every effort should be made to remove these eggs before 50% gestation. Decisions on exactly when and how to remove eggs and related nest management needs (e.g. replacing with dummy eggs) should always be made with consultation or at least prior knowledge or understanding of the Supervisor, members of the work team, and/or Curator. If the exact percentage of gestation is unknown and can't be reliably determined by candling, err on the side of caution and have the egg euthanized by a zoo veterinarian.

Chapter 9. Behavior Management

9.1 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 (US) that evokes an innate, often reflexive, response. 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. Institutions should follow a formal written animal training program that facilitates husbandry, science, and veterinary procedures and enhances the health and well-being of the animals (AZA Accreditation Standard 1.6.4).

> **AZA Accreditation Standard**
>
> **(1.6.4)** The institution should follow a formal written animal training program that facilitates husbandry, science, and veterinary procedures and enhances the overall health and well-being of the animals.

Alcids have been successfully trained using operant conditioning techniques. They have been successfully bridge trained, often using either a whistle or clicker, and are most often reinforced with food like small fish or mealworms. Alcids have been successfully trained on targeting, scale, kennel, and a specially designed “foot box” that allows keepers to monitor the bottom of their feet. In addition, a few AZA facilities have used alcids in educational programs and have trained the following behaviors: step up (onto hand or platform), growl (make a puffin growling noise), off (either off the scale or off a hand or platform), wings (spread wings in a wing display), target (touch beak to a specific item), kennel (step from hand/platform/floor into a transportation kennel), water (leave from hand/platform and fly to the water), station (stand on a particular spot), gape (open mouth in a gaping gesture similar to the courtship behavior), straight (stand up straight on platform/hand), come/follow, hold (hold a dollar bill with the intent to have the bird drop it in a receptacle, or a paintbrush for painting), and called with name out of water or into back holding. See Chapter 10 for more details about program animal training. Overall, alcids are very responsive to operant training and there are opportunities to advance the field of training with this species.

9.2 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, stimulate cognition, and promote natural behaviors. Stimuli, including natural and artificial objects, scents, and sounds, are presented in a safe way for alcids 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 alcids should take into account the natural history of the species, individual needs of the animals, and facility constraints. The alcid enrichment plan should include the following elements: goal setting, planning and approval process, implementation, documentation/record-keeping, evaluation, and subsequent program refinement. The alcid enrichment program should ensure that all environmental enrichment devices (EEDs) are “alcid safe” and are presented on a variable schedule to prevent habituation. AZA-accredited institutions must have a

> **AZA Accreditation Standard**
>
> **(1.6.1)** The institution must follow a formal written enrichment program that promotes species-appropriate behavioral opportunities.

formal written enrichment program that promotes alcid-appropriate behavioral opportunities (AZA Accreditation Standard 1.6.1). Enrichment activities must be documented and evaluated, and the program should be refined based on the results, if appropriate. Records must be kept current (AZA Accreditation Standard 1.6.3).

Alcid 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 a specific paid staff member(s) assigned to oversee, implement, assess, and coordinate interdepartmental enrichment programs (AZA Accreditation Standard 1.6.2).

AZA Accreditation Standard

(1.6.3) Enrichment activities must be documented and evaluated, and program refinements should be made based on the results, if appropriate. Records must be kept current.

AZA Accreditation Standard

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

Behavioral management programs should include training, enrichment, exhibit design, and husbandry practices. They should aim to promote natural behaviors of alcids, such as swimming, diving, porpoising, rafting, roosting, courtship behaviors, bathing, preening, foraging, exploring, and other forms of exercise such as flapping wings, climbing, or flying. For a more detailed description of all natural behaviors, see Chapter 2. As bumblefoot is a common health problem for alcids in zoos and aquariums, special care should be given to promote swimming as much as possible. Ideas of how to encourage various natural behaviors of alcids are included below in Table 16.

Table 16: Natural behavior encouragement ideas for alcids

Behaviors	Ideas to encourage those behaviors	Misc. notes
Basking/sunbathing	Heat lamp or direct sunlight	Care should be given to not change the overall temperature of the exhibit and give choice to birds to not be in it.
Bathing	Encourage in water - see diving and swimming notes	
	Provide shallow pans of water	
	Showers/misters	Showers should be limited to a set amount of time as to not waste water. Birds should be given a choice to get out of the shower.
Breeding/copulation/incubation	Appropriate diets	
	Appropriate light cycles	
	Even sex ratio	
	Nest boxes	
	Recorded bird vocalizations	
Climbing	Exhibit design - multiple climbing areas that birds can get to easily	
	Place enrichment items up high	
	Scatter nesting material during breeding season	
	Target or station training	
Diving/swimming	Live invertebrates, fish, etc. in the water	Exhibit should be designed in a way that allows appropriate hiding areas for fish.
	Currents/water motion - can use devices for moving water,	

	Enrichment items placed along outside of exhibit, along glass by water	
	Enrichment items placed in water	Care should be given as to not get stuck in overflow/weir box.
	Feeding of live fish	
	Live/fake kelp or grasses	They should be designed so they can be moved around by staff to keep exhibit dynamic.
	Scatter feeding in water (by tossing or use of feeding ports)	
	Training behaviors such as cueing into water	
Exploring/observation/alert	Hand feeding and operant conditioning	
	Moving exhibit furniture around, such as driftwood recorded bird vocalizations	
	Use of enrichment items	
Foraging	Feeding a portion of their daily diet in the water	
	Feeding live fish	
	Food inside enrichment devices	
	Live insects scattered	
	Scatter nesting material during breeding season around entire exhibit	
	Use of food dishes on land - change the location around on a regular basis	
	Varied times of feed and varied food items	
Molting	Provide appropriate diet	
	Provide natural light cycle	
Nest building	Appropriate light cycle	
	Even sex ratio	
	Provide nesting material, such as pebbles, shells, plant material, etc.	
	Provide nests on exhibit	
Porpoising	Encourage in water as much as possible - see swimming and diving	
	Exhibit designed with appropriate water size	
Preening	Encourage in the water as much as possible	

	Even sex ratio Provide bathing opportunities
Rafting	Logs or other enrichment devices floating at the surface of the water
Resting	Exhibit design with some "neutral" areas away from nest boxes Varied substrate
Using beak to carry items	Ice cubes Nesting material Small enrichment toys Train a behavior

Training is an important component of behavioral management programs. For example, by scattering more of the food, hand-feeding should be reduced, and therefore, a bird's caloric intake cannot be monitored precisely. Thus, having the birds scale trained is an essential tool to help monitor the birds' health. In addition, training birds to enter the water on command, target training, and/or training item retrieval are all behaviors that could encourage more swimming and/or diving.

All enrichment items should be assessed for various safety issues, such as entrapment of body parts, trip hazards, anything that could cause foot problems, choking hazards, dietary health issues, etc. In order to avoid overfeeding, facilities can consider removing the reinforcing food from the primary diet, only offering it during training sessions. In addition, as aspergillosis is another common health problem for managed alcids, giving live plant material should be done carefully, and air sampling should occur to monitor air spore samples. All enrichment items should go through an appropriate approval process that considers safety, behavior, and exhibit aesthetics. In addition, the response to the enrichment items should be routinely monitored and recorded. One method that has been used successfully is to schedule enrichment on monthly calendars in order to make sure the types of enrichment, as well as the days, are variable. See Appendices L and M for examples of enrichment approval and evaluation forms.

9.3 Staff and Animal Interactions

Animal training and environmental enrichment protocols and techniques should be based on interactions that promote safety for all involved. Space considerations for training should be considered when designing exhibits. In order to facilitate animal training and enrichment for alcids, the exhibit should have a flat space large enough to accommodate a scale or crate, room for the keeper feeding the birds, and room for most of the birds to gather around the area. Overall exhibit design should aim to promote the behaviors listed in the table in Section 9.2.

The water area should be large enough for all birds to comfortably fit in the water at the same time. In addition, it should be long enough to promote porpoising behavior as well as deep enough to promote diving (see Chapter 1 for more details). Water motion is another important design feature to promote swimming. Rockwork can be designed to hide features such as the hydrowizard. Lightweight, fake, movable rocks can be designed to anchor enrichment items underwater. Feeding ports can be designed into the exhibit. Swim steps throughout the entire exhibit will allow for more access points the birds have into/out of the water. This will help encourage the use of the water. Encouraging swimming is important to help decrease the chance of bumblefoot.

Staff and alcid interactions do not usually require any sort of protected contact, special gloves, etc. during training sessions. However, if you are catching up a bird to restrain it, gloves should be worn to protect from scratches and bites as well as to protect the bird's feather from human oils.

9.4 Staff Skills and Training

Alcid staff members should be trained in all areas of alcid 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.

Keepers should have an understanding of basic alcid behavior and be able to distinguish signs of stress. They should be familiar with natural history of the species they are working with. They should also have an understanding of operant conditioning techniques. Keepers new to animal training should be mentored by an experienced trainer while working with the birds.

Chapter 10. Ambassador Animals

10.1 Ambassador Animal Policy

AZA recognizes many public education and, ultimately, conservation benefits from ambassador animal presentations. AZA's Conservation Education Committee's Ambassador (previously called Program) Animal Position Statement (Appendix F) summarizes the value of ambassador animal presentations. For the purpose of this policy, an ambassador 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.

Ambassador 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 ambassador animal presentations to develop an institutional ambassador animal policy that clearly identifies and justifies those species and individuals approved as ambassador animals and details their long-term management plan and educational program objectives. The policy must incorporate the elements contained in AZA's "Recommendations For Developing an Institutional Ambassador Animal Policy". If an animal on loan from another facility is used as an ambassador animal, the owner's permission is to be obtained prior to program use.

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). All record-keeping requirements noted previously apply to ambassador animals (AZA Accreditation Standards 1.4.1, 1.4.2, 1.4.3, 1.4.4, 1.4.5, 1.4.6, and 1.4.7). In addition, providing ambassador animals with options to choose among a variety of conditions within their environment is essential to ensuring effective care, welfare, and management (AZA Accreditation Standard 1.5.2.2). 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.

AZA Accreditation Standard

(1.5.4) If ambassador animals are used, a written policy on the use of live animals in programs must be on file and incorporate the elements contained in AZA's "Recommendations For Developing an Institutional Ambassador Animal Policy" (see policy in the current edition of the *Accreditation Standards and Related Policies* booklet). An education, conservation, and welfare message must be an integral component of all programs. Animals in education programs must be maintained and cared for by paid and/or unpaid trained staff, and housing conditions must meet standards required for the remainder of the animals in the institution. While outside their primary enclosure, although the conditions may be different, animal safety and welfare need to be assured at all times.

Using alcids as program animals is a relatively new practice, and, to date, only a few AZA institutions are using them for programs. This area could use further research. Program animals should be kept in the exhibit with the rest of the colony. If a bird has become difficult to work with, needs to be a part of programs at a consistent specific time, or is for any other reason deemed important by the aviculturists, the bird can be pulled from habitat and kept in a holding pen for a limited amount of time, at most a few days. More specific off-exhibit holding recommendations can be found in Chapter 2. As with any off exhibit holding areas, these should be big enough to encourage exercise, including diving, swimming, and walking. They should also have a haul out area large enough for each individual housed in the enclosure to have their own space. This helps minimize any aggressive and/or territorial behaviors. Any birds housed in a holding area together should be observed closely for signs of aggression and separated if aggression occurs. In order to prevent foot issues, a soft substrate such as 3M Nomad™ matting should be put over all haul out areas. An enrichment program that includes various different types of enrichment is also important, see chapter 9.2 for more information about enrichment. Only birds that are known to have an amiable relationship should be housed in a holding pen together.

All program animals, regardless of location in habitat or in holding, should be trained daily. Any birds pulled from habitat should be part of a more rigorous training program in order to promote mental stimulation and should be trained a minimum of twice a day.

Wild alcids occupy cold water marine habitats and are consequently extremely good at staying warm and can quickly become overheated. Their water should be kept as cold as possible at all times as should the ambient air temperature of their pen. See Chapter 1 for more information on appropriate temperatures. If necessary, ice packs should be placed in the enclosure with the bird in order to keep the space cool. Alcids are very susceptible to fungal infections such as aspergillosis. High humidity increases the risk of a bird succumbing to aspergillosis, so birds should be kept in low humidity as well.

10.2 Institutional Ambassador 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 ambassador animals are required to develop their own Institutional Ambassador Animal Policy that articulates and evaluates the program benefits (see Appendix G for recommendations). Ambassador 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 ambassador animal demonstration (AZA Accreditation Standard 1.5.3).

AZA Accreditation Standard

(1.5.3) If animal demonstrations are a part of the institution's programs, an educational/conservation message must be an integral component.

Every presentation is recommended to promote stewardship of marine ecosystems. Education animals should serve an important role in the presentation by leaving the audience with a deeper amazement with the natural world. Important messages that can be addressed during educational programs include invasive species (rats eat adults, eggs, and take over nesting burrows; introduced cattle and horses trample burrows), overfishing (can reduce fish populations, which are alcid prey items), and sport fishing next to rookeries (can result in birds being caught by fishermen; these birds are often killed or severely injured).

Horned puffins, tufted puffins, and rhinoceros auklets are recommended for use in animal programs. These species do, however, make very strong pair bonds and can develop strong bonds with particular keepers, so special attention needs to be made in order to prevent adverse behaviors. Common murres are not recommended. This species can easily develop leg and foot sores when kept in holding areas. They also have an extremely strong flock mentality during the entire year, and separating them from the flock is difficult and stressful for the individual. Pigeon guillemots can be used as juveniles and on habitat for short keeper chats. However, their use is not recommended because they are a difficult species to manage. They are naturally very aggressive during the breeding season which makes them a challenge to work with during that time.

Food should be offered as a reward after each behavior presented during a presentation. While it is appropriate to manipulate when a program animal is fed in order to promote an adequate response during a presentation, the bird should always be offered at least 20% of its body weight every day.

Most birds used for animal programs are hand-raised, but this is not essential. However, birds that are not hand-raised require a much greater time commitment to be involved in animal programs. Consequently, utilizing primarily hand-raised birds tends to be the preferred method of animal selection. In order to prevent imprinting, it is recommended to allow parents to raise the chick until two weeks of age, and then have the keepers finish rearing the chick off habitat. A training program should be implemented as soon as feasibly possible with newly hatched chicks that are targeted for training programs. When using a parent-reared bird, the birds selected have been ones that have shown an interest and curiosity in the keepers from a fledgling age. This initial curiosity can be used to start implementing a positive training program. Conversely, a bird that is hand-raised to be a program animal may no longer be a good candidate for a presentation program once it finds a mate and starts raising chicks. In such cases, it is feasible to retire education animals at a relatively young age, or only use them outside of the breeding season. It is important to remember that although birds are targeted for training programs, not all birds will respond positively to keeper involvement, and it is best to not include birds with unfavorable dispositions in training programs. Consequently, it is desirable to target slightly more chicks and fledglings for animal programs than the target number of program individuals.

The breeding season from May to September is an important time for alcids and includes seasonal hormonal and behavioral changes. If birds are pulled from the habitat during the breeding season, it is important to accommodate these changes and promote healthy behaviors. Any alcids pulled into back-holding during the breeding season should have access to a nest site. This helps to promote naturally occurring breeding behaviors. If these natural behaviors are not easy for a bird to express, the individual will often become very aggressive. If at all possible, birds should be paired during the breeding season and not in a holding pen by themselves. Only juveniles can be safely housed alone and still maintain program behaviors. If an adult individual is housed alone during the breeding season, the individual will typically court keepers excessively. This can lead to a bird who cannot participate in educational programs, because the individual will consistently court keepers throughout the presentation. The presence of another bird, even if they are not paired, helps minimize this behavior. Keepers should not respond to courtship behaviors from presentation animals at any time. This only reinforces the courtship behavior which can very rapidly spiral out of control. If program animals are allowed to parent incubate and rear young, they should be pulled from participation in animal programs through the duration of the incubation and chick rearing period.

Animal care and education staff should be trained in ambassador animal-specific handling protocols, conservation, and education messaging techniques, and public interaction procedures. Paid and/or unpaid staff assigned to handle animals during demonstrations or educational programs must be trained in accordance with the institution's written animal handling protocols. Such training must take place before handling may occur (Accreditation Standard 1.5.12). These staff members should be competent in recognizing stress or discomfort behaviors exhibited by the ambassador animals and be able to address any safety issues that arise. Additionally, when in operation, animal contact areas must be supervised by trained paid and/or unpaid staff (AZA Accreditation Standard 1.5.13).

AZA Accreditation Standard

(1.5.12) Paid and/or unpaid staff assigned to handle animals during demonstrations or educational programs must be trained in accordance with the institution's written animal handling protocols. Such training must take place before handling may occur.

AZA Accreditation Standard

(1.5.13) When in operation, animal contact areas (petting zoos, touch tanks, etc.) must be supervised by trained, paid and/or unpaid staff.

All animal care staff involved in animal programs should have a background in avian programs. The particular program should be memorized, observed at least three times, and the presenter should be competent at presenting before they're allowed to do the presentation themselves. Presenters should have a minimum of six months of experience working with a particular bird before they take it off habitat for a presentation. Animal care staff should also have an adequate understanding of operant conditioning and positive reinforcement through both hands-on experience and literary supplementation. Staff should have re-training sessions biannually.

The health of both the animals and humans involved in programs should be a primary concern at all times. At the start of any presentation involving a bird, a safety talk should be given to the public. The public should not be allowed to touch or handle birds. In the event that a bird jumps onto a member of the public, the person should be asked to stay still and not touch the bird and the keeper should remove the bird. The keeper should always be the only one physically interacting with the bird. Any injuries that occur to keepers or the public should be documented with an incident report and security should be notified. If an animal is injured, an incident report should be filed and vet staff should be notified. A program should always immediately end if the safety of any humans or animals is jeopardized.

Human food and drink should also be kept away from the animals. However, alcids do not tend to be drawn to human food and drink, so bringing a bird into a banquet setting for a presentation does not pose a danger to the bird. However, since puffins are burrow nesters that project fecal material at a high velocity to keep it away from the nest, care should be taken to keep the birds a safe distance from any food or drink to prevent contamination.

Any signs of stress should be dealt with immediately. The most common signs come from a bird uncomfortable with a particular situation. This can be from a variety of changes to its environment that happen during an animal program, including the sudden appearance of a large audience. A bird might stop eating, show an uneasy body posture, or try to jump back into a kennel. These signs of stress are important to notice and accommodate, but they are not an immediate danger to the bird. Still, every program experience should be kept as positive as possible in order to prevent a bird from refusing to participate in animal programs, so these signs of uneasiness should be avoided if possible. These signs can be avoided by practicing the program with your animal as much as possible and making each practice program a positive experience. Always end a training session with something positive. If these behaviors occur during a presentation, try to put the individual animal back into a safe, cool location as soon as appropriately possible. Another sign of stress is overheating. Indicators of overheating include panting, droopy wings, and hot feet. If a bird starts to become overheated during a presentation, it should be put back into a cool kennel. If the bird starts panting heavily, it should be put back into a cool, stress-free habitat as soon as possible.

Ambassador 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).

Program animals may have a greater risk of being exposed to disease and illness. The most predominant infectious diseases in alcids are aspergillosis, pox, influenza and other viruses, and bacterial infections such as Salmonella, staph, and other skin infections. Of these, influenza and Salmonella are the most likely zoonotic problems for program animals. Before and after handling the birds, the staff person should wash their hands. All crates, mats, etc. should be kept clean and disinfected after every use. Access to veterinary care is important, and all program animals should have access to an on-call veterinarian seven days a week. Animals should have a thorough health check annually by veterinary staff, including monitoring CBC and weight. Thorough health exams should be performed as needed if health concerns arise.

Careful consideration must be given to the design and size of all ambassador 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, 1.5.2, 1.5.2.1).

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).

Animals should always be taken off habitat or out of its enclosure using operant conditioning and positive reinforcement. The bird should be asked to station on a hand or platform while it is walked off habitat or to the presentation area or it should be asked into a kennel and enter the kennel on its own. Both of these options are behaviors that a bird can and should be trained to perform. No individual should be used more than ten hours a week, three hours at a time, including all kenneling and transportation time. One individual should not be used for more than two presentations lasting longer than one hour in a week. The bird should not be asked to perform other behaviors such as wing displaying or head tossing for more than ten minutes total per presentation. Animals should be rotated so that the presentation duties are shared among individuals.

Transportation kennels for program animals should be kept cool at all times as well. Ice packs and a trampoline should always be in the kennel and in warm environments or, if the bird is going to be spending a longer period of time in the kennel, the kennel itself should be frozen and filled with ice soaked towels. Any ice packs should be covered with a trampoline and 3M Nomad™ matting or a towel to protect the individual's feet. Birds should not be kept in kennels for longer than three hours, including holding time and presentation time. Each individual in a program should have its own kennel; kennels should not be shared. Windows and holes in kennels are important for air circulation, but they should be covered with a light towel to keep the bird calm. No other items should be placed in the kennel with the bird other than those necessary for a cool environment. Birds should be kept in temperatures that will have them exhibit normal behavior—not excessively hot and panting, excessively cold, or tucking beak or feet in feathers. It is recommended to keep them in temperatures close to their holding temperatures.

10.3 Program Evaluation

AZA-accredited institutions that have an Institutional Ambassador Animal Plan are required to evaluate the efficacy of the plan routinely (see Appendix G 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 revised biannually. They should be reviewed annually before the peak program season. The success of programs can be measured by the amount of revenue a program brings in, the number sold, and how engaged the audience is. The success of guest interpretations can be measured by visitor feedback sheets and how many visitors leave with the take home message intended.

In the event of a violation of animal program policies, animal care staff should be pulled from programs, and their animal handling privileges revoked. The individual should undergo a mandatory period of exclusion from participation in animal programs and training, the duration of which can be determined on an individual basis. When the break period is over, training should begin at the most basic level of training and program interaction, and only under supervision. When the animal care personnel demonstrates a high quality of animal training and program performance, the individual can then start participating in animal programs again.

The welfare of animals participating in animal programs should be reassessed on a constant basis. Weight and behavioral changes, including breeding behaviors, should be monitored closely. The introduction of any adverse behaviors is an indication that a bird is being overworked or not worked enough, and the case should be assessed accordingly. An animal's performance during a presentation is also an indication of its welfare within the program parameters. Any animal not performing well should be assessed and potentially pulled from programming until the individual is once again performing well. If an animal is unwilling to participate in a program three times in a row, it should be given a break from the animal program. Any signs of sickness or stress are reason to pull the bird from participation.

Chapter 11. Research

11.1 Known Methodologies

> **AZA Accreditation Standard**
>
> **(5.3)** The institution should maximize the generation and dissemination of scientific knowledge gained. This might be achieved by participating in AZA TAG/SSP sponsored studies when applicable, conducting and publishing original research projects, affiliating with local universities, and/or employing staff with scientific credentials.

> **AZA Accreditation Standard**
>
> **(5.0)** The institution must have a demonstrated commitment to scientific study that is in proportion to the size and scope of its facilities, staff (paid and unpaid), and animals.

AZA believes that contemporary alcid 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 in both *in situ* and *ex situ* settings to advance scientific knowledge of the animals in our care and enhance the conservation of wild populations. Participating in AZA Taxon Advisory Group (TAG) or Species Survival Plan® (SSP) Program sponsored research when applicable, conducting and publishing original research projects, affiliating with local universities, and/or employing staff with scientific credentials could help achieve this (AZA Accreditation Standard 5.3). An AZA institution must demonstrate a commitment to scientific study that is in proportion to the size and scope of its facilities, staff, and animals (AZA Accreditation Standard 5.0).

All record-keeping requirements noted previously apply to most research animals, especially those which are part of the exhibit collection. When an animal on loan to a facility is subject to an invasive research procedure, including when done as part of a routine health exam, the owner's prior permission is to be obtained.

Not much is known about the worldwide populations of seabirds. They inhabit remote and difficult to access areas of the world. Because of this, there is a limited amount of information on population densities, behavior, and physiology. This group of birds spends a big portion of their year living in the open ocean and only comes to land to breed. This makes it challenging to house and breed these birds in an *ex situ* setting. Zoos can add to the body of knowledge by studying *ex situ* behavior and documenting physiological data. Zoos can also contribute by providing staff and resources to assist in population surveys. The Charadriiformes TAG covers the alcid group. The tufted puffin and common murre are Green SSPs; the Atlantic puffin and the horned puffin are Yellow SSPs.

One major source of studies and funding to help seabirds come from the Oiled Wildlife Care Network (OWCN) headquartered at the University of California at Davis. This is a collection of trained wildlife care providers, regulatory agencies, and academic institutions that respond to oiled wildlife emergencies. The Monterey Bay Aquarium collaborates with the OWCN on improving oiled bird rehabilitation.

Another research project in progress is Project Puffin. It was started by Dr. Stephen Kress and the Audubon Society to restore puffins to historic nesting islands in Maine, mainly Eastern Egg Rock Island. Project Puffin staff and volunteers conduct research on population counts, chick growth, survival, movement, and diet.

The University of Alaska and the Cincinnati Zoo collaborated on the study of alloanointing behavior in crested auklets. Swarthmore College and the Aquarium of the Pacific cooperated to study the scent of individuals and how scent relates to social status in the crested auklet. Researchers at the University of Florida are investigating seabird viruses and their impact on health.

Scientists involved with alcids research:

- Hannahrose Nevins, Colleen Young, and Melissa Miller with the Marine Wildlife Veterinary Care and Research Center and the OWCN have studied the impact of thermography and temperature reporting transponders of oiled seabird rehabilitation.
- Dr. Stephen Kress with Project Puffin pioneered techniques using decoys and calls to encourage birds to re-establish breeding in former nesting sites.
- Dr. Hector Douglas of the University of Alaska at Fairbanks has studied the role of the tangerine scent of the Crested Auklet at the Cincinnati Zoo (Douglas, 2008).

- Dr. Julie Hagelin of Swarthmore College has studied the crested auklet both in a zoo setting and in the wild (Hagelin, Jones, & Rasmussen, 2003).
- Dr. Jim Wellehan and Dr. Tom Waltzek at the University of Florida are studying species-specific DNA viruses that infect seabirds during oil spills and may be useful in health assessment. This is in cooperation with the OWCN.

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.

As mentioned above, scientists affiliated with the Aquarium of the Pacific and the Cincinnati Zoo have studied the role of tangerine-scented pheromones in the crested auklet. These studies have physiological and behavioral components. The auklet research at the Aquarium of the Pacific involved discrimination training. The birds were taught to touch their beak to the tip of a Y-shaped stick. They received food for touching a colored tip and then for touching the scented tip. The birds were then presented with a Y-shaped stick having two ends with two choices for the testing paradigm.

The OWCN supports research that increases survival of oiled seabirds. They have supported research on seabird viruses that may infect seabirds in areas at risk for oil spills. These viruses could cause medical conditions that affect care during rehabilitation. This is an example of a pathology investigation.

There is limited information at this time on research testing paradigms for alcids and seabirds. Any research on these subjects would be useful.

AZA-accredited institutions are required to follow a clearly written research policy that includes a process for the evaluation of project proposals and identifies the types of research being conducted, methods used, staff involved, evaluations of the projects, animals included, and guidelines for the reporting or publication of any findings (AZA Accreditation Standard 5.2). Institutions must designate a qualified staff member or committee to oversee and direct its research program (AZA Accreditation Standard 5.1).

An Institutional Animal Care and Use Committee (IACUC) should be established within the institution if animals are included in research or instructional programs. The IACUC should be responsible for reviewing all research protocols and conducting evaluations of the institution's animal care and use.

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 Plans® (SSP) Programs.

> **AZA Accreditation Standard**
>
> **(5.2)** The institution must follow a formal written policy that includes a process for the evaluation and approval of scientific project proposals, and outlines the type of studies it conducts, methods, staff (paid and unpaid) involvement, evaluations, animals that may be involved, and guidelines for publication of findings.

> **AZA Accreditation Standard**
>
> **(5.1)** Scientific studies must be under the direction of a paid or unpaid staff member or committee qualified to make informed decisions.

See Appendix N for Sample Research Policy Form.

The research team should be comprised of a combination of a project principal investigator (PI), who is the main person publishing the results and applying for grants, permits, etc. The person who would be serving as the program manager is expected to work underneath the PI and oversee the data collection and analysis for the research being conducted, or oversee the personnel doing the work with a strong involvement in the research. They are expected to have basic husbandry knowledge of the research target species and minimally oversee husbandry staff to ensure research goals are met. The program manager works closely with the departmental curator to ensure animal welfare and basic husbandry practices are met. Institutions can also consult with outside researchers to support in-house data collection. Basically, it is the project manager's responsibility to ensure that research goals are met, data is sufficient, and animal welfare is a top priority. The manager then assembles a team of husbandry, data collection, and analyst personnel unique to the project and the personnel's expertise. All projects should be vetted through their IACUC committee and senior science staff, if available.

11.2 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 these areas will maximize AZA-accredited institutions' capacity for excellence in animal care and welfare as well as enhance conservation initiatives for the species.

There is a big need for information on alcids and seabirds. Animal keepers in a zoo or aquarium setting can fill many of these knowledge gaps. Listed are but a few of the topics that need more research:

- How these birds handle freezing temperatures when on land, since wild alcids spend most of their time in the open ocean (see Chapter 1.1)
- Species-specific vision data (see Chapter 1.2)
- The function of seasonal ornaments, spectral evaluation of visual displays related to mate choice, and spectral sensitivity in alcids (see Chapter 1.2)
- Evaluation of alcids' ability or need to synthesize Vitamin D (see Chapter 1.2)
- The importance of light provision for alcid neonates (see Chapter 1.2)
- Investigating correlations between the various light provision schemes and alcid health and reproductive outcomes, as well as finding the most effective light sources for broad-spectrum light provision that includes UVA (see Chapter 1.2)
- Information on the value of UVA versus UVB when it comes to bird health (see Chapter 1.2)
- Determining if birds housed in freshwater exhibits can be sent to a saltwater exhibit, since it is thought that pelagic birds kept in a managed setting for long periods of time without regular intake of salt can lose the ability to utilize salt water (see Chapter 1.3)
- The maximum safe level of coliforms in exhibit water (see Chapter 1.3)
- Air and water exchange rates specific to alcid enclosures (see Chapter 1.3)
- How alcids respond to large noise sources and how such noises affect other aspects of their lives (see Chapter 1.4)
- Whether or not alcid groups form a typical multigenerational group *in situ*, as this information would be useful for the management of *ex situ* populations (see Chapter 5.1)
- Social recommendations for alcids used in conservation and education programs, as use of these animals in programs is rather new (see Chapter 5.1)
- More information about the nutritional requirements of managed alcids, including supplementation needs and how supplementation affects coloration (see Chapter 6.1)
- Levels of vitamin A that are necessary versus what is excessive, as high levels may antagonize the uptake of other fat-soluble vitamins, even with vitamin E (see Chapter 6.3)
- The pathogenicity of alcid parasites; tapeworm and roundworm infections have been associated with intestinal lesions, but the clinical significance is unknown (see Chapter 7.3)
- Significance of Newcastle disease virus and infectious bursal disease (seen serologically in wild Charadriiformes) (see Chapter 7.6)
- Specific studies on the safety of Meloxicam for alcids (see Chapter 7.6)
- Information about hormonal and chemical changes, and associated tracking methods, to better manage alcid breeding (see 8.1)
- Use of artificial insemination with alcids; at this time, natural conception is the only type of breeding that has been achieved with alcids (see Chapter 8.2)
- Incubation temperature and humidity (wet bulb temp) for various alcid species (see Chapter 8.3)
- Hand-reared alcid breeding success versus breeding success of parent-reared alcids (see Chapter 8.4)
- Research to flesh out the specifics of the use of alcids as program animals (see Chapter 10.1)
- Information about research testing paradigms (see Chapter 11.1)
- Average life span of wild and managed alcids
- Cataract issues with auklet species in human care
- Energy requirement calculations for the species, or an appropriate model to encompass energy requirements for a range of ages (infant, juvenile, reproductive adult, senescent adult)
- Information about the impact of climate change on alcid species

- Using managed populations to help understand bonds between pairs and nest site fidelity could have strong implications on a wild population's ability to adapt to rapidly changing food sources and climate in a specific geographic location.
- Keepers can add to the body of knowledge by observing and recording the behaviors associated with breeding; even something as simple as noting what nest material a species uses can be helpful.
- Research is needed on incubation period, pip-to-hatch times, weight gains and losses, parental behavior, fledging times, and any hand-rearing formulas, tips, and techniques. (Brackett (2013) discussed how they were able to acclimate tufted puffins chicks to the exhibit, as well as get regular weights and have birds with no behavioral issues.)
- The Migratory Connectivity Project may be interested in using alcid species in human care to test geolocators.

Chapter 12. Other Considerations

12.1 Surplus Animals

All SSP species held by institutions should be reported to the SSP Program Leaders. The SSP Program Leader should be responsible for making the decision as to whether or not specific animals are to be included in the managed population (e.g., over-represented animals or animals beyond reproductive age). Those animals not included in the managed population should be considered surplus to the managed population, but records still must be maintained on them to the same degree as those in the managed population. Instances in which individual animals may not be part of the managed population:

- Over-represented individuals
- Individuals beyond reproductive age
- Inbred individuals
- Individuals of unknown (UNK or MULT) parentage
- Individuals on contraception

All AZA managed alcid populations (Atlantic puffins, horned puffins, tufted puffins, and common murres) follow the current SSP categorical status of Green or Yellow, looking to maintain genetic diversity in a zoo or aquarium setting. The species studbook keeper, SSP coordinator, and Population Management Center (PMC) population biologists work together in determining any exclusions from the Breeding and Transfer Plan for each species. Please see the above reasons for exclusion.

According to each SSP publication, recommended pairings are determined with consideration of mean kinship, change in population gene diversity, maximum avoidance of inbreeding, demographic goals, and the wants/needs of individual institutions in attempt to increase and/or maintain genetic diversity for as long as possible. The MateRx software program creates matrices that prioritize individual pairings using the Mate Suitability Index (MSI). Institutions housing these species are encouraged to follow the recommended MSI rankings in order to meet the demographic goals of the population. Each species studbook keeper and SSP coordinator is responsible for providing updated facility species information to the PMC in order to generate current MSI pairings and SSP publications.

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- Cyclop-Eeze®
- Dean's Brooders
- Dri-Dek®
- Lighting Systems: http://chameleongrowsystems.com/
- Luxim® http://luxim.resilient.lighting/
- Mazuri® Supplements
- Nomad™
- Plexiglas®
- ProVet Logic®
- Purolater®
- Sky Kennel®
- Solatube® http://www.solatube.com/
- Solutia™ Matting
- StellarNet, Inc. https://www.stellarnet.us/
- TRACKS
- Turtle Tiles™
- Vari Kennel
- Wildfire® Lighting http://www.wildfirefx.com/products/lighting/viostorm.html
- ZIMS
- Zoo Med® Laboratories https://zoomed.com/category/avian-products/lighting/

Appendix A: Accreditation Standards by Chapter

The following specific standards of care relevant to alcids are taken from the AZA Accreditation Standards and Related Policies (AZA, 2017) 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/provincial, and federal wildlife laws and/or regulations. It is understood that, in some cases, AZA accreditation standards are more stringent than existing laws and/or regulations. In these cases the AZA standard must be met.

Chapter 1

(1.5.7) The animals must be protected or provided accommodation from weather or other conditions clearly known to be detrimental to their health or welfare.

(10.2.1) Critical life-support systems for the animals, 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. Warning mechanisms and emergency backup systems must be tested periodically.

(1.5.9) The institution must have a regular program of monitoring water quality for fish, marine mammals, and other aquatic animals. A written record must be maintained to document long-term water quality results and chemical additions.

Chapter 2

(1.5.1) All animals must be well cared for and presented in a manner reflecting modern zoological practices in exhibit design, balancing animals' welfare requirements with aesthetic and educational considerations.

(1.5.2) All animals must be housed in enclosures which are safe for the animals and meet their physical and psychological needs.

(1.5.2.1) All animals must be kept in appropriate groupings which meet their social and welfare needs.

(1.5.2.2) All animals should be provided the opportunity to choose among a variety of conditions within their environment.

(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. AZA housing guidelines outlined in the Animal Care Manuals should be followed.

(10.3.4) When sunlight is likely to cause overheating of or discomfort to the animals, sufficient shade (in addition to shelter structures) must be provided by natural or artificial means to allow all animals kept outdoors to protect themselves from direct sunlight.

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

(11.3.1) All animal exhibits and holding areas must be secured to prevent unintentional animal egress.

(1.5.15) All animal exhibit and holding area air and water inflows and outflows must be securely protected to prevent animal injury or egress.

(2.8.1) Pest control management programs must be administered in such a manner that the animals, paid and unpaid staff, the public, and wildlife are not threatened by the pests, contamination from pests, or the control methods used.

(11.3.6) There must be barriers in place (for example, guardrails, fences, walls, etc.) of sufficient strength and/or design to deter public entry into animal exhibits or holding areas, and to deter public contact with animals in all areas where such contact is not intended.

(11.2.4) All emergency procedures must be written and provided to appropriate paid and unpaid staff. Appropriate emergency procedures must be readily available for reference in the event of an actual emergency.

(11.2.5) Live-action emergency drills (functional exercises) must be conducted at least once annually for each of the four basic types of emergency (fire; weather or other environmental emergency appropriate to the region; injury to visitor or paid/unpaid staff; and animal escape). Four separate drills are required. These drills must be recorded and results evaluated for compliance with emergency procedures, efficacy of paid/unpaid staff training, aspects of the emergency response that are deemed adequate are reinforced, and those requiring improvement are identified and modified. (See 11.5.2 and 11.7.4 for other required drills).

(11.6.2) Security personnel, whether employed by 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).

(11.2.6) The institution must have a communication system that can be quickly accessed in case of an emergency.

(11.2.0) A paid staff member or a committee must be designated as responsible for ensuring that all required emergency drills are conducted, recorded, and evaluated in accordance with AZA accreditation standards (see 11.2.5, 11.5.2, and 11.7.4).

(11.2.7) 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 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.

(11.5.2) All areas housing venomous animals must be equipped with appropriate alarm systems, and/or have protocols and procedures in place which will notify paid and unpaid staff in the event of a bite injury, attack, or escape from the enclosure. These systems and/or protocols and procedures must be routinely checked to insure proper functionality, and periodic drills (at minimum annually) must be conducted to insure that appropriate paid and unpaid staff are notified (See 11.2.5 and 11.7.4 for other required drills).

(11.5.1) Institutions maintaining venomous animals must have appropriate antivenin readily available, and its location must be known by all paid and unpaid staff working in those areas. An individual must be responsible for inventory, disposal/replacement, and storage of antivenin.

Chapter 3

(1.4.0) The institution must show evidence of having a zoological records management program for managing animal records, veterinary records, and other relevant information.

(1.4.6) A paid 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 paid and unpaid animal care staff members apprised of relevant laws and regulations regarding the institution's animals.

(1.4.7) Animal and veterinary records must be kept current.

(1.4.4) Animal records, whether in electronic or paper form, must be duplicated and stored in a separate location. Animal records are defined as data, regardless of physical form or medium, providing information about individual animals, or samples or parts thereof, or groups of animals.

(1.4.5) At least one set of the institution's historical animal and veterinary records must be stored and protected. Those records should include permits, titles, declaration forms, and other pertinent information.

(1.4.1) An animal inventory must be compiled at least once a year and include data regarding acquisition, transfer, euthanasia, release, and reintroduction.

(1.4.2) All species owned by the institution must be listed on the inventory, including those animals on loan to and from the institution.

(1.4.3) Animals must be identifiable, whenever practical, and have corresponding ID numbers. For animals maintained in colonies/groups or other animals not considered readily identifiable, the institution must provide a statement explaining how record keeping is maintained.

Chapter 4

(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 laws and/or regulations must be adhered to.

(1.5.10) Temporary, seasonal and traveling live animal exhibits, programs, or presentations (regardless of ownership or contractual arrangements) must be maintained at the same level of care as the institution's permanent resident animals, with foremost attention to animal welfare considerations, both onsite and at the location where the animals are permanently housed.

Chapter 6

(2.6.2) The institution must follow a written nutrition program that meets the behavioral and nutritional needs of all species, individuals, and colonies/groups in the institution. Animal diets must be of a quality and quantity suitable for each animal's nutritional and psychological needs.

(2.6.1) Animal food preparation and storage must meet all applicable laws and/or regulations.

(2.6.3) The institution must assign at least one paid or unpaid staff member to oversee appropriate browse material for the animals (including aquatic animals).

Chapter 7

(2.1.1) A full-time staff veterinarian is recommended. In cases where such is not necessary because of the number and/or nature of the animals residing there, a consulting/part-time veterinarian must be under written contract to make at least twice monthly inspections of the animals and to respond as soon as possible to any emergencies.

(2.1.2) So that indications of disease, injury, or stress may be dealt with promptly, veterinary coverage must be available to the animals24 hours a day, 7 days a week.

(2.0.1) The institution should adopt the *Guidelines for Zoo and Aquarium Veterinary Medical Programs and Veterinary Hospitals,* and policies developed or supported by the American Association of Zoo Veterinarians (AAZV). The most recent edition of the medical programs and hospitals booklet is available at the AAZV website, under "Publications", at http://www.aazv.org/displaycommon.cfm?an=1&subarticlenbr=839, and can also be obtained in PDF format by contacting AZA staff.

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

(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. Quarantine duration should be assessed and determined by the pathogen risk and best practice for animal welfare.

(2.7.3) Quarantine, hospital, and isolation areas should be in compliance with standards/guidelines contained within the *Guidelines for Zoo and Aquarium Veterinary Medical Programs and Veterinary Hospitals* developed by the American Association of Zoo Veterinarians (AAZV), which can be obtained at: http://www.aazv.org/displaycommon.cfm?an=1&subarticlenbr=839.

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

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

(2.5.1) Deceased animals should be necropsied to determine the cause of death for tracking morbidity and mortality trends to strengthen the program of veterinary care and meet SSP-related requests.

(2.5.2) The institution should have an area dedicated to performing necropsies.

(2.5.3) Cadavers must be kept in a dedicated storage area before and after necropsy. Remains must be disposed of in accordance with local/federal laws.

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

(2.0.3) Institutions should be aware of and prepared for periodic disease outbreaks in wild or other domestic or exotic animal populations that might affect the institution's animals (ex – Avian Influenza, Eastern Equine Encephalitis Virus, etc.). Plans should be developed that outline steps to be taken to protect the institution's animals in these situations.

(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 animals at the institution from exposure to infectious agents.

(11.1.3) A tuberculin (TB) testing/surveillance program must be established for appropriate paid and unpaid staff in order to assure the health of both the paid and unpaid staff and the animals.

(2.3.1) Capture equipment must be in good working order and available to authorized, trained personnel at all times.

(2.1.3) Paid and unpaid animal care staff should be trained to assess welfare and recognize abnormal behavior and clinical signs 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, animal care staff (paid and unpaid) must not diagnose illnesses nor prescribe treatment.

(2.3.2) Institution facilities must have radiographic equipment or have access to radiographic services.

(1.5.8) The institution must develop and implement a clear and transparent process for identifying, communicating, and addressing animal welfare concerns from paid or unpaid staff within the institution in a timely manner, and without retribution.

Chapter 9

(1.6.4) The institution should follow a formal written animal training program that facilitates husbandry, science, and veterinary procedures and enhances the overall health and well-being of the animals.

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

(1.6.3) Enrichment activities must be documented and evaluated, and program refinements should be made based on the results, if appropriate. Records must be kept current.

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

Chapter 10

(1.5.4) If ambassador animals are used, a written policy on the use of live animals in programs must be on file and incorporate the elements contained in AZA's "Recommendations For Developing an Institutional Ambassador Animal Policy" (see policy in the current edition of the *Accreditation Standards and Related Policies* booklet). An education, conservation, and welfare message must be an integral component of all programs. Animals in education programs must be maintained and cared for by paid and/or unpaid trained staff, and housing conditions must meet standards required for the remainder of the animals in the institution. While outside their primary enclosure, although the conditions may be different, animal safety and welfare need to be assured at all times.

(1.5.3) If animal demonstrations are a part of the institution's programs, an educational/conservation message must be an integral component.

(1.5.12) Paid and/or unpaid staff assigned to handle animals during demonstrations or educational programs must be trained in accordance with the institution's written animal handling protocols. Such training must take place before handling may occur.

(1.5.13) When in operation, animal contact areas (petting zoos, touch tanks, etc.) must be supervised by trained, paid and/or unpaid staff.

Chapter 11

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

(5.0) The institution must have a demonstrated commitment to scientific study that is in proportion to the size and scope of its facilities, staff (paid and unpaid), and animals.

(5.2) The institution must follow a formal written policy that includes a process for the evaluation and approval of scientific project proposals, and outlines the type of studies it conducts, methods, staff (paid and unpaid) involvement, evaluations, animals that may be involved, and guidelines for publication of findings.

(5.1) Scientific studies must be under the direction of a paid or unpaid staff member or committee qualified to make informed decisions.

Appendix B: Recordkeeping Guidelines for Group Accessions

Developed by the AZA Institutional Data Management Scientific Advisory Group
Published 23 May 2014
Edited to replace the document entitled "Updated Data Entry for Groups" published 16 December 2002

Animals can be accessioned into a collection as either individuals or as part of a group. The term "group" has many definitions when used in zoos and aquariums, and is usually defined by its application, such as a social group or animals grouped for husbandry purposes. To provide a consistent language that can be used throughout the Association of Zoos and Aquariums (AZA), the term "group accession", as defined by the AZA Institutional Data Management Scientific Advisory Group (IDMAG),

- contains multiple animals of the same species or subspecies, which
- cannot be differentiated from one another, either physically (there are no scars or color pattern differences), artificially (they are not tagged or transpondered), or spatially (they are not held in separate enclosures), and
- are cared for as a whole.

Thus, no individually accessioned animals are included in a group accession and no individually *identifiable* animals are included in a group accession. As soon as an animal becomes individually identifiable, it is recommended that it be split from the group record and accessioned as an individual. For example, large clutches of amphibian tadpoles should first be accessioned as a group; then as individuals become identifiable, they should be removed from the group record and accessioned as individuals. Otherwise, information about an individual animal that could otherwise be tracked through the animal's life will be lost in the group record. An exception to this occurs occasionally when a group member is removed and temporarily held separately for medical treatment, with the expectation that it will be returned to the group when treatment ends. In this case, the animal remains part of the group even though separated from it. As with individual records, group record accession numbers should not duplicate any other accession number, and once a group accession number has been assigned, it should not be changed.

Group accession provides less information on specific individuals than does individual accession. Group records make information less retrievable, and often need more clarifying comments than individual records. Whenever information applies to only part of the group, notes should be used to indicate which animal(s) the information applies to. It is of utmost importance that these notes be thorough and clear so future readers can easily understand them. Examples of information needing additional notations in group records include, but are not limited to, parentage when not every member of the group has the ˘the group. Thus, though it is preferable to accession animals as individuals, a group accession can capture considerable information when individual accession is not appropriate.

Although colonies are often confused with groups, the term "colony" should be used to designate truly colonial organisms: those that must live and function as an intact unit, such as corals and eusocial insects. Individuals within a colony are components of a single entity rather than separate members of a group. Also, colony members generally cannot be counted and true census data is not possible, so for the purposes of inventory, a colony is a singular unit while a group is composed of a number of individuals. However, for accessioning purposes, colonies are treated in the same manner as are groups.

Examples of Appropriate Group Accessions

- A group of animals that are not individually identifiable and are the same species or subspecies.

  Your institution receives 50 Puerto Rican crested toad tadpoles to rear. Unless each tadpole is raised in a separate numbered tank, there is no way to tell one tadpole from another. All tadpoles housed together are accessioned as one group.
- Colonial species, such as coral or eusocial insects (e.g., some species of bees or ants).

  Your institution receives a piece of coral. Since the coral is in one piece, you accession it as a group of one. You make a note of the dimensions or mass of the piece to give an estimate of colony size, since it is not possible to count individual animals in the colony. In the inventory, the colony counts as one animal. When a section of the coral breaks off, you accession that new piece as a new colony.

- A self-sustaining, breeding group of small rodents or insects.

  Your institution has a large number of Cairo spiny mice. No daily count is made, though births and deaths increase and decrease the count. A census is taken periodically, and the new count is recorded by sex and life stage. Exact counts are made whenever possible – for example, when the group is moved to a new enclosure.

- Young born to several females of the same species or subspecies and raised together without means of identifying which offspring were born to which mother.

  A flock of 3.6 peafowl raise 25 chicks this year. Identity of the hens incubating each nest, hatch dates, and number of chicks hatched from each nest can be determined and recorded. However, unless the chicks are caught and banded at hatching, once the mothers and chicks join the main flock, it is no longer possible to tell which chicks belong to which females. All chicks in the flock have the same possible parents: all the peacocks and those peahens that incubated the nests. The chicks are accessioned as a group and are split out only when they are banded or tagged (and are thus individually identifiable).

- Historical records for a species or subspecies for which there is insufficient information to attribute events to specific individuals.

  Some of your historical records are found as simple lists of events. Though there are dates for all transactions, and maybe even specified vendors or recipients for those events, you cannot create individual records for any of these animals without additional information: there is nothing connecting any specific individual to both acquisition and disposition information. If additional information is uncovered that makes this connection, then that individual can be removed from the group accession and given an individual record.

Managing Group Records

Maintaining Group Records - As with individual records, group records should also be maintained and updated. Addition of animals through births or transactions such as loans, purchases, donations, or trades are entered as acquisitions. Subtraction of animals through deaths or transactions such as loans, sales, donations, or trades are entered as dispositions.

Weights and lengths can be entered into a group record even if that data cannot be attributed to a specific individual. This information is still useful in describing the overall condition of group members, although care should be given to describe the animal that the measurement came from. For example, is the animal a juvenile or a breeding adult? Is it healthy, or sickly? Alternatively, average and/or median measurements can be entered into the record to give an indication of what size a "normal" individual might be. In this case, notes should include the maximum and minimum measurements, and how many animals were measured to calculate the average or median.

Censuses - Groups should be censused at regular intervals - ideally, no longer than one inter-birth interval. Institutions should establish and follow a census schedule for each group. An inventory must be done at least once yearly (AZA Accreditation Standard 1.4.1) but the frequency at which a group is censused depends on species biology, husbandry protocols, and animal welfare. For species in which births/hatches and deaths tend to go undetected, or for species that have high fecundity and mortality (which makes counting every animal very difficult or impossible), census data should be obtained more frequently than for species with longer inter-birth intervals. These more frequent censuses should not be undertaken when intrusion on the group has a negative effect on the welfare of the group, e.g., disruption of maternal care.

Censuses should provide as much detail as possible by recording numbers in distinctive life stages (such as newborn, immature, adult) and/or sex ratio (such as male, female, unknown/undetermined). If the census count is estimated, the estimation method and (when possible) the accuracy of the estimate should be included. When updating the sex ratio, who sexed the animals and how they were sexed should also be recorded.

Splitting And Combining (Merging) Groups - Splitting animals from groups and combining groups together are realities of group management. Animals may be removed to create additional groups, or perhaps new animals are received from another institution. When new groups are created, new group records also need to be created. However, if the entire group moves to a new location (such as a different tank), it retains the same accession number, and notation of the change in location is made.

When a single group is split into two or more groups, one of the new groups keeps the original accession number and the others are assigned new accession numbers. This is also true if a portion of a group is sent to another institution: the subgroup making the transfer must have an accession number distinct from that of the main group. The accession number(s) for the new group(s) should follow institutional procedures for the assignment of new accession numbers. Note of the new group accession number(s) should appear in the originating group record, and the new group accession record(s) should contain the originating group number. The reason for the split should be entered into both the originating and new group records.

When two or more groups combine to form a larger group, all but one of the groups are deaccessioned and their counts brought to zero. Notes in all the group records should indicate why the groups were merged, as well as the accession numbers of all groups involved – both the closed (empty) groups and the remaining group.

In all cases of splits and merges, the date of creation of the new record should be the same as the date of removal from the previous group or individual. Detailed notes should explain the reasons for all splits and merges.

<u>Merging Individuals Into Groups and Splitting Individuals From Groups</u> - Good husbandry dictates the use of identification methods that allow animals to be tracked as individuals whenever possible (AZA Accreditation Standard 1.4.3). Thus, most institutions initially accession newly-acquired animals as individual animals with individual identifiers.

Despite the best intentions, individual identification sometimes becomes impossible. For example, birds in large aviaries lose their bands; small frogs in a large terrarium die and decompose without being noticed. When individual identification of several of the animals in the group is lost and can't be resolved in a reasonable amount of time, it is best to move all potentially unidentifiable animals to a group record, by either creating a new group or merging them into an existing group. As with splitting and merging groups, the group record should contain the identities of the originating individuals and the individual records should show the new group identity. If the animals in the group ever become individually identifiable again, they can be split back to individual records to better capture demographic information. If this occurs, new accession numbers are generally needed for the new individual records since it is rarely possible to know which old individual record would apply to the newly identifiable group member.

Conversely, if one or more group members become identifiable, for example, the previously unbanded young of the year are caught up and banded, they should be split from the group record and given individual accessions. The group record should include the individual numbers assigned, and the records of all individuals should show the number of the originating group. In the case of new individual records, information particular to the animal being given the individual record (if known) should be transferred to the individual record. This includes birth date, origin, parent identification, etc. As in the cases of splitting and merging groups, the date of creation of the new record is the same as the date of removal from the previous group or individual, and detailed notes should explain the reasons for all changes in accession type.

<u>Transfers Between Institutions</u> - When accessioning a number of animals that were received from another institution, the new animals should be accessioned using the same type of record that the sending institution used, regardless of how the animals will ultimately be managed. If a group is received but the members will be managed as individuals, they should be accessioned as a group first, then split out as individuals. Similarly, if a number of individuals are received but the plan is to manage them as a group, they should be accessioned as individuals, then merged into a group. Although this is an extra step in the accession process, it allows the records from both institutions to more seamlessly link.

<u>Removing Individuals From Historical Group Records</u> - The decision of whether to use individual or group accession for historical records should be made thoughtfully and carefully. As detailed above, group accession should be used if there is insufficient information to create an *accurate* individual record. The

use of group accession is preferable to the inclusion of “best guess” information, i.e. fiction, to fill the information necessary to complete an individual record.

If additional information is later found that allows the creation of an individual record for one of the members of a historical group record, the procedure for removal from the group is different from that for current records. This situation is treated differently because the historical individual was not truly part of a group accession – the information necessary for a complete individual record was merely not known and the group accession was used “temporarily” until the required information was found or learned. For this reason, the individual should NOT be split from the group, but all reference to the individual should instead be *deleted entirely* from the group, as if it were never part of the group. This will allow the individual record to begin with the initial acquisition (instead of the date of removal from a group) and will include the animal’s entire history in one record. It also prevents inflation of inventory numbers by eliminating the possible duplication of the same information in both the group and the individual records.

Appendix C: Guidelines for Creating and Sharing Animal and Collection Records

Developed by the AZA Institutional Data Management Scientific Advisory Group
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The goal of maintaining a centralized, compiled record for each animal cared for in a zoo or aquarium is ideal, however, oftentimes, information belonging in an animal record is spread across many departments and may originate with any member of the animal care staff. Therefore, it is important for zoos and aquariums to have a formal method for collecting or linking various pieces of information into the official records and that the roles and responsibilities for each named record type are clearly defined in written protocols for the reporting, recording, distribution, storage, and retrieval processes; there should also be a stated process of review for the accuracy and completeness of these records. For example, a recording/reporting protocol would state who reports births or deaths, to whom they are reported, in what manner and in what time frame they are reported, who officially records the information, and who reviews the resulting record for accuracy and completeness. Then, the maintenance and archiving protocol would state where the record is to be filed, who may have access, and how long the record is to be maintained before being archived or disposed of.

Information contained in animal records is essential not only to the immediate care of the individual animal but also as pooled data to manage larger concerns (e.g., providing norms for species-related veterinary and population management decisions, evidence of compliance with laws and regulations, showing trends in populations on every level from institutional to global, etc.). No matter what its use, it is critical for the information contained in an animal record to be factual, clear, complete, and documented. Because zoos and aquariums vary greatly in size and organizational structure, it is impossible to set defined procedures that would be applicable to all; therefore the following guidelines for creating and sharing animal records have been developed to assist with the establishment of written policies that best fit their own internal structure and protocols.

Animal and Collection Records – Definitions and Examples

The AZA Institutional Data Management Scientific Advisory Group (IDMAG) defines an animal record as: *"data, regardless of physical form or medium, providing information about individual animals, groups of animals, or samples or parts thereof".* An animal's record may include, but is not limited to, information about its provenance, history, daily care, activities, and condition; some may originate in non-animal care departments. Some examples of animal records are:

- transaction documents (including proof of legal ownership, purchase contracts, etc.)
- identification information
- reports of collection changes (including in-house moves)
- pedigrees/lineages
- veterinary information, including images, test results, etc.
- nutrition and body condition information
- information on sampling and parts/products distribution

In addition, the IDMAG defines collection records as: *"information, evidence, rationalizations about an animal collection as a whole that may supplement or explain information contained in an animal record"*. Collection records may include, but are not limited to, documentation of collection decisions and changes, evidence of structural change at the institution, evidence of building name changes, and documentation of institution level or unit level husbandry protocols and changes. Some examples of collection records are:

- collection plans
- permits
- annual inventories (which include reconciliation with the previous year)
- area journals/notebooks (including information to/from/between other animal care staff)
- keeper reports

- animal management protocols (e.g., species hand-rearing protocols, special care or treatments, etc.)
- enclosure maps/trees
- enclosure/exhibit information (monitoring, maintenance, modifications, etc.)
- research plans and published papers

Animal and Collection Records - Development

It is recommended that each zoo and aquarium develop written policies and procedures, applicable to all staff involved with animal care, that:

- define the types of records that are required.
  For example, daily keeper reports might be required from the keeper staff and weekly summaries of activities might be required from the animal curator and senior veterinarian.
- define the information that is to be included in each type of record.
  Following the example above, the institution would state the specific types of information to be recorded on the daily keeper report and the weekly summaries.
- define the primary location where each record can be found.
  For example, if a zoo does not employ a nutritionist, the policy or procedures might state that animal diet information will be found in keeper daily reports, curator-developed daily diets, and/or veterinarian-prescribed treatment diets.
- assign responsibility for the generation of each record type and set time limits for the their creation.
  For example, keepers might be held responsible for producing daily reports by the start of the next day and curators might be held responsible for producing weekly summaries by the Tuesday of the following week.
- define a process to review the accuracy of each record type and assign responsibility for that review process.
  For example, the identity of who will review each type of record, the date of reviews, and the review/correction processes might be included in the policy.
- define a process to identify official records and assign responsibility for the recording of, or linking of, information into these records.
  For example, the identity of who will be responsible for placing information into the official records and the processes of how to identify official records might be included in the policy.
- ensure entries in official records are never erased or deleted.
  For example, if an entry is determined to be erroneous, rather than deleting it, the entry should be amended and an audit trail should be created that identifies what data was changed, who made the change, the date it was changed, and the reason for the change.
- ensure records relating to specific animals in the collection, including the records of non--animal care departments, are permanently archived as part of the animal's record.
  For example, if your zoo or aquarium's records retention schedules differ from this recommendation every attempt should be made to exempt these records from schedules requiring their destruction.

Animal and Collection Records – Sharing of Information

Each zoo and aquarium should assess the ownership of their animal and collection records and determine the rights of employees and outside entities to the information contained in them. It is recommended that each zoo and aquarium develop written policies and procedures for the distribution and/or availability of the animal and collection records that:

- identify who has access to animal and collection records and under what conditions.
  For example, animal care staff whose duties require a direct need for information about specific animals or collection of animals should be identified as individuals who are allowed access to any or specified records, regardless of who created them or when they were created.
- assign responsibility for the distribution, archiving and retrieval of each record type.

For example, the recordkeeper or registrar might be held responsible for maintaining all past and current transaction documents and the curator might be held responsible for maintaining the daily keeper reports from his/her section.

- define a notification system that specifies what information will be provided in the notification, who will be notified, the date they will be notified by, and the mechanism that will be used to ensure the notification is communicated appropriately.

  For example, the shipment of an animal might require that written notice be made to the senior keeper in the animal's area, the curator, and the veterinarian at least 30 days prior to the move, and identifies the animal by group or individual identification/accession number, sex, and tag/transponder number, etc.
- define where each record type (stored or archived) is available and what format (paper or digital) it is in.

  For example, all original animal transaction documents might be kept in the registrar's office in fire-proof file cabinets but copies of the Animal Data Transfer Forms are kept in the appropriate keeper area.
- define a system for obtaining necessary information such that the information is available regardless of department and regardless of staffing issues

  For example, keeper daily reports might be maintained in an electronic database run on the institution's network, to which all animal care staff members have at least read-only access.

Implementation of these Recommendations

Well-written, consistent data-recording protocols and clear lines of communication will increase the quality of animal records and should be implemented by all institutions, regardless of technical resources. While the best option for availability of information is an electronic database system run on a computer network (intranet) to which all animal care staff members have unrestricted access, the above recommendations may also be adopted by zoos and aquariums without full electronic connections.

Appendix D: AZA Policy on Responsible Population Management

PREAMBLE

The stringent requirements for AZA accreditation, and high ethical standards of professional conduct, are unmatched by similar organizations and far surpass the United States Department of Agriculture's Animal and Plant Health Inspection Service's requirements for licensed animal exhibitors. Every AZA member must abide by a Code of Professional Ethics (https://www.aza.org/Ethics/). In order to continue these high standards, AZA-accredited institutions and certified related facilities should make it a priority, when possible, to acquire animals from and transfer them to other AZA member institutions, or members of other regional zoo associations that have professionally recognized accreditation programs.

AZA-accredited institutions and certified related facilities cannot fulfill their important missions of conservation, education, and science without live animals. Responsible management and the long-term sustainability of living animal populations necessitates that some individuals be acquired and transferred, reintroduced or even humanely euthanized at certain times. The acquisition and transfer of animals should be prioritized by the long-term sustainability needs of the species and AZA-managed populations among AZA-accredited and certified related facilities, and between AZA member institutions and non-AZA entities with animal care and welfare standards aligned with AZA. AZA member institutions that acquire animals from the wild, directly or through commercial vendors, should perform due diligence to ensure that such activities do not have a negative impact on species in the wild. Animals should only be acquired from non-AZA entities 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.

I. INTRODUCTION

This AZA Policy on Responsible Population Management provides guidance to AZA members to:

1. Assure that animals from AZA member institutions and certified related facilities are not transferred to individuals or organizations that lack the appropriate expertise or facilities to care for them [*see taxa specific appendices (in development)*],

2. Assure that the health and conservation of wild populations and ecosystems are carefully considered as appropriate,

3. Maintain a proper standard of conduct for AZA members during acquisition and transfer/reintroduction activities, including adherence to all applicable laws and regulations,

4. Assure that the health and welfare of individual animals is a priority during acquisition and transfer/reintroduction activities, and

5. Support the goals of AZA's cooperatively managed populations and associated Animal Programs [Species Survival Plans® (SSPs), Studbooks, and Taxon Advisory Groups (TAGs)].

This AZA Policy on Responsible Population Management will serve as the default policy for AZA member institutions. Institutions should develop their own Policy on Responsible Population Management in order to address specific local concerns. Any institutional policy must incorporate and not conflict with the AZA acquisition and transfer/transition standards.

II. LAWS, AUTHORITY, RECORD-KEEPING, IDENTIFICATION AND DOCUMENTATION

The following must be considered with regard to the acquisition or transfer/management of all living animals and specimens (their living and non-living parts, materials, and/or products):

1. Any acquisitions, transfers, euthanasia and reintroductions must meet the requirements of all applicable local, state, federal and international laws and regulations. Humane euthanasia must be performed in accordance with the established euthanasia policy of the institution and follow the recommendations of current AVMA Guidelines for the Euthanasia of Animals (2013 Edition https://www.avma.org/KB/Policies/Documents/euthanasia.pdf) or the AAZV's Guidelines on the Euthanasia of Non-Domestic Animals. Ownership and any applicable chain-of-custody must be documented. If such information does not exist, an explanation must be provided regarding such animals and specimens. Any acquisition of free-ranging animals must be done in accordance with all local, state, federal, and international laws and regulations and must not be detrimental to the long-term viability of the species in the wild.

2. The Director/Chief Executive Officer of the institution must have final authority for all acquisitions, transfers, and euthanasia.

3. Acquisitions or transfers/euthanasia/reintroductions must be documented through institutional record keeping systems. The ability to identify which animal is being transferred is very important and the method of identifying each individual animal should be documented. Any existing documentation must accompany all transfers. Institutional animal records data, records guidelines have been developed for certain species to standardize the process (https://www.aza.org/AnimalCare/detail.aspx?id=3150).

4. For some colonial, group-living, or prolific species, it may be impossible or highly impractical to identify individual animals when these individuals are maintained in a group. These species can be maintained, acquisitioned, transferred, and managed as a group or colony, or as part of a group or colony.

5. If the intended use of specimens from animals either living or non-living is to create live animal(s), their acquisition and transfer should follow the same guidelines. If germplasm is acquired or transferred with the intention of creating live animal(s), ownership of the offspring must be clearly defined in transaction documents (e.g., breeding loan agreements).

   Institutions acquiring, transferring or otherwise managing specimens should consider current and possible future uses as new technologies become available. All specimens from which nuclear DNA could be recovered should be carefully considered for preservation as these basic DNA extraction technologies already exist.

6. AZA member institutions must maintain transaction documents (e.g., confirmation forms, breeding agreements) which provide the terms and conditions of animal acquisitions, transfers and loans, including documentation for animal parts, products and materials. These documents should require the potential recipient or provider to adhere to the AZA Policy on Responsible Population Management, and the AZA Code of Professional Ethics, and must require compliance with the applicable laws and regulations of local, state, federal, and international authorities.

7. In the case of animals (living or non-living) and their parts, materials, or products (living or non-living) held on loan, the owner's written permission should be obtained prior to any transfer and documented in the institutional records.

8. AZA SSP and TAG necropsy and sampling protocols should be accommodated.

9. Some governments maintain ownership of the species naturally found within their borders. It is therefore incumbent on institutions to determine whether animals they are acquiring or transferring are owned by a government entity, foreign or domestic, and act accordingly by reviewing the government ownership policies available on the AZA website. In the case of government owned animals, proposals for and/or notifications of transfers must be sent to the species manager for the government owned species.

III. ACQUISITION REQUIREMENTS

A. General Acquisitions

1. 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 regarding the individual or species.

2. Animals (wild, feral, and domestic) may be held temporarily for reasons such as assisting governmental agencies or other institutions, rescue and/or rehabilitation, research, propagation or headstarting for reintroduction, or special exhibits.

3. Any receiving institution must have the necessary expertise and resources to support and provide for the professional care and management of the species, so that the physical, psychological, and social needs of individual animals and species are met.

4. If the acquisition involves a species managed by an AZA Animal Program, the institution should communicate with the Animal Program Leader and, in the case of Green SSP Programs, must adhere to the AZA Full Participation Policy (http://www.aza.org/full-participation-in-ssp-program-policy/).

5. AZA member institutions should consult AZA Wildlife Conservation and Management Committee (WCMC)-approved TAG Regional Collection Plans (RCPs), Animal Program Leaders, and AZA Animal Care Manuals (ACMs) when making acquisition decisions.

6. AZA member institutions that work with commercial vendors that acquire animals from the wild, must perform due diligence to assure the vendors' collection of animals is legal and using ethical practices. Commercial vendors should have conservation and animal welfare goals similar to those of AZA institutions.

7. AZA member institutions may acquire animals through public donations and other non-AZA entities when it is in the best interest of the animal and/or species.

B. Acquisitions from the Wild

Maintaining wild animal populations for exhibition, education and wildlife conservation purposes is a core function of AZA-member institutions. AZA zoos and aquariums have saving species and conservation of wildlife and wildlands as a basic part of their public mission. As such, the AZA recognizes that there are circumstances where acquisitions from the wild are needed in order to maintain healthy, diverse animal populations. Healthy, sustainable populations support the objectives of managed species programs and the core mission of AZA members. In some cases, acquiring individuals from the wild may be a viable option in addition to, or instead of, relying on breeding programs with animals already in human care.

Acquiring animals from the wild can result in socioeconomic benefit and environmental protection and therefore the AZA supports environmentally sustainable/beneficial acquisition from the wild when conservation is a positive outcome.

1. Before acquiring animals from the wild, institutions are encouraged to examine alternative sources including other AZA institutions and other regional zoological associations or other non-AZA entities.

2. When acquiring animals from the wild, both the long-term health and welfare impacts on the wild population as well as on individual animals must be considered. In crisis situations, when the survival of a population is at risk, rescue decisions will be made on a case-by-case basis by the appropriate agency and institution.

3. AZA zoos and aquariums may assist wildlife agencies by providing homes for animals born in nature if they are incapable of surviving on their own (e.g., in case of orphaned or injured animals) or by euthanizing the animals because they pose a risk to humans or for humane reasons.
4. Institutions should only accept animals from the wild after a risk assessment determines the zoo/aquarium can mitigate any potential adverse impacts on the health, care and maintenance of the existing animals already being housed at the zoo or aquarium, and the new animals being acquired.

IV. TRANSFER, EUTHANASIA AND REINTRODUCTION REQUIREMENTS

A. Living Animals

Successful conservation and animal management relies on the cooperation of many entities, both AZA and non-AZA. While preference is given to placing animals with AZA-accredited institutions or certified related facilities, it is important to foster a cooperative culture among those who share AZA's mission of saving species and excellence in animal care.

1. AZA members should assure that all animals in their care are transferred, humanely euthanized and/or reintroduced in a manner that meets the standards of AZA, and that animals are not transferred to those not qualified to care for them properly. Refer to IV.12, below, for further requirements regarding euthanasia.

2. If the transfer of animals or their specimens (parts, materials, and products) involves a species managed by an AZA Animal Program, the institution should communicate with that Animal Program Leader and, in the case of Green SSP Programs must adhere to the AZA Full Participation Policy (http://www.aza.org/full-participation-in-ssp-program-policy/).

3. AZA member institutions should consult WCMC-approved TAG Regional Collection Plans, Animal Program Leaders, and Animal Care Manuals when making transfer decisions.

4. Animals acquired solely as a food source for animals in the institution's care are not typically accessioned. There may be occasions, however, when it is appropriate to use accessioned animals that exceed population carrying capacity as feeder animals to support other animals. In some cases, accessioned animals may have their status changed to "feeder animal" status by the institution as part of their program for long-term sustained population management of the species.

5. In transfers to non-AZA entities, AZA members must perform due diligence and should have documented validation, including one or more letters of reference, for example from an appropriate AZA Professional Fellow or other trusted source with expertise in animal care and welfare, who is familiar with the proposed recipient and their current practices, and that the recipient has the expertise and resources required to properly care for and maintain the animals. Any recipient must have the necessary expertise and resources to support and provide for the professional care and management of the species, so that the physical, psychological, and social needs of individual animals and species are met within the parameters of modern zoological philosophy and practice. Supporting documentation must be kept at the AZA member institution (see #IV.9 below).

6. Domestic animals should be transferred in accordance with locally acceptable humane farming practices, including auctions, and must be subject to all relevant laws and regulations.

7. AZA members must not send any non-domestic animal to auction or to any organization or individual that may display or sell the animal at an animal auction. *See certain taxa-specific appendices to this Policy (in development) for information regarding exceptions*.

8. Animals must not be sent to organizations or individuals that allow the hunting of these individual animals; that is, no individual animal transferred from an AZA institution may be hunted. For purposes of maintaining genetically healthy, sustainable zoo and aquarium populations, AZA-accredited institutions and certified related facilities may send animals to non-AZA organizations or individuals

(refer to #IV.5 above). These non-AZA entities (for instance, ranching operations) should follow appropriate ranch management practices and other conservation minded practices to support population sustainability.

9. Every loaning institution must annually monitor and document the conditions of any loaned specimen(s) and the ability of the recipient(s) to provide proper care (refer to #IV.5 above). If the conditions and care of animals are in violation of the loan agreement, the loaning institution must recall the animal or assure prompt correction of the situation. Furthermore, an institution's loaning policy must not be in conflict with this AZA Policy on Responsible Population Management.

10. If living animals are sent to a non-AZA entity for research purposes, it must be a registered research facility by the U.S. Department of Agriculture and accredited by the Association for the Assessment & Accreditation of Laboratory Animal Care, International (AAALAC), if eligible. For international transactions, the receiving facility must be registered by that country's equivalent body having enforcement over animal welfare. In cases where research is conducted, but governmental oversight is not required, institutions should do due diligence to assure the welfare of the animals during the research.

11. Reintroductions and release of animals into the wild must meet all applicable local, state, and international laws and regulations. Any reintroduction requires adherence to best health and veterinary practices to ensure that non-native pathogens are not released into the environment exposing naive wild animals to danger. Reintroductions may be a part of a recovery program and must be compatible with the IUCN Reintroduction Specialist Group's Reintroduction Guidelines (http://www.iucnsscrsg.org/index.php).

12. Humane euthanasia may be employed for medical reasons to address quality of life issues for animals or to prevent the transmission of disease. AZA also recognizes that humane euthanasia may be employed for managing the demographics, genetics, and diversity of animal populations. Humane euthanasia must be performed in accordance with the established euthanasia policy of the institution and follow the recommendations of current AVMA Guidelines for the Euthanasia of Animals (2013 Edition https://www.avma.org/KB/Policies/Documents/euthanasia.pdf) or the AAZV's Guidelines on the Euthanasia of Non-Domestic Animals.

B. Non-Living Animals and Specimens

AZA members should optimize the use and recovery of animal remains. All transfers must meet the requirements of all applicable laws and regulations.

1. Optimal recovery of animal remains may include performing a complete necropsy including, if possible, histologic evaluation of tissues which should take priority over specimens' use in education/exhibits. AZA SSP and TAG necropsy and sampling protocols should be accommodated. This information should be available to SSP Programs for population management.

2. The educational use of non-living animals, parts, materials, and products should be maximized, and their use in Animal Program sponsored projects and other scientific projects that provide data for species management and/or conservation must be considered.

3. Non-living animals, if handled properly to protect the health of the recipient animals, may be utilized as feeder animals to support other animals as deemed appropriate by the institution.

4. AZA members should consult with AZA Animal Program Leaders prior to transferring or disposing of remains/samples to determine if existing projects or protocols are in place to optimize use.

5. AZA member institutions should develop agreements for the transfer or donation of non-living animals, parts, materials, products, and specimens and associated documentation, to non-AZA entities such as universities and museums. These agreements should be made with entities that have

appropriate long term curation/collections capacity and research protocols, or needs for educational programs and/or exhibits.

DEFINITIONS

Acquisition: Acquisition of animals can occur through breeding (births, hatchings, cloning, and division of marine invertebrates = “fragging”), trade, donation, lease, loan, transfer (inter- and intra-institution), purchase, collection, confiscation, appearing on zoo property, or rescue and/or rehabilitation for release.

Annual monitoring and Due diligence: Due diligence for the health of animals on loan is important. Examples of annual monitoring and documentation include and are not limited to inventory records, health records, photos of the recipient’s facilities, and direct inspections by AZA professionals with knowledge of animal care. The level of due diligence will depend on professional relationships.

AZA member institution: In this Policy “AZA member institutions” refers to AZA-accredited institutions and certified related facilities (zoological parks and aquariums). “AZA members” may refer to either institutions or individuals.

Data sharing: When specimens are transferred, the transferring and receiving institutions should agree on data that must be transferred with the specimen(s). Examples of associated documentation include provenance of the animal, original permits, tags and other metadata, life history data for the animal, how and when specimens were collected and conserved, etc.

Dispose: “Dispose/Disposing of” in this document is limited to complete and permanent removal of an individual via incineration, burying or other means of permanent destruction

Documentation: Examples of documentation include ZIMS records, “Breeding Loan” agreements, chain-of-custody logs, letters of reference, transfer agreements, and transaction documents. This is documentation that maximizes data sharing.

Domestic animal: Examples of domestic animals may include certain camelids, cattle, cats, dogs, ferrets, goats, pigs, reindeer, rodents, sheep, budgerigars, chickens, doves, ducks, geese, pheasants, turkeys, and goldfish or koi.

Ethics of Acquisition/Transfer/Euthanasia: Attempts by members to circumvent AZA Animal Programs in the acquisition of animals can be detrimental to the Association and its Animal Programs. Such action may also be detrimental to the species involved and may be a violation of the Association’s Code of Professional Ethics. Attempts by members to circumvent AZA Animal Programs in the transfer, euthanasia or reintroduction of animals may be detrimental to the Association and its Animal Programs (unless the animal or animals are deemed extra in the Animal Program population by the Animal Program Coordinator). Such action may be detrimental to the species involved and may be a violation of the Association’s Code of Professional Ethics.

“Extra” or Surplus: AZA’s scientifically-managed Animal Programs, including SSPs, have successfully bred and reintroduced critically endangered species for the benefit of humankind. To accomplish these critical conservation goals, populations must be managed within “carrying capacity” limits. At times, the number of individual animals in a population exceeds carrying capacity, and while meaning no disrespect for these individual animals, we refer to these individual animals as “extra” within the managed population.

Euthanasia: Humane death. This act removes an animal from the managed population. Specimens can be maintained in museums or cryopreserved collections. Humane euthanasia must be performed in accordance with the established euthanasia policy of the institution and follow the recommendations of current AVMA Guidelines for the Euthanasia of Animals (2013 Edition https://www.avma.org/KB/Policies/Documents/euthanasia.pdf) or the AAZV’s Guidelines on the Euthanasia of Non-Domestic Animals.

Feral: Feral animals are animals that have escaped from domestication or have been abandoned to the wild and have become wild, and the offspring of such animals. Feral animals may be acquired for temporary or permanent reasons.

Group: Examples of colonial, group-living, or prolific species include and are not limited to certain terrestrial and aquatic invertebrates, fish, sharks/rays, amphibians, reptiles, birds, rodents, bats, big herds, and other mammals,

Lacey act: The Lacey Act prohibits the importation, exportation, transportation, sale, receipt, acquisition or purchase of wildlife taken or possessed in violation of any law, treaty or regulation of the United States or any Indian tribal law of wildlife law. In cases when there is no documentation accompanying an acquisition, the animal(s) may not be transferred across state lines. If the animal was illegally acquired at any time then any movement across state or international borders would be a violation of the Lacey Act.

Museum: It is best practice for modern zoos and aquariums to establish relationships with nearby museums or other biorepositories, so that they can maximize the value of animals when they die (e.g., knowing who to call when they have an animal in necropsy, or specimens for cryopreservation). Natural history museums that are members of the Natural Science Collections Alliance (NSCA) and frozen biorepositories that are members of the International Society of Biological and Environmental Repositories (ISBER) are potential collaborators that could help zoos find appropriate repositories for biological specimens.

Non-AZA entity: Non-AZA entities includes facilities not accredited or certified by the AZA, facilities in other zoological regions, academic institutions, museums, research facilities, private individuals, etc.

Reintroduction: Examples of transfers outside of a living zoological population include movements of animals from zoo/aquarium populations to the wild through reintroductions or other legal means.

Specimen: Examples of specimens include animal parts, materials and products including bodily fluids, cell lines, clones, digestive content, DNA, feces, marine invertebrate (coral) fragments ("frags"), germplasm, and tissues.

Transaction documents: Transaction documents must be signed by the authorized representatives of both parties, and copies must be retained by both parties*. In the case of loans, the owner's permission for appropriate activities should be documented in the institutional records. This document(s) should be completed prior to any transfer. In the case of rescue, confiscation, and evacuation due to natural disasters, it is understood that documents may not be available until after acceptance or shipping. In this case documentation (e.g., a log) must be kept to reconcile the inventory and chain of custody after the event occurs. (*In the case of government owned animals, notification of transfers must be sent to species manager for the government owned species).

Transfer: Transfer occurs when an animal leaves the institution for any reason. Reasons for transfer or euthanasia may include cooperative population management (genetic, demographic or behavioral management), animal welfare or behavior management reasons (including sexual maturation and individual management needs). Types of transfer include withdrawal through donation, trade, lease, loan, inter- and intra-institution transfers, sale, escape, theft. Reintroduction to the wild, humane euthanasia or natural death are other possible individual animal changes in a population.

RECIPIENT PROFILE EXAMPLE

Example questions for transfers to non-AZA entities (from AZA-member Recipient Profile documents):

Has your organization, or any of its officers, been indicted, convicted, or fined by a State or Federal agency for any statute or regulation involving the care or welfare of animals housed at your facility? (If yes, please explain on a separate sheet).

Recipients agree that the specimen(s) or their offspring will not be utilized, sold or traded for any purpose contrary to the Association of Zoos and Aquariums (AZA) Code of Ethics (enclosed).

References, other than (LOCAL ZOO/AQUARIUM) employees, 2 minimum (please provide additional references on separate sheet):

Reference Name		Phone	
Facility		Fax	
Address		E-mail	
City	State		Zip
Country		AZA Member?	

Reference Name		Phone	
Facility		Fax	
Address		E-mail	
City	State		Zip
Country		AZA Member?	

Veterinary Information:

Veterinarian		Phone	
Clinic/Practice		Fax	
Address		E-mail	
City	State		Zip
Country			

How are animals identified at your facility? If animals are not identified at your facility, please provide an explanation about why they are not here:

Where do you acquire and send animals? (Select all that apply)

AZA Institutions	Non-AZA Institutions	Exotic Animal Auctions	Pet Stores
Hunting Ranches	Dealers	Private Breeders	Non-hunting Game Ranches
Entertainment Industry	Hobbyists	Research Labs	Wild
Other			

What specific criteria are used to evaluate if a facility is appropriate to receive animals from you?

Please provide all of the documents listed below:
Required:

1. Please provide a brief statement of intent for the specimens requested.
2. Resumes of primary caretakers and those who will be responsible for the husbandry and management of animals.
3. Description (including photographs) of facilities and exhibits where animals will be housed.
4. Copy of your current animal inventory.

Only if Applicable:

5. Copies of your last two USDA inspection reports (if applicable).
6. Copies of current federal and state permits.

7. Copy of your institutional acquisition/disposition policy.

(In-house use only) In-Person Inspection of this facility (Staff member/Date, attach notes):

(Local institution: provide Legal language certifying that the information contained herein is true and correct)

(Validity of this: This document and all materials associated will be valid for a period of 2 years from date of signature.)

Example agreement for Receiving institution (agrees to following condition upon signing):
RECIPIENT AGREES THAT THE ANIMAL(S) AND ITS (THEIR) OFFSPRING WILL NOT BE UTILIZED, SOLD OR TRADED FOR THE PURPOSE OF COMMERCE OR SPORT HUNTING, OR FOR USE IN ANY STRESSFUL OR TERMINAL RESEARCH OR SENT TO ANY ANIMAL AUCTION. RECIPIENT FURTHER AGREES THAT IN THE EVENT THE RECIPIENT INTENDS TO DISPOSE OF AN ANIMAL DONATED BY (INSITUTION), RECIPIENT WILL FIRST NOTIFY (INSTITUTION) OF THE IDENTITY OF THE PROPOSED TRANSFEREE AND THE TERMS AND CONDITIONS OF SUCH DISPOSITION AND WILL PROVIDE (INSTITUTION) THE OPPORTUNITY TO ACQUIRE THE ANIMAL(S) WITHOUT CHARGE. IF (INSTITUTION) ELECTS NOT TO RECLAIM THE ANIMAL WITHIN TEN (10) BUSINESS DAYS FOLLOWING SUCH NOTIFICATION, THEN, IN SUCH EVENT, (INSTITUTION) WAIVES ANY RIGHT IT MAY HAVE TO THE ANIMAL AND RECIPIENT MAY DISPOSE OF THE ANIMAL AS PROPOSED.

Institutional note: The text above is similar to the language most dog breeders use in their contracts when they sell a puppy. If people can provide that protection to the puppies they place, zoos/aquariums can provide it for animals that we place too! Some entities have been reluctant to sign it, and in that case we revert to a loan and our institution retains ownership of the animal. Either way, we are advised of the animal's eventual placement and location.

Appendix E: 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 institutions 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 alcids:

- Required:
    - Direct and floatation fecals
    - Vaccinate as appropriate
- Strongly recommended:
    - CBC/sera profile
    - Urinalysis
    - Appropriate serology (FIP, FeLV, FIV)
    - Heartworm testing in appropriate species

Appendix F: Ambassador (Program) Animal Policy and Position Statement

Ambassador (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 ambassador animal presentations. AZA's Conservation Education Committee's *Ambassador Animal Position Statement* summarizes the value of ambassador animal presentations (see pages 42–44).

For the purpose of this policy, an Ambassador 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 Ambassador Animals on a full-time basis, while others are designated as such only occasionally. Ambassador Animal-related Accreditation Standards are applicable to all animals during the times that they are designated as Ambassador Animals.

There are three main categories of Ambassador Animal interactions:

1. On Grounds with the Ambassador Animal Inside the Exhibit/Enclosure:
    a. Public access outside the exhibit/enclosure. Public may interact with animals from outside the exhibit/enclosure (e.g., giraffe feeding, touch tanks).
    b. 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 Ambassador Animal Outside the Exhibit/Enclosure:
    a. Minimal handling and training techniques are used to present Ambassador Animals to the public. Public has minimal or no opportunity to directly interact with Ambassador Animals when they are outside the exhibit/enclosure (e.g., raptors on the glove, reptiles held "presentation style").
    b. Moderate handling and training techniques are used to present Ambassador Animals to the public. Public may be in close proximity to, or have direct contact with, Ambassador Animals when they're outside the exhibit/enclosure (e.g., media, fund raising, photo, and/or touch opportunities).
    c. Significant handling and training techniques are used to present Ambassador Animals to the public. Public may have direct contact with Ambassador Animals or simply observe the in-depth presentations when they're outside the exhibit/enclosure (e.g., wildlife education shows).
3. Off Grounds:
    a. Handling and training techniques are used to present Ambassador 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 Ambassador 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 Ambassador Animals and the periods during which the Ambassador Animal-related Accreditation Standards are applicable. In addition, these Ambassador Animal categories establish a framework for understanding increasing degrees of an animal's involvement in Ambassador Animal activities.

Ambassador 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 Ambassador Animal presentations to develop an institutional Ambassador Animal policy that clearly identifies and justifies those species and individuals approved as Ambassador 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 Ambassador 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 Ambassador 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.

Ambassador 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 Ambassador 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 Ambassador 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 & Hodgkinson, 1999). The use of Ambassador 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.

Ambassador 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 & Benefield, 1986, 1988; Wolf & 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 & 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 Ambassador Animals in shows or informal presentations can be effective in lengthening the potential time period for learning and overall impact.

Ambassador 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 Ambassador Animals. A growing body of evidence supports the validity of using Ambassador 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 Ambassador 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

Ambassador 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 Ambassador 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 Ambassador 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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Appendix G: Developing an Institutional Ambassador Animal Policy

Last revision 2003
Re-authorized by the Board, June 2011

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 **Ambassador Animal Position Statement** describes the research underpinning the appropriate use of Ambassador 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 Ambassador Animals conveys intended and/or conflicting messages and to modify and improve programs accordingly and to ensure that all Ambassador Animals have the best possible welfare.

When utilizing Ambassador 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 Ambassador 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 Ambassador Animal policy that articulates and evaluates program benefits. The following recommendations are offered to assist each institution in formulating its own Institutional Ambassador Animal Policy, which incorporates the AZA Ambassador 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 Ambassador 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 Ambassador 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 Ambassador Animal policies should include a philosophical statement addressing the above, and should relate the use of Ambassador Animals to the institution's overall mission statement.

II. Appropriate Settings

The Ambassador Animal Policy should include a listing of all settings both on and off site, where Ambassador 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:

1. On-site programming
    a. Informal and non-registrants:
        i. On-grounds programming with animals being brought out (demonstrations, lectures, parties, special events, and media)
        ii. Children's zoos and contact yards
        iii. Behind-the-scenes open houses
        iv. Shows
        v. Touch pools
    b. Formal (registration involved) and controlled settings
        i. School group programs
        i. Summer camps
        ii. Overnights
        iii. Birthday parties
        iv. Animal rides
        v. Public animal feeding programs
    c. Offsite and outreach
        i. PR events (TV, radio)
        ii. Fundraising events
        iii. Field programs involving the public
        iv. School visits
        v. Library visits
        vi. Nursing home visits (therapy)
        vii. Hospital visits
        viii. Senior centers
        ix. 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 Ambassador Animal Policy address compliance with appropriate regulations and AZA Accreditation Standards.

IV. Collection Planning

AZA accredited institutions should have a collection planning process in place. Ambassador 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 Ambassador Animals:

1. Listing of approved Ambassador Animals (to be periodically amended as collection changes). Justification of each species should be based upon criteria such as:
    a. Temperament and suitability for program use
    b. Husbandry requirements
    c. Husbandry expertise
    d. Veterinary issues and concerns
    e. Ease and means of acquisition / disposition according to the AZA code of ethics
    f. Educational value and intended conservation message
    g. Conservation Status
    h. 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 Ambassador Animal Policy should address the specific messages related to the use of Ambassador 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 Ambassador 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 Ambassador 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 Ambassador 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., hand washing 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 Ambassador 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 Ambassador 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 Ambassador Animals is consistent with that of other animals in the collection.
7. Whenever possible have a “cradle to grave” plan for each Ambassador Animal to ensure that the animal can be taken care of properly when not used as a Ambassador Animal anymore.
8. If lengthy “down” times in Ambassador 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 Ambassador 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.
3. Signs of stress, stress factors, distress and discomfort behaviors.

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):

1. Guidelines for disinfecting surfaces, transport carriers, enclosures, etc. using environmentally safe chemicals and cleaners where possible.
2. Animal facts and conservation information.
3. Limitations and restrictions regarding ambient temperatures and or weather conditions.
4. Time limitations (including animal rotation and rest periods, as appropriate, duration of time each animal can participate, and restrictions on travel distances).
5. The number of trained personnel required to ensure the health and welfare of the animals, handlers and public.
6. The level of training and experience required for handling this species
7. Taxon/species-specific guidelines on animal health.
8. 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 Ambassador Animals, including:

1. Where and how the Ambassador 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 Ambassador 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 Ambassador 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.

Appendix H: Sample Transport Checklists

Item	Responsibility	Notes/Date Complete
COLLECTION PLANNING Contact Zoo/Aquarium and/or source of animal Ask for specimen report, terms (sale, trade, loan, donation, etc), and if the animal has any medical issues Start to fill in the ATP If needed, ask again for specimen reports For new species, include Collection Plan with ATP If animal is from a non AZA accredited institution, send a vendor profile and explain that it will need be submitted and approved prior to contracts/ATCs being generated	Curator / Supervisor	
ANIMAL TRANSACTION PROPOSAL ATP submitted to Registrar for submission code On ATP, use comment field to indicate the following: Status of vendor profile: Curator needs to get a copy of a signed vendor profile to the Registrar if applicable Specimen report received: must have specimen report from zoo to submit with ATP If no specimen report is available, explain why and when the report was originally requested by the Curator or have it in writing that the zoo won't provide one There are some "Special Circumstances" that the Registrars might need to be made aware of: Alert Registrar if payment needs to be done prior to receiving animals (this will require that the Curator or the Registrar, at the Curator's request, get an invoice from the sending institution) Alert Registrar if payment for shipping is required prior to receiving shipment (this will require that the Curator or the Registrar, at the Curator's request, get an invoice from the sending institution) Provide estimated or actual birth date of animal if Registrar does not receive a specimen report or specimen report is not available (i.e. private breeder, etc.) Alert Registrar if animal will go in to a group (existing or set-up a new group) or individual number	Curator / Supervisor	
CONTRACT After ATP is approved, Registrars will generate an Animal Transaction Confirmation/ATC and Curators will be required to sign both copies and return to Registrars. The following information might be needed for Curator signature depending on the type of transaction proposed: Breeding loan terms (signed by Rick or Sharon) TPWD donation form (this form is required by the state of Texas for all incoming and outgoing Texas native animals regardless of terms of the transaction). Prior to setting up a time-frame for receiving the shipment: The Registrars will email the section and the Vet clinic staff when the ATC and other necessary forms (permits, breeding loans, etc.) are received. Registrar must confirm a signed ATC or breeding loan is received before setting up shipment for arrival Registrar must confirm if a signed TPWD is on file (if applicable)	Curator / Supervisor	

Notable Exception: Some zoos will send Curators contracts for incoming animals. These contracts must be submitted to the Registrars if they are mailed to Curator or Supervisor Registrar does not write contracts for incoming animals. Typically this is done by the sending zoo/aquarium; however there are some that don't. Registrar will write contracts for private individuals/dealers and some zoos (for example, National Zoo and San Diego Zoo do not generally provide contracts)		
MEDICAL RECORDS Incoming Vet contacts sending institution for medical records and preship results	Vets	
QUARANTINE & DIET ARRANGEMENTS Medical records have been received Arrival / quarantine date determined with Vet Clinic Husbandry, training, and enrichment needs determined Quarantine set-up verified with Vet Clinic Diet information verified with Animal Commissary Diet Sheet sent to Incoming Vet and Clinic Keeper/Supervisor Delivery date set-up with Commissary Animal data transfer forms received from sending facility prior to the animal's arrival	Curator / Supervisor	
SHIPPING ARRANGEMENTS Prior to confirming final shipping arrangements: Curators must email Everyone in Registrars and Incoming Vet as well as the Clinic Keepers/Supervisor to confirm that all necessary paperwork has been obtained and shipment is cleared to arrive Ship date determined and confirmed with sending facility Vet staff and Registrars notified of confirmed shipping date Shipping container provided if necessary Arrival / Pick-up arranged There are some "Special Circumstances" that the Registrars might need to be made aware of: Payment method of shipping/purchase should be determined prior to shipment: i.e. will shipping be paid out of Section's budget, VP of Animal Programs budget, donor, etc. Gather all paperwork and submit to Registrars including the original airway bill - Registrar receives all shipping documents first Acquisition/Arrival notice entered on Section daily report Inform Registrar when crate is returned to sending institution or if payment is needed for the crate	Curator / Supervisor	

Appendix I: Sample AZA-Accredited Institutional Fungal Air Spore Sampling Protocol

Purpose: Sampling provides a snapshot picture of the fungal air spore population in our avian exhibits. Routine testing is performed twice a year in spring (April/May) and fall (October/November). We are most concerned with the population of *Aspergillus* sp., as these organisms cause respiratory infections in birds; we also examine the trends in total air spores, as this may indicate a contamination problem in one of the exhibits.

Equipment:
- Aerotech® 6 sampler with tubing and pump
- Extension cord
- Isopropyl Alcohol
- Lint-Free Litho Pads
- CMA+ plates (enough for duplicate samples and a sample blank at each location)
- Stopwatch
- Pen and Sharpie®
- Paper
- Gloves
- Small Ziploc® bags

Sampling Procedure:
1) Choose an appropriate location.
2) Connect the tubing from the pump to the sampler.
3) Plug in the sampler and turn it on.
4) Adjust the air flow gauge to 28.3 L/min (figure it as best you can).
5) Turn the pump off and sterilize the sampler. With gloves on, wipe the surfaces of the sampler with isopropyl alcohol using a lint-free Litho pad or other sterile gauze. Start from the inside and work your way out to reduce the risk of contamination.
6) To load the sampler, place a sample plate on the base of the sampler so it rests on the three raised pins. Immediately place the classification stage (the one with all the little pin pricks in it) on top so it is seated snugly with the O-ring. Then place the inlet cone (the cone part, this goes on the top) into its groove. Secure with the three springs, making sure there is a good seal and the lid is on straight.
7) Take a sample blank at each sample location by loading an uncapped agar plate into the sampler and letting it sit without turning it on for a few seconds. Label this plate as the sample location blank and the date.
8) Load the sampler with another agar plate.
9) Turn on the pump and sample for 2 minutes.
10) Remove the sample plate, recover with its lid, label with the sample info (location, date, replicate number), and seal it in a Ziploc® bag.
11) Take a replicate sample following the above procedure.
12) Sterilization of the sampler isn't necessary between each sample but it should be performed at each sample location.
13) Note the time of sampling for each location to keep in your records.
14) Before putting the sampler away, fully wipe down the instrument with alcohol and then store in its special box.

Incubation, Identification and Shipping: If you are going to attempt to count and ID the plates yourself: This is time consuming and not highly recommended. If you are a mycologist, then this may be for you. If you aren't then I would suggest sending the plates out for identification instead. This technique is sometimes used on special samples where we are only looking for a particular genus/species of fungi:
1) Incubate the plates at 25ºC (or leave them in the laminar flow hood at room temperature).
2) Incubation may take anywhere from 7-14 days.

3) Count and identify the colonies that you can. Check the references we have for help with identification of colonies.
4) Report the results as colony forming units (cfu) per liter of air sampled.

If you are shipping these samples out for someone else to ID (recommended):

1) Retain the sample blanks since there should be little to no growth on these plates and there is a charge for processing them.

Make sure you sample on a Monday–Thursday so you can ship the samples overnight to Aerotech® Labs. Label all plates clearly with distinct identifications. Seal the samples with tape to secure the lids and prevent contamination during shipment. I also keep the duplicate samples in their own Ziploc® bags for further protection. Fill out the Chain of Command sheet that Aerotech® has provided with the sample ID, type of analysis requested (A003.14 for environmental fungi count on cornmeal agar), total volume sampled for each sample (should be 56.6L if 2 minute sample time was used), and any special instructions (like we use corn meal agar plates and we are especially looking for *Aspergillus* spp. identification).

The Report: When the results are in, calculate the averages and standard deviations of the two replicate samples for each sample location. Use these values to give a short description of the results from each sampling area. Try to include notes on trends and differences noted from previous sampling sessions. The tables included in the report should include these categories: *Aspergillus* spp., *Cladosporium* spp., *Penicillium* spp., other fungi, and total fungi with all results reported as cfu/L of air sampled. The figures should include bar graphs for each sample location demonstrating the results from the table.

Appendix J: Representative ingredients commonly fed to alcids (NPAL values, supplied by AZA institution).

Ingredient	H_2O	Protein	Fat	Ash	CHO	Kcal	Ca	P	Mg	Vit A	Vit D	Vit E
Wet weight basis.	**%**	**%**	**%**	**%**	**%**	**kcal/100g**	**g/100g**	**%**	**%**	**IU A/lb**	**IU D/g**	**mg/100g**
Albacore, frozen (whole, head off, gutted)	57	19.4	21	3.91	<1.0	270						
Cichlids	81	11.9	4.3	3.81	<0.5	86.2						
Clam, Whole Surfclam (hand shucked) Lot #757587	78	14.2	0.6	1.93	5.6	84.6						
Clam, Surf, Whole Lot #765548	76	18.5	1.3	1.36	2.5	95.9						1.36
Cod, Hoki	77	15.9	5.2	0.98	0.7	113						
Crickets	72	17.3	5.3	2.59	3	129	0.034	0.25				
Crickets (enriched)	73	17.8	5.1	2.75	1.2	122	0.315	0.27				
Cricktets with vitamins	71	17.5		3.63			1.03	0.26				
Goldfish	84	11.5	1.3	5.2	<0.5	57.5						
Krill, Pacifica, Lot #777547	79	14.5	3.5	2.66	<0.5	89.3						
Krill, Pacifica, (rinsed) Lot #712004	85	11.7	1.4	1.9	<0.5	59.8	0.313	0.28		10300	<0.250	2.53
Krill, superba (fresh) with vitamins	80	13.1	3.8	2.91	<0.5	86.4	0.309	0.24				
Krill, Superba (Fresh) Lot #737760	87	9.31	2.3	1.89	<0.5	57.6	0.326	0.28				
Krill, Shake, (with Banquet Flakes)												
Krill, Superba (blanched) Lot #712171	86	8.95	3.2	2.21	<0.5	64.3						3.45
Mackeral, whole (race street foods)	71	21.4	6.3	2.33	<0.5	142						
Mealworms	58	19.9	9.1	1.94	11	205	<.01	0.23				
Mealworms (enriched)	62	19.9	13	1.52	4	212	0.06	0.34				
Mealworms with vitamins	57	21.4	12	2.34	7.2	226	0.396	0.34				
Natural Balance cat food, weight management	9.5	30	12	8	41	389						
Nightsmelt	81	15.9	1.7	2.12	<0.5	79.1	0.56	0.41	0.33	769	0.36	
Sardines, IQF (MFC),*sardinops sagax*	66	16.9	17	1.83	<0.5	218						<0.400
Seabird Diet							2.46	1.27				

Shrimp, Arizona Desert Sweet	80	15	2	2	0.8	81						
Shrimp, Black Tiger P&D	85	12.6	0.5	2.12	<0.5	55.1						
Shrimp, Georgia, browns (shell off)	77	19.8	0.8	1.17	0.9	90.3						
Shrimp, Georgia, browns (shell on, Tail off)	77	19	0.9	1.83	0.9	87.7						
Shrimp, Ocean Garden 26/30 (shell on, Tail off)	77	19.9	0.9	1.44	0.8	91.3						
Shrimp, Ocean Garden 26/30 (shell off)	77	19.9	1	1.27	1.3	93.4						1.87
Silversides, Whole, Lot #755913	78	14.2	5.9	2.6	<0.5	110	0.379	0.33	0.35	1390	1.59	0.888
Squid,*loligo opalescens*, Whole	81	15.3	1.9	1.43	<0.5	78.6						2.49
Trout, boned, frozen (Idaho brand)	70	18.4	8.7	1.48	1.3	157						
Trout, whole (IQF) Lot#779194	74	15.4	9.4	2072	<.05	146						
Tubifex Worms	91	5.33	1.7	0.39	1.2	41.5	0.011	0.11				
Waxworms	64	15.2	18	0.95	2	233	0.025	0.2				
Waxworms with vitamins	58	14.3		1.55			0.461	0.19				

Appendix K: Reference Values (mean ± SD) for Hematological and Serum Biochemistry Parameters of Selected Species (ISIS, 2007).

Parameter	**Common puffin (*Fratercula arctica*) (*N*)**	**Horned puffin (*Fratercula corniculata*) (*N*)**	**Tufted puffin (*Fratercula cirrhata*) (*N*)**	**Common murre (*Uria aalge*) (*N*)**
Erythrocytes ($x10^6/\mu L$)	2.70±0.50 (73)	2.46±0.40 (21)	7.38±0.00 (1)	2.40±0.50 (14)
Leukocytes ($x10^3/\mu L$)	7.06±2.97 (29)	6.77±5.37 (20)	9.66±2.99 (26)	6.81±2.47 (16)
Hemoglobin (g/dL)	12.4±3.5 (9)	12.7±2.3 (5)	14.7±3.5 (22)	11.0±3.9 (8)
Hematocrit (%)	47.3±4.3 (85)	42.3±6.6 (21)	43.0±11.5 (60)	45.7±3.3 (23)
MCH (pg)	42.3±9.9 (9)	51.9±17.6 (5)	28.5±0.00 (1)	41.6±13.8 (8)
MCHC (g/dL)	24.2±6.7 (9)	32.8±7.5 (5)	32.3±8.3 (20)	23.3±8.2 (8)
MCV (fl)	181.2±40.2 (73)	176.0±28.7 (20)	83.3±0.0 (1)	194.0±39.3 (13)
Heterophils ($x10^3/\mu L$)	3.35±1.46 (29)	3.15±3.34 (20)	6.00±2.76 (23)	3.81±1.02 (16)
Lymphocytes ($x10^3/\mu L$)	3.08±1.92 (29)	2.97±2.09 (20)	3.25±1.43 (23)	2.20±1.61 (16)
Monocytes ($x10^3/\mu L$)	0.29±0.40 (26)	0.35±0.45 (20)	0.28±0.15 (14)	0.25±0.23 (11)
Eosinophils ($x10^3/\mu L$)	0.27±0.34 (4)	0.21±0.00 (1)	0.37±0.15 (2)	0.64±0.61 (10)
Basophils ($x10^3/\mu L$)	0.70±0.61 (14)	0.31±0.35 (20)	0.53±0.00 (1)	0.73±0.60 (5)

MCH, mean corpuscular hemoglobin; MCHC, mean corpuscular hemoglobin concentration; MCV, mean corpuscular volume.

Parameter	**Common puffin (*Fratercula arctica*) (*N*)**	**Horned puffin (*Fratercula corniculata*) (*N*)**	**Tufted puffin (*Fratercula cirrhata*) (*N*)**	**Common murre (*Uria aalge*) (*N*)**
Glucose (mg/dL)	334±50 (69)	337±67 (5)	350±46 (20)	326±43 (10)
Blood Urea Nitrogen (mg/dL)	6±3 (19)		3±1 (11)	
Creatinine (mg/dL)	0.5±0.2 (18)		0.2±0.2 (11)	
Uric Acid (mg/dL)	6.7±3.4 (75)	12.8±9.6 (6)	14.0±13.0 (24)	22.1±18.5 (4)
Calcium (mg/dL)	9.2±1.5 (63)	9.7±0.7 (6)	10.0±0.9 (20)	7.8±0.5 (2)
Phosphorous (mg/dL)	3.1±1.1 (40)	4.3±1.8 (4)		0.9±0.1 (2)
Sodium (mEq/L)	131±0 (1)		167±6 (12)	148±0 (1)
Potassium (mEq/L)	3.9±0.0 (1)		2.3±0.7 (12)	1.9±0.3 (2)
Cholesterol (mg/dL)	346±50 (33)		280±113 (50)	228±69 (5)
Triglycerides (mg/dL)	146±26 (16)		136±57 (8)	
Total Protein (g/dL)	2.7±0.5 (48)	4.6±0.6 (7)	3.9±0.6 (33)	
Albumin (g/dL)	1.1±0.3 (25)	1.4±0.3 (5)		
Globulin (g/dL)	1.3±0.5 (25)	2.9±0.2 (5)		
AST (IU/L)	162±106 (70)	126±62 (5)	159±54 (17)	88±36 (6)
ALT (IU/L)	45±14 (20)	148±12 (2)	53±13 (7)	26±11 (6)
Total bilirubin (mg/dL)	0.4±0.4 (41)		1.7±2.1 (11)	
Amylase (U/L)	457±129 (2)		2172±0 (1)	
ALP (IU/L)	44±20 (38)		68±51 (3)	129±0 (1)
LDH (IU/L)	470±284 (36)		1145±1134 (3)	
CPK (IU/L)	881±575 (13)		1201±876(14)	883±731 (2)

AST, aspartate aminotransferase; ALT, alanine aminotransferase; ALP, alkaline phoshatase; LDH, lactate dehydrogenase; CPK, creatine phosphokinase.

Appendix L: Sample Enrichment Approval Form

Animal____

Submitted by____

Item (include picture)____

Behavior to encourage____

Type: food____ Environmental_____ Social _______ Sensory_________ Manipulative________

Description____
____

Duration____
____

Preopening_______ Open, sign_____________ Open, no sign____________ Off exhibit_________

Duration____

Approved_____________

Not approved__________

Comments____
____

Curator____________________

Vet________________________

Appendix M: Sample Enrichment Evaluation Form

New Enrichment Evaluation Form

Animal: ____

Type of Enrichment: ____

Date	Response	Comments

RESPONSE EVALUATION:

1 = animal runs/flees from enrichment
2 = animal appears to ignore enrichment
3 = animal orients to/looks at, but does not physically contact enrichment
4 = animal makes brief contact (e.g. pecks, picks up, etc.)
5 = animal makes substantial or repeated contact with enrichment
6 = no direct interaction seen, but evidence interaction with enrichment (e.g. items moved or feces by item)

Appendix N: Sample Research Policy Form

RESEARCH OVERSIGHT POLICY
August 2007

I. INTRODUCTION

The (insert name of institution) is committed to supporting, facilitating and conducting scientific research in the life sciences in pursuit of its mission to inspire conservation of the oceans. Effective conservation efforts have their roots in science, and the aquarium supports research efforts to benefit the wild relatives of the species in our care and their habitats.

(insert name of institution)has developed this administrative oversight framework to ensure that research conducted under the auspices of (insert name of institution) is credible, defensible and meritorious. This framework will also:

- Encourage and facilitate research at the aquarium/zoo, and maintain an effective and minimally burdensome review, approval and tracking process;
- Hold researchers accountable for progress, outputs and outcomes;
- Meet AZA requirements and guidelines regarding research oversight.

II. SCOPE OF ACTIVITIES COVERED

For the purposes of this oversight framework, "scientific research" is defined as diligent, systematic and experimentally-based investigation in the life sciences, outside the scope of normal plant and animal management and clinical care, in order to discover or revise facts, theories and applications. Scientific research covered by this policy framework includes research by non-staff investigators requiring (insert name of institution) assets and resources. It also covers research by (insert name of institution) staff when one or more of the following applies:

- There is potential for animal health concerns
- A significant financial or resource commitment is required
- There are human health and safety concerns
- The research involves the use of the (insert name of institution) "brand"
- The research has either high public visibility or affects the visitor experience
- Oversight is requested by VP of Husbandry, Director of Conservation Research, the Animal Welfare Committee, or by the investigator

III. CRITERIA FOR APPROVAL OF RESEARCH INITIATIVES

All research covered by this policy will be critically reviewed. While the specific parameters evaluated during this review process may vary depending upon the nature of the proposed research, all proposals will be evaluated by the following criteria:

- Relevance to the aquarium's mission
- Scientific rigor and defensibility
- Scientific merit

In addition, in order to be considered for approval, all research proposals must:

- Comply with all applicable internal and external regulatory requirements, including, but not limited to, federal, state, local, and institutional permitting
- Meet scientific ethical standards
- Be financially and logistically feasible with available MBA resources
- Identify a qualified scientific advisor
- Not be redundant with on-going institutional research without adequate and specific justification

IV. PROTOCOLS AND PROCEDURES FOR RESEARCH OVERSIGHT

A. Research Committee

Research Committee is comprised of the following members:

Director of Conservation Research (chair)
Research Biologist
Staff Veterinarian
Vice President of Husbandry
Husbandry Curator
Outside research scientist without vested interest in aquarium research initiatives

This committee oversees the implementation and administration of the research oversight program at the aquarium. The charge of the committee is to:

- Monitor and, when necessary, revise the aquarium's research policies and procedures
- Review and evaluate research proposals, status reports and final reports
- Maintain an ongoing list of pending and current research projects conducted under the auspices of this policy, and provide an annual summary report to senior management
- Review, approve and track external requests for biological samples

B. Research Approval Process

Investigators wishing to conduct research at the aquarium must submit a research proposal on the aquarium's standardized form. Proposals are submitted to the Research Biologist, who will serve as the administrative lead on reviewing, approving and tracking research proposals. For projects that require significant aquarium funding, proposals should be submitted in synchrony with the aquarium's annual goal-setting and budget process.

All proposals will be reviewed by the Research Committee chair and forwarded to all committee members. The chair may recommend that specific research proposals be reviewed by the full committee, by one or more designated reviewer(s) or by one or more external reviewer(s). Any member of the Research Committee can request review of a proposal by the full committee. The committee may request a meeting with the investigator and others to clarify aspects of the proposal or to address reviewers' concerns. Approval, denial or continuance for further information or clarification is by consensus of the committee. The Research Biologist will oversee the communication between the investigator and the committee regarding the status and disposition of research proposals.

C. Requirements and Expectations

Annual progress reports are required for all research conducted under this policy. Internal investigator research reports are due by August 1 of each year, allowing for evaluation of on-going research initiatives prior to budget preparation for the following year.

Final reports are due upon completion of all research projects. Broad dissemination of research findings, including publication of research results, is encouraged. Manuscripts intended for publication must be reviewed and approved by the Research Committee prior to submission. The aquarium should be appropriately acknowledged in publications stemming from research conducted under this policy. Copies of written products (published proceedings, gray literature reports, reprints of published reports, journal articles, books and book chapters, dissertations and theses) stemming from research projects must be sent to the committee chair when they are published, and will be placed in the aquarium's library holdings.

At the committee's discretion, primary investigators are required to present the status and findings of their research to aquarium staff and volunteers in a lunchtime seminar or other venue.