ASSOCIATION OF ZOOS & AQUARIUMS

SHOREBIRDS (Charadriiformes*) CARE MANUAL

*Does not include Alcidae

CREATED BY
AZA CHARADRIIFORMES TAXON ADVISORY GROUP
IN ASSOCIATION WITH
AZA ANIMAL WELFARE COMMITTEE

Shorebirds (Charadriiformes) Care Manual

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

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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 Charadriiformes

Classification	Taxonomy	Additional information
Kingdom	Animalia	
Phylum	Chordata	
Class	Aves	
Order	Charadriiformes	
Family	Burnhinidae (Thick-knees) Charadriidae (Plovers) Chionididae (Sheathbills) Dromadidae (Crab plover) Frynchopidae (Skimmers) Glareolidae (Coursers and pratincoles) Haematopodidae (Oystercatchers) Ibidorhynchidae (Ibisbill) Jacanidae (Jacanas) Pedionomidae (Plains-wanderer) Pluvianellidae (Magellanic plover) Recurvirostridae (Stilts and avocets) Rostratulidae (Painted snipes) Scolopacidae (Snipes, sandpipers, and phalaropes) Stercorariidae (Skuas) Thinocoridae (Seedsnipes)	

Genus, Species, and Status

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

Genus	Species	Common Name	USA Status	IUCN Status	AZA Status
Family: Jacanidae (Jacanas)					
Actophilornis	*africanus*	African jacana		Least concern	Studbook
Hydrophasianus	*chirurgus*	Pheasant-tailed jacana		Least concern	
Jacana	*jacana*	Wattled jacana		Least concern	
Jacana	*spinosa*	Northern jacana			
Metopidius	*indicus*	Bronze-winged jacana		Least concern	
Family: Rostratulidae (Painted snipes)					
Rostratula	*benghalensis*	Greater painted-snipe			
Family: Dromadidae (Crab plover)					
Dromas	*ardeola*	Crab plover		Least concern	
Family: Haematopodidae (Oystercatchers)					
Haematopus	*bachmani*	American black oystercatcher	Species of concern	Least concern	
Haematopus	*palliatus*	American oystercatcher	Species of high concern	Least concern	
Family: Ibidorhynchidae (Ibisbill)					
Ibidoryncha	*struthersii*	Ibisbill		Least concern	

Family: Recurvirostridae (Stilts and avocets)					
Himantopus h.	*mexicanus*	Black-necked stilt		Least concern	SSP
Himantopus h.	*melanurus*	Black-tailed stilt			
Recurvirostra	*americana*	American avocet		Least concern	
Recurvirostra	*avosetta*	Pied avocet		Least concern	
Family: Burnhinidae (Thick-knees)					
Burhinus	*capensis*	Spotted dikkop		Least concern	SSP
Burhinus	*bistriatus*	Double-striped Thick-knee		Least concern	
Burhinus	*oedicnemus*	Stone-curlew		Least concern	
Burhinus	*superciliaris*	Peruvian thick-knee		Least concern	
Burhinus	*grallarius*	Bush thick-knee		Near threatened	
Esacus	*magnirostris*	Beach thick-knee		Near threatened	
Family: Glareolidae (Coursers and pratincoles)					
Glareola	*maldivarum*	Oriental pratincole		Least concern	
Pluvianus	*aegyptius*	Egyptian plover		Least concern	
Family: Charadriidae (Plovers)					
Anitibyx	*armatus*	Blacksmith plover		Least concern	
Charadrius	*melodus*	Piping plover	Endangered and threatened	Near threatened	
Charadrius	*semipalmatus*	Semipalmated plover		Least concern	
Charadrius	*montanus*	Mountain plover		Near threatened	
Charadrius	*vociferus*	Killdeer		Least concern	
Charadrius a.	*nivosus*	Snowy plover	Threatened	Least concern	
Charadrius a.	*occidentalis*	Snowy plover		Least concern	
Pluvialis	*dominica*	American golden plover		Least concern	
Pluvialis	*fulva*	Pacific golden plover		Least concern	
Pluvialis	*squatarola*	Black-bellied plover		Least concern	
Vanellus	*crassirostris*	Long-toed lapwing		Least concern	
Vanellus	*miles*	Masked lapwing		Least concern	SSP
Vanellus	*spinosus*	Spur-winged Lapwing		Least concern	Studbook
Vanellus	*chilensis*	Southern lapwing		Least concern	
Vanellus	*senegallus*	African wattled lapwing		Least concern	
Vanellus	*albiceps*	White-headed lapwing		Least concern	
Family: Chionididae (Sheathbills)					
Chionis	*albus*	Snowy sheathbill			
Chionis	*minor*	Black-faced sheathbill	Not evaluated		
Family: Scolopacidae (Snipes, sandpipers, and phalaropes)					
Actitis	*macularia*	Spotted sandpiper		Least concern	
Aphriza	*virgata*	Surfbird		Least concern	
Arenaria	*interpres*	Ruddy turnstone		Least concern	
Arenaria	*melanocephala*	Black turnstone		Least concern	
Bartramia	*longicauda*	Upland sandpiper		Least concern	
Calidris	*alpina*	Dunlin		Least concern	
Calidris	*canutus*	Red knot	Candidate	Least concern	
Calidris	*alba*	Sanderling		Least concern	
Calidris	*mauri*	Western sandpiper		Least concern	
Calidris	*minutilla*	Least sandpiper		Least concern	
Calidris	*fusciollis*	White-rumped sandpiper		Least concern	
Calidris	*bairdii*	Baird’s sandpiper		Least concern	
Calidris	*melanotos*	Pectoral sandpiper		Least concern	
Calidris	*maritima*	Purple sandpiper		Least concern	
Calidris	*ptilocnemus*	Rock sandpiper		Least concern	
Calidris	*pusilla*	Semipalmated		Near	

		sandpiper		Threatened	
Catoptrophorus	*semipalmatus*	Willet		Least concern	
Gallinago	*gallinago*	Common snipe		Least concern	
Gallinago	*nemoricola*	Wood snipe		Vulnerable	
Heteroscelus	*incanus*	Wandering tattler		Least concern	
Limnodromas	*scolopaceus*	Long-billed dowitcher		Least concern	
Limnodromas	*griseus*	Short-billed dowitcher		Least concern	
Limosa	*haemastica*	Hudsonian godwit		Least concern	
Limosa	*fedoa*	Marbled godwit		Least concern	
Micropalama	*himantopus*	Stilt sandpiper		Least concern	
Numenius	*phaeopus*	Whimbrel		Least concern	
Numenius	*americanus*	Long-billed curlew		Least concern	
Numenius	*borealis*	Eskimo curlew	Endangered	Critically endangered	
Phalaropus	*lobatus*	Red-necked phalarope		Least concern	
Phalaropus	*fulicularia*	Red phalarope		Least concern	
Philomachus	*pugnax*	Ruff		Least concern	
Scolopax	*minor*	American woodcock		Least concern	
Steganopus	*tricolor*	Wilson’s phalarope		Least concern	
Tringa	*melanoleuca*	Greater yellowlegs		Least concern	
Tringa	*flaviceps*	Lesser yellowlegs		Least concern	
Tringa	*solitaria*	Solitary sandpiper		Least concern	
Tryngites	*subruficollis*	Buff-breasted sandpiper		Near threatened	
Family: Stercorariidae (Skuas)					
Catharacta	*antarctica*	Brown skua		Least concern	
Stercorarius	*parasiticus*	Arctic skua		Least concern	
Family: Laridae (Gulls)					
Larus	*atricilla*	Laughing gull		Least concern	
Larus	*novaehollandiae*	Silver gull		Least concern	
Larus	*pipixcan*	Franklin’s gull		Least concern	
Larus	*philadelphia*	Bonaparte’s gull		Least concern	
Larus	*modestus*	Grey gull		Least concern	
Larus	*heermanni*	Heermann’s gull		Near threatened	
Larus	*delawarensis*	Ring-billed gull		Least Concern	
Larus	*cirrocephalus*	Grey-headed gull		Least Concern	
Larus	*maculipennis*	Brown-hooded gull		Least Concern	
Larus	*californicus*	California gull		Least concern	
Larus	*marinus*	Great black-billed gull		Least Concern	
Larus	*dominicanus*	Kelp gull		Least concern	
Larus	*occidentalis*	Western gull		Least concern	
Larus	*argentatus*	Herring gull		Least concern	
Rissa	*brevirostris*	Red-legged Kittiwake		Vulnerable	
Rissa	*tridactyla*	Black-legged kittiwake		Least concern	
Family: Pedionomidae					
Pedionomus	*torquatus*	Plains-wanderer		Endangered	
Family: Pluvianellidae					
Pluvianellus	*socialis*	Magellanic plover		Near threatened	
Family: Sternidae (Terns)					
Gygis	*alba*	White tern		Least concern	
Hydroprogne	*caspia*	Caspian tern		Least concern	
Larosterna	*inca*	Inca tern		Near threatened	SSP
Sterna	*hirundo*	Common tern		Least concern	
Sterna	*forsteri*	Forster’s tern		Least concern	
Sterna	*paradisaea*	Arctic tern		Least concern	
Sterna	*antillarum*	Least tern		Least concern	

Sterna	*fuscata*	Sooty tern	Least concern
Thalasseus	*maximus*	Royal tern	Least concern
Thalasseus	*sandvicensis*	Sandwich tern	Least concern
Family: Rynchopidae (Skimmers)			
Rynchops	*niger*	Black skimmer	Least concern
Family: Thinocoridae (Seedsnipes)			
Attagis	*gayi*	Rufous-bellied seedsnipe	Least concern
Attagis	*malouinus*	White-bellied seedsnipe	Least concern
Thinocorus	*orbignyianus*	Gray-breasted seedsnipe	Least concern
Thinocorus	*rumicivorus*	Least seedsnipe	Least concern

General Information

The information contained within this Animal Care Manual (ACM) provides a compilation of animal care and management knowledge that has been gained from recognized species experts, including AZA Taxon Advisory Groups (TAGs), Species Survival Plan® Programs (SSPs), Studbook Programs, biologists, veterinarians, nutritionists, reproduction physiologists, behaviorists and researchers. They are based on the most current science, practices, and technologies used in animal care and management and are valuable resources that enhance animal welfare by providing information about the basic requirements needed and best practices known for caring for *ex situ* Charadriiformes 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 Charadriiformes must comply with all relevant local, state, and federal wildlife laws and regulations; AZA accreditation standards that are more stringent than these laws and regulations must be met (AZA Accreditation Standard 1.1.1).

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

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

Natural history: Charadriiformes are a diverse group of shorebirds that live, breed, and forage along the water's edge. Worldwide, there are over 200 species in 18 families; their closest relatives are gulls and terns. Most shorebirds are gregarious and are frequently seen in mixed flocks of several species. Further research is needed to ascertain the average life span of shorebirds in natural settings and whether or not that trend continues in AZA facilities.

Habitat preferences: Most shorebirds are found around bodies of water such as ocean beaches, estuaries, salt marshes, and fresh water lakes and wetlands. The birds rely on these habitats for nest selection, rearing young, and for feeding. The type of habitat where shorebirds are found depends on their intended activity, such as feeding or breeding. Birds such as plovers, willets, and sandpipers will forage on intertidal mudflats, estuaries and beaches, or they may be found inland around fresh water wetlands, mudflats, and sparsely vegetated agricultural areas. Each habitat has its own characteristic species.

During the breeding season, shorebirds are limited by available food supply and nest sites, and these constraints dictate their choice of habitat. Most North American shorebird species nest on the open tundra areas of the Arctic and Subarctic. Species that breed in coastal areas seek sandy beaches, gravelly areas, or vegetated marshes. Birds such as killdeer, which breed in the interior regions, generally nest near wetlands with sand or gravel substrates. They may also breed in wet meadows or marshes, wooded ponds, and streams found throughout the region. During migration and wintertime, shorebirds have distinct habitat preferences. Some species show marked partiality for one type of habitat; others frequent a range of habitats. In the nonbreeding season, they may collect along the coastline in greater densities (Colwell, 2010).

Feeding habits: Shorebird bills have an amazing variety of shapes and sizes, from the long, down-curved bill of the curlew, to the short, stubby bill of plovers. Shorebird bills dictate not only what the birds eat, but also how they catch their prey, as each bill is perfectly adapted to its task. Shorebirds use two basic methods of feeding: "picking"—plucking prey from the surface of the water or land; and "probing"—immersing the bill into a particular substrate. Some shorebirds use only one method of feeding year-round; others vary their way of foraging according to changes in the seasons, locales, and availability of food. In general, sandpipers probe and plovers pick. Shorebird diets also change with the seasons and habitats. For example, during breeding season in the Arctic, many species eat seeds early in the season and berries later on, when the insect population has diminished. When insects are plentiful, they become the main prey for many species on the breeding grounds. Chick hatching almost always coincides with periods of insect abundance. Adult and larval insects make up the bulk of most species diets at this time.

The tips of many shorebird bills are flexible and can be opened without opening the base of the bill, permitting the birds to grasp prey while simultaneously probing in the ground. Specialized cells in the tips of the bills are highly sensitive to touch and movement, allowing the birds to feel a prey item, as well the prey's motion. Because of the great variation of bill length among shorebirds, many different species can feed on the same area without depleting the food source.

Migration: Migration represents an important segment of the annual cycle of shorebirds. Individuals spend considerable time preparing for and completing it, and often require immense energy storage to reach their destination. Over 60% of shorebirds migrate (Colwell, 2010). Several species that breed in the more northern latitudes can migrate great distances to their wintering areas, which can be as far as south of the equator. For example, the bar-tailed godwit flies nonstop on the trans-Pacific route from Alaska to New Zealand, around 11,000 k (6,835 mi). Other species, usually the more temperate and tropical species, can be partial migrants with a mix of year-round residents and migrants in a single population. Examples of these types of migrants include the Pacific Coast population of the snowy plover, black oystercatcher, and killdeer. Populations move various distances among suitable habitats that vary from wetlands to grasslands. These staging areas offer rich food resources for individuals to recoup the energy reserves necessary to complete their journey. The timing can vary according to species, but generally the bulk of shorebird populations move north over a short window of time, from a few weeks to 1 or 2 months. Southbound migrations are more drawn-out and can last several months (Colwell, 2010).

History, habitat, and vulnerability: While some shorebird populations remain stable, many are declining; some species are listed as threatened or endangered. For instance, Eskimo curlews were historically overhunted for meat in the 1800s. This overhunting combined with the destruction of native grass prairies in Central North America contributed to the species decline, and the population has never recovered—the species may be extinct (Colwell, 2010). According the U.S. Fish and Wildlife Service (USFWS), only the snipe and woodcock may be hunted legally; the Migratory Treaty Act protects all other shorebird species in the United States.

Habitat loss is a widespread problem for shorebirds. It is estimated that about 50% of the natural wetlands in the United States have been filled or drained, and we continue to lose about 90.6 k^2 (35 mi^2) of wetlands each year. Native grasslands also have suffered great losses, resulting in restricted habitat for the shorebirds that use those areas as rest stops or nesting in the prairies (O'Brien et al., 2006).

Pollution also poses a major threat to the shorebird population. Chemicals and solid waste have been dumped in estuaries where the birds feed. This contaminates the food and can destroy the habitat. Oil spills can also affect the birds by either contaminating the food source or directly getting on the bird.
Direct competition with humans is another problem for the birds. The beaches and shorelines are critical habitats for feeding or nesting, and although the shorebirds attempt to squeeze in around humans, they often get flushed away from their nest or forage site, inhibiting them from properly gaining the energy needed for migration. Humans have also affected food sources, such as overharvesting the horseshoe crab. Horseshoe crab eggs provide an important food source for several species of migrating shorebirds, particularly the red knots. It is estimated that red knots eat about 18,000 eggs a day, which will double

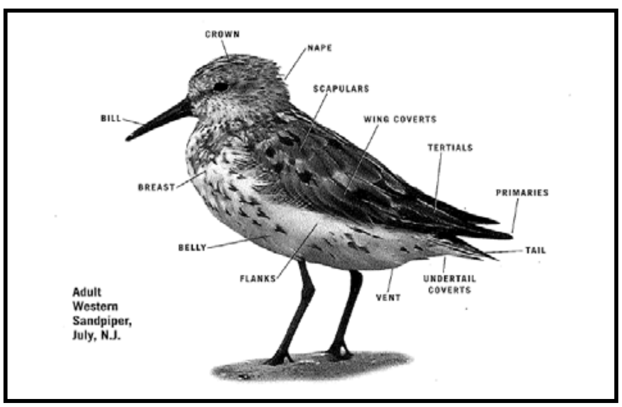
their body weight in order to make their migration. The overharvesting of these crabs has caused declines in the red knot populations (O'Brien et al., 2006).

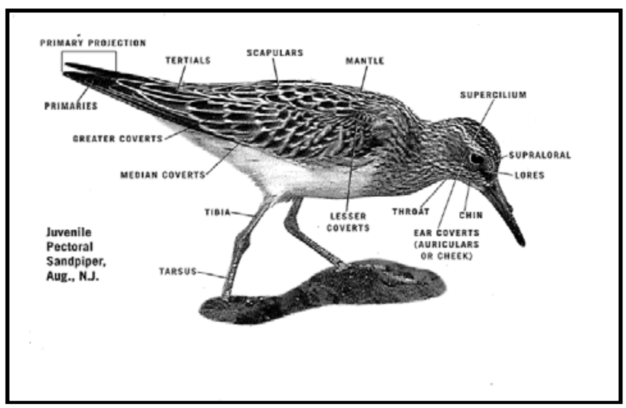
Breeding and life cycle: Most shorebirds in North America breed only in the Arctic and Subarctic, although some are temperate breeders and a few species breed in both boreal and temperate zones. Birds arrive on the breeding grounds in the spring, and immediately declare territory and advertise for a mate by displaying in flight or on the ground. Shorebird display flights exhibit a variety of patterns, styles, heights, and distances.

Most species are monogamous—at least seasonally. Nests are typically simple scrapes on bare earth or in short vegetation. Egg clutches are usually made up of two to four mottled, speckled eggs with a cream, gray, pale green, or brown background. The eggs are pear-shaped so that they pack snugly into the nests and can be easily incubated. Development of the embryo is rapid; eggs hatch 3–4 weeks after fertilization. Hatching may be as short as 17 days in smaller species or as long as 30 days in larger species, such as the lapwings and oystercatchers. Most shorebirds incubate 22–24 days. Egg and chick mortality is high; many predators such as gulls, skuas, owls, raptors, foxes, and river otters feed on the eggs or chicks. Ground-nesting birds are also susceptible to destruction by human activity or livestock that may trample the nests. For the eggs of killdeers and plovers, off-road vehicles may run over the nests on beaches. In salt marshes, exceptionally high tides may flood the nests (Colwell, 2010).

Hatchlings of most species are precocial, which means they are able to run about and feed themselves soon after birth. Most species leave the nest within 24 hours after hatching. They still require parental care to lead them to food sources, help keep them warm by brooding, and protect them from predators. The chicks are unable to maintain their body temperature and usually need to be brooded for about the first 2 weeks. At least one parent will usually stay with the chick until they fledge.

Juvenile feathers begin to appear anywhere from 4–7 days after hatching. The molt to juvenile plumage is complete in 10–30 days, and generally occurs faster in smaller species. Young can fly in 2–6 weeks, and at this point the adults will begin their migration, leaving the chicks behind. The adults leaving may actually benefit the chicks, as it ensures more food is available for the young after the adults migrate. The fledglings will remain on the breeding grounds, feeding heavily and gaining weight, before starting their own migration in the fall (Colwell, 2010).





Figures 1 & 2. Shorebird anatomy (O'Brien et al., 2006)

Definitions of common words that are used when working with shorebirds:

- Peeps: Any of the seven smallest members of the genus *Calidris* (O'Brien et al., 2006).
- Rhynchokinesis: Ability possessed by some birds to flex their upper beak. It involves flexing at a point some way along the upper beak—either upwards, in which case the upper beak and lower beak diverge, resembling a yawn—or downwards, in which case the tips of the beaks remain together while a gap opens up between them at their midpoint.
- Scrape: A type of nest that is a simple depression in the ground or leaf litter.
- Herbst corpuscles: Type of sensory organ that are typically found in the bill, oral cavity, and other parts of the birds. They are thought to be able receive several types of external tactile sensations, such as pain, pressure, and vibration (Berger, 1976).

- Distraction display: Anti-predator behaviors used to attract the attention from an object, typically the nest or young, that is being protected.
- Semipalmated: Having partial webbing between the toes.

Regulations: Native shorebirds are managed by the USFWS under the Migratory Bird Treaty Act. Most of the shorebirds are acquired from local wildlife rehabilitation centers and are considered non-releasable. Please consult with a local representative from USFWS, as well as check for any specific state laws when acquiring or transferring shorebirds that came from the wild.

Chapter 1. Ambient Environment

1.1 Temperature and Humidity

> **AZA Accreditation Standard**
>
> **(1.5.7)** The animals must be protected from weather, and any adverse environmental conditions.

The animals must be protected from weather, and any adverse environmental 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.

Shorebirds are unusual in the way in which they meet energetic challenges of thermoregulation. Shorebirds use more energy compared to other birds, have greater daily energy expenditures, and maintain higher metabolic rates. Basal metabolic rates of various shorebirds are roughly 40% higher than that reported for other non-passerines of comparable size (Colwell, 2010).

Regardless of the temperature at which an exhibit is maintained, birds should always be able to choose from several temperature gradients. These can be created by: (1) screens/netting allowing outside air to recirculate throughout the exhibit; (2) fans to create breezes; (3) exhibit furniture such as plants (dune grass, twinberry, etc.), logs, rocks, etc. to create shade; (4) wide areas of sand where birds can sun themselves; and (5) water sources such as freshwater or saltwater ponds, streams, bathing areas, and misters the birds can use to cool off. Both indoor and outdoor exhibits should have exhibit furniture to provide shelter from bright light or high temperatures. Examples include, but are not limited to: driftwood, beach plants such as dune grass or twinberry, and large rocks. Artificial devices such as shade cloth or panels can also be utilized. Likewise, areas should be provided for birds to loaf in. These areas can consist of open sandy areas or exposed rocks. A period of adjustment, which could range from a few days up to a week, is recommended when moving birds from one temperature extreme to the other. Most importantly, all birds should be given the opportunity to move in and out of warmer/cooler areas.

Cold weather: If institutional area temperatures fall below 0 °C (32 °F), it is strongly recommended that institutions have indoor winter holding facilities. Given the colder and exposed environments many species experience during the year in outdoor exhibits, they will need a heat source provided during colder parts of the year. Cold birds can exhibit the following behaviors: tucking in beak and/or leg(s) into feathers, shivering, fluffing up feathers, and lethargy. In general, most shorebirds are known to tolerate a wide range of temperatures, but care should be given to older birds, which may require a spot heat lamp in cooler temperatures. For young chicks, a spot heat source should also be provided. Hand-raised stone-curlew chicks at 1 month of age in favorable weather have been left outside overnight as long as the nighttime temperature is above 18 °C (64.4 °F) (Cwiertnia, 1999). Heat sources are recommended when temperatures fall below 4.5–7 °C (40–45 °F), particularly if a collection consists of smaller species such as dunlin or snowy plovers that cannot withstand the low temperatures.

The feet of shorebirds may suffer if allowed to remain on frozen ground for prolonged periods. If temperatures are higher than 4.5–7 °C (40–45 °F), but rain and/or wind are present, heat sources such as spot heat lamps should be available to combat the wind chill factor. Saltwater pools inside exhibits do not normally require protection from freezing, but any outdoor fresh water ponds, bathing pans, or drinking bowls will. Shorebirds can go overnight without a water source for drinking, but if water continues to freeze during the day, the water should either be changed frequently or have a heating element added to it. This can be accomplished by the use of heat pads or increased flow to the water container/pool. Again, for institutions with outdoor exhibits that the birds inhabit year-round, procedures should be in place to transfer the collection to holding areas if unusual weather conditions are predicted that could possibly endanger the birds. Even if the institution does not experience cold weather on a routine basis, it is still recommended to have a holding area, as it can be invaluable if the exhibit needs repairs or a bird needs to be confined for medical treatment or from storm protection.

Warm weather: In hot weather, exhibits should offer a balance of direct sunlight and shade to protect birds from excessive heat. There should be enough shade so that all birds in the exhibit have shade available without competition. Excessive heat can also be a potential problem with some birds in that they have trouble dissipating heat. It was found that great knots studied in the wild have to dissipate heat due to: (1) fat loads gained for migration; (2) darker plumage which absorbed more solar energy; (3)

increased metabolism during migratory periods (Colwell, 2010). Exhibit features, such as shade and water sources, mitigate potential heat problems. Water should be provided for bathing and thermoregulation during high temperature conditions. Misters have been used to provide heat relief for temperatures over 38 °C (100 °F).

Avocets and stilts: These species of shorebird have been historically kept in exhibit areas with a range of 0–38 °C (32–100 °F), with appropriate heat and water sources, and shade. If the weather is raining, sleeting, or snowing, the birds may need to be moved to a heated shelter at a higher temperature. The ambient temperature of off exhibit holding should be around 10–21 °C (50–70 °F).

Dikkops: Although the African dikkop is a relatively hardy bird, it is recommended that in extreme climates it should be housed indoors (i.e., if the temperatures remain below 10 °C [50 °F] for an extended period of time) (Holland, 2007). If the weather is raining, sleeting, or snowing, the birds may need to be moved to a heated shelter at a higher temperature. The ambient temperature in the holding area should be in the range of 10–15.5 °C (50–60 °F). The heat bulb should be enclosed by a wire mesh to protect the bird from contacting the bulb with any part of its body.

Gull and terns: Gulls are the ideal inhabitants of outdoor exhibits due to their tolerance of many weather conditions as long as open water is available. A breeding group of silver gulls that are native to Australia were kept at a range of temperatures from -28–38 °C (-18–100 °F) (Bohmke & Fisher, 1992). If temperatures are below -27 °C (-18 °F), birds should be brought indoor to an off-exhibit holding with a temperature range of 5–21 °C (41–70 °F). If it is a drastic temperature change from the outdoor to the indoor holding, gradually acclimate them to new temperature over a period of 3–7 days.

Jacanas: These birds are sub-tropical and it is recommended that they be exhibited at an ambient temperature of 20–30 °C (68–86 °F) during the daytime, and at 20 °C (68 °F) during the nighttime. The recommended water temperature within indoor or outdoor enclosures is 26 °C (75 °F), and a high relative humidity of 80–100%. Since jacanas are from sub-tropical regions, the exhibits should reflect the humidity and temperatures from those habitats.

Lapwings: Vince (1996) reported successfully keeping lapwings outside in snow and ice. During winter evenings they were shut in their heated housing, but let out the next day with no ill effects. More research is needed for specific temperature ranges.

Oystercatchers: Oystercatchers are known to be tolerant of a wide range of temperatures from -7–27 °C (20–80 °F), although a narrower temperature range needs to be considered, such as an optimum range of 4.5–18.5 °C (40–65 °F). Facilities that house oystercatchers in ambient conditions should always maintain the option of bringing them indoors during the colder months. It had been determined that -7 °C (20 °F) is the lowest temperature that they can safely tolerate, so when ambient conditions start approaching this temperature, moving the oystercatchers indoors is recommended. Heat lamps should also be provided during the colder months in case of an unpredicted cold snap or wind chill effect. Oystercatchers have been held successfully in zoos and aquariums with relative humidity levels between 20% and 90%, and may be considered to have a high tolerance to fluctuating humidity.

Phalaropes: In the wild, this species of bird can be found in areas that range from 4.5–21 °C (40–70 °F). Institutions should try to replicate this temperature range. They have been successfully kept by a limited number of zoological institutions so there is limited data on their tolerance range in *ex situ* situations; more research is needed.

Sandpipers and plovers: Sandpipers and plovers have been exhibited successfully in both indoor and outdoor exhibits throughout North America. Sandpipers and plovers are cosmopolitan migrants and also experience a wide range of temperatures. They have been kept in outdoor exhibits in temperatures ranging from -8–46 °C (18–115 °F). These are the extremes; most outdoor exhibits have ranges of 0–32 °C (32–90 °F). For the most part, sandpipers and plovers do well with higher temperatures and are frequently seen sunbathing even during summer months (M. Carlson, personal communication, 2012). However, since most North American species migrate to Central and South America for the winter, any outdoor exhibit should have heating devices such as heat lamps, radiant heaters, or heat pads if temperatures drop below 4.4 °C (40 °F). Sandpipers and plovers exhibited in outdoor enclosures in parts of North America with harsh winter conditions should be transferred indoors for the duration of the season into a holding area where they can get warm and remain out of the elements. Although sandpipers and plovers sometimes encounter snow in their breeding areas in the wild, it is advisable to take them off exhibit if snow might endanger the exhibit netting and/or if the snowfall is high enough to make it difficult for birds, particularly smaller species, to move towards heat sources or food. Humidity has not been studied or recorded in zoos and aquariums, but given the wide range of environments sandpipers and plovers inhabit, their humidity range is probably quite variable. Where humidity requirements were not listed above for specific species, further research is needed to determine humidity ranges and air turnover rates.

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

> **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. All mechanical equipment must be kept in working order and should be under a preventative maintenance program as evidenced through a record-keeping system. Special equipment should be maintained under a maintenance agreement, or a training record should show that staff members are trained for specified maintenance of special equipment.

There are a number of facilities that successfully house shorebirds 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 in addition to an air testing protocol and schedule of 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 facilities with climate-controlled shorebird habitats, 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 to maintain regular checks.

1.2 Light

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

Light is essential for bird health in three ways. First, light affects birds on a daily basis. A basic daily light schedule for birds is 12 hours of daylight and 12 hours of darkness, but can vary according to the season and the birds' natural range. For instance, a longer schedule of daylight may be needed to induce breeding. During darkness, birds need a restful sleep to minimize stress, reduce fatigue, and maintain a strong immune system. The second way that light affects birds is on a seasonal basis. The photoperiod is one of the cues the birds use to induce changes in breeding, molting, and when to migrate. The third way is on a physiological level, as birds need light to help synthesize vitamin D and calcium.

When possible, providing access to natural sunlight is preferable. If natural daylight is not available, then full spectrum lighting may be used. Full spectrum lighting would be best as it promotes appropriate molting and plumage quality. If full spectrum lighting is not available, indoor exhibits have used a variety of other types of lights successfully including fluorescent, low pressure sodium, and metal halide skylights in conjunction with artificial lighting. For dim light or nighttime conditions, blue-colored light bulbs or gels that create a good nighttime light similar to moonlight should be installed within the exhibit for use in avian or life support emergencies. Full spectrum bulbs that emit light in the ultra violet wavelength mounted 30–46 cm (12–18 in.) above the bird should provide the basic light requirements (Foster & Smith, 2010). Ideally, all artificial lighting—especially emergency lighting—should be on dimmer switches to avoid startling birds more than necessary should the need arise to turn lights on and off during dark hours. The day length can vary according to the season and the species, and care should be given to provide a dawn and dusk period as well. Regular and consistent light cycles are vital.

In regards to outdoor exhibits, any exhibit work should ideally take place after natural light has lit the area. Outdoor exhibit lighting cycles should not extend beyond what is normal for that time of year. In general, it is recommended that outdoor exhibits remain closed for night events in order to help keep birds on their natural light cycle. In addition, if the shorebird exhibit is indoors near an area where the institution regularly hosts nighttime events, adjustable curtains can be installed on the exhibit windows for situations that require light to be blocked at night.

Avocets and stilts: These birds are found in a wide range of latitudes, but typically longer daylight hours should be provided during the summer and shorter day lengths during the winter. If possible, natural daylight cycles should be mimicked.

Dikkops: In the wild, they range widely through sub-Saharan Africa. There are no standards for light requirements with chick rearing for the spotted dikkop. However, in the kori bustard it has been documented that three chicks developed cataracts after being reared under standard incandescent bulbs. No cataracts were observed after the bulbs were changed to 60W dull-emitter bulbs (Bailey et al., 1997).

Gulls and terns: Natural daylight cycles should be mimicked.

Jacanas: Light suggestions in relation to this species are to mimic the light patterns in the regions in which the jacana is naturally found; Southern Africa typically has 12 hours of sunlight. Exceptions to housing jacanas in natural light conditions would be the 30-day quarantine, temporary housing, medical reasons, or transportation. More research is needed in this area.

Oystercatchers: Oystercatchers occur in a wide range of latitudes from Baja, CA to South Central Alaska. There is a significant difference in the seasonal day lengths between these areas, allowing for some flexibility in mimicking the day lengths in indoor habitats. Even if the exhibit has natural sunlight, the lighting should also be supplemented with full spectrum lights. Placing lights on timers will allow for mimicry of the natural light cycle and can be adjusted for seasonal changes.

Phalaropes: They should have longer daylight hours during the summer and shorter ones during the winter to promote proper molting and breeding activity.

Sandpipers and plovers: Wild sandpipers and plovers experience a wide range of changing light intensities and durations depending on the species. Species that winter in North America (i.e., Long-billed curlews) will experience shorter photoperiods than those that winter in Central or South America (i.e., dunlin), and will have more variability in light cycles. In zoos and aquariums, piping plovers should have supplementary or manually controlled lighting within exhibits if necessary. In one AZA-accredited institution, the piping plover exhibit contains skylights that provide the normal lengthening and shortening of daylight. These skylights are supplemented with metal halide, multi-vapor lamps of 400W each. An artificial timer is used to adjust light durations by 15–30 minutes once a week during early fall or spring. This ensures that the maximum or minimum additional light durations are achieved.

At this institution, maximum light durations of 14 hours are provided between March and May, and minimum light durations of 10 hours are provided between September and November (Brown et al., 2006). Outdoor sandpiper and plover exhibits use ambient light conditions with artificial lights for emergency use. Some exhibits, due to their orientation, have supplemental lighting for darker days or periods of the day. Normally, outdoor exhibits have the same photoperiod as the species local geographical location. More research is needed to determine if and how light cycles affect premigratory foraging and weight changes in birds held in zoos and aquariums.

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, pinnipeds, cetaceans, and other aquatic animals. A written record must be maintained to document long-term water quality results and chemical additions.

Water quality: Water quality can be assessed for bacteria levels. Pertinent water parameters include testing for pH and coliforms. Records should be kept according to AZA standards 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 informed if there are any concerns or anticipated needs for changes. To meet basic water needs, a low pan of water that birds can dip their bill into and drink from will suffice in an exhibit. This water should be cleaned at least once a day, and more often if droppings or food particles soil the water.

If an exhibit has small pools, these should be drained and cleaned at least every 3 days. Moving, recirculating water promotes more natural behaviors such as bathing and foraging. These flows can assist with: (1) limiting surface buildup that could affect bird's plumage; (2) reduction/elimination of mosquito breeding; and (3) keeping water clean for drinking/bathing. If there is more than one pool in the habitat, each pool should be tested and the water samples collected from at least 60 cm (2 ft) below the surface if possible. Use a sterilized container and refrigerate the sample immediately afterwards. It is recommended that the coliform counts should be below 1000MPN per 100 ml.

If a pool fails the water quality recommendations, appropriate life support staff should be informed immediately and measures to drain and clean the pool should be taken. Additionally, filtration, cleaning, and water turn over may need to be increased. If pool water is recirculated through filters, using sand and gravel filters with ozone and/or UV sterilization treatments is 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 shorebirds.

Air quality: In outdoor exhibits, there are no standards for air quality; however, air quality can be improved by allowing for natural air flows/currents either through exhibit design, care in arranging exhibit furniture, or by the use of screens or air vents. To provide good air quality in indoor holding areas there should be some sort of air ventilation such as screened vents, a ventilation system, or fans to move fresh air. Air turnover rates should be maximized in indoor facilities to maintain air quality while still maintaining recommended air temperatures. The specific design of an air-system needs to balance the tradeoffs between filter efficiency and airflow, fresh air exchange, and temperature regulation capacity (Beall et al., 2005). For shorebirds housed in closed indoor habitats, an HVAC system with air filters would be necessary to maintain good air quality, especially when housed with multiple birds.

Air should be tested for fungal spores about every 6 months. If fungal problems arise, a good quality HEPA filter system may reduce infection rates. Normal ranges and high levels for fungal air spores are: (1) indoor air spore numbers should be lower than outdoor or outdoor exposed areas; (2) environmentally controlled spaces are expected to have a greater than 90% reduction in air spores; and (3) any *Aspergillus* sp. are noted and addressed if need be; any result over 60 cfu/cubic meter is taken very seriously. If high counts are found, notify vet staff immediately. They may choose to put the birds on an anti-fungal med. In addition, removing the birds from the area needs to be considered, and trying to filter the air to help remove the *Aspergillus* sp. and/or modify the exhibit to decrease the counts. Air quality can be tested by using Petri dishes with appropriate media placed randomly in the exhibit, ensuring that the birds do not have access to the media. All air quality test results and measures should be recorded. Further research is needed to determine minimum water exchange rates and air exchange rates for shorebirds. See Appendix K for sample air spore sampling protocol.

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. Shorebirds 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 shorebirds are often seen resting comfortably in close proximity to large groups of visitors. Sounds that are not routine may be stressful to the birds, such as unscheduled maintenance work or construction projects. 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 stress such as running, crouching, or standing stock still, and to 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 sound-proof box. There may be waterfalls in an aquatic habitat exhibit near a shorebird exhibit, the sound of which should be monitored to make sure it does not contribute to pushing the total ambient sound levels above normally accepted levels. Events in close proximity to shorebird habitats that utilize loud music and/or PA systems need to be considered, 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, the intermittent nature of visitors to a habitat is not considered an important noise factor for most shorebirds. 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.

Jacanas: It is unknown what amount of sound and vibration exposure would be considered safe for jacanas. As with any wild animals, it is best to keep sounds and vibrations to a minimum. More research is needed in this area.

Oystercatchers: Oystercatchers are no more sensitive to noise or vibration than humans. Up to 80 decibels (dB) is considered a normal and acceptable noise level for long term exposure. Oystercatchers themselves have a very loud, shrill call that can be heard up to a mile away by humans. This call may be considered loud for some other species to tolerate. Further research is needed to determine what level of noise would be too high for oystercatchers to tolerate for extended periods.

Sandpipers and plovers: Judging by their behavior on exhibit and in holding, these birds are affected by sounds and vibrations. When a loud, unexpected sound occurs, both groups of birds will often stop their normal behavior and stand rigidly; however, further research is needed to more accurately determine the sound sensitivity of sandpipers or plovers. Although these birds can become acclimated to visitor noise, they should be shielded from loud intermittent noises either through distance from visitor pathways, barriers such as windows, or volunteers who can monitor crowd noise levels whenever possible. Birds should have access to areas of the exhibit where they can shield themselves from loud noises if physical barriers are not present. Examples of noises that sandpipers and plovers should be shielded from are pumps, pressure washers, and equipment being used late at night when they are normally resting/sleeping.

It is known that shorebirds and plovers are sensitive to natural vibrations. Sandpipers have large numbers of Herbst corpuscles in their bills, which are used for detecting prey items, and plovers are known for tapping the substrate with their foot to stir up prey. To encourage these natural behaviors it is recommended that large amounts of vibration be minimized as much as possible. Additional research on the hearing of and sensitivity to sound and vibration in shorebird species could provide more quantitative guidelines for this species.

Chapter 2. Habitat Design and Containment

2.1 Space and Complexity

Careful consideration should be given to exhibit design so that all areas meet the physical, social, behavioral, and psychological needs of the species. Animals should be presented in a manner reflecting modern zoological practices in exhibit design (AZA Accreditation Standard 1.5.1). All animals must be housed in enclosures and in appropriate groupings which meet their physical, psychological, and social needs. (AZA Accreditation Standard 1.5.2).

AZA Accreditation Standard

(1.5.1) Animals should be presented in a manner reflecting modern zoological practices in exhibit design, balancing animals' functional welfare requirements with aesthetic and educational considerations.

AZA Accreditation Standard

(1.5.2) All animals must be housed in enclosures and in appropriate groupings which meet their physical, psychological, and social needs. Wherever possible and appropriate, animals should be provided the opportunity to choose among a variety of conditions within their environment. Display of single specimens should be avoided unless biologically correct for the species involved.

Species appropriate behaviors: Listed below are specific species behaviors for each of the major groups of shorebirds.

<u>Avocets and stilts</u>: These birds spend a lot of time wading in shallow pools, and avocets can be seen scything in the water for food. They also spend a lot of time resting on land, often with one leg tucked up underneath their body.

<u>Dikkops:</u> Although dikkops are strong flyers, they spend most of their time walking on the ground. In the wild, dikkops tend to be nocturnal or crepuscular. In zoos and aquariums, they have typically adjusted to being active during the day. For instance, while studying double-striped thick-knees housed in a zoological institution, Pate (1983) found that these birds were breeding, laying eggs, and engaging in agonistic behavior during the day.

<u>Gulls and terns</u>: They are generalist feeders that can swim, fly, and walk very well. They can hover and take off rapidly with little space.

<u>Jacanas:</u> In zoos and aquariums, jacanas should be given the opportunity to perform species appropriate vigilance behaviors. A jacana will frequently turn its heads on its side and peer at the sky, to track aerial predators. They also stand upright with their necks outstretched forward at a 60° angle above horizontal, the feathers on the back of the head raised and the bill pointed downward, as they peer into the water apparently looking at potential aquatic predators. The elongated toes and claws of jacanas allow them to walk on floating aquatic vegetation, where they glean food, establish territories, pair, rest, raise their young, and fend off potential predators. Jacanas are also good swimmers and can swim beneath the surface. When jacanas fly, which is infrequently, they will only do so for short distances.

<u>Lapwings</u>: While quite capable of flying long distances, lapwings are mostly ground-dwelling birds. They live in varied habitats like marshes, mudflats, pastures, and fields. They stamp their feet to draw invertebrates to the surface so they can feed on them.

<u>Oystercatchers</u>: Oystercatchers inhabit beaches along sandy, gravelly, and rocky coastlines, feeding on small invertebrates and vertebrates in the intertidal zones, using their long sharp beaks to probe for food on the beach, in shallow water, and on the rocks. Providing an exhibit that allows for these feeding habits is essential and would require a sandy/gravelly/rocky area, with shallow and preferably fluctuating water levels. Most oystercatchers are housed in covered habitats allowing them to be flighted. Although the ability to fly is beneficial to the oystercatchers, non-flighted oystercatchers have also been held successfully in zoos and aquariums. A sand or gravel beach is beneficial in that it allows for natural wear and tear on the fast growing bill, eliminating the need for regular bill trimming. Oystercatchers nest on stony beaches so an area (at least 1.5 m x 1.5 m [5 ft x 5 ft]) covered in similarly sized (2.5–5 cm [12 in.] diameter) pebbles would provide ample nesting area. When resting, oystercatchers habitually perch on a vantage point slightly higher than the surrounding beaches such as a prominent rock from which they can scan their territory. Providing a choice of such vantage points will ensure the oystercatchers feel comfortable in their habitat.

<u>Phalaropes</u>: In general, phalaropes spend most of their time swimming and very little time on land, except for the summer nesting season. If food is provided in shallow water, such as freshwater tubifex, they can

be stimulated to engage in their natural feeding behavior of spinning. The spinning behavior consists of spinning in the water, creating a vortex to bring food up to the surface. They will also pick prey from the surface of the ground.

Sandpipers and plovers: Behaviors most commonly exhibited by both groups include, but are not limited to: (1) chasing; (2) pecking/stabbing; (3) body slamming (dunlins); (4) grabbing and pinning down exhibit mate. Sandpipers and plovers each have two distinct methods of searching for food, which are probing and stitching. (There is some overlap; neither group exclusively uses one method.) Many sandpiper species such as the long-billed dowitchers employ stitching. Other species with long, decurved bills such as long-billed curlew will use their bills to probe for food in invertebrate tunnels or under the edges of rocks and driftwood. Black and ruddy turnstones will flip over small stones or plant materials in search of food underneath. Plovers generally use a visual method of foraging in which they raise their heads, look for prey and upon sighting, will quickly walk over and peck it. Both groups exhibit walking, wading, running, flying, and swimming. Of the five, swimming is the least observed while flying is the most prevalent

Table 3. Summation of species-appropriate behaviors (Avocets=1, Jacanas=2, Lapwings=3, Oystercatchers=4, Phalaropes=5, Plovers=6 Sandpipers=7, Stilts=8, Thick-knees=9)

General behavior categories	Individual behaviors	Description	Bird groups exhibiting behavior
Comfort and maintenance	Bathing	Birds can bathe by standing in shallow water, by hopping in and out of the water, and/or bathing in the rain.	All groups
	Loafing or sunbathing	While sunbathing, a bird typically fluffs its feathers, leans to one side, opens its bill, spreads its tail feathers and either droops or extends one wing or both wings.	All groups
	Preening	Birds use their bill to straighten the feathers on their breast, neck tail, legs, or wings. Preening is performed while sitting or standing and bird's eyes are often closed.	All groups
	Scratching	Bird scratches body (i.e., neck and head areas) using a toe.	All groups
	Sleeping	Postures can vary among birds; most common posture involves turning the head backward and resting the bill on, or buried in, the scapular feathers. Some species commonly sleep standing with one leg held up, tucked underneath the body.	All groups
	Stretching	Bird stretches its leg, wing, or body, and typically involves stretching the wing and leg on the same side of the body.	All groups
Feeding and foraging behaviors	Bill pursuit	Individual rapidly opens and closes bill while moving it erratically along shallow water surface.	1
	Defecation	Fecal matter is excreted mainly while walking, or during short pauses.	All groups
	Drinking	Birds consume water by using tongue and/or sucking motion to suck in water.	All groups
	Filtering	Bill opens and closes rapidly while moving over mud; feeding birds pauses to swallow.	1, 5
	Hammering	Using the bill to rapidly hit a bivalve shell in order to create a hole and/or break open the bivalve and then extracting the meat.	4
	Pecking	Visual search for prey while standing still or walking slowly, followed by a quick jab of the bill to capture prey on mud or near water surface; head does not go under water.	1, 2, 4, 6, 7, 8
	Plunging	Head and upper breast enter water to capture food from within the water column.	1, 6, 7, 8
	Probing	Using the bill to poke into the substrate or under rocks, logs,	4, 6, 7,

		etc. to search for prey items.	
	Scraping	Extending of neck to move bill 5–10 cm forward through the mud followed by swallowing.	1
	Spinning	While swimming, top-like spinning on the surface of the water, rarely seen in moving water. Thought to stir up prey from bottom in shallow water or stimulate prey immobilized in cold water.	5
	Stabbing	Waiting for a bivalve to gape, followed by inserting bill into bivalve and cutting its adductor muscles so bivalve falls open, enabling bird to extract the meat.	4
	Stomping	Commonly pats the ground or bottom in shallow water with one foot with quivering motion. It is thought this behavior was intended to bring prey items to the surface.	6, 7
	Scything (single)	Bill is held open slightly at the level of the muddy substrate and moved from one side to the other; one step occurs between each swipe, and the swipe moves toward the leading foot.	1, 8
	Scything (multiple)	Resembles single scything except the bill is not raised in between steps.	1, 8
	Scything (dabble)	Similar to single scything, but is performed while swimming; bird tips up from a swimming or deep wading position to bring bill in contact with substrate. A backward kick of the feet maintains the tipped position during dabble scything.	1
	Stitching	A tactile method of foraging by which the bird repeatedly probes soft substrates with its bill while slowly walking along the area it is searching.	7
	Picking	Precisely picking prey off the surface of the ground with the bill.	1, 2, 5, 8, 9
	Turning stones, plants	Flipping small stones or pieces of seaweed/plants over so bird can capture prey underneath object.	7 (Ruddy & black turnstones)
Locomotion	Flying	Birds take off into the air by flapping their wings.	All groups
	Swimming	Swimming at the surface of the water.	1, 5, 6, 7, 8
	Walking	Bird moves about on the ground at a leisurely pace.	All groups
	Wading	Wading through shallow water from depths from 1/4 in. up to the bird's chest.	1, 2, 5, 7, 8
Miscellaneous	Agonistic	Any behavior appearing in conflict between animals, including fighting and escape behavior. Can include chasing, pecking/stabbing, body slamming, grabbing, and pinning down another bird.	All groups
	Head bobbing	In-between foraging bouts, bird lifts head in a jerking motion. This motion results in the whole body including the tail, tilting back. Occasionally quiet vocalizations accompany this behavior. This behavior may be communicative in purpose.	4
	Skyward looking	Birds frequently look around for predators, often with head tilted to one side, looking up in the air.	All groups
	Tail wagging	Mostly executed while walking and sometimes accompanied by defecation. A swift side-to-side movement of the tail, often three times to each side.	4
Reproductive	Chick care	Birds often seen brooding the chicks or leading them to food, protection from other birds. Often get very territorial when defending chicks and/or eggs.	All groups

	Courtship	Any behavior pattern that brings the exes together and leads to copulation.	All groups
	Distraction displays	Any behavior pattern of an adult bird that tends to divert an intruder from the eggs or young. A common pattern is the "broken-wing" display, where the bird typically runs off a short distance, falls to the ground, leans to one side, spreads its tail and rapidly flaps one wing or both wings, while emitting alarm notes. The bird may then stand up, run a few feet, and repeat the display.	1, 6, 7, 8, 9
	Incubation	Sitting on eggs after all have been laid. Males and/or females will incubate eggs depending on species.	All groups
	Mating and/or copulation	Copulation is the act of transferring sperm from the male to the cloaca of the female, which is accomplished by cloacal contact as the male balances on the female's back.	All groups
	Nest building	Construction of nest by parent(s) by various methods depending on species.	All groups
	Territoriality	Limited area defended by a bird, especially against members of its own species and sex during at least part of the breeding cycle. It may defend its mate, young, nest, or food supply. Territories are defended against intruders not only by physical attack or threat of it, but often merely by vocalization or the conspicuous presence of the defending bird.	All groups
Social behavior	Aggressive displacement	One bird chases another bird by lowering its head, ruffling its plumage slightly and aiming its body towards it. Pursuit will often continue until the other bird is out of sight.	All groups
	Non-aggressive displacement	One bird walks towards another bird causing the second bird to vacate its position and move elsewhere.	All groups
	Fighting	Birds grasp each other's bills or one bird will grab the back of the head of the other bird. Birds may shove each other. Wings will be spread, feathers fluffed, and tail may be up.	All groups
	Tail lifting	Tail feathers will be lifted up (threat display).	9
	Threat posture	Bird may do one or all of the following: lift tail, spread tail feathers, spread wings, lower head, extend neck.	All groups
Thermo-regulation	Fluffing	Fluffing of body feathers in order to trap and warm air underneath them.	All groups
	Standing on one leg	Bird will stand on one leg and keep the second one tucked against its body and/or under its feathers in order to keep leg warm. Bird will sometimes hop several times in this position before placing tucked up leg on the ground again.	1, 6, 7, 8, 9

Enclosure design: When designing exhibits, it is important to keep the species' natural foraging behaviors in mind and provide opportunities for them to exhibit natural behaviors, such as probing in sand/mud, wading, swimming, pecking, turning over stones, etc. Also, as most wild habitats can be static, providing opportunities to move things around, such as sand dunes, plants, driftwood, tidal changes, waves, water motion, etc. can help provide the opportunity to simulate natural habitats. Regular keeper observations are essential in monitoring overall shorebird health and behavior.

For monitoring shorebirds, it is useful to band individual birds with identification. If the birds are normally flighted, they would need to be flight restrained in an open environment. As most of the shorebirds come from rehab facilities and are non-releasable due to wing injuries, many of the birds will be unable to fly. Therefore, 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 required.

In regards to pools, all should allow easy access in and out of water. If deep-water pools are in the exhibit, sloped areas should be created so the bird will not be trapped in the pool. In regards to glass windows, they can be hazardous when birds fly into them. Installation of adjustable curtains should be considered so that they can be lowered when catching birds. This is to help prevent the birds from crashing into the windows. If glass is used, it should be taped or soaped when new birds are introduced to the exhibit until they are able to recognize it as a barrier. If glass is used anywhere in an avian exhibit, measures should be taken to ensure that reflections in the glass from lighting (natural or artificial) do not create the illusion for the birds that they can fly through the glass to the other side, as this kind of impact would have negative implications. When piano wire is used as a barrier, the wire should be close together so birds cannot be caught between strands or slip through the strands. Below are specific enclosure design recommendations for each group of shorebird species.

Avocets and stilts: These birds should be provided with wading pools with easy in and out access. The size of the exhibit can vary, depending on the number of total birds in the aviary. Plenty of space should be provided to give birds areas where they can be visually separated from each other, and get away from keeper during exhibit maintenance.

Dikkops: Generally, dikkops are appropriately housed in a large free-flight aviary with lots of choices for resting and nesting places. They can also be housed in smaller enclosures as long as there is a mix of open spaces and spaces with cover. Holland (2007) suggests that dikkops be kept in an aviary of 10 m x 3 m x 2 m (33 ft x 10 ft x 6.5 ft). It is best to house these birds in a covered area since this protects them from predators and parasite transmission from wild animals.

Gulls and terns: Five species of gulls have been kept successfully at an AZA-accredited institution in an aviary that was 46 m x 17 m x 23 m (152 ft x 55 ft x 75 ft). A breeding group of silver gulls was kept at another institution in an aviary 69 m x 25.5 m x 15m (227 ft x 84 ft x 50 ft). Another AZA institution successfully maintained a flock of herring gulls at liberty for many years (Holland, 2007). Another institution housed several species of gulls in an aviary constructed over a natural rock formation (Holland, 2007).

Jacanas: The need for a land area, and a large surface of water covered with aquatic, floating vegetation is vital to the keeping and breeding of jacanas. Jacanas live in a water dependent habitat. The water temperature for all indoor and outdoor pools should be at least 25 °C (75 °F). Recommended types of floating vegetation to be used include watersalad (*Pistia strateoides)*, waterlilies (*Nymphaea)*, *Ludwigia, Salvinia, Potamogeton,* and *Polygonum.* Other vegetation may be used for hiding but not for nesting (e.g., bulrushes, reeds, papyrus, and robust stands of water hyacinth). Jacanas prefer low-profile floating vegetation, combined with a few dispersed emergent. In addition to floating vegetation, dense vegetation hanging over the water is recommended since this attracts all kinds of insects and promotes natural foraging.

Lapwings: The ideal lapwing exhibit will have plenty of open, sandy areas combined with short grass areas. In addition, the lapwing exhibit should have an area of well-drained turf and a gradually deepening pool with running water (Vince, 1996). A few shrubs will provide shade and a sense of security (Holland, 2007).

Oystercatchers: Sandy, gravelly, or rocky beaches are an essential part of oystercatcher habitats and should be provided in their exhibit. Shallow water of 2.5–7.6 cm (1–3 in.) on the edge of the beaches where food can be offered will encourage natural foraging behaviors. A gravel or pebbled area is necessary for nesting. It would be advantageous if this pebbled area were visually blocked from the rest of the habitat by driftwood or similar visual barriers so as to give the nesting birds privacy.

Phalaropes: Phalaropes require plenty of water and space to swim; they prefer access to both salt and freshwater. They should have a gradual slope in and out of the water, and the sloping area should be texturized to prevent slipping, but not so rough that they will scrape their feet. The water depth should be a minimum of about 13 cm (5 in.). Providing food in shallow water will encourage natural foraging behaviors. They can be kept in a mixed aviary, and should be kept with a minimum of three other species of phalaropes, plus other shorebirds of similar size.

Sandpipers and plovers: Roof or netting height should be set at a level that allows the birds to fly without hitting them. When startled, many species will fly almost 2.4–3 m (8–10 ft) straight up, requiring overhead structures to be higher than this distance. Large open areas where birds can lie down, be able to see around them, and have access to have either natural light or an artificial heat source should be provided. Large logs/driftwood, large rocks, fence posts, and tall hills for birds to stand upon are also highly recommended. Territoriality among sandpipers and plovers can be decreased by offering: (1) adequate space; and (2) spaces broken up by exhibit furniture (e.g., large rocks or boulders, driftwood logs, grasses, dune grass, hills, variety of vegetation, etc.) where birds can hide from an aggressor. The same furniture also promotes resting behaviors and gives individuals cover from the public.

Additionally, in these types of enclosures, it is recommended that various water sources such as bathing pans, shallow streams, ponds, and large bodies of water, wide areas of shallow depth be provided (preferably with running water). For example, waves in larger bodies of water would help to promote the quick walking/running behavior of sanderlings. Both freshwater and saltwater should be available. To promote longer billed species to probe deeply for food, it is recommended that food dispensers (i.e., mealworm or cricket dispensers), logs and/or plants to scatter food around and within be provided. Whole or sections of beach consisting of small stones (2.5–5 cm [1–2 in.]) should be available to promote the stone flipping behavior of black and ruddy turnstones.

Enclosure complexity: Typically, shorebirds are kept in small numbers of about two to four of each species, but with a mix of other shorebirds or non-shorebird species. Typically, most shorebirds are territorial. If paired, they can become aggressive during spring with other birds. Often, during breeding season, the pairs may need to be removed from the other birds, especially if they have access to other smaller, non-flighted birds.

<u>Avocets and stilts</u>: As they can be aggressive birds, food and wading areas should be spread out in a variety of places to allow multiple birds the opportunity to access them. Enough food and wading areas should be provided so that the aggressive birds cannot guard all of the areas. The exhibit should be designed to make sure that all birds, especially small, non-flighted birds, can get away from aggression and not get trapped in a corner.

<u>Dikkops</u>: Dikkops can show territorial behavior so it is important that multiple food and water areas be present when they are housed with other birds. Unlike other shorebirds, dikkops are very terrestrial and only need water for regular drinking and bathing purposes. It is beneficial to give them a dry, pebbly area and areas of low vegetation (Vince, 1996).

<u>Gulls and terns</u>: This group of birds can be aggressive to other birds, but if the exhibit is designed properly they can be successfully housed with other birds. One successful exhibit at an AZA-accredited institution has housed grey gulls, Inca terns, and Humboldt penguins together with breeding success from all species. This exhibit is roomy, well lit, and made to represent an abandoned guano mine on a sea cliff (Lindholm, 2007)

<u>Lapwings</u>: If the lapwing species is more terrestrial in nature—like the crowned lapwing—then the enclosure should have open sandy areas with mowed lawns and a few shrubs for shade and shelter. If the lapwing species is more aquatic—like the blacksmith lapwing—then a shallow pond a few inches deep with a mud bottom and a few sedges planted around the edge of the pond will provide an ideal environment (Holland, 2007).

<u>Oystercatchers</u>: In the wild, breeding pairs tend to be aggressive towards other oystercatchers. They will protect their territory against other pairs, as well as other bird species. This tendency is even stronger in zoo settings where the subjects of their aggression cannot fly away as in the wild. For this reason, oystercatchers are generally kept in low numbers in zoological institutions, and ideally in pairs.

<u>Phalaropes</u>: To help decrease the chance of sores developing on their feet, the amount of walking they have to do should be minimized by designing their exhibit with plenty of land space along with a food source placed close to the water. The water space needs to have plenty of access points that are easy to get in and out of.

<u>Sandpipers and plovers</u>: Exhibits with these birds should have multiple food, water, and bathing pools available. It is recommended that there be a variety of potential hiding places where a harassed or threatened bird can hide and/or get out of visual sight. Hiding places can be provided by exhibit furniture such as plants, driftwood, boulders/rocks, sand drifts, etc. Birds should have enough space so they cannot be trapped in an area with no escape when a keeper is working in the exhibit, or if another bird

chases it. If the exhibit pool is large and deep enough, it can be used as a barrier for non-flighted birds (if they need to be separated). The beaches on each side of the pool can function as separate exhibits, and birds can be transferred between them depending on their interactions with each other. If breeding is planned, adequate space is required to give breeding pair(s) room for their nest. The amount of space around a nest can vary depending on the species, with smaller plovers and sandpipers requiring 0.914 m (3 ft) and larger species needing more room.

Water sources: All water areas should be designed with easy access in and out of the water, making sure the substrate is not slippery or too rough that it could scrape their feet. The depth of the water should at least allow the bird to stand about 1/3 of its height. For more species-specific information, see below.

<u>Avocets and stilts</u>: These birds have been kept successfully in just freshwater, but observations from one AZA-accredited institution revealed that their feet seem to do better when they also have access to saltwater wading pools. Access to saltwater has been shown to reduce foot problems like bumblefoot in wading birds (Holland, 2007).

<u>Gulls and terns</u>: This bird group is highly adaptable, but if possible, access to fresh and saltwater would be ideal.

<u>Jacanas</u>: Again, jacanas live in a water dependent habitat. The water temperature for all indoor and outdoor pools should be at least 25 °C (75 °F). A large surface of water covered with aquatic, floating vegetation is vital. If the water surface ratio is relatively small, the species will be limited to monogamous breeding. Ideally this should be avoided, but it is an area requiring more research.

<u>Lapwings</u>: For lapwings, a shallow pond surrounded by a few plants is highly recommended. Fresh, flowing water is best to prevent botulism, but a pool that can be cleaned daily is acceptable. Bumblefoot can be averted by placing the food bowl in the center of a shallow footbath containing a saline solution of 3.5%. This strengthens the skin on the feet (Holland, 2007).

<u>Oystercatcher</u>: These habitats should allow access to shallow water (2.5–7.6 cm [1–3 in.] deep), preferably saltwater. It should be considered essential for oystercatchers held in zoos and aquariums to have access to saltwater. Oystercatchers have salt glands that enable them to extract and secrete salt from water they consume, and if they do not have access to saltwater, these glands may become inactive, which can lead to secondary health issues. This has not been documented in oystercatchers yet, but has been in other seabird species. If providing saltwater in a pool is not feasible, providing a bowl of saltwater (at the same salinity as sea water [i.e., 3.5%] would be sufficient). It may be necessary to serve some of the oystercatchers' food in this saltwater bowl to encourage use.

<u>Phalaropes</u>: This species prefers access to salt and freshwater pools. The size of the pool should be large enough that all the phalaropes can fit in it at one time, with about .09 m^2 (1 ft^2) of space per bird. Each pool should be designed with easy access in and out of the water. The depth of the pool should be deep enough for the birds to swim, roughly a minimum of about 15.2 cm (6 in.).

<u>Sandpipers and plovers</u>: Both groups should have access to both fresh and saltwater sources. These sources can consist of small or large shallow puddles, streams, ponds, etc. Moving water is preferable as it keeps the water and substrate cleaner. Good mechanical skimming is also essential to prevent the accumulation of surface materials that might negatively affect a bird's plumage. The water should not move so quickly or suddenly that a bird might be carried away. Pools should be of a depth appropriate to the height of the bird(s) exhibited. Pools with gradually sloping edges will make the pool usable for more species and will allow the birds easier access—different species have different depth preferences. If pools have artificial bottoms, the pool floor should not be rough or too flat as foot problems may develop. Saltwater and freshwater temperatures in the wild have wide ranges. In zoos and aquariums, saltwater temperatures generally range from 4.5–10 °C (40–50 °F).

Enclosure substrates: The following list exemplifies what substrate materials are best suited to a particular shorebird species managed in zoos and aquariums.

<u>Avocets and stilts</u>: These birds have been kept successfully in enclosures that employ a variety of substrates such as sand, natural ground with grass and dirt, small gravel, and some areas of concrete. Foot problems have been noted if they spend too much time on concrete.

Dikkops: Since dikkops are a terrestrial species, a variety of furnishings and substrates are best. This species does prefer a raised, dry area to nest. They will abandon a nest if it becomes too moist from landscape irrigation. A pair of dikkops at an AZA-accredited institution never nested in a tropical rainforest exhibit until a raised, dry area of mixed sand and dirt was created for them (Jones, 1999).

Gulls and terns: It is best to give gulls and terns a variety of substrates such as sand, dirt, grass gravel, and mulch. Most birds in this group will not use a lot of nest material but will use pebbles and grass.

Lapwings: The lapwings prefer a sandy, pebbly area to nest. In the wild, lapwings readily commute between grassland areas, farmlands, and shore habitats. A variety of substrates such as soil, sand, gravel, and pebbles would be beneficial.

Oystercatchers: A variety of sand, gravel, pebble, and rock substrates are recommended for oystercatchers. This will help prevent bumblefoot and will closely mimic the substrates they encounter in the wild. If breeding is encouraged, a pebbled or gravel area near the water needs to be provided.

Phalaropes: Soft substrate, such as sand and/or dirt should be provided for the birds. They will typically throw bits of vegetation towards nest, such as grass, leaves, etc. In the wild, they typically make a scrape in the substrate near water and vegetation.

Sandpipers and plovers: Both groups can be exhibited on a variety of substrates such as sand, small gravel, cobble, mixture of sand/gravel and/or cobble. This of course depends on the species (e.g., whether the species is a sand or rocky specialist, etc.). Although mud substrates such as mudflats are utilized by many species of shorebirds, mud is not advised for zoo habitats due to the difficulty in cleaning and maintaining them, as well as the potential for bacterial growth (K. Anderson, personal communication 2005). For the stitching method of acquiring food, soft areas consisting of sand, small gravel, etc. allow for excellent probing. Some of these areas should also be under shallow water approximating a beach line.

Beach lines with shallow depths gradually becoming deeper allows for species of different heights and leg lengths to use the same shoreline. The ideal height of water for probing and wading for many species is 2.5–5 cm (.5–2 in.). Wading requires pools of the appropriate depth and gradual slope so a bird can gradually wade up to its chest. Shorelines that fall away quickly make it more difficult for birds to probe and reduce the area available for probing. Appropriate substrates such as sand, small gravel/cobble, or rocks/boulders for rocky specialists promotes walking. Regardless of which substrate is used and whether natural or artificial, substrates should not be so rough as to possibly abrade shorebird feet. In regards to nesting, moss, plant materials, small stones should be available for lining nest.

Enclosure cleaning: Typically, most birds will become very aggressive when nesting. Care should be taken to provide nesting birds a wider distance from keepers than usual during cleaning. Many times, areas close to the nest will be left dirty as to not stress the birds.

Avocets and stilts: When a keeper comes too close to the nest site, these birds make alarm calls and may attempt to attack the keeper. Rather than disturb the birds, it is recommended to put off cleaning in the immediate area until the chicks have fledged. When the birds are not nesting, their behavior should return to normal and regular cleaning can be resumed.

Dikkop: Dikkop behavior changes radically when they are nesting. When a keeper comes too close to the nest site, the birds make loud, growling alarm calls and spread their wings out to present their bold black and white pattern. If the keeper does not retreat, the birds will peck. Rather than disturb the birds, it is recommended to put off cleaning in the immediate area until the chicks have fledged.

Gulls and terns: During nesting season, gulls and terns will mob keepers that get too close to the nest. If possible, reduce the amount of cleaning until the chicks have fledged, then regular daily cleaning routines can be resumed.

Oystercatchers: Oystercatcher habitat should be cleaned daily. Roosting areas should be hosed/rinsed daily and disinfected weekly. Feeding areas should be hosed/rinsed daily and disinfected every 3 days. The food that they are fed (mostly bivalves) can decompose very quickly and can result in bacterial growth as well as attract flies if the area is not kept exceptionally clean. Pebbled areas should be thoroughly cleaned on a monthly basis. Sandy areas need to be raked and sifted daily, and feces and old food removed. Rocks in the habitat should be scrubbed daily with either a disinfectant or a commercially available product that removes bird feces such as Poop Off®. If nesting is in progress, cleaning the nesting area should be avoided so as not to disturb incubating birds.

Phalaropes: Land or any hard surfaces in the enclosure should be kept clean of feces by sifting the sand or sweeping up the soiled dirt. The water should have good mechanical skimming on the surface to keep it free of oils and other debris. The birds will need an area to get away from the keeper, so the holding should be large enough to provide plenty of space between the birds and the keeper while cleaning. They have not nested in zoos and/or aquariums so more research is needed about cleaning around nesting birds.

Sandpipers and plovers: Exhibit substrates, furniture, and water features should be cleaned daily as standard operating procedures to reduce/eliminate the potential for disease, foot problems, rodent attractants, or other factors. Substrates should be sprayed and/or raked to break up and eliminate fecal material and to "fluff up" sand/dirt so it does not become too compacted. Dried fecal material can also be swept up with a hand broom and pan. Elimination of fecal material prevents disease and along with the "fluffing" of the substrate, promotes healthy feet by reducing the chance of foot infected and by providing a softer substrate.

Exhibit furniture should be sprayed and hand scrubbed to eliminate fecal material for the same reasons as described above. Furniture should be checked for mold or fungi, which should be eliminated if found either by manually removing via hand scrubbing/scraping or killing it with salt water or a disinfectant. Elimination of molds/fungi aids in the prevention of respiratory diseases.

Water features with hard bottoms should be scrubbed daily to eliminate algal growth, which could foul the water surface, which in turn could negatively affect the birds' plumage. Pools should also be flushed out daily to eliminate all surface contaminants. Factors influencing the above standard operating procedures include individual bird stress levels, reproductive behaviors, or the introduction of new birds. Keepers in close proximity in the exhibit can easily result in stress for sandpipers and plovers (sandpipers more so than plovers). Care should be taken to always leave them a path so they can get away from the keeper and not be inadvertently caught in a corner with no escape. They can also be intimidated by the large size of keepers, so staff should take care not to directly stare at the birds while performing their duties. Despite these precautions, some individuals may be easily stressed or on some particular day, are stressed by normal husbandry maintenance for unknown reasons. If bird(s) are too stressed, it is best for the keeper to discontinue with the exhibit cleaning and do it the next day.

Sandpipers and plovers during a breeding season may need extra space and undisturbed periods for courtship, mating, nest building, incubation and chick care. All these can cause changes in the daily exhibit cleaning.

The same careful consideration regarding exhibit size and complexity and its relationship to the Charadriiformes overall well-being must be given to the design and size 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 Standard10.3.4).

AZA Accreditation Standard

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

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).

If the public has direct access with the birds, it is recommended to station a person in the exhibit to observe and ensure the public interacts appropriately with the birds, and be trained to assist the birds should they get in the public space and

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 selected, monitored, and treated humanely at all times.

are unable to get out on their own. Secondary containment or revolving doors should be used so birds do not have direct access outside the area; this will help prevent animal escapes. Shorebirds are not a threat to the public in any way, but if they are nesting, the public should remain at least 4.5 m (15 ft) from the nesting area. Closer interaction with the incubating birds may cause them to desert the eggs or spend less time than required on the eggs for successful hatch.

Shorebirds have been kept in a variety of aviaries, from walkthrough enclosures with no barriers, to physical barriers such as handrails, piano wire, glass, etc. ½" x ½" wire mesh is suitable as well as zoomesh and piano wire. Glass will also contain most shorebirds, but if the bird is not accustomed to glass and is being introduced to a glass exhibit, the glass should be soaped and gradually removed so the bird will not fly into the glass and injure itself. If flighted shorebirds are housed in public walkthrough aviaries, allowance should be made for the birds to be able to fly back to the habitat area. The birds should be discouraged to go to the public access areas by making those areas not suitable shorebird habitat.

Trimming flight feathers is an option for keeping birds from flying. Flight retraining techniques for birds would be wing clipping or pinioning. Some individuals in zoos and aquariums have been determined non-releasable rehabilitation patients and as such cannot fly and would be good candidates for open habitats. To determine how to select species of this taxon, please see Chapter 4 for more information.

Animal exhibits and holding areas in all AZA-accredited institutions must be secured to prevent unintentional animal egress (AZA Accreditation Standard 11.3.1). Pest control methods must be administered so there is no threat to the animals, staff, and public (AZA Accreditation Standard 2.8.1). Exhibit design must 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.

Although shorebirds can be safely held in secured outdoor enclosures, care should still be taken to prevent potential predators such as birds of prey, rodents, raccoons, or river otters from entering the exhibit by gaining access through netting. Institutions should have a facility wide pest control program to reduce the possibility of non-collection animals from gaining access to the avian exhibits. Since shorebirds held in an open topped habitat would not be flighted, the risk of avian and other predators would be high. Measures to keep predators out would need to be taken. Fencing with electrification keeps mammalian predators out and overhead wires would keep avian predators at bay. If the birds have all day access to the exhibit anti-barriers should be installed to prevent predators from digging into the exhibit.

Exhibits in which the visiting public is not intended to have contact with animals must have a guardrail/barrier that separates the two (AZA Accreditation Standard 11.3.6).

All emergency safety procedures must be clearly written, provided to appropriate staff and volunteers, and readily available for reference in the event of an actual emergency (AZA Accreditation Standard 11.2.4).

There should be enough crates and nets on site to be able to quickly transport all your birds in case of emergency evacuations. There should be a written evacuation plan that includes alternate locations to hold the animal should your facility have to be evacuated.

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).

AZA Accreditation Standard

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

AZA Accreditation Standard

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

AZA Accreditation Standard

(11.3.6) In areas where the public is not intended to have contact with animals, some means of deterring public contact with animals (e.g., guardrails/barriers) must be in place.

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 staff of the institution, or a provided and/or contracted service, must be trained to handle all emergencies in full accordance with the policies and procedures of the institution. In some cases, it is recognized that Security personnel may be in charge of the respective emergency (i.e. shooting teams).

Emergency drills should 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 should be recorded and evaluated to ensure that procedures are being followed, that staff training is effective and that what is learned is used to correct and/or improve the emergency procedures. Records of these drills should be maintained and improvements in the procedures duly noted whenever such are identified (AZA 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).

Each institution should have its own emergency response procedure that would work best for its aviary. Shorebird habitats should be designed in such a way that the birds can always be caught up within a 10-minute response period. All areas of the habitat should be accessible to keepers so that catching can be accomplished quickly and safely and animals removed from the pending danger. Enough crates/transport containers should be kept on site that all the birds could be removed in an emergency. Generally, shorebirds are small enough for one person to restrain, per bird. Care should be taken to avoid getting bitten or scratched by the bird as well as to avoid hurting the bird. Their legs are susceptible to bone breakages and depending on the time of year and how far into a molt they are, blood feathers are subject to breaking during handling

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 which care for potentially dangerous animals must have appropriate safety procedures in place to prevent attacks and injuries by these animals (AZA Accreditation Standard 11.5.3; AZA Accreditation Standard 11.5.2).

Secondary containment doors should be used to help prevent bird escapes. All avian keepers that care for the birds should be trained on safely catching, restraining, and transporting them.

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

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

AZA Accreditation Standard

(11.2.5) Live-action emergency drills must be conducted at least once annually for each of the four basic types of emergency (fire; weather/environment appropriate to the region; injury to staff or a visitor; animal escape). Four separate drills are required. These drills must be recorded and evaluated to determine 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 documented whenever such are identified.

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

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

AZA Accreditation Standard

(11.5.3) Institutions maintaining potentially dangerous animals (e.g. large carnivores, large reptiles, medium to large primates, large hoofstock, killer whales, sharks, venomous animals, and others, etc.) must have appropriate safety procedures in place to prevent attacks and injuries by these animals. Appropriate response procedures must also be in place to deal with an attack resulting in an injury. These procedures must be practiced routinely per the emergency drill requirements contained in these standards. Whenever injuries result from these incidents, a written account outlining the cause of the incident, how the injury was handled, and a description of any resulting changes to either the safety procedures or the physical facility must be prepared and maintained for five years from the date of the incident.

known by all staff members working in areas containing venomous animals (AZA Accreditation Standard 11.5.1).

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. Security personnel should be trained to call the appropriate staff members via an established phone tree, should an emergency arise for which catching and moving the bird is required. It is not necessary for security personnel to be trained to handle the birds, as they will have other duties to accomplish during emergency procedures. However, shorebirds do not pose a threat in terms of human safety and do not need emergency attack drills.

AZA Accreditation Standard

(11.5.2) All areas housing venomous animals, or animals which pose a serious threat of catastrophic injury and/or death (e.g., large carnivores, large reptiles, medium to large primates, large hoofstock, killer whales, sharks, venomous animals, and others, etc.) must be equipped with appropriate alarm systems, and/or have protocols and procedures in place which will notify 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 must be conducted to insure that appropriate staff members are notified.

AZA Accreditation Standard

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

Chapter 3. Transport

3.1 Preparations

Animal transportation must be conducted in a manner that adheres to all laws, is safe, and minimizes risk to the animal(s), employees, and general public (AZA Accreditation Standard 1.5.11). All temporary, seasonal, and traveling live animal exhibits must meet the same accreditation standards as the institution's permanent resident animals (AZA Accreditation Standard 1.5.10). Safe animal transport requires the use of appropriate conveyance and equipment that is in good working order.

> **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 local, state, and federal laws must be adhered to. Planning and coordination for animal transport requires good communication among all involved parties, plans for a variety of emergencies and contingencies that may arise, and timely execution of the transport. At no time should the animal(s) or people be subjected to unnecessary risk or danger.

> **AZA Accreditation Standard**
>
> **(1.5.10)** Temporary, seasonal and traveling live animal exhibits (regardless of ownership or contractual arrangements) must meet the same accreditation standards as the institution's permanent resident animals.

IATA regulations require that the transport container allow a shorebird being transported to stand fully erect without touching the roof and sides of the container. IATA regulations can be found at www.iata.org. 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.]) plastic Vari Kennels® have been used successfully to transport most shorebirds. As with the kori bustard, close confinement seems to calm most shorebirds and reduce struggling (Hallager et al., 2009).

There are different methods that can be used to transport shorebirds that are dependent on the individual species, the animals' health, and their overall temperament. In general, soft, absorbent, and non-slippery materials should to be used. Examples of suitable options are nomad matting, or a trampoline made of suspended soft mesh, constructed to cover the base of the kennel. The mesh should be of such 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. It is recommended that the pad is changed every 8 hours, or as flight restraints allow. Some species, such as the dikkops, can use soft hay or wood chips.

The inside roof of the kennel should be padded with foam covered in fabric with no loose ends or threads. The best way to attach the covering is via zip ties to the roof since duct tape could become loose and stick to the bird. The padding serves to protect the birds from hurting themselves if they attempt to fly in the kennel. The standard ventilation openings on the kennel should be covered in a dark shade cloth that has mesh with holes approximately 1–2 mm 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 closed and secured 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, should it pop out of the holes. Small food and water bowls should be fastened so they don't tip over to the inside of the kennel to comply with IATA regulations. The water bowl should have a hard sponge in it to reduce water spillage. Food should be chilled on ice and should be changed every 12 hours or as flight restraints allow.

On short trips (less than 12 hours) inspection of the kennel and bird is not necessary, nor does the bird need to be accompanied by a keeper. On extended trips (such as multiple flights or an overseas transport) the bird may need to be accompanied by a keeper (on the same flights) so that absorbent pads and food can be changed and the condition of the bird evaluated at opportune times between flights. These inspections would need to be done in an enclosed room at an airport cargo warehouse and by a qualified keeper. For flighted birds, necessary precautions should be taken to prevent escape during kennel inspection. On extended trips, hydration of the bird should be checked and if it is determined that the bird needs hydration, it should be hydrated with a hydrating fluid such as Pedialyte®. For instance, during transport, jacanas may "play dead;" customs and other authorities should be warned about this behavior. If this happens, they should be left alone until they are up and moving around.

The equipment must provide for the adequate containment, life support, comfort, temperature control, food/water, and safety of the animal(s).

Table 4. Necessary equipment needed for a shorebird transport (short or long 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	Towel for restraint, if needed
"This way up" and "Live bird" labels	Medical supplies, such as quick stop
	Spare absorbent pads, trash bags

*This equipment is in addition to the items listed under "short trips"

Temperature range tolerance by most shorebirds is very good, but in the event that a temperature spike is expected in any area the bird is traveling, ice packs may need to be included in the kennel under the floor substrate. Care should be taken to ensure that the shorebird's feet do not come into contact with the ice pack so as to avoid cold damage. Multiple, large "temperature recommendation" labels should be clearly taped onto the kennels so that airport personnel have guidelines within which to work. The recommendations should allow for some flexibility: 4.5–24 °C (40–75 °F). Multiple large labels with the clearly written words "this way up" and "live bird" 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).

Usually no staff person is required to accompany transport if the trip is less than 12 hours. At most, one person would be needed. A trained staff person accompanying the bird on the trip needs to be be familiar with that bird's behavior, signs of stress, and handling protocol. In order to reduce risk to shorebirds and staff handling birds during transport, the following recommendations are useful:

- Conveyance and equipment that is in good working order
- Obtaining the most direct flight
- Performing transports when temperatures are the best for species (seasonal and and/or daily) in the area where they are coming from as well as where they are going
- Avoiding transports during a 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

3.2 Protocols

Transport protocols should be well defined and clear to all animal care staff. It is best to have the birds well fed and watered before transport.

To catch a bird for transport, use a catch cage or net. 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 for some species, such as dikkops, wood shavings or soft hay can be used. Optimal temperatures for shipping most shorebirds are between 7 °C to 24 °C (45 °F to 75 °F). 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 bird. This covering should not restrict airflow. It is good to ship early in the day to avoid high temperatures and give the bird enough time to reach the destination.

Group size during transport is typically one bird per crate, but this can be dependent on species/individual bird. Again, if a transport lasts longer than 12 hours, a trained staff person should accompany the bird to monitor and add food/water as needed. If a staff person is accompanying a bird and can safely access the bird in a secure area, as mentioned above, to provide food/water, the bird may be kept in the kennel for up to 24-48 hours, but this can vary on the species/individual temperament of a bird.

Once the kennel has reached its final destination, but before releasing the bird from the crate, it is important to make sure that the bird has not been injured in shipment. After that has been determined, the weight of the crate (still with the bird inside) should be taken and recorded. Once the bird has been released, the crate should be weighed again to get a baseline weight on the bird without having to restrain it. For release, only trained staff should open the crate door. It is less stressful if the bird can be allowed to exit the crate on its own timetable. Each institution will have its own protocol for post-transport release.

Chapter 4. Social Environment

4.1 Group Structure and Size

Careful consideration should be given to ensure that animal group structures and sizes meet the social, physical, and psychological well-being of those animals and facilitate species-appropriate behaviors. Shorebirds have successfully been kept in multispecies exhibits. When deciding what birds can be mixed, the following factors should be considered: sex ratios, time of the year, the age of the birds, a bird's origin, and a bird's size. Several species such as dikkops, gulls, and terns can be more aggressive than others. For the more aggressive species, either having an even sex ratio—all female or an all-male group—may be needed. Certain species or individuals may need to be placed in an off exhibit area during a breeding season. Therefore, adequate off-exhibit space needs to be available.

In regards to a bird's age, once individuals reach sexual maturity (generally around 1 year of age), the dynamic of the entire group within an exhibit could change. Also, older birds that cannot get around as well may be more susceptible to aggression from other birds due to their reduced ability to move. In reference to a bird's origin, zoo-bred and parent-reared birds tend to be calmer and more acclimated to exhibit mates (if reared on exhibit). Rehab birds, whether flighted or not, are often more sensitive due to recently living in the wild, particularly during their introductory period and may require closer observations to ascertain they are compatible with their exhibit mates. If they are not compatible, then they may need to be moved to a different exhibit or area. Certain birds that are non-flighted may not be a good mix with flighted birds. Non-flighted birds may be more susceptible to aggression and less likely to be able to get away from flighted birds. Lastly, shorebirds typically like to flock together and if an institution has a single species in a particular exhibit, other birds of similar size and habitat should also be included. The following is a list of recommended group structure and sex structures for Charadriiformes that should try and be replicated in AZA-accredited institutions.

Avocets and stilts: The general rule of thumb to follow in regards to housing avocets and stilts is that they can be kept in a mixed species exhibit with only one additional species of stilt/avocet, because they do better if they have other similar sized birds around them. Having either an even sex ration or two birds of the same sex works best. They can be kept in single sexed groups; however, problems with aggression have been seen with more frequently between males than females.

Dikkops: Since these birds do not perch, they are ideal to mix with arboreal, perching birds. It is important that the aviary is covered to prevent escape; if it is not covered, dikkops will need to be feather-clipped. In general, dikkops can be kept in pairs or larger groups, and in single-sexed or all male groups. They are gregarious when not in breeding season. The following examples demonstrate that when not in breeding season, these birds tend to form flocks, but then pair off during the breeding season. The spotted dikkop occurs in flocks of up to 50 birds (MacLean, 1993). The bush thick-knee has been seen to form flocks of up to 40 birds (Blakers et al., 1984). Additionally, in Israel, the stone-curlew has been observed in flocks of more than 100 birds (Paz, 1987). In one AZA-accredited institution, all unpaired and young stone-curlews were kept together in a group aviary. Members of the *Burhinus* genus are aggressive to each other at times, and one AZA institution separated their double-striped thick-knees from their peruvian thick-knees (Jones, 1999).

Group structure can change in the breeding season and keepers should be wary of aggression during a breeding season if dikkops are not kept in pairs. For example, the spotted dikkop tends to be solitary when nesting, but during the non-breeding season will form loose groups (del Hoyo, 1996). Then it may be advisable to give the breeding pair their own area either in the exhibit or in a holding area. In zoos and aquariums, nesting dikkops have been observed being aggressive towards other ground dwelling birds such as crowned pigeons and glossy ibis (Pate, 1983). This aggression seems to be limited to chasing and pecking by the parent thick-knee. In one institution, a pair of purple glossy-starlings was observed pecking open the eggs of a pair of spotted dikkops (Jones, 1999).

Gulls and terns: Gulls and terns are best kept in a flock setting and have been successfully kept with other bird species. One zoo raised 72 laughing gulls over an approximately 40-year time span in an exhibit with cattle egrets, green herons, snowy egrets, and roseate spoonbills. The other birds exhibited with the gulls raised young (Zoological Society of London, 1979). In another instance, 20 silver gulls were hatched and raised in an exhibit with 30 species of mainly aquatic birds (Bohmke & Fisher, 1992). The only interspecific aggression by the gulls was in defense of the immediate nest area. Lastly, a third AZA-accredited institution houses 10 grey gulls, 25 Inca terns and 30 Humboldt penguins, illustrating that many gull and tern species can be housed in one exhibit with a high degree of cooperation (Lindholm, 2007) All three species reproduce successfully. An estimate of the optimum flock size would be between 10–25 individuals, depending on the amount of space in the exhibit. They can be kept in single-sexed groups.

Jacanas: In a zoo setting, it is optimal to have 1.1 jacana. At one AZA-accredited institution it was found that a jacana pair could be kept with other species and were easily displaced by other species. However, during breeding season they would become very aggressive towards any other jacana. This aggression has been seen in enclosures as large as 60 m x 50 m x 15 m (196.8 ft x 164 ft x 49.2 ft) (Holland, 2007).

Lapwings: Lapwings are happiest in an exhibit if a sex balance is maintained, and they are best kept in breeding pairs. It is unknown how lapwings react in single-sexed groups. They are generally not aggressive to unrelated species. Crowned and blacksmith lapwings have been mixed with no problems in a large aviary. However, keep in mind that most aviaries may not be large enough to mix lapwing species (Holland, 2007). Most aviaries are not large enough to keep lapwing species from fighting over territories.

Oystercatchers: While it can be challenging to get oystercatchers to bond, as it seems to depend largely on the temperament of the individual birds, oystercatchers are best housed in bonded pairs. Since oystercatchers are long-lived in managed settings (known to live up to 35), it would be ideal for them to be matched quite closely in age. Other combinations such as housing two females together, trios, or single birds have also been successful. However, there are very few oystercatchers in in zoos and aquariums, so keeping them singularly should be considered acceptable. There are oystercatchers in the wild that do not form bonds nor congregate in large flocks. In institutions with oystercatchers, there are a number of single-sexed female flocks of up to two birds. This generally does not present a problem, but in some instances, aggression can occur to the extent that the two females cannot be housed together.

There is no record of single-sexed male groups. Generally, in zoos and aquariums, breeding pairs will be very aggressive towards newly fledged juveniles and the juveniles have to be removed from the breeding habitat for safety reasons, so multigenerational flocks are not found. The tendency for breeding pairs to be aggressive towards other oystercatchers is strong in managed settings, and so for this reason oystercatchers are kept in low numbers.

Although oystercatchers are unlikely to be considered good candidates for conservation and education programs, if they are to be used as program animals, they can be housed successfully as individuals. Their holding pen should be set up so as to ensure ample enrichment as well as to allow for natural foraging mechanisms.

Phalaropes: From an AZA-accredited institution's observation, phalaropes do not tend to be too aggressive with each other. Because of their low tendency towards aggression, the sex ratio can vary. It is recommended to have a minimum of about .09 m^2 (1 ft^2) of space per bird, both on land and in the water, with plenty of space to be able to get away from keeper staff if necessary. If they have other species of birds in their exhibit, the minimum number of phalaropes that should be kept is three. If they are the only birds in the exhibit, there should be at least six phalaropes. These can be a mix any of the phalarope species. They can be kept in single sexed groups.

Sandpipers and plovers: In zoos and aquariums, most sandpiper species do well mixed together. Smaller species such as the dunlin do better if they are held with species of the same or similar size. While larger species such as long-billed curlews do well in mixed flocks, they should not be exhibited with more than one of their kind unless they are a mated pair. Marbled godwits have been successfully exhibited with three individuals in one exhibit. Sandpipers can be kept in single sex groups of either sex.

Plovers do well in mixed flocks, but they should not be exhibited in large numbers of same species exhibits, as they tend to be more aggressive towards other plovers. As a rule of thumb, no more than two individuals of the same species should be exhibited together. It is recommended that sex ratios for plovers be even or consisting of mostly females, but no all-male groups. Some aggression may occur between male plovers, but only with some species and within a certain range of aggression, this is normal. The aggression should not escalate to the point that a bird is bleeding or not allowed to eat. It has been observed that male piping plovers do not always obtain good breeding colors without some aggression between exhibit mates (K. Smith, personal communication, 2006). Smaller species of both groups do better if they are exhibited with similar sized individuals.

4.2 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.

Avocets and stilts: These birds can be kept with a variety of birds, including other shorebirds. These birds include waterfowl, egrets, spoonbills, ibis, small herons, gulls, pelicans, parrots, pheasants, doves, and songbirds (Vince, 1996). They have been known to be aggressive with other birds during breeding season, especially smaller birds.

Dikkops: Dikkops have been successfully housed with coucals, owls, and hornbills (Holland, 2007). At one AZA institution, they were housed with blue-bellied rollers and bearded barbets with success (C. Pinger, personal communication, 2011). Disputes with conspecifics seem to be limited to the breeding season and with particular larger species like crowned pigeons and glossy ibis (Pate, 1983). The typical negative interaction will involve chasing, loud vocalizations, wing flashing, and pecking.

Gulls and terns: Many different gull species have been kept with multiple other species. Specifically, laughing gulls have been kept with cattle egrets, green herons, snowy egrets, and roseate spoonbills. Also, grey gulls have been kept with Inca terns and Humboldt penguins. Grey gulls have been housed with an extensive array of other species such as with Inca, common, and sandwich terns, European avocets, ruffs, redshanks, black-tailed godwits, black-necked stilts, black scoters, spectacled and common eiders, barrow's goldeneyes, and gentoo and South African penguins.

Jacanas: Jacanas can be mixed with several other species of reptiles/amphibians, birds, (e.g., crakes, kingfisher, starlings, hammerkop, and ibis), small mammals, and tropical fish. Care should be taken to make sure that there are no large predatory fish present (e.g., tilapia), as they may prey on jacana chicks. Crakes and rails are known to destroy nests and should not be present when jacanas are breeding.

Oystercatchers: When housing oystercatchers with other species, it is important to make sure that other species do not monopolize the food supply in a mixed species habitat. If food is offered for the oystercatchers in a way that stimulates natural feeding behaviors, there is less chance of the other birds finding the oystercatchers food. Oystercatchers have been housed successfully with the following species:

- American widgeon
- Arctic terns
- Avocets
- Black skimmer
- Black-necked stilts
- Brown pelican
- Common goldeneye
- Common murre
- Common terns
- Dunlins
- Gadwall
- Godwit spp.
- Harlequin duck
- Horned puffin
- King eider
- Long-tailed duck
- Marbled teal
- Northern pintail
- Northern shoveller
- Pigeon guillemot
- Plover spp.
- Red-legged kittiwake
- Rhinoceros auklet
- Roseate spoonbill
- Sacred ibis
- Sandpiper spp.
- Scarlet ibis
- Tufted puffin
- White ibis

Several instances of aggression have been recorded in relation to breeding events. In one instance, between one female oystercatcher and a breeding pair of tufted puffins, the oystercatcher approached the puffin nest and one of the puffins defended the nest by biting the oystercatcher's head; the oystercatcher lost her eye as a result of the attack. In a second instance, where oystercatchers are housed with avocets, the avocets are typically moved to another habitat during their breeding season as aggression from the avocets towards oystercatchers can be severe. There is no record of the other species listed of negative interactions with the oystercatchers. Another example of aggression in relation to breeding territory involved a male oystercatcher. When exhibited with the main shorebird collection, he built a scrape and defended a wide territory around it (about 2.4–3 m [8–10 ft]), which resulted in the other shorebirds having only about 25–30% of the exhibit to utilize.

Phalaropes: These birds have been kept successfully in enclosures with other shorebird species. Examples of these shorebird species are sanderlings, dunlins, plovers, avocets, stilts, dowitchers, godwits, dunlins and smaller songbirds, such as sparrows and pipits. Phalaropes cannot be housed successfully with ducks because as one AZA-accredited institution reported, when phalaropes have direct access to ducks, the ducks were often times drowned. When housing phalaropes with other species, it is important to make sure there is enough water space for other birds to wade and all phalaropes swimming at the same time.

Sandpipers and plovers: Plovers and sandpipers have been successfully exhibited in multispecies displays with a multitude of different species. Examples include but are not limited to: black crown night heron, snowy egret, flickers, orange weavers, gold-fronted leafbird, yellow-bellied sapsucker, red-winged blackbirds, yellow-headed blackbirds, Bonaparte's gulls, phalaropes, avocets, and stilts. In some managed situations, they have been displayed successfully with waterfowl (baikal teal, greenwinged teal, European shellduck, etc.), but in other instances, aggression has occurred between the shorebirds and waterfowl to the point where the waterfowl had to be removed. Even smaller waterfowl species such as ruddy ducks or hooded mergansers have exhibited aggressive and/or territorial behavior such as chasing, staking out territory, pulling feathers, etc. Most of these interactions occurred during breeding season. Smaller species of plovers and sandpipers do better when exhibited with species of like size, whether their own species or others. However, for the most part, interactions with other species are neutral.

Trained keepers should always keep an eye on all interactions, both positive and negative, in all shorebird collections. Many smaller species of shorebirds, especially non-flighted ones, have had negative interactions with ducks, egrets and herons, leading to death. Other positive and negative interactions between conspecifics include: chasing, pecking, neck and head feathers missing, and displacement at food area.

Oystercatchers: When hand-rearing oystercatchers for use as program animals, the risk of imprinting is high. It would be preferable to allow the chicks to be parent-reared up to 3 weeks of age before completing the rearing by hand. If an oystercatcher is housed alone for use as a program animal, there is a chance that the oystercatcher will bond with a human keeper as it becomes mature. This can lead to aggressive behavior towards other keepers and possibly the public. Having more than one keeper working with the bird can prevent bonds like this, but it is not always possible to prevent.

4.3 Introductions and Reintroductions

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

Introductions and reintroductions should be made outside of the breeding season, if at all possible. Birds should be released away from glass barriers and water sources. Also consider releasing birds during days with lower visitor attendance, avoiding weekend crowds. All birds should be monitored closely after introductions to watch for aggression. Both introductions and reintroductions should be done at a time of day where staff will be around to observe for several hours. Additional visual barriers may be needed, such as plants, driftwood, tarps etc. The exhibit should be designed in a way that no bird can become trapped in a tight spot in the exhibit and can't escape from another bird. If more aggressive birds are on exhibit, consideration should be given to remove them while the introduction is happening, giving the new bird a chance to settle in before reintroducing the more aggressive birds.

Depending on species and individual birds' temperament, "howdy" cages can be used or birds can just be introduced straight onto exhibit. If no plans to clip a flighted bird are in place, any glass barriers should be soaped, taped, etc. on the public side to allow the bird to learn where barrier is located Soap will gradually wear off over the course of a few weeks but tape needs to be removed on a regular schedule.

Depending on the birds' temperament, an institution may need to set up stanchions to keep visitors away from the glass, with signs explaining the need for them to stand back and keep their voices low. Stanchions can be gradually moved towards the glass railing as the new bird becomes acclimated to the exhibit and its visitors. Movement and later removal of stanchions depend on how well bird is acclimating to the exhibit.

Avocets and stilts: The best time for introductions are during the non-breeding season, as aggression levels are lower during this time. With the black-winged stilt, an introduction strategy called "flock mating" has been described by Holland (2007). The best time for this is in late winter. If you are introducing multiple birds, all birds in the flock need to be reliably sexed and banded. Two pairs should be housed together for 2–3 hours on neutral territory. The aviary should be at least 400 m^2 (4,305.5 ft^2). By closely watching the birds, the dominant pair will be obvious within a few hours. This dominant pair should be moved to their own breeding aviary. This method could also work for avocets. If you are introducing a single bird into the exhibit, you can either try the "howdy" cage method, or introduce the bird straight on exhibit and monitor for signs of aggression. If the new bird continues to be harassed, consider removing the harasser for a few days until the new bird gets settled in. As pairs tend to be very territorial, introducing an extra male or female to an existing pair can be difficult.

Dikkops: The best time for introducing new birds is during the non-breeding season. During the non-breeding season aggression levels are low and the birds are inclined to form flocks and accept new members. It is always the best practice to allow new birds and established birds to be slowly accustomed to each other by the use of a "howdy" cage. This allows the new bird to interact with the other birds and become familiar with the layout of the exhibit. On some occasions, a "howdy" situation may not be possible. If this is the case, then keepers should closely monitor the introduction and intervene if the aggression level escalates to unacceptable levels.

Gulls and terns: There is no information available on the introduction of gulls and terns specifically. It would be best to follow the general guidelines given in this manual.

Jacanas: The complexity of territoriality in jacanas complicates introductions. Males may compete with other males for suitable breeding territories; females may compete with other females for ownership of one or more males and their territories; and females may also compete for large territories that can be subdivided by males. For this reason, the introduction of new birds to existing situations where established pairs have already formed is virtually impossible in small zoo-based enclosures. Even introducing an extra male or female to an existing pair or trio is very difficult.

Lapwings: It is best to introduce lapwings to an exhibit all at the same time and if possible, during the non-breeding season. Expect behavior changes in the groups during the breeding season. Keep a close eye out for aggression such as pecking and chasing.

Oystercatchers: It is not recommended to introduce young birds into exhibits that house adults, as adults tend to be very aggressive towards young birds. One successful introduction technique that has been used to bond oystercatchers with a new comer in an existing habitant is to first introduce and house them together in a neutral housing pen that is not the habitat, like a howdy cage or adjoining holding cage. Staging introductions in a space other than the established habitat seems to offset the aggression since both birds are equally new to the staged area and have no perceived territorial rights. Once they have been together in this neutral area for at least 4 weeks, an attempt to move them to the established habitat can be made.

Another possible introduction method was attempted at one institution, where two female oystercatchers were gradually introduced to each other by using a barrier constructed of netting and driftwood. Over a period of several weeks, the barrier was gradually reduced and the two birds were able to see each other more clearly. The birds are then allowed to move closer together by adjusting the barrier. Eventually the barrier was removed completely and the birds were allowed to interact. Both of the methods described above require close observation.

Phalaropes: It is recommended to make introductions outside of the breeding season. Phalaropes can be introduced into the exhibit without use of a howdy, but still need to be monitored closely for signs of aggression. Mild displacement may occur for a few days by other birds but they typically get along well.

Sandpipers and plovers: It is recommended to make introductions outside of the breeding season, and all introductions monitored closely. Prior to introducing the new bird, aggressive individuals need to be removed and held off exhibit until the new bird seems comfortable, usually about a week. While a howdy cage can be used, it usually isn't necessary. Typically they can be put right on exhibit.

Chapter 5. Nutrition

5.1 Nutritional Requirements

A formal nutrition program is recommended to meet the nutritional and behavioral needs of all Charadriiformes (AZA Accreditation Standard 2.6.2). Diets should be developed using the recommendations of nutritionists, the Nutrition Scientific Advisory Group (NAG) feeding guidelines (http://www.nagonline.net/Feeding%20Guidelines/feeding_guidelines.htm), 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.

> **AZA Accreditation Standard**
>
> **(2.6.2)** The institution should have 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.

The diet of shorebirds are dominated by invertebrates, but some species may also eat small vertebrates such as fish, amphibians and reptiles, seeds, tubers and small fruits during part of the year (Cowell, 2010).

The diets of wild shorebirds vary greatly both among and within the species. This variation correlates with associated structures necessary to digest the soft-bodied and hard-bodied prey items. The sections of the digestive system responsible for glandular and mechanical digestion are the proventriculus and the gizzard, which make up the stomach. According to Colwell, the proventriculus represents a much smaller proportion of the stomach mass than the gizzards. Stomach mass increases proportionate to body size. There is even considerable intraspecific variation in stomach mass associated with seasonal changes in the diet. Stomachs can also change size according to time of year related to migration. Some of the smallest stomachs obtained from wild caught knots are just before long-distance migrations. It is thought their stomachs had shrunk as a weight conservation measure for reducing wing-loading and effecting greater flight efficiency (Cowell, 2010).

In the wild, with a few exceptions, shorebird diets are dominated by invertebrates, especially soft-bodied prey. They eat mostly freshwater and marine invertebrates from the classes Insecta, Malacostraca, Gastropoda, Polychaeta, and Bivalvia (Cowell, 2010). Larger shorebirds, such as thick-knees and curlews, will occasionally eat small vertebrates such as fishes, amphibians, reptiles, and even small mammals. Other species, such as some of the sandpipers occasionally include seeds or tubers (Cowell, 2010).

Depending on the species of shorebird, most AZA institutions feed their shorebirds a mixture of the following items:

- Bird-of-prey meat
- Krill
- Soaked dog food
- Small bird maintenance
- Finch seed
- Parakeet seed
- Hard boiled eggs
- Chopped greens
- Live mealworms
- Live waxworms
- Live crickets
- Live tubifex
- Smelt fish
- Silverside fish
- Flamingo fare

Diets should be offered in a way that will encourage natural feeding behaviors. Most institutions scatter food so the birds can forage and have other food items available in shallow dishes. Several dishes should be used and spread out to allow all birds access to the food. The exact number of dishes used will vary according to the number of birds, but one bird shouldn't be able to be able to guard the food and not allows others to eat. Make sure to consult with a veterinarian to plan a diet best suited for all species. A variety of natural diet items should be offered to supplement other diet items offered. The feeding schedule should be varied as well as the types of food offered every day for both enrichment purposes and to keep the digestive system healthy.

Depending on the range of ambient temperatures that the shorebirds are exposed to, they may require an increased amount of food during colder temperatures. Food consumption needs to be monitored daily and amounts adjusted accordingly. Below certain temperatures, shorebird food consumption drops dramatically; this is a sign that they should be caught up and put in warmer indoor housing. That temperature can vary according to species and age of the bird, but the keeper should be aware of this potential occurrence, and watch for associated behavior. Amounts of food offered will also change with molting status; they will eat more during molt. More research is needed to suggest species-specific nutrient target ranges for all life stages and energy requirement calculations for shorebirds.

Avocets and stilts: These birds primarily feed on a variety of invertebrates and small vertebrates that are picked off the surface of water or mud. Avocets frequently feed by skimming the water surface with a distinctive side-to-side swipe of the bill and can sometimes swim to pursue food.

Dikkops: The spotted dikkop forages by running forward, stopping, and jabbing the food item with its bill. While these birds primarily feed on insects (e.g., crickets, grasshoppers, and mealworms), they are quite diverse eaters. Here is a list of the food items that have been recorded in the diet for wild birds:

<u>Invertebrates</u>

- Insects
- Coleoptera (beetles)
- Curculionidae (weevils)
- Carabidae (ground beetles)
- Tenebrionidae (darkling beetles)
- Dermaptera (earwigs)
- Hemiptera (bugs)
- Hymenoptera (wasps, bees, and ants)
- Termites
- Lodotermes mossambicus (Northern harvester termite)
- Lepidoptera (moths and butterflies)
- Mantids
- Crickets (Orthoptera)
- Gastropods
- Spiders
- Sun spiders
- Giant millipedes
- Worms
- Scorpions

<u>Vertebrates</u>

- Small amphibians
- Small mammals
- Leptotyphlops (thread snakes)
- Eggs and chicks of White-fronted plover (*Charadrius marginatus*) (Hockey et al., 2005)

The amount of live insects should be increased by at least twice the regular amount when an institution is dealing with dikkop chicks. The diets for dikkops in zoological institutions vary widely as seen in the diet list below. For instance, Holland (2007) described a diet that consisted of minced heart and bird of prey diet meat mixed with commercial softbill food and moistened cat food. They have also been known to eat mollusks (snails and slugs), crustaceans, lizards, boiled egg, cooked rice, white cheese ground meat, minced fish, and occasionally small mice (Cramp & Simmons, 1983; Cwiertnia, 1999). The birds preferred the mice followed by boiled egg and ground meat. A multi-vitamin supplement should be given twice a week. According to Bates & Busenbark (1970) dikkops need an acclimation period to diets presented to them in managed settings. This acclimation diet should consist of live foods like mealworms and small fish. After an adjustment period that could range from 7–21 days, they should begin eating a more balanced diet. Below are sample diets that other AZA-accredited institutions have used to feed dikkops:

- Diet 1: 38% powdered softbill pellets, 20% chopped hard-boiled egg, 15% soaked dog or cat chow, 15and Bird of Prey diet or lean hamburger, and 12% live food including chopped earthworms All ingredients be mixed together and the meat pieces should be very small, so it easy for the birds to pick them up (Vince, 1996).
- Diet 2: Soaked dog food (30%), Bird of Prey diet (30%), superworms (10%), and gamebird chow (30%) to thick-knees (Padron, 2010).
- Diet 3: Bird of prey diet and pinkie mice with vitamins (VV-13).
- Diet 4: Flamingo chow complete (3/4 c), soaked dog food (1/4 c), Natural Balance 5% fat (30 grams), and live insects mealworms, waxworms, and crickets (20–30 insects).
- Diet 5: Chopped silversides or smelt, fuzzy or pinkie mice, and meat product mixed with insectivore pellets.
- Diet 6: Nebraska Brand Bird of Prey diet in tablespoon size pieces. Mice, crickets, mealworms, and earthworms were also fed occasionally (Jones, 1999).

Gulls and terns: These birds eat a wide variety of foods from insects, crustaceans, fish, and mollusks to eggs, worms, other birds, invertebrates, and even garbage. An AZA-accredited institution feeds their grey gulls and Inca terns a variety of fish including capelin, silversides, lake smelt, and marine smelt. Handlers supplement these fish with Stuart Thiamine E paste. When they are fed fish, it is presented to them two times a day in stainless steel cake pans. If chicks are present, the birds are fed three times a day (T. Snyder, personal communication, 2011).

Jacanas: Recommended diets in zoos and aquariums for jacanas include insects, snails, and small fish. Additional items include a soft-billed mixture that is usually given with small amounts of meat diet, vegetable (e.g., endive, carrot), insects, and fruit (e.g., apple, banana, grapes). For example, a diet for wattled jacanas given at one AZA accredited institution consisted of 36 grams of food per day per bird, of which 44.5% was soft-billed mixture, 11% fish (sprat), 11% mealworms, 11% vegetables, and 23.5% fruit. The nutrient analysis summary for this diet is listed below in Table 5.

Table 5. Nutritional breakdown of an AZA-accredited institution wattled jacana diet

Nutrition Category	Nutrient Type	Quantity
Energy	ME Primate	0.19 kcal/g
Carbohydrates	ADF	0.24%
Fat	Crude fat	1.73%
	Linoleic acid	0.03%
Protein	Crude protein	4.52%
Vitamins	Folacin	1.10 mg/kg
	Niacin	1.35 mg/kg
	Pantothemic acid	0.82 mg/kg
	Riboflavin	0.19 mg/kg
	Thiamin	0.21 mg/kg
	Vitamin A	17.26 IU A/g or RE/g
	Vitamin B12	0.00 mcg/g
	Vitamin B6 Pyridoxine	0.55 mg/kg
	Vitamin C Ascorbic acid	27.58 mg/kg
	Vitamin E	1.50 mg/kg
Ash/Minerals	Ash	0.36%

Calcium	0.01%
Copper	1.04mg/kg
Iron	3.07 mg/kg
Magnesium	0.01%
Manganese	1.07 mg/kg
Phosporus	0.07%
Potassium	0.12%
Selenium	0.01 mg/kg
Sodium	0.01%

In regards to a diet for jacana chicks, which are precocial and self-feeding, it is essential that they are fed adequately. But often, it is difficult to entice them to accept food. Providing a variety of foods, including live prey items, helps ensure that they will recognize and consume appropriate food items when they are older. The following items are recommended for starter diets, and should be provided ad libitum:

- Live pinhead crickets, fed on a high calcium diet for 24–48 hours before fed out
- Live wingless fruit flies, fed on a high calcium diet for 24–48 hours before use
- Live brine shrimp in water/brine
- Mouse pinkies, finely minced, moistened with distilled water
- Tubifex, freeze dried, in water
- Mazzuri pheasant starter pellets, crushed

Food items can be offered in petri dishes and/or scattered in the brooder. Guppy flakes and/or freeze-dried plankton may also be offered in water if the above items are not consumed in sufficient quantity. All dishes containing water should be shallow (<1.3 cm [.5 in.]), as younger chicks may drown. In the forced-air brooder, food dishes should be changed every 1–3 hours. In the larger, precocial brooder, food dishes should be placed away from heat sources and should be changed every 4–6 hours. Freshly molted mealworms can be offered to chicks when they are able to consume food items this size. As chicks develop further, they will be able to consume unmolted mealworms and larger crickets. At around 30 days, the chick diet will begin to transition to the adult diet. Hand-reared chicks may develop a vitamin D and calcium deficiency. More research is required to determine the causal factors for these deficiencies.

Lapwings: Lapwings are a diverse group whose beaks vary greatly in shape and size. Free-ranging lapwings eat invertebrates including earthworms, beetles, flies, and caterpillars. One zoological institution feeds equal portions of soaked cat kibble, minced heart, and insects such as mealworms and bloodworms (Holland, 2007).

Oystercatchers: Free-ranging oystercatchers eat intertidal marine invertebrates, particularly bivalves (e.g., mussels and oysters) and other mollusks (e.g., limpets, whelks, and chitins), crabs, sea urchins, isopods and barnacles. The oystercatcher bill morphology is critical for prey capture, as their beak enables them to pry open live mollusk shells and dig in a beach's sand gravel substrates for up to 9 cm (3.5 in.). They use their bills to sever the adductor mussel of the bivalve and then remove the fleshy body from between the shells. Indigestible parts of their prey are sometimes regurgitated in a cast pellet in an unconsolidated form. Below are sample diets that other AZA-accredited institutions have used to feed oystercatchers:

- Diet 1: 40% mussels, 40% oysters, 8% fish, 8% clams, 4% krill
- Diet 2: Mixed fish including herring, capelin, smelt, silversides, salmon smolt, krill, live mussels growing on rocks, and if they are raising chicks, additional shellfish chopped up
- Diet 3: Insectivorous diet, bird of prey-like diet and insects
- Diet 4: Dog food, flamingo fare, chopped fish, apple paradise, silversides and pinky mice
- Diet 5: Mealworms, wax worms, krill, black worms, crickets
- Diet 6: Mussels, krill, silversides, capelin, plankton, shredded seaweed, crickets, wax worms, mealworms, and gel diet

Phalaropes: Phalaropes feed visually while swimming, picking prey items from the surface of the water and occasionally submerging head to secure prey. The most conspicuous and well-known phalarope feeding behavior is a top-like spinning on surface of water. They spin at the surface to create a vortex, drawing nutrients and prey items to the surface. They can also forage on foot when walking slowly on land, picking prey from the surface of the mud or water.

Sandpipers and plovers: Similar to lapwings, this is a widespread group with much variation in beak size and shape. Some species locate their food visually, while many others find their food through tactile clues by detected by Herbst corpuscles embedded in the tips of the bill (although there can be overlap between the two groups). In general, plovers have shorter, stouter bills and visually forage for prey and capture it by pecking. Pecking can be described as plovers lifting their heads and visually searching for prey items, quickly running towards the item, and pecking into the substrate with their bill.

Sandpipers have longer, more slender bills, some of which are decurved, and are tactile probers that capture prey by probing. Sandpipers often wade into water up to their bellies while they probe for food. A common method of probing is known as “stitching,” where they methodically probe the substrate as they walk along the shore both above and below water line. Sandpipers with long decurved bills often utilize them as forceps as they thrust their bill into wormholes to extract prey or to delve under logs and seaweed. However, ruddy and black turnstones are sandpipers whose bills are stout and conical shaped—more reminiscent of plovers. These birds use their bills to flip over objects such as small stones (hence their name), seaweed, beach debris, etc. in search of invertebrates such as sand fleas.

At least one month before egg-laying, it is recommended to give shorebirds and plovers supplemental calcium. Deficiency of vitamin E is associated with low fertility and low hatchability of eggs (Dierenfeld, 1989). During breeding and chick-rearing season, more food—as much as double the normal amount—should be offered for the condition of the parents and the added intake for the chicks. Diet for both groups consist of a wide variety of marine invertebrates, insects, worms and a few other miscellaneous items such as seeds, berries, eggs, etc. These latter items constitute a minor portion of their diet. Examples of marine invertebrates include but are not limited to: small bivalves such as clams, marine worms, barnacles, beetles, earthworms, fly larvae, etc. Most places feed one or two items from several different categories. Live prey needs to be fed a diet that will encourage high calcium to phosphorus ratio, preferably 2:1. For instance, information regarding blacksmith and shore plovers states that they should receive 20 mealworms per day during most of the year, and 30 mealworms twice a day during breeding season (Holland, 2007). The table below illustrates some sample prey items typically fed to shorebirds by AZA institutions. They are typically fed minimum twice per day and variety of any of the following items:

Table 6. Sample of food items fed in other AZA institutions

Live Prey	**Previously Frozen**	**Grains**	**Fruits/Vegetables**	**Supplementation**	**Other**
Mealworms	Krill	Soaked Marion® Red Apple Jungle pellets	Shredded mixed greens	Nekton	Soaked dog food
Superworms	Chopped smelt	Soaked Mazuri® low iron softbill pellets	Carrot/cabbage mix	Calcium	Finch seed
Waxworms	Pinkies	Small bird maintenance	Fruit mix consisting of diced apple and cantaloupe, cut grapes, chopped orange without the peel, diced cooked sweet potato and chopped banana without the peel		Chopped hardboiled egg with shell
Blackworms	Sandlance	Parrot chow Flamingo chow	Novel fruit such as thawed frozen fruit (blueberries, peaches or strawberries), fresh papaya or other seasonal fruits is added daily		
Crickets	Silversides	Wild gamebird			

		maintenance
Fly larvae	Mussels	
Butterworms	Oysters	
	Clams (razor and surf)	
	Sandeels	

Based on Table 6, these are sample diets for shorebirds that have proved successful from other AZA institutions.

- Diet 1: 38% krill, 38% chopped clam, 24% chopped sandeels. Ingredients are mixed together and offered two times a day. Live foods (e.g., crickets, tubifex, mealworms, waxworms and fly larvae) are given as enrichment items
- Diet 2: Soaked dog chow, chopped smelt, chopped silversides, mealworms, crickets fed 2x day
- Diet 3: All live food (e.g., crickets, mealworms, waxworms, fly larvae, tubifex worms) given twice a day

More information is needed for specific energy requirement calculations of most shorebird species. According to Cowell, shorebirds use more energy, have greater daily energy expenditures and maintain higher metabolic rates. The basal metabolic rate of various shorebird species are rougly 40% higher than that reported for other nonpassarines of comparable size. It is thought it is a consequence of relatively sparse/low insulating properties of their plumages as well as tendency to occupy open habitats, such as Arctic tundra and coastal beaches. Consequently, the higher metabolic rates effect thermoregulation (Cowell, 2010).

5.2 Diets

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

Table 7. Sample diet analysis from N.P. Analytical laboratories (2008)

Sample	**Moisture %**	**Protein %**	**Fat %**	**Ash %**	**Carbohydrate %**	**Calorie kcal/100g**	**Calcium g/100g**	**Phosphorus %**
Crickets	71.80	17.30	5.33	2.59	2.98	129.00	0.0343	0.245
Crickets (enriched)	73.10	17.80	5.13	2.75	1.22	122.00	0.315	0.269
Crickets with vitamins	71.00	17.50		3.63			1.03	0.261
Fly larvae	68.10	11.70	10.70	1.23	8.27	176.00	0.0576	0.229
Fly larvae with vitamins	73.50	11.70	8.33	1.66	4.81	141.00	0.332	0.223
Krill, Pacifica, Lot #777547	79.20	14.50	3.48	2.66	<0.5	89.30		
Krill, Pacifica, (rinsed) Lot #712004	84.90	11.70	1.44	1.90	<0.5	59.80	0.313	0.275
Krill, Superba (fresh) with vitamins	79.90	13.10	3.78	2.91	<0.5	86.40	0.309	0.243

Krill, Superba (Fresh) Lot #737760	87.30	9.31	2.26	1.89	<0.5	57.60	0.326	0.282
Mealworms	58.30	19.90	9.13	1.94	10.70	205.00	<.01	0.227
Mealworms (enriched)	61.70	19.90	12.90	1.52	3.98	212.00	0.0599	0.336
Mealworms with vitamins	56.70	21.40	12.40	2.34	7.16	226.00	0.396	0.335
Silversides, Whole, Lot #755913	77.90	14.20	5.91	2.60	<0.5	110.00	0.379	0.325
Tubifex Worms	91.40	5.33	1.72	0.39	1.16	41.50	0.0109	0.109
Waxworms	63.5	15.2	18.3	.949	2.0	233	0.0253	0.199
Waxworms with vitamins	58.30	14.30		1.55			0.461	0.192

Table 8. Nutrient analysis (Michelson Laboratories Inc. March 2011 on food supplied by ASLC)

Nutrition Category	**Krill**	**Mussels**	**Oyster**	**Silversides**	**Herring**
Moisture (%)	80.6	83.45	84.07	75.01	76.38
Protein (%)	10.98	10.91	7.54	15.77	16.41
Fat (%)	5.38	1.58	2.58	6.69	4.86
Ash (%)	3.14	2.78	2.33	2.52	2.08
Carbohydrates (%)	0	1.28	3.48	0.01	0.27
Calories (per 100 g)	92	63	67	123	110

The following labs, both located at 6280 Chalet Drive, Commerce, CA, 90040-3761, can perform nutritional analyses:

- N.P. Analytical Laboratories
- Michelson Laboratories Inc.

The following is a list of reputable companies in which appropriate diet/foods or related supplies can be obtained from (other companies may be found locally):

Table 9. List of companies that supply acceptable shorebird food

Company	**Food type**	**Phone number**
Animal Specialties	Flamingo diet	503-981-4738
Aquatic Foods		559-291-0623
Arbico	Fly larvae	800-827-2847
Atlantic Pacific	Fish	401-294-9570
Bassett's Cricket Ranch	Crickets	800-634-2445
Bionic Bait		
Fluker Farms	Insects	800-735-8537
Krill Canada Corp	Krill (*Superba, Euphausia* spp.)	604-533-0038
Natural Balance Pet Foods	Carnivore diet	800-829-4493
Mazzuri		
Nature's Way	Carnivore diet	800-829-4493
Northwest Zoological Supply	Insects	425-776-0724
Penn Cove Shellfish	Mussels	425-743-2033
Rainbow Mealworms	Insects	310-635-1494
RodentPro		812-867-7598
Roudybush		800-326-1726
Xanadu Seafoods	Fish	425-718-7752
Ziegler	Insects	717-677-6181

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 the taxa or species specified. Diet preparation staff should remain current on food recalls, updates, and regulations per USDA/FDA. Remove food within a maximum of 24 hours of being offered unless state or federal regulations specify otherwise and dispose of per USDA guidelines.

AZA Accreditation Standard

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

AZA Accreditation Standard

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

If browse plants are used within the animal's diet or for enrichment, all plants must be identified and assessed for safety. The responsibility for approval of plants and oversight of the program should be assigned to at least one qualified individual (AZA Accreditation Standard 2.6.3). The program should identify if the plants have been treated with any chemicals or near any point sources of pollution and if the plants are safe for the Charadriiformes. If animals have access to plants in and around their exhibits, there should be a staff member responsible for ensuring that toxic plants are not available.

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 need to 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 to oystercatchers as an edible enrichment item.

5.3 Nutritional Evaluations

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

- Regular weights
- Bill measurements
- Feather quality
- Blood draws checking CBC and biochemical profiles
- Fecal testing for parasitic ova

More research is needed to recommend values for serum and tissue nutrient levels for shorebirds.

Chapter 6. Veterinary Care

6.1 Veterinary Services

Veterinary services are a vital component of excellent animal care practices. A full-time staff veterinarian is recommended, however, in cases where this is not practical, a consulting/part-time veterinarian must be under contract to make at least twice monthly inspections of the animal collection and to any emergencies (AZA Accreditation Standard 2.1.1). 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). The AZA Accreditation Standards recommend that AZA-accredited institutions adopt the guidelines for medical programs developed by the American Association of Zoo Veterinarians (AAZV): (http://aazv.affiniscape.com/associations/6442/files/veterinary_standards_2009_final.docx).

AZA Accreditation Standard

(2.1.1) A full-time staff veterinarian is recommended. In cases where such is not practical, 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 animal collection 24 hours a day, 7 days a week.

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. Routine health inspections by a veterinarian are recommended at least annually, 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.

Protocols for the use and security of drugs used for veterinary purposes must be formally written and available to animal care staff (AZA Accreditation Standard 2.2.1). Procedures should include, but are not limited to: a list of persons authorized to administer animal drugs, situations in which they are to be utilized, location of animal drugs and those persons with access to them, and emergency procedures in the event of accidental human exposure.

AZA Accreditation Standard

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

Commonly used drugs for Charadriiformes include, but are not limited to:

- Anesthetic gases (i.e., isoflurane, sevoflurane)
- Antibiotics (e.g., amoxicillin, cephalexin, clindamycin, enrofloxacin, metronidazole, trimethoprim-sulfamethoxazole)
- Antifungals (e.g., 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.

Animal recordkeeping is an important element of animal care and ensures that information about individual animals and their treatment is always available. A designated staff member should be responsible for maintaining an animal record keeping system and for conveying relevant laws and regulations to the animal

AZA Accreditation Standard

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

AZA Accreditation Standard

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

care staff (AZA Accreditation Standard 1.4.6). Recordkeeping must be accurate and documented on a daily basis (AZA Accreditation Standard 1.4.7). Complete and up-to-date animal records must be retained in a fireproof container within the institution (AZA Accreditation Standard 1.4.5) as well as be duplicated and stored at a separate location (AZA Accreditation Standard 1.4.4).

Many institutions use ARKS and MedARKS, although other systems are also used. 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.

AZA Accreditation Standard

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

AZA Accreditation Standard

(1.4.4) Animal records, whether in electronic or paper form, including health records, must be duplicated and stored in a separate location.

All US birds of this taxa fall under the protection of the Migratory Bird Species Act and certain birds, such as the Piping Plovers and Western Snowy plovers also fall under the U.S. Endangered Species Act. Institutions should check current regulations

6.2 Identification Methods

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

Commonly used identification methods for Charadriiformes include leg bands or implantable transponder chips. Leg bands are easier to place and less invasive but should be monitored closely as they can cause constrictions. This can occur if they slide proximally and get stuck or when placed on young birds as they become tight as the bird matures. Leg bands may be placed either above or below the tarsus. In wading birds it is often more easy to visualize bands from a distance that are above the tarsus. Transponders can be placed subcutaneously between the shoulders or intramuscularly in the breast muscle. Subcutaneously placed transponders are more likely to migrate. Due to the large size of the needle required to implant a transponder, anesthesia may be advisable, particularly if other procedures such as radiographs are going to be performed at the same time.

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.

Figures 3 & 4. Options for band placement.
Photos courtesy of S. McCain

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

AZA Accreditation Standard

(1.4.1) An animal inventory must be compiled at least once a year and include data regarding acquisitions and dispositions at the institution.

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. In both cases, notations should be made on the inventory.

Below is a sample transaction form used by an institution caring for shorebirds. The name of the institution has been blocked out.

[redacted]

Date Transaction Initi	**Initiators Name (Print)**		**Type of Transaction** ☐ Incoming		☐ Outgoing
Institute Information	**Institution Name**	**Contact Person:**		**Phone Number**	
Street		**City**	**State**		**Zip**
AZA Accredited	**Program Consideratio** ☐ SSP ☐ PMP ☐ None	**Addl Information (Terms of Loan, Conditional Tests, perr**			

Sex	**Common Name**	**ISIS Number**	**Notes:**

Animal Info (Check one):	☐ Trade	☐ Sale	☐ Donation	☐ Loan	Amount Paid	Payment made to:	

Incoming Animals Only

Purpose of Transaction: ☐ Exhibit Only ☐ Programs ☐ Breeding			
Is this species included in the ☐ Yes ☐ No	**Estimated Yearly Cost of Dietary**	**Where Will the Species be held?**	
Is there winter holding available? (If yes, describe)	☐ Yes ☐ No		
additional training? (if yes, please describe)	☐ Yes ☐ No		
Additional Information:			

Director	Date	Mammal Curator	Date	Bird Curator	Date
General Curator	Date	Veterinarian	Date	Registrar	Date
Trails Curator	Date				

6.3 Transfer Examination and Diagnostic Testing Recommendations

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

All Charadriiformes 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 F for reference values for hematological serum biochemistry parameters of selected species. Weights can vary according to individuals, time of year, etc. The following is a weight range of selected species complied from the CRC handbook of avian body masses. This is a general guide only, weights can change according to time of year, etc.

Table 10. Weight range of selected Charadriiformes species compiled from CRC Handbook of Avian Body Masses

Species	**Weight range male (grams)**	**Weight range female (grams)**
Avocet, American *Recurvostra americana*	Average = 316.0	Average = 316.0
Dowitcher, Short-Billed *Limnodromas griseus*	73.0–152.0	82.5–154.0
Dunlin *Calidris alpina*	Average = 55.4	Average = 59.7
Dikkop *Burhinus capensis*	400–500	400–500
Godwit, Marbled *Limosa fedoa*	281.0–362.0	240.0–510.0
Gull, Laughing *Larus atricilla*	270–400	270–400
Jacana, Northern *Jacana spinosa*	70–85	102–122
Killdeer *Charadrius vociferus*	83.9–109.0	87.7–121.0
Oystercatcher, Black *Haematopus bachmani*	555.0–648	618.0–750.0
Phalarope, Red *Phalaropus fulicarius*	Average = 50.2	Average = 61.1
Phalarope, Red-Necked *Phalaropus lobatus*	Average = 32.7	Average = 34.9
Plover, Black-Bellied *Pluvialis squatarola*	181.0–263.0	181.0–263.0
Plover, Semipalmated *Charadrius semipalmatus*	37.6–57.4	39.2–56.5
Plover, Snowy *Charadrius nivosus*	37.0–49.0	37.0–49.0
Sanderling *Calidris alba*	47.0–72.5	47.0–72.5
Sandpiper, Least *Calidris minutilla*	19.0–30.0	19.0–30.0
Sandpiper, Semipalmated *Calidris pusilla*	Average = 31.3	Average = 31.3

Stilt, Black-Necked *Hemantopus mexicanus*	Average = 166.0	Average = 166.0
Tern, Common *Sterna hirundo*	103–145	103–145
Turnstone, Ruddy *Arenaria interpres*	Average = 110.0	Average = 120.0
Willet *Tringa semipalmata*	Average = 215.0	Average = 215.0

6.4 Quarantine

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

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.

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/associations/6442/files/veterinary_standards_2009_final.docx.

AZA Accreditation Standard

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

Quarantine and hospitalization should take place in appropriate sized enclosures that allow for inclusion of a shallow pool, see Chapter 2 for more details about enclosure recommendations. A sandy shore or soft soil that allows normal feeding behavior is desirable. Newly arrived birds may refuse to eat. Adequate privacy and hiding places should be provided. It is best to keep noise to a minimum.

Wild caught birds may take longer to adjust to new surroundings than zoo or aquarium born birds. While it can be tempting to try and monitor birds closely, often it is best to leave them alone for a few days as much as possible. Housing birds with at least one other individual can also be beneficial. A mirror can also be added in lieu of a companion if it is necessary to house a single animal. Housing for animals should be easily disinfected, and allow birds to be maintained in their preferred environmental parameters. Substrate choices should minimize trauma to the feet that could predispose a bird to pododermatitis.

An area separate from the exhibited population is ideal, including a separate water and air supply in case of a transmittable disease. All quarantine 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 and matched to the exhibit photoperiod. A skylight can add additional lighting to the room, but may also increase the air temperature.

A pool is an important part of any quarantine room and 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 flush to the pool when a keeper enters the room.

If quarantine facilities are not available for your taxa, new birds should be held off exhibit, away from other exhibit shorebirds. Ideally, quarantine staff will be separate from exhibit staff. Some facilities will use mammal staff to care for quarantine birds. If quarantine staff cannot be kept separate from exhibit staff, they should wear coveralls, coat and gloves when cleaning/handling quarantine birds. A footbath should be used. Staff should have minimum contact with quarantine birds until after all exhibit birds are cleaned and cared for.

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

AZA Accreditation Standard

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

Ideally, a dedicated staff member that does not care for other birds in the collection should care for Charadriiformes while in quarantine. Alternatively, staff should change clothes and shoes (or wear shoe covers) before and after entering quarantine. There should also be a disinfectant footbath to minimize carrying potential disease into or out of the quarantine area. A variety of disinfectants can be used, including virkon, diluted bleach solution, and quat sanitizer. Work with the staff veterinarian to come up with the best disinfectant for your needs.

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

During the 30-day quarantine period, specific diagnostic tests should be conducted with each animal if possible or from a representative sample of a larger population (e.g., birds in an aviary or frogs in a terrarium) (see Appendix C). If new birds are added to the group after quarantine period has started, it will need to start over again. Minimum 30-day quarantine period is recommended. 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.

AZA Accreditation Standard

(11.1.3) A tuberculin (TB) testing and surveillance program must be established for appropriate staff in order to ensure the health of both the employees and the animals. Each institution must have an employee occupational health and safety program.

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

During the 30-day quarantine period a complete exam should occur. This exam should include a complete physical exam including body weight and evaluation for ectoparasites, confirmation or placement of individual identification method, blood collection if size allows (for complete blood count and biochemical panel, +/- protein electrophoresis), and radiographs. See Appendix F for reference values for hematological and serum biochemistry parameters of selected species. Radiographs allow establishment of a baseline for individuals and also assessment of any abnormalities. Aspergillosis serological testing may be performed, but titers can be difficult to interpret or fail to show evidence of subclinical disease. In larger birds for which a greater blood volume can be collected, extra serum or plasma should be banked at -80 °C (-112 °F) for future disease testing or research purposes. A fecal sample should be obtained for fecal culture and fecal parasite screen. Endoparasites can be treated with pyrantel, fenbendazole, or ivermectin, as appropriate. Capillaria is a relatively common parasitic disease of Charadriiformes and can be difficult to get rid of (Ball, 2003). Ectoparasites can be treated with ivermectin. Testing for avian TB should occur. There are currently no recommended vaccinations for Charadriiformes. Release from quarantine should be contingent upon normal results from diagnostic testing and a minimum of two negative fecal parasite screens performed at least two weeks apart. 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.

Quarantine 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 cardboard box) is recommended, and offering favorite food items can also be considered. The utilization of mirrors has also been successful as an alternative to a live companion bird. Stress behaviors could include pacing, lack of appetite, flying/crashing into walls and/or lethargy.

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

> **AZA Accreditation Standard**
>
> **(2.5.1)** Deceased animals should be necropsied to determine the cause of death. Cadavers must be stored in a dedicated storage area. Disposal after necropsy must be done in accordance with local/federal laws.

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 and birds are more susceptible to disease such as Aspergillosis. Birds that die during quarantine should be necropsied as soon as possible. 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, 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. A pathologist familiar with avian pathology should perform histopathology.

6.5 Preventive Medicine

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

> **AZA Accreditation Standard**
>
> **(2.4.1)** The veterinary care program must emphasize disease prevention.

Routine health inspections by a veterinarian are recommended. Often these are performed opportunistically, although if possible, should be performed annually. A complete exam should include a thorough physical exam including body weight, blood collection (if size allows) for complete blood count and biochemical panel, and radiographs. No specialized equipment is needed for preventive medical procedures for Charadriiformes. Fecal parasite screens should be performed twice yearly, followed by appropriate deworming. It may be appropriate to take a representative group fecal sample where individuals are housed together. No specific vaccinations are recommended at this time, although Charadriiformes have been shown to be competent reservoirs for West Nile virus and vaccination may be considered (Travis, 2008).

Blood samples can be collected under manual restraint or general anesthesia. See Appendix F for reference values for hematological and serum biochemistry parameters of selected species. The jugular vein, ulnar vein, and metatarsal vein are all commonly used sites. Care should be taken to provide adequate hemostasis when using the ulnar or metatarsal vein, as these sites tend to have prolonged hemorrhage when compared to the jugular vein. A wrap can be applied to the leg for a brief period if necessary.

The main methods of restricting flight include pinioning chicks, regular wing trimming, and the use of covered enclosures. While pinioning is more invasive, regular clipping requires frequent restraint, which can result in injury. The pros and cons should be discussed at each institution. Many shorebirds kept in zoos and aquariums are non-releasable due to wing injuries and may not require additional flight restriction methods. Handling during molting should be avoided or limited. If possible, shipment should not occur during molt.

Neonates: 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 the 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. Chicks have been successfully raised in exhibits, but problems can occur because of aggression from adults or unfamiliarity with the exhibit.

<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 3–5 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. Early hobbling of the chick's legs can often minimize splay leg syndrome. 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 bacterial 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 also been noted in geriatric Charadriiformes. Cases of gout may be related to age-related renal changes. Treatment is similar to other avian species. Caution is warranted with non-steroidal anti-inflammatory agents that can result in adverse renal or gastrointestinal side effects. Meloxicam at 0.1 mg/kg (with a single loading dose of 0.2 mg/kg) once daily for 5–7 days has been successfully used in hydrated Charadriiformes without renal compromise.

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

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

Salmonella spp., *Erysipelothrix* spp., *Campylobacter* spp., and *Yersinia* spp. have been reported in Chardriiformes and have zoonotic potential (Ball, 2003). Sick birds should be isolated to limit spread of disease. Good hygiene is important in limiting spread of disease. Use of gloves when handling infected birds or their feces can help prevent transmission to humans. Disinfectant footbaths should also be used.

Ideally, a dedicated staff member that does not care for other birds in the collection should care for Charadriiformes that are in quarantine or while sick. Alternatively, staff should change clothes and shoes (or wear shoe covers) before and after entering quarantine or sick bird areas. There should also be a disinfectant footbath 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 need to be appropriately disinfected, as designated by the veterinarian supervising quarantine before use with resident animals.

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).

> **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.

While currently there are no known Charadriiformes used for off-grounds programs, the potential dose exist. Such birds should be housed separately from collection Charadriiformes 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**
>
> **(11.1.3)** A tuberculin (TB) testing and surveillance program must be established for appropriate staff in order to ensure the health of both the employees and the animals. Each institution must have an employee occupational health and safety program.

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

TB testing is not required for animal care staff to work with Charadriiformes. *Mycobacterium avium* should be ruled out for any bony lesion.

6.6 Capture, Restraint, and Immobilization

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

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

Manual restraint is appropriate for most Charadriiformes. If possible, the target individual should be restricted to a small area rather than caught in a large enclosure. The smaller the area and the fewer birds in the area, the easier it is to catch the desired bird. A net may be used.. A light towel may be helpful for restraint. 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. If possible, 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.

In larger Charadriiformes, the wings should be secured by holding the body tucked under one arm to help prevent any damage. A finger should be placed between the legs for better control and so that they don't rub together. The head should be restrained with the other hand. Care should be taken with the beak, which can induce injury, however avoid covering the nares. In some cases a second person may be helpful to restrain the head. In small Charadriiformes, the bird can be held upright against the handler's body with one hand securing the wings and one hand securing the beak (see Figure 5). In even smaller species, such as plovers, the restrainer can hold the bird in one hand with the back of the bird against the palm of the hand and the head between the index and middle finger.

Figure 5. Restraining an American avocet
Photo courtesy of A. Greenebaum

Care needs to 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.

The most common anesthetic agents used for Charadriiformes are isoflurane or sevoflurane. These can be administered via facemask after manual restraint. Facemasks can be fabricated from empty syringe cases with part of a disposable glove placed over the end to better-fit long beaked birds (see Figure 6). Intubation should be performed when control of the airway is desired such as in prolonged procedures or if the bird is not ventilating well on its own.

Figure 6. Example of facemask for anesthesia
Photo courtesy of S. McCain

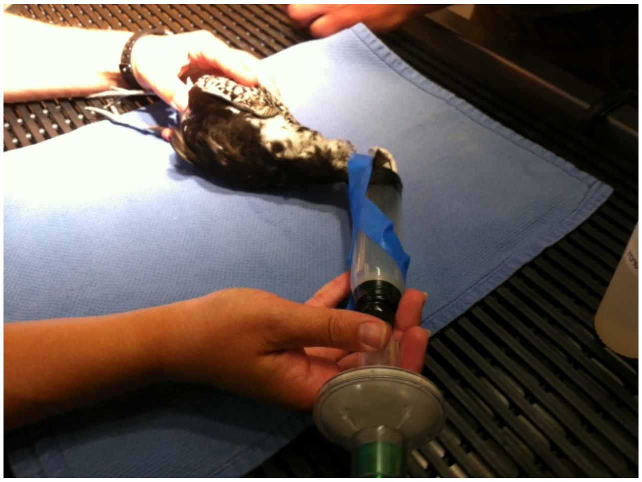


Figure 7. Black bellied plover under anesthesia
Photo courtesy of A. Greenebaum

Anesthetic monitoring is similar to other birds. Esophageal or cloacal temperature probes can be used. Modalities such as electrocardiography, Doppler, pulse oximetry, capnography, and blood gas analysis have potential application in Charadriiformes. Birds should be restrained during recovery and not allowed to recover in a crate, as they tend to flap as they recover and can injure themselves. The same restraint methods should be used as just described; making sure keel movement is not restricted.

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

AZA-accredited institutions should have an extensive veterinary program that manages animal diseases, disorders, or injuries and has the ability to isolate these animals in a hospital setting for treatment if necessary. Charadriiformes keepers should be trained for meeting the animal's dietary, husbandry, and enrichment needs, as well as in restraint techniques, and recognizing behavioral indicators animals may display if their health becomes compromised (AZA Accreditation Standard 2.4.2). Protocols should be established for reporting these observations to the veterinary department. Charadriiformes hospital facilities should 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

(2.4.2) Keepers should be trained to 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, keepers should not diagnose illnesses nor prescribe treatment.

AZA Accreditation Standard

(2.3.2) Hospital facilities should have radiographic equipment or have access to radiographic services.

Charadriiformes 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.

Quarantine and hospitalization should take place in appropriately sized stalls that allow for inclusion of a pool. A pool is an important part of any holding room and should be kept clean at all times. 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. Housing for ill animals needs to allow for ease of disinfection, and permit birds to be maintained within their preferred environmental parameters. For birds with limited mobility or bandages, pool access may need to be initially restricted until the bird is more recuperated. It is recommended that substrate minimize potential foot trauma 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 wet birds to dry with ease.

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 and matched to the exhibit photoperiod. A skylight can add additional lighting to the room, but 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 can be considered, as this might minimize stress to the compromised bird. However, this action requires careful consideration since 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, that this may reduce the stress of separation from the colony. The risk of disease transmission, however, needs to be weighed against the social and psychological benefit of cooperative housing.

Bacterial diseases: Bacterial infections can be primary or secondary. *Salmonella* spp., *Erysipelothrix* spp., *Campylobacter* spp., *Chlamydophila* spp., and *Yersinia* spp. have been reported in Chardriiformes and have zoonotic potential (Ball, 2003). *Mycoplasma* spp. are also occasionally encountered. *Mycobacterium avium* infection is uncommon, but 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* often is implicated in cases of respiratory tract disease in Charadriiformes kept in zoos and aquariums, and was implicated in chick mortality of black stilt chicks (Reed et al., 2000).

Culture and sensitivity of suspected bacterial infections is indicated. In cases where bacterial infections result in disease in Charadriiformes, systemic antibiotics can be administered intramuscularly or orally though medicated fish or gruel.

Viral diseases: Avian influenza is not uncommonly found in Charadriiformes. Typically it causes an in apparent or subclinical infection. A mortality event occurred in wild common terns in South Africa in 1961, but this is not typical (Friend, 1999). Ten of the 15 hemagglutination, and eight of the nine neuraminidase types, have been identified in shorebirds, and many of the combinations are unique to Charadriiformes. The H9 and H13 types predominate. The avian influenza type H5N1 has been identified in shorebirds.

Newcastle disease virus and infectious bursal disease have been serologically in wild Charadriiformes. The significance of these diseases is unknown. A mortality event has been associated with a reovirus in American woodcock, and avian pox has been diagnosed in Sanderlings (Docherty, 1994; 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).

Fungal diseases: Aspergillosis is not uncommon in Charadriiformes kept in zoos and aquariums, 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 in zoo and aquarium settings. 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 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 inappetence. 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 waterproofing, and on auscultation, an audible pulmonic/tracheal click or decreased respiratory sounds.

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 Charadriiformes. 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, 1999).

Young birds are particularly at risk for candidiasis, with hand-reared birds being the 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.

Parasitic diseases: The most common reports include cestodes and capillaria (Ball, 2003). Cestodes are more common in wild-caught birds than zoo born, 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. *Cyclocoelum* sp. trematodes have been seen in shorebirds and may be associated with respiratory infection, airway obstruction, and acute death.

Lice do not generally affect feather quality. Occasionally, however, these parasites can cause pruritis. Treatment with oral or injectable ivermectin, or dusting with a pyrethrin-based insecticide is effective for pediculosis.

Non-infectious diseases: The most common non-infectious diseases in zoo or aquarium housed Charadriiformes include trauma, pododermatitis, and circumferential foreign body entanglement of the digits. Trauma is not uncommon, either accidental or from inter-specific aggression. Treatment of fractures or ligament repair needs to allow the bird to be functional. Water access restrictions need to 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 an animal 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. Sand substrate has been used successfully and needs to be cleaned of debris and feces at least daily. Sand depth ought to be adequate to ensure proper drainage. Pine shavings need to be used with caution, as they have been associated with pododermatitis in shorebirds. 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-term 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.

Foreign objects such as thread, hair, and other string-like material can wrap around digits and cause avascular necrosis. If identified quickly, damage can be minimal; however, it is not uncommon to require partial amputation. Walkthrough exhibits should be monitored closely for this reason.

Metabolic bone disease has been seen in American avocets and black-winged stilts. Scissor bill and curly toe can result from inappropriate incubation and are seen in avocets and stilts. Curly toe can often be corrected with splints and appropriate substrate. Corrective trimming can be attempted to realign the bill, although some deviation may persist. It is important to ensure adequate nutritional intake during this time.

Neoplasms have not been commonly reported. There is a single report of a suspected teratoma in a black-headed gull (Baker, 1981). Environmental contaminants such as pesticides, heavy metals, industrial chemicals, and petroleum products, while common in free-ranging shorebirds, are not typically seen in zoo or aquarium settings.

AZA-accredited institutions must have a clear process for identifying and addressing Charadriiformes 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 a clear process for identifying, communicating, and addressing animal welfare concerns within the institution in a timely manner, and without retribution.

There are no specific protocols for reporting welfare concerns for Charadriiformes and each individual institution's policies should be followed. If necessary, the appropriate SSP should be contacted.

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

AZA Accreditation Standard

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

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: 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. It is required that the histopathology be performed by a pathologist familiar with avian pathology. For reference values (mean ± SD) of hematological and serum biochemistry parameters of selected species, please see Appendix F.

Chapter 7. Reproduction

7.1 Reproductive Physiology and Behavior

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

While the information below highlights breeding behaviors of shorebirds typically found in AZA institutions, there is still much to be learned about the behaviors these animals engage in as they begin to reproduce. Typically, shorebirds can use several cues for the breeding season, including light cycle, food availability, breeding plumage, and/or the presence of nesting areas and nesting substrate. It is important to note that many shorebirds in institutions are non-releasable because of wing injuries, and these injuries may prevent the males from being able to copulate.

Normal hormonal values for shorebirds needs further research but you should see an increase in calcium levels for females during egg laying season. See Table 11 for more information about age of sexual maturity.

Avocets and stilts: May have pre- and post-copulatory displays. Pre-copulatory display can be initiated by female solicitation posture or by male and female sexual preening. For example, the female American avocet assumes a posture in which the body is held parallel to the water with head and neck extended. As the sequence advances, the male moves from one side of their mate to the other, always from behind. Each time the male sidles next to his mate, he preens his breast feathers in an upright posture. As the preening becomes more exaggerated, the male will sometimes splash the female with water. The male jumps on the female’s back for copulation. Immediately after copulation, the pair cross bills, extend wings over each other’s back and walk briskly for a short distance through the shallow water until the part and resume feeding. Stilts also have very similar displays (Colwell, 2010). They typically have a clutch size of 3–4 eggs, an average of a 26-day incubation period, and the chicks fledge around 27 days of age.

Dikkops: The spotted dikkop is monogamous and usually a territorial, solitary nester, sometimes forming loose colonies. It is likely that the spotted dikkop will undergo a very radical personality change when starting to nest. They tend to make loud, growling noises and spread their wings out above the ground. If pushed, they will physically attack the leg of a keeper. This behavior has been noted at AZA-accredited institutions (Jones, 1999). Dikkop keepers need to be aware that when they see this behavior, they need to tread carefully and keep a close eye out for a nest.

The eggs and nest are very subtle and will easily blend into the substrate. The nest is a simple, shallow scrape in the ground. It can be unlined or lined with twigs, leaves, stones, or animal droppings. The egg-laying season is from August to April with the peak of egg production in September through January. A clutch of eggs can range from one to three eggs. Both sexes incubate the eggs for between 24–30 days. More specifically for the spotted dikkop, one egg is laid 56.52% of the time and two eggs are laid 43.48% of the time. According to the AZA Spotted Dikkop studbook, the incubation period is 24 days. Bigalke (1933) has also reported this incubation period (Pinger, 2012).

Chicks leave the nest around 24 hours after hatching and both parents feed the chicks. The average weight of a newly hatched spotted dikkop is 25 g. This is an average from specimen reports from three AZA-accredited zoos. The chicks fledge at around 8 weeks. The chicks are also hard to see and will flatten themselves on the ground at the least sign of danger. In the wild, the spotted dikkop will use distraction techniques to protect the chicks, if a predator threatens. The chicks crouch and remain still, and the adults pretend to have a broken back, wing, or leg to draw the predator away (Hockney et al., 2005). At first glance, the chick may appear to be dead (Jones, 1999). This chick behavior has been observed in zoos in the stone-curlew. In this species, the chicks may lay on their side with the legs stretched out straight (Cwiertnia, 1999).

Breeding behavior in the stone-curlew starts with nest construction and the “deep-bow” display by the male and female. The male selects the site by pointing at the area with his bill and the female begins to scrape the area. Both birds then sit in the nest scrape and shuffle and pick up pebbles and throw them over their shoulders. The birds may make several nest scrapes before they choose one. Then the “neck-arch” display is seen, a behavior in which both birds stand upright and arch their necks downwards. Copulation will usually follow after this display (Cramp & Simmons, 1983). When a pair is formed, they begin to chase other birds away and then the pair is moved to their own breeding aviary (Cwiertnia, 1999). Dikkop chicks can fly by 6–7 weeks and may become members of other flocks of dikkops. In the wild, stone-curlew chicks gain weight very slowly for the first few days and then rapidly gain weight (Westwood, 1983).

Gulls and terns: Pre-copulatory behaviors in the gull and tern group are: courtship feeding of the female by the male, nest site selection, and nest construction. Both groups can be aggressive during the breeding season and mob anything that enters their nesting territory. In temperate and polar zones, most gulls and terns breed at the same time every year. All species are monogamous and both male and female incubate, defend a territory, and care for the young. The normal clutch size is 2–3 eggs and the incubation period for the group ranges between 20–30 days. Young gulls do not remain with their parents as long as young terns. Young terns remain with parents longer so they can learn how to plunge-dive for food. In the wild, gull nests are usually mats of vegetation with a central nest cup. It is usually built on the ground but some build nests on cliffs and some nest in trees. On the other hand, tern nests in the wild are generally unlined scrapes on the ground. Some species may put a loose collection of sticks together for a nest.

Jacanas: Jacanas have complicated polyandrous mating systems, whereby a female has access to several mates simultaneously or sequentially and holds territories against other females. The average territory size is .36 acres for males, and .88 acres for females (Jenni & Collier, 1972). An individual female may have up to four or more mates simultaneously, with an average of two or three. As the male's territory is very large, in situations where the breeding habitat is small or the quality is not optimal, and the female has no opportunity to monopolize more than one male, then the mating is monogamous.

These birds breed throughout the year, but in the wild, the highest amount of breeding and egg-laying occurs during the summer months, starting in the early part of summer to late summer (Tarboton, 1993). Nests are usually built at least 50 m (164 ft) from each other (Tarboton, 1993). In zoos, nests are usually built of all sorts of plant matter, which is trodden down and collected. Nesting occurs at the edge of the pool or on tiny islands in the pool. Usually the birds re-use the same nest all the time. The jacanas at one institution were seen using a floating island constructed of styropore, covered with coating, soil, and plant matter. Islands need to be fixed to land with a strong wire to prevent floating away.

Depending on the size of the territory and enclosure provided, fledglings may need to be removed if the adults become intolerant towards the chicks. This is possible when the adults have a second clutch. Since the males may not be able to cope with two clutches at the same time, the chicks of the first clutch should be removed as soon as chicks from the second clutch begin to hatch. Since chicks have a much higher feeding rate when accompanied by the adult male, it is recommended that chicks stay with the males for as long as possible. Although adults never feed the chicks, they do lead them to feed and point to the food with their bills.

The following is specific breeding information for the wattled and African jacana. In the wild, the wattled jacana uses a small, often partially submerged, collection of stems and aquatic vegetation for a nest. In the wild, females defend and breed with 1–3 (or more) neighboring, territorial males. The number of mates is variable and dependent on the size and quality of the habitat, with a greater number of smaller territories occurring in areas of dense, mixed-species mats of floating vegetation. Females vigorously exclude other females and assist their mates to drive out intruders. The age of sexual maturity is assumed to be at least 1–2 years of age. Wattled jacanas fledge between 42–84 days.

In the wild, the African jacana's nest is a flimsy, mostly submerged pad of aquatic vegetation, extending to 2 cm (.78 in.) above the water surface, and usually located over deeper water. These birds are polyandrous, but their breeding strategy is highly variable. Males hold nesting, breeding, feeding, and chick rearing territories while females, together with one to several adjacent males, hold their territories against other females. Some males are sequentially polygynous, because new females keep replacing their former mates. Sexual role reversal is complete with males performing most nest building, all incubation, and all care of the precocial young. African jacanas have been known to be able to breed at 1 year of age.

A male will attract a female by calling from a potential nest area, which may contain a few plants to suggest a future nesting site. The male may also put on a display by hopping up and down to catch a female's attention. If she accepts his invitation, she will stand with her bill pointed downward while he walks around her, poking her with his bill. He then hops on her back and breeds with her. Females only invite males to their nesting area less than 30% of the time. Females usually breed with more than one male during a 1–2 hour breeding period that day.

Males are the primary nest builder; a nest consists of water plant materials. It is possible that the male may have to build several nests before one is accepted. If there is an increase of water levels and the eggs begin to float, the males are able to push the eggs to a new nest if necessary. There are generally 3–5 eggs in a clutch. Usually one egg is laid per day until all the eggs for that clutch is laid. The eggs are glossy, tan, and have thick black brown lines. Each female lays a similar egg design throughout her life. Eggs are generally 7.9–10.5 g each. A female may lay up to 10 clutches per year. There may be 4–28 days between clutches. Males usually take over incubation when the third egg has been laid. When chicks are ready to hatch, calls can be heard 24–48 hours beforehand. The eggs generally hatch in the order they were laid. The male will show a great deal of excitement when hatching occurs and will also begin to remove the eggshells once the chick has hatched.

The male will care for the young by leading them to food; the chicks will stay between the wings and the body of the male, which will occur for up to 18 days of age for the chicks. He may also not be the father of all or any of the chicks. The male is very attentive for the first 14 days and then his attention begins to decrease from that point on until 40–50 days of age when the chicks are on their own. Chicks become flighted at 39–33 days of age. Most males will brood two clutches per season.

Lapwings: Both sexes of lapwings make a shallow nest scrape with a few pebbles added. Parents share incubation and defense of the nest. The clutch size is typically 2–4 eggs and they are olive-clay colored mottled with brown. They fledge around 8 weeks. The young birds can remain on exhibit with the parents until the next breeding season; then they are seen as intruders and should be separated. This has been seen in the spur-winged lapwings (*Vanellus spinosus*) (Holland, 2007).

Oystercatchers: Once oystercatcher chicks have been parent reared, the adults have a tendency to show aggression towards the juveniles and they normally have to be removed at fledgling age.

Phalaropes: In the wild, courtship behavior is characterized by several different things: first, aerial chases occur where the initiating bird, usually the female, attempts to remain close to, exhibits appeasement behaviors to, and persistently follows the potential mate; then the initiating bird attempts to drive away same-sex conspecifics. Some aggression between potential pair members may be seen and should be monitored closely. Finally, there is gradual acceptance of the initiating bird by the potential mate. Pair formation can take as little as a few hours and will end with either the last egg laid or persist up to 13 days post laying (Poole, 2005).

Sandpipers/plovers: They typically lay 3–4 eggs. Depending on the species, the average range of incubation can vary from 21–30 days, and chicks will fledge from 18–45 days of age. Most species, once paired, exhibit scraping, a type of courtship display. It occurs early in the breeding season as part of courtship, and continues throughout. In most species, the male usually starts the scraping ceremony by bowing with his bill directed downward and using his feet to scrape a shallow depression in the substrate. Depending on the species and habitat, they may toss small twigs, pieces of vegetation or pebbles as they sit in the scrape. They may create several scrapes but gradually focus their attention on one where the eggs are eventually laid. As very few shorebirds have been breeding in zoos and aquariums; further research is needed for breeding in *ex situ* settings.

7.2 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, taxa and should be investigated further. At this time, artificial insemination techniques have not been used in any capacity with shorebirds.

7.3 Pregnancy & Egg-laying

It is extremely important to understand the physiological and behavioral changes that occur throughout an animal's pregnancy. Gestation and incubation periods for Charadriiformes vary widely (see Table 12 for information). Prior to egg-laying, females tend to lay in their nest more frequently. Parents usually get more aggressive to other birds (and sometimes each other) before and after laying eggs; oftentimes they have to be removed from access to other animals. Often pairs need to be removed when nesting. During parturition, lactation and rearing, certain species may need to be removed from large groups of birds to prevent aggression to the other birds. Equipment used for parturition includes incubator, brooder, heat lamps, and tweezers for if hatching assistance is needed. Changes in appetite can also be seen with egg-laying females and sometimes there is an increase in the amount of food consumed a few days before laying. Also, females' calcium levels tend to fluctuate with reproductive state.

Staff should have experience/knowledge of incubating techniques such as candling, monitoring weight loss, and watching for developmental issues and assistant hatching. Medical problems during reproduction are not unique to Charadriiformes and may include egg binding, thin-walled eggs, and egg yolk peritonitis. Standard diagnostic and treatment techniques used in other avian species are appropriate.

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. The sicker the bird at presentation the more aggressive 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 but instead let the removal of the contents collapse the egg. Once this is complete, the shell should pass by itself. 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.

In dealing with thin-walled eggs, diet should be evaluated for calcium content. In addition, the age of the bird could also be a factor. Older birds may have thinner eggs. If eggs are thin, it should be considered to remove from parent and incubated artificially.

Indicators for egg peritonitis can 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. Endoscopic surgery may be preferred over traditional celiotomy.

Table 11. Gestation and incubation periods of shorebirds

Bird Group	Average clutch size	Average incubation period	Average age at fledging	Incubation temperature	Humidity	Age of sexual maturity	Other notes
Avocets	3–4	26 days from 1st egg laid (temp. dependent)	27 days	37.5 °C (99.5 °F)	55%	1–2 years	
Stilts	4	25 days	27–31 days	37.5 °C (99.5 °F)	55%	1–2 year	
Dikkops	1–3	24–30 days	42–49 days	37.5 °C (99.5 °F)	40–50%	Generally at 2–3 years, but can breed at 1 year	
Gulls and terns	2–3	21–23 days	22–29 days	N/A	N/A	N/A	
Jacanas	3–5	22–23 days	Wattled: 42–84 days African: 40–70 days	37.5 °C (99.5 °F)	80–100%	1 year of age	
Lapwing	2–4	28–32 days	1 month	37.2 °C (99 °F)	60%	18 months	
Oystercatcher	1–3	26–28 days after clutch complete	35 days	N/A	N/A	3–4 years	
Phalaropes	4	17–26; avg. 19 days	18 days	37.5 dry bulb 29.4 wet bulb	N/A	1–2 year	Males incubate and raise chicks
Sandpipers	4	Most spp. 21–23, Larger spp. 27–30	Most spp. 18–21 Larger spp. 32–45	37.5 °C (99.5 °F)	45%	1 year	
Plovers	3–4	Most spp. 23–28	Most spp. 21–28 Larger spp. 34–45	Killdeer/piping plover: 37.4 °C (99.4 °F) Killdeer: 37.8 °C (100 °F) Piping plover: 37.5 °C (99.5 °F) Blacksmith/ shore plovers: 37.2 °C (99 °F)	45-50%	1 year	

7.4 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.”

Due to aggression by the parents, it his highly probably you will have to remove pair off exhibit for incubating and rearing of chicks. If you are artificially incubating, several AZA institutions have a special “incubator room” off exhibit used for incubating eggs. In addition, they should have available off exhibit brooding and chick rearing space.

If they are left with the parents, check for species-specific information for appropriate nesting material, but in general, most shorebirds will use small grasses, leaves, etc. or some just make a scrape in the sand. If parents are removed off exhibit with the nest, the holding should be checked to make sure that all water sources are easy to get in and out of for chicks. The holding needs to be adequate such that small chicks cannot escape or get stuck somewhere.

Every facility that will be incubating eggs should have a separate incubator room. This room should be kept clean and sterile, have filtered air, and be able to candle the egg inside the room (the room should be able to get dark). If the parents are going to be removed from the exhibit, it should be done before breeding/nesting occurs—well before she is expected to lay. It is usually done at the start of the breeding season, typically in the spring. The parents could take up to several weeks to acclimate to the off exhibit holding before they would be comfortable enough for breeding, nest building and copulation. Once they lay the eggs, you can either leave the eggs with the parents or artificial incubate them. If you remove the eggs, fake eggs should be given to the parents to avoid reclutching. If they are artificially incubated, they will need to be hatched out in a hatcher, then moved to a brooder for raising.

7.5 Assisted Rearing

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

Shorebirds are typically successful in giving birth and caring for offspring. Chicks are precocial, so they do not need to be directly fed by the parents. Parents typically help brood to keep the chicks warm and lead them to food. Staff should be familiar with species natural history, appropriate substrate and food items.

Shorebirds have been successfully raised off-exhibit without parents by providing a heat source and plenty of food that is size appropriate and easy to find. Some places will hang a feather duster for chicks to stand under to simulate parents. Care should be taken to ensure no loose or straggly pieces are available that could wrap around the chicks. If a chick is by itself, consider adding a mirror so the chick feels like it has companions.

If the parents are not incubating the eggs, the eggs may need to be removed and placed in incubator. If the egg needs to be artificially incubated, at one AZA-accredited institution, the incubator parameters of the wet bulb temperature was 29.5 °C (85.1 °F) and the dry bulb temperature was 37.5 °C (99.5 °F). Two days before hatching, the eggs were put into a hatcher; the humidity was slightly increased and the temperature decreased. The chicks stayed in the hatcher for 1 day until they were dry. Another institution reported that within 48 hours of being in the air cell, and within 24 hours of the first external pip, the chicks were hatched out of the egg.

Once it is time to introduce the fledged chicks into the exhibit, keepers may have to remove some of the more aggressive animals until the new birds have a chance to acclimate. It is recommended to wait until after breeding season is finished so the exhibit birds are less aggressive. Depending on the birds' temperament, keeper staff will have to decide between releasing the birds directly into the exhibit and doing a howdy process. All new birds should be monitored closely for acclimation.

Jacanas: A sample hand-rearing protocol for jacanas follows. The post-hatching rate for jacanas is extremely slow, although this rate is highly variable between and within broods. Hand-raised Jacanas can show signs of imprinting. It is recommended that a puppet be used for the first 3–5 days to resemble the parent.

Initially at 0–2 weeks, healthy chicks can be housed in a forced-air brooder at a temperature of 35–36 °C (95–97 °F) with a relative humidity of greater than 50%. Chicks can easily be held together as they are not aggressive to one another. Temperature in the forced-air brooder can be gradually reduced to approximately 32 °C (90 °F) by the time chicks are 2 weeks old. Two or three layers of terrycloth toweling placed on top of the brooder will insulate against temperature fluctuations. The brooder floor should also be covered with terrycloth towels, a few layers thick. When using an Animal Intensive Care Unit (AICU) brooder, be sure that the towel is banked on the front of the brooder to prevent toes from getting caught in the sliding door channels. Toweling should be covered with a non-slip matt, such as light-colored rubber shelf liner or bar mat. A feather duster should be suspended in one corner of the brooder.

At approximately 14 days, chicks can be moved from the AICU brooder to a larger precocial brooder, with a temperature gradient created by use of a localized radiant heat source (ceramic and/or infrared bulbs). Temperature under the center of the heat should be 35–38 °C (95–100 °F), and the minimum temperature away from the heat source should be 24 °C (75 °F). The bottom of the brooder should be lined with non-slip matting. An uneven walking surface, created by placing rolled toweling under the surface towels, will help to prevent foot and joint problems. If a clear-walled brooder box is used, at least three sides should be covered in opaque material as a sight barrier to reduce stress and prevent pacing. Open brooders are typically covered with small mesh, as birds are able to fly before 60 days. Non-toxic, artificial plants and grasses, serving as both sight barriers and walking substrates, should be placed in the brooder. Plants chosen should not have parts that can be pulled off and eaten. It is recommended to provide appropriate-seized perching (>1.3 cm [.5 in.] deep), preferably with a rough texture to aid with foot conditions. A larger, shallow dish, filled with water 2.5–5.1 cm (1–2 in.) deep, will serve as a small pool.

By 60 days, chicks should be transferred to an outdoor aviary, weather permitting. Birds kept indoors for long periods are prone to cracking on the plantar surfaces of their feet.

Lapwings: A sample hand-rearing protocol for lapwings follows. At hatching, temperature in the brooder needs to be reduced to 36.5 °C (97.7 °F), and the relative humidity should be increased to 66–70%. Movement of the egg and vocalizations can be seen and heard within 2 days of hatching. Leave the chick in the incubator for the first 8–10 hours until down is dry. Then, the chicks can be carefully moved a brooder box with a temperature from 32–34 °C (89.6–93.2 °F). Chicks may be inactive for up to 36 hours before they move around and feed. Gradually reduce the temperature for 10 days until it is around room temperature (25 °C [77 °F]), while still providing heat at one end of the brooder. River sand, brooder matting, or towels are all good substrates.

Offer live food like mealworms on top of pellets to encourage feeding. A shallow bowl of water with pebbles in it should be provided from the first day. Starting on the fifth day, a bowl of moistened softbill pellets, minced up meat, and live insects can be offered. The aim should be to provide the chicks with approximately 24% protein. If the protein level is too high, leg problems will develop. If possible, put the brooder box outside in mottled sunlight to help provide vitamin D. The amount of time in sunlight can vary according to the outside temperature; the chick should not be allowed to get too cold or too hot, but a minimum of 10 minutes a day is recommended.

Lastly, to serve as a reference for institutions that may be involved in hand rearing masked plovers, here is a sample chart that tracks the weights of masked plovers as they age.

Table 12. Sample weights (g) of masked plovers over time

Day	Weight (g)
0	18
5	25
10	30
15	47
20	65
25	100
30	130
35	180
40	225
45	280

(Holland, 2007)

7.6 Contraception

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

Typically for contraception, the real eggs can be replaced with dummy eggs, allowing the birds to incubate the fakes for the normal incubation range. If eggs are pulled without being replaced by dummy eggs, then the female bird may repeatedly attempt to lay another clutch, which could cause depletion in calcium levels. After the normal incubation period is over, if the parents have not deserted the nest, then the dummy eggs should be pulled. Some birds have been known to continue to incubate dummy eggs long after the incubation period is over which could lead to potential husbandry problems, such as leg problems from laying down so much.

Based on recommendations made by the American Association of Zoo Veterinarians, avian embryos older than 50% gestation should be killed using decapitation, an overdose of anesthetic, or other methods considered appropriate for hatched birds. This is because by 50% gestation, the neural tube has sufficient development for pain perception.

For eggs younger than 50% of gestation, the egg needs to be pulled and cooled, and then a determination about whether it is fertile or not should be made. Cooling is a relatively simple method, and if it is done early enough in embryo development, it is humane. One drawback is that “egg age” should be monitored closely. This is still an area where more research would be beneficial.

Chapter 8. Behavior Management

8.1 Animal Training

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

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

AZA-accredited institutions are expected to utilize reinforcing conditioning techniques to facilitate husbandry procedures and behavioral research investigations. While developing a training program for a shorebird collection is still quite new in most zoological settings, it is a completely worthwhile investment. It provides the birds with the opportunity to voluntarily participate in their own health care using less invasive procedures (e.g., the bird voluntarily enters a holding cage or steps on a scale without being grabbed up by a keeper), and provides them with additional behavioral enrichment.

For most facilities, incorporating a husbandry-training program as part of environmental enrichment for shorebird collections may be a completely new development. Altering husbandry practices that allow species individuals to choose to participate in their own health care adds the element of cognition; birds think about what is being asked, make decisions, and learn what actions are being positively reinforced.

Adding in husbandry objects such stations, scales, kennels, etc. changes the normal everyday shorebird exhibit. Through training sessions, unusual environmental objects become less intimidating when being paired with desired food items. Each training session provides an opportunity for a change in environment and behavior. Several factors, such as time of day of training session, training location, length of session, etc. can create variable bird behavior. Training sessions also allow a shorebird collection’s diet to be delivered in an alternate way. Rather than being given food or searching for food, the bird has the opportunity to earn food.

With vision being the dominant sense in most birds, and because of the skittish nature of shorebirds, a gentle food toss has worked best for primary reinforcement delivery and can be used rather than requiring individuals to hand feed. A low, calm, verbal “good” with gentle arm movements can be used when delivering reinforcement. Clickers and whistles have also been used successfully as a bridge. If there are known leeriness and skittish tendencies in the collection, baiting can be used with food being tossed near the station/scale, then encourage or lead birds toward them.

Stationing: Depending on the number of individuals in a facility’s shorebird collection, station locations can be established at different areas on exhibit to reduce competition and aggression between birds.

Scale training: To avoid excessive handling of shorebirds while monitoring general health and documenting seasonal weight changes, a scale can be used to weigh birds on a monthly (or as needed) basis. The scale platform should be positioned in an area where the birds feel comfortable. A familiar substrate, such as nomad matting, can be placed over the scale in order to hide the bright silver color of the scale, and provide the birds with good footing when they step onto the platform. Scale training can make monthly weighing a relatively quick process, without the need to handle or restrain the birds. More information is still needed in this area.

Catch cages: Having the shorebirds routinely enter a catch cage—which will reduce fear of it—has been used successfully for capturing.

8.2 Environmental Enrichment

Environmental enrichment, also called behavioral enrichment, refers to the practice of providing a variety of stimuli to the animal's environment, or changing the environment itself to increase physical activity, stimulate cognition, and promote natural behaviors. Stimuli, including natural and artificial objects, scents, and sounds are presented in a safe way for the Charadriiformes 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.

Behavioral management programs should include training, enrichment, exhibit design, and husbandry practices. They should be designed to promote natural behaviors of shorebirds, such as foraging, flying, preening, bathing, wading, swimming, thermoregulation, courtship, molting, observations/alert behavior. For a more detailed description of all natural behaviors, see Chapter 2. As bumblefoot is a common health problem shorebirds have in captivity, special care should be given to promote wading as much as possible. All behavioral programs should be evaluated on a regular basis.

Training is an important component to behavioral management programs. For example, by scattering more of the food, keepers are not able to hand feed as much, which removes the keeper's ability to monitor a bird's exact caloric intake. Having the birds scale trained is an important tool to help monitor the birds' health. The types of enrichment listed in Table 13 have been used at other AZA institutions, but all items should be approved by your veterinary staff for safety concerns such as entanglement, ingestion, entrapment or dietary concerns.

AZA-accredited institutions must have a formal written enrichment program that promotes Charadriiformes-appropriate behavioral opportunities (AZA Accreditation Standard 1.6.1).

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

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. All enrichment items should go through an appropriate approval process that considers safety, behavioral 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 Appendix I for examples of written enrichment sample plans and new enrichment evaluation forms. Table 13 illustrates several examples of enrichment devices typically used with shorebirds and what behaviors those devices elicit.

Table 13. Examples of enrichment devices typically used with shorebirds and associated elicited behaviors*

Enrichment technique/device	**Description**	**Behavior(s) elicited**
Bird vocalizations	Play recordings of bird vocalizations. *Do not use recordings that might unduly stress the collection, (i.e., do not use overt aggressive or territorial vocalizations).	Vocalizations and search behaviors
Scatter feed or use of PVC dispensers	Use methods to feed items such as insects or worms around exhibit *in lieu* of food bowls. Food can be broadcast or hidden in a variety of ways: (1) scatter on surface of exhibit substrate; (2) scattered amongst vegetation; (3) gently raked under an inch or less of exhibit substrates such as sand or dirt; (4) hidden along the edges of logs or stones; (5) placed in PVC dispensers that are either laid on the ground or hung overhead.	Natural foraging behaviors such as stitching, probing, etc.
Mirrors	Setup small mirrors in the exhibit or holding, change mirror locations if left in more than a day. *Watch for aggressive behavior towards mirror and remove if needed.	Vocalizations and display behaviors

Mists/showers	Mists and/or showers provided on a random schedule.	Bathing and preening
Rearrange exhibit furniture	Change location and arrangement of exhibit furniture (logs, rocks, plants, bathing pans, etc.).	Stimulates investigation of furniture as well as promoting the use of more of the exhibit
Water features: change depth or add movement	Change size of bathing pans, tidal changes, etc.	Foraging, wading, walking, probing, stitching
Offer food items on a variable schedule and/or offer different food choices	Offer random feeding times and/or different food choices.	Increased foraging
Use of live food	Offer live foods such as bivalves, insects, annelids, etc. Offer bivalves in the shell.	Walking, running, foraging (e.g., stabbing, pecking, probing, stitching, hammering). Increases time required for foraging and eating.
Turning over substrate or making small sand piles	Either turn substrate over with a shovel or create small mounds or hills. The latter can have live food such as mealworms added to them, or be created from pond sand with live saltwater organisms if available.	Walking, foraging (stabbing, pecking, probing, stitching). Increases time required for foraging and eating.

*Information for this table incorporates enrichment ideas from several different AZA-accredited institutions

8.3 Staff and Animal Interactions

Animal training and environmental enrichment protocols and techniques should be based on interactions that promote safety for all involved.

Due to their small size, shorebirds do not pose much of a threat to animal care staff members. In contrast, care should be taken so that the reverse is not true. Staff members should be aware of the hesitant, skittish nature of shorebird species, and take caution not to create unnecessarily stressful situations. Slow approximations and patience are an important factor in developing a trusting relationship while training. It is preferable for staff members to work with a shorebird collection in a "free contact" setting; however, increasing proximity toward individuals in a collection will depend on the comfort level of the bird. A squatting body posture position while training will improve the bird's comfort level as well. There are no known human safety issues working with shorebirds and keeper staff can have direct contact with the birds without protection.

Exhibit designs that include holding areas or visual barriers to separate individuals can benefit animal management, and may also enhance training in competitive group dynamics. A small, flat surface appropriate for scale placement that would also give the keeper access to the birds within a comfortable distance would be an excellent exhibit addition.

Often, the more aggressive birds can take over a training area. The size of the exhibit needs to be large enough that it permits work with multiple birds while allowing the birds to remain a comfortable distance from the keeper and conspecifics. In addition, leaving space for props, such as scales and crates, are important in the overall exhibit design. Some zoos and aquariums have designated permanent areas for catch cages within the exhibit where birds can be habituated to the catch; sometimes they are even fed in it. This allows the birds to be extremely comfortable when entering the catch cages, which eases the bird catching process.

8.4 Staff Skills and Training

Charadriiformes staff members should be trained in all areas of Charadriiformes 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.

Staff should be familiar with shorebird natural history, behavior, biology and husbandry techniques, as well as training in the basics of operant conditioning. It is recommended that staff be familiar with their range of vocalizations and postures/displays. They should be able to capture and restrain the bird quickly and safely when needed. New keepers should be trained by keepers experienced in all aspects of shorebird husbandry, training, etc. whenever possible. The AZA Shorebird TAG does not have any specific recommendations for certifications and qualifications needed by animal care staff working with shorebirds, but encourage all institutions to provide opportunities for animal caretakers to gain additional experience in all fields of animal management and care.

All staff members working with a shorebird collection should be aware of all behavioral tendencies exhibited by the birds. If and when a shorebird-training program is developed, staff members should be familiar with basic operant conditioning principles with an emphasis on positive reinforcement. A training plan should outline the overall goal of the behavior, the steps used to approximate obtaining that behavior, and the benefit or purpose of the behavior. Animal and staff safety is a top priority for training and enrichment programs, and appropriate protocols should be approved prior to implementation by management and husbandry staff.

Chapter 9. Program Animals

9.1 Program Animal Policy

AZA recognizes many public education and, ultimately, conservation benefits from program animal presentations. AZA's Conservation Education Committee's Program Animal Position Statement (Appendix D) summarizes the value of program animal presentations.

For the purpose of this policy, a program animal is described as an animal presented either within or outside of its normal exhibit or holding area that is intended to have regular proximity to or physical contact with trainers, handlers, the public, or will be part of an ongoing conservation education/outreach program.

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

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

AZA Accreditation Standard

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

At this time, no known birds from these families are used in animal programs. This is an area that needs further research.

9.2 Institutional Program Animal Plans

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

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

AZA 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.

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-accredited institutions that have designated program animals are required to develop their own Institutional Program Animal Policy that articulates and evaluates the program benefits (see Appendix E for recommendations). Program animals should be consistently maintained in a manner that meets their social, physical, behavioral, and nutritional needs. Education and conservation messaging must be an integral component of any program animal demonstration (AZA Accreditation Standard 1.5.3).

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

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

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

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

9.3 Program Evaluation

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

AZA Accreditation Standard

(10.3.3) All animal enclosures (exhibits, holding areas, hospital, and quarantine/isolation) must be of a size and complexity sufficient to provide for the animal's physical, social, and psychological well-being; and exhibit enclosures must include provisions for the behavioral enrichment of the animals. AZA housing guidelines outlined in the Animal Care Manuals should be followed.

AZA Accreditation Standard

(1.5.2) All animals must be housed in enclosures and in appropriate groupings which meet their physical, psychological, and social needs. Wherever possible and appropriate, animals should be provided the opportunity to choose among a variety of conditions within their environment. Display of single specimens should be avoided unless biologically correct for the species involved.

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 local, state, and federal laws must be adhered to. Planning and coordination for animal transport requires good communication among all involved parties, plans for a variety of emergencies and contingencies that may arise, and timely execution of the transport. At no time should the animal(s) or people be subjected to unnecessary risk or danger.

Chapter 10. Research

10.1 Known Methodologies

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

AZA Accreditation Standard

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

Many North American shorebird species are in steep decline. To reverse this trend and achieve stable populations, a basic knowledge of shorebird biology is needed. Only a few species have valid estimates of population size. Population limiting factors of most species are unknown. The main goal of the National Shorebird Research Program (NSRP) (shorebirdplan.fws.gov) is the maintenance of stable, self-sustaining populations. This is similar to the goals of the AZA population management groups. Partnerships between federal, state, and non-governmental organizations are encouraged to achieve the goals of stable shorebird populations. Zoos can assist by developing partnerships to help with shorebird population surveys, and to study the behavior and physiology of shorebirds in a zoo or aquarium setting. The NSRP stated one research topic is to increase productivity; this includes *ex situ* breeding and reintroduction programs.

The Charadriiformes TAG includes the shorebirds and alcids. Within the TAG there are three Green SSPs: the Inca tern and the Atlantic and tufted puffins; and four Yellow SSPs: the common murre, the black-necked stilt, the spotted dikkop, and the masked lapwing; and two Red SSPs: the African jacana and the spur-winged lapwing. Depending on population evaluations by the Population Management Center, the horned puffin and Peruvian thick-knee may become yellow SSPs.

The University of Minnesota and the University of Michigan Biological Station are both involved with shorebird research in the Great Lakes region. These two universities partner with the Detroit Zoo and the USFWS in the Great Lakes Piping Plover Project. Lake Superior State University and the Sustain Our Great Lakes Conservation Group also support the Great Lakes Piping Plover Project. Sustain Our Great Lakes is a bi-national, public-private partnership that sustains, restores and protects fish, wildlife, and habitat in the Great Lakes Basin by leveraging funding, building and conservation capacity and focusing partners and resources toward key ecological issues.

- The Detroit Zoo and the Detroit Water and Sewerage Department and USFWS have partnered in creating Common Tern habitat to draw nesting terns back to former nesting grounds on Belle Isle.
- The Monterey Bay Aquarium and the Point Blue Conservation Science combine efforts on the Snowy Plover Program. Since 2000 the Monterey Bay Aquarium has taken released over 100 birds back into the wild.

Scientists involved with Charadriiformes research:

- Dr. Francie Cuthbert of the University of Minnesota has been conducting shorebird research in the Great Lakes Area for many years.
- Dr. Terry Norton D.V.M. of the St. Catherine's Island Foundation has performed health assessments on wild American oystercatchers in Georgia and South Carolina. Dr. Norton is the vet advisor for wild Charadriiformes for the TAG.

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.

- Roche, Cuthbert, & Arnold (2009) performed an observational study on the relative fitness of wild and zoo-reared piping plovers to determine if egg salvage contributed to the recovery of the Great Lakes population. This group is investigating the effectiveness of the egg salvage program and this is directly tied to benefiting the wild population.
- The work on the American oystercatchers by Carson-Bremer et al. (2010) was a physiological study that gave mean values for hematology, plasma, biochemistry, hormone levels, and exposure to certain pathogens. This gave valuable baseline information on the American oystercatcher and will aid in future monitoring and conservation.
- 'Saunders, Ong, and Cuthbert (2013) performed a study investigating threat recognition of zoo/aquarium-reared piping plover chicks to determine if individuals adequately responded to predators. This group found that zoo/aquarium-reared chicks had innate recognition of avian threats, suggesting that individuals would be capable of responding to predators when released into the wild.'
- Neuman et al (2013) performed a study evaluating the success of zoo/aquarium-rearing for the Western Snowy Plover by comparing the survival and reproductive success of zoo/aquarium and wild-reared individuals.

At this time there is not much information on research testing paradigms for this TAG. Any research on these subjects would be useful.

Several articles have been published on the differences between zoo-reared and wild plovers, and how to further develop and refine techniques. One article compared the breeding of hand and wild-reared plovers in Page et al. (1989). Powell et al. (1997) published an article about *ex situ*-rearing piping plovers and developing techniques to augment wild populations. Other research has been done on the innate anti-predator behavior in *ex situ*-reared piping plovers, and the contributions that an egg salvage program makes in the recovery of the Great Lakes Piping Plover population (Saunders et al., 2013; Roche et al., 2009). These researchers are quantifying how *ex situ* rearing programs impact reintroduction programs.

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

AZA Accreditation Standard

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

AZA Accreditation Standard

(5.1) Research activities must be under the direction of a person qualified to make informed decisions regarding research.

Sample Research Policy:

RESEARCH OVERSIGHT POLICY
AQUARIUM A
I. INTRODUCTION
Aquarium A 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 its care and their habitats.
The Aquarium has developed this administrative oversight framework to ensure that research conducted under the auspices of the Aquarium is credible, defensible and meritorious. This framework will also:

- Encourage and facilitate research at the Aquarium, 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-Aquarium investigators requiring Aquarium assets and resources. It also covers research by Aquarium 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 Monterey Bay Aquarium "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 should:
- 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 Aquarium 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

The Aquarium's Research Committee is comprised of the following members:

- Director of Conservation Research (chair)
- Senior Coordinator of Special Conservation Research Projects
- Staff Veterinarian
- Vice President of Husbandry
- Husbandry Director
- 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 should submit a research proposal on the Aquarium's standardized form. Proposals are submitted to the Senior Coordinator, 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, or any committee member may request, 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). No external review will be sought by committee members without prior approval of the full committee. All communications with external reviewers is through the chair, Senior Coordinator, or designee. 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 Senior Coordinator 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. 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 should 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 may be required to present the status and findings of their research to Aquarium staff and volunteers in a lunchtime seminar or other venue.

The Piping Plover Project managed by the Detroit Zoo and the Snowy Plover Project managed by the Monterey Bay Aquarium are both priority conservation initiatives. The Detroit Zoo has contributed greatly to conservation goals through the Piping Plover Project and the Tern habitat restoration program. The Monterey Bay Aquarium has contributed to AZA conservation goals through the Snowy Plover program.

Monterey Bay Aquarium Snowy Plover Program: The same staff is used for exhibit care and rehab care. To decrease the chance of disease transmission, all exhibits are cleaned before taking care of the rehab birds. Once exhibits are cleaned, staff puts on the rehab gear, which includes coverall and jackets to cover clothes, gloves and use of footbath in and out of the rehab cages and rehab birds are fed. They are not cleaned until near the end of the day, after all exhibit work is done. All cleaning tools, dishes, etc. are kept separated from exhibit supplies.

Detroit Zoo Piping Plover Program: The program manager is the primary point person who is in contact with keepers in the field at the biological station on a daily basis (3–4 times a day via phone and email). This point person is sent egg weights and chick weights daily so they can guide the field staff through bouncing eggs back and forth in incubators or separating and feeding chicks differently for weight gains. They are provided paperwork and daily checklist in the field so they do have guidelines to follow. The rearing protocol is updated yearly, if needed. The program manager goes up to set up the entire facility (takes 3–4 days) and then sometimes that person is in charge of breaking down the facility. A lot of the equipment is stored over the winter in the biological station. A standard medical kit that their vets put together is left in the biological station all season. Accounts with vendors are set up so they can order food as needed. A seasonal cell phone for the zookeepers is provided that they carry at all times so that they are always available to pick up eggs. Portable field incubators are supplied for the field biologist. The program manager spends a lot of time in April preparing for the program, sets up in May, and starts receiving eggs in May/June. It runs until first week of August and then we break down. In the winter months, the program manager usually puts all of the data and paperwork together in a binder so we can use it the next year as a point of reference. The program manager estimates spending about one hour a day during the plover season consulting the keepers and a couple of weeks at the biological station.

Conservation Projects Supported by the TAG:
<u>The Great Lakes Piping Plover project Salvage Rearing Program</u>: The Great Lakes Piping Plover project Salvage Rearing Program is directed by Tom Schneider, the Bird Curator at the Detroit Zoo. In 1986 there were only 17 nesting pairs of the piping plover in the Great Lakes, and the United States Fish and Wildlife Service established a recovery program. Scientists found that some of the plovers were abandoning their eggs. By salvaging these eggs, a significant contribution could be made toward the species recovery. Because of the Detroit Zoo's expertise in bird care and incubation, an *ex situ* breeding program was coordinated to hatch out abandoned eggs and rear the chicks until they can be released to join the wild plovers. Eggs are brought to the rearing facility at The University of Michigan's Biological Station in Pellston, MI. At this time, the Detroit Zoo bird staff oversees the hatching and rearing of the chicks, and organizes the participation of bird care staff from zoos across the country. The chicks are reared in pens on the beach to protect them from predators as they acclimate to living in the wild. Piping plovers can fly when they are 4 weeks old, and at that time they are released at sites with adult plovers and their almost grown chicks. They quickly join these groups and migrate to the southern U.S. to spend the winter. The Great Lakes piping plover population is now about 60 breeding pairs. The *ex situ*-reared birds and their descendants have made a significant contribution to the population. The Detroit Zoo's

Piping Plover program was awarded the AZA 2009 Significant Achievement Award in North American Conservation.

This is a great program and is a good way for zoo staff to use their talents to make a contribution to the Great Lakes piping plover population. For further information about this program please contact Tom Schneider, Curator of Birds at the Detroit Zoo at tscheider@detroitzoo.org.

Figure 8. Piping plover
Photo courtesy of Roger Eriksson

Common Tern Nesting Habitat: Another project at the Detroit Zoo is the rehabilitation of the common tern nesting habitat on Belle Isle. During the 1960's, common terns nested by the thousands on islands in the Detroit River. Habitat loss, nesting disturbance, and competition from gulls have caused a significant population decline. At this time only two nesting colonies remain on the Detroit River, both at the Grosse Isle bridges.

The zoo has partnered with the Detroit Water and Sewage Department (DWSD) and the Detroit River International Wildlife Refuge to develop a tern nesting site on the east end of Belle Isle on a spit of land owned by DWSD. Common terns prefer to nest on areas with gravel substrate and a small amount of low vegetation. The zoo and other partners have removed tall trees and added gravel to the Belle Isle site and made it a much more suitable tern habitat. Tern decoys have been placed in the area and recorded tern calls are played on speakers in the nesting site. It is the zoo staff's hope that the terns will return to the area and build nests on Belle Isle for the first time in many years. For more information about this project please contact Tom Schneider, Curator of Birds at the Detroit Zoo at tschneider@detroitzoo.org. The website is www.detroitzoo.org.

Figure 9. Snowy plover
Photo courtesy of Monterey Bay Aquarium

Snowy Plover Recovery Program: In 2000, the Monterey Bay Aquarium began the Snowy Plover Recovery Program. This program was created to work alongside Point Blue Conservation Science (formerly PRBO) in an effort to help protect endangered snowy plovers. The work that PRBO does with snowy plovers ranges from determining population size in the wild, dispersal patterns, protecting nesting sites from people and predators, and developing management programs for the recovery of these birds. Over the years, the aquarium has received snowy plovers of all ages that are in need of rehabilitation, with ages ranging from eggs, chicks, and adults. These birds are coming from a variety of locations: PRBO, state park rangers and volunteers, local wildlife rehabilitators, and the general public. Once at the aquarium, aviculture staff cares for the birds with the hope that they can be released back into the wild.

Behind the scenes, staff feed, clean, and provide any necessary medical treatment to the rehabilitate the birds. After their stay at the aquarium—and staff determines that the birds are ready for release—staff from Point Blue are contacted. Point Blue staff will locate an optimal location for release with specific criteria (e.g., other snowy plovers in the area, low predation, and minimal human impact). The birds are banded in order for Point Blue to monitor each released individual and track their survival in the wild. For more information on the Snowy Plover Recovery Program please contact Aimee Greenebaum at the Monterey Bay Aquarium: agreenebaum@mbayaq.org.

10.2 Future Research Needs

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

There is a big shortage of knowledge about shorebirds and many knowledge gaps that need to be filled. People working with these birds in a zoo or aquarium setting can fill many of these gaps. Listed are but a few of the topics that need attention.

- Breeding behaviors: 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.
- Growth and development: This includes incubation period, pip-to-hatch times, weight gains and losses, parental behavior, fledging times, and any hand-rearing formulas, tips, and techniques.
- Training techniques: Scale training birds allows a no-stress method of obtaining a weight that adds data to the average weight range for a species.
- Population surveys: If possible, contact the local Audubon Society and offer assistance in counting birds. This helps give an idea of population size of birds in the wild.
- Average life span of wild and zoo-kept birds.
- For most species, a nutrient analysis of specific diets compared to target nutrient ranges
- 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).
- Any research about the behaviors of wild shorebirds or those kept in zoos and aquariums will add to the body of knowledge and help us better manage this group of birds.

Chapter 11. Other Considerations

11.1 Additional Information

Very few species regularly breed in managed settings. Most birds are acquired as non-releasable from rehab centers. It can be very challenging to find/acquire birds. It is highly recommended for each facility to have a good relationship with local rehab centers to help facilitate contact when they find a non-releasable animal. In addition, the following rehab centers have online placement needs and you can send request for animals wanted or look for animals available:

- International Wildlife Rehabilitation Council (IWRC) http://theiwrc.org/
- California Council for Wildlife Rehabilitators (CCWR) http://www.ccwr.org/index.html

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Personal Communications

Karen Anderson, Long Beach Aquarium of the Pacific, 2005
Mary Carlson, Seattle Aquarium, 2005
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Serge Pépin, Biodôme de Montréal, 2005

Cindy Pinger, Birmingham Zoo 2011
Kim Smith, Oregon Zoo, 2006
Tim Snyder, Brookfield Zoo, 2011

Appendix A: Accreditation Standards by Chapter

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

General Information

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

Chapter 1

(1.5.7) The animals must be protected from weather, and any adverse environmental conditions.

(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. All mechanical equipment must be kept in working order and should be under a preventative maintenance program as evidenced through a recordkeeping system. Special equipment should be maintained under a maintenance agreement, or a training record should show that staff members are trained for specified maintenance of special equipment.

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

Chapter 2

(1.5.1) Animals should be presented in a manner reflecting modern zoological practices in exhibit design, balancing animals' functional welfare requirements with aesthetic and educational considerations.

(1.5.2) All animals must be housed in enclosures and in appropriate groupings which meet their physical, psychological, and social needs. Wherever possible and appropriate, animals should be provided the opportunity to choose among a variety of conditions within their environment. Display of single animals should be avoided unless biologically correct for the species.

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

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

(11.3.6) In areas where the public is not intended to have contact with animals, some means of deterring public contact with animals (e.g., guardrails/barriers) must be in place.

(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.

(11.2.5) Live-action emergency drills must be conducted at least once annually for each of the four basic types of emergency (fire; weather/environment appropriate to the region; injury to staff or a visitor;

animal escape). Four separate drills are required. These drills must be recorded and evaluated to determine 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 documented whenever such are identified.

(11.6.2) Security personnel, whether staff of the institution, or a provided and/or contracted service, must be trained to handle all emergencies in full accordance with the policies and procedures of the institution. In some cases, it is recognized that Security personnel may be in charge of the respective emergency (i.e. shooting teams).

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

(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 (e.g. large carnivores, large reptiles, medium to large primates, large hoofstock, killer whales, sharks, venomous animals, and others, etc.) must have appropriate safety procedures in place to prevent attacks and injuries by these animals. Appropriate response procedures must also be in place to deal with an attack resulting in an injury. These procedures must be practiced routinely per the emergency drill requirements contained in these standards. Whenever injuries result from these incidents, a written account outlining the cause of the incident, how the injury was handled, and a description of any resulting changes to either the safety procedures or the physical facility must be prepared and maintained for five years from the date of the incident.

(11.5.2) All areas housing venomous animals, or animals which pose a serious threat of catastrophic injury and/or death (e.g. large carnivores, large reptiles, medium to large primates, large hoofstock, killer whales, sharks, venomous animals, and others, etc.) must be equipped with appropriate alarm systems, and/or have protocols and procedures in place which will notify 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 must be conducted to insure that appropriate staff members are notified.

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

Chapter 3

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

(1.5.10) Temporary, seasonal and traveling live animal exhibits (regardless of ownership or contractual arrangements) must meet the same accreditation standards as the institution's permanent resident animals.

Chapter 5

(2.6.2) The institution should have 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 preparations must meet all applicable laws and regulations.

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

Chapter 6

(2.1.1) A full-time staff veterinarian is recommended. In cases where such is not practical, 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 animal collection 24 hours a day, 7 days a week.

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

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

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

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

(1.4.4) Animal records, whether in electronic or paper form, including health records, must be duplicated and stored in a separate location.

(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.

(1.4.1) An animal inventory must be compiled at least once a year and include data regarding acquisitions and dispositions at the institution.

(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. In both cases, notations should be made on the inventory.

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

(2.7.3) Quarantine, hospital, and isolation areas should be in compliance with standards/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/associations/6442/files/veterinary_standards_2009_final.docx.

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

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

(11.1.3) A tuberculin (TB) testing/surveillance program must be established for appropriate staff in order to ensure the health of both the employees and the animals. Each institution must have an employee occupational health and safety program.

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

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

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

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

(2.4.2) Keepers should be trained to recognize abnormal behavior and clinical 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, keepers should not diagnose illnesses nor prescribe treatment.

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

(1.5.8) The institution must develop a clear process for identifying, communicating, and addressing animal welfare concerns within the institution in a timely manner, and without retribution.

Chapter 8

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

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

Chapter 9

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

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

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

(10.3.3) All animal enclosures (exhibits, holding areas, hospital, and quarantine/isolation) must be of a size and complexity sufficient to provide for the animal's physical, social, and psychological well-being; and exhibit enclosures must include provisions for the behavioral enrichment of the animals. AZA housing guidelines outlined in the Animal Care Manuals should b e followed.

(1.5.2) All animals must be housed in enclosures and in appropriate groupings which meet their physical, psychological, and social needs. Wherever possible and appropriate, animals should be provided the opportunity to choose among a variety of conditions within their environment. Display of single animals should be avoided unless biologically correct for the species.

(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. Planning and coordination for animal transport requires good communication among all involved parties, plans for a variety of emergencies and contingencies that may arise, and timely execution of the transport. At no time should the animal(s) or people be subjected to unnecessary risk or danger.

Chapter 10

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

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

(5.1) Research activities must be under the direction of a person qualified to make informed decisions regarding research.

Appendix B: Acquisition/Disposition Policy

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

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

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

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

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

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

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

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

III. Germplasm: Acquisition and disposition of germplasm should follow the same guidelines outlined in this document if its intended use is to create live animal(s). Ownership of germplasm and any resulting animals should be clearly defined. Institutions acquiring or dispositioning germplasm or any animal parts or samples should consider not only its current use, but also future possible uses as new technologies become available.

IV(a). General Acquisitions: Animals are to be acquisitioned into an AZA member institution's collection if the following conditions are met:

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

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

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

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

An accredited AZA member balances public display, recreation, and entertainment with demonstrated efforts in education, conservation, and science. While some non-AZA member organizations may meet minimum daily standards of animal care for wildlife, the AZA recognizes that this, by itself, is insufficient to warrant either AZA membership or participation in AZA's cooperative animal management programs.

When an animal is sent to a non-member of AZA, it is imperative that the member be confident that the animal will be cared for properly.

Animals may only be disposed of from an AZA member institution's collection if the following conditions are met:

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

14. In dispositions to non-AZA members, the AZA members must be convinced that the recipient has the expertise, records management practices, financial stability, facilities, and resources required to properly care for and maintain the animals and their offspring. It is recommended that this documentation be kept in the permanent record of the animals at the AZA member institution.

15. If living animals are sent to a non-AZA member research institution, the institution must be registered under the Animal Welfare Act by the U.S. Department of Agriculture Animal and Plant Health Inspection Service. For international transactions, the receiving facility should be registered by that country's equivalent body with enforcement over animal welfare.

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

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

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

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

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

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

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

1. Dispositions of dead specimens must meet the requirements of all applicable local, state, federal and international regulations and laws.

2. Maximum utilization is to be made of the remains, which could include use in educational programs or exhibits.

3. Consideration is given to scientific projects that provide data for species management and/or conservation.

4. Records (including ownership information) are to be kept on all dispositions, including animal body parts, when possible.

5. SSP and TAG necropsy protocols are to be accommodated insofar as possible.

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

Appendix C: Recommended Quarantine Procedures

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

Such separation should be obligatory for primates, small mammals, birds, and reptiles, and attempted wherever possible with larger mammals such as large ungulates and carnivores, marine mammals, and cetaceans. If the receiving institution lacks appropriate facilities for isolation of large primates, pre-shipment quarantine at an AZA or American Association for Laboratory Animal Science (AALAS) accredited institution may be applied to the receiving 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 Charadriiformes:

Charadriiformes:

Required:

1. Direct and floatation fecals
2. Vaccinate as appropriate

Strongly recommended:

1. CBC/sera profile
2. Urinalysis
3. Appropriate serology (FIP, FeLV, FIV)
4. Heartworm testing in appropriate species

Appendix D: Program Animal Policy and Position Statement

Program Animal Policy

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

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

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

There are three main categories of Program Animal interactions:

1. On Grounds with the Program Animal Inside the Exhibit/Enclosure:
    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 Program Animal Outside the Exhibit/Enclosure:
    a. Minimal handling and training techniques are used to present Program Animals to the public. Public has minimal or no opportunity to directly interact with Program Animals when they are outside the exhibit/enclosure (e.g., raptors on the glove, reptiles held "presentation style").
    b. Moderate handling and training techniques are used to present Program Animals to the public. Public may be in close proximity to, or have direct contact with, Program Animals when they're outside the exhibit/enclosure (e.g., media, fund raising, photo, and/or touch opportunities).
    c. Significant handling and training techniques are used to present Program Animals to the public. Public may have direct contact with Program Animals or simply observe the in-depth presentations when they're outside the exhibit/enclosure (e.g., wildlife education shows).
3. Off Grounds:
    a. Handling and training techniques are used to present Program Animals to the public outside of the zoo/aquarium grounds. Public may have minimal contact or be in close proximity to and have direct contact with Program Animals (e.g., animals transported to schools, media, fund raising events).

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

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

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

Program Animal Position Statement

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

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

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

Program animals are powerful catalysts for learning for a variety of reasons. They are generally active, easily viewed, and usually presented in close proximity to the public. These factors have proven to contribute to increasing the length of time that people spend watching animals in zoo exhibits (Bitgood, Patterson & 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 program animals in shows or informal presentations can be effective in lengthening the potential time period for learning and overall impact.

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

Knowledge Acquisition
Improving our visitors' knowledge and understanding regarding wildlife and wildlife conservation is a fundamental goal for many zoo educators using program animals. A growing body of evidence supports the validity of using program animals to enhance delivery of these cognitive messages as well.

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

Enhanced Environmental Attitudes

Program animals have been clearly demonstrated to increase affective learning and attitudinal change.

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

Conclusion

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

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Appendix E: Developing an Institutional Program Animal Policy

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Rationale

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

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

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

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

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

The Policy Development Process

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

- The Education Department
- The Animal Husbandry Department
- The Veterinary and Animal Health Department
- The Conservation & Science Department
- The Behavioral Husbandry Department
- Any animal show staff (if in a separate department)
- Departments that frequently request special program animal situations (e.g., special events, development, marketing, zoo or aquarium society, administration)

Additionally, staff from all levels of the organization should be involved in this development (e.g., curators, keepers, education managers, interpreters, volunteer coordinators).

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

I. Philosophy

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

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

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

II. Appropriate Settings

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

- I. On-site programming
    - a. Informal and non-registrants:
        - 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 Program 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. Program animals are part of an institution's overall collection and must be included in the overall collection planning process. The AZA Guide to Accreditation contains specific requirements for the institution collection plan. For more information about collection planning in general, please see the Collection Management pages in the Members Only section.

The following recommendations apply to program animals:

1. Listing of approved program animals (to be periodically amended as collection changes). Justification of each species should be based upon criteria such as:
    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 Program Animal Policy should address the specific messages related to the use of program animals, as well as the need to be cautious about hidden or conflicting messages (e.g., "petting" an animal while stating verbally that it makes a poor pet). This section may include or reference the AZA Conservation Messages.

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

VI. Human Health and Safety

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

1. Minimization of the possibility of disease transfer from non-human animals to humans, and vice-versa (e.g., 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 Program Animal Policy should make a strong statement on the importance of animal welfare. The policy should address:

1. General housing, husbandry, and animal health concerns (e.g. that the housing and husbandry for program animals meets or exceeds general AZA standards and that the physical, social and psychological needs of the individual animal, such as adequate rest periods, provision of enrichment, visual cover, contact with conspecifics as appropriate, etc., are accommodated).

2. Where ever possible provide a choice for animal program participation, e.g., retreat areas for touch tanks or contact yards, evaluation of willingness/readiness to participate by handler, etc.)

3. The empowerment of handlers to make decisions related to animal health and welfare; such as withdrawing animals from a situation if safety or health is in danger of being compromised.
4. Requirements for supervision of contact areas and touch tanks by trained staff and volunteers.
5. Frequent evaluation of human / animal interactions to assess safety, health, welfare, etc.
6. Ensure that the level of health care for the program animals is consistent with that of other animals in the collection.
7. Whenever possible have a "cradle to grave" plan for each program animal to ensure that the animal can be taken care of properly when not used as a program animal anymore.
8. If lengthy "down" times in program animal use occur, staff should ensure that animals accustomed to regular human interactions can still maintain such contact and receive the same level of care when not used in programs.

VIII. Taxon Specific Protocols

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

Taxon and species -specific protocols should address:

1. How to remove the individual animal from and return it to its permanent enclosure, including suggestions for operant conditioning training.
2. How to crate and transport animals.
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 program animals, including:

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

X. Staff Training

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

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

1. Personnel authorized to handle and present animals.
2. Handling protocol during quarantine.
3. The process for training, qualifying and assessing handlers including who is authorized to train handlers.
4. The frequency of required re-training sessions for handlers.

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

XI. Review of Institutional Policies

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

XII. TAG and SSP Recommendations

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

Appendix F: Reference Values for Hematological and Serum Biochemistry Parameters of Selected Species

Parameter (mean ± SD)	**American avocet (*Recurvirostra americana*) (*N*)**	**Black-winged stilt (*Himantopus himantopus*) (*N*)**	**Cape thick-knee (*Burhinus capensis*) (*N*)**	**Inca tern (*Larosterna inca*) (*N*)**	**Masked lapwing (*Vanellus miles*) (*N*)**
Erythrocytes ($x10^6/\mu L$)	3.48±0.77 (8)	3.45±0.39 (16)	2.06±0.00 (1)	3.60±0.55 (31)	2.73±0.50 (12)
Leukocytes ($x10^3/\mu L$)	8.26±4.82 (8)	7.78±3.27 (40)	13.35±5.93 (38)	11.39±6.87 (245)	8.41±4.43 (22)
Hemoglobin (g/dL)	13.4±1.2 (3)	15.0±3.4 (11)	13.3±0.9 (13)	15.7±1.4 (41)	26.4±5.2 (6)
Hematocrit (%)	46.2±5.0 (10)	50.3±6.3 (47)	46.0±6.0 (30)	50.0±4.6 (255)	47.8±4.0 (21)
MCH (pg)	43.2±4.0 (3)	45.2±7.2 (8)			94.8±9.8 (6)
MCHC (g/dL)	32.6±4.7 (3)	29.7±3.9 (11)	28.6±1.2 (13)	30.9±2.8 (40)	49.2±5.7 (5)
MCV (fl)	135.3±25.7 (8)	148.8±20.7 (16)	174.8±0.0 (1)	136.7±19.6 (30)	182.9±24.2 (11)
Heterophils ($x10^3/\mu L$)	5.54±3.70 (8)	3.47±2.54 (39)	6.22±4.03 (38)	4.46±3.17 (244)	3.05±1.62 (22)
Lymphocytes ($x10^3/\mu L$)	2.57±1.52 (8)	3.84±2.22 (39)	4.52±3.34 (38)	5.63±5.06 (244)	4.38±2.49 (22)
Monocytes ($x10^3/\mu L$)	0.15±0.00 (4)	0.24±0.18 (23)	0.76±0.53 (35)	0.61±0.66 (222)	0.19±0.16 (14)
Eosinophils ($x10^3/\mu L$)	0.09±0.05 (3)	0.41±0.33 (33)	1.55±1.31 (37)	0.27±0.30 (116)	0.78±0.96 (19)
Basophils ($x10^3/\mu L$)	0.11±0.06 (3)	0.21±0.15 (22)	0.50±0.34 (32)	0.76±0.57 (206)	0.38±0.40 (10)

MCH, mean corpuscular hemoglobin; MCHC, mean corpuscular hemoglobin concentration; MCV, mean corpuscular volume. (ISIS, 1999)

Parameter	**American avocet (*Recurvirostra americana*) (*N*)**	**Black-winged stilt (*Himantopus himantopus*) (*N*)**	**Cape thick-knee (*Burhinus capensis*) (*N*)**	**Inca tern (*Larosterna inca*) (*N*)**	**Masked lapwing (*Vanellus miles*) (*N*)**
Glucose (mg/dL)	266±58 (9)	275±57 (26)	295±79 (35)	324±40 (170)	286±28 (9)
Blood Urea Nitrogen (mg/dL)	10±8 (2)	3±1 (5)	3±1 (20)	3±2 (61)	5±2 (2)
Creatinine (mg/dL)	0.4±0.0 (4)	0.2±0.0 (1)	0.4±0.2 (25)	0.5±0.3 (69)	0.4±0.0 (1)
Uric Acid (mg/dL)	7.4±4.4 (9)	7.0±4.9 (28)	6.9±4.6 (35)	8.5±6.4 (175)	8.8±3.3 (9)
Calcium (mg/dL)	8.7±0.6 (8)	8.9±0.9 (23)	9.4±1.1 (31)	9.5±1.0 (107)	9.1±0.8 (9)
Phosphorous (mg/dL)	2.5±0.4 (2)	2.3±2.3 (14)	3.8±1.7 (22)	2.8±2.2 (90)	1.3±0.6 (5)
Sodium (mEq/L)	155±4 (6)	147±6 (3)	153±4 (23)	148±4 (81)	154±4 (5)
Potassium (mEq/L)	3.2±2.0 (6)	2.8±0.0 (1)	1.9±0.8 (23)	3.3±2.0 (82)	3.6±1.3 (5)
Chloride (mEq/L)	115±16 (4)	113±6 (3)	118±5 (18)	117±5 (78)	121±2 (5)
Cholesterol (mg/dL)	260±41 (4)	200±72 (9)	292±77 (31)	312±67 (74)	178±35 (7)
Triglycerides (mg/dL)	134±0 (1)	101±26 (4)	122±55 (8)	47±12 (38)	309±194 (2)

Total Protein (g/dL)	3.0±0.5 (9)	3.6±0.6 (23)	3.6±0.6 (31)	3.7±0.7 (181)	3.5±0.5 (10)
Albumin (g/dL)	1.2±0.3 (4)	2.2±0.5 (5)	1.4±0.3 (22)	1.5±0.5 (77)	
Globulin (g/dL)	1.8±0.5 (4)	1.3±0.6 (5)	2.2±0.6 (22)	2.0±0.5 (77)	
AST (IU/L)	225±78 (8)	206±107 (23)	490±549 (33)	276±123 (136)	274±84 (8)
ALT (IU/L)	36±13 (3)	42±8 (5)	62±20 (20)	31±21 (83)	99±48 (2)
Total bilirubin (mg/dL)	0.4±0.2 (2)	0.2±0.0 (5)	0.5±0.8 (17)	0.6±0.3 (81)	0.2±0.1 (2)
Amylase (U/L)	302±47 (2)	827±96 (4)			
ALP (IU/L)	136±52 (2)	249±134 (8)	81±94 (23)	183±106 (78)	90±60 (7)
LDH (IU/L)	786±409 (5)	504±183 (4)	900±559 (18)	399±189 (47)	2141±949 (6)
CPK (IU/L)	777±1033 (5)	252±155 (13)	1116±1664 (20)	326±290 (110)	1101±568 (6)

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

Appendix G: Sample Hand Rearing Protocol

A) Example of Snowy Plover and Killdeer Hand Rearing Protocol from an accredited AZA institution (for release into the wild):

*This protocol is a general guide line. Each chick will be evaluated by overall behavior and weight gain. The protocol may be adjusted to ensure their well-being.

Incubation

Egg Age: 1–26 days
- Temperature: 37.7 °C (100 °F)
- Humidity: 40–50% (25.5–28 °C [77.9–82.4 °F])
- Turning Rate: 180 degrees every 2 hours
- Weigh eggs when received and then every 3 days

Egg age: 26 days +
- Temperature: 37.7 °C (100 °F)
- Humidity: 65% (31 °C [87.8 °F])
- Do not turn eggs
- Play recorded chirps,

Hatching

*Turner should be turned off 2–3 days before hatch date

Signs of Hatching
- Fine cracks appear at the large end of the egg up to 4 days before hatching (Difficult to see without magnification)
- Audile tapping 2 days before hatching
- Audible peeping 1–2 days before hatching
- Encourage chicks to hatch by pepping back to them/play recording

Hatching
- Eggs can hatch day or night
- Very distinct hole usually visible about 20 min before hatching
- Chicks usually hatch within 1 hour of initial pip, but can take up to 24–48 hrs.
- Move chick to I.C.U. or brooder once the chick is mostly dry and is walking around, usually within 3 hours of hatching.
- Unabsorbed yolk sac is usually absorbed within a few hours of hatching
- Weigh chick(s) and place in ICU when ready. If more than one chick, either band or mark the chicks with marker on head or toe for ID.

Hatching Problems
- May need assistance with hatching if there is no progress or activity 24 hours after initial pip. Monitor chick closely for signs of progress or weakness/ inactivity.
- If chick is showing signs of needing assistance, contact the veterinarian or an experienced aviculturist to aid the chick.
- It is important to keep the humidity as high possible during hatching. Try to avoid unnecessarily opening the lid.
- Lethargic chicks may need 50% dextrose or Ensure and rehydrating fluids (Pedialyte, LRS) orally

Intensive Care Unit (ICU)

Measurements 45.7 cm x 61 cm x 45.7 cm (18 in. x 24 in. x 18 in.)
Minimum weight: 4 g
Minimum age: 1 hour
Behavior: Mostly dry, walking around good, eyes open, yolk sac absorbed

Set-up
- Make sure ICU is completely disinfected
- Fill Water tray half full with distilled water

- Temperature should be set at 37.2–37.8 °C (99–100 °F)
- Place pillowcases in ICU 2.5–5 cm (1–2 in.) thick
- Place a feather duster and a stuffed animal in ICU, make sure the stuffed animal is placed in a way that the chick can get all the way around it with ease
- If a single chick, add a mirror
- Weigh chick before placing in ICU
- If there are multiple chicks, place a temporary band on leg for identification or using marker, mark on head or toe
- Add a full spectrum light above the holding, making sure timer is set to current light cycle
- Place towel over outside of the door to minimize human impact, make sure vents on the side are not covered

Diet

- Fly larvae, small mealworms (if available) and black worms
- Use small shallow dishes ¾ of food and scatter ¼ of food around of towel
- Sprinkle calcium powder on all food
- Add shallow water dish

Cleaning and maintenance

- Weigh chick(s) daily in the morning
- Change pillowcase daily (coordinate with chick weighing so chicks are contained during cleaning)
- Change stuffed animal as needed
- Feed 3–4 times daily or as needed. If the chick isn't eating, you may need to put some black worms directly in front of the chick on the sheet and/or gently place its beak in the food so it gets the idea.
- Decrease temperature 1–2 degrees daily, depending on chicks behavior

Indoor Tank

Measurements: 58.4 cm x 162.5 cm x 50.8 cm (23 in. x 64 in. x 20 in.)
Snowy plover minimum weight: 6 g
Killdeer minimum weight: 9 g
Minimum age: 2 days
Behavior: Eating, gaining weight, walking around good

Set-up

- Sand 5.08 cm (2 in.) thick
- 1–2 Full spectrum lights, on timer, above tank
- Heat lamp(s) with one spot 35 °C (95 °F).
- Feather duster, if without adult
- Mirror, if by themselves
- Driftwood and fake plant(s) placed in it

Diet

- Fly larvae, black worms, mini-mealworms, regular mealworms, chopped krill (PM only), and small crickets.
- Scatter 1/4 of dry food around tank. Avoid placing food/water directly under the heat lamp(s).
- Make sure dishes are shallow enough that the chicks can stand in them and easily get out of them
- Sprinkle calcium and Nekton vitamin on food
- Shallow water dish

Cleaning and maintenance

- Weight chick(s) daily
- Sift sand daily to remove debris (coordinate with weighing so chicks are contained during cleaning)
- Scrub Driftwood as needed
- Feed three times per day or as needed
- Record everything on the Avian Rehabilitation Notes Sheet

Outdoor flight cages (Phase I and II)
Measurements 1.67 m x 5.48 m x 3.04 m (5.5 ft x 18 ft x 10 ft)

Phase I
Snowy plover minimum weight: 13 g
Killdeer minimum weight: 20 g
Minimum age: 10 days

Set-up
- Place clean #30 sand in at least 5.08 cm (2 in.) thick
- Wooden shelter
- Secure heat lamp(s) so one spot is 35 °C (95 °F).
- Monitor temperature with the thermostat
- Hang a feather duster on shelter if chick is without adult
- Add artificial or real plants in the tank
- Place driftwood throughout tank
- If a single chick, add a mirror
- Fill pool with freshwater about 2.54–5.08 cm (1–2 in.) (so chicks can still stand in it)
- Add shallow wading dish for saltwater

Diet
- Fly larvae, black worms, mini-mealworms, regular mealworms, waxworms, small-medium sized crickets, and krill (PM only)
- Sprinkle calcium and Nekton vitamin on food.
- Place 1/4 of dry food in a dish; scatter 3/4 of dry food on sand. Avoid placing food/water directly under the heat lamps.
- Shallow freshwater dish for drinking

Cleaning and maintenance
- All water gets changed/cleaned daily
- Weigh chick(s) twice a week
- Sift sand twice per week to remove debris (coordinate with weighing so chicks are contained during cleaning)
- Scrub driftwood twice a week
- Feed two times per day or as needed

Phase II
Snowy plover minimum weight: 25 g
Killdeer minimum weight 50 g
Minimum age: 25 days

Set-up
- Remove heat lamp
- Remove feather duster and stuffed animal
- Fill pool completely full with freshwater
- Remove wooden shelter
- The rest stays the same

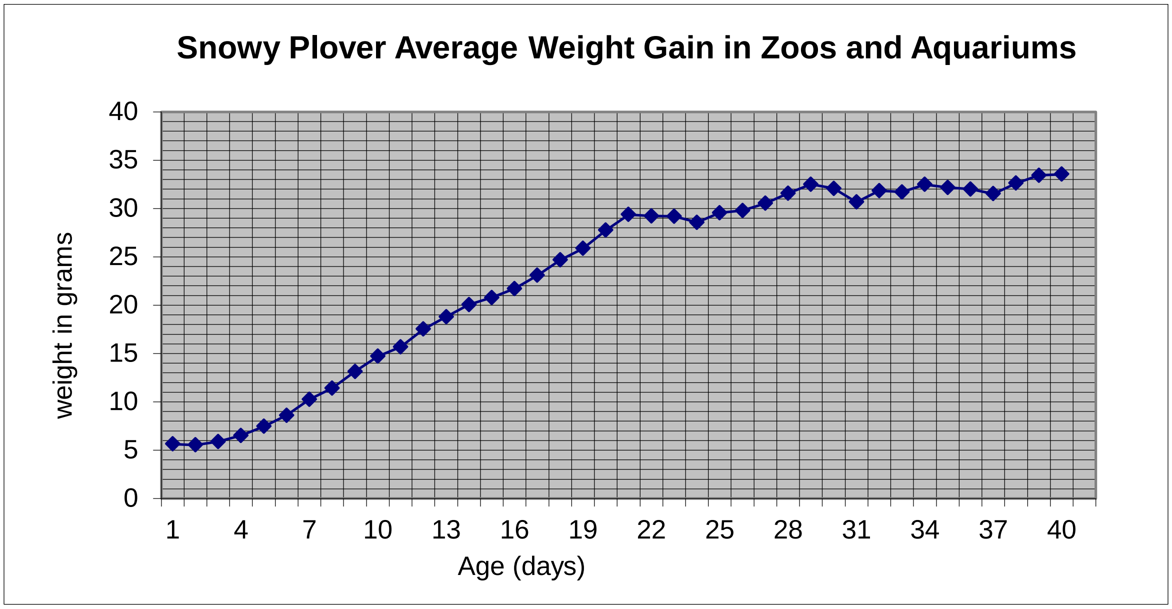
Diet
- Scatter all dry food in sand, only wet food in dish
- Other components remain the same

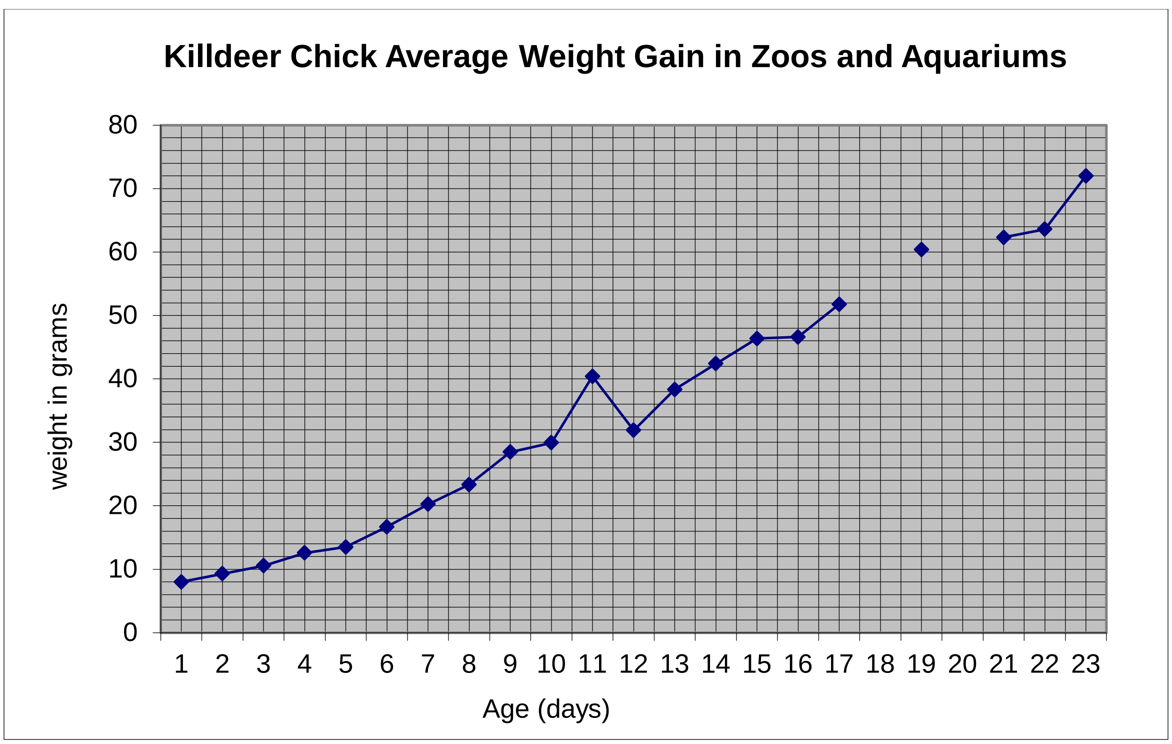
Cleaning and maintenance
- Remains the same.

Release criteria
Snowy plover minimum weight: 30 g
Killdeer minimum weight 65 g
Minimum age: 35 days

Behavior:

- Excellent health
- Excellent flyer
- Waterproof
- Appropriate wariness of humans
- Appropriate foraging behavior





B) **Example of Spotted Dikkop Chick Feeding Protocol**

*This protocol is a general guide line. This protocol is based on the diet that an AZA-accredited facility feeds their spotted dikkop chicks.

Feeding times:
For the first 1-3 days we will aim for 7 feedings (7:00am, 9:00am, 11:00am, 1:00pm, 3:00pm, 5:00pm, 8:00pm). We can start decreasing the feedings once we confirm that the chick is eating well and gaining sufficient weight (around 10%/day).

Food items:
BOP, apple paradise, hard-boiled egg yolk, wax worms/mealworms, pinkies.

- BOP and egg should be discarded after 24 hours.
- Apple paradise should be soaked in the AM before feeding. Discard at the end of the day.
- Only feed pinky viscera for the first 3 days. We will determine if the chick is ready for pieces containing bone/cartilage after that. Throw away uneaten pinky pieces after each feeding.
- We will start introducing mealworms after day 3

Feeding:

- Warm the pinky/pinky viscera before feeding
- Weigh out each food item before feeding (except worms, they can be counted)
- Try to give equal parts of all food items (it doesn't have to be exact)
- At each feeding, dip the first food item you feed the chick into calcium. No more calcium is needed at that feed.
- BOP should be fed in pea size pieces. Avoid feeding "stringy" or "chewy" pieces.
- Feed until the chick appears full/has stopped begging.
- Weight leftover food items and record amount eaten in laptop log.

Be sure to communicate with each other if there are several people feeding during the course of the day.

Appendix H: Sample Feeding Protocol

Sample feeding protocol (Galliforme)

		Ingredient	Form fed	Quantity offered by Volume	Quantity offered by weight (g)	Frequency offered
Common name	American avocet	Galliforme mix		1/4 cup	36 g	AM/PM
Scientific name	*Recurvirostra americana*	Bird of prey	Very small pieces		10 g	AM/PM
		Regular mealworms	Whole	10	0.7 g	AM/PM

Galliforme diets:
Ground dwelling birds such as turkeys, guineafowl, pheasants, curassows, tinamou, etc. receive a galliforme mix.

Galliforme mix*:* 2 parts: Mazzuri® Exotic Gamebird pellets
1 part: Scenic® Tropical Bits pellets
1 part: soaked Mazuri® parrot pellets
1/2 part: chopped hardboiled egg
1 part: chopped romaine lettuce
1 part: passerine fruit: apples (finely chopped) banana, steamed yam, melon, papaya

Birds may be offered insects during the day and offered a diet in the PM or be offered a diet AM and PM.

It is accepted that the weight of individually portioned amounts (tablespoons, cups, etc.) can vary due to the mix itself and the preparer of the diet. Therefore the following weights will be used when assessing diets that use the Galliforme mix:

1 Tablespoon: 9 g
1/4 cup: 36 g
1/2cup: 72 g
1 cup: 144 g

The pellets offered will be rotated on the schedule below:
March 1st – Sept. 1st : Mazuri® Exotic Gamebird Breeder
Sept 1st – March 1st : Mazuri® Exotic Gamebird Maintenance

Appendix I: Sample Enrichment Forms

Written Enrichment Plans-Samples

Enrichment Request Form

Animal: ______________________ Requested By: ________________________________

Enrichment:

Food ☐ Environmental ☐ Social ☐ Manipulative ☐ Sensory ☐

Pre-Open ☐ Open-Sign ☐ Open-No Sign ☐ Overnight ☐

Description:
Duration ____________________________________
Comments:
Approved: ☐

NOT Approved**:** ☐

Re-submit with these modifications for possible approval: ☐

Comments:
Associate Curator Signature____ Date: ____________________
Enrichment Coordinator Signature: ___________________________________ Date: ____________________
Veterinarian Signature:____ Date: ____________________
Exhibits Signature:____ Date:____________________

Sample enrichment evaluation form

Species:
Enrichment:

Date	**Response**	**Comments**

Response evaluation:
1= animal flees from item
2=animal ignores item
3= animal orients/looks at item but no contact made
4= animal makes brief contact with item
5=animal makes repeated or substantial contact with item
6=no direct contact seen but evidence suggest they did interact with item (item moved or feces around item, etc.)

Appendix J: Sample Research Policy Form

Research oversight policy

I. Introduction

We are 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 its care and their habitats. The Aquarium has developed this administrative oversight framework to ensure that research conducted under the auspices of the Aquarium is credible, defensible and meritorious. This framework will also:

- Encourage and facilitate research at the Aquarium, 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-Aquarium investigators requiring Aquarium assets and resources. It also covers research by Aquarium 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 our “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 Aquarium 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

The Aquarium’s Research Committee is comprised of the following members:

- Director of Conservation Research (chair)
- Senior Coordinator of Special Conservation Research Projects
- Staff Veterinarian
- Vice President of Husbandry
- Husbandry Director
- 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 Senior Coordinator, 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, or any committee member may request, 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). No external review will be sought by committee members without prior approval of the full committee. All communications with external reviewers is through the chair, Senior Coordinator, or designee. 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 Senior Coordinator 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. 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 should 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 may be required to present the status and findings of their research to Aquarium staff and volunteers in a lunchtime seminar or other venue.

Appendix K: Sample AZA-accredited Intuitional 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* spp., as these organisms cause respiratory infections in birds but 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)
- Stop watch
- Pen and Sharpie
- Paper
- Gloves
- Small Ziploc bags

Procedures:
<u>Sampling Procedure</u>

1) Choose an appropriate location. See below for the usual sampling spots.
2) Connect the tubing from the pump to the sampler.
3) Plug in the sampler and turn it on.
4) Adjust the airflow 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 it is suggested to send 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 25 °C (77 °F)(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)

- 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.