ASSOCIATION OF ZOOS & AQUARIUMS

BUSTARD (*Otididae*) CARE MANUAL

2nd Edition

CREATED BY THE

AZA Kori Bustard & Buff-crested Bustard Species Survival Plan®

IN ASSOCIATION WITH THE

AZA Gruiformes Taxon Advisory Group

Bustard Care Manual

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Cover Photo Credit:

Left, kori bustard adult male: Photo by Jessie Cohen
Right, buff-crested bustard adult male: Photo by Cathy Burkey

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Introduction

Preamble

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

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

Taxonomic Classification

Table 1. Taxonomic classification for kori bustards and buff-crested bustards

Classification	Taxonomy
Kingdom	Animalia
Phylum	Chordata
Class	Aves
Order	Gruiformes
Family	Otididae

Genus, Species, and Status

Table 2. Genus, species, and status information for bustards housed in the United States

Genus	Species	Common Name	USA Status	IUCN Status	CITES status	AZA Status
Ardeotis	*kori struthiunculus (eastern Africa)* *kori kori (southern Africa)*	Kori bustard	Not Listed	Near Threatened	Appendix II	Yellow SSP
Lophotis	*gindiana*	Buff-crested bustard	Not listed	Least Concern	Appendix II	Red SSP

General Information

The information contained within this Animal Care Manual (ACM) provides a compilation of animal care and management knowledge that has been gained from recognized species experts, including AZA Taxon Advisory Groups (TAGs), Species Survival Plan® Programs (SSPs), biologists, veterinarians, nutritionists, reproduction physiologists, behaviorists, and researchers. This ACM is based on the most current science, practices, and technologies used in animal care and management and is a valuable resource that enhances animal welfare by providing information about the basic requirements needed and best practices known for caring for *ex situ* kori bustard and buff-crested bustard populations. This ACM is considered a living document that is updated as new information becomes available and at a minimum of every five years. This manual was adapted from the *Kori Bustard (Ardeotis kori) Care Manual* (2009) and now includes information on buff-crested bustards. While some AZA-accredited zoos still manage white-bellied bustards (*Eupodotis senegalensis*), the Gruiformes TAG has recommended that these programs be phased out. Husbandry of white-bellied bustards is similar to buff-crested bustards. Some aspects of caring for bustards are the same across taxa. When this is not the case, sub-sections for each species have been included with relevant information.

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

AZA Accreditation Standard

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

sidebar boxes. AZA-accredited institutions which care for bustards must comply with all relevant local, state/provincial, and federal wildlife laws and/or regulations; AZA accreditation standards that are more stringent than these laws and/or regulations must be met (AZA Accreditation Standard 1.1.1).

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

Kori bustard:

Natural history: Kori bustards are large, terrestrial, omnivorous birds that inhabit open plains and semi-desert areas within their natural habitats. Kori bustards are indigenous to the grasslands and lightly wooded savannas of southern and eastern Africa (Johnsgard, 1991). In eastern Africa, *A. k. struthiunculus* can be found in Ethiopia, Uganda, Sudan, Kenya, and Tanzania (del Hoyo, Elliot, & Saragatal, 1996). *A. k. kori* can be found in Botswana, Zimbabwe, Namibia, southern Angola, South Africa, and Mozambique (Johnsgard, 1991). Natural predators of kori bustards in these environments include the black-backed jackal (*Canis mesomelas*), spotted hyena (*Crocuta crocuta*), martial eagle (*Polemaetus bellicosus*), tawny eagle (*Aquila rapax*), Verreaux's eagle (*Aquila verreauxii*), leopard (*Panthera pardus*), lion (*Panthera leo*), and caracal (*Caracal caracal*) (Hallager & Boylan, 2004).

Kori bustards are omnivorous, and have been observed consuming flowers, leaves, seeds, fruits, pods, acacia gum, insect prey (e.g., Hymenoptera, Orthoptera, Coleoptera, Lepidoptera), and non-insect prey (e.g., Chilopoda, Diplopoda, Annelida, Reptilia) (Mwangi, 1988; Osborne, 1998). When drinking from water sources, kori bustards use an unusual sucking action (Hallager, 1994), which may be an adaptation to arid climates where water can be limited (Hallager & Boylan, 2004).

The daily activity of wild kori bustards involves periods of activity in the morning and evening (0900h and 1700h), with birds awaking 30 minutes before sunrise to begin feeding, and periods of rest during the heat of the day (1130h to 1530h) (Mwangi & Karanja, 1993; Osborne, 1998). Preening and dustbathing were observed most frequently during the middle of the day (Mwangi, 1988). During the breeding season, males also spend time performing courtship displays during the early morning and late afternoon/evening periods (Hallager & Boylan, 2004). Kori bustards spend much of their time on the ground, generally moving about in a slow walk; running and flying are performed to escape from danger (Mwangi, 1988).

In the wild, kori bustards have a lek-like breeding system, where males gather either singly or in loose lek-like formations during the breeding season and perform "balloon" displays to attract females (Hallager & Boylan, 2004). During a male's display, the esophagus can be inflated up to four times its normal size, and with the neck expanded, the tail and wing feathers pointed downward, and the crest erected, the male emits a low-pitched six-note booming vocalization as he snaps his bill open and shut (Hallager & Boylan, 2004). Prior to copulation, males often spend 5–10 minutes pecking the head of the recumbent females; copulation itself lasts no more than a few seconds (Hallager & Boylan, 2004). Males play no role in egg or chick care, and continue to display to other females after copulation; males do not associate with females outside of breeding interactions (Hallager & Boylan, 2004).

The chicks of kori bustards are precocial, have open eyes at hatching, and are able to stand within hours (Hallager & Boylan, 2004). Average brood size of wild birds is 1.52 chicks (Osborne & Osborne, 1998). Females are thought to feed newly hatched young with insects as the primary food item. Chicks remain with the dam until the start of the next breeding season, when they disperse (Hallager & Boylan, 2004). Adult and sub-adult males have been found to disperse widely (up to 120 km (74.6 mi.)) following the breeding season, while juvenile females were found to emigrate only 2–5 km (1.2–3.1 miles) from their natal areas (Osborne & Osborne 1998, 1999, 2000).

Physical description: The following information on the physical appearance of kori bustards has been adapted in part from Johnsgard (1991). Table 3 provides ranges for kori bustard size and weight parameters.

Table 3. Size and weight parameters for *A. k. struthiunculus* (Johnsgard, 1991)

	Male	**Female**
Weight	10–15 kg (22–33.1 lb.)	5.5–7 kg (12.1–15.4 lb.)
Breeding season weight	15–19 kg (33.1–41.9 lb.)	Unknown for wild birds
Tail	370–387 mm (14.6–15.2 in)	280–342 mm (11–13.5 in)
Wingspan	752–767 mm (29.6–30.2 in)	600–655 mm (23.6–25.8 in)
Culmen	95–120 mm (3.7–4.7 in)	81–95 mm (3.2–3.7 in)
Tarsus	230–247 mm (9.1–9.7 in)	181–205 mm (7.1–8.1 in)

Adult male: Kori bustards show size-based sexual dimorphism, with adult males being larger than females; the weight of adult males range from 7–14 kg (15.4–30.9 lb.), while adult females usually weigh less than 7 kg (15.4 lb.) (Bailey & Hallager, 2003). Sub-adult males are similar in size to adult males but have a thinner neck (Osborne & Osborne, 2001). During the breeding season, male kori bustards (*A. k. struthiunculs*) have a darkened throat patch, which becomes less black following post-breeding molt. Plumage coloration is generally similar between the sexes.

Adult female: Adult females are smaller than males, approximately 3–6 kg (6.6–13.2 lb.) (Bailey & Hallager, 2003), with black on the crown and with the eye stripe somewhat reduced (Urban, Fry, & Keith, 1986). Juvenile females have a slighter appearance with a slimmer bill, thinner legs, and a brownish back (Osborne & Osborne, 2001). Table 4 provides a general description of male and female coloration.

Table 4. Coloration of kori bustards (Johnsgard, 1991; Hallager & Boylan, 2004)

Body part	**Coloration**
General coloration (back)	Dark sandy brown, with blackish vermiculation and a slight grayish shade
Mantle and upper back feathers	As above, but generally more blackish
Lower back, rump, and upper tail coverts	Similar to back, but upper tail coverts are rather more coarsely freckled
Lesser wing coverts	Similar to back
Median coverts	Mostly white, coarsely mottled with black or grey freckles, with a broad black subterminal bar and a white tip
Greater coverts	Similar to median coverts, but more thickly mottled with black or grey vermiculation
Alula	Similar to median or greater coverts, but subterminal bar not so strongly indicated
Primary coverts	Ashy brown, with the inner ones mottled and broadly tipped with white
Remiges	Brown, with two outer ones scarcely freckled with white on the outer web, but inner ones becoming whiter on the inner web, barred with bluish grey, and tipped with white
Inner primaries	Some are checkered with sandy buff on the outer webs
Secondaries	Bluish-gray, mottled with white, with a white tip, and a subterminal bar of blackish-brown; the innermost secondaries are like the back
Rectrices	Ashy brown at base, crossed by two broad bands of white, separated from each other by black bands, one broad and one narrow, the latter band is followed by an indistinct white band which merges into the sandy brown ending of the tail. This portion has a narrow band of black, a broader subterminal band of black, and a white tip
Crown	Strongly crested, black, with a grayish band of feathers down the center, and a black post-ocular stripe (stripe reduced or lacking in *A. k. kori*)

Body part	Coloration
Nape and sides of posterior crown	Grayish white and barred with black, exactly like the neck
Sides of face, throat, streak over eye, patch in front of eye, anterior cheeks, and chin	White
Foreneck	A crescentic band of black is partly concealed by the long barred feathers of the lower throat
Sides of the upper breast	Marked with black
Axillaries and under wing coverts	White
Lower primary coverts	Ashy freckled with white
Iris	Lemon yellow to orange brown
Bill	Light horn color, darker brown above, and yellowish below
Tarsi and toes	Light yellowish

Conservation issues: Kori bustards are listed in Appendix II of the Convention on International Trade in Endangered Species (CITES) and are listed by IUCN as Near Threatened. Any importation of kori bustards to the United States requires a CITES import permit and a CITES export permit from the sending country. The southern race, *Ardeotis kori kori,* is listed as Vulnerable in the *South African Red Data Book* (Brooke, 1984), as well as in the *Eskom Red Data Book of Birds of South Africa, Lesotho, and Swaziland* (Barnes, 2000). The kori bustard population in AZA-accredited facilities is managed as a Yellow SSP®.

Since the late 1800s, kori bustard ranges have contracted more than 20% in East Africa and close to 10% in southern Africa, and population sizes are reduced throughout this species' range. The causes of kori bustard decline are largely unknown but are thought to be multifactorial, including habitat loss, hunting pressure, generally poor tolerance of human activity, and a low breeding rate sensitive to natural and unnatural environmental changes (Senyatso, Collar, & Dolman, 2012). The steady decrease in kori bustard numbers in the wild indicates the need for successful captive management of this species in order to advance the current understanding of its biology and provide recommendations with which to improve its survival.

The main threats to wild kori bustards are in the form of human-induced factors, and include habitat destruction through increasing agricultural development (Dale, 1990; Ottichilo, Leeuw, & Prins, 2001), bush encroachment caused by over-grazing from livestock, hunting for the bushmeat trade, and trapping for private collections in the Gulf States. Poison used to control locusts is generally toxic to birds, and may be affecting kori bustard populations (Barnes, 2000). Although the kori bustard is listed as "protected game," it continues to be hunted over much of its range. In Namibia, it is commonly referred to as the "Christmas turkey" (Osborne & Osborne, 2001), and in South Africa it is called the "Kalahari Kentucky" (Barnes, 2000).

Collisions with overhead power lines are also a serious problem for kori bustards, with one 10 km (6 mi.) stretch of overhead power lines in the Karoo (South Africa) leading to the death of 22 kori bustards during a 5-month period (Van Rooyen, 2000). Very little is currently known about natural health and disease issues affecting wild kori bustards, or how their health status relates to declining populations. Recent studies have begun to collect such valuable baseline information on free ranging kori bustards.

Natural factors affecting populations of kori bustards in the wild include an inherently low reproductive rate and reduced breeding activity in dry years, despite a constant predation pressure. In addition, favored areas such as tree-lined watercourses are becoming unsuitable for kori bustards because they are being invaded by alien plant species (Barnes, 2000).

Present range of *Ardeotis kori struthiunculus*: The present range of this subspecies is smaller than in previous times. In Ethiopia, the species is now found only south of 9º latitude. From there, the range extends west to the extreme southeastern part of Sudan and south to western Kenya and northeastern Uganda. There are no records of birds in Somalia since 1970. In Tanzania, it is restricted to the northern plains. They are scarce around the coastal lowlands of Tanzania and Kenya (Zimmerman, 1996).

Historical distribution of *Ardeotis kori struthiunculus*: The subspecies historically ranged throughout most of Ethiopia (Ash, 1989) and southeastern Sudan (below 9º latitude). The subspecies also ranged southeast to northwestern Somalia, and west and south to northern Uganda, Kenya, and the highlands north of the Singida providence in Tanzania.

Present range of *Ardeotis kori kori*: The present range of this subspecies is smaller than in previous times. It is now distributed in the semi-arid areas in the western half of southern Africa, including Namibia, extreme southern Angola (rarely), western Zambia, Botswana, western Zimbabwe, South Africa, and the Limpopo Valley of Mozambique.

In South Africa, it is found mainly in the Transvaal Lowveld and the northern Cape Province, as well as Kruger and Kalahari Gemsbok National Parks (Kemp, 1980), although it is very scarce along the eastern border of Kruger National Park near Mozambique (Barnes, 2000). *A. k. kori* is a vagrant in Lesotho (P. Goriup, personal communication, 2007). Allan (1988) reported that the subspecies has declined in the Transvaal, Orange Free State, and parts of Cape Province; Parker (1994) noted that this subspecies went extinct in Swaziland prior to 1960.

Historical distribution of *Ardeotis kori kori*: This southern subspecies historically ranged throughout most of southern Africa, including Zimbabwe, Botswana, southern Angola, Namibia, South Africa, southern Mozambique (Johnsgard, 1991) and Swaziland (Harrison et al., 1997).

Population: Throughout its range, the kori bustard (including both subspecies) is uncommon to locally common, but generally declining (Urban et al., 1986). The habitat of both subspecies is under threat from the spread of agriculture, high human densities, illegal hunting, overgrazing by livestock, and bush encroachment. According to del Hoyo et al. (1996), the kori bustard is showing signs of chronic decline and local extinction over its entire range. Total population size has not been reported for either subspecies.

The entire East African region is currently undergoing widespread ecological changes as a result of increased agricultural practices and other forms of land use (Mwangi, 1988). Since 1950, the area of land used for agriculture has increased by 26% (Happold, 1995). Lado (1996) states that habitat destruction and/or alteration the most serious threat to the future of wildlife in Kenya. As an example, the area used for wheat production in the Masai Mara has grown from 4,875 ha in 1975 to over 50,000 ha in 1995. In the nearby Loita plains (where kori bustards are known to frequent), wheat production continues to expand as the human population grows, and as farmers realize the agricultural potential of the land. As areas used for agriculture expand in Kenya, it can be expected that the numbers of wildlife, including kori bustards, will decline (Ottichilo, Leeuw, & Prins, 2001).

The spread of agriculture, urbanization, pollution, pesticides (including those that are banned in other countries), and other consequences of an exploding human population, are all contributing to a deteriorating situation for many species of wildlife, including kori bustards. Table 5 provides information on the conservation status of kori bustards in range countries.

Table 5. Status of *Ardeotis kori struthiunculus* and *Ardeotis kori kori* in range countries

Country	**Population size/status**
Ardeotis kori struthiunculus	
Sudan	Breeding populations exist in the extreme southeastern area of the country, but total population size is unknown. Kori bustards may be only a dry season visitor to this country (Nikolaus, 1987).
Kenya	Kori bustards are most numerous in the dry grassland areas of northern and western Kenya, and in the Rift Valley highlands south to Mara Game Reserve, Loita Plains, Nairobi National Park, and Amboseli National Park. They are scarce and localized from the Tana River south to Tsavo West and Tsavo East National Park (Zimmerman, 1996). Total population size is unknown. Mwangi (1988) estimated 0.3 birds per km^2 in Nairobi National Park in 1986/87.
Uganda	Breeding populations exist in Acholi, Lango, and Kidepo National Park. Total population size is unknown.
Ethiopia	Kori bustards were formerly common in Ethiopia south of 9º latitude, but numbers have declined (P. Goriup, personal communication). Total population size is unknown.
Somalia	There are no records of birds in Somalia since 1970.
Tanzania	The Serengeti National Park, Ngorongoro Conservation Area, Tarangire National Park, Maswa Game Reserve, Arusha National Park, and Mkomazi Game Reserve offer long-term protection to kori bustards, and viable populations of birds can be found in these protected areas. Kori bustards

Country	Population size/status
	are still relatively common in the Rift Valley highlands. There is a small and isolated population in central Tanzania, which occupies a small area at low densities (N. Baker, personal communication). This subspecies is regarded as scarce around the coast (Zimmerman, 1996). The birds are hunted around the Lake Eyasi Basin, Lake Natron, and in the foothills of Mt. Kilimnajaro (N. Baker, personal communication). Total population size is unknown.
Ardeotis kori kori	
Botswana	Despite low human densities, kori bustards in Botswana are under severe pressure from habitat loss. Nonetheless, strongholds for the species include the Kalahari Gemsbok National Park (with an estimated population size of 100–140 birds (Barnes, 2000)), Central Kalahari Game Reserve, Nxai Pan National Park, and the Chobe National Park, where road counts found 1 bird/106 km. However, in unprotected areas, the density level dropped to 1 bird/4,356 km (Harrison et al., 1997). Suitable habitat for kori bustards has been lost due to grazing by livestock (Herremans, 1998), which has increased dramatically over the past 100 years. Livestock numbers continue to grow despite reports of overgrazing and forecasts of devastating long-term land degradation since the early 1970s. Total population size of kori bustards is unknown.
Namibia	The kori bustard stronghold in Namibia, and possibly the world, is Etosha National Park. There, Osborne and Osborne (1998) found 1 bird/16 km. However, birds are hunted outside park boundaries.
Zimbabwe	Suitable habitat for kori bustards is deteriorating through overgrazing by livestock, and the situation is similar to Botswana. Matabeleland is the stronghold for the species in Zimbabwe (Rockingham-Gill, 1983). The species has decreased in several areas, most noticeably in the Mashonaland plateau (Harrison et al., 1997), where birds are hunted. The decline in this area was first noticed in the 1920s (Irwin, 1981). Total population size has been variously reported in 1980, when an estimation of 10,700 birds was given by Rockingham-Gill (1983), although Dale (1990) reported 5,000 birds, and Mundy (1989) estimated 2,000 birds, and states that Rockingham-Gill's 1983 assessment is vastly over estimated. Total population size is unknown.
South Africa	Numbers declined in the 20th century, but the extent of the decline is unknown (Brooke, 1984). Kruger National Park supports 100–250 individuals (Barnes, 2000). Outside protected areas, kori bustards are found in relatively large numbers only in the Platberg-Karoo Conservancy in South Africa (Barnes, 2000). Allan (1988) reported that the species has declined in the Transvaal, Orange Free State (where it is uncommon to rare), and in parts of Cape Province. Total population size is estimated to be between 2,000–5,000 birds.
Mozambique	The kori bustard population is locally threatened (hunting is the greatest threat), and probably numbers less than 100 birds (Parker, 1999).
Other	Parker (1994) noted that this subspecies went extinct in Swaziland prior to 1960. In Angola, the species is rare. In Zambia, kori bustards are found only west of the Zambezi River, though their status there is unclear. The Sioma Ngwezi National Park may offer some protection. This subspecies is considered very sparse in Natal with one sighting reported in 1976 (Cyrus & Robson, 1980).

Buff-crested bustard:

Natural history: The buff-crested bustard *Lophotis gindiana* is found in East Africa from Ethiopia and Somalia south through Kenya to northeastern Uganda and central Tanzania. The closely related Savile's bustard (*Lophotis savilei*) is found from Senegal east to Sudan. Another member of the same genus, the red-crested bustard (*Lophotis ruficrista*), is found in southern Angola, Namibia through Botswana, Zambia, southeast to Mozambique, and northern South Africa. Buff-crested bustards live in arid to semi-arid habitats that offer some cover. They are rarely found in open areas. In Kenya and parts of East Africa, they are found at elevations of 1500 m (0.93 mi.) or less. Movements in response to rainfall and food supply have been recorded, but the species is not truly migratory.

Taxonomic note: *Lophotis gindiana* (del Hoyo & Collar, 2014) was previously placed in the genus *Eupodotis*. *Lophotis (Eupodotis) ruficrista, L. savilei*, *and L. gindiana* (Sibley & Monroe 1990, 1993) are retained as separate species contra Dowsett and Forbes-Watson (1993) who included *L. savilei* and *L.*

gindiana as subspecies of *E. ruficrista*. For the purposes of AZA management, the three populations are considered one species comprised of three subspecies (BirdLife International, 2016).

Physical description: *Lophotis gindiana* is tawny-brown above, marked with large, dark brown pointed spots and finer irregular brown and buff barring. Its head is plain sandy buff, but slightly greyer around the eyes. Its crown is light brown with buff and dark brown speckling and has a dropping rufous buff crest noticeable in display. A narrow black throat stripe extends down the foreneck to the black belly that contrasts with the white on the wing coverts. Most of its neck is buff grey and its breast is grey with white patches at the sides. Its tail has vermiculated black and white feathers that appear grey in flight except for the broad black tips. The bill is dark grey, and the eyes are pale cream. The female differs from the male in being more buffy colored on the crown and nape. The buff colored neck is heavily flecked and barred with brown and lacks a black foreneck streak of the male (Zimmerman, 1996).

Like all bustards, buff-crested bustards have no preen gland. Instead of using an oil gland to keep their feathers clean, they employ dustbathing. Their feathers contain light sensitive porphyrins, which give them a pink tinge at the base. As with all bustards, they lack a hind toe.

Conservation issues: Buff-crested bustards are listed by IUCN as a species of Least Concern. Any importation of buff crested bustards to the United States require a CITES import permit. The AZA buff-crested bustard population is managed as a Red SSP®.

Present range of *Lophotis gindiana*: The buff-crested bustard is native to Djibouti, Ethiopia, Kenya, Somalia, South Sudan, Sudan, Tanzania, and Uganda. The global population size has not been quantified, but the species is reported to be common throughout its range except in Sudan where it is generally uncommon (del Hoyo et al., 1996). The population is believed to be stable in the absence of evidence for any declines or substantial threats (BirdLife International, 2016; del Hoyo, et al., 1996).

Historical distribution of *Lophotis gindiana*: The historical distribution of the buff-crested bustard is presumed to be similar to its current range.

Table 6. Status of *Lophotis gindiana* in range countries

Country	Population Size/Status
Lophotis gindiana	
Ethiopia	Total population size is unknown; but population is suspected to be stable in the absence of evidence for any declines or substantial threats.
Somalia	Total population size is unknown; but population is suspected to be stable in the absence of evidence for any declines or substantial threats.
Uganda	Total population size is unknown; but population is suspected to be stable in the absence of evidence for any declines or substantial threats.
Kenya	Fairly common and widespread in dry northern and eastern Kenya, mainly below 1,250 m (0.8 mi.). South of the highlands, it ranges west into the Rift Valley only around Olorgesailie (Zimmerman, 1996).
Tanzania	In northern Tanzania, common on the Masai Steppe west to Lake Natron and Tarangire National Park (Zimmerman, 1996).
Djibouti	Total population size is unknown; but population is suspected to be stable in the absence of evidence for any declines or substantial threats.
South Sudan	Generally uncommon; An estimate of 25–40 females per 10,000 ha in suitable habitat.
Sudan	Total population size is unknown; but population is suspected to be stable in the absence of evidence for any declines or substantial threats.

Chapter 1. Ambient Environment

1.1 Temperature and Humidity

The animals must be protected or provided accommodation from weather, and any adverse conditions. (AZA Accreditation Standard 1.5.7). Animals not normally exposed to cold weather/water temperatures should be provided heated enclosures/pool water. Likewise, protection from excessive cold weather/water temperatures should be provided to those animals normally living in warmer climates/water temperatures.

> **AZA Accreditation Standard**
>
> **(1.5.7)** The animal collection must be protected from weather detrimental to their health.

Cold weather: Bustards are susceptible to frostbite (Bailey & Hallager, 2003). In climatic zones where temperatures fall below 0 °C (32 °F), both species of bustard require winter holding facilities for housing birds during inclement weather. To avoid frostbite, it is also strongly recommended that institutions provide their bustards with supplemental heat when temperatures fall below 4 °C (40 °F). Birds should be locked into heated winter holding quarters when the wind-chill temperature is forecasted to be below -6 °C (20 °F), especially when it is raining or sleeting, and always when it is snowing (Hallager & Boylan, 2004). Bustards should not be left outside during periods of freezing rain or snow. Winter holding facilities should be heated to 10–15.5 °C (50–60 °F). When heat bulbs are used, they should be encased in protective wiring to prevent the birds from breaking the bulbs with their bodies or beaks where contact is possible. Both wild-caught birds and birds born in zoos will readily utilize heated straw piles in their enclosure when temperatures fall below 4 ºC (40 ºF). The availability of heated pads (e.g., pig warmers) covered with straw will also allow birds to remain outside for longer. This is especially advantageous when working with wild-caught birds, which can be more reluctant to utilize indoor shelters. The straw should be replaced when it becomes wet, as bustards will not use wet straw piles. As the feathers of bustards are not particularly waterproof, care should be taken to avoid the birds becoming wet during any cold conditions.

Even institutions that do not experience extremely cold weather should have a shelter available for times when enclosure repairs are needed, for medical confinement, to house temperature-sensitive chicks, to minimize food loss from wild birds during feeding, or when birds have to be caught and restrained. For the first couple of months of their lives, bustard chicks are sensitive to the cold (T. Bailey, personal communication, 2007). Care is needed to provide sufficient heating, especially to debilitated chicks that are hospitalized. Managers should follow the advice of the referring aviculturist or veterinarian for temperature guidelines for chicks. Under sub-optimal temperature conditions (21ºC (70ºF)), bustard chicks and even juvenile bustards can suffer from hypothermia (see section 7.4). For more details on the size and design of winter holding facilities, see section 2.1.

Hot weather: Exhibits should offer some degree of direct sunlight (see section 1.2), but areas of shade that all birds within a group can utilize also need to be available, especially in hotter climates. Hyperthermia has caused the death of a number of bustards at the National Avian Research Center (NARC), and can occur when chicks are moved prematurely from air-conditioned rearing facilities to outdoor aviaries in the summer, without a period of acclimatization (T. Bailey, personal communication, 2007). See section 7.4 for additional information on hyperthermia.

Humidity: Bustards do not thrive in climates that are consistently wet, rainy, and damp. These conditions lead to poor feather condition (bustards do not have a preen gland to oil their feathers) and unhealthy birds. Warm, dry shelters and areas of full sun that allow animals to dry themselves are recommended (see section 2.1).

AZA institutions with exhibits which rely on climate control must have critical life-support systems for the animal collection and emergency backup systems available. Warning mechanisms and backup systems must be tested periodically (AZA Accreditation Standard 10.2.1).

> **AZA Accreditation Standard**
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> **(10.2.1)** Critical life-support systems for the animals, including but not limited to plumbing, heating, cooling, aeration, and filtration, must be equipped with a warning mechanism, and emergency backup systems must be available. Warning mechanisms and emergency backup systems must be tested periodically.

Climate control systems: The AZA Gruiformes TAG, Kori Bustard SSP®, and Buff-crested Bustard SSP® recommend that each institution identify the most appropriate climate control systems suitable for their bustard enclosures in order to meet the temperature and humidity recommendations provided above.

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. Bustards need areas of full sun to allow them to dry off damp plumage, and to engage in sunning behaviors. Sunning, characterized by the spreading of one or both wings in response to direct sunlight, is very important to bustards for maintaining good feather condition. If exhibits contain multiple birds, multiple sunning spots are necessary to ensure that each individual has access to a sunning area, and that no one individual can prevent another from gaining access to these areas. Birds demonstrate preferred sunning areas. Data from a behavioral assessment of kori bustards at one AZA-accredited institution (Fernandes & Hallager, 2007) found that kori bustards exhibit sunning behaviors intermittently from 1100h to 1400h on sunny days. Birds will sometimes sun themselves until they pant heavily, at which point they usually move to a shady area of the enclosure. In the shade, the birds typically then preen for upwards of 20 minutes.

The proposed functions of sunning behavior in various bird species includes ectoparasite control, thermoregulation, drying of wet plumage, assistance in molting, soothing of skin irritated by molting, and increasing the flow of preen gland secretions (Simmons, 1986). Since kori bustards preen intensely following sunning, there is some evidence that suggests that this behavior is involved in feather maintenance and ectoparasite control. Kori bustards lack preen glands, so their sunning activity cannot increase the flow of preen gland secretions or aid in vitamin D production. Sunning also does not appear to be a major thermoregulation mechanism for kori bustards, as it is in some other bird species. Some kori bustards do not sunbathe in the colder months but do perform sunning once temperatures are above 7.2 °C (45 °F). Sunning is most prevalent during the summer months and is performed at a median temperature of 23.9 °C (75 °F) (Fernandes & Hallager, 2007).

As kori bustards should not be kept inside for extended periods, weather permitting, providing artificial UV-emitting lights is not necessary within indoor enclosures. Similarly, artificial seasonal changes in indoor light intensity and duration do not seem to be necessary, as kori bustards are not maintained indoors throughout the year. Zoos that bring kori bustards into indoor enclosures for the winter should have outside holding pens available for use by the birds during warm days, in order to provide them with full spectrum lighting.

Buff-crested bustards are often kept completely indoors and should be provided with appropriate lighting. UVB lights may improve breeding.

Chick rearing: Standard incandescent bulbs should not be used for brooding hand-reared, bustard chicks, as at least three chicks reared under incandescent bulbs at National Aviary Research Center in 1993 developed cataracts that may have been associated with the use of these bulbs (Bailey, T. A., Naldo, J., Samour, J. H., Sleigh, I. M., & Howlett, J. C. (1997). b; T. Bailey, personal communication, 2007). No further cases of cataracts were observed at National Aviary Research Center once the lighting was changed, and when the chicks were reared under 60 W ceramic dull-emitter bulbs (Bailey, T. A., Naldo, J., Samour, J. H., Sleigh, I. M., & Howlett, J. C. (1997). However, further research is needed to investigate possible links between light type during rearing and cataract development.

The relative importance of sunlight for bustard chick development is unknown. The Kori Bustard SSP® recommends that chicks are provided access to natural light as soon as possible. In the absence of natural light, full-spectrum bird type lamps should be provided. These should be monitored with UV meters and lamps replaced when needed. Popular practice suggests that chicks be exposed to sunlight to some degree on a daily basis starting at day seven. However, the exact day that this exposure should begin is subjective. Some birds that were not exposed to natural light until they were two months of age have developed satisfactorily.

1.3 Water and Air Quality

There are no recommendations or regulations specific to air and water quality for bustards in zoos. Any clean water source that is considered suitable for livestock is acceptable for bustards. Water in bowls and drinkers should be changed once a day at minimum, but more frequently if the water becomes contaminated. Bustards require only small areas of water from which to drink, as they are not heavy drinkers, but they do drink on a daily basis. Heated water dispensers for northern zones are recommended to prevent water from freezing. Bustards do not bathe in water (they dustbathe), so pools are not needed within enclosures other than for aesthetic reasons.

From an air quality perspective, holding areas and indoor or winter enclosures should not be sealed so tightly that they prevent fresh air from entering the area, as this can adversely affect air quality within these enclosures. A ventilation system, small windows with screens, and/or the ability to open windows, will allow fresh air to enter these areas, and will discourage the formation of fungal spores. Air exchange rates, air filters or HVAC systems for closed indoor systems are not applicable to bustard housing situations and are therefore not provided in this manual.

1.4 Sound and Vibration

Consideration should be given to controlling sounds and vibrations that can be heard by bustards in their indoor and outdoor enclosures. Bustards can become habituated to the sounds of routine zoo operation (e.g., trash trucks, nearby construction, leaf blowers), as well as to other environmental sound stimuli (e.g., overhead aircraft, traffic noise, etc.). However, some individuals remain skittish at such sounds throughout their lifetime. Unusual sounds, however, can act as stressors, and may cause birds to react negatively by running or crouching. Breeding activities can also be interrupted by novel sounds. The timing of planned construction work near to bustard enclosures should coincide with the non-breeding season to minimize stress on the birds (see section 8.1 for information on reproduction). Workers should be cautioned that their activities might stress the birds. Work should be stopped at once if it is causing obvious stress to the birds (e.g., birds not eating, running within their enclosure, or remaining in the crouch position), and the situation should be re-evaluated. In some cases, birds may need to be temporarily housed or even relocated to another exhibit, although the pros and cons of moving animals should be carefully discussed.

Little is known about the hearing sensitivity of bustards, and there is no information available on whether there are certain frequencies of sounds or decibels that will have the greatest negative effect on the welfare of bustards. Bustards should be carefully monitored in any situations where loud, atypical sounds can be heard by caretakers around bustard enclosures. Additional research on hearing in bustards would provide some guidance for creating more objective recommendations for managing sound stimuli for these taxa.

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

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

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

> **AZA Accreditation Standard**
>
> **(1.5.2.1)** All animals must be kept in appropriate groupings which meet their social and welfare needs.

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

Species-appropriate behaviors: All bustard enclosures and husbandry management programs in zoos should be designed with knowledge of the species' natural behaviors in mind. Appendix G provides a comprehensive ethogram of kori bustard behavior that should be carefully considered from an enclosure design and enrichment perspective (see section 9.2 for more information on environmental enrichment). An ethogram for the buff-crested bustard does not exist, but behaviors are similar to that of kori bustard.

Kori bustard enclosure space: Large paddock-like enclosures are the most appropriate type of space for kori bustards, and exhibits up to 4,645 m^2 (50,000 ft^2) have been provided to this species. Kori bustards will be more likely to thrive if provided with a few hectares of space (Hallager & Boylan, 2004), opportunities for their full range of species-appropriate behaviors, and a complex environment to meet their physical and social needs. The recommended minimum space for outdoor enclosures is 13 m x 20 m (42.7 ft. x 65.6 ft.) per bird (Siegel, Hallager, & Bailey, 2007). All enclosures should be large enough for bustards to avoid animal caretakers when enclosures are cleaned, re-provisioned, etc., and to maintain their preferred flight distance from animal care staff. Indicators that the size and complexity of the enclosures are not meeting the needs of the birds may include poor physical health, pacing along fence lines, and increased behavioral displacements between females or between males and females.

Buff-crested bustard enclosure space: The minimum size range for enclosures housing a single breeding pair of buff-crested bustards is between 5 m x 3 m x 2.5 m (16.4 ft. x 9.8 ft. x 8.2 ft.) (Siegel, Hallager, & Bailey, 2007) and 9 m x 9 m x 2.5 m (29.5 ft. x 29.5 ft. x 8.2 ft.) (Anderson, 1998a). The minimum height should be carefully considered, as many cases of death and physical trauma (e.g., fractured necks) have been related to birds hitting themselves against the ceiling of their enclosures (Anderson, 1998a). Even pinioned birds can propel themselves vertically to a height of 2.5 m (and possibly beyond). It may be possible to have a pair of breeding buff-crested bustards that can be housed together year-round, although this is rare. Serious aggression problems can occur during the breeding season, such that males and females may need to be separated and allowed access to each other only for copulations. Where males and females are housed in adjacent pens, a small gate should be built into the dividing wall; the gate should be 13 cm (5.1 in.) wide and 20 cm (7.9 in.) high, and have the option to be fastened open (Siegel, Hallager, & Bailey, 2007).

Enclosure design for bustards:
Bustards are mostly terrestrial birds but do fly. Kori bustards generally move about in a slow walk; running and flying behaviors are generally only performed to escape from danger (Mwangi, 1988). Unless the enclosure is completely covered, birds should be flight restrained (see section 2.2). Covered enclosures have the added benefit of keeping out unwanted pests or potential predators (see section 2.2), and can help to minimize the spread of parasites and diseases from wild animals to the kori bustards. Buff-crested bustards are typically kept in covered pens; if not, they should also be flight restrained. As bustards are mostly terrestrial, all unnecessary obstacles should be avoided within enclosures. The enclosures should also be as flat as possible to avoid long-term leg and hip problems that may develop if birds are housed in

areas with hills. However, some low hills can provide additional cover and serve as courtship display areas for males (see section 8.1 for more information on courtship displays).

To minimize trip hazards, enclosures should be kept as free as possible from non-plant furnishings (e.g., rocks, tree stumps). There should be ample bare ground as bustards generally do not like to walk on turf, especially if it is moist (e.g., from dew). Access to sandy patches can help to promote dustbathing, sunning, and preening behaviors. Bustards are not perching birds, lacking a hind toe with which to perch, so there is no need to provide opportunities for perching within enclosures. If pools are present in enclosures, they should be shallow enough that a bird can walk through the water, and the sides should gradually slope to the deepest portion. The coating on the sides of the pool should be of a non-slip nature. Trauma is a prominent cause of morbidity and mortality for all bustards in zoos, so care should be taken to use 'soft' materials when enclosures are constructed.

Bustards can be housed with a variety of plant furnishings, although there are some considerations to keep in mind. Plants that have large thorns and/or thick flower stems should not be used, as birds can injure themselves. Additionally, bamboo and ornamental grasses with thick, hard stems should be avoided, because birds have impaled themselves on these stems (Hallager & Boylan, 2004). Large, soft-stemmed grasses and small shrubs scattered throughout an exhibit will provide the birds with ample cover, and visual barriers for enclosures housing multiple birds. Large shrubs and mature trees, which offer shade as well as limited protection from inclement weather, are also desirable. Furthermore, nervous birds are less likely to injure themselves by flying into the ceiling if they have vegetation in which to hide when animal caretakers are servicing the enclosure (Siegel, Hallager, & Bailey, 2007).

As omnivores, bustards will also utilize natural plantings to forage for fruits, berries, and any associated small vertebrate and invertebrate prey. Plants with edible, non-toxic fruits that can be eaten by the birds can be planted in the enclosure with approval from institutional veterinarians and nutritionists (see section 6.2 for additional information on the use of browse), but nutritional value, fruit size, and attractiveness of the fruit to unwanted pest species should be carefully considered. Plants with fruits large enough to cause an impaction should be avoided. Along with natural vegetation, the addition of alfalfa beds provides birds with a direct food source and a cover to hide in, and encourages foraging for invertebrates attracted to the vegetation (Siegel, Hallager, & Bailey, 2007). Some areas of the enclosure should be left as bare dirt or sand to allow birds to perform dustbathing.

Enclosure substrates: Substrate used in all bustard enclosures should be natural and non-abrasive for the feet and hock joints of the birds. Bustards should be provided with substrates that promote a wide range of species-appropriate behaviors (e.g., dustbathing, preening, foraging, nest building, etc.). Areas that provide the opportunity for dustbathing should be provided in both indoor and outdoor enclosures, as this is a behavior commonly seen in wild birds (Mwangi, 1988). These areas can contain sand, mulch, peat moss, or coarse oyster shell.

Bare floors lacking natural substrates, specifically substrates promoting manipulation and/or foraging, may encourage coprophagy in bustards (Huchzermeyer, 1998). Bustard chicks are known to be coprophagic (T. Bailey, personal communication, 2007). Ingestion of feces can lead to the rapid spread of diseases and parasites within a group of birds. As such, care should be taken to remove feces from enclosures as soon as possible (i.e., at least daily), especially when chicks are present.

<u>Indoor substrates:</u> The floor of indoor enclosures can be dirt or sand that is covered with bedding hay (straw). Concrete is not recommended, as birds can slip if startled. Enclosures with concrete floors should be covered with non-slip materials (e.g., indoor/outdoor carpeting).

<u>Outdoor substrates:</u> A natural soil or grass substrate is the most appropriate substrate for outdoor exhibits, although sand and gravel have also been used successfully (T. Bailey, personal communication, 2007). Damp, marshy areas within enclosures should be avoided and there should be ample bare ground that is not covered with grasses or turf. Ingestion of foreign bodies including nails, galvanized wire, and pieces of plastic-coated chain-linked fencing is not uncommon in bustards (see section 2.2).

Minimizing stressors: Attention to the design of facilities and the behavior of staff members working with bustards is important to minimize stressors and trauma-related problems. Bustards are generally nervous and alert when on the ground, and will move into cover in response to aversive stimuli or at the first sign of danger (Hallager & Boylan, 2004). Various stimuli within the zoo environment have been reported as potential stressors for bustards, including capture and restraint of birds, animal caretakers or other staff working in or near the enclosure (especially with loud machinery such as line trimmers), native birds competing for food, animal introductions, aggressive behavior between birds, feeding time, and the presence of several keepers in visual range of the birds. High visitor levels have also been shown to be stressful to some birds (Brostek, Hallaers, & Powell, 2003). Visual barriers, such as thick shrubs, placed 1.3–2.5 m (4.3–8.2 ft.) in from the enclosure perimeter, may provide an increased sense of security for birds housed in small enclosures (Hallager & Boylan, 2004), and may help mitigate some of the stressors listed above. Ensuring that visitors are kept at least 1.5 m (5 ft.) from the perimeter of bustard enclosures may also be beneficial (S. Hallager, personal communication, 2006). Using shade-cloth or tension netting on the roof and sides of aviaries and within holding barns can minimize visual stressors and cushion any impact resulting from birds flying within an enclosure due to stressors.

Typical vocal indicators of stress include growling and barking, although young birds may be generally more vocal even in the absence of specific stressors. Non-vocal indicators of stress include running/chasing, stereotypical pacing (though this can also occur naturally prior to egg laying), fluffing, positioning tail in up/alert position, and tucking (Hallager & Boylan, 2004). A common response to stressors is hiding. Hiding can be induced by high crowd levels, which may cause subordinate birds to seek areas away from visitors. New bustard enclosures should limit visitor access to no more than two sides of the enclosure, in order to prevent excessive visitor presence around the enclosure perimeter. Stress can also be potentially decreased by reducing the number of non-essential people who enter enclosures, or the off-exhibit areas directly around these enclosures. Bustards are shy birds by nature and should have areas of privacy to retreat to when crowd levels are high (Brostek, Hallaers, & Powell, 2003). These areas can also provide secure nesting spots for females during the breeding season.

The same careful consideration regarding exhibit size and complexity and its relationship to the bustard's overall well-being must be given to the design and size of all enclosures, including those used in exhibits, holding areas (e.g., winter holding), hospital/treatment rooms, and quarantine/isolation enclosures (AZA Accreditation Standard 10.3.3). Sufficient shade must be provided by natural or artificial means when sunlight is likely to cause overheating or discomfort to the animals (AZA Accreditation Standard 10.3.4).

The design of all enclosures for bustards should minimize negative stressors, allow for efficient handling and restraint when necessary, provide access for emergency and routine procedures, maximize the potential for social interaction (and separation when needed) and a full range of species-appropriate behaviors, and should effectively integrate enrichment and animal training (see Chapter 9) into the daily husbandry routine.

The use of bustards in conservation/education programs is generally limited due to behavioral and health concerns for the animals (see Chapter 10) and there are no additional recommendations for enclosures specific to animals used in these programs.

AZA Accreditation Standard

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

AZA Accreditation Standard

(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.

Winter holding areas: All bustard species currently kept in North America (kori bustard, buff-crested bustard, and white-bellied bustard) are susceptible to frostbite. In climatic zones where temperatures fall below 0 °C (32 °F), institutions should have winter holding facilities available for housing birds during inclement weather. To avoid frostbite, it is also strongly recommended that these institutions provide their bustards with supplemental heat. Bustards should not be left outside during periods of freezing rain or snow.

<u>Kori bustard winter housing</u>: For winter holding of two compatible kori bustards, the minimum recommended space per bird is:

- 2.4 m x 3 m (8 ft. x 10 ft.) for overnight holding
- 2.4 m x 4.9 m (8 ft. x 16 ft.) for housing up to 7 days
- 3 m x 6.1 m (10 ft. x 20 ft.) for housing longer than 7 days, should also include an outside holding yard so that birds can be given outside access

The recommended oblong dimensions provide some exercise space, and allow the birds to distance themselves from keepers during enclosure cleaning. Birds, especially wild caught individuals, will usually move away from animal caretakers who are closer than 1.8 m (6 ft.) away (S. Hallager, personal communication, 2006). These size recommendations are highly dependent on the compatibility of conspecifics. Larger dimensions or separate shelters may be required for birds that have a lower degree of social compatibility. Winter holding areas should have the capability to be divided in the event that birds are not compatible in small areas, as can be especially common when males and females are housed together.

Sheds should be heated to a maximum of 10–15.5 °C (50–60 °F), and less if heating lamps and/or pads provide warmer areas. Heat bulbs, if used, should be encased in protective wiring to prevent bulb breakage in case a bird contacts the bulb. Care should be taken to secure and/or elevate in cords to reduce trip hazards. Skylights within the winter holding area can be advantageous for birds that have to be housed for extended periods during the winter, but artificial lights should also be installed to provide adequate light during the day. A small night-light turned on at night will provide some degree of light for the birds. The floor of the shed can be dirt or sand that can be covered with bedding hay (straw). Concrete is not recommended, as birds can slip if startled. Non-abrasive walls are important to minimize wing damage caused by any pacing. Sliding doors, operated from outside of the holding area, are useful for controlling the location of birds within the shelter.

<u>Buff-crested bustard winter housing</u>: For winter holding of one buff-crested bustard, the minimum recommended space is:

- 2.1 m x 3.7 m x 2.4 m (7 ft. x 12 ft. x 8 ft.) for housing less than 1 week
- 2.1 m x 3.7 m x 2.4 m (7 ft. x 12 ft. x 8 ft.) for housing longer than 7 days, should also include an outside holding yard of the same size as indoor holding so that birds can be given outside access
- Interior holding sheds should include a soft ceiling mesh as males can jump quite high and incur head damage.

The recommended oblong dimensions provide some exercise space, and allow the birds to distance themselves from keepers during enclosure cleaning. Birds (especially wild caught individuals) will move away from animal caretakers who are closer than 3 m (10 ft.) away (M. Herry, personal communication, 2015). These size recommendations are highly dependent on the compatibility of conspecifics. Larger dimensions or separate shelters may be required for birds that have a lower degree of social compatibility. Winter holding areas should have the capability to be divided in the event that birds are not compatible.

Holding areas should be heated to 10–15.5 °C (50–60 °F), and less if heating lamps and/or pads provide warmer areas. Heat bulbs, if used, should be encased in protective wiring to prevent bulb breakage in case a bird makes contact with the bulb. Skylights within the winter holding area can be advantageous for birds that have to be housed for extended periods during the winter, but artificial lights should also be installed to provide adequate light during the day. A small night-light turned on at night will provide some degree of light for the birds. Rugs or bedding hay (straw) may be used to add additional warmth to the floor. Sliding doors, operated from outside of the holding area, are always useful for controlling the location of birds within the shelter.

2.2 Safety and Containment

Bustards 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).

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

AZA Accreditation Standard

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

AZA Accreditation Standard

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

AZA Accreditation Standard

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

AZA Accreditation Standard

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

animals, staff, public, and wildlife (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.

Flight restraint: The three main methods of flight restraint for bustards are pinioning, feather clipping, and the use of covered enclosures. Each method poses some risk to the birds. Pinioning bustards may leave them prone to injury because the removal of a large section of the wing can make them unbalanced when they jump and land. Frequent feather clipping requires that birds be captured and restrained on a regular basis, and can lead to physical injuries and trauma (see section 7.5). Allowing full-winged birds to be housed in completely covered aviaries may also result in injuries if birds attempt to fly within the restricted space and collide with the containment barriers of the enclosure.

The Kori Bustard SSP® and the Buff-crested Bustard SSP® do not recommend pinioning as method of flight restraint. However, if facilities pinion, the procedure should be carried out at no later than 3 days of age. Research has shown that reproduction in kori bustards and buff-crested bustards is not compromised by pinioning, as copulation in both species occurs while lying on the ground (Hallager & Boylan, 2004). However, it is the recommendation of the Kori Bustard SSP® and the Buff-crested Bustard SSP® that adult, full-winged bustards should not be pinioned. Unlike for chicks, the procedure in adult birds is very difficult, stressful, and can lead to complications from the surgery and further injury as birds learn to adapt to an altered wing. Regular feather clipping is the recommended procedure for rendering non-pinioned adult birds flightless. Buff-crested bustards should be clipped on both sides (J. Oosterhuis, personal communication, 2016) while kori bustards may be clipped on one side (S. Hallager, personal communication, 2018). Educating handlers on the proper techniques of catching adult birds is necessary to minimize trauma to the birds during feather clipping (see section 7.5). Additional research on the welfare of flight-restricted bustards is still needed in order to develop the most effective animal care recommendations for housing these animals in zoos. For more information, see Appendix N "Recommendations for Developing an Institutional Flight Restriction Policy" document (developed by the Avian SAG in December 2013).

<u>Kori bustard</u>: The preferred primary containment boundary bustard enclosures is 2.5 cm (1 in.) chain link mesh, durable nylon mesh, or Invisnet® for kori bustards. For birds less than 6 months, enclosure mesh should be smaller than 1.27 cm x 7.62 cm (½ in. x 3 in.) to prevent wings from slipping through and causing breaks. This size mesh reduces large rodents and small predators getting in. The smaller size also eliminates any chance of a bird getting a leg caught in the fence during a capture. If nylon mesh and/or Invisnet® is used, it should be of a strength to withstand a coyote bite. Even though some birds are successfully maintained in exhibits with lower, public-friendly fences, the recommended height of fencing is 3–3.7 m (10–12 ft.). Kori bustards are powerful flyers, and even flight-restrained birds can escape a 2.4 m (8 ft.) fence on a windy day or when startled. Covered pens, with anti-dig barriers to prevent predators or pests from gaining access to the enclosure, are recommended for facilities where kori bustards are allowed 24/7 access to outside enclosures.

<u>Buff-crested bustard</u>: The preferred primary containment boundary bustard enclosures is 2.5 cm (1 in.) mesh for buff-crested bustards. This size mesh reduces the chances of chicks getting out of enclosures, and large rodents and small predators getting in. The recommended height of fencing is 2.5 m (8 ft.). Covered pens, with anti-dig barriers to prevent predators or pests from gaining access to the enclosure, are recommended.

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

> **AZA Accreditation Standard**
>
> **(11.3.6)** Guardrails/barriers must be constructed in all areas where the visiting public could have contact with other than handleable animals.

<u>Kori bustard</u>: To prevent any physical interaction between kori bustards within their enclosures and members of the public outside the enclosure, there must be a guardrail/barrier that separates the two in addition to the primary containment suggested in section 2.2 (e.g., wire mesh). To prevent physical contact, and to provide birds with a greater sense of security within their enclosures, it is recommended that visitors be kept at least 1.5 m (5 ft.) from the perimeter of kori enclosures (S. Hallager, personal communication, 2006).

Walk through aviaries containing kori bustards are not recommended, as kori bustards are shy by nature. Hand-reared birds can also be aggressive to their caretakers (aggression can increase during breeding season), and consequently should not be allowed direct exposure to visitors in any situation.

Buff-crested bustard: Walk through aviaries for buff-crested bustards have been successful, provided there are areas for the bustards to retreat. To prevent physical contact, and to provide birds with a greater sense of security within their enclosures, it is recommended that the distance between the visitors and birds be no less than 3 m (10 ft.); greater distances are recommended for nervous birds. Well-planted aviaries will afford places to hide.

Exhibit safety: Any small holes that develop in the soil within any bustard enclosure (either from erosion or rodent activity) should be filled as soon as possible; bustard feet are small, and broken toes and legs can result if birds trip or fall in these holes. On a daily basis, keepers should also inspect the areas of the enclosure where birds have the closest access to the public, and immediately remove any foreign materials. Bustards will readily ingest objects such as nails, batteries, broken glass, and coins, and there can be serious health consequences associated with ingestion of these items (see section 7.6). Bustards should be regularly monitored for signs of impaction and zinc toxicity. Hand-reared bustards may tolerate a metal detector device held against the abdomen to check for the presence of ingested metal, especially if this is part of a husbandry training program (see section 9.1 for additional information on operant conditioning). Plants in and around enclosures should also be carefully selected to ensure that they do not have any poisonous properties, or do not pose any risk of physical injury to the birds (e.g., from thorns) (see section 6.2 for additional information on browse selection).

Pest and predator control: Keepers should check enclosures each day for signs of rodent activity. Spilled food should be removed on a daily basis to aid in rodent control. Poison should not be used inside bustard exhibits. If snap traps need to be set, they should be covered so that the birds are unable to see or reach the trap. Bustards are curious and will investigate a trap if they can see it.

Wild birds and rodents can pose a problem for bustards, especially at feeding time, as most bustards will not aggressively defend their food. Keepers may need to compensate for this by using special feeders to discourage wild birds and squirrels, or by providing extra food. However, providing extra food may exacerbate the initial problem and attract more pest species. For pelleted food (see section 6.2 for sample diets), feeders that have a platform that closes when a wild bird (e.g., starling, pigeon) lands on it work very well. For discouraging mice, large metal feeders that can hold a food pan work well. Where possible, it is advisable to work out a pest control program with a qualified pest control officer.

Predators: Native/feral predators are dangerous for both adult and young bustards. Foxes and/or raccoons have attacked adult birds (even adult kori bustards), sometimes fatally, and small chicks left out in unsecured exhibits are at great risk. All enclosures should be built to minimize predator access. Digging predators (e.g., dogs, foxes) can be excluded by burying the base of the boundary 0.3 m (1 ft.) in the ground. Surrounding the upper and lower portion of the enclosure with electrical wire can deter climbing predators (e.g., raccoons). In areas where large predators (e.g., coyote, bobcat, cougar, etc.) are common, birds may need to be housed indoors at night if pens are not covered. However, it should be noted that shifting and housing birds indoors each day might negatively affect breeding success (S. Hallager, personal communication, 2016).

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

Staff training for responses to emergencies must be undertaken, and records of such training maintained. Both animal care staff members and 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).

Transport crates should be readily available to move bustards

AZA Accreditation Standard

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

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).

in the event of a fire or other natural disaster that requires their immediate relocation. There should be one crate per bird to ensure that translocation can be performed quickly, if needed. For zoos in hurricane prone zones, birds should be housed in sheds or cement structures (e.g., basements, bathrooms) that can withstand hurricane force winds during the storm. Institutions should develop protocols that provide step-by-step instructions for where, how, when, and by whom birds should be moved. Non-perishable food and sufficient water should be left with the bustards in their shelters in the event staff cannot immediately service the birds after the storm.

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

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 Accreditation Standard

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

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.

Chapter 3. Records

3.1 Definitions

In the zoo and aquarium world, animal records are defined as "data, regardless of physical form or medium, providing information about individual animals, samples or parts thereof, or groups of animals". Most animals in zoo and aquarium collections are recorded as (referred to as) individuals, though some types of animals are recorded as (referred to as) groups or colonies of animals, particularly with invertebrates and in aquariums (see Appendix B for definitions and Recordkeeping Guidelines for Group Accessions). The decision about how to record its animals usually resides with each institution, but in certain cases, the AZA Animal Program Leader (i.e., TAG Chair, SSP Coordinator, or Studbook Keeper) may request that animals be recorded in a certain manner, whether as individuals or as groups. The Kori Bustard SSP®, Buff-crested Bustard SSP®, and the Gruiformes TAG manage all animals on the individual level.

3.2 Types of Records

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

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

For bustards, nothing beyond the normal record keeping of any bird species is necessary.

A designated paid staff member must be responsible for maintaining the animal record-keeping system and for conveying relevant laws and regulations to the animal care staff (AZA Accreditation Standard 1.4.6). Recordkeeping must be accurate and current (AZA Accreditation Standard 1.4.7). Complete and up-to-date animal records must be duplicated and stored at a separate location (AZA Accreditation Standard 1.4.4) and at least one set of historical records safely stored and protected (AZA Accreditation Standard 1.4.5). No special protocols are needed for effective records management of bustards.

AZA member institutions must inventory their bustard population at least annually and document all acquisitions, acquisitions, transfers, euthanasias, releases, and reintroductions (AZA Accreditation Standard 1.4.1). All bustards owned by an AZA institution must be listed on the inventory, including those animals on loan to and from the institution (AZA Accreditation Standard 1.4.2). All AZA-accredited institutions must abide by the AZA Policy on Responsible Population Management (Appendix

AZA Accreditation Standard

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

AZA Accreditation Standard

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

AZA Accreditation Standard

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

AZA Accreditation Standard

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

AZA Accreditation Standard

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

AZA Accreditation Standard

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

AZA Accreditation Standard

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

C) and the long-term welfare of animals should be considered in all acquisition, transfer, and transition decisions.

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 D), 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 bustard births and deaths should be reported to relevant AZA Bustard SSPs. All bustard shipments between institutions should be determined by the recommendations of the SSPs, and all breeding efforts should be based on the AZA Kori Bustard SSP®, Buff-crested Bustard SSP®, and AZA Gruiformes TAG Regional Collection Plan (RCP) in accordance with the dictates of the owner if a bird is on loan to a facility.

3.3 Permit Considerations

All bustards are regulated by the federal government. Bustards are also covered under the Wild Bird Conservation Act (WBCA). Any bird that is imported will require a WBCA permit. Depending on the agency involved, the application and approval process may take a few days to many months. These permits must be received by the applicant before the proposed possession or activity can occur. Both kori bustards and buff-crested bustards are listed in Appendix II of the Convention on International Trade in Endangered Species (CITES) and require a CITES export permit. State-specific permits may be required. Facilities should allow at least 6 months for permit application processing. Import of bustards into the U.S. may also require a USDA import permit. Contact USDA to determine if a permit is required. Import from certain countries may be prohibited. Therefore USDA should be contacted with the country of origin to confirm the permit requirements for any import.] https://www.aphis.usda.gov/aphis/ourfocus/animalhealth/animal-and-animal-product-import-information/ct_animal_imports_home

Paperwork required to accompany the animal includes, but may not be limited to: two copies of a health certificate from the shipper's veterinarian; animal health records with the animal(s) studbook and transponder number. USFWS and CITES permits may also be needed, and state-specific permits may be required. Animal data transfer forms, available through the American Association of Zookeepers (AAZK), should also be included. Labels for the outside of the crate showing the shipper's name and address, the receiver's name and address, and any additional instructions, should be attached to the crate.

3.4 Identification

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

> **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.

Intangible identifiers (called 'logical identifiers' in the Zoological Information Management System [ZIMS]): These include, but are not limited to, institutional accession number, house name, public name, studbook number, and ZIMS Global Accession Number.

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

A basic requirement for successful management of, and research on, bustards in zoos is individual animal identification. Techniques used to identify bustards include leg bands and passive induced transponders (PIT). Colored, metal, or plastic leg bands placed above the hock are recommended for those zoos with multiple birds, to aid in fast, easy identification. Bands should be placed above the hock to reduce ice buildup under the band in climatic zones that experience snow and ice during the winter. Transponders can be injected by syringe under the skin where they can be detected and read by an electronic scanner. For kori bustards, passive induced transponders can be placed in the inner crural region of the leg (Bailey & Hallager, 2003). Transponders can be inserted in the left pectoral muscle or thigh when kori bustards are one month old or even sooner depending upon the preferences of the attending veterinarians at each institution. Transponders for buff-crested bustards should be inserted when birds are closer to adult weight.

Chapter 4. Transport

4.1 Preparations

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.

AZA Accreditation Standard

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

Bustard transportation must be conducted in a manner that adheres to all laws, is safe, and minimizes risk to the animal(s), employees, and general public (AZA Accreditation Standard 1.5.11). All temporary, seasonal, and traveling live animal exhibits must meet the same accreditation standards as the institution's permanent resident animals, with foremost attention to animal welfare considerations (AZA Accreditation Standard 1.5.10). Safe animal transport requires the use of appropriate conveyance and equipment that is in good working order. Include copies of appropriate permits and authorizations in transport documentation. If the animal is not owned by the shipping institution, permission is to be obtained from the owner well in advance of the move.

One must check with the State Veterinarian of the destination state (and if shipped by ground, the states through which the bird travels) any relevant wildlife agencies. Birds imported from other countries may be subject to federal quarantine.

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

Transport crates: All crates for shipping bustards by air should meet International Air Transport Association (IATA) recommendations. Information on specific shipping recommendations related to bustards can be found in the IATA Live Animal Regulations. Bustards moved by ground should be contained in IATA approved crates since these provide the most secure containment for these taxa.

<u>Kori bustard</u>: Shipping crates should be made specifically for transporting kori bustards (typically necessary for transporting adult males). Crate size will vary according to the sex and age of the bird being shipped (see Table 7). Crates should not be too large. Kori bustards tend to be nervous during transport, and relatively close confinement will help them to retain their balance and reduce struggling. Regardless of crate type, the inside roof of the crate should be padded to protect the top of the bird's head. In general, it is recommended that a crate be padded on top, tall enough for the bird to stand upright, and just narrow enough to restrict a large amount of movement. It is not necessary for the kori bustard to be able to turn around easily within its crate. Air holes should be 2.54cm (1 in.) in diameter and around the entire top and bottom of the crate. The holes should be covered with window screening so that bills cannot poke through.

<u>Buff-crested bustard</u>: Shipping crates are typically plastic airline dog kennel of appropriate size (see Table 7). Crates should not be too large. Like kori bustards, buff-crested bustards tend to be nervous during transport, and relatively close confinement will help them to retain their balance and reduce struggling. The inside roof of the crate should be padded with a double layer of foam or soft material which can be affixed by heavy duty staples to protect the top of the bird's head (staples must be secure to prevent ingestion). In general, it is recommended that a crate be padded on top, tall enough for the bird to stand upright, and just narrow enough to restrict a large amount of movement. Crates should be dark with extra thick padding on the roof (e.g., two layers of padding).

Table 7. Recommended transport crate sizes for bustards

Sex/Age	Crate type	Crate dimensions (L x W x H)
Females kori bustard (all ages)	Wooden crate	86 cm x 53 cm x 83 cm (34 in. x 21 in. x 33 in.)
Juvenile kori bustard males (<6 months)	Wooden crate	86 cm x 53 cm x 83 cm (34 in. x 21 in. x 33 in.)
Adult kori bustard males (>6 months)	Wooden crate	86 cm x 53 cm x 83 cm (34 in. x 21 in. x 33 in.)

Male and female buff-crested bustard (adult)	Sky Kennel®	38.1 cm x 53.34 cm x 40.64 cm (15 in x 21 in x 16 in.)

Equipment and supplies: Transport protocols should be developed to ensure the safe transport of bustards between shipping and receiving institutions, and both institutions should have appropriate equipment and supplies to care for birds immediately before and after the birds are loaded onto aircraft or into transport vans for transport. For domestic shipments, sufficient diet should be shipped with the animal or ahead of the shipment to allow for a gradual transition to a new diet at the receiving institution. Note that food provided for international shipments will be subject to USDA regulations. Two people may be required to lift kori bustard crates on and off transport vans. Ensure crates stay level at all times to prevent birds from slipping or falling in the crates and move slowly.

Contingencies: 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).

Shipping birds via airlines is a common method for travel because it is the fastest. Direct flights via the shortest route should be scheduled whenever possible. Problems that could arise include delayed flights and/or missed flight connections. Flights should be carefully monitored, and the animal's location should be tracked. Contact the airline at once if any problems arise. It may be advisable to contact a colleague in the city of transfer to be on standby and ready to assist if problems arise. Bustards can overheat so birds should preferentially be transported in cooler months. In order to reduce the risk to bustards during transport, the following recommendations are useful:

- Obtaining the most direct flight
- Performing transports when temperatures are the best for species (seasonally and/or daily) in the area where they are coming from, as well as where they are going and at any transfer point
- Avoiding transports during breeding season (especially if female could be carrying eggs) or when a bird is molting
- Having an in-house protocol established for shipping
- Shipping eggs
- Shipping young birds 6-9 months of age

Capture myopathy: Both wild caught and captive reared bustards are susceptible to capture myopathy (Bailey, T. A. (2008). Along with other measures such as minimalizing handling and keeping temperatures low (e.g., shipping in early morning), supplementing birds with oral vitamin E can be utilized as a preventative measure. While supplementation has not been explicitly proven to reduce the incidence of capture myopathy, there is anecdotal evidence to suggest vitamin E administration has been helpful in other species and thus is recommended for kori bustards three days prior to shipment (S. Murray personal communication 2017). Facilities housing buff-crested bustards should evaluate the effectiveness of vitamin E therapy for this species.

4.2 Protocols

Transport protocols should be well defined and clear to all animal care staff.

General guidelines: Bustards should always be shipped singly in crates. There is no requirement for animal caretakers to accompany bustards during shipping (with the exception of egg transport), although some zoos have their own requirements for keepers to accompany birds on international flights. Where possible, bustards should not be held in a crate longer than 24 hours. Transport crates should not be opened during transport, unless there is a medical emergency, and it is recommended that they be opened under the supervision of a veterinarian at the shipping or receiving institution. Once the kennel has reached its final destination, but before releasing the birds, it is important to make sure that the birds have not been injured in shipment. Once that has been determined, the weight of the crate (still with a bird inside) should be taken and recorded. Once the bird has been released, weigh the crate again to get a baseline weight on the bird without having to restrain it. If a blood draw is scheduled for health screening, this should not be conducted on the first day of quarantine. Always allow the birds to recover from the stress of the travel to avoid accumulative stress and skewed results.

Equipment such as a medical kit and cellular phone should available to caretakers accompanying bustards during transport. Contact details for zoological institutions along the transport route (for ground transport) or at layover locations (for air transport) should also be available in case expert assistance is needed during the shipment, or if there is an unexpected delay.

Crating animals for transport: Protocols for successfully capturing and crating birds in preparation for transport are described in section 7.5. The use of operant conditioning (e.g. crate training) to train birds to enter crates without the need for capture is discussed in section 9.1.

Food and water: Food and water are not needed in the crate unless the bird will be held in the crate for more than 24 hours. Birds should be well fed prior to transport (the afternoon before, or the morning of shipment), as they may not eat immediately upon arrival at their new destination. Wild-caught birds, even those previously held in zoos, may not eat for several days after shipping. Hand-reared birds will likely eat within a day of arrival (if not sooner). Water should be immediately available once the bird has arrived at the receiving institution.

If it is known that transport will take longer than 24 hours, transport crates should be designed to allow food and water to be provided to the bird through appropriate access ports. For transport longer than 24 hours, bustards can be offered water in a small bowl placed in the crate. The water should be removed after an hour to prevent the bird from tripping. Pieces of watermelon can also be offered as a source of moisture, but the birds may not recognize these as food if watermelon is not part of their usual diet. Mice, sized appropriately for the bird/species and part of their normal diet, are also food items that can be provided during transport.

Substrate and bedding: The floor of the transport crate should be covered with a non-slip material such as indoor/outdoor carpeting that is glued to the crate bottom so that it does not move. Hay or straw should be placed on top of non-slip materials, as these materials do not provide enough traction when placed directly on a plywood or plastic floor. Bustards have very small feet compared to the size of their bodies, and can lose their balance easily. Hay and straw can also be used effectively to absorb urates and feces during transport.

Temperature, light, and sound: Kori bustards are large, and their body heat can increase the interior temperature of crates considerably. This is especially significant for shipping that takes place during summer months. For all bustard species, it is recommended that shipments be carried out early in the day, especially during the warmer summer months, so that the birds are not subjected to extreme temperatures while in their crates waiting to be loaded onto planes or trucks. Temperatures within the crate should not exceed 29.4 °C (85 °F) to ensure the safety and comfort of the birds. Low light conditions may help to minimize the stress associated with transport. The bird within the crate should not be able to see out of the crate easily. The shade-cloth material should not restrict ventilation within the crate. Under no circumstances should transport crates containing animals be left for any period of time in direct sunlight. During the ground transportation of bustards, all ground transport vehicles should be well ventilated, and the ambient temperature monitored so that the animals do not become over-heated or chilled. Placement of transport crates during shipments should avoid areas of excessive noise and commotion.

Post-transport release: When recovering from transport (or anesthesia within a crate), it is important to make sure the bird is safe from injury and has recovered completely before it is released, although release should occur as soon as possible after prolonged transport. Release the bird with the crate door directed towards an open space within the bird’s new enclosure (e.g., quarantine enclosure) that the bird can see. The bird should be allowed to step out of the transport crate of its own accord. Once the bird has moved away from the crate, the crate can be removed while the bird continues to be watched for any negative reactions associated with the shipping experience.

Chapter 5. Social Environment

5.1 Group Structure and Size

Careful consideration should be given to ensure that animal group structures and sizes meet the social, physical, and psychological well-being of both kori and buff-crested bustards, and facilitate opportunities for them to perform a full range of species-appropriate behaviors.

General group structure/size guidelines:

Kori bustard: By nature, kori bustards are generally solitary animals, except for females with chicks. In some instances, kori bustards in the wild have been observed to form small, semi-social groups during the non-breeding season (Hallager & Boylan, 2004). In the wild, chicks separate from their mother at the start of the following year's breeding season. In zoos, data are limited, but it is also recommended that young be removed from the enclosure before the onset of the next year's breeding season. In general terms, at least two kori bustards should be housed together (with the exception of two adult males who should not be housed together due to aggression) to allow for the opportunity of social interactions, but this will be dependent on the sex and temperament of the birds, as well as the space provided (e.g., while in winter housing). Maintaining single birds is not recommended on a long-term basis; optimum group size for kori bustards is 1–2 males and 2–3 females, depending on the enclosure facilities available. Adult males (>3 years old) are not compatible and should never be housed together.

Kori bustards show clear dominance hierarchies within genders. Adult birds (male and females) are dominant over juvenile birds. Kori bustard adult males are always dominant over females. Males generally show stable hierarchies over time. Where multiple males are housed together, the age of a male does not predict his rank (Hallager & Boylan, 2004). It is recommended that adult males be housed separately at all times (see below for exceptions to this recommendation). Female rank can change with breeding status, so dominant females during the breeding season may not remain so during the non-breeding season (Hallager & Boylan, 2004).

When females are housed together, it is recommended that each bird be provided with at least 250 m^2 (2,691 ft^2) of useable space, and that suitable visual barriers (e.g., thick shrubs) are available within the enclosure. Areas within enclosures that allow individuals to get out of sight from one another are advantageous (e.g., areas containing bushes, or low relief—see section 2.1), as they allow subordinate birds to hide from more dominant individuals. This will minimize aggressive interactions between the birds. Subordinate females can occasionally receive attacks on the head and neck from dominant birds (Hallager & Boylan, 2004). Care should be taken to monitor for actual physical injuries that result from dominance interactions.

See the *Ex situ* Kori Bustard Report on the ASAG website (https://de0046ae-67da-48da-9d60-85f9fe01aee2.filesusr.com/ugd/df9b5b_422089b2c6114efdbc81d4579544019c.pdf) for more information on the impact of social housing on kori bustard breeding behavior.

Buff-crested bustard: Little information is available on the natural social structure of breeding and non-breeding buff-crested bustards. The breeding system of this species is most likely monogamous (Johnsgard, 1991). Wild buff-crested bustards are not thought to form permanent pair-bonds, nor are they observed to maintain social groupings outside of the breeding season (Kemp & Tarboton, 1976). It is recommended that buff-crested bustards be bred in managed care as pairs, but there has been some breeding success with these bustards when housed as polygynous groups of three females a male in a large aviary (Siegel, Hallager, & Bailey, 2007). Juvenile buff-crested bustards can be housed in groups of up to five individuals (Bailey & Hallager, 2003).

Breeding season (see section 8.1 for more information):

Kori bustard: During the breeding season, multiple females can be housed with a single male, although females may become less compatible with one another during the breeding season. Previously, it was recommended to keep two males in the breeding flock so that they would stimulate each other into breeding behavior, as males do tend to display synchronously. However, recent breeding in flocks with only one male negates this strict recommendation. Animal caretakers should monitor male/male compatibly constantly if males are housed together in the non-breeding season. It is strongly recommended that adult males (>3 years old) be housed separately during the breeding season (regardless of their compatibility), because dominant males may kill or severely wound subordinate males.

Providing females with the opportunity to choose a mate has been attempted in some institutions by housing females in a central enclosure adjoined by two or more enclosures housing single males (Hallager & Boylan, 2004). The males are stimulated to display by the sight and sound of their competitors, and females have the option to select males by passing through a 75 cm x 57 cm (29.5 in. x 22.4 in.) opening to each enclosure with a male inside. The size of the opening is too small for males to pass through, although some males have become stuck in these holes trying to gain access to females, and have injured themselves (Hallager & Boylan, 2004). These openings should be used with extreme caution, if at all.

<u>Buff-crested bustard</u>: Females should have an area separate from the male for nesting, as breeding males will chase females from their nesting sites. It is unknown if multiple females can be housed together during the breeding season.

Non-breeding season:

<u>Kori bustard</u>: The appropriate size and composition of social groupings during the non-breeding season is dependent upon the age, sex, and personality of individual birds. Multiple females and one male can be housed year-round in the same enclosure. Juvenile males (<3 years) that have grown up together are generally compatible; although once they reach sexual maturity they should be separated.

<u>Buff-crested bustard</u>: The appropriate size and composition of social groupings during the non-breeding season is dependent upon the age, sex, and personality of individual birds. Multiple females and one male can be housed year-round in the same enclosure, but it is rare. Juvenile males (<3 years) that have grown up together are generally compatible, although once they reach sexual maturity aggression tends to escalate and the addition of a female will likely alter the compatibility of the males. In managed care facilities, it is safest to house adult males separately from other males year-round.

Winter housing and temporary holding: Winter housing is usually smaller or more restrictive than regular housing, although the complexity of the enclosure and the range of behavioral opportunities available to the birds should match that of their primary enclosures (see section 2.1).

<u>Kori bustard</u>: Social group options in smaller holding areas are dependent on individual bird personalities. If females are compatible, temporary holding areas that are 2.4 m x 2.4 m (8 ft. x 8 ft.) are sufficient. However, aggressively dominant females can harm subordinate females, especially when confined in small areas. Adult males should never be housed in the same holding stall together, even if they are considered compatible. However, juvenile males (<3 years) may be housed together depending on individual bird personalities. Aggressive males should always be housed alone. Multiple females can usually be housed with a single male unless the male is overly aggressive. Animal caretakers should rely on their knowledge of each bird's behavior when determining housing arrangements.

<u>Buff crested bustard</u>: Social group options in smaller holding areas are dependent on individual bird personalities. Adult females should not be housed together with the exception of dams with chicks. Chicks should be removed at day 45 to prevent aggression from the dam (M. Herry personal communication, 2016). Holding areas that are 2.1 m x 3.7 m (7 ft. x 12 ft.) are sufficient for one adult bird. Adult males should never be housed in the same holding stall together, even if they are considered compatible, with the exception of juvenile males (<5 months) and depending on individual bird personalities. Animal caretakers should rely on their knowledge of each bird's behavior when determining housing arrangements. At the National Avian Research Center (NARC), aggression between sibling groups of juveniles as young as 4–5 months was observed (National Aviary Research Center internal report).

5.2 Influence of Conspecifics and Other Species

Animals cared for by AZA-accredited institutions are often found residing with other animals of their species (conspecifics) but may also be found residing with other species. Consideration should also be given to the nature of other species housed next to bustard enclosures. Species that could injure or kill a bustard should not be housed adjacent to bustard enclosures if the potential exists for a bustard to escape and inadvertently enter a neighboring yard (e.g., an uncovered enclosure). Containment barriers between bustards and other species should also prevent any form of physical interaction between the animals.

Kori bustard: If two males are housed in adjacent enclosures during the breeding season, a visual barrier may need to be erected to prevent the dominant male from attempting to be aggressive towards the subordinate male through the physical barrier. The visual barrier will prevent the dominant bird from injuring himself, and may enhance a sense of security for the subordinate male. Some subordinate males may be sexually inhibited by the visual presence of the dominant male. In some cases, however, males may need only a physical barrier; some subordinate males have bred successfully within sight of the dominant male. Visual access may even act to stimulate one or both males to display/breed. The dynamics of each pair of dominant-subordinate males in their separate enclosures is different, and management should be adjusted to minimize aggression and promote breeding. If an adult male is not involved in breeding, it is not recommended to house the bird next to an exhibit containing females, especially when there is no visual barrier between the birds. A male's frustrated attempts to gain access to the females may lead to physical injuries or chronic stress.

Buff-crested bustard: Successful breeding by some buff-crested bustard males at the National Avian Research Center (NARC) may have been reduced when pairs of breeding birds were housed in adjacent exhibits allowing full visibility through a single layer of shade cloth (Anderson, 1998a). Interactions between males were observed, and this may have resulted in breeding interference. A double layer of shade cloth reduced visibility between the breeding pairs and the amount of interactions (e.g. chasing) between the males. Solid barriers to eliminate physical and visual interactions between breeding pairs of buff-crested bustards in adjacent enclosures should be considered.

Mixed-species exhibits: Some species have been housed successfully with bustards in mixed-species enclosures. Before integrating other species with bustards, it is recommended that AZA institutions with successful mixed-species exhibits be contacted to determine specific exhibit parameters necessary to keep all species involved safe and comfortable. The SSP Coordinator should also be contacted prior to integrating species for advice on compatibility.

- Kori bustard: Kori bustards and other species can only be mixed under certain circumstances. Facilities interested in housing kori bustards with any species should contact the SSP for specifics.
- Buff-crested bustard: Smaller bustard species can be exhibited successfully in mixed-species aviaries (Bailey & Hallager, 2003). However, animal caretakers must be aware that bustards are opportunistic carnivores, and their presence does pose a risk to small fledglings of other species. Species successfully exhibited with buff-crested bustards include barbets, rollers, pigeons, guinea fowl, waterfowl, waxbills, starlings, bee-eaters, wood-hoopoes, turacos, Egyptian plovers, robin chats, lovebirds, hamerkop, small hornbill species, African grey parrot, emerald starling, white crowned robin chat, common bulbul, cattle egret, blue-breasted kingfisher, magpie shrike, Hadada ibis, thick-knee, purple gallinule, Von der Decken's hornbill, African spoonbill, Abdim's stork, weaver, gonolek, Inca tern, scarlet ibis, roseate spoonbill, sunbittern, green wood hoopoe.

5.3 Introductions

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.

General guidelines for bustard introductions: The best time for introducing new birds together is during the non-breeding season (in the US, typically Sept-Feb). In the non-breeding season, birds are calmer and less aggressive, and long introduction periods are typically unnecessary. New birds should never be introduced to an enclosure occupied by other bustards during the breeding season, as levels of aggression are at their highest during this time. Introduction of new birds to a breeding flock will also negatively impact the breeding members of the flock, and the new birds will be unduly subjected to abnormal levels of aggression.

Ideally, an introduction protocol should consist of housing the new bird next to its intended exhibit mates for at least a few days, with a physical barrier that allows for visual contact, before full physical introductions are then attempted. The following step-by-step approach is recommended for all introductions. When this approach is not possible, a bird may need to be placed directly into the new situation with other animals. Although this procedure is not recommended, when it is used, it is crucial for animal caretakers to know individual bird characteristics so that they can identify which birds are more likely to become aggressive towards new arrivals. Sufficient staff members should be present to monitor the birds for the first few days of the initial introduction, as the dynamics of the social group will change. Specific recommendations for different types of introductions are provided below.

<u>Kori bustard male/female and female/female introductions</u>: When introducing a new male or female to another female (or group of females), the new animal should be housed in a pen that has visual (but not physical) access to the females for at least a few days prior to complete physical introductions. Males or females should not be introduced to an existing flock during the breeding season unless the flock is a non-breeding flock (e.g., males are not displaying, and females are not laying). Once new females are added, there will be a re-ordering of the social hierarchy. Animal caretakers should monitor aggression levels, and action may need to be taken if individuals show physical injuries (e.g., bleeding, lacerations) or limping and exhaustion from excessive chasing. The same approach can be taken when introducing a new female to a male. The new female should be housed in a pen so that she has visual (but not physical) access to the male for at least a few days, and introductions should not take place during the breeding season.

<u>Kori bustard introduction of a male to a male</u>: Males should be introduced by visual contact only during the non-breeding season. If physical introductions are attempted, an extended period (7–10 days) of visual contact is recommended, and a physical introduction should not be attempted if one male is persistently seen trying to be aggressive to the other male during the visual contact period. Adult males should never be housed together.

<u>Kori bustard introduction of juvenile birds (1–3 years) to adult birds</u>: When introducing young birds (male or females) to an existing flock, the new birds should be housed in a pen so that they have visual (but not physical) access to the other birds for at least a few days. Young birds should not be introduced to an existing flock during the breeding season unless the flock is a non-breeding flock. Young birds entering an established flock will likely be the most subordinate members of the flock, and so it is important that these birds be monitored closely for overly aggressive actions by other birds. Animal caretakers should expect some aggression to occur as the flock sorts out its new hierarchy, but this should decrease over time.

<u>Kori bustard introduction of hand-reared chicks to other chicks</u>: At 5 days, hand-reared chicks can be placed with other chicks provided that the oldest chick is less than two weeks old. Before 7 days of age, chicks should not be placed with chicks older than 2 weeks of age, as the older chicks have the capacity to injure severely (and possibly fatally) the younger chicks (Hallager & Boylan, 2004). All chicks can eventually be placed together when the youngest chick is at least 3 weeks old. When introducing young chicks together, the older chicks will typically be aggressive towards the younger chicks, but the period of aggression is usually limited to the first few hours after the introduction (Hallager & Boylan, 2004). Chicks should be observed carefully during the brief introduction period.

<u>Kori bustard introduction of birds to mixed-species enclosures</u>: If birds are to be placed in a mixed-species enclosure, the ideal approach would be to allow the kori bustards to set up residency in an empty enclosure first, and then slowly introduce other appropriate species to that enclosure.

<u>Buff-crested bustard male/female and female/female introductions</u>: When introducing a new male or female to another female (or group of females), the new animal should be housed in a pen that has visual (but not physical) access to the females for at least a few days prior to complete physical introductions. Males or females should not be introduced to an existing flock during the breeding season unless the flock is a non-breeding flock (e.g., males are not displaying, and females are not laying). Once new females are added, there will be a re-ordering of the social hierarchy. Animal caretakers should monitor aggression levels, and action may need to be taken if individuals show physical injuries (e.g., bleeding, lacerations) or limping and exhaustion from excessive chasing. The same approach can be taken when introducing a new female to a male. The new female should be housed in a pen so that she has visual

(but not physical) access to the male for at least a few days, and introductions should not take place during the breeding season.

Buff-crested bustard introduction of a male to a male: The safest situation is to house adult males separately even during the non-breeding season. Adult male buff-crested bustards are not compatible.

Buff-crested bustard Introduction of juvenile birds (1–2 years) to adult birds: When introducing young birds (male or females) to an existing flock, the new birds should be housed in a pen so that they have visual (but not physical) access to the other birds for at least a few days. Young birds should not be introduced to an existing flock during the breeding season unless the flock is a non-breeding flock. Young birds entering an established flock will likely be the most subordinate members of the flock, and so it is important that these birds be monitored closely for overly aggressive actions by other birds. Animal caretakers should expect some aggression to occur as the flock sorts out its new hierarchy, but this should decrease over time.

Buff-crested bustard introduction of hand-reared chicks to other chicks: At 5 days, hand-reared chicks can be placed with other chicks provided that the oldest chick is less than two weeks old. Before 7 days of age, chicks should not be placed with chicks older than 2 weeks of age, as the older chicks have the capacity to injure severely (and possibly fatally) the younger chicks (Hallager & Boylan, 2004). All chicks can eventually be placed together when the youngest chick is at least 3 weeks old. When introducing young chicks together, the older chicks will typically be aggressive towards the younger chicks, but the period of aggression is usually limited to the first few hours after the introduction (Hallager & Boylan, 2004). Chicks should be observed carefully during the brief introduction period.

Buff-crested bustard introduction of birds to mixed-species enclosures: If birds are to be placed in a mixed-species enclosure, the new animal should be housed in a covered pen or adjoining enclosure that has visual (but not physical) access to the other species for at least a few days prior to complete physical introductions. Well-planted aviaries that give the birds many places to hide from enclosure-mates are ideal. A particularly nervous individual may need a large space. Buff-crested bustards fly up to avoid danger and can be seriously injured by flying into the ceiling of shorter housing.

Chapter 6. Nutrition

6.1 Nutritional Requirements

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

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

Bustards are generally considered omnivorous—opportunistically taking prey items and plants that are locally abundant (Bailey & Hallager, 2003). In some environments, insects make up a large proportion of their diet (Bailey & Hallager, 2003). General observations of free-ranging kori bustards indicate that although they consume mainly insects, they also consume lizards, leaves, seeds, acacia gum, and flowers (Osborne, 1998; Osborne & Osborne, 1999). Mwangi (1988) recorded kori bustards in East Africa mainly consuming flowers, seeds, fruits, and pods. Insect prey consumed by wild kori bustards consists of Hymenoptera, Orthoptera, Coleoptera, and Lepidoptera. Non-insect prey consumed includes Chilopoda, Diplopoda, Annelida, and Reptilia. Most recently, Lopes, Fernandes, Medeiros, and Marini (2016) classified them as exudativores based on their regular consumption of acacia gum. The diet of wild buff-crested bustards is poorly studied, but individuals from this species are known to eat seeds, vegetation, berries, acacia gum, and insects (e.g., scarabs, beetle larvae, grasshoppers, ants) (Siegel, Hallager, & Bailey, 2007).

Information on the gastrointestinal tract (GIT) of various bustard species is described in detail by Bailey, T. A., Mensah-Brown, E. P., Samour, J. H., Naldo, J. L., Lawrence, P., & Garner, A. (1997), who characterize the functional elements of the GIT for kori bustards as those of opportunistic omnivores. Bustards lack a crop, although their long esophagus and longer proventriculus and ventriculus may serve a functional role in food storage. Bustards are commonly found with stones in their ventriculi. These may be purposefully ingested and play a role in grinding up food within the ventriculus (Bailey, T. A., Mensah-Brown, E. P., Samour, J. H., Naldo, J. L., Lawrence, P., & Garner, A., 1997).

<u>Kori bustards</u>: Although the GI tract of the kori bustard is typical of an insectivorous bird (Maloiy, Warui, & Clemens, 1987), they have been historically fed in zoos as "primarily carnivorous" omnivores (Hallager & Boylan, 2004). Most kori bustard diets in managed-care facilities are a combination of mice, beef or horsemeat, and avian nutritionally complete pellets (e.g., gamebird or crane pellets), along with a variety of produce items and insects. All food items provided within the diet should be included in the nutritional analysis of the complete diet. Whereas free-ranging insectivores have a myriad of insect prey choices, the variety of commercially available insects is limited. It is important that the nutrient content of the insects chosen is known. There are excellent references that provide the nutrient content of invertebrate prey (Bernard & Allen, 1997), and these should be carefully reviewed when formulating a diet for kori bustards. 'Gut-loading' has been the method of choice to improve the nutrient profile of commercially available insects, but dusting insects with a vitamin/mineral mix can also be an effective strategy (Bernard & Allen, 1997). For mother-reared chicks, insects are always the preferred food item, although dams will also feed pinkies and meatballs (beef or horsemeat mixed with avian pellets) to chicks. Pinkie mice should be used instead of full-grown mice in the mother's diet, until the chick weighs 1 kg (2.2 lb.), as chicks that ingest too much mouse fur can experience gut impaction. Giant mealworms should also be removed from the adult's diet to prevent them from being fed to the chicks, and the back legs of any crickets that are provided should be removed.

<u>Buff-crested bustards</u>: In managed care, general food items provided to buff-crested bustards include nutritionally complete pelleted or extruded feeds (e.g., production pellets, Mazuri® Zulife Softbill, Marion® Scenic Apple Paradise, dry dog food), ground meat (e.g., beef, fortified meat mixes), whole prey (e.g. pinkie or fuzzy mice), invertebrates (e.g. crickets, mealworms), chopped fruit and vegetables (e.g., fruits,

carrots, greens), and some institutions add vitamin and mineral supplements (e.g. Vionate, Osteoform). Analysis in February 2018 of diets provided at U.S. AZA institutions found concentrated feeds and produce comprise the majority of diet items provided, with pinkie/fuzzy mice, meat mixes, and invertebrates offered as a smaller proportion of diets. However, assessment of consumption at these zoos finds the most consistently consumed food items are whole prey, invertebrates, meat mixes, and dry dog food; most avian pellets are only partially consumed, and fruits and vegetables appear to be the least preferred items. Quantities of items fed to buff-crested bustards in U.S. zoos appear to vary widely, with diets offered providing a range of 88–201 g/d per bird (135–428 kcal/d per bird); diets as consumed appear to range even more widely, from 39–190 g/d per bird (87–400 kcal/d per bird). Given the variability in the amounts reportedly fed and consumed, regularly weighing individual birds will help monitor and evaluate adequate diet quantities. Based on the variety of appropriate food items for this species, several combinations of food categories and items are possible to build a diet that meets nutrient targets (see Tables 16, 17, and 18 below). However, care must be taken to consider all food items (including those offered as enrichment) in the nutritional analysis of the diet, and to account for the diet as consumed to prevent birds from selecting against fortified feeds (e.g. pellets, meat mixes).

The target nutrient levels established for bustard diets were derived from several domestic and exotic species (e.g., pheasants, quail, geese, and cranes) (National Research Council [NRC], 1994; Anderson, 1995). In the table below, target values are expressed as ranges, the low end represents a maintenance requirement, the high end (marked by a double asterisk) represents a breeding requirement (Table 8). Growth requirements tend towards the high end of the range.

Table 8. Proposed nutrient guidelines for kori and buff-crested bustards on a dry matter basis*

Nutrient	Proposed nutrient guidelines, 2004
Protein, %	16.5–30.0**[1]
Fat, %	---
Crude Fiber, %	---
Ca, %	0.66–2.75**
P, %	0.33–1.0**
Ca:P	---
K, %	0.44–0.72
Na, %	0.13–0.18
Mg, %	0.05–0.06
Cu, mg/kg	5.5–8.8
Fe, mg/kg	55–77
Zn, mg/kg	55–70
Mn, mg/kg	66–72
Se, mg/kg	0.2
I, mg/kg	0.33–0.44
Vitamin A, IU/kg	1650–5500
Vitamin D3, IU/kg	220–1200
Vitamin E, IU/kg	11.0–27.5
Thiamin, mg/kg	2.2
Riboflavin, mg/kg	2.75–4.4
Pyridoxine, mg/kg	3.3–5.0
Vitamin B12, mcg/kg	3–10
Biotin, mg/kg	0.11–0.25
Choline, mg/kg	990–1650
Folacin, mg/kg	0.8–1.1
Niacin, mg/kg	22–71.5
Pantothenic acid, mg/kg	10.5–17.6

*Target values based on NRC (1994) and Anderson (1995).

**Values at high end of range for breeding only.

[1]Available data indicate that breeding diets for kori bustards that contain 26.4% crude protein on a dry matter basis should be adequate (Hallager et al., 2002). Recommendations for dietary crude protein levels of no more than 24% on a dry matter basis for growing sandhill cranes (Serafin, 1982) may also be appropriate for growing kori bustards.

There are currently no established bustard-specific daily energy requirements or energy requirement calculations/equations, but available equations based on similar-sized or comparable foraging strategy birds (Nagy et al 1999, Lasiewski and Dawson 1967, Swengel and Carpenter 2000) yield possible maintenance energy requirement ranges of 360-1436 kcal/d for kori bustards (360-752 kcal/d for females weighing 5-7 kg and 641-1436 kcal/d for males weighing 11-17 kg) and 81-169 kcal/d for buff-crested bustards (body weight range 650-900 g). Additional research that focuses on exact daily food intake, energy expenditure, and diet at all life stages (e.g., chick, juvenile, reproductive adult, senescent adult), will be important to developing more specific nutritional and energy requirements and recommendations.

Factors influencing nutritional requirements: The following factors affect the nutritional requirements of bustards and should be carefully considered when formulating appropriate diets.

Hand-reared chicks: Limited published data are available regarding successful hand-rearing diets for kori bustard chicks (Maslanka & Ward, 2003; Hallager, 2005) or buff-crested bustards. However, nearly all hand-reared kori bustard chicks prior to 2008 have developed angel wing, a condition linked to high protein diets in waterfowl and cranes (Serafin, 1982; Kear, 1986). Growth rates of previously hand-reared kori bustard chicks that developed angel wing ranged from 5.7–8.1% (mean 6.8%) of body weight on a daily basis (Hallager et al., 2002). Whereas fast growth is important for the production of commercial birds with significant muscle mass, it is not the goal for exotic birds housed in zoos. Angel wing in waterfowl and cranes can be successfully “treated” by reducing the crude protein content of the diet offered if caught early enough in development. For hand-reared kori bustards, dietary protein levels that facilitate normal growth in waterfowl and cranes may be most appropriate. It is recommended that diets for hand-reared kori chicks contain between 18–22% crude protein on a dry matter basis, and growth should not exceed 5% of body weight per day, to avoid angel wing.

Reproductive status: If their diet does not already meet the upper end of calcium needs (Table 8), breeding female bustards should be supplemented with calcium starting at least one month prior to the beginning of egg laying. Appropriate vitamin E levels are essential for successful reproduction in most bird species, and deficiency of this vitamin has been associated with both low fertility and low hatchability (Dierenfeld, 1989). Chicks and juvenile birds may also have a higher requirement for vitamin E, as researchers have found plasma concentrations of vitamin E to be lower in juvenile birds despite a higher vitamin E intake by these juveniles in their diets (Anderson et al., 2002).

Season: In northern latitudes, where temperatures regularly fall below 0 ºC (32 ºF), birds should be offered more food in colder months than in warmer months to accommodate a higher caloric requirement. As such, foods high in fat (e.g., peanuts or peanut butter) can be offered as occasional enrichment items during winter months. Increases in diet amounts may be based on observed behavior changes or increased consumption, but should be made in coordination with institution veterinarians and nutritionists. As with most birds, protein requirements for bustards increase during the breeding season, and birds should be placed on a breeder pellet instead of a maintainer pellet. Females should be provided additional calcium, which may be in the form of a nutritionally complete avian breeder feed, oyster shell, or supplemental calcium mixed into a fortified meat mix or produce.

6.2 Diets

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

Recommended diets for bustards in captivity include nutritionally complete feed(s), whole prey (vertebrate and invertebrate), and produce. An example of a typical zoo diet following these recommendations might include apple, cabbage, mealworms and crickets, mice, beef or horsemeat, and a formulated gamebird pellet (Bailey & Hallager, 2003).

Sample diets: Based on the reported omnivorous foraging strategy of free-ranging bustards and available appropriate food items, proposed diet proportion guidelines are presented in Tables 9 and 10. These guidelines can assist with diet formulation by proportion, in order to ensure that nutrient needs are met and levels of specific nutrients are not grossly exceeded (i.e., protein). Diets can be formulated by using the table to select the desired proportions of items present in smaller amounts (e.g., vertebrate prey, invertebrate prey, and produce), and then a nutritionally complete food (see Tables 11 and 15) can be used to round out 100% of the total diet. The total amount of food required will vary based on individual bird size, seasonal factors (see section 6.3), feeding environment (e.g. mixed-species exhibits, access to food by wild animals in enclosure) and other considerations. A review of diets fed at U.S.

institutions suggests 350-700 grams daily total food may be sufficient for kori bustards (approximately 350-500 g/d for females and 400-700 g/d for males), and approximately 40-200 grams daily total food may be sufficient for buff-crested bustards. Diets higher in more energy-dense foods like nutritionally complete pellets would facilitate feeding less total food than diets higher in less-dense foods like produce and prey items. Bustard activity level remains fairly stable year-round, and food amounts are likely to remain stable throughout the year.

Table 9. Kori bustard recommended diet proportion guidelines (as fed basis)

Item	Minimum % of diet	Maximum % of diet
Vertebrate prey	0	25
Invertebrate prey	5	30
Nutritionally complete feeds*	40	55**
Produce	10	20

*Nutritionally complete feeds are those designed to meet specific recommended nutrient levels. Specifications are provided in section 6.1 (Table 8).

**Diets that exceed 55% complete feeds can be considered. A diet comprised of 75% complete feed has successfully maintained kori bustards in zoos and aquariums (Anderson, 1995).

Table 10. Buff-crested bustard recommended diet proportion guidelines (as fed basis)

Item	Minimum % of diet	Maximum % of diet
Vertebrate prey	0	35
Invertebrate prey	1–2	15
Nutritionally complete feeds*	30	55**
Fortified meat mixes	0	45–50
Produce	5	20

*Nutritionally complete feeds are those designed to meet specific recommended nutrient levels. Specifications are provided in section 6.1 (Table 8).

**Diets containing 55% complete feeds should consider a combination of feeds including a dog food, based on reported consumption preferences of this species in U.S. zoos, as >40% of a single non-dog food nutritionally complete feed may result in significant waste (analysis of U.S. institution BCB diets for this ACM).

Nutritionally complete items should comprise the backbone of the diet. There are a range of nutritionally complete foods that are capable of meeting the nutritional requirements of kori bustards, and that can be successfully included within their diets with approval from institutional veterinarians and nutritionists. See Tables 11 and 15 for examples of nutritionally complete feeds capable of meeting target nutrient values within the framework provided by the recommended diet proportions listed in Tables 9 and 10 above. All of these feeds may be used for any bustard species but are listed separately based on diets reportedly fed to kori and buff-crested bustards (differences in the size of these species likely influence which of these feeds each species prefers or will consume). Inclusion in these tables does not indicate specific endorsement of any of these products.

Table 11. A sample of nutritionally complete feeds suitable for kori bustard diets (as recommended by institutional veterinarians and nutritionists) as part of the overall diet (values on a dry mater basis)

Nutrient	Mazuri® Exotic Gamebird Maint[1]	Zeigler® Avian Maint[2]	Mazuri® Waterfowl Maint	Mazuri® Ratite	Zeigler® Ratite Gr/Mai	Zeigler® Crane Breeder	NARC Production Pellet[3]	Kock Pellet[4]
Protein, %	13.9	13.9*	15.6	16.7	17.8*	24.4*	24.3	17.8-22.1
Fat, %	3.6	2.2*	4.1	4.6	4.4*	5.6*	-	-
Ca, %	0.9	0.9	1.3	1.8	1.0	3.1	3.7	1.4-3.9
P, %	0.3	0.6	0.4	0.9	0.8	0.9	0.9	0.9-1.9
Ca:P	3.0	1.5	3.2	2.1	1.25	7.8	4.6	1.7-2.1
K, %	0.6	0.6	0.6	0.8	1.3	0.8	0.9	0.9-1.0
Na, %	0.1	0.1	0.2	0.3	0.2	0.3	0.2	0.4-0.5
Mg, %	0.2	0.2	0.2	0.3	0.3	0.3	0.4	0.3-0.4
Cu, mg/kg	10.0	11.5	10.0	20.0	24.4	16.8	16.6	9.6-9.5
Fe, mg/kg	144.4	158.9	155.6	455.6	282.4	348.2	442.9	131.1-191.8

Nutrient	Mazuri® Exotic Gamebird Maint[1]	Zeigler® Avian Maint[2]	Mazuri® Waterfowl Maint	Mazuri® Ratite	Zeigler® Ratite Gr/Mai	Zeigler® Crane Breeder	NARC Production Pellet[3]	Kock Pellet[4]
Zn, mg/kg	100.0	49.1	101.1	142.2	175.7	136.9	72.8	69.5-167.2
Mn, mg/kg	111.1	48.5	108.9	122.2	192.1	147.1	80.6	70.7-139.4
Se, mg/kg	0.5	0.4	0.5	0.6	0.6	0.5	0.3	0.2
I, mg/kg	1.2	0.6	1.3	1.2	0.6	0.4	0.9	1.4-2.3
Vitamin A, IU/g	6.7	8.2	10.8	11.3	24.6	19.2	13.8	6.6-7.1
Vitamin D3, IU/g	2.5	0.6	2.5	1.7	1.5	2.1	3.3	1-3
Vitamin E, IU/kg	138.9	140.5	133.3	188.9	173.8	78.6	37.2	75.6-222.0
Thiamin, mg/kg	11.0	6.7	6.9	9.6	12.3	16.3	5.9	12.4-16.2
Riboflavin, mg/kg	6.1	5.7	3.4	11.1	9.4	14.3	8.4	8.6-13.8
Pyridoxine, mg/kg	6.4	10.1	5.4	6.7	126.3	15.2	8.8	9.2-13.7
Vitamin B12, mcg/kg	16.7	^	10	2	^	^	2	40-50
Biotin, mg/kg	0.4	0.4	0.3	0.5	0.5	0.7	0.2	0.3-1.2
Choline, mg/kg	7888.9	1751.7	1133.3	1555	1637.6	2188.7	896.4	976-1494
Folacin, mg/kg	3.0	2.0	1.7	6.3	5.4	5.3	1.1	2.5-9.4
Niacin, mg/kg	103.3	88.3	86.7	121.1	126.3	136.2	68.2	75.5-88.4
Pantothenic acid, mg/kg	11.1	29.1	20.0	28.9	41.1	40.2	25.1	34.7-50.8

[1]PMI Nutrition International, LLC. Brentwood, MO 63144
[2]Zeigler Brothers, Gardners, PA 17324
[3]Production Pellet (Anderson, 1995).
[4]Richard A. Kock Pelleted Diets (Kock, 1990). Values expressed as a range of maintenance-breeder.
*Value represents guaranteed minimum
^Missing values unavailable from manufacturer.

The diets listed in Table 12 (see Table 13 for nutrient analyses of the example diets) are not recommended diets, but examples of how the proportions listed in Table 9 can be used to formulate diets that meet nutrient guidelines. A variety of ingredients can be chosen based upon availability, palatability, and management needs.

Table 12. Examples of kori bustard diets using recommended diet proportions

Diet	Vertebrate prey	Invertebrates (Crickets)	Nutritionally complete foods	Produce
1	25% (mice)	25%	35% (Zeigler® Avian Maintenance[1])	15%
2	15% (mice)	25%	40% (Mazuri® Waterfowl Maintenance[2])	20%
3	0%	25%	55% (Zeigler® Ratite Grower/Maintenance[1])	20%
4	20% (mice)	25%	55% (Mazuri® Exotic Gamebird Maintenance[2])	0%
5	10% (beef)	0%	75% (Kock Kori Production Pellets[3])	15%

[1]Zeigler Brothers, Gardners, PA 17324
[2]PMI International, LLC, Brentwood, MO
[3]Kock, 1990

The following table (Table 13) provides a nutrient analysis of each of the example diets listed in Table 12 above, with a comparison to the proposed nutrient guidelines (Table 8) listed in section 6.1.

Table 13. Example diets (see Table 12) that meet proposed nutrient guidelines (as of 2004) for kori bustards (analysis on a dry matter basis)

	Example diets[1]					Proposed nutrient guidelines, 2004[2]
Nutrient	Diet 1	Diet 2	Diet 3	Diet 4	Diet 5	
Protein, %	29.1	27.9	24.3	21.2	23.6	16.5–30.0**
Fat, %	7.5	7.6	5.5	4.7	2.8	---
Crude Fiber, %	5.3	5.0	14.4	5.3	4.3	---
Ca, %	1.1	1.2	0.9	0.8	3.6	0.66–2.75**
P, %	0.9	0.8	0.9	0.7	1.8	0.33–1.0**

	Example diets[1]					Proposed nutrient guidelines, 2004[2]
Nutrient	Diet 1	Diet 2	Diet 3	Diet 4	Diet 5	
Ca:P	1.2	1.5	1.0	1.1	2.0	---
K, %	0.6	0.7	1.4	0.7	1.1	0.44–0.72
Na, %	0.1	0.1	0.2	0.1	0.5	0.13–0.18
Mg, %	0.1	0.2	0.3	0.2	0.4	0.05–0.06
Cu, mg/kg	13.1	13.8	25.6	14.2	8.9	5.5–8.8
Fe, mg/kg	187.4	107.7	291.0	264.4	182.0	55–77
Zn, mg/kg	93.2	121.6	194.9	114.7	158.5	55–70.1
Mn, mg/kg	42.7	83.4	183.1	5.3	127.1	66–72
Se, mg/kg	0.3	0.3	0.8	0.4	0.2	0.2
Vitamin A, IU/g	7.9	11.4	5.5	8.8	7.8	1.65–5.5
Vitamin D3, IU/g	0.5	1.8	1.4	2.1	2.7	0.22–1.2
Vitamin E, IU/kg	105.8	96.1	162.3	118.0	203.8	11.0–27.5
Thiamin, mg/kg	5.1	5.1	11.5	8.5	15.1	2.2
Riboflavin, mg/kg	4.4	2.7	8.9	5.3	13.0	2.75–4.4
Pyridoxine, mg/kg	7.6	4.0	11.9	5.5	12.8	3.3–5.0
Vitamin B12, mcg/kg	10	10	20	10	3	3–10
Biotin, mg/kg	0.3	0.2	0.4	0.4	1.1	0.11–0.25
Folacin, mg/kg	1.6	1.5	5.4	2.8	0.1	0.8–1.1
Niacin, mg/kg	66.8	63.0	118.7	88.1	88.8	22–71.5
Pantothenic acid, mg/kg	22.1	14.8	38.8	9.8*	47.3	10.5–17.6

[1]See Table 12
[2]See Table 8
*Values generated as a result of missing values in database.
**Values at high end of range for breeding only.

Kori bustards and buff-crested bustards at the National Aviary Research Center (Abu Dhabi) were successfully maintained on a diet composed of 75% (as fed) nutritionally-complete pellet formulated specifically for bustards (recipe and ingredients not available), with the overall diet providing 390 g/d total food per kori bustard and 46 g/d total food per buff-crested bustard (Sleigh & Samour, 1996). While a diet this high in pelleted feed is not representative of diets fed to either species in the U.S. institutions and likely is not practical based on reported consumption of pelleted feeds by these species in U.S. institutions, it may be considered and would meet nutrient targets for all bustards if consumed. It is possible the bustard-specific pellet used at the NARC was more palatable to bustards than available avian pellets in the U.S..

Table 15. A sample of nutritionally complete feeds suitable for buff-crested bustard diets (as recommended by institutional veterinarians and nutritionists) as part of the overall diet (values on a dry mater basis)**

Nutrient	**Mazuri® Exotic Gamebird Maint. 5643/ Breeder 5639[1]**	**Mazuri® Crane Diet 5646[1]**	**Mazuri® Parrot Breeder 56A9[1]**	**Mazuri® Zulife Softbill 5MI2[1]**	**Marion® Scenic Paradise or Jungle Apple[2]**	**Purina® One Small Bites dog food[3,*]**
Protein, %	13.0–21.7	21.5	21.1	22.2	24.0	29.1
Fat, %	2.7–5.1	5.4	7.2	8.9	8.5	13.4
Crude Fiber, %	5.0–7.6	7.5	5.0	5.6	4.2	3.3
Ca, %	0.9–3.0	1.6	1.3	1.6	1.2	1.5
P, %	0.7–1.0	1.1	1.0	0.9	0.8	1.1
Ca:P	1.3–2.9	1.5	1.4	1.7	1.5	1.4
K, %	0.7–1.0	1.1	0.7	0.6	0.7	0.7
Na, %	0.2	0.3	0.1	0.2	0.3	0.4

Nutrient	Mazuri® Exotic Gamebird Maint. 5643/ Breeder 5639[1]	Mazuri® Crane Diet 5646[1]	Mazuri® Parrot Breeder 56A9[1]	Mazuri® Zulife Softbill 5MI2[1]	Marion® Scenic Paradise or Jungle Apple[2]	Purina® One Small Bites dog food[3,*]
Mg, %	0.2	0.3	0.2	0.1	0.2	0.1
Cu, mg/kg	10.8–17.3	21.5	15.6	18.9	19.7	15.2
Fe, mg/kg	163.0–373.8	284.5	388.9	97.2	186.9	403.7
Zn, mg/kg	97.8–130.0	161.0	111.1	77.8	110.4	256.8
Mn, mg/kg	114.1–162.5	150.3	111.1	94.4	50.3	70.7
Se, mg/kg	0.3	0.3	0.2	0.2	0.38	0.57
I, mg/kg	1.4–1.8	1.8	1.4	0.9	1.1	2.5
Vitamin A, IU/g	8.7–19.9	12.5	14.9	7.0	15.1	16.6
Vitamin D3, IU/g	2.3–2.4	4.7	2.0	2.1	3.9	2.5
Vitamin E, IU/kg	114.1–167.9	214.7	177.8	316.7	344.3	646.9
Thiamin, mg/kg	8.7–10.3	18.2	11.1	8.9	8.5	21.1
Riboflavin, mg/kg	5.1–10.3	12.9	15.6	15.6	9.8	15.1
Pyridoxine, mg/kg	6.1–8.6	15.0	12.2	16.7	9.6	12.9
Vitamin B12, mcg/kg	15.2–22.8	52.6	45.6	68.9	29.5	87.1
Biotin, mg/kg	0.2–0.4	1.2	0.8	1.0	0.4	0.2
Choline, mg/kg	1032.6–1543.9	2012.7	1416.7	2005.6	1532.2	1402.6
Folacin, mg/kg	3.0–3.1	9.4	5.0	4.8	5792.3	4.7
Niacin, mg/kg	70.7–82.3	67.6	95.6	94.4	74.3	130.6
Pantothenic acid, mg/kg	10.8–20.6	49.4	24.4	21.1	26.2	39.2

[1]PMI Nutrition International, LLC. Brentwood, MO 63144. Values expressed as a range of maintenance-breeder where both versions exist for a feed.
[2]Marion Zoological Inc., Plymouth, MN 55447. Values reflect analysis at time of manufacture.
[3]Nestle Purina Petcare Company, St. Louis, MO 63164
*Given as representative example of a suitable dry dog food; any commercial dog food meeting AAFCO guidelines should have approximately similar nutrient profile
**Any of the feeds listed in Table 11 are also suitable for buff-crested bustards
^Missing values unavailable from manufacturer.

The diets listed in Table 16 (see Table 17 for nutrient analyses of the example diets) are not recommended diets, but examples of how the proportions listed in Table 10 can be used to formulate diets that meet nutrient guidelines. A variety of ingredients can be chosen based upon availability, palatability, and management needs.

Table 16. Examples of buff-crested bustard diets using recommended diet proportions

Diet	Vertebrate prey	Invertebrates (crickets, worms)	Fortified meat mixes	Nutritionally complete foods	Produce
1	0%	15%	45% (Milliken™ Small Carnivore[1])	30% (Mazuri® Crane Diet[2])	10%
2	10% (fuzzy mice)	10%	35% (Nebraska® Bird of Prey[3])	40% (Mazuri® Zulife Softbill[2])	5%
3	5% (pinkie mice)	2%	25% (Nebraska® Bird of Prey)	55% (Mazuri® Exotic Gamebird Breeder[2] + Purina One Small Bites[4] dog food)	13%
4	20% (fuzzy mice)	10%	0%	50% (Marion® SPA + Mazuri Exotic Gamebird Maintenance[2])	20%
5	35% (fuzzy mice)	5%	0%	45% (Mazuri® Parrot Breeder[2] + Purina® One Small Bites)	15%

*Range of Maintenance-Breeder diets
[1]Milliken Meat Products, Ltd; Markham, ON L6G 1C4, Canada
[2]PMI International, LLC, Brentwood, MO 63144
[3]Central Nebraska Packing, Inc., North Platte, NE 69103
[4]Nestle Purina Petcare Company, St. Louis, MO 63164
[5]SPA=Scenic Paradise Apple; Marion Zoological, Inc., Plymouth, MN 55447

The following table (Table 17) provides a nutrient analysis of each of the example diets listed in Table 16 above, with a comparison to the proposed nutrient guidelines (Table 8) listed in section 6.1.

Table 17. Example diets (see Table 16) that meet proposed nutrient guidelines (as of 2004) for buff-crested bustards (analysis on a dry matter basis)

	Example diets[1]					Proposed nutrient guidelines, 2004[2]
Nutrient	Diet 1	Diet 2	Diet 3	Diet 4	Diet 5	
Protein, %	36.4	31.7	29.1	21.7	28.6	16.5–30.0*
Fat, %	15.8	14.7	10.4	9.2	13.8	---
Crude Fiber, %	5.2	4.7	4.9	4.2	3.3	---
Ca, %	1.20	1.38	2.08	0.95	1.34	0.66–2.75*
P, %	0.88	0.97	1.05	0.72	1.01	0.33–1.0*
Ca:P	1.36	1.42	1.99	1.32	1.33	>1.0
K, %	1.02	0.7	0.92	0.75	0.78	0.44–0.72
Na, %	0.28	0.27	0.35	0.22	0.33	0.13–0.18
Mg, %	0.22	0.11	0.17	0.19	0.13	0.05–0.06
Cu, mg/kg	15.0	15.90	14.8	14.6	14.6	5.5–8.8
Fe, mg/kg	209.6	121.6	342.4	157.2	338.1	55–77
Zn, mg/kg	140.3	82.4	165.0	99.5	179.9	55–70.1
Mn, mg/kg	92.5	67.3	99.3	69.8	68.9	66–72
Se, mg/kg	0.41	0.32	0.43	0.33	0.38	0.2
Iodine, mg/kg	1.1	0.6	1.7	1.1	1.7	0.33–0.44
Vitamin A, IU/g	20.8	11.5	24.4	25.1	27.2	1.65–5.5
Vitamin D3, IU/g	3.3	1.6	2.1	2.6	1.8	0.22–1.2
Vitamin E, IU/kg	268	326	372	211	429	11.0–27.5
Thiamin, mg/kg	10.8	9.4	13.4	8.1	14.5	2.2
Riboflavin, mg/kg	13.8	11.6	10.2	10.0	13.0	2.75–4.4
Pyridoxine, mg/kg	10.3	12.4	8.8	7.7	11.1	3.3–5.0
Vitamin B12, mcg/kg	13.8	11.6	10.2	10.0	13.0	3–10
Biotin, mg/kg	0.74	0.67	0.17	0.38	0.31	0.11–0.25

	Example diets[1]					Proposed nutrient guidelines, 2004[2]
Nutrient	Diet 1	Diet 2	Diet 3	Diet 4	Diet 5	
Choline, mg/kg	1664	2174	1575	1374	1315	990–1700
Folacin, mg/kg	5.62	3.40	3.11	3.80	3.98	0.8–1.1
Niacin, mg/kg	51.2	69.0	84.7	68.0	99.8	22–71.5
Pantothenic acid, mg/kg	35.9	18.6	24.2	20.3	30.4	10.5–17.6

[1]See Table 16
[2]See Table 8
*Values at high end of range for breeding only.

Diet presentation and preparation:

When bustards are provided their diets, food items can be offered in pans, tubs, buckets, platforms, etc., or hand-fed to individual birds in a group. Pellet dispensers should be placed in pens to encourage the consumption of appropriate dry, nutritionally complete feeds. Dispensers and other feeding presentations should be designed to minimize the consumption of the bustard food by pest species (Hallager & Boylan, 2004). During feeding sessions, dominant birds may displace subordinate birds from the food with short chases and sometimes even biting. During the breeding season, subordinate females can be displaced from food dispensers by the dominant female and/or dominant male. It may be necessary to provide several, well-spaced feeders to allow all birds to have access to the complete diet.

Generally, bustards are easy to medicate by putting a pill or liquid inside of a dead mouse. There are times when birds will refuse the medicated item, however, and alternative approaches need to be considered such as: peanuts in the shell (see Table 19), earthworms (work well for thin liquid medications), large mealworms, banana, grapes, horsemeat meatballs, or a compounded, flavored medication (when possible). It is recommended that animal caretakers become familiar with favored food items before a bird becomes ill, so that appropriate food that will be readily accepted by sick/injured birds can be provided, thus increasing the likelihood that medication will be taken successfully.

> **AZA Accreditation Standard**
>
> **(2.6.1)** Animal food preparations must meet all local, state/provincial, and federal regulations.

Bustard food preparation must be performed in accordance with all relevant federal, state, or local regulations (AZA Accreditation Standard 2.6.1) and an appropriate hazard analysis and critical control points (HACCP) food safety protocols for the diet ingredients, diet preparation, and diet administration should be established. 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. As bustards do eat meat, any meat fed on site should be processed following all USDA standards. Bustard diets containing raw meat need careful consideration in terms of preparation, handling, and provision; meat is a perishable food item, and spoilage can occur for a variety of reasons, including growth and activity of micro-organisms, insects or parasites, natural enzyme action in meat, chemical reactions, and physical changes (Hallager & Boylan, 2004).

The potential for spoilage is based on the type/number of micro-organisms present on the meat, in the storage and preparation areas, or transferred by the handler or 'pest' species with access to the bustard enclosures (Frazier & Westhoff, 1988). Meat and whole prey items should be held at appropriate temperatures during periods of thawing, preparation, and storage, and meat items should not remain at temperatures capable of promoting excessive microbial growth for extended periods of time once fed to the animals (Crissey et al., 2001). Uneaten meat should be disposed of according to local or state requirements.

Feeding schedules: Observations by Osborne and Osborne (1998) show that kori bustards feed around 0900h and again around 1700h, resting during the heat of the day. In zoos, bustards should be fed at least twice per day, but additional feedings should be considered to allow for necessary husbandry management and to promote behavioral opportunities for foraging and feeding throughout the day. As long as birds have adequate time to consume the diet, the period of access to diet can range from several hours to all day. Efforts to minimize the presence of pest species and their consumption of the diet may shorten the periods of time when the diet is offered to the birds, unless pest-proof feeders are provided (see section 2.2). The presence of pests should always be considered when determining the period of time the bustards have access to the diet.

Species-appropriate feeding and foraging: Bustards are curious, intelligent animals. Table 19 lists a range of food items that can promote foraging behavior (for a complete list, see Hallager & Boylan, 2004). Most of the items can be scattered around enclosures to encourage foraging/searching and object manipulation behaviors. It is important to note that different individuals in a group will respond differently to different items, and observations on preferred items that promote species-appropriate behaviors should be recorded for each individual. Approval from area veterinarians, managers, and nutritionists should be obtained if the following feeding approaches are considered.

Table 19. List of enrichment initiatives to promote foraging behaviors (Hallager & Boylan, 2004)

Food item	Description
Live insects	Bustards respond well to live insects, such as super worms, crickets, regular mealworms, and waxworms. Birds that are off their food for various medical reasons will often start eating again if live insects are offered.
Whole peanuts in the shell	Whole peanuts are also useful for medicating birds, especially kori bustards. A small portion of the top of the peanut can be taken off, the nut inside removed, and a pill inserted in its place. The peanut shell can be replaced and secured with peanut butter. Peanuts covered with peanut butter work well for medicating birds when individuals become suspicious of medicated mice.
Peanut butter	A few tablespoons of peanut butter can be spread on the trunks of trees in the wintertime as a source of extra calories. The behavior required by the birds to obtain this food item replicates the behavior of wild birds eating sap from acacia trees.
Alfalfa	Hanging bunches of alfalfa or other browse items from trees or other enclosure structures can also promote foraging.

While it is recommended to obtain approval from area veterinarians and nutritionists, the following food items can also be provided to bustards (e.g., hidden or scattered throughout the enclosure) to promote foraging behavior:

- Apple
- Banana
- Cherry tomatoes
- Chopped cantaloupe
- Chopped fruit/berries
- Cooked sweet potato
- Grapes (bunches or scattered)
- Mixed vegetables (e.g., peas, carrots, and corn)
- Watermelon

Many plant species have been observed in fecal samples from wild kori bustards (Mwangi, 1988). 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 must 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 species. 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.

AZA Accreditation Standard

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

Kori bustards have physically injured themselves on naturally growing browse in or around the perimeter of their enclosures. In one case, an individual bird experienced recurring injuries from the thorns of a barberry (*Berberis sp.*) plant that had grown into the enclosure (Hallager & Boylan, 2004).

Bustards are not browsers, however, individual institutions housing them may have appropriate locale-specific lists of browse materials used for enrichment. Any browse items known to have led to an adverse reaction in bustards should be reported to the SSP Coordinator so that these can be formally documented and the information widely disseminated. It is also important that institutions determine the nutrient content of any plant material and produce offered, and these data should be incorporated into the nutrient analysis of the entire diet if significant portions are consumed (Hallager & Boylan, 2004). There are several resources available which report nutrient content of readily available produce items (e.g., NRC, 2003), and a number of laboratories are equipped to perform nutrient assays.

6.3 Nutritional Evaluations

At one institution, kori bustards are weighed on a monthly basis to determine accurate body weight measures. The birds are scale trained and step onto a scale loaded with mealworms. Using this approach, Hallager (2005) has identified that males increase in body mass during the breeding season (Table 20), with the alpha male showing a significantly greater body weight increase than the beta male. This phenomenon has been confirmed at other institutions (S. Hallager, personal communication, 2018). Monitoring weight increases in males is one way to determine when reproductive hormones are beginning to increase (see section 8.1). Body weight evaluations are important, especially for zoos that house multiple males in the same enclosure during the non-breeding season, as weight increase in the alpha bird signify that it is time to separate the males prior to the breeding season. Seasonal weight fluctuations have not been reported in buff-crested bustards.

Table 20. Breeding and non-breeding weights (kg) for male kori bustards at Institution A

Dominant male						
Month	**1999***	**2000**	**2001**	**2002**	**2003**	**Average**
Jan.	---	---	---	14.4	---	14.4
Feb.	10.6	---	---	15.0	14.6	13.4
Mar.	11.1	14.5	14.2	15.1	14.8	13.9
Apr.	11.8	14.4	16.3	16.6	15.8	15.0
May	14.3	14.4	17.5	17.6	18.6	16.5
June	14.1	16.2	18.5	18.0	18.8	17.1
July	15.1	16.1	18.0	17.4	18.2	16.9
Aug.	15.1	17.3	16.3	14.8	18.6	16.4
Sep.	16.2	14.3	16.7	14.2	18.9	16.0
Oct.	15.3	14.6	15.3	14.2	18.0	15.5
Nov.	15.2	---	15.2	15.4	16.0	15.4
Dec.	13.8	---	15	15.4	14.8	14.7
Subordinate male						
Month	**1999**	**2000**	**2001**	**2002**		**Average**
Jan.	---	---	---	11.4	---	11.4
Feb.	10.3	12.1	---	11.9	---	11.4
Mar.	11.3	12.9	12.5	12.5	---	12.3
Apr.	11.3	12.5	13.4	12.6	---	12.4
May	11.3	13.5	14.1	11.0	---	12.5
June	12.1	14.5	---	11.6	---	12.7
July	13.0	14.7	14.5	11.8	---	13.5
Aug.	13.3	12.0	---	13.2	---	12.8
Sep.	12.7	---	---	13.2	---	12.9
Oct.	12.3	11.8	---	13.3	---	12.4
Nov.	12.6	---	11.5	11.0	---	11.7
Dec.	---	---	11.3	10.4	---	10.8

*The dominant male in 1999 was different than in the other years.

Other than body weight evaluations, there are currently no clinically valid nutritional evaluations that have been developed for bustards to assess growth, seasonal changes, etc. Body condition scores and fecal condition scores have not been used in assessments of these birds, but these scores should be developed.

Health issues: One of the most common signs of stress in bustards is decreased food consumption. Decreased food consumption should be monitored very closely, as it may not only be caused by environmental stressors, but also by impaction or illness. If a bird does not eat for more than one day, a veterinarian should be notified immediately.

Parent-reared chicks in large aviaries should be regularly checked for signs of metabolic bone disease, as this has been commonly seen at one institution (T. Bailey, personal communication, 2007). Kori bustards seem to be especially susceptible to nutritional bone disease (NBD), based on clinical findings of angular deformities of the metatarsi and laxity and swelling of the hock joint in animals housed in zoo conditions (Bailey, T. A., Nicholls, P. K., Samour, J. H., Naldo, J., Wernery, U., & Howlett, J. 1996). Careful monitoring of calcium and vitamin D levels in the diets of growing bustards has been shown to decrease the incidence of this musculoskeletal disorder (Bailey, T. A., Nicholls, P. K., Samour, J. H., Naldo, J., Wernery, U., & Howlett, J. 1996). In general, birds that are not provided with appropriate levels of nutrients may also show poor feather conditions, low weights, and lack of reproductive activity.

For hand-reared chicks, it is strongly recommended that individual food items be weighed when diets are prepared, so that a more accurate determination of nutrient content can be made when assessing the diet during the early growth period of the chicks. This is an important approach for hand-reared chicks, as nutrient content and growth rate need to be carefully monitored to minimize the occurrence of 'angel wing'

Target serum and tissue nutrient evaluations: Currently, there are insufficient data available to develop valid target serum and tissue nutrient values for bustards managed in AZA-accredited zoos. There are also no known model species that can be used to develop appropriate target serum and nutrient values for bustards. Additional information is needed and should be obtained from opportunistic blood samples taken from clinically 'normal' animals (e.g., during routine physicals) and standardized nutrient assays. A robust dataset will need to be developed before target values and related recommendations can be made. The Kori Bustard SSP® Nutrition and Veterinary Advisors can be consulted for information on institutions with laboratories that are able to perform nutrient assays suitable for these evaluations.

Chapter 7. Veterinary Care

7.1 Veterinary Services

Veterinary services are a vital component of excellent animal care practices. A full-time staff veterinarian is recommended; however, in cases where this is not necessary, a consulting/part-time veterinarian must be under contract to make at least twice monthly inspections of the animal collection and to respond to any emergencies (AZA Accreditation Standard 2.1.1). In some instances, because of their size or nature, exceptions may be made to the twice-monthly inspection requirement for certain institutions (e.g., insects only, etc.). Veterinary coverage must also be available at all times so that any indications of disease, injury, or stress may be responded to in a timely manner (AZA Accreditation Standard 2.1.2). All AZA-accredited institutions should adopt the guidelines for medical programs developed by the American Association of Zoo Veterinarians (AAZV), available at the AAZV website under "Publications", at http://www.aazv.org/displaycommon.cfm?an=1&subarticlenbr=839 (AZA Accreditation Standard 2.0.1).

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

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

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

The AZA Gruiformes TAG, Kori Bustard SSP®, and Buff-crested Bustard SSP® have Veterinary Advisors knowledgeable about veterinary care and management specifically applicable to bustards. They are also aware of veterinary research that still needs to be conducted to address current knowledge gaps. Generally, no specific training programs are necessary for veterinarians planning to work with bustards, as most general avian principles apply. However, it may be beneficial for first-time bustard veterinarians to contact the relevant SSP Veterinary Advisor or another experienced bustard veterinarian with any questions prior to performing a procedure. The AZA Gruiformes TAG Chair, Buff-crested Bustard SSP® Coordinator, and AZA Kori Bustard SSP® Coordinator can be contacted to obtain contact information for these Veterinary Advisors.

Daily assessments of activity, attitude, appetite, fecal output, and new concerns should be made by animal keeper staff. Routine veterinary health assessments should be performed every 2–3 years for each individual, or when otherwise indicated by signs of illness. These assessments may include physical examination, blood collection for a complete blood cell count, chemistry panel and plasma/serum banking, radiographs, fecal parasite screen, and fecal culture. Additionally, regular body weights should be obtained to enable more sensitive monitoring of possible disease. No unique equipment or technologies are necessary for performing routine health assessments in bustards.

Protocols for the use and security of drugs used for veterinary purposes must be formally written and available to paid and unpaid animal care staff (AZA Accreditation Standard 2.2.1). Procedures should include, but are not limited to: a list of persons authorized to administer animal drugs, situations in which they are to be utilized, location of animal drugs and those persons with access to them, and emergency procedures in the event of accidental human exposure. The AZA Gruiformes TAG, Buff-crested Bustard SSP®, and Kori Bustard SSP® recommend that veterinarians at each institution be involved in formulating their own institutional protocols for the storage and use of drugs that could be used in the care and management of bustards. Given the wide variation in veterinary practices, veterinary staff, and equipment available to veterinarians at different institutions, no bustard-specific recommendations can be made. Institutional veterinarians should also determine which drugs and medications are important for the treatment of bustard groups and individual animals. Recommendations for drugs used in parasite control, vaccinations, and anesthesia are

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

provided in the sections within this chapter. Other commonly used drugs include anthelmintics (ivermectin, pyrantel); analgesics (NSAIDS and steroids); and antibiotics (cephalosporins, beta-lactams, fluroquinolones, aminoglycosides). As these drugs are not unique to bustard management, institutions should follow their current protocols regarding storage and administration of these drugs.

Veterinary recordkeeping is an important element of animal care and ensures that information about individual animals and their treatment is always available. A designated staff member should be responsible for maintaining accurate animal veterinary record keeping. Health records should include all physical examination findings (including body weights), fecal examinations, blood work, radiograph interpretations, and other ancillary tests. Many institutions use ZIMS for record keeping, but other systems are also used. Institutions should keep continuous records of animal diet, housing, reproductive history, weight, behavior, molting, and medical history. Data from any veterinary exam should be entered into the institutional record keeping system. This should include history, anesthesia if used, physical exam findings, blood work results and interpretations, radiographic findings, fecal parasite check results, and any other diagnostics performed. Any medical problems and prescribed medications and treatments should also be recorded. In the event of death, necropsy findings should be included.

Animal development recordkeeping is an important part of an on-going effort to increase knowledge of bustard biology. Body measurements for each bustard should be recorded within the animals' medical records, as these can be used to add to the scientific knowledge base, and will continue to play an important role in taxonomic and eco-morphological investigations. Measurements that should be recorded for all bustards include:

- Wing length: Measure from the "wrist" to the tip of the longest primary.
- Tail length: Insert a ruler centrally between the longest tail feathers and the under-tail coverts until the ruler stops.
- Tarsus length: Measure on the front of the leg from the joint of the tibiotarsus with the tarsometatarsus to the lower end at the foot (last scute).
- Skull: Maximum length from the rear of the skull to the tip of the bill should be recorded, and the skull width (across the postorbital bones) should be measured.
- Culmen length: Measure from the tip of bill to base of skull.
- Toe length (inner, middle, and outer): Measure from the tip of the nail to the joint of the toe with the tarsometatarsus (Hallager & Boylan, 2004).

The morphometric measures listed above should be taken from birds at weekly intervals during their development, beginning with chicks from the day after hatching, and using the methods described by Hallager and Boylan (2004). Diagrams for how to take these body measurements properly can be found in Appendix H. Measurements should be recorded within the animals' institutional records, and information on each animal's sex, age, collection date, sexual condition, and weight should also be recorded and sent to the AZA Kori Bustard SSP® and Buff-crested Bustard SSP® Coordinators. Records for the entire life of each bird should be kept within institutional records, and should include information on:

- Diet: Dietary components, amount of food fed, and method of feeding.
- Housing: Dates when birds are moved indoors, outdoors, or to new enclosures.
- Egg production and reproduction: Yearly onset of egg laying, male display, copulation observations, egg fertility, egg measurements.
- Weight of adult birds: Weights can be taken from adult birds using the scale training method described in Chapter 9, section 9.1 (see also Hallager & Boylan, 2004).
- Behavior: Observations of aggressive behavior and animals involved.
- Medical problems: As required by institutional veterinary programs.
- Cause of death.

Additionally, any other information which animal caretakers at an institution consider to be pertinent, and which may improve husbandry standards for the species, should be included within each individual's medical record.

7.2 Transfer Examination and Diagnostic Testing Recommendations

The transfer of bustards between AZA-accredited institutions or certified related facilities due to SSP 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. Pre-shipment and transfer examinations and diagnostic tests should include a physical examination, blood collection for a complete blood cell count, chemistry panel and plasma/serum banking, radiographs, fecal parasite screen, and fecal culture.

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

- Regular weights
- Body condition by palpation of the pectoral muscles and keel as well as evaluation of the thoracic inlet
- Feather quality
- Blood draws checking CBC and biochemical profiles
- Fecal testing for parasitic ova

7.3 Quarantine

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

AZA Accreditation Standard

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

Birds originating from outside the United States will first need to enter International Quarantine. Upon completion, it is up to the receiving institution whether they enter an additional quarantine period at the receiving facility. Upon arrival at the receiving institution, bustards should be let out of their shipping crates as soon as possible into the quarantine area, and a visual inspection of the bird by a veterinarian or a keeper familiar with bustards should be performed to ensure that there are no obvious injuries or trauma acquired during transport. Water should be immediately available within the quarantine enclosure. When newly arrived bustards are placed in quarantine after shipping, they may initially refuse to eat; privacy and a continuous abundance of live insects and baby mice are recommended to promote feeding (Siegel, Hallager, & Bailey, 2007). Wild caught birds, even those previously held in zoos, may not eat for several days. Hand-reared birds will likely eat within a day of arrival, if not sooner.

Quarantine facilities for kori bustards should consist of at least a 3.7 m x 3.7 m (12 ft. x 12 ft.) fully enclosed stall (with outside access if possible) per bird, with a soft substrate (e.g., hay) and a shelter large enough for the bird to hide. For buff-crested bustards, a 1.2 m x 2.4 m x 2.4 m (4 ft. x 8 ft. x 8 ft.) per bird should be fine. Each stall should be cleaned daily by a dedicated staff member who will ideally not be interacting with other birds throughout the day. Staff should change clothes (i.e., scrubs and booties) before and after they enter the quarantine area. Additionally, there should be a disinfectant footbath at the entrance and exit of the quarantine area to minimize carrying potential disease into or out of the quarantine area.

If quarantine facilities are not available, it is preferable to physically separate new birds for the duration of quarantine. Quarantine exams should still take place.

AZA Accreditation Standard

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

AZA Accreditation Standard

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

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

Bustards can be carriers of *Salmonella* within their digestive tract, and *E. coli* is the most prevalent bacterial organism within bustard feces and cloacae (D'Aloia et al., 1996). Both of these bacteria represent potential hazards to human caretakers.

The AZA Gruiformes TAG, AZA Kori Bustard SSP®, and the Buff-crested Bustard SSP® recommend that veterinarians at each institution develop their own specific zoonotic disease and disinfection protocols for animal caretakers, animal management equipment, and enrichment initiatives provided in quarantine and hospital facilities. Care should be taken to ensure that those keepers working with sick or quarantine animals 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. Effective measures that help prevent the transmission of diseases between animals include (as designated by veterinarians at each institution): 1) washing hands between and after handling animals, fecals and urates, other bodily fluids or secretions, or animal diets; 2) wearing gloves, goggles, and a mask when cleaning animal enclosures; and 3) wearing gloves when handling any animal tissues. Disinfection protocols should take into consideration the material to be disinfected, and should ensure that disinfectants are thoroughly rinsed off or neutralized before the equipment or enrichment is used again with the birds.

Quarantine should last 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. The recommended quarantine period for bustards is 30 days (unless otherwise directed by the staff veterinarian). If additional birds are introduced into the quarantine area containing bustards during the 30-day quarantine, the minimum quarantine period should begin over again for all individuals unless the attending veterinarian says otherwise.

During the quarantine period, bustards should be given a full physical examination and tested/treated for fecal parasites. The AZA Gruiformes TAG recommends that all institutions follow AZA quarantine guidelines (Appendix E), and that veterinarians should develop appropriate quarantine testing protocols for their bustards. A complete physical should be performed. The birds should be evaluated for endoparasites and ectoparasites and treated accordingly. Endoparasites can be treated with pyrantel, ivermectin, or fenbendazole. Ectoparasites can be treated with dilute pyrethrin spray topically or with systemic ivermectin. Fecal samples should be collected and analyzed for gastrointestinal parasites. Blood should be taken for a complete blood count (CBC) and chemistry panel. Blood should be collected, analyzed, and then sera banked in either a -70 °C (-94 °F) freezer or a frost-free -20 °C (-4 °F) freezer for retrospective evaluation. Preliminary work to assess nutritional status and disease exposure is underway, and banked blood samples could help supplement these projects. Several publications provide hematological reference values for mature and growing bustards and are summarized in Appendix I, which should be consulted and compared with values taken during quarantine assessments.

When there is an indication, viral testing may also be appropriate (see section 7.4). 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. There are currently no recommended bustard-specific vaccination protocols or regulations to follow within the United States although many zoos choose to vaccinate against West Nile virus (WNV). Birds can also be anesthetized for radiographs in order to establish a "normal" radiograph baseline, but also to check for any abnormalities, including the presence of any foreign bodies that the birds may have ingested in their previous environment. While birds are anesthetized for their physical assessment, they should also be permanently identified (see section 3.4), if this has not been done before.

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.

Hand-reared bustards are generally more tolerant of quarantine conditions than wild-caught birds, and are less prone to developing social or behavioral problems during the quarantine period. The most commonly observed behavioral problems (e.g., pacing, low appetite, barking) arise when birds are responding to perceived stressors in the environment. Common stressors include loud noises, sudden noises, excessive vibrations, and separation from exhibit mates. Hand-reared birds may benefit from increased visits by keepers, installation of a mirror within the quarantine enclosure, an increase in favored food items, and quiet surroundings. Wild-caught birds may benefit from a reduction in keeper presence and environments as free from noise and disruption as possible.

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

Bustard deaths during quarantine are a very rare occurrence. 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 Appendix J for the Kori Bustard SSP® standardized necropsy protocol. Information on the gastro-intestinal tract of various bustard species is described in detail by Bailey, T. A., Mensah-Brown, E. P., Samour, J. H., Naldo, J. L., Lawrence, P., & Garner, A. (1997) and can be used for comparison with institutional data collected from bustard necropsies.

AZA Accreditation Standard

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

AZA Accreditation Standard

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

AZA Accreditation Standard

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

AZA Accreditation Standard

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

AZA Accreditation Standard

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

7.4 Preventive Medicine

AZA-accredited institutions should have an extensive veterinary program that must emphasize disease prevention (AZA

Accreditation Standard 2.0.2). AZA institutions should be aware of and prepared for periodic disease outbreaks in other animal populations that might affect the institution's animals, and should develop plans to protect the institution's animals in these situations (AZA Accreditation Standard 2.0.3). The American Association of Zoo Veterinarians (AAZV) has developed an outline of an effective preventative veterinary medicine program that should be implemented to ensure proactive veterinary care for all animals: https://cdn.ymaws.com/www.aazv.org/resource/resmgr/files/aazvveterinaryguidelines2016.pdf.

Parasite screening: Bustards should be screened biannually for parasites and de-wormed if necessary. A saline swab of the oropharynx and upper esophagus can be effective for cytology/parasitology testing (T. Bailey, personal communication, 2007). Anthelmintic and antiprotozoal medication can be given in the water or food when needed (Bailey & Hallager, 2003). See section 7.6 for a description of common parasites for bustards. It is recommended that parasite screenings be performed during health-assessment catch-ups that are scheduled for 1–2 months prior to the start of the breeding season (Bailey & Hallager, 2003).

Vaccinations: Vaccination policies for bustards depend upon individual institution policies that are generally based on a risk/benefit analysis. This analysis usually involves the prevalence of the specific disease, subsequent threat of exposure, efficacy and safety of a vaccine, and the risk to the bird. Studies evaluating the susceptibility of kori bustards to West Nile virus (WNV) have not yet been conducted and, given the relatively low incidence of reported morbidity and mortality of WNV in kori bustards, this may not be a necessary vaccination. Annual vaccination with inactivated Newcastle disease vaccine and live canary pox vaccine is provided in animal holding facilities in the Middle East (Bailey & Hallager, 2003), but typically not in zoos in the United States. When vaccinations are provided, it is recommended that they be given during health-assessment catch-ups performed 1–2 months prior to the start of the breeding season (Bailey & Hallager, 2003).

Blood sampling: Blood can be taken from bustards when they are appropriately restrained (see section 7.5), and blood sampling plays an important role in assessing the overall health of individual animals. If blood samples are taken from the leg of birds when the temperature is below 4°C (40°F), it is recommended that birds first be housed in a warm area for a minimum of 30 minutes to allow their legs to warm up. The increased temperature will allow veins to dilate and will facilitate blood collection. Blood samples may also be collected from the jugular or ulnar veins, but this requires a greater degree of skill and expertise in safely restraining the bird and locating the vessels. Appendix I lists normal kori bustard blood values, and hematological reference values for mature and growing bustards. These references should be consulted to compare them with the current health status of birds as part of each institution's preventative veterinary health program.

Medical management of neonate bustards: Information on some of the common issues encountered in the veterinary management of parent- and hand-reared kori bustard and buff-crested bustard chicks in zoos is provided below.

Dehydration: Newly hatched chicks are prone to dehydration for the first 2–3 days of life. Hand-reared chicks can be properly hydrated by feeding watermelon or dipping food items in water immediately prior to feeding. Subcutaneous fluids may need to be administered if oral hydration is not sufficient. When the female is feeding parent-reared chicks, she salivates copiously, and this may potentially be a source of water for the chicks.

Curled toes: Kori bustard chicks may hatch with inward pointing toes. This condition typically corrects itself over time. Taping the toes into the correct position is only necessary in cases where the condition is severe or worsens with time. The condition has not been reported in buff-crested bustards.

Angel wing: This condition begins between days 7–11 (see section 6.1). Taping of the affected primaries in a natural position at the first sign of outward turning will permanently correct the deformity. Although parent-reared chicks seem to have higher growth rates than hand-reared chicks during the first week of life, parent-reared kori bustard chicks do not usually develop angel wing. More research is needed to determine why parent-reared chicks do not develop angel wing. Angel wing in parent-reared buff-crested bustards has been reported in chicks between the ages of 10–12 days but was quickly corrected by taping the affected wing (M. Herry, personal communication, 2016).

Ingestion of foreign objects: Chicks normally eat small pebbles to aid in digestion. When exercising on natural substrates, chicks should be monitored closely to ensure that they do not consume too large or too many pebbles, leading to impaction. Hand-reared bustards should be provided with some grit or small gravel particles, as “overload ventriculus” has been identified as a cause of death in great bustard hand-reared chicks that were fed without grit (Siedel, 1995).

Eye pecking: Occasionally, one kori bustard chick may peck at the eyes of another bird, leading to physical injuries. The aggressor should be removed until the eyelid of the injured bird has completely healed. Providing the birds with food items to pick at (e.g., watermelon, tomatoes etc.) may reduce this behavior, as it may be a form of redirected or displaced foraging behavior. The behavior is nearly non-existent in buff-crested bustard chicks, given that there is usually only one chick in the clutch.

Weighing and handling: After hatching, parent-reared chicks should be weighed on the 2nd, 4th, and 6th day, and then every four days until the chick becomes too large for the scale (Hallager & Boylan, 2004). Unless an operant conditioning program includes scale training for the birds, healthy chicks should not be weighed daily, as increased injuries may result from frequent handling. Hand-reared chicks should be weighed daily. In the first two days, chicks may lose 10% of their body weight, but should gain 2–18% per day from then onwards (Hallager & Boylan, 2004). If chicks show continued weight loss, then supplemental feedings or medical treatment may be needed. By the time that chicks reach one-month of age, males can usually be distinguished from females by their larger size; based on zoo data, chicks greater than 2.2 kg (4.9 lb.) at 65 days of age are likely to be males, and individuals less than 2.2 kg (4.9 lb.) are likely to be females (Hallager & Boylan, 2004). If necessary for social housing considerations, earlier determination of sex can occur by taking a small amount of blood for DNA sexing.

Hypothermia: For the first couple of months after hatching, bustard chicks are sensitive to the cold. Care should be taken to provide sufficient heating, especially to debilitated chicks that are hospitalized. Managers should follow the advice of the referring aviculturalist or veterinarian for temperature guidelines. Under sub-optimal temperature conditions, bustard chicks and even juvenile bustards can suffer from hypothermia. Hypothermic bustards will not feed until their body temperature has returned to normal again (T. Bailey, personal communication, 2007).

Hyperthermia: Hyperthermia has caused the death of a number of bustards. Hyperthermia occurred in a group of white-bellied bustard chicks that were moved prematurely from air-conditioned rearing facilities to outdoor aviaries in the summer, without a proper period of acclimatization (T. Bailey, personal communication, 2007).

Medical management of molt: The molt cycle of bustards has not been closely studied. However, it is known that in kori bustards feathers tend to be shed mainly in the spring (before breeding season) and fall (after breeding season) (S. Hallager, personal communication, 2016). Males often (but not always) molt out their neck feathers during these periods. Given the visual importance of the throat area in male courtship displays, the replacement of new feathers may be critical to the breeding success of the male. Naturally molted feathers should be picked up and recorded as they are discovered so that the normal molt pattern of bustards can be understood, and this includes all primaries (note whether left or right), secondaries, tail, alular quills, greater and secondary coverts, underwing, and undertail feathers (Hallager & Boylan, 2004). Smaller feathers such as the neck, crest, and very small coverts, are too numerous to record, and can simply be noted when found. Molt seasonality in buff-crested bustards is not as apparent (M. Herry, personal communication, 2016) and requires more observation. Information on molt patterns should be recorded with the animal’s medical records, and a summary provided to the AZA Kori Bustard SSP® and AZA Buff-crested Bustard SSP® Coordinators on an annual basis.

Medical management of geriatrics: The greatest recorded lifespan of a kori bustard in zoos to date is a wild caught female who has been managed in a zoo for 37 years old (Hallager, 2016). This species may be capable of living longer given their delayed sexual maturity and low reproduction rate. Recent improvements in husbandry should yield longer lifespans.

The greatest recorded lifespan of a buff-crested bustard in zoos to date is a wild caught male who was estimated to be 30 years at the time of his death (Hallager, 2016). The oldest birds born into managed care was a male who died at 25 years old and a female who died at 21 years old. This species may be capable of living longer given its delayed sexual maturity and low reproduction rate. Recent improvements in husbandry should yield longer life spans.

Older birds may need to be housed inside more often than younger birds during inclement weather and may need to be supplied with supplemental heat earlier and later in the season. Assessments of the behavioral response of older birds to changing temperatures should be used to evaluate the needs of the animals. When catching or herding older birds, they should be moved slowly, as some may experience arthritic symptoms in their legs. Older birds may benefit from a daily supplement of nutraceuticals such as Cosequin® (a patented combination of glucosamine, purified chondroitin sulfate, and manganese ascorbate). Cosequin® is considered an adjunctive therapy for osteoarthritis in many species, but its use should be based on recommendations made by veterinarians. Some older birds may also be partially or totally deaf. If this is the case, animal caretakers should ensure that these birds are not startled during daily management by maintaining visual contact with the birds when working within or near the enclosure. Geriatric females may need to be isolated from breeding males during the breeding season, as overly aggressive males may harass them in an attempt to copulate.

Animals that are taken off zoo/aquarium grounds for any purpose have the potential to be exposed to infectious agents that could spread to the rest of the institution's healthy population. AZA-accredited institutions must have adequate protocols in place to avoid this (AZA Accreditation Standard 1.5.5). While currently there are no known bustards used for off grounds programs, the potential does exist. Such birds should be housed separately from collection bustards 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 may be required to re-enter a quarantine period but this will at the discretion of the attending veterinarian. Any abnormalities consistent with mycobacterial disease (i.e., severe leukocytosis, bone or pulmonary lesions on radiographs) detected on exam of a healthy or ill bird should be investigated further.

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.

7.5 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). Crate training can minimize stress and injury and is encouraged for all bustard species.

AZA Accreditation Standard

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

General: Bustards, especially adult male koris, are very powerful birds, and require somewhat different handling skills from other long-legged birds of similar size. There are three important characteristics of bustards that should be considered when handling these animals:

1. The physical strength of kori bustards, especially adult males, which can weigh as much as 20 kg (44.1 lb.) during the breeding season.
2. The thinness of their skin; bustard skin is very thin and rips easily when a bird is struggling. Torn skin will result in the need for further handling for veterinary treatment (e.g., sutures), and can lead to maggot infestation.
3. Their inherent nature to shed feathers when captured; bustard feathers are very loosely embedded in the follicles, and even the best handling will result in some feather loss. Improper handling can cause major feather loss. The wings or tails of the birds should never be grabbed during a capture attempt, as this will lead to significant feather loss and an increasingly fearful bird.

Bustards are highly sensitive to stress. In addition to the considerations listed above, poorly planned capture events and incorrect handling can also lead to:

- Temporary or permanent neural damage
- Hyperthermia
- Fractures of legs or wings

- Skin lacerations, bruising, and feather loss
- Luxation of the tibiotarsal bones
- Dislocation of the cervical vertebrae
- Compression of the trachea and internal organs
- Capture myopathy
- Stress, the progression of a disease process, and even death (Siegel, Hallager, & Bailey, 2007)

Kori bustard capture protocol: A specific plan for the capture and release of kori bustards, and the personnel and equipment needed, should be discussed prior to the actual capture event. In order to avoid injuries to birds and personnel, it is recommended that two people are involved in the capture and restraint of kori bustards, and this is very strongly recommended when capturing a male. Equipment such as hoods for bird, protective eyewear, and gloves should be readily available for animal caretakers. Although kori bustards are generally silent (outside of male courtship displays), both sexes produce a barking sound when they are alarmed, and individuals can make loud, roaring noises during capture (Hallager & Boylan, 2004). The pre-capture planning should consider where and when captures take place. Important issues to consider for a capture event include:

<u>Temperature</u>: Capturing the birds in the early morning hours (especially at southern latitudes during the summer months) is recommended in order to reduce the risk of heat stress to the birds.

<u>Preferred capture site</u>: It is recommended that captures be performed in a small, darkened and enclosed area, such as a shed or holding stall with solid walls, so that the chance of the animals evading capture is greatly reduced. Cornering a bird against chain-link or welded-mesh fences is not recommended, as the birds are more likely to receive trauma to their beaks, heads, carpal joints, and feet if they try to “climb” the see-through barriers. Padded walls may be beneficial in a capture area to reduce the chance of trauma. Fixing corrugated cardboard to the walls of a capture area can also be beneficial, although there are plastic padding alternatives available for use that is more permanent. In the absence of a recommended indoor capture area, a small area with either trees or brush can be used to assist in directing the animals into a catch area. A funneling system or the boundaries of the exhibit (such as fences or walls) can also be used to direct the animal to the catch area.

<u>Containment</u>: If birds are to be captured in an outside enclosure, then the fence line at the capture area should be at least 2.1 m (7 ft.) high, preferably higher. When kori bustards are cornered against a solid wall, most birds will generally attempt to leap into the air, and even a pinioned bird can jump at least 1.8 m (6 ft.). Ideally, the bird should be caught before it takes a leap. If it does leap upwards, the bird should be caught as soon as its feet get back onto the ground, before it can leap again.

<u>Safety of caretakers</u>: The main potential cause of injury to caretakers during restraint of a kori bustard will be from the bird’s feet, as they are very strong-legged birds. Two caretakers are generally needed to ensure proper safety of the bird as well as the keepers involved. When two people are involved, one person captures the bird, while the other takes control of the legs and wings. Goggles can be worn as an added safety measure to protect the eyes of the animal care staff, as kori bustards do occasionally peck at their holder’s face.

<u>Capture techniques</u>: The ease of capture will vary widely, depending on exhibit design and the temperament of the birds. If the birds are accustomed to coming into a shed or holding stall on a daily basis for their food, then this can reduce the stress of a capture, as the birds can be moved into the capture area using food reinforcement. Once birds are in the containment area, it is very important that they be caught quickly. The less the bird struggles and attempts to avoid capture, the less it will be stressed and the less the chance of resulting injury. The use of nets is not recommended to capture kori bustards, as the risk of injury escalates greatly with this capture technique. Injuries will be greatly reduced if birds can be guided into darkened areas and then caught by hand, as described above. Once the bird has been captured, and the body and legs are controlled, it is necessary to lift or walk the bird to the area of the husbandry or veterinary procedure. If this is at an off-site area, the bird may need to be crated (see the sub-sections below for additional information).

Kori bustard restraint protocol: The preferred method of restraint for kori bustards is to tuck the body of the bird under one arm with the head facing back. The weight of the bird should rest primarily on the holder’s forearm, while the other hand is used to restrain the legs. The legs should generally not be tucked up under the bird's body, as it is possible for a bird to break its leg if it struggles when tightly restrained. The legs should be restrained at the tarsal joint with at least one finger keeping the legs apart so that they do not abrade the joints against each other. Kori bustards also have powerful wings, and it is important to ensure that the wings are tucked firmly against the body of the bird so that they cannot flap them. Once the bird is secured, a second handler should immediately cover the eyes of the bird, without blocking the nares or holding the beak shut, as this can have a calming effect. Cotton bags can be used to cover the heads of kori bustards, but these bags should have an opening over the nostrils to allow breathing. Hand-reared birds often prefer to remain un-hooded (S. Hallager, personal communication, 2007). While it is not necessary to restrain the head of kori bustards, it may be in the best interest of the restrainer to have the head under control.

During the first 7 days, hand-reared kori bustard chicks are less stressed by restraint if handling by animal caretakers simulates brooding. Chicks can be cupped in one hand and covered with the other hand, kept in a "nest" with a feather duster placed on top, or covered with a breathable cloth. When restrained, care should be taken to keep the chicks' feet away from their bodies, as they have very sharp nails, and can inflict injuries to themselves (e.g., a ripped neck) if they are able to kick with their feet (Hallager & Boylan, 2004).

When moving restrained kori bustards within an institution, caretakers should rely on their knowledge of the bird. Hand-reared birds may prefer to be hand carried. Crates can work very well for transporting birds even over short distances, but removing a bird from a crate so that it can be handled can be challenging. If bustards do need to be recaptured once released from a crate, it is recommended that the bird first be released into a small, darkened area to let it calm down, before recapturing it again using the techniques described above. If a bird is being captured and restrained for a procedure that will involve anesthesia, it may be helpful to anesthetize the bird at its enclosure to decrease the stress involved during transport from the enclosure to the clinic when the bird is awake. The effectiveness of this approach depends upon a variety of factors, including the veterinarian's comfort level with this technique, available monitoring and portable anesthetic equipment, proposed time under anesthesia, length of travel time to the clinic, training of animal care staff, and the bird's presenting complaint. The final decision regarding which approach to use rests with the attending veterinarian.

When manually restrained bustards are released, it is important that the bird be stabilized before it is released. The bird should be positioned so that it legs are touching the ground, keeping in mind they can jump with power, and it should be facing a clear area. Birds should be released in the direction of a clearing that allows it time to gain its balance and get its bearings. Once the bird is stabilized, the restrainer should simply release the bird from their control and slowly step back. The bird will most likely move away, but individuals can remain standing, looking at the caretakers. In either case, the best practice is for caretakers to move towards the exit in a slow and deliberate manner.

Kori bustard immobilization: The oral sacs of kori bustard males in breeding condition can occlude the glottis during anesthesia, potentially leading to anoxia and death (Fowler & Miller, 2012). The use of an endotracheal tube during anesthesia in these birds is essential. Anesthesia techniques for kori bustards have also been described in Naldo et al. (1997, 1998) and Bailey, Werney, Howlett, Naldo, and Samour. (1999). Bailey, Kinne, Naldo, Silvanose, and Howlett (2001) used a combination of isoflurane (5% with mask for induction, and 3-3.5% with endotracheal tube for maintenance) and oxygen (3 L per minute). It is recommended that institution-specific anesthesia protocols be developed for kori bustards by veterinarians at each institution, and that effective protocols should be shared with the AZA Gruiformes TAG and AZA Kori Bustard SSP for wider dissemination.

Kori bustard release/recovery: When recovering a bird from anesthesia, it will be necessary to restrain the bird for a longer period than after manual restraint. Birds recovering from anesthesia should be manually restrained using firm, constant pressure, and the body and legs of the animals should be carefully controlled to restrict any sudden outbursts of energy. Covering the eyes of the bird with a hood or towel will help in reducing this reaction (ensuring that the nares are not covered). Once the animal has fully recovered from anesthesia, it can be released as described above, ensuring that the bird has regained sufficient control of its legs to be stable. After prolonged capture, restraint, or immobilization, it is recommended to treat bustards with fluids, vitamin E, selenium, and physiotherapy, to help the birds recover (Siegel, Hallager, & Bailey, 2007). Some pre-capture treatments of vitamin E and selenium may also be beneficial. It is also important to reduce conditions that increase the likelihood of capture myopathy occurring, such as extended pursuit times, transport, excessive handling, and over-exertion during times of high heat.

Buff-crested bustard capture protocol: A specific plan for the capture and release of buff-crested bustards, and the personnel and equipment needed, should be discussed prior to the actual capture event. Some pre-capture treatments of vitamin E and selenium may also be beneficial. It is ~~also~~ important to reduce conditions that increase the likelihood of capture myopathy occurring, such as extended pursuit times, transport, excessive handling, and over-exertion during times of high heat. The pre-capture planning should consider where and when captures take place. Important issues to consider for a capture event include:

<u>Temperature</u>: Capturing the birds in the early morning hours (especially at southern latitudes during the summer months) is recommended in order to reduce the risk of heat stress to the birds. When whole flocks of small bustards need to be captured, the use of a catching corral is recommended: a blind-ended funnel should be set-up that leads to a circular catching area at the end (Siegel, Hallager, & Bailey, 2007). Small and medium-sized bustards in the circular catching area can be caught with nets (Bailey & Hallager 2003). Hand-held nets with a string mesh bag and lightweight frame are appropriate for buff-crested bustards (Anderson, 1998a, 1998b). Out on exhibit, individual small or medium bustards can also be caught with nets, while tamer bustards within smaller indoor aviaries can be herded into a corner and caught by hand.

<u>Preferred capture site</u>: It is recommended that captures be performed in a small, darkened and enclosed area, such as a shed or holding stall with solid walls, so that the chance of the animals evading capture is greatly reduced. Buff-crested bustards will try to evade capture by jumping straight up. Make sure that the capture area is clear of overhead perching or obstacles that would create a hazard for the bird. Cornering a bird against chain-link or welded-mesh fences is not recommended, as the birds are more likely to receive trauma to their beaks, heads, carpal joints, and feet if they try to “climb” the see-through barriers. Padded walls may be beneficial in a capture area to reduce the chance of trauma. Fixing corrugated cardboard to the walls of a capture area can also be beneficial, although there are plastic padding alternatives available for use that is more permanent. In the absence of a recommended indoor capture area, a small area with either trees or brush can be used to assist in directing the animals into a catch area. A funneling system or the boundaries of the exhibit (such as fences or walls) can also be used to direct the animal to the catch area.

<u>Containment</u>: Out on exhibit, individual small or medium bustards can also be caught with nets, while tamer bustards within smaller indoor aviaries can be herded into a corner and caught by hand.

<u>Safety of caretakers</u>: Goggles can be worn as an added safety measure to protect the eyes of the animal care staff.

<u>Capture techniques</u>: The ease of capture will vary widely, depending on exhibit design and the temperament of the birds. If the birds are accustomed to coming into a shed or holding stall on a daily basis for their food, then this can reduce the stress of a capture, as the birds can be moved into the capture area using food reinforcement. Once birds are in the containment area, it is very important that they be caught quickly. The less the bird struggles and attempts to avoid capture, the less it will be stressed and the less the chance of resulting injury. Injuries will be greatly reduced if birds can be guided into darkened areas and then caught by hand, as described above. Once the bird has been captured, and the body and legs are controlled walk the bird to the area of the husbandry or veterinary procedure. If this is at an off-site area, the bird may need to be crated.

Buff-crested bustard restraint protocol: When restrained, small or medium-sized bustards should be held firmly against the handler’s body, the wings should be held closed, and the legs held together with the handler’s fingers between them to prevent the legs from rubbing (Bailey & Hallager, 2003). The legs can be allowed to bend at the tibiotarsal joint, and some movement can be allowed to prevent tension-induced strains (Anderson, 1998a, 1998b), without letting the birds kick in an uncontrolled manner. The use of hoods or cloth bags (with holes for the nares) can calm birds down during restraint (hand-reared birds may be calmer without hoods), and the birds should be held as sternally as possible to minimize stress (Bailey & Hallager, 2003). Once the bird is secured, a second handler should immediately cover the eyes of the bird, without blocking the nares or holding the beak shut, as this can have a calming effect. Cotton bags can be used to cover the heads of bustards, but these bags should have an opening over the nostrils to allow breathing. While it is not necessary to restrain the head of buff-crested bustards, it may be in the best interest of the restrainer to have the head under control.

When moving restrained buff-crested bustards within an institution, caretakers should rely on their knowledge of the bird. Hand-reared birds may prefer to be hand carried. Crates can work very well for transporting birds even over short distances, but removing a bird from a crate so that it can be handled can be challenging. If bustards do need to be recaptured once released from a crate, it is recommended that the bird first be released into a small, darkened area to let it calm down, before recapturing it again using the techniques described above.The effectiveness of this approach depends upon a variety of factors, including the veterinarian's comfort level with this technique, available monitoring and portable anesthetic equipment, proposed time under anesthesia, length of travel time to the clinic, training of animal care staff, and the bird's presenting complaint. The final decision regarding which approach to use rests with the attending veterinarian.

When manually restrained bustards are released, it is important that the bird be stabilized before it is released. The bird should be positioned so that it legs are touching the ground, keeping in mind they can jump with power, and it should be facing a clear area. Birds should be released in the direction of a clearing that allows it time to gain its balance and get its bearings. Once the bird is stabilized, the restrainer should simply release the bird from their control and slowly step back. The bird will most likely move away, but individuals can remain standing, looking at the caretakers. In either case, the best practice is for caretakers to move towards the exit in a slow and deliberate manner.

Buff-crested bustard release/recovery: When recovering a bird from anesthesia, it will be necessary to restrain the bird for a longer period than after manual restraint. Birds recovering from anesthesia should be manually restrained using firm, constant pressure, and the body and legs of the animals should be carefully controlled to restrict any sudden outbursts of energy. Covering the eyes of the bird with a hood or towel will help in reducing this reaction (ensuring that the nares are not covered). Once the animal has fully recovered from anesthesia, it can be released as described above, ensuring that the bird has regained sufficient control of its legs to be stable. After prolonged capture, restraint, or immobilization, it is recommended to treat bustards with fluids, vitamin E, selenium, and physiotherapy, to help the birds recover (Siegel, Hallager, & Bailey, 2007).

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

AZA-accredited institutions should have an extensive veterinary program that manages animal diseases, disorders, or injuries and has the ability to isolate these animals in a hospital setting for treatment if necessary. The owner of an animal on loan at a facility is to be consulted prior to any elective invasive procedures, including permanent contraception.

Bustard care staff should be trained in meeting the animal's dietary, husbandry, and enrichment needs, as well as in restraint techniques. Staff should also be trained to assess animal welfare and recognize behavioral indicators animals may display if their health becomes compromised, however, animal care staff should not diagnose illnesses nor prescribe treatment (AZA Accreditation Standard 2.1.3). Protocols should be established for reporting these observations to the veterinary department. Hospital facilities for bustards must have radiographic equipment or access to radiographic services (AZA Accreditation Standard 2.3.2), contain appropriate equipment and supplies on hand for treatment of diseases, disorders or injuries, and have staff available that are trained to address health issues, manage short and long term medical treatments and control for zoonotic disease transmission.

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(2.1.3) Paid and unpaid animal care staff should be trained to assess welfare and recognize abnormal behavior and clinical signs of illness and have knowledge of the diets, husbandry (including enrichment items and strategies), and restraint procedures required for the animals under their care. However, animal care staff (paid and unpaid) must not diagnose illnesses nor prescribe treatment.

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

The following information in this section describes common diseases and disorders experienced by bustards in zoos, and diagnosis and treatment options for these medical concerns. New knowledge gained should be reported to the AZA Kori Bustard SSP® and the AZA Buff-crested Bustard SSP® for dissemination.

Kori bustard trauma: Most large bustards are generally hardy birds, but individuals can sustain life-threatening trauma such as puncture wounds, or compound fractures of legs or wings. The complications resulting from these injuries can be made worse if individuals are housed with incompatible exhibit-mates (Siegel, Hallager, & Bailey, 2007). Where open wounds are present, bustards often do not preen fly eggs or maggots from their wounds, and it is recommended that wounded animals be captured and placed into a fly-free room, or that the wound be covered in fly-repellent gel.

In response to stressors in the environment, kori bustards will run into or pace against the perimeter fencing or walls of their enclosures in attempts to flee from the stressor (or potential stressor). If the containment barriers are not smooth, then there is a greater likelihood that individuals will sustain abrasions to their faces, wings, or pinion sites (Siegel, Hallager, & Bailey, 2007). Excessive pacing may also lead to compaction of the soil, which can be a contributing factor to lameness and pododermatitis in these birds.

Buff-crested bustard trauma: Bailey, T. A., Nicholls, P. K., Samour, J. H., Naldo, J., Wernery, U., & Howlett, J. (1996) and Bailey, T. A., Samour, J. H., Naldo, J., Howlett, J. C., & Tarik, M. (1996) identified intra-specific trauma, fractured necks, dislocations, and aviary-related trauma to account for 57.1% of the deaths of adult buff-crested bustards at the National Avian Research Center (Abu Dhabi). Birds responding to stressors in their environments, and those that have neither been pinioned nor have had their primary flight feathers clipped, are especially susceptible to trauma (Siegel, Hallager, & Bailey, 2007). Male buff-crested bustards may also injure their heads during courtship displays that involve flying directly upwards; and females may receive severe injuries by overly aggressive or hand-reared males during the breeding season (Siegel, Hallager, & Bailey, 2007). Open wounds will likely become infested with flies.

Disturbed buff-crested bustards often fly away vertically to avoid a potential stressor and are very prone to broken necks (Siegel, Hallager, & Bailey, 2007). The provision of well-designed enclosures, minimizing disturbances, and the use of husbandry training to reduce the sensitivity of the bustards to the sights and sounds of the captive environment are all recommended. Trauma can be reduced in the following ways.

<u>Padding</u>: Adding plastic-coated foam padding to the sides and tops of enclosures (e.g., quarantine and hospital enclosures) can reduce abrasion damage to wingtips for both species.

<u>Netting</u>: In outdoor aviaries, adding shade-cloth or tension netting to the roof and sides of aviaries can help to cushion the impact resulting from birds flying within a pen.

<u>Training</u>: Desensitizing nervous individuals to stimuli that occur frequently within the captive environment by using operant conditioning may reduce fearful behavior, as will housing nervous birds in naturalistic pens with plenty of cover.

<u>Husbandry</u>: Birds that are not pinioned should have their primary feathers regularly trimmed. Reducing the number of unnecessary disturbances (e.g., non-essential people) around the birds may also prevent flighty or fearful behavior.

Viral diseases: The following viral diseases have been associated with bustard species (Bailey, 2008) and so represent potential health concerns for bustards. The significance of these viruses for the care and management of bustards in zoos remains unknown, and additional research is needed on the prevalence and serious of these viruses to bustards in the United States:

- Adenovirus
- Avian influenza
- Avipox
- Herpesvirus
- Infectious bursal disease
- Lymphoid leucosis
- Marek's disease
- Newcastle disease
- Pigeon herpes
- PMV-2
- Reovirus

Bacterial diseases: Wild and managed bustards are colonized by several species of bacteria as part of their normal aerobic intestinal flora (e.g., *Proteus spp*., *Enterobacter spp*., *Escherichia coli*, *Klebsiella spp*., *Aerococcus spp*., *Pseudomonas sp*., *Serratia sp*., and *Enterococcus spp*.) (D'Aloia, M., Bailey, T. A., Samour, J. H., Naldo, J. L., & Howlett, J. C., 1996).; Silvanose et al. 1998a). The following bacteria have also been isolated from the conjunctiva of healthy kori bustards: *Micrococcus spp*., *Staphylococcus xylosus*, and *Bacillus spp*.; and from the nasal cavities of healthy animals: *Micrococcus spp*., *Bacillus spp*., *Staphylococcus sciuri*, and *Aerococcus spp*. (Silvanose, Bailey, Naldo, & Howlett, 2001). Birds from which these samples were taken showed no clinical signs of disease. Aerobic bacteria (including *Klebsiella sp*. and *E. coli*) have also been found in cloacal swabs taken from clinically healthy kori bustards (D'Aloia, M., Bailey, T. A., Samour, J. H., Naldo, J. L., & Howlett, J. C. 1996). Both Gram-positive cocci and Gram-negative bacilli can be found in fecal samples collected from clinically healthy kori bustard chicks housed in typical zoo conditions, and may represent the normal aerobic microflora associated with these birds (Naldo, J.L., Silvanose, C.D., Samour, J. H., & Bailey, T. A., 1998). Bacteria have also been isolated from the wing skin of clinically healthy kori bustards, and the species identified from this area include *Staphylococcus sp*., *Bacillus sp*., *Citrobacter sp*., and *Pseudomonas sp*. (D'Aloia, M., Bailey, T. A., Samour, J. H., Naldo, J. L., & Howlett, J. C., 1996).

Some clinically active bacterial infections that have been reported in kori bustards include colisepticaemia, mycobacteriosis, clostridiosis, salmonellosis, yersiniosis, pseudomoniasis, erysipelothrix, and chlamydophilosis (Siegel, Hallager, & Bailey, 2007). *Pseudomonas sp*. infections can present in kori bustards with clinical signs such as mucopurulent nasal and choanal discharge, mucoid ocular discharge, coughing, choanal inflammation, sinus swelling, and anorexia (Bailey et al., 2000). Hematology results from birds presenting with clinical signs of pseudomoniasis are presented by Bailey et al. (2000), along with potential treatment regimens. Control measures include the proper cleaning and sanitization of food and water containers, where the bacteria can proliferate in warm conditions (Bailey et al., 2000). If treatment is warranted for any bacterial diseases, antibiotic selection should be based upon culture and bacterial sensitivity results. One kori bustard died after a 24 hour period of mild lethargy, the only gross necropsy findings being petechiae and ecchymoses of the internal organs. Histopathologic examination revealed acute bacterial septicemia, and *Campylobacter* sp. was isolated in pure culture from the liver. (C. Bonar, personal communication, 2019).

Fungal diseases: Aspergillosis (*Aspergillus fumigatus*) can occur in bustard chicks reared in zoos (Siegel, Hallager, & Bailey, 2007), and has been found in wild and managed great bustards (Garcia-Montijano et al., 2002). Yeast infections (*Candida albicans*) of the oropharyngeal cavity and oesophagus have been reported in birds on antibiotic treatment. While this type of infection typically occurs in juvenile birds with immature immune systems, it can also occur in adult birds (Siegel, Hallager, & Bailey, 2007). A survey of clinically healthy kori bustards found no latent fungal infections (D'Aloia, M., Bailey, T. A., Samour, J. H., Naldo, J. L., & Howlett, J. C., 1996).

Protozoal diseases: Trichomoniasis is reported as a significant protozoal disease for captive bustards in the Middle East (Naldo, Bailey, & Samour, 2000a), and has led to fatalities for animals in the United States as well. Clinical signs of trichomoniasis include: lesions in the oro-pharynx and esophagus, foul-smelling odor from the mouth, anorexia, lethargy, dyspnoea, emaciation, oral and lacrimal discharges, and inflammation of the oro-pharynx, tongue, or beak margin (Naldo, Bailey, & Samour, 2000a). Transmission of the protozoa is through ingestion of contaminated food and water sources (Naldo, Bailey, & Samour, 2000a). Treatment options for trichomoniasis include metronidazole and supportive therapy, as well as the use of dimetridazole and ronidazole as preventative medication (Naldo, Bailey, & Samour, 2000a). Other protozoa that can cause diseases of the digestive tract include: *Eimeria* spp., *Giardia* spp., *Histomonas* spp., and *Entamoeba anatis* (Silvanose et al., 1998a).

In general, clinical signs associated with protozoal oro-pharyngeal diseases in kori bustards can include weight loss, weakness, white-yellow lesions, mucus discharge, and exfoliation of epithelial cells (Silvanose et al., 1998b). The nature of clinical signs showed by infected birds is dependent on the level of parasitism, duration of infection, and any secondary bacterial or fungal diseases (Silvanose et al., 1998b).

Parasites: Helminth and cestode infestations can cause morbidity in some bustards (Jones et al., 1996). Since birds taken from the wild show significant internal parasite loads, it is recommended that all newly arriving birds be treated with anti-parasitic medication during quarantine periods (Jones et al., 1996;

Bailey et al. 2000; Schuster & Kinne, 2003). A single dose of praziquantel (10 mg/kg p.o.) has been shown to eliminate gastrointestinal cestodes in kori bustards (Jones et al., 1996), although the authors also recommend a second dose after 14 days. To eliminate unnecessary handling of the animals, birds can be treated with anthelmintic tablets placed in preferred food items (e.g., mice) that are offered to the birds (Jones et al. 1996), especially if birds are treated as part of a routine, preventative program.

Metabolic disorders: Hemosiderosis has been reported in some kori bustards in zoos, but the cause of this is not yet known. Two hypotheses that should be explored are a possible dietary etiology or a genetic predisposition (S. Murray, personal communication, 2007). Kori bustards are also susceptible to fatty liver disease (Nicholls, Bailey, & Samour, 1997). Capture and transport of bustards has been shown to be associated with an increased prevalence of this disease; the clinical biochemical findings linked to fatty liver syndrome are further described by Nicholls et al. (1997). The use of serum bile acid (SBA) concentration in the blood as a diagnostic tool for liver function in kori bustards has been proposed by Howlett et al. (1999), and the authors present reference value ranges for SBA concentrations.

Musculoskeletal disorders: Musculoskeletal disorders can significantly affect the health and development of kori bustards (Naldo, Bailey, & Samour, 2000b). Common disorders include: angel wing, rotational and angular wing deformities, spraddle legs, rolled toes, and bone fractures (Naldo, Bailey, & Tamour, 1998c), especially in birds experiencing metabolic bone disease (Naldo, Bailey, & Samour, 2000b). Musculoskeletal disorders are most common in the first 26 weeks of life, when bone growth rates are at their greatest (Naldo, Bailey, & Tamour, 1998c). Assessment of chick growth rate, body weight changes, and food intake, as well as performing routine physical examinations, is important to detect musculoskeletal disorders early on; early detection increases the likelihood of successful treatment (Naldo, Bailey, & Samour, 2000b). Growth rates, periods, and bone sizes are detailed for kori bustards by Naldo, Bailey, & Samour (2000b), and can be used as reference values for developing chicks.

Hereditary issues: No known hereditary diseases or disorders have been reported in kori or buff-crested bustards, although it is possible that hemosiderosis may have a genetic link.

Other health-related issues: Ingestion of certain non-food items by bustards can lead to perforation or impaction of the gastrointestinal tract, and can be a significant source of morbidity and mortality for bustards (Bailey et al., 2001). Clinical signs of possible gastrointestinal trauma associated with the ingestion of foreign objects can include decreased appetite, poor pectoral muscle condition, weight loss, palpable abscesses in the abdominal wall, and hematological indicators associated with an inflammatory response (Bailey et al., 2001). Foreign bodies that penetrate the ventriculus of birds once ingested can lead to decreased muscular contractions and poor digestion of food (Lumeij, 1994). The use of rigid endoscopy to remove foreign bodies from the ventriculus is generally not possible with kori bustards, given the length of the esophagus, and so ventriculostomy or proventriculostomy are the best techniques to use (Bailey et al., 2001). A ventral laparotomy has also been used successfully to remove wire ingested by a kori bustard (Bailey et al., 2001).

Hospitalization: Wild caught bustards that have to be hospitalized benefit from surroundings that are free from noise and disturbance. While hand-reared birds are not as stress prone, they should not be housed in areas where sudden noise or frequent disturbances are a possibility. Sudden and unexpected noises have led to injury and death to birds in unfamiliar surroundings. During hospitalization, hand-reared birds may benefit from increased visits by keepers, the installation of a mirror within their hospital enclosure, an increase in favored food items, and quiet surroundings. The response of the animal to a mirror should be monitored to ensure that it is not seen as an additional stressor. Wild-caught birds may benefit from a reduction in keeper presence, and environments as free from noise and disruption as possible.

Hospitalization facilities for bustards do not need to be elaborate or expensive. In larger enclosures and naturalistic aviaries, small holding pens (6 m x 6 m (19.7 ft. x 19.7 ft.)) made of shade cloth are suitable for isolating kori bustards (T. Bailey, personal communication, 2007). For buff-crested bustards, pens measuring 1.2m x 2.4m x 2.4m (4 ft. x 8 ft. x 8 ft.) are appropriate and should contain areas to hide behind to reduce nervousness as well as soft mesh on the ceiling to prevent head injuries caused when birds jump (M. Herry, personal communication, 2016).

When birds are housed within specific hospital enclosures, the provision of padded rooms should be considered. Hospital stays should be as short as possible, and the birds returned to their normal enclosures as soon as possible. While housed in hospital facilities, favored food items should be offered to encourage eating. The presence of a familiar keeper may also help some birds adjust to their temporary quarters.

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

AZA Accreditation Standard

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

Given the wide variety of zoos that house bustards, the AZA Gruiformes TAG, the AZA Kori Bustard SSP®, and the AZA Buff-crested Bustard SSP® cannot provide specific recommendations for the best approaches to take to communicate animal welfare issues effectively within every institution. All animal caretakers that work with bustards should be aware of institutional protocols in place for them to identify, communicate, and hopefully address potential animal welfare issues that are associated with the care and management of these animals.

Bustards can be very good at hiding an illness. For this reason, animal caretakers should be especially vigilant, and immediately communicate their concerns to a curator and/or veterinarian, as required by institutional protocols (Hallager & Boylan, 2004). Animal caretakers should report any signs of illness, especially a reduced appetite, as soon as possible. Bustards usually have a very good appetite, and any deviation from this behavior is unusual and should be reported at once. Any lameness issues observed should also be reported and monitored, as bustards can develop serious foot and leg problems; if these musculoskeletal health issues are detected early, more serious physical health complications can possibly be avoided (Hallager & Boylan, 2004). Familiarity with individual birds is essential when caring for these species effectively.

Euthanasia and necropsy: AZA-accredited zoos and aquariums provide superior daily care and husbandry routines, high quality diets, and regular veterinary care, to support bustard 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 bustard both in their care and in the wild. As stated earlier, necropsies should be conducted on deceased bustard to determine their cause of death, and the subsequent disposal of the body must be done in accordance with local, state, or federal laws (AZA Accreditation Standards 2.5.1 and 2.5.3). If the animal is on loan from another facility, the loan agreement should be consulted as to the owner's wishes for disposition of the carcass; if nothing is stated, the owner should be consulted. Necropsies should include a detailed external and internal gross morphological examination and representative tissue samples from the body organs should be submitted for histopathological examination. Many institutions utilize private labs, partner with Universities or have their own in-house pathology department to analyze these samples. The AZA and American Association of Zoo Veterinarians (AAZV) website should be checked for any AZA bustard SSP Program approved active research requests that could be filled from a necropsy.

An extensive survey was conducted in which husbandry and medical data were collected for the majority of captive kori bustards (n=198) housed at zoos and wild animal parks throughout the United States between 1988 and 2008. The main objectives of this survey were: 1) to determine causes of morbidity and mortality in the US captive kori bustard population, 2) to collect information on demographic variables and husbandry practices currently employed in US zoos that may affect the health of these birds, and finally, 3) to identify potential risk factors for kori bustard mortality in US zoos.

Common causes of kori bustard death were summarized by Hanselmann et al. (2012). The most common clinical and pathologic findings observed were lameness (48 cases), gastrointestinal parasitism (45 cases), and wing integumentary trauma (32 cases). Trauma was a very common cause of morbidity (135 cases) and was the most common cause of mortality (52 individuals, 40% of deceased birds). Considering the high prevalence of traumatic injury and death observed historically in this population, captive management of kori bustards should focus on developing strategies that minimize opportunity for injury. Priorities include preventing exposure to potentially hostile exhibit males, decreasing stress associated with human interactions, and researching the effects of diet on skeletal development of young birds. A similar in-depth study for buff-crested bustards would be very useful.

The AZA Gruiformes TAG, AZA Kori Bustard SSP®, and Buff-crested Bustard SSP® do not currently have any specific recommended protocols for bustard euthanasia within zoos. Veterinarians at each institution are encouraged to contact the AZA Kori Bustard SSP® Veterinary Advisor or the AAZV (American Association of Zoo Veterinarians) for more specific information or advice on the most effective, safe, and humane approaches to utilize. Each institution housing bustards should have a euthanasia protocol in place, developed by the veterinary team, in case euthanasia becomes necessary in a particular situation. The AZA Animal Welfare Committee also encourages each institution to develop a process to determine when elective euthanasia might be appropriate from a quality of life perspective, taking into account behavioral, health, social, nutritional, and animal caretaker perspectives. Examples of approaches used by institutions are available from the AZA Animal Welfare Committee.

Eggs that did not hatch should be opened and checked for fertility and age of embryonic death. Bacterial cultures should be taken of the yolk/albumin or embryo to identify bacterial infection as a cause of embryonic death. Representative tissue samples from the body organs should be submitted for histopathologic examination. Thorough necropsy examination and records will aid assessment of the overall health, and causes of morbidity and mortality in bustard collections. In turn this should lead to better husbandry, management and treatment of the collection.

See Appendix J for the AZA Kori Bustard SSP® standardized necropsy protocol. This protocol could be used for buff-crested bustards if desired. Bustards display normal avian anatomy, which should be reflected in histopathological results.

Chapter 8. Reproduction

8.1 Reproductive Physiology and Behavior

It is important to have a comprehensive understanding of the reproductive physiology and behaviors of the bustards in our care. This knowledge facilitates all aspects of reproduction, artificial insemination, egg laying, hatching/rearing, and even contraception efforts that AZA-accredited zoos strive to achieve. At this time, natural conception is the only type of breeding that has been achieved with bustards. More research is needed into hormonal changes and tracking methods to better manage bustard breeding. See the *Ex situ* Kori Bustard Report on the ASAG website (https://de0046ae-67da-48da-9d60-85f9fe01aee2.filesusr.com/ugd/df9b5b_422089b2c6114efdbc81d4579544019c.pdf) for information on a study comparing the success of hand reared vs parent reared kori bustards.

Kori bustard: In the wild, kori bustards are polygynous, and males engage in lek-like behavior during the breeding season (Hallager & Boylan, 2004). Male kori bustards become sexually mature between 4–6 years of age, and females sexually mature between 3–4 years of age, although sexual maturity may occur earlier in zoos (Hallager & Boylan, 2004). The oldest male to breed successfully in a zoo was 27 years old, while a 28-year-old female was the oldest to lay a viable, fertile egg (Hallager & Boylan, 2004). The timing of the breeding season in animals housed in zoos varies throughout the United States, with breeding commencing as early as February in southern zones, and ending as late as October in northern zones. Once started, male reproductive displays reach their maximum several weeks after their initial onset. Monthly weighing of males (see Figure 2 below) has demonstrated seasonal weight gains of as much as 4 kg (8.82 lb.) immediately before and during this period.

In the wild, males tend to associate with females only during the breeding season (Osborne & Osborne, 1998). However, in zoos, single males can be housed with groups of females throughout the year (see section 5.1). One month prior to the breeding season, males that are housed together should be separated from each other. If monthly weights are being monitored, keepers should separate males when there is an observed weight increase in any of the males, and especially if it is shown by the alpha male. Reproductively active males and females can become more aggressive to both conspecifics and keepers during the breeding season. Separated males should be housed in enclosures that continue to meet the behavioral needs of the animals, as outlined in section 2.1.

During the breeding season, displaying males produce a low-pitched 6-noted booming noise as part of their mating display (Hallager & Boylan, 2004). Males will display in the presence and absence of females, but displays typically intensify when males are able to see females (Hallager & Boylan, 2004). The various stages of copulation are initiated by a receptive female in response to the male's display. These stages include (Hallager, 2003):

- Pre-copulation: The sequence of copulation begins with the female laying down near the displaying male. Once seated, the male approaches from behind either in full display (neck inflated and wings drooping) or in partial display (neck inflated and tail feathers raised). The male stands over the female and pecks at the back and sides of her head in a slow, deliberate manner, and with his tail and crest feathers raised slightly. The male stands for 5–10 minutes, alternately pecking at the female's head and stepping from side-to-side behind her, before he sits down on his hocks and continues pecking at the back of her head for another 5 minutes as the female remains seated.

- Copulation: After 10–15 minutes of head pecking, the seated male moves closer towards the sitting female using his hocks to progress forward. When close enough, the male spreads his wings and mounts the female from behind. Actual copulation lasts no more than a few seconds.

- Post-copulation: Immediately following copulation, both birds rise and part quickly, violently shaking their feathers. Females often make a "barking" sound as they move away from the male. Following copulation, both birds resume other activities, with the male often resuming courtship display behaviors.

Since copulation involves a lengthy period of head pecking by the male on the female (Hallager 2003), females should be closely monitored during the breeding season for signs of head injuries. While injuries will likely be minimal (feather plucking, bruising, and abrasions), keepers should watch for more serious injuries to the head.

Females typically begin laying 4–6 weeks after males have begun to display. Females can lay eggs as early as February in southern zones, and as early as May in northern zones. Some females lay only one egg per clutch, while others lay two eggs; the average clutch size is 1.5 eggs (Hallager, S. 2018). Females will lay replacement clutches if previous clutches are pulled. Aggressive behavior during the breeding season (between males, or directed from males to females) does not apply solely to adult males; adult females exhibit aggressive behaviors as well. Unlike males, fatal aggression between females has not been reported. Hens will aggressively defend their nesting site from other females, males, and animal caretakers. Females that are compatible with other females or males during the non-breeding season can become aggressive to other birds during breeding, and in some cases will not tolerate other females (e.g., subordinate birds) being in close proximity (Hallager & Boylan, 2004). In some cases, nesting females may need to be provided with a separate area to set up a nest, with the other females blocked from entering this area.

The following table (Table 21) provides details on incubation, pipping, and hatching durations of bustard eggs:

Table 21. Incubation data for kori, buff-crested, and white-bellied bustards (Bailey & Anderson, 2000)

Bustard species	**Incubation start to internal pip (days)**	**Time to external pip (hours)**	**Time to hatch from external pip (hours)**
Kori	21 (20–22) n=10	13 (3–24) n=10	26 (6–39) n=20
Buff-crested	19 (17–24) n=15	13 (4–24) n=14	18 (6–44) n=34
White-bellied	19 (19–19) n=3	19 (14–24) n=2	26 (8–38) n=5

AZA Institutions breeding kori bustards should submit an annual egg report to the AZA Kori Bustard SSP® Coordinator detailing egg size and general egg production (see Appendix H) parameters. Information on the egg size, dimension, and composition of kori bustard eggs (specifically infertile eggs) is provided by Anderson and Deeming (2002), and these data can be used for reference.

Buff-crested bustard: Sexual maturity has been documented in a buff-crested female in managed care that bred at the age of 21 months at the San Antonio Zoo, TX and 22 months at the National Avian Research Center, Abu Dhabi (Hallager, S. 2017). Longevity records for buff-crested bustards in managed care approach over 25 years.

Buff-crested bustards are generally found alone. Reports of pairs may pertain to females with older chicks. There is no indication of permanent pair bonding. In Kenya, breeding season starts around March and extends to August. In Uganda, breeding runs from December through March, while in Ethiopia and Somalia breeding commences in March and runs through June.

The breeding display of the male is quite spectacular, and is of the 'aerial' type. The male begins the display by calling in volume. It is thought that males call from traditional display areas. The call has been described in detail by Chappuis, Erare, and Morel (1979). After calling, the male flies into the air upwards of 30 m (98.4 ft.) and then turns on his back with his feet facing up. Next, he flips over and falls to the ground with his wings spread out. The display is geared towards attracting a female and may also define territories among males. When a female is encountered, the feathers on the crown of the male are erected to form a crest. The throat and neck feathers are puffed out. After copulation, the male leaves and resumes displaying to attract another female.

Since male buff-crested bustards perform spectacular aerial displays of great height during the breeding season (Johnsgard, 1991), appropriately tall enclosures must be provided to allow birds to engage in this courtship behavior while also prevent the birds from injuring themselves against the ceiling. The presence of the sight and sound of several males may be important in stimulating reproductive behavior in buff-crested bustards (Siegel, Hallager, & Bailey, 2007). Females can be housed in enclosures adjoined by two or more other enclosures containing single males. In this case, the males are stimulated by the vocalizations of the other males, and providing females with the choice of multiple males may also be stimulating. Buff-crested bustards have also bred successfully when housed in a compatible male-female pair within a small (3 m diameter) public exhibit (Siegel, Hallager, & Bailey, 2007).

Female buff-crested bustards make no nest in the traditional sense. Rather, the eggs are laid on the ground in a shallow scrape. Often, the nest is near a clump of grass. The usual clutch is two eggs. The eggs are olive brown to pinkish-buff with dark brown marks. Incubation periods range from 19–21 days. The chicks are precocial and able to follow their mother around a day or so after hatching. Fledging occurs at about four or five weeks, although the young often remain with the female for several months after this event. In 1992, Dallas Zoo first reported that chicks were observed being carried by their mothers under her wing (Falzone, 1992). This occurred most often when the female sensed danger.

In buff-crested bustards, the laying of the first infertile egg of the season can be used as an indicator that the female is ready to be introduced to males for breeding (Siegel, Hallager, & Bailey, 2007). At this time, females should be given access to the male's enclosure. Typically, males will enter the female's enclosure and copulation will occur. The male and female can be separated between 30 minutes and three hours after introduction. These short periods of introductions can be continued until the female lays her next egg. At this point, introductions should be stopped.

Female buff-crested bustards will usually stay on the nest scrape for 1–2 days before laying (Siegel, Hallager, & Bailey, 2007). Two eggs are typically produced, and each egg weighs approximately 45 g (0.1 lb.). Incubation lasts 19–22 days and is performed entirely by the female. Females will leave the nest site once or twice daily to eat and preen. Egg laying by female buff-crested bustards at NARC occurred in all months between January and September (Anderson, 1998a). Chicks can stay with their mother until the beginning of the next breeding season (Siegel, Hallager, & Bailey, 2007).

Figure 1. Buff-crested bustard chick under wing of dam. Photo credit C. Falzone

Reproductive monitoring in bustards: Monitoring weight increase in male kori bustards is an effective, non-invasive approach to determine when levels of reproductive hormones are beginning to increase. Sexually active male koris show an increase in body mass during the breeding season, and it appears that the alpha-male shows a significantly greater increase in body weight than the beta-male (where other males are present). Monitoring changes in the weight of males is especially important for institutions that house male kori bustards in physical contact during the non-breeding season; when weights in the alpha-male begin to rise, the males should be separated to prevent aggression. Preliminary analysis of testosterone levels in a male kori bustard at one institution (Figure 2) shows testosterone levels rising at the same time male weight gain begins (Hallager & Lichtenberg, 2007).

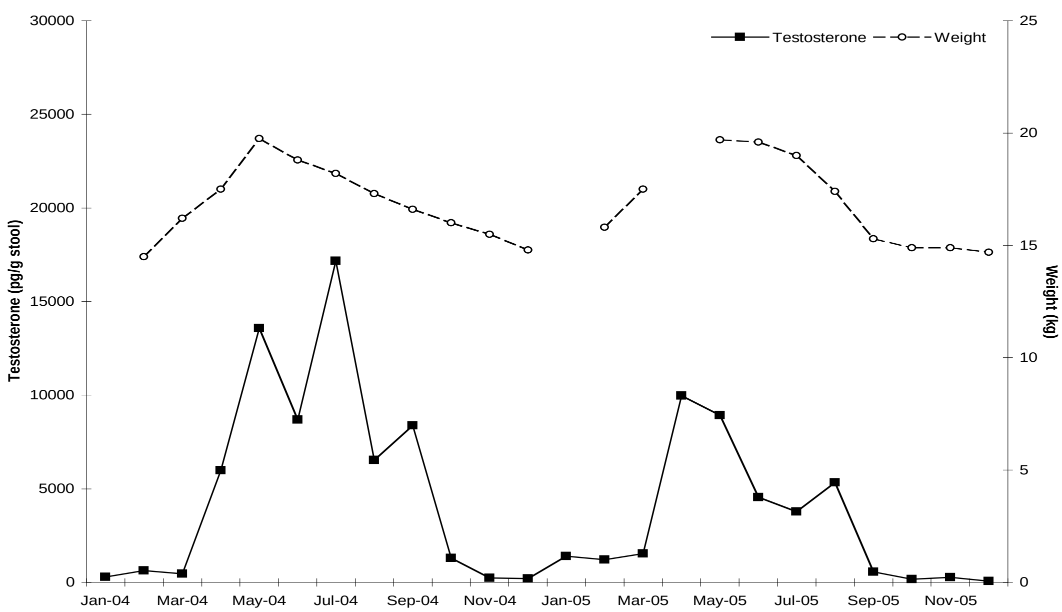

Figure 2. Testosterone and weight changes in a male kori bustard at Institution A
(Hallager & Lichtenberg, 2007)

8.2 Assisted Reproductive Technology

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

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

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

Due to the complexity and inherent risk of transporting bustards between institutions for breeding attempts, use of AI stands as a beneficial means to improve the mean kinship of bustards in zoos but requires research into all areas associated with the development of using chilled and cryopreserved semen in order for semen to be shipped. In particular, the characterization of female reproductive hormones and temporal relationships of such hormones with physiological events such as ovulation.

Hand-reared males are excellent candidates for semen collection, as they show a willingness to copulate with objects that resemble a seated female, and are not disturbed by the presence of nearby caretakers attempting to collect the semen. The process of inseminating females does require handling, which can increase the risk of injuries to both animals and their human caretakers, but some females may be conditioned to present their cloaca to familiar caretakers (L. Murphy, personal observation). The time of insemination should coincide with the female's most fertile period, and this is best determined by running hormonal assays. The expense and the need for facilities and equipment to perform this type of hormonal analysis is a limiting factor for the use of AI techniques in zoos with bustards.

Bustard semen collection, insemination, and hatching have been successfully in three species of bustard: the houbara bustard (*Chlamydotis undulata macqueenii* and *Chlamydotis undulata undulata*), great bustard (*Otis tarda*), and rufous-crested bustard (*Lophotis ruficrista*). Houbara bustard semen collection techniques have been successful in the United Arab Emirates (Jalme & van Heezik, 1996). At the National Aviary Research Center (NARC), semen was collected on rufous-crested bustards and females were successfully inseminated (anonymous). Researchers in Spain have been artificially inseminating great bustard (*Otis tarda*) using methods similar to houbara.

Jalme, Gaucher, and Paillat (1994) provide some specific information on the results of Houbara bustard artificial insemination approaches at Prince Saud al-Faisal Wildlife Research Center in Taif:

- Mean volume of ejaculate was recorded as 0.08 ml
- Mean sperm concentration was 3.5×10^8 spermatozoa ml^{-1}
- Mean number of spermatozoa per ejaculate was approximately 2.0×10^7
- Mean quantity of spermatozoa produced per week by fully sexually mature houbara bustards was 165×10^6
- The intra-individual variation in the number of spermatozoa per ejaculate was attributed to seasonal variation
- There was a statistically significant positive correlation between egg fertility and the quantity of sperm inseminated
- The median sperm storage duration for females was 10 days, with a maximum storage duration of 22 days
- The duration of sperm storage was related to the number of spermatozoa inseminated

- The greatest proportion of fertile eggs was obtained when >10^6 spermatozoa were inseminated between 3–6 days before laying
- Embryo mortality was found to increase when inseminations were performed more than 10 days before laying

At NARC, semen characteristics of five male rufous-crested bustards include:

- Mean volume of ejaculate was 0.02 ml
- Mean sperm concentration was 6.62 × 10^9/ml
- Mean number of spermatozoa per ejaculate was 1.32 x 10^8 million

The hand-reared males at NARC displayed around and copulated with objects without the need to modify them to resemble a female bird as found to be necessary in *Chlamydotis undulata macqueenii* and *Chlamydotis undulata undulata*. In rufous-crested bustards, differences were observed between males in terms of what they accepted as a copulatory stimulus. Some males copulated with a gardening glove, while others would only copulate with footwear and some discriminated between different kinds of shoes.

At Centro de Cría de Aves Esteparias in Spain, artificial insemination has been successful with hand-reared great bustard (*Otis tarda*) males (see http://www.avutardas.com/Inseminacion.html for more information).

Further research is needed to determine if these research findings are applicable to all bustards and to develop appropriate protocols for the use of artificial insemination with kori bustard and buff-crested bustards. General information on semen collection, artificial insemination, and cryopreservation of semen in non-domestic birds is provided by Gee et al. (2004).

8.3 Egg-laying/Incubation/Hatching

It is extremely important to understand the physiological and behavioral changes that occur throughout an animal's oviposition.

Kori bustard: Female kori bustards typically begin laying in May in northern zones of the United States, and begin laying as early as February in southern zones. Egg laying generally occurs 3–6 weeks after males begin booming as part of their courtship displays. Kori bustard females often pace excessively 2–3 days prior to egg laying, and typically do so around the area where egg laying will occur (Hallager & Boylan, 2004). Keepers should monitor this behavior and observe the location of the pacing so that the area can be inspected daily for eggs. However, not all females pace prior to egg-laying, and more research is needed to identify other behaviors that may reliably indicate imminent egg-laying. Males should generally be moved to another enclosure when females are incubating eggs, and other females may have to be separated as well, depending on the behavioral interactions shown by the birds (see sections 8.1 and 8.4).

Kori bustard eggs generally hatch on the 23rd day of incubation. The average clutch size for kori bustards is 1.4 eggs (Hallager & Boylan, 2004). Most females lay only one egg per clutch. The shortest inter-clutch interval for kori bustards in North American zoos has been documented as 16 days (Hallager & Boylan, 2004). On average, kori bustard eggs are 149 g (0.3 lb.), 57.6 mm (2.3 in.) wide, and 82.2 mm (3.2 in.) long (Hallager & Boylan, 2004). Not all females incubate their eggs after laying. Unattended eggs should be placed in an incubator as soon as they are found to minimize breakage and reduce bacterial infection. Keepers should continue to inspect the area where the first egg was found, as some females lay a second egg two days after the first. Egg binding has not been reported in kori bustards.

If eggs have been artificially incubated while the female sits on dummy eggs (see section 8.5 for additional information on artificial incubation), the eggs should be returned to the brooding female at internal pip if the chick is to be parent-reared (Hallager & Boylan, 2004). The expected pip-to-hatch time is 8–24 hours. No assistance should be provided to hatching chicks until at least 24 hours following external pipping (Hallager & Boylan, 2004), or if the chick appears to be weak. If the chick has not hatched after 24 hours, radiography, endoscopy, and ovotomy can be performed to determine the status of the embryo. Assisted hatching techniques can be successful, but survival of chicks is lower if they are used (Bailey & Anderson, 2000). Following assisted hatches, chicks may require supportive care in the form of subcutaneous fluids, vitamin injections, and antibiotics.

Once chicks have hatched, they should be left with their dam for 24 hours to allow normal imprinting and bonding; a quick medical assessment should be performed after 24 hours have passed, so that chicks can be examined, weighed, the umbilicus cleaned, and a determination made as to whether appropriate yolk-sac reabsorption is occurring (Hallager & Boylan, 2004). Morphometric measures can also be taken at this time. The chick should be returned to its mother as quickly as possible.

When chicks are being fed or brooded, they produce a light chirp or purring sound (Hallager & Boylan, 2004). In response to a negative stressor in the environment, chicks produce a cry that sounds like a long, sad whistle that can escalate into loud wailing cry (Hallager & Boylan, 2004). Chicks as young as 2 weeks of age will also bark when alarmed.

Buff-crested bustard: Hatch seasonality for buff-crested bustards is typically April through August in North America, though they have been documented to lay year-round (Hallager, 2015). In buff-crested bustards, the laying of the first infertile egg of the season can be used as an indicator that the female is ready to be introduced to males for breeding (Siegel, Hallager, & Bailey, 2007). At this time, females should be given access to the male's enclosure. Typically, males will enter the female's enclosure and copulation will occur. The male and female can be separated between 30 minutes and three hours after introduction. These short periods of introductions can be continued until the female lays her next egg. At this point, introductions should be stopped (Siegel, Hallager, & Bailey, 2007). Males should be moved to another enclosure when females are incubating eggs, and other females may have to be separated as well, depending on the behavioral interactions shown by the birds.

Buff-crested bustard eggs generally hatch after 19–21 days of incubation. The average clutch size is one egg (Hallager, 2017). The shortest inter-clutch interval has been documented as 11 days (Hallager, 2017). On average, eggs are 47.04 g (0.1 lb.) (n=32 eggs), 41.8 mm (1.6 in.) wide, and 51.03 mm (2.0 in.) long) (n=33 eggs) (M. Herry, personal communication, 2016). Not all females incubate their eggs after laying. Unattended eggs should be placed in an incubator as soon as they are found to minimize breakage and reduce bacterial infection. Keepers should continue to inspect the area where the first egg was found, as some females lay a second egg two days after the first.

If eggs have been artificially incubated while the female sits on dummy eggs, the eggs may be returned to the brooding female at internal pip if the chick is to be parent-reared. The expected pip-to-hatch time is approximately 24 hours. A 24-hour period between internal and external pip and 24 hours between external pip and hatch has been observed (M. Herry personal communication). No assistance should be provided to hatching chicks until at least 24 hours following external pipping or if the chick appears to be weak. If the chick has not hatched after 24 hours, radiography, endoscopy, and ovotomy can be performed to determine the status of the embryo. Assisted hatching techniques can be successful, but survival of chicks is lower if they are used (Bailey & Anderson, 2000). Following assisted hatches, chicks may require supportive care in the form of subcutaneous fluids, vitamin injections, and antibiotics.

Once chicks have hatched, they should be left with the dam for 24 hours to allow normal imprinting and bonding; a quick medical assessment should be performed after 24 hours have passed, so that chicks can be examined, weighed, the umbilicus cleaned, and a determination made as to whether appropriate yolk-sac reabsorption is occurring. Morphometric measures can also be taken at this time. The chick should be returned to its mother as quickly as possible.

When chicks are being fed or brooded, they produce a light chirp or purring sound. In response to a negative stressor in the environment, chicks produce a cry that sounds like a long, sad whistle that can escalate into loud wailing cry.

At hatching, the mother's diet should be switched from fuzzies to pinkies on a weight basis; all other items will be finely chopped for the first week. Live crickets will be discontinued for the first two weeks, and giant mealworms are discontinued for the first three weeks. The entire diet will then be divided into 3 feedings per day with the final feeding left until dark. The dam's diet consists of:

- ¼ cup Soaked softbill pellets
- 2 soaked Marion® pellets
- 1/8 cup Chopped fruit mix
- 1 tbsp. Mixed vegetables
- 2 tbsp. Mixed greens
- 1 tsp. Mazuri® Exotic Gamebird Maintenance
- 1 tsp. Scratch grains
- ½ tsp. Chopped boiled egg
- 14 g (0.5 oz.) Fuzzies
- 10 Small mealworms

- 10 Crickets, waxworms, giant mealworms (do not feed giant mealworms for 3 weeks post hatch) or 10 live crickets (do not feed live crickets for 2 weeks post hatch)

Weights for the chick should be taken 24 hours after hatch and then every other day for the first week. After that, they continue every 4 days. At Day 61, the chick should be fully feathered and adult-sized. At that time, the chick can be switched to an adult buff-crested bustard diet.

8.4 Hatching Facilities

As oviposition approaches, animal care staff should ensure that the dam is comfortable in the area where the hatch will take place, and that this area is "chick-proofed." The area should be considered "chickproofed" when the chicks do not have access to the water and there are no small areas that chicks can get stuck in. During the breeding season, modifications to exhibit husbandry may be necessary to ensure the health of eggs and chicks, and to minimize disturbance to nesting birds. Females that are suspected of getting ready to lay an egg should not be removed from the exhibit or handled as it could cause egg bound issues. Nests should be monitored so that eggs are found and documented when they are laid. To decrease the stress on the birds, consider installing nest cams. Keepers should monitor nest activity throughout the day. Checking for eggs should be performed cautiously, to avoid flushing the birds from their nests. Frequency and timing of egg checking can be based on a schedule or dictated by the birds' behavior.

New eggs can be weighed and measured at the onset of incubation but caution is advised as this may cause the dam to desert her nest. Fertility in bustard eggs is very challenging to detect due to the dark coloration and often the only sign the egg is fertile, is the movement of the egg when placed on a hard surface 7 days prior to hatch. Frequency of weighing and candling the egg during development should be based on the bird's behavior. Extra care should be taken when removing the egg from an incubating parent, as some birds will peck at the handlers' face. Safety glasses may be worn for eye protection. Placing a warmed dummy egg in the nest during the process of checking the egg will reduce the risk of nest abandonment. Dummy eggs are warmed in the keeper's hands. All weighing and candling equipment should be ready before the egg is removed so that the whole process can be completed in under a minute.

Kori bustard: The Kori Bustard SSP® recommends hand-rearing kori bustards in pens that are uncovered. Parent rearing, although not common, is also appropriate but should only be undertaken in covered pens. To encourage laying, females should have a separate area from the male for nesting, as breeding males will chase females from their nesting sites. In the wild, kori bustard nests are typically shallow scrapes in the soil that are sometimes lined with sticks, grass stems, and mammal pellets (Mwangi, 1988). The nests are often partially hidden and usually located near a grass clump or a rock. Osborne and Osborne (1999) and Mwangi (1988) provide evidence that the location of kori bustard nests may make use of plants and trees that offer shade throughout the day, and that partially obscure the sight of the nest. During incubation, females have been observed to throw sticks, grasses, and leaves onto their back, and this behavior may also be related to camouflage (S. Hallager, personal observation, 2007).

In managed care, kori bustard females will also make a small scrape in the ground where the eggs will eventually be laid. The females spend increasing amounts of time in the area, and become protective of it. Once the eggs have been laid, incubating females rarely leave the nest during the 23-day incubation period. When females do leave the nest (e.g., at feeding times or to perform comfort activities such as sunning or dustbathing), they are generally aggressive towards keepers and other birds, and will return very quickly to the nest site. If another bird approaches the incubating female, she will aggressively drive them away or run away to distract the aggressor (Hallager, 2009). During nesting/incubation, multiple females can remain together as long as no aggression is observed. If aggression is seen between birds, then the non-nesting bird should be removed to a separate enclosure.

During the breeding season, care should be taken to minimize all environmental stressors and invasive animal management procedures. All non-essential work within and around the enclosure should stop, and entry into the enclosure should be restricted only to those personnel that need to be there for essential animal management, care, or enclosure maintenance reasons. During feeding times, food bowls can be placed as close to the incubating female as possible, without actually disturbing the hen (Hallager & Boylan, 2004) although often times females will not eat during incubation. Some females can be hand fed while incubating (Hallager, personal communication). Female aggressiveness towards keepers varies between individual animals. Some individuals will become aggressive when caretakers are 1.5–3 m (5–10 ft.) away; others will let animal caretakers remove the eggs from beneath them. Animal caretakers should wear protective eyewear when attempting to remove an egg from under an incubating female, and should take great care as females can defend their nests aggressively. Some females may defend their nest more aggressively than others may, and two keepers may be needed to remove the egg. If eggs do need to be removed (e.g., for candling, artificial incubation, etc.), the best approach is to reach under the female from behind.

The dam's diet should be switched from mice to fuzzies on a weight basis when the chick hatches and all other items are finely chopped for the first week. The entire diet is divided into three feedings per day with the final feeding left until dark. A typical sample diet for parent reared kori bustards is:

- Soaked pellet (use pellet fed to adults, will vary by institution but typically a crane pellet). Amounts should be adjusted to based on environmental, cage mate presence and wild birds/rodents. A good starting point is:
- 1/2 cup Chopped fruit mix
- 2 tbsp. Mixed vegetables
- 5 tbsp. Mixed greens
- 1 tsp. Chopped boiled egg
- 5 Fuzzies, chopped the first 2 weeks
- 30 Small mealworms
- 30 frozen, thawed Crickets

No live crickets or giant mealworms should be fed for the first three weeks

After a few days, chicks begin picking up food on their own, but still rely on the dam for the majority of their food intake. The dam feeds the chick by picking up a food item and feeding it directly to the chick. Chicks reach adult size in about at 8 months. A dish of live food items (dusted with vitamin and mineral supplements) should be provided to the female and chicks three times a day for the first month (Anderson 1998b). The provision of live food can be reduced to once a day as the chicks begin to feed for themselves.

Weights for the chick should be taken 24 hours after hatch (if tolerated by the female) and then every other day for the first week. After that, continue every 4 days as tolerated by the dam. At Day 60, the chick can be switched to an adult kori bustard diet.

Buff-crested bustard: The Buff-crested Bustard SSP® recommends parent rearing buff-crested bustards when possible. A nesting female will search out a nest site and lay her single egg either directly on the ground, or in a shallow scrape in the soil. The egg is cryptically colored. Chicks are cryptically colored and precocial at hatch, but depend on the dam for their entire diet for the first few days of life. Males play no part in incubation or chick rearing and should be removed from the enclosure housing the female following copulation. Twenty-four hours after hatch, the chick may be examined to ensure it is healthy. An umbilical swabbing and weight should be obtained at this time. The dam's diet should be switched from fuzzies to pinkies on a weight basis, all other items are finely chopped for the first week. The entire diet is divided into three feedings per day with the final feeding left until dark. At one institution, the dam's diet consisted of:

- ¼ cup Soaked softbill pellets
- 2 soaked Marion® pellets
- 1/8 cup Chopped fruit mix
- 1 tbsp. Mixed vegetables
- 2 tbsp. Mixed greens
- 1 tsp. Mazuri® Exotic Gamebird Maintenance
- 1 tsp. Scratch grains
- ½ tsp. Chopped boiled egg
- 14 g (0.5 oz.) Fuzzies
- 10 Small mealworms
- 10 Crickets

The live crickets were discontinued for the first two weeks, and the giant mealworms were discontinued for the first three weeks.

After a few days, chicks begin picking up food on their own, but still rely on the dam for the majority of their food intake. The dam feeds the chick by picking up a food item and feeding it directly to the chick. Chicks reach adult size in about 4–5 weeks. A dish of live food items (dusted with vitamin and mineral supplements) should be provided to the female and chicks three times a day for the first month (Anderson 1998b). Mealworm larvae are a recommended food item, and immature house crickets can be offered when the chicks are older. The provision of live food can be reduced to once a day as the chicks begin to feed for themselves. Females may also strip seeds from grass seed heads growing in the enclosure, and feed these to the chick (Siegel, Hallager, & Bailey, 2007).

Weights for the chick should be taken 24 hours after hatch and then every other day for the first week. After that, continue every 4 days. At Day 61, the chick should be fully feathered and adult sized. At that time, the chick can be switched to an adult buff-crested bustard diet. Caretakers should watch for signs of aggression from the dam to the chick, which can occur as early as 45 days (M. Herry, personal communication, 2017).

Vitamins: Breeder vitamins (50% Rep-Cal® without vitamin D; 50% Vionate®, by weight) should be added at a rate of 1/8 tsp. for every 10 g (0.35 oz.) of the chick's food (see the table below). Note, the adult's diet is nutritionally sound and does not require additional supplementation. The vitamins should be added directly to the chick's and dam's daily amount of insects (the dam primarily feeds the chick insects during rearing).

Table 22. Additional items added to female buff-crested bustard diet when a chick is present

Day	Items	Amount
1–7	Chopped pinkies, cricket bodies, small mealworms, and waxworms	10 g (0.35 oz.) (2.5 g of each)
	Vitamins	1/8 tsp.
8–14	Chopped pinkies, cricket bodies, small mealworms, and waxworms	20 g (0.7 oz.) (5 g of each)
	Vitamins	¼ tsp.
15–20	Fuzzies, crickets, small mealworms, and waxworms	30 g (7.5 g of each)
	Vitamins	3/8 tsp.
21–28	Fuzzies, crickets, small mealworms, waxworms, and giant mealworms	50 g (1.8 oz.) (10 g of each)
	Vitamins	½ tsp.
29–60	Fuzzies, crickets, small mealworms, waxworms, giant mealworms, chopped fruit, soaked softbill pellets, mixed veggies, gamebird maintenance, scratch grains	85 g (3 oz.) (8.5 g of each)
	Vitamins	5/8 tsp.

8.5 Assisted Rearing

Although mothers may successfully hatch young, 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 when deemed necessary. Intervention may be warranted in cases where the dam has a health concern, or where she is not observed to regularly feed a chick or a chick fails to thrive.

Institutions should be familiar with expected incubation behavior for bustards as described in section 8.3, in order to properly manage eggs on the nest.

Kori bustard and buff-crested bustards:

1) On the first day after being laid, eggs should be taken from the incubating female for weighing, measuring, and candling. During this procedure, the egg taken from the female should be replaced with a dummy egg warmed to 37.5 °C (99.5 °F). The removed egg should be placed in an artificial incubator (e.g., Grumbach) at 37.5 °C (99.5 °F) and 55% relative humidity.

2) The target weight loss for bustard eggs is 12–15%. The humidity in the incubator should be adjusted to result in this weight loss. Eggs should be turned every two hours. Humidity should be increased to 70–80% once candling shows that the air cell within the egg begins to drop down. However, many kori bustard eggs are too dark to candle effectively, and in these cases, humidity should be increased at internal pip.

The following tables (Table 23 and Table 24) provide a summary of the artificial incubation protocols for kori bustard and buff-crested bustard eggs used at several institutions. While the incubators used at these facilities have been successful, other models may be appropriate as well.

Table 23. Artificial incubation protocols for kori bustard eggs

	Institution A protocol	Institution B protocol
Incubator	Grumbach® incubator	Petersime® model 1 or Humidaire® model 21 incubator
Temperature	37.5 °C (99.5 °F)	37.5 °C (99.5 °F)
Humidity/wet bulb	50–55%	26.7–30°C (80–86°F)
Egg turning	Every 4 hours	Every 1–2 hours, with additional 180° turn twice daily
Egg cleaning	No	Wiped with dry sponge
Egg weight loss	---	15%
Moved to hatchery	Internal or external pip	Day 21–22

Table 24. Artificial incubation protocols for buff-crested bustard eggs

	Institution A protocol	Institution B protocol	Institution C protocol
Incubator	Grumbach® incubator	Petersime® model 1 or Humidaire® model 21 incubator	Grumbach® incubator
Temperature	37.5 °C (99.5 °F)	37.5 °C (99.5 °F)	37.2 °C (98.9 °F)
Humidity/wet bulb	50–55% RH	26.7–30 °C (80–86 °F)	40% RH
Egg turning	Every 1 hour	Every 1–2 hours, with additional 180° turn twice daily	Every 30 mins
Egg cleaning	No	Wiped with dry sponge	No
Egg weight loss	12–15%	15%	15–18%
Moved to hatchery	Internal pip	Day 21–22	When chick is pushing on air cell

If artificial incubation is performed, delayed incubation of bustard eggs is generally not recommended. Delayed incubation of houbara bustard (*Chlamydotis undulata*) eggs, where the eggs were stored for a period of time before being transferred to incubators, resulted in a 19% lower hatchability rate, with higher mortality recorded between 3–5 days of development (Jalme & Van Heezik, 1996). Bustard eggs should be placed in an incubator as soon as they are discovered if they are to be artificially incubated.

If artificially incubated bustard chicks do not hatch within established pipping intervals, or seem to be weak, then it is possible to perform radiography, endoscopy, and ovotomy, to determine the status of the embryo. Assisted hatching techniques can be successful, but survival of chicks is lower if they are used (Bailey & Anderson, 2000). Table 25 and Table 26 provide a summary of the hatching protocols for kori bustard eggs and buff-crested bustard eggs used at two institutions. While these protocols have been successful for these facilities, other models may be appropriate as well.

Table 25. Artificial hatching protocols for kori bustard eggs

	Institution A protocol	Institution B protocol
Hatcher	Grumbach® incubator	Leahy® hatcher
Temperature	37.4 °C (99.3 °F)	36.9 °C (98.4 °F)
Humidity/wet bulb	70–75%	31.1–32.2 °C (88–90 °F)
Egg turning	None	None

Table 26. Artificial hatching protocols for buff-crested bustard eggs

	Institution A protocol	Institution B protocol
Hatcher	Grumbach® incubator	GQF® or ReptiPro®
Temperature	37.2 °C (98.96 °F)	36.6 °C (98 °F)
Humidity/wet bulb	70–75%	65–70%
Egg turning	None	None

Hand-rearing kori bustards: Hand-rearing is recommended for kori bustards in uncovered enclosures. Kori bustard chicks are vulnerable to predation by bird and mammal species that commonly occur in and around zoos in the United States. Accumulating evidence indicates that hand-rearing of kori bustards does not negatively impact the future breeding success of the chicks, and may result in animals with a better temperament for management within zoos (e.g., more tractable and more accepting of caretaker/visitor presence) (Hallager & Boylan, 2004). Numerous hand-reared kori bustards have become viable breeding birds.

Hand-rearing kori bustard protocol: With only rare exceptions, hatching occurs on the 23rd day of incubation. Hatch weights for chicks range from 77–116 g (0.17–0.26 lb.), with an average of 98 g (0.2 lb.) in chicks from an AZA-accredited institution (n=33). After hatching, the chick's umbilicus should be cleaned with povidone-iodine (100%) solution, and the chick should be weighed. If the chick is clinically dehydrated or has had difficulty hatching, 2 ml of half-strength Lactated Ringer's and 2.5% dextrose solution should be provided subcutaneously (Hallager & Boylan, 2004). After the initial assessment, the chick should be allowed to rest for several hours in the hatcher, before being moved to a brooder. Brooders that are 69.9 cm x 33 cm x 35.5 cm (27.5 in. x 13 in. x 14 in.) deep and that are kept at 36.1 ºC (97 °F) have been successfully used to hand-rear chicks. The floor of the brooder should be carpeted, and a feather duster (preferably made out of kori bustard feathers to avoid chicks getting entangled in softer feathers) hung in one corner to simulate the mother. A small mirror affixed in the brooder can also act as a visual stimulus for the chick. When brooder conditions are appropriate and chicks are provided with sufficient food and tactile stimulation, they will often produce a light chirp or purring sound, especially when feeding (Hallager & Boylan, 2004). Chicks in conditions that are not meeting their needs can produce a long, sad whistle that can escalate into a loud wailing. The nutritional requirements of hand-reared chicks are covered in sections 6.1 and 6.2.

At 4–5 days after hatching, hand-reared chicks can be placed with other chicks, provided that the older chick is less than two weeks old. Chicks less than 1 week old should not be placed with other chicks that are >2 weeks of age, as older chicks have the capacity to severely (and possibly fatally) wound younger chicks. When chicks are 3 weeks old, they can be housed together with slightly older chicks, but should be carefully monitored. When introducing young chicks to each other, the older of the chicks will be aggressive towards the younger chick, but the period of aggression is generally limited to the first several hours after the initial introduction. Chicks should be observed carefully during the brief introduction period. Raising hand-reared chicks with a sibling or similarly aged conspecific reduces the likelihood of imprinting.

Chicks housed alone benefit from a mirror or a heterospecific companion (e.g., quail or guinea fowl chick) in their enclosure; the mirror acts as a calming agent, and serves to reduce the degree of imprinting that occurs. Typically, chicks reared alone have the highest degree of imprinting on their caretakers (e.g., approaching animal caretakers, readily accepting food from them). Placing the chicks in visual contact with adult kori bustards may also reduce human imprinting to some degree.

Cross-fostering and shared-rearing techniques have not been used with kori bustards in zoos in the United States, but further investigation into these approaches might be useful to determine if they are applicable to this species. For cross-fostering to be successful, the timing needs to be appropriate for the animals involved. Shared rearing is less likely to be appropriate for kori bustards, as chicks imprint on their dam, and females may attack a chick that is not imprinted. This technique has been used in Australian bustards (*Ardeotis australis*), where some highly capable females were regularly used to foster chicks from less competent mothers (P. Goriup, personal communication, 2007).

As hand-reared kori bustard chicks grow, they should be exercised following each feeding session and as often as possible throughout the day. This will minimize musculoskeletal health problems such as slipped tendons and angel wing. Access to larger, more complex enclosures should be provided as the chicks continue to develop. Containment for hand-reared chicks should follow recommendations made in section 2.2. To minimize injuries, chicks should not be housed in enclosures with barriers and obstacles that the birds can impact or trip over (Hallager & Boylan, 2004). Enclosure mesh should be smaller than 1.27 cm x 7.62 cm (½ in. x 3 in.) to prevent wings from slipping through and causing breaks. Whenever hand-reared chicks are introduced to a new enclosure, a familiar keeper should acquaint them to the new space immediately after releasing them by walking around with the birds and showing them the resources available within the enclosure (e.g., plants, fences, walls, dishes, etc.).

Complete day-to-day hand-rearing protocols for kori bustard chicks and buff crested bustard chicks are summarized in Appendix L and Appendix M. Bailey, T. A., Naldo, J., Samour, J. H., Sleigh, I. M., & Howlett, J. C. (1997) provide recommended approaches to maximize the health of hand-reared bustard chicks.

Hand-rearing buff-crested bustards: Hand-rearing is not recommended for buff-crested bustards, as there is an increased possibility of reduced breeding success caused by the chicks imprinting on animal caretakers. Male buff-crested bustards that are imprinted will display to humans, and have been known to scalp or kill females during breeding interactions (Siegel, Hallager, & Bailey, 2007). Imprinting can be reduced using isolation rearing techniques or adding a mirror to the brooder.

When buff-crested bustard chicks need to be hand-reared, the following protocols (Siegel, Hallager, & Bailey, 2007) are recommended. As soon as chicks hatch, their umbilical area should be disinfected with a solution of betadine, and placed in a hatcher to dry for 3–4 hours (Anderson, 1998b). A well-baby exam should be done 24 hours after hatch. Some facilities swab the umbilical area with betadine for the first three days after hatch. Barring any medical problems, the chick will receive its first feeding after the yolk has resorbed (24 after hatch.) The chick is expected to lose 10% of its body weight per day for the first 2 days. Chicks should gain between 2–18% body weight per day, with 10–12% (Siegel, Hallager, & Bailey, 2007) or 5–10% (Anderson 1998a, 1998b) being optimal (see Table 27 below). If diets are too high in protein, or if growth rates are too high, then bone developmental abnormalities such as angel wing or slipped leg tendons may result (see section 6.1).

Individual chicks can be raised inside brooders measuring 60 cm x 42 cm x 35 cm (26.6 in. x 16.5 in. x 13.8 in.), or in large group brooders measuring 120 cm x 60 cm x 40 cm (47.3 in. x 26.6 in. x 15.8 in.). A cotton cloth substrate should be provided for the chicks (Anderson 1998a). The temperature of the brooder should be set at 32.2–35 °C (96–99 °F) initially and reduced to 29.4 °C (85 °F) by the end of the second week (Siegel, Hallager, & Bailey, 2007). Providing a large, sterilized feather duster (preferably made out of buff-crested bustard feathers to avoid chicks getting entangled in softer feathers) in one corner of the brooder will provide the chick with a surrogate mother; the presence of a mirror or another similarly aged hatchling bustard will also decrease imprinting. By 10–20 days of age, chicks can be moved to a larger outside area (3 m x 3 m (9.8 ft. x 9.8 ft.), with a sand substrate, and heat bulbs positioned to provide variable temperature gradients within the enclosure. Patches of fine sand enable the birds to engage in dust-bathing behaviors.

Table 27. Growth rates of bustards reared in zoos

Species	Growth Rates	Source
Kori	7.5–10% for the first month. Adult bodyweight achieved in females at 300–450 days. Adult bodyweight not achieved in males by 330 days	Anderson,1998a
Buff-crested	5–7.5% for the first month[1]	Anderson,1998b
White-bellied	7.5-10% for the first month. Adult bodyweight reached by 6 months	Anderson,1998c

[1]Anderson (1998c) reports that musculoskeletal problems were seen in buff-crested bustards with growth rates in excess of 10% during one season; recommended growth rates are less than 10% (Anderson 1998c).

<u>Feeding protocol</u>: The chick should be weighed first thing every morning. Chicks should not be fed for the first 24 hours after hatching. After 24 hours, food can be offered with tweezers, taking care to limit the amount of fur or chitin offered, as these may cause impaction. Food should be dipped in water and lightly dipped in a vitamin powder before being fed to the chick. Chicks will begin feeding on their own between 4–14 days of age. Chicks should be encouraged to feed by themselves as early as possible, and should be self-feeding on pellets and other diet items by one month of age (Anderson, 1998). Hand-reared

juveniles should not be provided with the full adult diet until they are three months of age (Anderson, 1998a).

Water can be offered in droplets from an eyedropper, or in a small, shallow dish containing a rock, large enough to avoid ingestion. Water in the dish should be no deeper than 1 cm (0.4 in.) to prevent drowning, and birds can be stimulated to drink from the dish by tapping the pebbles with tweezers. Appendix M provides a day-by-day protocol for hand-rearing this species.

If possible, chicks should have a minimum of 20 minutes outdoors daily after Day 10 if above 60 ˚F (watch for chick's response to the outdoor temperatures). More time is allowed if the chick is tolerating it well. Some institutions do not let bustard chicks outside until they have received West Nile virus (WNV) vaccinations so each facility must determine their comfort level with outside access.

The occurrences of splayed legs, curled toes, and slipped wing are related to genetics, growth rates, the protein content of diets, and the substrates provided to the chicks in the hatcher and brooder (Anderson, 1998a, 1998b). Treatment of these issues involved preventative and reactive treatments. In many cases, addressing multiple variables is necessary to prevent these types of developmental abnormalities from occurring. For example, high growth rates and high energy or protein diets have been linked to the development of slipped wings in buff-crested and white-bellied bustards (Anderson 1998a, 1998b). However, slipped wing issues were eliminated in hand-reared Australian bustard (*Ardeotis australis*) chicks by periodically reducing brooder temperatures to more closely mimic female brooding behavior (White, 1985). More research is needed on buff-crested bustards to determine if more constant brooding temperatures for chicks affects the prevalence of slipped wings. Care must also be taken when handling bustard chicks, as trauma to the growth plates of chicks (Wise and Jennings, 1973) during apparently routine capture and handling may lead to the twisting or bowing of long bones.

It is recommended that hand-reared chicks be reared in pairs or small groups of similarly-aged conspecifics, as socialization leads to birds that feed better, are calmer, and that are less likely to imprint on human caretakers (Anderson, 1998a). Hand-reared buff-crested bustards show a strong propensity to imprint on their human caretakers, especially if housed singly during their development (Anderson, 1998a). Males seem especially susceptible to imprinting, and this can lead to behavioral issues (e.g., aggression) occurring in the adults that reduce the possibility or success of breeding. At the National Aviary Research Center (NARC) in Abu Dhabi, some male buff-crested bustards could be housed with females without aggression problems, but the eggs produced by these pairs of birds were not fertile (Anderson, 1998a).

8.6 Contraception

Many animals cared for in AZA-accredited institutions breed so successfully that contraception techniques are implemented to ensure that the population remains at a healthy size. In the case of an animal on loan from another facility, consult the loan agreement or owner regarding authority to contracept. In the case of permanent contraception, prior permission of the animal's owner must be obtained.

The use of invasive contraceptive methods with bustards has not been described. Bustards, as with other birds, provide easy contraception management via the removal of eggs immediately at lay. If a recommendation is made not to breed a female bustard, any fertile eggs that are laid should be removed as soon as they are laid and replaced with dummy eggs. The female should be allowed to sit on her nest until she abandons it at the end of the breeding season. Eggs that are removed should always be replaced with dummy eggs in order to stop the female from laying an excessive number of eggs, as females will re-clutch in some cases. If an egg is removed, it is usually replaced with a dummy egg to discourage second clutching. The dummy egg should be warmed up in a keeper's hand before placing it underneath the bird. The dummy egg is then removed after the normal incubation period for that species. Any egg pulled should be examined to determine if it is fertile or not. In addition, the weight of the egg, the thickness of the shell, and any egg/embryo malformations should be noted. All eggs should be recorded, whether fertile or not, in support of fecundity information.

Avian egg embryo euthanasia: The disposal of fertile eggs should be done with proper consideration for animal welfare implications The AZA Kori Bustard SSP® and AZA Buff-crested Bustard SSP® and the American Association of Zoo Veterinarians (AAZV) recommend that institutions adopt the guidelines that state: "by 50% gestation the neural tube of avian embryos has developed sufficiently for pain perception, and therefore any bird embryos that old or older should be euthanized using methods appropriate for hatched birds (i.e., chemical).” The AZA Kori Bustard SSP® and AZA Buff-crested Bustard SSP® Veterinary Advisors can provide additional information and advice on effective, safe, and humane approaches to utilize. Veterinarians at each institution should develop their own euthanasia protocols that also include egg embryo euthanasia.

Chapter 9. Behavior Management

9.1 Animal Training

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

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

AZA-accredited institutions are expected to utilize reinforcing conditioning techniques to facilitate husbandry procedures and behavioral research investigations. Institutions should follow a formal written animal training program that facilitates husbandry, science, and veterinary procedures and enhances the health and well-being of the animals (AZA Accreditation Standard 1.6.4).

AZA Accreditation Standard

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

Historically, daily husbandry practices involving bustards (e.g., crating, shifting, weighing, restraint etc.) have led to health and injury problems in managed care facilities. Behavioral management of bustards utilizing basic operant conditioning techniques can help prevent these types of injuries. Bustards can be trained to step on scales so that their weight can be monitored routinely (see protocol below). Target training has also been performed to facilitate the training of animals to move to and station on a platform scale to allow their weights to be taken. The training of additional husbandry behaviors using operant conditioning is very feasible with these animals, and bustards would benefit from being trained to enter sheds/holding areas on cue (especially useful for northern facilities that have to move birds indoors routinely in colder months). Training bustards to take hand-tossed food items is useful for medicating birds. Crate training would also be useful for the occasional situations when birds have to be relocated or transferred for medical treatment, and would reduce the likelihood that birds would be injured during manual capture and restraint procedures that are otherwise used (see section 7.5). Overall, bustards are very responsive to operant training and there are opportunities to advance the field of training with these taxa.

To avoid excessive handling of kori bustards while monitoring general health and documenting seasonal weight changes in males, a 1.2 m x 1.2 m (4 ft. x 4 ft.) scale can be used to weigh birds on a monthly (or as needed) basis. Recording weight changes can assist in the social and reproductive management of these birds (see section 7.1). The scale platform should be positioned in an area where the birds feel comfortable (e.g., in front of a feeder), near a dry area where the scale indicator can be located, and in a location where keepers can remain out of sight while still being able to identify the bird and read the indicator. Indoor/outdoor carpet 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. The process of scale training can begin by placing mealworms or some other favored food item on the scale to encourage the birds to step onto it (Hallager & Boylan, 2004). Target training may also be beneficial to help move the birds to the platform within their enclosures. Once the birds feel comfortable stepping onto the scale to reach food items, routine weighing can begin. The birds should be reinforced for standing still on the platform where possible. Scale training can make monthly weighing a relatively quick process, without the need to handle or restrain the birds. Similar benefits apply to buff-crested bustards.

9.2 Environmental Enrichment

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

AZA Accreditation Standard

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

Enrichment programs for bustards should take into account the natural history of the species, individual needs of the animals, and facility constraints. The bustard enrichment plan should include the following elements: goal setting, planning and approval process, implementation, documentation/record-keeping, evaluation, and subsequent program refinement. The bustard enrichment program should ensure that all environmental enrichment devices (EEDs) are "bustard" safe and are presented on a variable schedule to prevent habituation. AZA-accredited institutions must have a formal written enrichment program that promotes bustard-appropriate behavioral opportunities (AZA Accreditation Standard 1.6.1). Enrichment activities must be documented and evaluated, and the program should be refined based on the results, if appropriate. Records must be kept current (AZA Accreditation Standard 1.6.3).

AZA Accreditation Standard

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

AZA Accreditation Standard

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

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

For bustards, some variation in their environment is important to satisfy their natural curiosity and intelligence. With approval from institution veterinarians and nutritionists, food items may be offered as a form of enrichment (see section 6.2 for additional information on food-based enrichment initiatives). Non-food enrichment initiatives can also be developed as part of a formalized enrichment program (see www.animalenrichment.org), and should provide opportunities for the birds to express their full range of behaviors (see Appendix G for a kori bustard ethogram. Behaviors for other bustard species are similar). Keepers should observe the animals' interactions with enrichment initiatives to ensure that there are no health or safety concerns. As with all taxa, safety is of utmost concern with environmental enrichment devices. Carefully examine all devices for small, ingestible pieces, parts that could easily be broken off, entanglement issues and so on. New devices should always be monitored after presentation to assure that they are safe. Food enrichment should be appropriate for the species and follow the institutional approval process prior to offering. It is also important to be sure that the devices do not cause undue stress on the animals. All devices should be examined on a regular basis to assure that there has been no degradation and if there has been they should be disposed of.

Behavioral management programs should include training, enrichment, exhibit design, and husbandry practices. They should aim to promote natural behaviors of bustards, such as walking, sunning, courtship behaviors, preening, foraging, exploring, and other forms of exercise such as flapping wings and running. Beyond normal stimuli in a zoo environment, bustards generally tend to respond with curiosity to novel objects and increase their exploratory behavior. Enrichment does not require elaborate or costly apparatus. Some facilities report good success with the use of different feeding strategies, such as multiple feedings, extended feedings, and scatter feedings.

Participation in training programs and in behavioral research programs can be enriching as they allow the bird to have differing cognitive stimulations from the normal zoo experience. Interaction and mental stimulation are important aspects of training and are essentially enriching. Training reinforcers can include items that the birds find enriching such as novel foods or favorite devices. Training and enrichment can also be utilized to address issues such as veterinary or nutritional needs. Lack of activity can be addressed by enrichment and offering different food choices and presentations can be used to deal with nutritional requirements. Training can make necessary interactions more cooperative and create an environment of choice and control.

9.3 Staff and Animal Interactions

Animal training and environmental enrichment protocols and techniques should be based on interactions that promote safety for all involved. Bustards are not aggressive by nature. The main potential for injury to an animal caretaker from a kori bustard will be during capture and restraint of the birds, and especially when males push out with their feet while they are being held. Kori bustards are very strong-legged birds, and it can be difficult to tuck them into a position to carry them safely. It is recommended to have two keepers present during capture and restraint to ensure proper safety of the bird as well as the keepers involved (see section 7.5). Although kori bustards rarely strike out with their bills, bouts of struggling during restraint procedures could result in a bird inadvertently pecking the face of its handlers. Goggles can be worn to minimize the risk of eye injuries during these procedures. Leather gloves are not recommended during restraint procedures, as they inhibit the handler's ability to hold the bird properly. Buff-crested bustards pose little threat to handlers due to their diminutive size.

Unlike cranes, kori bustards are not usually aggressive, and unprotected free contact management of these animals is the most common form of interaction between the keeper and the bird. Males can be aggressive to keepers during the breeding season, and keepers may opt to carry a broom or other soft/rigid object that can be used to push the bird away. In some cases, the use of the broom may increase aggression, as some birds focus their aggressive responses solely on the broom.

Attention to the design of enclosures and facilities housing bustards, and to the behavior of staff members working with these birds, is important to minimize trauma-related problems during human-animal interactions, such as capture/restraint and animal training. In the daily management of bustards, the likelihood of trauma to the birds can be reduced in the following ways (T. Bailey, personal communication, 2007):

- Using plastic coated foam padding to surround the sides of enclosures or pens, especially in areas where birds are regularly caught, such as in hospital or quarantine pens. Padding minimizes abrasion injuries to wingtips.
- Using shade-cloth or tension netting on the roof and sides of aviaries to cushion the impact that may result from birds flying within an enclosure (birds can still attempt flight whether they are flight restricted or not).
- Modifying behavior of the birds by habituating nervous individuals to common stimuli that may occur within the zoo environment (e.g., noises, presence of veterinarians, visitors, etc.), or housing such birds in naturalistic pens with plenty of cover to allow them to avoid potential negative stressors.
- Ensuring that birds that have not been pinioned are regularly feather trimmed to prevent the birds from getting airborne (see Appendix N for information on flight restraint methods).
- Minimizing stress by reducing the number of non-essential people who enter bustard enclosures or the off-exhibit areas directly around these enclosures.

9.4 Staff Skills and Training

Bustard staff members should be trained in all areas of bustard 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.

Animal care staff should have a complete understanding of the natural history, behavior, and biology of bustards. For the effective management of the animals, animal caretakers should be familiar with their range of vocalizations (e.g., barks, growls, and booms) and their postures/displays and should be able to capture and restrain birds quickly and safely when needed. New keepers should be trained by keepers experienced in bustard handling, whenever possible. The AZA Gruiformes TAG and bustard SSPs do not have any specific recommendations for certifications and qualifications needed by animal care staff working with bustards, but encourage all institutions to provide opportunities for animal caretakers to gain additional experience in all fields of animal management and care.

Chapter 10. Ambassador Animals

10.1 Program Animal Policy

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

Ambassador animal presentations bring a host of responsibilities, including the welfare of the animals involved, the safety of the animal handler and public, and accountability for the take-home, educational messages received by the audience. Therefore, AZA requires all accredited institutions that give ambassador animal presentations to develop an institutional ambassador animal policy that clearly identifies and justifies those species and individuals approved as ambassador animals and details their long-term management plan and educational program objectives. The policy must incorporate the elements contained in AZA's "Recommendations For Developing an Institutional Ambassador Animal Policy". If an animal on loan from another facility is used as an ambassador animal, the owner's permission is to be obtained prior to program use.

AZA's accreditation standards require that the conditions and treatment of animals in education programs must meet standards set for the remainder of the animal collection, including species-appropriate shelter, exercise, sound and environmental enrichment, access to veterinary care, nutrition, and other related standards (AZA Accreditation Standard 1.5.4). All record-keeping requirements noted previously apply to ambassador animals (AZA Accreditation Standards 1.4.1, 1.4.2, 1.4.3, 1.4.4, 1.4.5, 1.4.6, and 1.4.7). In addition, providing ambassador animals with options to choose among a variety of conditions within their environment is essential to ensuring effective care, welfare, and management (AZA Accreditation Standard 1.5.2.2). Some of these requirements can be met outside of the primary exhibit enclosure while the animal is involved in a program or is being transported. For example, housing may be reduced in size compared to a primary enclosure as long as the animal's physical and psychological needs are being met during the program; upon return to the facility the animal should be returned to its species-appropriate housing as described above.

AZA Accreditation Standard

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

10.2 Institutional Program Animal Plans

Bustards are not commonly used in formal conservation/education programs, and the AZA Kori Bustard SSP® and AZA Buff-crested Bustard SSP® have no specific recommendations for the use of either bustard species as program animals beyond the general care recommendations included within this manual. Any future recommendations will be added to this manual as they are developed by the AZA Gruiformes TAG and bustard SSPs. Institutions that utilize bustards in formal or informal programs should provide updates to the appropriate bustard SSPs in regards to the approaches used with either species.

Chapter 11. Research

11.1 Known Methodologies

AZA believes that contemporary bustard management, husbandry, veterinary care, and conservation practices should be based in science, and that a commitment to scientific research, both basic and applied, is a trademark of the modern zoological park and aquarium. AZA-accredited institutions have the invaluable opportunity, and are expected, to conduct or facilitate research in both *in situ* and *ex situ* settings to advance scientific knowledge of the animals in our care and enhance the conservation of wild populations. Participating in AZA Taxon Advisory Group (TAG) or Species Survival Plan® (SSP) Program sponsored research when applicable, conducting and publishing original research projects, affiliating with local universities, and/or employing staff with scientific credentials could help achieve this (AZA Accreditation Standard 5.3). An AZA institution must demonstrate a commitment to scientific study that is in proportion to the size and scope of its facilities, staff, and animals (AZA Accreditation Standard 5.0).

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

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

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

Not much is known about many of the 25 bustard species. They inhabit remote and difficult to access areas of the world. Because of this, there is a limited amount of information on population densities, behavior, and physiology. This group of is generally secretive and elusive. This makes it challenging to house and breed these birds in an ex situ setting. Zoos can add to the body of knowledge by studying ex situ behavior and documenting physiological data. Zoos can also contribute by providing staff and resources to assist in research in the wild.

Groups and organizations working on bustard research:

- The International Union for the Conservation of Nature (IUCN) Bustard Specialist Group (www.iucn.org)
- The Great Bustard Group http://greatbustard.org/
- Eurasian Bustard Alliance http://www.asiangreatbustard.org/
- International Fund for Houbara Conservation http://houbarafund.org/en/home
- Centro de Cría de Aves Esteparias, Spain http://www.avutardas.com/Inicio.html
- Wildlife Institute of India http://www.wii.gov.in/

Keeper research: Keepers are in a great position to contribute to bustard management and husbandry advances and research, as they work with the species on a daily basis. Some areas where keeper input and participation can be very valuable include:

- Instituting scale training so that birds can be routinely weighed in order to assess seasonal and lifetime changes in body mass.
- Documenting physical development of chicks, including data collection on weight, specific diet ingredient intake (weighed amounts), morphometrics, and plumage changes.
- Recording weight, culmen length, skull length, tibiotarsus length, tail length, and wing chord when adult birds are handled.
- Weighing and measuring all eggs (fresh weight and length/width).
- Collecting data on activity budgets of adults and chicks.
- Determining food preferences of females when feeding chicks.
- Recording space utilization by monitoring types of areas preferred by bustards, and by comparing on-exhibit birds with off-exhibit birds.
- Documenting personalities of individuals.

- Documenting molt patterns.

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

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

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

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

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

If institutions are not able to conduct in-house research investigations, they are strongly encouraged to provide financial, personnel, logistical, and other support for priority research and conservation initiatives identified by Taxon Advisory Groups (TAGs) or Species Survival Plans® (SSP) Programs.

11.2 Future Research Needs

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

Chapter 1: Ambient Environment

<u>Section 1.2 Light</u>

A study done by Fernandes and Hallager (2007) demonstrated that the act of sunbathing plays an integral role in the health and feather condition of captive kori bustards and suggests that ectoparasite control is the most plausible cause for sunning in captive kori bustards. However, additional observations are needed of birds in the wild to further elucidate the significance, frequency, and occurrence of sunning in kori bustards.

The relative importance of sunlight for bustard chick development is unknown, and more research is needed in this area. The AZA Kori Bustard SSP® recommends that chicks be provided access to natural light as soon as possible. Popular practice suggests that chicks be exposed to sunlight to some degree on a daily basis starting at Day 7. However, the exact day that this exposure should begin is subjective. Some birds that were not exposed to natural light until they were two months of age have developed satisfactorily.

<u>Section 1.4. Sound and Vibration</u>

Little is known about the hearing sensitivity of kori bustards, and additional research on hearing would provide some guidance for creating more objective recommendations for managing sound stimuli for this species.

Chapter 2: Habitat Design and Containment

Section 2.2. Safety and Containment

Covered enclosures are strongly recommended if kori bustard hens are allowed to raise chicks naturally, and it is possible that completely covered enclosures may help to minimize the risk of avian flu transmission. Further research is needed to determine the role that covered aviaries can play in minimizing the transmission of diseases from wild birds.

Pinioning kori bustards as chicks may make them more prone to injury and trauma within their enclosures, but more research is needed to determine the incidence of injuries in flight restrained and free-flighted individuals throughout the population of kori bustards managed in zoos.

Chapter 4: Transport

Section 4.1 Capture myopathy

Both wild caught and captive reared bustards are susceptible to capture myopathy (Bailey, T. A. (2008). Along with other measures such as minimalizing handling and keeping temperatures low (e.g., shipping in early morning), supplementing birds with oral vitamin E can be utilized as a preventative measure. While supplementation has not been explicitly proven to reduce the incidence of capture myopathy, there is anecdotal evidence to suggest vitamin E administration has been helpful in other species and thus is recommended for kori bustards three days prior to shipment (S. Murray personal communication 2017). Facilities housing buff-crested bustards should evaluate the effectiveness of vitamin E therapy for this species.

Chapter 5: Social Environment

Section 5.1. Group Structure and Size

There is a need to keep adult male kori bustards (>3 years old) separate during the breeding season, because dominant males may kill or severely wound subordinate males. It is recommended that adult males be housed separately at all times. Where multiple males are maintained together, more research is recommended to determine the appropriate conditions (if any) for being able to house these males together all year round.

By nature, kori bustards are generally solitary animals, except for females with chicks. In some instances, kori bustards in the wild have been observed to form small, semi-social groups during the non-breeding season (Hallager & Boylan, 2004). In the wild, chicks separate from their mother at the start of the following year's breeding season. In managed care facilities, data are limited, but it is also recommended that young be removed from the enclosure before the onset of the next year's breeding season. More research could be done on this topic.

Chapter 6: Nutrition Requirements

Section 6.1. Nutritional Requirements

Additional research that focuses on exact daily food intake and energy expenditure for bustards, and that covers all life stages (e.g., chick, juvenile, reproductive adult, senescent adult), will be important to perform so that more specific nutritional requirements and recommendations can be developed for kori bustards. Likewise, limited published data are available regarding successful hand-rearing diets for kori bustard chicks (Maslanka & Ward, 2003; Hallager, 2005) or buff-crested bustards.

Section 6.2 Diets

The diet of wild buff-crested bustards is poorly studied, but individuals from this species are known to eat seeds, vegetation, berries, acacia gum, and insects (e.g., scarabs, beetle larvae, grasshoppers) (Siegel, Hallager, & Bailey, 2007.). More information regarding the specifics of buff-crested bustard diets in the wild would be beneficial.

Section 6.3. Nutritional Evaluations

More information is needed from blood samples collected from clinically 'normal' kori bustards. It is recommended that blood samples be taken opportunistically (e.g., during routine physicals), and analyzed for nutritionally related information that can be used to develop appropriate target serum and nutrient values.

Other than body weight evaluations, there are currently no clinically valid nutritional evaluations that have been developed for kori bustards to assess growth, seasonal changes, etc. Huchzermeyer (1998) provides a scale (1–10) for scoring body condition in ostrich and Bailey (2008) provides descriptive text for assessing weight, hydration, cere, nares, beak, oropharynx, eyes, ears, pectoral muscle condition, neck, saccus oralis, body, coelomic space, vent, thoracic and pelvid limbs, feathers, and skin. A grading system for overall body condition that takes into account the entire body of the bird should be developed for bustards to assist in proper husbandry.

Chapter 7: Veterinary Care

<u>Section 7.3 Quarantine</u>

Preliminary work to assess nutritional status and disease exposure is underway, and banked blood samples could help supplement these projects.

<u>Section 7.4 Preventative Medicine</u>

Additional information is needed to determine the natural molt pattern of kori bustards, and whether this pattern is affected by diet, local environmental conditions, the social environment, etc. Naturally molted feathers should be picked up and recorded as they are discovered so that the normal molt pattern of kori bustards can be described at different institutions.

<u>Section 7.6. Management of Diseases, Disorders, Injuries and/or Isolation</u>

Hemosiderosis has been reported in some kori bustards managed in zoos, but the causal factors associated with this disorder are not yet known. More research is needed to test hypotheses that link this disorder with a possible dietary etiology, or a genetic predisposition.

Additionally, the following viral diseases have been associated with bustard species, but the significance of these viruses for the care and management of bustards in zoos remains unknown. More research is needed on the prevalence and treatment of these diseases within the zoo population.

- Adenovirus
- Avian influenza
- Avipox
- Herpesvirus
- Infectious bursal disease
- Lymphoid leucosis
- Marek’s disease
- Newcastle disease
- Pigeon herpes
- PMV-2
- Reovirus

Chapter 8: Reproduction

<u>Section 8.3. Egg-laying/Incubating/Hatching</u>

Kori bustard females often pace excessively 2–3 days prior to egg laying, and typically do so around the area where egg laying will occur. However, not all females pace prior to egg laying, and more research is needed to identify other behaviors that may reliably indicate imminent egg laying.

<u>Section 8.2. Assisted Reproductive Technology</u>

Artificial insemination has not been performed with bustards in AZA-accredited institutions, although it remains a feasible approach to take in future reproductive efforts. Houbara bustard semen collection techniques have been successful in the United Arab Emirates, and might provide a useful foundation for future research if the technique is applied to kori bustards, but more research is needed on bustards to develop suitable semen collection, storage, and insemination practices that are specific to kori bustards and buff-crested bustards. General information on semen collection, artificial insemination, and cryopreservation of semen in non-domestic birds is provided by Gee (2004).

<u>Section 8.5. Assisted Rearing</u>

Cross-fostering and shared-rearing techniques have not been used with kori bustards in zoos in the United States, but further investigation into these approaches might be useful to determine if they are applicable to this species. Research should focus on the timing of cross-fostering attempts in kori bustards and related species, when this approach has been attempted, and the influence that imprinting has on the success of shared-rearing techniques.

Angel wing in chicks can begin to occur between 7–11 days post-hatch. Although parent-reared chicks seem to have higher growth rates than hand-reared chicks during the first week of life, parent-reared chicks tend not to develop angel wing. More research is needed to determine why parent-reared chicks do not develop angel wing but hand-reared chicks do. Institutions should carefully monitor the growth rate of hand- and parent-reared chicks, and should maintain detailed records on the nutrient composition of the diets provided to hand-reared chicks. More research is also needed on the ways in which both diet and exercise affect growth and development of kori bustard chicks.

Preliminary research has shown that the mortality of female chicks within the first year has been found to be higher than mortality rates for males, and further research is needed to determine if this is a phenomenon seen in wild populations of kori bustards, or if it represents sub-optimal husbandry and management within zoo environments.

Chapter 9: Behavior

Section 9.2. Environmental Enrichment

Most zoos employ some form of enrichment with their kori bustards. Research is needed to determine the efficacy of the enrichment as well as the required frequency.

Section 9.1. Animal Training

Additional behavioral research that focuses on the behavior of wild kori bustards, and that can be used to make general comparisons with the behavior of kori bustards in zoos (e.g., based on daily activity budgets) in different social and physical conditions, will always be beneficial for improving appropriate animal management recommendations.

In 2007, the Kori Bustard Species Survival Plan® (SSP) Program sponsored a behavioral study on *ex situ* kori bustards housed at nine facilities in the United States. Over 75,000 behavioral observations were collected on 50 birds over five years using the Colonel Stanley R. McNeil Foundation's EthoTrak Observation System, a Palm®-based program. These data were used to investigate three areas of interest and to make management recommendations. The results of this study are published on the ASAG website (https://de0046ae-67da-48da-9d60-85f9fe01aee2.filesusr.com/ugd/df9b5b_422089b2c6114efdbc81d4579544019c.pdf).

Chapter 12. Other Considerations

12.1 Surplus Animals

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

All bustards housed at AZA institutions are considered part of the SSP. All bustards owned by AZA facilities are considered part of the SSP. If a situation arises where an SSP bustard is recommended for placement at a non-AZA facility the decision to deem the individual temporarily surplus to the SSP will be made by the SSP management group and communicated in writing via the Population Analysis & Breeding and Transfer Plan.

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

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Personal Communications:
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Paul Goriup, Managing Director, Fieldfare International Ecological Development, 2007
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Appendix A: Accreditation Standards by Chapter

The following specific standards of care relevant to bustards are taken from the AZA Accreditation Standards and Related Policies (AZA, 2017) and are referenced fully within the chapters of this animal care manual:

General Information

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

Chapter 1

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

(10.2.1) Critical life-support systems for the animals, including but not limited to plumbing, heating, cooling, aeration, and filtration, must be equipped with a warning mechanism, and emergency backup systems must be available. Warning mechanisms and emergency backup systems must be tested periodically.

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

Chapter 2

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

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

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

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

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

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

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

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

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

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

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

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

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

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

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

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

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

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

Chapter 3

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

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

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

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

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

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

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

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

Chapter 4

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

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

Chapter 6

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

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

Chapter 7

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

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

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

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

(2.7.1) The institution must have holding facilities or procedures for the quarantine of newly arrived animals and isolation facilities or procedures for the treatment of sick/injured animals. Quarantine duration should be assessed and determined by the pathogen risk and best practice for animal welfare.

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

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

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

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

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

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

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

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

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

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

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

(2.1.3) Paid and unpaid animal care staff should be trained to assess welfare and recognize abnormal behavior and clinical signs of illness and have knowledge of the diets, husbandry (including enrichment items and strategies), and restraint procedures required for the animals under their care. However, animal care staff (paid and unpaid) must not diagnose illnesses nor prescribe treatment.

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

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

Chapter 9

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

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

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

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

Chapter 10

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

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

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

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

(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.

Chapter 11

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

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

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

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

Appendix B: Recordkeeping Guidelines for Group Accessions

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

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

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

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

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

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

Examples of Appropriate Group Accessions

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

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

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

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

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

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

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

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

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

Managing Group Records

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

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

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

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

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

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

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

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

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

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

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

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

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

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

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

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

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

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

Animal and Collection Records – Definitions and Examples

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

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

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

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

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

Animal and Collection Records - Development

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

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

Animal and Collection Records – Sharing of Information

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

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

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

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

    For example, the shipment of an animal might require that written notice be made to the senior keeper in the animal's area, the curator, and the veterinarian at least 30 days prior to the move, and identifies the animal by group or individual identification/accession number, sex, and tag/transponder number, etc.

- define where each record type (stored or archived) is available and what format (paper or digital) it is in.

    For example, all original animal transaction documents might be kept in the registrar's office in fire-proof file cabinets but copies of the Animal Data Transfer Forms are kept in the appropriate keeper area.

- define a system for obtaining necessary information such that the information is available regardless of department and regardless of staffing issues

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

Implementation of these Recommendations

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

Appendix D: AZA Policy on Responsible Population Management

PREAMBLE

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

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

I. INTRODUCTION

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

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

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

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

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

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

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

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

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

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

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

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

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

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

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

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

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

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

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

III. ACQUISITION REQUIREMENTS

A. General Acquisitions

1. Acquisitions must be consistent with the mission of the institution, as reflected in its Institutional Collection Plan, by addressing its exhibition/education, conservation, and/or scientific goals regarding the individual or species.

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

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

4. If the acquisition involves a species managed by an AZA Animal Program, the institution should communicate with the Animal Program Leader and, in the case of Green SSP Programs, must adhere to the AZA Full Participation Policy (https://www.aza.org/board-approved-policies-and-position-statements).

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

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

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

B. Acquisitions from the Wild

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

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

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

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

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

IV. TRANSFER, EUTHANASIA AND REINTRODUCTION REQUIREMENTS

A. Living Animals

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

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

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

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

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

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

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

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

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

institutions and certified related facilities may send animals to non-AZA organizations or individuals (refer to #IV.5 above). These non-AZA entities (for instance, ranching operations) should follow appropriate ranch management practices and other conservation minded practices to support population sustainability.

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

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

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

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

B. Non-Living Animals and Specimens

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

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

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

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

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

5. AZA member institutions should develop agreements for the transfer or donation of non-living animals, parts, materials, products, and specimens and associated documentation, to non-AZA

entities such as universities and museums. These agreements should be made with entities that have appropriate long term curation/collections capacity and research protocols, or needs for educational programs and/or exhibits.

DEFINITIONS

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

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

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

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

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

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

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

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

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

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

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

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

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

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

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

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

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

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

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

RECIPIENT PROFILE EXAMPLE

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

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

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

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

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

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

Veterinary Information:

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

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

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

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

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

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

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

7. Copy of your institutional acquisition/disposition policy.

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

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

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

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

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

Appendix E: Recommended Quarantine Procedures

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

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

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

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

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

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

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

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

Quarantine procedures: The following are recommendations and suggestions for appropriate quarantine procedures for bustards:

Quarantine procedures**:** The following are recommendations and suggestions for appropriate quarantine procedures for birds that are applicable to kori bustards:

Required:

1. Direct and floatation fecals
2. Evaluate for ectoparasites
3. Appropriate serological tests for psittacosis, and if positive, confirmed by culture vaccinate as appropriate

Strongly Recommended:

1. CBC/sera profile
2. Fecal culture for *Salmonella sp.*
3. Fecal gram stain

Appendix F: Ambassador Animal Policy and Position Statement

Ambassador (Program) Animal Policy

Originally approved by the AZA Board of Directors—2003
Updated and approved by the Board—July 2008 & June 2011
Modified from "Program Animal" to "Ambassador Animal" to avoid confusion with "Animal Programs," approved by the CEC; no change to meaning of these terms - January 2015

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

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

There are three main categories of Ambassador Animal interactions:

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

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

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

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

Ambassador Animal Position Statement

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

The Conservation Education Committee (CEC) of the Association of Zoos and Aquariums supports the appropriate use of Ambassador Animals as an important and powerful educational tool that provides a variety of benefits to zoo and aquarium educators seeking to convey cognitive and affective (emotional) messages about conservation, wildlife and animal welfare.

Utilizing these animals allows educators to strongly engage audiences. As discussed below, the use of Ambassador Animals has been demonstrated to result in lengthened learning periods, increased knowledge acquisition and retention, enhanced environmental attitudes, and the creation of positive perceptions concerning zoo and aquarium animals.

Audience Engagement

Zoos and aquariums are ideal venues for developing emotional ties to wildlife and fostering an appreciation for the natural world. However, developing and delivering effective educational messages in the free-choice learning environments of zoos and aquariums is a difficult task.

Zoo and aquarium educators are constantly challenged to develop methods for engaging and teaching visitors who often view a trip to the zoo as a social or recreational experience (Morgan & Hodgkinson, 1999). The use of Ambassador Animals can provide the compelling experience necessary to attract and maintain personal connections with visitors of all motivations, thus preparing them for learning and reflection on their own relationships with nature.

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

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

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

Knowledge Acquisition

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

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

Enhanced Environmental Attitudes

Ambassador Animals have been clearly demonstrated to increase affective learning and attitudinal change.

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

Conclusion

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

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Appendix G: Kori Bustard Ethogram

Table _. Kori bustard ethogram (Lichtenberg & Hallager, 2007)

Behavior	**Description**
Resting behaviors	
Alert	Bird sits on the ground or stands with its eyes open.
Resting	Bird sits or stands with its eyes closed.
Hock sitting	Bird sits with its tarsi on the ground and tibias vertical, leaving the belly raised off the ground.
Wet weather standing	During periods of light to heavy rain, bird tucks its head tightly against the back of its neck, giving it a hunched appearance. During poor weather, bird may stand under a bush for protection from the elements.
Comfort/maintenance behaviors	
Scratching	Bird scratches body (i.e., neck and head areas) using a toe.
Stretching	Bird stretches its leg, wing, or body, and typically involves stretching the wing and leg on the same side of the body. During a body stretch, neck is extended forward, body is lowered slightly, and wings are raised.
Wing flapping	Bird extends its wings and flaps them several times, often in conjunction with stretching or jumping.
Body fluffing	Bird briefly erects feathers on the neck, wings, and back.
Ruffling	Bird shakes its body, with movement passing as a wave from head to tail, which is usually slightly fanned.
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 the birds' eyes are often closed.
Toe picking	Bird uses bill to pull at a toe or nail, while standing on one leg.
Dustbathing	Bird lies flat and rubs its belly, head, neck, and wings on the ground, in a sandy or dusty depression. Feathers are ruffled during bathing.
Sun bathing	Bird sits in the direct sun (often near a bush or short grass clump) with one or both of its wings spread horizontal to the ground. Sun bathing often leads to heavy panting, and is always followed by preening.
Bill gaping	Bird briefly opens mouth wide.
Bill open	Bird partially opens bill for several seconds.
Panting	Bill is slightly open and the gular skin is moved back and forth. Panting is a cooling mechanism.
Tail erection	Birds raise their tails vertically and fan their tail feathers out. Tail erection occurs commonly after rain has ended, and may help in drying.
Locomotion	
Walking	Bird moves about on the ground at a leisurely pace. Walking is the primary mode of locomotion for kori bustards.
Running	Bird moves faster (at running human speed), with head held high and extended, or low and horizontal to the ground. Wings may be extended or held close to body. Running is used for avoiding predators.

Behavior	Description
Flying	Birds take off into the wind following a short run and wing flapping. Flying is also used for predator avoidance.
Jumping	Bird will jump into the air (up to 7 feet) without a running start. Jumping may be a means of evading terrestrial predators.
<u>Feeding behavior</u>	
Drinking	Birds drink while standing or hock sitting by submerging the beak in water, using tongue or throat motions to suck in water, and then raising the head to around a 45° angle to gulp down the water.
Foraging	Birds search for food while walking and looking down at the ground. Birds can also jump for food that is out of their reach.
Feeding	Birds peck at food within their reach or snap at jumping or flying prey with their bill. Larger vertebrates are shaken or struck against the ground before being swallowed whole; small items are just swallowed.
Grit eating	Birds peck at and ingest grit or small pebbles to aid in digestion.
Bill wiping	Bird rubs the sides of its bill along the ground, a tree trunk, or other object, to remove any debris attached to the bill.
Defecation	Fecal matter is excreted mainly while walking, or during short pauses, and typically when the bird wakes in the morning. Smaller amounts are excreted throughout the day. Defecation is also part of a fear response.
<u>Social behavior</u>	
Aggressive displacement	One bird chases another bird, by lowering its head, raising its head crest, ruffling its plumage slightly, and aiming its body towards it. Pursuit will often continue until the other bird it is out of sight.
Non-aggressive displacement	One bird walks toward another bird, causing the second bird to vacate its position and move elsewhere. Females do not displace adult males.
Fighting (males)	Birds grasp each other' beaks and shove each other, with feathers fluffed out and tails raised. May establish dominance during breeding.
Aggressive head pecking	A dominant bird pecks at the head of a subordinate bird for a short period of time, usually with no injuries.
Tail lifting	Sitting or standing bird lifts tail up to a 90º angle, fans out tail feathers, and then lowers the tail. Behavior may be repeated multiple times, and may be accompanied by erection of the head crest feathers. Occurs when birds approach. Males do not tail-lift to approaching females.
Threat posture	A standing bird lifts its tail and fans its tail feathers; its wings are outstretched, its plumage ruffled, and its head extended forward. The wings and tail may be vibrated.
<u>Inter-specific threat response</u>	
Skyward looking	Alert bird extends its head and cocks it upward, and may also tilt the head sideways so only one eye is facing upwards.
Crest up	Bird erects head crest feathers in response to a potential threat.
Neck fluffing	Bird erects neck feathers and head crest to increase its apparent size.
Tail spreading	Bird raises tail vertically and fans tail feathers as a defense posture against aerial predators.
Predator defense display	Bird crouches with its tail raised and fanned, wings loosely tucked to the body, and

Behavior	Description
	with its head and neck extended upwards in response to a potential or perceived predator flying overhead.
<u>Sexual behavior</u>	
Chasing females (males)	Males chase females with raised head crest and tail. Pursuit will often cease if females run away out of view.
Tail up position (males)	Males stands or walks with tail raised and fanned, wings held close to the body, and head crest erect, to attract the attention of females.
Partial balloon display (males)	Males stand with neck partially inflated, tail raised up, and head crest erect.
Balloon display (males)	Male extends neck and fully inflates the esophageal pouch (up to 4 times normal size) with the bill pointed upward. The tail and wing feathers point downward and the head crest is erect. Bird emits a low-pitched booming sound as bill is snapped open and shut. The balloon display is the most intense form of courtship.
Copulation initiation (females)	Female initiates copulation by sitting down near a displaying male to allow him to approach her from behind.
Head pecking (males)	Male approaches sitting female, and pecks (for 5–10 minutes) at the back of her head, stepping from one side of the female to the other.
Squatting and pecking (males)	Male squats on his hocks while continuing to peck at the female's head. This element of copulation generally lasts 5 minutes.
Mounting (males)	Male spreads wings, climbs onto the back of the seated female and transfers sperm (taking only a few seconds).
Post-copulation feather shaking	Males and females stand up after copulations and vigorously shake their feathers before resuming normal activities.
<u>Maternal behavior</u>	
Pacing (females)	The female walks back and forth in a particular area, faster than when just walking, for a few minutes or for several hours prior to egg-laying.
Incubating (females)	Females sit on eggs after first or subsequent eggs are laid. Incubating females rarely leave the nest, but will do so for behaviors such as feeding, dustbathing, and sunbathing.
Egg turning (females)	Females stand over the nest and rotate their eggs with their bills several times throughout the day and night.
Nest building (females)	Sitting female throws leaf litter and small sticks onto her back while incubating.
Parental care behavior (females)	Females in zoos are highly protective of the chicks and will act aggressively towards other females and keepers. Female will pick up food and present it to the chicks, bending her head and neck down while holding the food in her bill, and salivating copiously. The female produces a quiet vocalization when feeding the chicks.
<u>Vocalizations</u>	
Barking	Nervous or startled birds produce a soft, gruff "bark", also called a "gronk" call. Barking birds typically stand, remain focused on the cause of concern, and may move away from this object.
Growling	A soft "rrrrrr" sound, similar to a cat's growl. Females may growl when defending eggs or chicks, or when handled by keepers.
Booming (males)	A low-pitched booming vocalization produced by males in the breeding season when the bill is snapped open and shut. Males emit 6 calls in rapid succession, and may

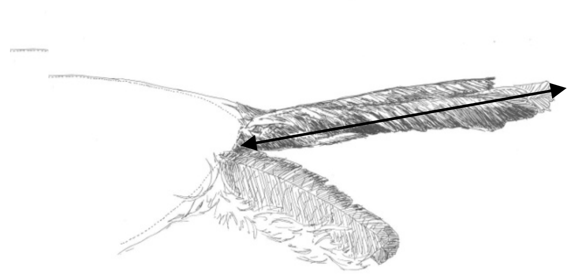
Behavior	Description
	repeat the 6-call cycles for several minutes.
Chick chirp	A light chirp or purring sound produced by chicks being fed or brooded.
Chick cry	Stressed, chicks produce a long, sad whistle that can escalate into a loud wailing.
Grunting (females)	A low- to high-pitched sound produced by females to call their chicks, especially when food is available.
Roaring	A very loud vocalization similar to a lion's roar, often occurring when birds are initially captured.

Appendix H: Measuring Adult Kori Bustards

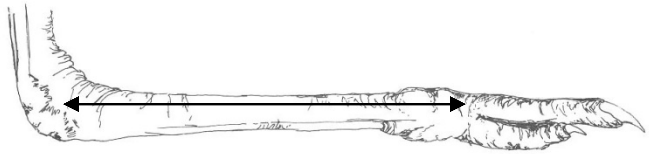
It is important to take body measurements on every kori bustard at some point during the animal's life. These measurements are of important use in taxonomy, species characterization, and eco-morphology (the study of the relationship between body form and ecology). In addition to the measurements taken below (drawings provided courtesy of Debi Talbott) collectors should also record the sex, age, collection date, sexual condition, weight, and their (the collector's) name. Please send these measurements to the Kori Bustard SSP® Coordinator.

Wing length: Measure from the "wrist" to the tip of the longest primary.

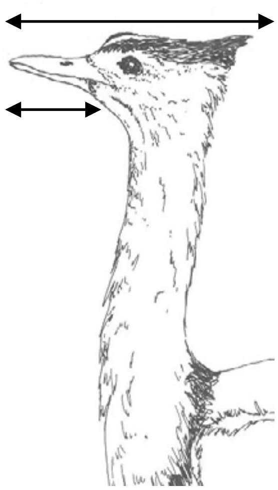
Tail length: Insert a ruler centrally between the longest tail feathers and the under-tail coverts until the ruler stops.



Tarsus: Measure on the front of the leg from the joint of the tibiotarsus with the tarsometatarsus to the lower end at the foot (last scute).

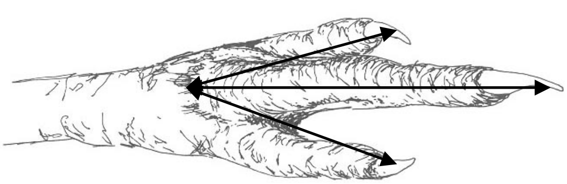


Skull: Maximum length from the rear of the skull to the tip of the bill. Also measure skull width (across the postorbital bones).



Culmen: Tip of bill to base of skull.

Toe, inner, middle and outer: Measure from the tip of the nail to the joint of the toe with the tarsometatarsus.



Quarantine procedures**:** The following are recommendations and suggestions for appropriate quarantine procedures for birds that are applicable to kori bustards:

Required:

4. Direct and floatation fecals
5. Evaluate for ectoparasites
6. Appropriate serological tests for psittacosis, and if positive, confirmed by culture vaccinate as appropriate

Strongly Recommended:

4. CBC/sera profile
5. Fecal culture for *Salmonella sp.*
6. Fecal gram stain

Appendix I: Hematological Reference Values

A few publications providing hematological reference values for mature and growing kori bustards and have been included here (Tables 28–40) for consultation and comparison with values taken during quarantine and when examining the current health status of birds as part of each institution's preventative veterinary health program.

Table 28. Hematological reference values for mature and growing kori bustards (Howlett et al.,1995)

Cell description (μm)	**Mean ± SEM* (minimum--maximum)**	***N***
Erythrocytes length x width	13.5±0.05 x 7.3±0.06 (13–15) x (6–8)	100
Erythrocytes nucleus length x width	6.4±0.05 x 2±0.01 (5.5–7.5) x (2–2.5)	100
Heterophils diameter	11.5±0.21 (10–15)	50
Eosinophils diameter	11.8±0.17 (9–15)	50
Basophils diameter	9.5±0.27 (7–12)	30
'Small' lymphocytes diameter	7.2±0.12 (6–9)	50
'Large' lymphocytes diameter	10.7±0.16 (9–13)	50
Monocytes diameter	14±0.2 (12–20)	50
'Small' thrombocytes length x width	5.6±0.14 x 5.6±0.14 (5–7) x (5–7)	25
'Large' thrombocytes length x width	8.8±0.19 x 8.1±0.16 (7–10) x (7–10)	25
'Mean' thrombocytes length x width	7.3±0.26 x 6.8±0.21 (5–11) x (5–10)	50
'Mean' thrombocyte nucleus length x width	5.1±0.13 x 4.7±0.10 (4–8) x (4–6)	50

* Mean ± standard error of mean

Table 29. Normal adult kori bustard haematological reference values (Howlett et al., 1995)

	Mean ± SEM* (minimum--maximum)	***N***
RBC $x10^{12}/l$	2.30±0.06 (1.74–2.95)	28
Hb g/dl	14.10±0.16 (11.9–15.9)	28
Hct l/l	0.47±0.05 (0.395–0.525)	28
MCV fl	208.5±5.1 (161.9–275.4)	28
MCH pg	62.4±1.6 (48.0–84.6)	28
MCHC g/dl	30.0±0.4 (29.7–34.9)	28
WBC $x10^{9}/l$	7.29±0.42 (3.05–12.85)	28
Heterophils $x10^{9}/l$	3.98±0.32 (0.95–9.25)	28
Lymphocytes $x10^{9}/l$	2.21±0.24 (0.41–5.45)	28
Monocytes $x10^{9}/l$	0.60±0.07 (0.0–1.57)	28
Eosinophils $x 10^{9}/l$	0.35±0.05	27

	(0.0–1.15)	
Basophils x 10^9/l	0.20±0.03 (0.0–0.80)	28
Thrombocytes x 10^9/l	5.5±0.7 (1.49–18.0)	25
Fibrinogen g/l	2.42±0.10 (1.42–4.5)	27

*Mean ± standard error of mean
Hb, haemoglobin; Hct, haematocrit; MCV, mean cell volume;MCH, mean cell haemoglobin; MCHC, mean cell haemoglobin concentration

Table 30. Kori bustard (*Ardeotis kori*): Haematological findings in birds of different ages (1-5 months) (Howlett et al., 1998)

	1 month	***n*1**	**2 months**	***n*2**	**3 months**	***n*3**	**4 months**	***n* 4**	**5 months**	***n*5**
RBC x 10^{12}/l	1.28±0.06* (1.04–1.61)**	9	1.57±0.06 (1.22–2.01)	12	1.76±0.08 (1.31–2.4)	14	2.06±0.08 (1.39–2.63)	15	2.07±0.08 (1.79–2.59)	11
HB g/dl	7.5±0.2 (6.8–8.3)	9	9.7±0.3 (7.4–10.9)	12	10.9±0.3 (9.6–13.1)	14	12.1±0.3 (10.3–14.0)	15	12.1±0.4 (9.1–14.0)	11
Hct l/l	0.23±0.7 (0.195v0.26)	9	0.30±0.9 (0.24–0.34)	12	35.1±0.7 (0.30–0.41)	14	0.374±0.7 (0.32–0.41)	15	0.396±0.9 (0.33–0.44)	11
MCV fl	178.4±7.9 (152.2–219.6)	9	195.2±6.6 (159.2–225.4)	12	204.3±8.1 (168.8–262.8)	14	185.2±7.9 (144.0–241.0)	15	194.2±7.8 (152.5–225.4)	11
MCH pg	59.3±3.4 (42.2–77.6)	9	62.0±1.5 (52.2–69.0)	12	63.5±2.4 (49.2–79.4)	14	59.6±2.3 (45.9–74.1)	15	59.5±3.1 (42.1–73.7)	11
MCH g/dl	33.2±1.0 (27.8–37.2)	9	32.0±0.9 (28.2–40.4)	12	31.2±0.8 (27.5–38.1)	14	32.3±0.5 (29.2–35.3)	15	30.5±0.7 (24.3–33)	11
THROMB x 10^9/l	7.03±1.79 (3.1–15.0)	6	8.1±0.8 (4.07–15.6)	12	6.7±0.4 (4.6–9.2)	14	7.9±0.7 (3.7–15.0)	15	7.35±0.61 (3.7–10.8)	11
WBC x 10^9/l	8.78±0.45 (6.65–10.85)	9	10.2±0.6 (6.1–14.75)	12	10.7±0.7 (7.05–16.2)	14	12.7±0.7 (8–18.8)	15	12.5±0.4 (10.35–15.15)	11
HET x 10^9/l	5.42±0.38 (3.72v6.94)	9	4.28±0.30 (2.62–6.34)	12	5.14±0.65 (2.58–11.8)	14	6.2±0.6 (2.9–10.2)	15	5.1±0.5 (1.5–7.5)	11
LYMPH x 10^9/l	2.63±0.26 1.37–3.70)	9	4.64±0.33 (2.50–6.97)	12	4.19±0.27 (3.08–6.35)	14	5.1±0.4 (2.9–7.8)	15	5.49±0.45 (3.60–9.05)	11
MONO x 10^9/l	0.24±0.06 (0.08–0.63)	9	0.66±0.15 (0.24–1.92)	12	0.80±0.12 (0.00–1.58)	14	0.81±0.08 (0.35–1.50)	15	1.13±0.16 (0.49–2.03)	11
EOS x 10^9/l	0.28±0.08 (0.00–0.65)	9	0.32±0.04 (0.1–0.5)	12	0.38±0.08 (0.07–1.03)	14	0.35±0.05 (0.00–0.78)	15	0.42±0.08 (0.00–1.06)	11
BAS x 10^9/l	0.21±0.06 (0.00–0.47)	9	0.12±0.04 (0.00–0.43)	12	0.04±0.01 (0.00–0.13)	14	0.11±0.04 (0.00–0.48)	15	0.05±0.01 (0.00–0.16)	11
FIB g/l	1.76±0.18 (1.1–2.6)	8	2.0±0.1 (1.2–2.8)	12	2.25±0.2 (1.6–3.8)	12	2.57±0.3 (1.6–4.0)	8	2.58±0.49 (1.6–4.8)	8

RBC, red blood cells; HB, haemoglobin; Hct, haematocrit; MCV, mean corpuscular volume; MCH, mean corpuscular haemoglobin; MCHC, mean corpuscular haemoglobin concentration; THROMB, thrombocytes; WBC, white blood cells; HET, heterophils; LYMPH, lymphocytes; MONO, monocytes; EOS, eosinophils; BAS, basophils; FIB, fibrinogen.
* Mean ± standard error of mean
** Minimum--maximum
n = no. of samples

Table 31. Kori bustard (*Ardeotis kori*): Haematological findings in birds of different ages (6–15 months) (Howlett et al., 1998)

	6 months	*n*6	7 months	*n*7	8 months	*n*8	9 months	*n*9	15 months	*n*15
RBC x 10^{12}/l	2.14±0.07* (1.84–2.46)**	10	2.12±0.07 (1.79–2.61)	12	2.0±0.1 (1.72–2.6)	10	2.1±0.04 (1.91–2.23)	8	2.08±0.06 (1.81–2.47)	13
HB g/dl	11.4±0.4 (9.9–13.2)	10	11.7±0.3 (10.5–13.1)	12	12.5±0.4 (10.9–14.3)	10	12.8±0.4 (10.9–14.2)	8	14.2±0.4 (12.1–16.1)	13
Hct l/l	0.38±0.9 (0.34–0.42)	10	0.396±0.9 (0.35–0.45)	12	0.39±1.0 (0.36–0.45)	10	0.39±0.9 (0.36–0.43)	8	0.47±0.9 (0.41–0.51)	13
MCV fl	179.9±5.0 (160.6–210.1)	10	188.2±5.6 (165.1–220.7)	12	200.5±7.4 (138.5–219.3)	10	186.5±3.6 (168.2–200.5)	8	226.5±7.3 (195.5–273.5)	13
MCH pg	53.7±2.1 (40.2–66.5)	10	55.8±2.1 (43.7–66.5)	12	64.4±2.5 (45.4–74.6)	10	60.5±1.3 (57.1–68.6)	8	68.7±2.4 (58.2–84.0)	13
MCH g/dl	29.8±0.7 (25.1–32.4)	10	29.6±0.6 (25.6–32.3)	12	32.2±0.6 (29.5–36.3)	10	32.5±0.7 (29.8–35.5)	8	30.3±0.5 (27.6–33.2)	13
THROMB x 109/l	9.98±1.09 (4.9–1.7)	10	11.61±1.0 (5.5–16.7)	12	10.6±0.9 (6.3–15)	10	7.0±1.0 (4.8–9.5)	4	6.52±0.54 (4.05–10.2)	13
WBC x 10^9/l	13.5±0.7 (9.8–16.1)	10	15.6±0.7 (9.2–18.5)	12	13.5±0.9 (9.25–17.05)	10	14.5±0.5 (12.8–6.9)	8	14.10±0.61 (11.25–17.8)	13
HET x 10^9/l	5.1±0.9 (1.9–11.4)	10	6.34±0.58 (2.76–8.83)	12	5.41±0.58 (2.59–8.35)	10	6.27±0.76 (4.08–10.8)	8	5.84±0.61 (2.70–10.63)	13
LYMPH x 10^9/l	6.7±0.6 (3.0–10.5)	10	7.32±0.59 (4.50–11.84)	12	6.38±0.52 (3.80–9.29)	10	6.49±0.59 (2.85–8.45)	8	6.49±0.43 (4.37–9.43)	13
MONO x 10^9/l	0.93±0.14 (0.14–1.64)	10	1.12±0.13 (0.45–1.78)	12	1.18±0.13 (0.63–1.70)	10	0.75±0.17 (0.13–1.41)	8	1.13±0.07 (0.80–1.53)	13
EOS x 10^9/l	0.36±0.07 (0.14–1.64)	10	0.48±0.08 (0.0–0.89)	12	0.21±0.06 (0.0–0.63)	10	0.53±0.09 (0.13–0.92)	8	0.35±0.05 (0.13–0.68)	13
BAS x 10^9/l	0.23±0.07 (0.0–0.57)	10	0.08±0.01 (0.0–0.14)	12	0.03±0.01 (0.0–0.08)	10	0.08±0.01 (0.02–0.14)	8	0.29±0.06 (0.0–0.69)	12
FIB g/l	2.83±0.22 (1.7–4.0)	10	3.0±0.2 (2.0–4.7)	11	2.7±0.3 (1.1–5.4)	10	2.65±0.27 (2.0–3.3)	4	3.0±0.3 (1.3–5.3)	13

RBC, red blood cells; HB, haemoglobin; Hct, haematocrit; MCV, mean corpuscular volume; MCH, mean corpuscular haemoglobin; MCHC, mean corpuscular haemoglobin concentration; THROMB, thrombocytes; WBC, white blood cells; HET, heterophils; LYMPH, lymphocytes; MONO, monocytes; EOS, eosinophils; BAS, basophils; FIB, fibrinogen.
*Mean ± SEM
**Minimum–maximum
n = no. of samples

Table 32. Haematology values for adult kori (*Ardeotis kori*) (n=28), houbara (*Chlamydotis macqueenii*) (n=34), Heuglin's (*Neotis heuglinii*) (n=5), black (*Afrotis afra*) (n=4), buff-crested (*Lophotis gindiana*) (n=14) and white-bellied bustards *(Eupodotis senegalensis*) (n=3) (Compiled by Jaime Samour and Judith Howlett)

	Species (source)					
Hematology values	**[1]Kori bustard**	**[2]Houbara bustard**	**[3]Heuglin's bustard**	**[3]Black bustard**	**[4]Buff-crested bustard**	**[3]White-bellied bustard**
Red blood cells (x 10^{12}/l)	2.30[a] ± 0.06[b] (1.74–2.95)[c]	2.53 ± 0.09 (1.95–3.15)	2.18 ± 0.05 (2.05–2.35)	2.68 ± 0.18 (2.12–2.9)	2.89 ± 0.19 (2.00–4.27)	2.31 ± 0.08 (2.16–2.47)
Haemoglobin (g/dl)	14.10 ± 0.16 (11.9–15.9)	14.72 ± 0.14 (13.7–15.7)	12.48 ± 0.37 (11.1–13.3)	14.62 ± 0.67 (13.6–16.6)	17.62 ± 0.52 (15.50–22.20)	15.23 ± 0.17 (14.9–15.5)
Haematocrit (l/l)	0.47 ± 0.05 (0.395–0.525)	0.47 ± 0.08 (0.42–0.51)	0.43 ± 0.005 (0.42– 0.45)	0.44 ± 0.01 (0.42–0.49)	0.47 ± 0.01 (0.40–0.50)	0.47 ± 0.01 (0.45–0.50)
Mean cell volume (fl)	208.5 ± 5.1 (161.9–275.4)	189.7 ± 8.85 (146.3–259.1)	198.14 ± 3.85 (185.1–206.3)	170.17 ± 14.04 (147.1–207.5)	172.85 ± 9.36 (105.39–220.00)	205.7 ± 8.3 (190.2–218.6)
Mean cell haemoglobin (pg)	62.4 ± 1.6 (48.0–84.6)	58.9 ± 2.44 (46.6–74.3)	57.3 ± 2.57 (49.3–64.8)	55.32 ± 4.26 (47.1–65.5)	65.21 ± 4.46 (40.71–97.50)	66.03 ± 3.06 (60.3–70.8)
Mean cell haemoglobin concentration (g/dl)	30.0 ± 0.4 (29.7–34.9)	31.16 ± 0.54 (26.1–34.1)	28.88 ± 1.15 (24.6–31.6)	32.52 ± 0.46 (31.5– 33.5)	37.63 ± 1.18 (31.31–47.56)	32.1 ± 1.0 (30.6–34)
Thrombocytes (x 10^9/l)	5.5 ± 0.7 (1.49–18.0)	6.82 ± 0.59 (2.76–9.88)	5.11 ± 0.45 (3.85–6.57)	10.48 ± 2.99 (4.68–18.49)	8.81 ± 1.04 (4.00–15.00)	5.99 ± 2.9 (3.06–11.8)
Fibrinogen (g/l)	2.42 ± 0.10 (1.42–4.5)	1.87 ± 0.26 (0.8–4.8)	1.71 ± 0.16 (1.12–2.11)	1.38 ± 0.19 (0.8–1.63)	1.7 ± 0.23 (0.66–4.31)	2.0 ± 0.15 (1.7–2.19)
White blood cells (x 10^9/l)	7.29 ± 0.42 (3.05–12.85)	5.81 ± 0.29 (4.25–7.6)	4.24 ± 0.3 (3.41–5.2)	7.85 ± 2.22 (3.81–13.8)	5.66 ± 0.38 (4.00–9.80)	6.26 ± 0.7 (5.2–7.6)
Heterophils (x 10^9/l)	3.98 ± 0.32 (0.95–9.25)	3.64 ± 0.24 (1.99–4.82)	1.55 ± 0.17 (1.34–2.25)	2.92 ± 0.53 (1.52–4.21)	3.32 ± 0.32 (1.44–5.88)	2.73 ± 0.75 (1.92–4.25)
Lymphocytes (x 10^9/l)	2.21 ± 0.24 (0.41–5.45)	1.84 ± 0.15 (0.97–3.24)	1.91 ± 0.22 (1.33–2.44)	3.66 ± 1.56 (1.48–8.14)	1.11 ± 0.20 (0.31–3.03)	2.51 ± 0.17 (2.18–2.73)
Monocytes (x 10^9/l)	0.60 ± 0.07 (0.0–1.57)	0.15 ± 0.03 (0.0–0.42)	0.41 ± 0.1 (0.07–0.72)	0.68 ± 0.23 (0.42–1.38)	0.42 ± 0.10 (0.04–1.30)	0.45 ± 0.14 (0.22–0.72)
Eosinophils (x 10^9/l)	0.35 ± 0.05 (0.0–1.15)	0.07 ± 0.01 (0.0–0.23)	0.12 ± 0.02 (0.04–0.208)	0.17 ± 0.08 (0.07–0.41)	0.24 ± 0.04 (0.00–0.62)	0.24 ± 0.09 (0.06–0.36)
Basophils (x 10^9/l)	0.20 ± 0.03 (0.0–0.80)	0.07 ± 0.02 (0.0–0.26)	0.13 ± 0.089 (0.0–0.46)	0.4 ± 0.09 (0.26–0.69)	0.44 ± 0.08 (0.10–1.23)	0.31.± 0.13 (0.07–0.54)

Mean [a] ± Standard Error of Mean [b] (Minimum - Maximum) [c] [1]Howlett et al. 1995, [2]Samour et al. 1994, [3]D'Aloia et al. 1996d, [4]D'Aloia et al. 1995

Table 33. Haematology values in buff-crested bustard (*Lophotis gindiana*) (n=5-8) of different ages. Bailey 2008.

Haematology values	**< 4 wk.**	**4–8 wk.**	**8–12 wk.**	**12–16 wk.**	**16–20 wk.**	**20–24 wk.**
Red blood cells ($x10^{12}$/l)	1.76[a] ±0.08[b]	1.87±0.09	1.97±0.09	2.37±0.10	2.59±0.05	2.86±0.18
	(1.51–1.89)[c]	(1.38–2.10)	(1.61–2.41)	(1.93–2.67)	(2.32–2.74)	(2.82–3.45)
Haemoglobin (g/dl)	7.72±0.45	9.55±0.17	10.64±0.36	13.15±0.31	14.34±0.29	14.24±0.61
	(6.80–9.30)	(7.90–11.30)	(8.90–12.00)	(11.90–14.50)	(13.60–16.20)	(12.40–16.20)
Haematocrit (l/l)	0.30±2.18	0.35±1.57	0.38±1.26	0.43±0.99	0.48±0.73	0.50±0.55
	(0.26–0.37)	(0.28–0.42)	(0.37–0.43)	(0.40–0.47)	(0.46–0.51)	(0.49–0.51)
Mean cell volume (fl)	172.35±7.50	185.30±5.25	193.42±6.18	185.18±8.68	185.14±4.24	176.04±10.54
	(150.54–195.77)	(162.30–205.00)	(164.14–217.88)	(150.94–229.06)	(169.71–202.43)	(147.83–211.30)
Mean cell haemoglobin (pg)	43.93±1.77	51.47±20.65	54.81±1.87	55.47±2.10	55.59±1.46	50.65±3.75
	(37.04–49.21)	(42.70–63.35)	(43.22–65.92)	(48.99–66.99)	(49.64–62.07)	(41.74–57.74)
Mean cell haemoglobin concentration (g/dl)	25.76±1.75	27.70±0.86	28.37±0.77	30.12±0.9	30.04±0.51	28.71±2.0
	(18.92–28.93)	(24.31–30.91)	(21.45–33.65)	(26.23–34.35)	(25.20–32.09)	(25.57–32.40)
Thrombocytes ($x10^9$/l)	8.74±2.46	6.50±0.99	9.22±1.21	8.40±1.54	6.28±1.04	9.20±2.11
	(3.40–17.88)	(2.20–9.72)	(3.07–17.92)	(4.40–17.85)	(2.28–11.60)	(3.90–16.80)
Fibrinogen (g/l)	1.68±0.24	1.88±0.21	1.73±0.18	2.21±0.14	2.39±0.28	2.20±0.32
	(1.02–2.50)	(1.50–3.30)	(1.20–3.52)	(1.80–2.60)	(1.15–3.33)	(1.60–3.33)
White Blood Cells ($x10^9$/l)	10.55±1.51	9.15±0.94	10.27±0.69	8.81±1.23	9.44±0.56	10.71±1.10
	(5.20–14.00)	(5.20–12.95)	(5.70–14.00)	(3.10–14.75)	(7.50–11.70)	(7.50–14.00)
Heterophils ($x10^9$/l)	5.2±1.17	3.72±0.53	3.31±0.46	3.37±1.13	2.80±0.10	2.77±0.54
	(0.94–7.56)	(1.64–5.97)	(0.51–6.44)	(0.56–9.44)	(2.31–3.16)	(1.04–4.20)

Lymphocytes (x10⁹/l)	4.25±0.53	4.33±0.56	5.49±0.66	4.14±0.61	4.89±0.55	6.27±0.56
	(3.02–5.85)	(2.50–7.90)	(2.16–8.82)	(1.92–6.50)	(3.17–7.14)	(4.50–7.70)
Monocytes (x10⁹/l)	0.29±0.03	0.31±0.09	0.57±0.10	0.53±0.10	0.63±0.13	1.00±0.26
	(0.21–0.39)	(0.00–0.75)	(0.11–1.47)	(0.19–0.95)	(0.20–1.18)	(0.30–1.80)
Eosinophils (x10⁹/l)	0.55±0.15	0.47±0.05	0.66±0.17	0.29±0.10	0.83±0.33	0.54±15
	(0.13–0.98)	(0.15–0.62)	(0.11–1.41)	(0.10–0.92)	(0.08–2.29)	(0.47–0.84)
Basophils (x10⁹/l)	0.27±0.11	0.34±0.07	0.20±0.07	0.48±0.16	0.34±0.08	0.13±0.07
	(0.00–0.63)	(0.00–0.63)	(0.00–0.64)	(0.00–1.48)	(0.11–0.70)	(0.0–0.36)

Mean [a] ± Standard Error of Mean [b] (Minimum - Maximum) [c]

Table 34. Haematology values in white-bellied bustard (*Eupodotis senegalensis*) (n=5) of different ages. Bailey 2008.

Haematology values	< 4 wk	4-8 wk	8-12 wk	12-16 wk	16-20 wk	20-24 wk
Red blood cells ($x10^{12}/l$)	1.45[a]±0.13[b]	1.76±0.12	1.95±0.04	2.04±0.10	2.31±0.13	2.58±0.13
	(1.16–1.68)[c]	(1.47–2.04)	(1.88–2.05)	(1.76–2.30)	(2.05–2.72)	(2.15–2.98)
Haemoglobin (g/dl)	8.75±0.96	10.53±0.8	11.31±0.6	11.84±0.27	13.98±0.86	14.24±0.35
	(7.00–10.70)	(9.30–12.90)	(9.40–12.30)	(11.20–12.60)	(11.90–16.30)	(13.20–14.90)
Haematocrit (l/l)	0.26±2.98	0.31±1.7	0.35±2.25	0.39±0.46	0.42±0.86	0.44±0.93
	(0.20–0.32)	(0.27–0.35)	(0.29–0.40)	(0.38–0.40)	(0.40–0.44)	(0.41–0.46)
Mean cell volume (fl)	175.21±6.21	177.73±4.1	182.52±13.47	191.95±9.75	183.62±9.88	171.23±6.69
	(159.09–187.50)	(169.12–187.88)	(146.46–210.11)	(171.74–215.91)	(159.93–213.59)	(147.65–188.37)
Mean cell haemoglobin (pg)	60.05±1.83	60.03±2.3	58.23±3.85	58.94±3.74	60.91±3.68	55.57±2.51
	(56.13–64.67)	(55.91–64.63)	(47.47–65.43)	(50.87–71.59)	(55.23–75.59)	(49.3–63.26)
Mean cell haemoglobin concentration (g/dl)	34.33±0.98	33.86±1.7	31.96±0.35	30.69±1.03	33.32±1.43	32.45±0.66
	(31.43–35.67)	(30.00–37.39)	(31.14–32.52)	(28.00–33.16)	(29.38–37.47)	(30.00–33.58)
Thrombocytes ($x10^9/l$)	9.45±2.70	8.22±2.4	7.20±2.07	7.60±1.08	6.34±1.18	9.14±1.12

	(4.80–15.00)	(4.60–15.20)	(3.64–12.10)	(5.10–10.50)	(3.70–8.80)	(7.0–11.9)
Fibrinogen (g/l)	1.13±0.09	2.02±0.3	2.19±0.33	2.44±0.58	1.88±0.40	2.20±0.70
	(1.00–1.30)	(1.37–2.48)	(1.40–2.72)	(1.60–4.80)	(1.00–2.90)	(1.06–4.00)
White blood cells ($x10^9/l$)	8.10±0.86	9.01±2.0	9.53±2.5	11.10±0.99	10.88±0.84	12.26±1.15
	(6.15–10.10)	(5.60–14.78)	(5.20–16.00)	(8.75–14.13)	(9.35–14.15)	(9.10–14.75)
Heterophils ($x10^9/l$)	3.80±0.21	4.77±1.1	3.83±1.51	4.61±1.06	5.43±0.91	4.91±0.81
	(3.30–4.20)	(2.69–7.97)	(2.20–8.36)	(1.39–7.21)	(3.77–8.77)	(1.91–6.81)
Lymphocytes ($x10^9/l$)	3.61±0.54	3.39±0.8	4.17±0.98	5.08±0.66	4.23±0.37	5.66±0.62
	(2.58–5.15)	(1.85–5.61)	(2.20–6.36)	(2.74– 6.50)	(2.81–4.85)	(4.20–7.32)
Monocytes ($x10^9/l$)	0.15±0.06	0.25±0.09	0.79±0.38	0.68±0.33	0.69±0.13	0.97±0.26
	(0.00–0.30)	(0.06–0.44)	(0.10–1.73)	(0.09–1.71)	(0.40–1.17)	(0.39–1.77)
Eosinophils ($x10^9/l$)	0.27±0.08	0.36±0.06	0.40±0.15	0.40±0.18	0.29±0.17	0.13±0.06
	(0.06–0.44)	(0.27–0.53)	(0.00–0.71)	(0.12–1.07)	(0.00–0.92)	(0.00–0.30)
Basophils ($x10^9/l$)	0.27±0.15	0.24±0.07	0.33±0.12	0.32±0.10	0.25±0.09	0.56±0.20
	(0.00–0.62)	(0.09–0.44)	(0.14–0.64)	(0.09–0.64)	(0.00–0.52)	(0.00–1.05)

Mean [a] ± Standard Error of Mean [b] (Minimum - Maximum) [c]

Table 35. Haematology values in kori bustards (*Ardeotis kori*) of different ages. Bailey 2008.

Parameter	6 months	7 months	8 months	9 months	15 months
Red blood cells ($x10^{12}/l$)	2.14 [a] ±0.07 [b]	2.12±0.07	2.0±0.1	2.1±0.04	2.08±0.06
	[10; 1.84–2.46] [c]	[12; 1.79–2.61]	[10; 1.72–2.6]	[8; 1.91–2.23]	[13; 1.81–2.47]
Haemoglobin (g/dl)	11.4±0.4	11.7±0.3	12.5±0.4	12.8±0.4	14.2±0.4
	[10; 9.9–13.2]	[12; 10.5–13.1]	[10; 10.9–14.3]	[8; 10.9–14.2]	[13; 12.1–16.1]
Haematocrit (l/l)	0.38±0.9	0.396±0.9	0.39±1.0	0.39±0.9	0.47±0.9
	[10; 0.34–0.42]	[12; 0.35–0.45]	[10; 0.36–0.45]	[8; 0.36–0.43]	[13; 0.41–0.51]

Mean cell volume (fl)	179.9±5.0 [10; 160.6–210.1]	188.2±5.6 [12; 165.1–220.7]	200.5±7.4 [10; 138.5–219.3]	186.5±3.6 [8; 168.2–200.5]	226.5±7.3 [13; 195.5–273.5]
Mean cell haemoglobin (pg)	53.7±2.1 [10; 40.2–66.5]	55.8±2.1 [12; 43.7–66.5]	64.4±2.5 [10; 45.4–74.6]	60.5±1.3 [8; 57.1–68.6]	68.7±2.4 [13; 58.2–84.0]
Mean cell haemoglobin conc (g/dl)	29.8±0.7 [10; 25.1–32.4]	29.6±0.6 [12; 25.6–32.3]	32.2±0.6 [10; 29.5–36.3]	32.5±0.7 [8; 29.8–35.5]	30.3±0.5 [13; 27.6–33.2]
Thrombocytes (x 10^9/l)	9.98±1.09 [10; 4.9–17]	11.61±1.0 [12; 5.5–16.7]	10.6±0.9 [10; 6.3–15]	7.0±1.0 [4; 4.8–9.5]	6.52±0.54 [4.05–10.2]
White Blood Cells (x10^9/l)	13.5±0.7 [10; 9.8–16.1]	15.6±0.7 [12; 9.2–18.5]	13.5±0.9 [10; 9.25–17.05]	14.5±0.5 [8; 12.8–16.9]	14.10±0.61 [13; 11.25–17.8]
Heterophils (x10^9/l)	5.1±0.9 [10; 1.9–11.4]	6.34±0.58 [12; 2.76–8.83]	5.41±0.58 [10; 2.59–8.35]	6.27±0.76 [8; 4.08–10.8]	5.84±0.61 [13; 2.70–10.63]
Lymphocytes (x10^9/l)	6.7±0.6 [10; 3.0–10.5]	7.32±0.59 [12; 4.50–11.84]	6.38±0.52 [10; 3.80–9.29]	6.49±0.59 [8; 2.85–8.45]	6.49±0.43 [13; 4.37–9.43]
Monocytes (x10^9/l)	0.93±0.14 [10; 0.14–1.64]	1.12±0.13 [12; 0.45–1.78]	1.18±0.13 [10; 0.63–1.70]	0.75±0.17 [8; 0.13–1.41]	1.13±0.07 [13; 0.80–1.53]
Eosinophils (x10^9/l)	0.36±0.07 [10; 0.14–0.76]	0.48±0.08 [12; 0.0–0.89]	0.21±0.06 [10; 0.0–0.63]	0.53±0.09 [8; 0.13–0.92]	0.35±0.05 [13; 0.13–0.68]
Basophils (x10^9/l)	0.23±0.07 [10; 0.0–0.57]	0.08±0.01 [12; 0.0–0.14]	0.03±0.01 [10; 0.0–0.08]	0.08±0.01 [8; 0.02–0.14]	0.29±0.06 [12; 0.0–0.69]
Fibrinogen (g/l)	2.83±0.22 [10; 1.7–4.0]	3.0±0.2 [11; 2.0–4.7]	2.7±0.3 [10; 1.1–5.4]	2.65±0.27 [4; 2.0–3.3]	3.0±0.3 [13; 1.3–5.3]

Mean [a] ± Standard Error of Mean [b] [Number of samples; Minimum – Maximum][c]

Table 36. Biochemical values for clinically normal adult and six age classes of juvenile kori bustards (*Ardeotis kori*). Bailey 2008.

	Age						
Assay	**4–8 wks (n=2-4)**	**9–16 wks (n=6–8)**	**17–24 wks (n=7–14)**	**25–32 wks (n=6–13)**	**33–40 wks (n=8–10)**	**41–52 wks (n=6–7)**	**>1 yr (adult) (n=28)**
Glucose (mmol/l)	20.87 ± 0.33[b]	17.61 ± 0.57	17.74 ± 0.45	18.14 ± 0.49	18.26 ± 0.72	16.43 ± 0.52	14.16 ± 0.33
	(20.26–21.21)[c]	(15.32–19.43)	(15.43–19.87)	(16.38–20.26)	(14.65–22.37)	(15.38–18.87)	(11.1–17.65)
Uric acid (μmol/l)	233.76 ± 46.57	422.31 ± 63.12	548.11 ± 46.04	433.07 ± 33.49	480.0 ± 36.69	518.43 ± 67.27	534.25 ± 38.36
	(148.7–309.30)	(178.44–797.03)	(333.08–808.93)	(273.61–594.8)	(297.4–612.64)	(321.19–785.14)	(237.92–969.52)
Total protein (g/l)	17.25 ± 1.25	24.63 ± 1.22	35.86 ± 1.98	35.84 ± 1.22	37.0 ± 1.64	32.83 ± 1.25	30.0 ± 0.82
	(14.0–20.0)	(19.0–28.0)	(24.0–51.0)	(29.0–43.0)	(28.0–43.0)	(29.0–38.0)	(23.0–40.0)
Albumin (g/l)	ND	ND	ND	ND	ND	ND	11.0 ± 0.3
							(8.0–15.0)
Globulin (g/l)	ND	ND	ND	ND	ND	ND	1.9 ± 0.06
							(1.5–3.1)
Albumin/globulin ratio	ND	ND	ND	ND	ND	ND	0.58 ± 0.01
							(0.29–0.73)
ALKP (U/l)	272.5 ± 34.5	182.75 ± 17.24	147.5 ± 16.35	130.75 ± 3.51	95.25 ± 5.92	56.67 ± 4.35	65.9 ± 2.6
	(238–307)	(117–239)	(55–219)	(119–144)	(75–119)	(47–78)	(37–98)
ALT (U/l)	27	27.29 ± 1.99	27.71 ± 2.69	28 ± 2.27	33.7 ± 1.54	25.33 ± 1.31	34.4 ± 3.43

	n=1	(20–35)	(20–39)	(20–34)	(24–39)	(23–31)	(20–120)
AST (U/l)	221.5 ± 8.5	273 ± 21.94	323.57 ± 16.39	294.67 ± 16.88	282.75 ± 13.94	228.67 ± 12.08	207 ± 7.1
	(213–230)	(208–408)	(285–409)	(231–400)	(224–355)	(191–271)	(168–369)
LDH (U/l)	1114 ± 17	1080.5 ± 48.29	1315.14 ± 167.47	996.88 ± 35.80	994.88 ± 37.1	887.83 ± 42.91	818.2 ± 50.8
	(1124–1158)	(874–1172)	(1008–2307)	(840–1117)	(848–1163)	(771–1050)	(543–1921)
CK (U/l)	ND	ND	ND	ND	ND	ND	275 ± 87.07
							(35–2527)
Magnesium (mmol/l)	ND	ND	ND	ND	ND	ND	1.05 ± 0.02
							(0.85–1.27)
Calcium (mmol/l)	0.83 ± 0.1	1.41 ± 0.12	1.42 ± 0.07	1.67 ± 0.08	1.98 ± 0.1	1.94 ± 0.1	2.34 ± 0.07
	(0.7–1.03)	(1.0–1.8)	(1.1–1.83)	(1.25–2.2)	(1.5–2.4)	(1.5–2.28)	(1.71–3.44)
Cholesterol (mmol/l)	ND	ND	ND	ND	ND	ND	3.7 ± 0.13
							(2.4–5.15)

[b]mean ± standard error of the mean, [c]minimum – maximum, ND - not determined.

Table 37. Blood chemistry findings for clinically normal adult and three age classes of juvenile buff-crested bustards (*Lophotis gindiana*). Bailey 2008.

	Age			
Assay	**2–8 wks.**	**9–16 wks.**	**17–24 wks.**	**>1 yr. (adult)**
Glucose (mmol/l)	14.93 ± 0.45[a] (14.11–15.67)[b] 3[c]	18.17 ± 0.81 (15.83–22.17) 8	16.32 ± 0.68 (13.94–17.78) 5	20.36 ± 0.55 (15.28–28.83) 25
Uric acid (µmol/l)	392.86 ± 47.14 (172.61–589.28) 7	337.5 ± 39.58 (113.1–726.19) 14	573.21 ± 47.29 (238.1–779.76) 12	533.93 ± 38.97 (202.38–928.57) 26
Total protein (g/l)	18.14 ± 1.37 (11.0–22.0) 7	26.88 ± 1.76 (13.0–39.0) 17	30.67 ± 1.56 (23.0–42.0) 12	32.81 ± 0.83 (25.0–41.0) 26
Albumin (g/l)	ND	ND	ND	12.91 ± 0.37 (10.0–16.0) 23
Globulin (g/l)	ND	ND	ND	19.61 ± 0.64 (14.0–26.0) 23
Albumin: globulin ratio	ND	ND	ND	0.67 ± 0.02 (0.52–0.79) 23
ALKP (U/l)	975.33 ± 367.33 (511–2926) 3	1080.81 ± 143.73 (392–2132) 11	561.5 ± 143.94 (155–790) 4	475.16 ± 45.71 (160–868) 26
ALT (U/l)	26 ± 7 (19–40) 3	28.25 ± 6.93 (17–76) 8	23.2 ± 2.48 (18–32) 5	ND
AST (U/l)	248.2 ± 20.17 (184–310) 5	256.46 ± 20.23 (151–388) 13	269 ± 29.51 (154–469) 9	334.96 ± 11.91 (217–482) 26
LDH (U/l)	381.5 ± 183.5 (198–565) 2	447.57 ± 79.65 (220–831) 7	415 ± 130.1 (278–675) 3	373.96 ± 29.72 (152–788) 26
CK (U/l)	ND	ND	ND	361.65 ± 41.27 (115–797) 26
Magnesium (mmol/l)	ND	ND	ND	1.05 ± 0,03

				(0.83–1.36)
				26
Calcium (mmol/l)	1.49 ± 0.11	1.61 ± 0.09	2.05 ± 0.19	2.55 ± 0.06
	(1.23–1.73)	(1.1–1.95)	(1.23–2.83)	(2.02– 3.51)
	5	8	7	26
Cholesterol (mmol/l)	ND	ND	ND	3.64 ± 0.15
				(2.19– 5.37)
				26

[a]mean ± standard error of the mean, [b]minimum - maximum, [c]number of samples, ND - not determined.

Table 38. Plasma chemistry values for adult kori bustards (*Ardeotis kori*). Bailey 2008.

Species	**Kori bustard[1]**
Assay	
Glucose (mg/dl)	238.66[a] ± 8.43[b]
	(166.0–357.0) [c]
	24[d]
Uric acid (mg/dl)	7.89 ± 0.50
	(3.50–14.30)
	26
Creatinine (mmol/dl)	0.57 ± 0.04
	(0.20–1.10)
	24
Total bilirubin (mg/dl)	0.70 ± 0.03
	(0.30–1.30)
	25
Total protein (g/dl)	2.96 ± 0.16
	(2.0–5.20)
	25
Albumin (g/dl)	1.59 ± 0.08
	(1.20–3.10)
	25
Globulin (g/dl)	1.30 ± 0.08
	(0.80–2.40)
	22
Albumin/globulin ratio	1.20 ± 0.47
	(0.70–2.10)
	23
GGT (u/l)	13.25 ± 0.47
	(12.0–14.0)
	4

ALT (u/l)	16.17 ± 2.24 (4.0–52.0) 23
ALKP (u/l)	ND
AST (u/l)	226.50 ± 10.80 (200.0–251.0) 4
LDH (u/l)	3862.50 ± 307.0 (2637.0–4689.0) 6
CK (u/l)	135.60 ± 20.90 (47.0–510.0) 24
Ammonia (µmol/l)	465.90 ± 47.40 (172.0–932.0) 21
Carbon dioxide (mmol/l)	27.47 ± 4.41 (10.0–94) 19
Magnesium (mg/dl)	0.87 ± 0.07 (0.30–1.90) 24
Phosphorus (mg/dl)	4.15 ± 0.26 (2.60–7.40) 26
Calcium (mg/dl)	12.47 ± 0.81 (6.10–21.0) 24
Potassium (mmol/l)	2.94 ± 0.19 (1.80–6.10) 25
Sodium (mmol/l)	154.48 ± 1.42 (145.0–174.0) 25
Chloride (mmol/l)	115.34 ± 0.98 (109.0–127.0) 23
Cholesterol (mg/dl)	120.34 ± 6.90 (66.0–193.0)

	26
Triglycerides (mg/dl)	107.24 ± 8.32 (61.0–225.0) 25
VLDL (mg/dl)	21.18 ± 1.44 (12.0–37.0) 22

[a]mean ± [b]standard error of the mean, [c]minimum - maximum, [d]number of samples, ND - not determined. [1]D'Aloia et al. 1996b., [2]D'Aloia et al. 1996a

Table 39. Serum bile acid concentrations for three bustard species using the colormetric assay. Bailey 2008.

Bustard species	**Serum bile acid (µmol/l)**
Houbara (*Chlamydotis macqueenii*)	35.8[a]±2.8[b] [6.4–89.6] [c] 50[d]
Kori (*Ardeotis kori*)	51.1±5.0 [9.6–131.7] 33
Buff-crested (*Lophotis gindiana*)	18.4±2.1 [4.8–41.8] 25
White-bellied (*Eupodotis senegalensis*)	20.8±5.4 [3.3–69.2] 13

[a]mean ± [b]standard error of the mean, [c]minimum - maximum, [d]number of samples, ND - no data.

Table 40. Serum copper, magnesium and zinc concentrations in six species of captive bustards in the United Arab Emirates (Bailey et al., 2004)

Species	**n[a]**	**Copper (µg/dl)**	**Magnesium (mmol/l)**	**Zinc (µg/dl)**
Houbara bustard (*Chlamydotis macqueenii*)	56	86.05[b] ± 0.71 [c] (76.7–98.1) [d]	1.2 ± 0.04 (0.63–1.95)	161.55 ± 2.53 (111.1–215)
Kori bustard (*Ardeotis kori*)	45	82.91 ± 0.88 (67.8–101.6)	1.08 ± 0.05 (0.63–1.84)	159.38 ± 5.53 (104–293.4)
White-bellied bustard (*Eupodotis senegalensis*)	34	84.20 ± 0.59 (77.4–93.3)	1.12 ± 0.04 (0.83–1.72)	175.5 ± 4.33 (121.5–220.2)
Buff-crested bustard (*Lophotis gindiana*)	31	81.61 ± 0.82 (70.9–89.9)	1.01 ± 0.03 (0.71–1.27)	174 ± 3,37 (129.7–205)
Black bustard (*Afrotis afra*)	3	83.7 ± 0.81 (82.4–85.2)	1.03 ± 0.09 (0.93–1.21)	171.9 ± 8.84 (156.3–186.9)
Heuglin's bustard (*Neotis heuglinii*)	4	82.43 ± 4.14 (71.6–91)	1.12 ± 0.05 (0.97–1.22)	201.33 ± 4.92 (192–215.2)

[a]number of samples, [b]mean ± [c]SE, [d] range.

Appendix J: AZA Kori Bustard SSP Necropsy Form – Revised December 2008

For best results, all dead birds should be necropsied as soon as possible. Carcasses should be refrigerated (never frozen) until the time of necropsy. Post-mortem examinations should be conducted in a routine fashion and all findings should be recorded on the AZA SSP Kori Bustard Necropsy Form presented below (or on a comparable form).

The Tissue Check List should be consulted to ensure that all samples have been collected. For proper tissue preservation, the volume of 10% buffered formalin used must be at least 10 times the volume of the tissue samples. After fixation (at least 24 hours) excess formalin can be poured off to facilitate shipping. Enough formalin should remain to keep tissues moist. One set of fixed tissues should be sent to the institution's Primary Pathologist for evaluation. If possible a duplicate set of tissue samples, along with copies of the Necropsy Form and Primary Pathologist's final report. Splitting of samples and lesions may not be advantageous. Alternatively, a duplicate set of histology slides (if available) and the original paraffin blocks (if agreeable to the laboratory), should be sent to the AZA Kori Bustard SSP Veterinary Advisor for storage and reference.

In addition to basic necropsy and histology, there should be a thorough examination of the vascular system including the large arteries and small peripheral ones associated with the intestines and distant sites such as skin. Also, any heart abnormalities should be noted and sampled. Some changes in the ventriculus (gizzard) such as distension, impaction, and muscle lesions have been recorded, and so recording the contents and appearance would be helpful. If pallor is noted in muscle, heart, or ventriculus, additional samples of frozen liver should be retained for possible nutritional evaluation after histopathology is completed.

If any questions arise regarding this protocol, please contact the AZA Kori Bustard SSP Veterinary Advisor, <u>before</u> proceeding with the necropsy.

SSP Veterinary Advisor:
Dr. Suzan Murray
Smithsonian's National Zoo
3001 Connecticut Ave NW
Washington DC 20008
Phone 202-633-3192; Email: murrays@si.edu

Please send a copy of the final pathology report to the AZA Kori Bustard SSP® Coordinator:
Sara Hallager
Smithsonian's National Zoo
3001 Connecticut Ave NW
Washington DC 20008
Phone (202-633-3088); Email: hallagers@si.edu

INSTITUTION/OWNER ____

ADDRESS______________________________________ COUNTRY ___________

ID# ___________ ISIS# _________ STUDBOOK# _________ SEX ________

AGE ___Y ___M ___D (Actual or estimate?) CAPTIVE-BORN OR WILD-CAUGHT?

WEIGHT (IN GRAMS OR KILOGRAMS) _______________

DEATH DATE ____________ NECROPSY DATE _____________

DEATH-NECROPSY INTERVAL (HRS) _______

DEATH LOCATION _______________ NECROPSY LOCATION ________________

EUTHANIZED? y / n . If yes, what method?_______________________________

HISTORY (Include clinical signs, circumstances of death, clinical labwork, diet and housing)

GROSS EXAMINATION

If no abnormalities are noted mark as normal or not examined (NE). Please perform complete necropsy and sample collection regardless if cause of death is known or grossly obvious.

GENERAL EXAM (Amount of subcutaneous and celomic fat, muscle condition pectoral/legs, skin, plumage, body orifices)

MUSCULOSKELETAL SYSTEM (Bones, marrow, joints, muscle)

RESPIRATORY SYSTEM (Nasal passages, pharynx, larynx, trachea, bronchi, lungs, air sacs). Please note the extent of the gular pouch in both genders.

CARDIOVASCULAR SYSTEM (heart, pericardial sac, great vessels, valves)

DIGESTIVE SYSTEM (mouth, beak, tongue, esophagus, proventriculus gizzard/ventriculus, intestines, cloaca, liver and gallbladder, pancreas). Please note the thickness of the ventricular wall and contents of the ventriculus.

SPLEEN AND THYMUS

URINARY SYSTEM (kidneys, ureters)

REPRODUCTIVE SYSTEM (gonads, oviduct)

ENDOCRINE SYSTEM (thyroids, parathyroids, adrenals, pituitary)

NERVOUS SYSTEM (nerves, brain, meninges, spinal cord, eyes)

ADDITIONAL COMMENTS OR OBSERVATIONS:

Prossector: ______________________ Date: __________

SUMMARIZE GROSS FINDINGS OR DIAGNOSES:

LABORATORY STUDIES: Results of cytology, fluid analysis, urinalysis, serum chemistries, bacteriology, mycology, virology, parasitology, radiography, etc.

Tissue Check List
Where possible freeze 1–3 cm (0.4–1.2 in.) blocks of tissue from major organs (e.g., liver, kidney, spleen, intestine) in small plastic bags, preferably to be kept ultrafrozen at -70 °C (-94 °F); freezing at conventional temperatures is acceptable if there is no access to an ultrafreezer. An additional sample of organs with a lesion should be taken. Additional liver (and possibly other organs) should be frozen if nutritional or toxin analysis is anticipated.

Preserve the following tissues in 10% buffered formalin at a ratio of approximately 1 part tissue to 10 parts solution. Large organs should have multiple samples taken from different areas. Where possible, always try to include junctions of different mucosal types (e.g., proventricular-ventricular junction; ileocecal junction, etc.). Tissues should be no thicker than 0.5–1 cm (0.2–0.4 in.). If possible, take two sets of fixed tissue, one for the Primary pathologist, and the other for the SSP Advisor. Alternatively, duplicate slides and blocks could be prepared for the SSP Advisors by the pathology facility. Tissues required for diagnosis should be sent to the SSP Advisor, who should be contacted for further instructions.

NOTE: There is generally no need to fix and label each tissue separately.

- Skin (with feathers)	- Cecum	- Trachea
- Muscle (leg and pectoral)	- Cloaca (with bursa of fabricius)	- Lung (bilateral)
- Nerve (Sciatic)	- Liver	- Air Sac
- Tongue	- Heart (atria, R/L ventricle, septum)	- Gallbladder
- Esophagus	- Blood vessels (aorta, femoral, etc.)	- Pancreas
- Proventriculus	- Thyroid (bilateral)	- Spleen
- Kidney	- Gizzard (full thickness)	- Parathyroid
- Duodenum	- Adrenal	- Thymus
- Jejunum	- Testis	- Brain (whole)
- Ileum	- Ovary	- Pituitary
- Colon	- Oviduct	- Eye
- Spinal cord*	- Bone with metaphyseal marrow	

* If neurologic deficits are suspect

PRIMARY PATHOLOGIST:
Name: ________________________________
Lab: ________________________________
Address: ________________________________
____
Phone: ________________________________

Please attach a copy of the final pathology report and send with the duplicate set of fixed tissues to the AZA Kori Bustard SSP Veterinary Advisor.

Appendix K: Sample Bustard Egg Production Annual Update Report

Egg Sizes

Egg Number	Sire Studbook Number	Dam Studbook Number	Date Egg Laid	Length	Width	Lay Weight
Ex.	143	119	DD/MO/YR			
1						
2						
3						
4						
5						
6						
7						
8						
9						
10						

If you ARTIFICALLY incubate any eggs, please provide your incubator settings and Incubator type ________________________________

Temperature_______________(F) or (C)

Humidity (wet bulb)_____________

OR, Relative Humidity_______________%

Please make any comments on the back of this sheet. Please copy this sheet as needed.

General Egg Production

				Incubation					Fertility			Results						Rearing				Chick Sex		
Egg Number	**Sire Studbook Number**	**Dam Studbook Number**	**Date Egg Laid**	**Parental**	**Incubator**	**Fostered**	**Combination**	**Not Incubated**	**Fertile**	**Infertile**	**Unknown**	**Hatched**	**Died in Shell**	**Broken**	**Missing**	**Unknown**	**Result Date or Hatch Date**	**Parental**	**Hand**	**Combination**	**Chick ID #**	**Male**	**Female**	**Unknown**
Ex.	143	119	D		x				x			x					DD/MO/YR		x		213964		x	
1																								
2																								
3																								
4																								
5																								
6																								
7																								
8																								
9																								
10																								

If you ARTIFICALLY incubate any eggs, please provide your incubator settings / Incubator type______________________________

Temperature______________(F) or (C) Humidity (wet bulb)____________ OR Relative Humidity______________%

Please make any comments on the back of this sheet. Please copy this sheet as needed.

Appendix L: Kori Bustard Chick Hand-rearing Protocol

Day	Brooder/Temp.	# of feedings	Notes
0	• 36.1°C (97 °F) • Brooder: 69.85 cm x 33 cm x 35.5 cm (27.5 in. x 13 in. x 14 in.) deep, floor is carpeted and a feather duster is hung in a corner • Single chicks are given a mirror	11	• Chicks should be fed as soon as they demonstrate a feeding response. This can be as early as 8 hours post hatch. After this period, chicks should be fed every 60–90 minutes for the first 24 hours post hatch. Night feedings may be necessary if a chick has hatched after 1700hrs. • Each feeding on Day 0: o 1-2 cricket abdomens (remove heads and legs) o 1-2 small pieces watermelon o 1 green bean • Later feedings on Day 0: o Add pellets (no more than 5 pellets). • Water is not necessary for the first three days following hatch provided watermelon is fed and chick hydration is monitored. After three days, chicks will begin drinking from a bowl with encouragement. • Crickets must be maintained on a high calcium insect diet for 72 hours prior to being fed. • Healthy chicks will attempt to grab offered food. Birds that do not show interest in food may be dehydrated. Puffy legs are a good sign of hydration, if skin on the legs appears tight, hydration is poor and chicks must be given SQ fluids. • Chicks will often have poor aim when attempting to eat, but this improves on Day 3. • Keepers should brood chicks at every feeding, and as often as possible during the first 7 days. • Do not offer mealworms until Day 21 as some chicks have impacted on this food.
1-2	• Same as for 'Day 0'	6	• Chicks are fed at two-hour intervals from 0630h to 1730h. Refer to Tables 26, 27, and 28 for daily amounts of food to be. Chicks will demonstrate a preference of fruits, crickets, vegetables over pellets so pellets should be fed first followed by the remaining diet. • Healthy chicks normally lose 3–5 g (0.1- 0.2 oz.) on Day 1, but their weights stabilize on Day 2 and increase thereafter. • In addition to crickets, two waxworms per day should be offered. The number is limited to two per day due to high fat content. Mealworms are considered enrichment items and are not offered until day 21 to avoid impaction • Chicks can stand and walk on Day 2. • Night feedings are not necessary provided the chick is gaining weight.

Day	Brooder/Temp.	# of feedings	Notes
3	• 35 °C (95 °F) • Carpeted nursery area (1.8 m x 3.7 m (5.9 ft. x 12 ft.)) • Three heat bulbs suspended from the ceiling so that three separate sections of floor are kept at 35 ºC (95 °F). • Several feather dusters are suspended near the heat bulbs. • Single chicks are given a mirror. Mirrors can be used with multiple chicks, but this may cause some chicks to be agitated; the mirror should be removed if this is the case.	• 6	• Feed every 2 hours (0630h to 1830h). • Chicks must be encouraged to exercise in the pen following each feeding to avoid problems such as slipped tendon. • Watermelon should be discontinued. Keepers should offer additional fruits (see Table 28). • Chicks should be trained to drink from a dish. Encouraging the birds to peck at shallow water dishes by using floating greens can be successful. • Use a shallow water bowl and place a rock in the bowl to prevent chicks from falling in and becoming wet. Do not use marbles as chicks can easily ingest these. • Egg (including pulverized shell) introduced into diet (scrambled, microwaved and cut into small pieces).
4–5	• Same as 'Day 4'	6	• Feed every 2 hours (0630h to 1830h) • By Day 5, chicks are keen to pick food floating in water, so to encourage self-feeding some food is left in shallow water bowls during the day. • general note on amounts of offered diet: chicks will rarely (if ever) consume all the pellets. They will however, usually eat all of the offered non-pellet items. • The need for brooding diminishes around Day 6—chicks will object strongly when brooded. • Chicks may now be offered whole crickets (no need to remove head and legs). • A mix of greens is essential for proper vitamin levels.
6–12	• Same as 'Day 4'	6	• Feed every 2 hours (0630h to 1830h). • Hand-reared chicks may develop slipped wings (i.e., an outward turning of the manus) anywhere from Day 7–11. Slipped wing is easily and permanently corrected if the primaries of the affected wing(s) are taped to the body in a natural position for 7–10 days at the first sign of the problem.
13	• Same as 'Day 4'	5	• Feedings may be reduced to five per day. • Chicks are taken outside for the day if the temperature is above 24 °C (75 °F). Once chicks have been given access to outside yards, they should be carefully monitored for the ingestion of foreign material that could result in impaction. The chicks should be watched to ensure that they do not consume too many pebbles, or stones that are too large.

Day	Brooder/Temp.	# of feedings	Notes
			• Ensuring that chicks continue to defecate normally is important. • Once outside, chicks should be monitored frequently for internal parasites and treated as necessary.
14–30	• Same as 'Day 4'	4	• Move the heat lamps up as the chicks grow so that they do not burn the top of their head. Also, beware of chicks jumping- they can jump very high and if heat lamps are too low, they will hit the bulb and break it. • Mealworms may now be introduced into the diet as an enrichment food, but limit the total number fed each day to <10 to reduce the risk of impaction. Mealworms must be maintained on a high calcium insect diet 72h prior to feeding. • Feed chicks 4 times per day.
30–60	• When chicks are 30–40 days old, they are moved outside to a covered yard measuring 5 m x 15 m (16.4 ft. x 49.2 ft.). The young birds spend the day outside and are housed in a heated shed at night.	3	• If chicks are scale trained, weights may be continued. Otherwise, daily weights can be discontinued at around 30 days to minimize the risks associated with repeated handling. • Maintain daily diet components in same approximate percentages as Day 30. • Three feedings per day is sufficient for chicks 30 days and older. • Chicks will likely demonstrate decreased interest in fruit and vegetables by 60 days. Greens are relished up until 8–12 months. • At 60+ days, add pellet-carnivore diet "meatballs" (see Table 29). Although chicks have been successfully reared without mammalian whole prey and carnivore diet introduced into their diet, at 60 days fuzzies may be introduced for behavioral management (scale training, close up viewing, etc.) but amounts should be limited to four per day.

Table 41. Daily quantities of food items offered to one kori bustard chick, based on 35% BW intake

Days of age	Average BW (g)	Total food offered (g)	Pellet (g)	Insects ⇨ (g)	Crickets each	Waxworms each	Egg (g)	Greens (g)	Vegetables (g)	Fruits (g)
0	99.7									
1	93.4	32.7	6.5	3.3 g =	10	2	0.0	1.6	8.2	13.1
2	98.7	34.5	6.9	3.5 g =	11	2	0.0	1.7	8.6	13.8
3	102.0	35.7	8.9	3.6 g =	12	2	1.8	3.6	8.9	8.9
4	110.8	38.8	9.7	3.9 g =	13	2	1.9	3.9	9.7	9.7
5	123.0	43.1	10.8	4.3 g =	15	2	2.2	4.3	10.8	10.8
6	130.3	45.6	11.4	4.6 g =	16	2	2.3	4.6	11.4	11.4
7	145.8	51.0	12.8	5.1 g =	18	2	2.6	5.1	12.8	12.8
8	157.3	55.0	13.8	5.5 g =	20	2	2.8	5.5	13.8	13.8
9	175.0	61.3	15.3	6.1 g =	22	2	3.1	6.1	15.3	15.3
10	192.5	67.4	16.8	6.7 g =	24	2	3.4	6.7	16.8	16.8
11	217.8	76.2	25.2	5.3 g =	18	2	3.8	11.4	19.1	11.4
12	240.0	84.0	27.7	5.9 g =	20	2	4.2	12.6	21.0	12.6
13	253.5	88.7	29.3	6.2 g =	22	2	4.4	13.3	22.2	13.3
14	287.5	100.6	33.2	7.0 g =	25	2	5.0	15.1	25.2	15.1
15	316.3	110.7	36.5	7.7 g =	27	2	5.5	16.6	27.7	16.6
16	338.8	118.6	39.1	8.3 g =	29	2	5.9	17.8	29.6	17.8
17	358.5	125.5	41.4	8.8 g =	31	2	6.3	18.8	31.4	18.8
18	390.0	136.5	45.0	9.6 g =	34	2	6.8	20.5	34.1	20.5
19	418.8	146.6	48.4	10.3 g =	37	2	7.3	22.0	36.6	22.0
20	444.5	155.6	51.3	10.9 g =	39	2	7.8	23.3	38.9	23.3
21	475.0	166.3	54.9	11.6 g =	41	2	8.3	24.9	41.6	24.9
22	513.3	179.6	59.3	12.6 g =	44	2	9.0	35.9	44.9	18.0
23	560.8	196.3	64.8	13.7 g =	48	2	9.8	39.3	49.1	19.6
24	596.5	208.8	68.9	14.6 g =	51	2	10.4	41.8	52.2	20.9
25	631.3	220.9	72.9	15.5 g =	54	2	11.0	44.2	55.2	22.1
26	665.3	232.8	76.8	16.3 g =	57	2	11.6	46.6	58.2	23.3
27	700.0	245.0	80.9	17.2 g =	60	2	12.3	49.0	61.3	24.5
28	743.0	260.1	85.8	18.2 g =	64	2	13.0	52.0	65.0	26.0
29	786.3	275.2	90.8	19.3 g =	68	2	13.8	55.0	68.8	27.5
30	825.8	289.0	95.4	20.2 g =	71	2	14.5	57.8	72.3	28.9

Table 42. Relative proportions of dietary components, fresh weight

Dietary component	Day (d) 0–2	d 3–10	d 11–21	d 22–30
Pellet	20%	25%	33%	33%
Insects (crickets, waxworms)	10%	10%	7%	7%
Egg	0%	5%	5%	5%
Greens	5%	10%	15%	20%
Vegetables	25%	25%	25%	25%
Fruits	40%	25%	15%	10%

Table 43. Kori bustard chick hand-rearing diet, food items and feeding information by dietary component category

Dietary component	Food items and feeding information
Pellet	Mixture of 50% Ratite + 50% Gamebird Maintenance **or** Crane Maintenance, by weight Product Examples: Mazuri® Ratite Diet (5647) Mazuri® Exotic Gamebird Maintenance (5643) Zeigler® Crane Maintenance
Insects	Crickets + Waxworms, offered daily / Mealworms occasionally after d20 Crickets and mealworms must be maintained on a high calcium insect diet 72h prior to feeding Product Examples: Marion® Zoological Insect Supplement Mazuri® Hi-Ca Cricket Diet Zeigler® Hi-Cal Cricket
Egg	Whole egg, including finely chopped shell, scrambled and cooked (microwave)
Greens	Offer a mixture of 2+ varieties: Chicory (Endive) Dandelion Kale Romaine Spinach Chicks will readily consume increasing quantities of greens through ~d45 and as through the first year
Vegetables	Offer a mixture: Peas, frozen, thawed Green beans, frozen, thawed
Fruits	Offer a mixture of 2+ varieties beginning d3: Watermelon: offer as 100% of fruit allotment days 0–2 (for hydration) Apple Banana Cantaloupe Grapes Honeydew Papaya Fruit initially serves as an important source of hydration, and is then decreased with an increased emphasis on the other diet components.

Table 44. Example proportions for kori bustard pellet-carnivore diet "meatballs"

Amount (g)	Meatball ingredients and information
150 g	Mazuri® Exotic Gamebird
50 g	Mazuri® Ratite Diet
250 g	Water Allow water to fully absorb, store mixture under refrigeration overnight
150 g	Commercial carnivore diet Thaw under refrigeration overnight

	<u>Product Examples:</u> Natural Balance® Meat-Eating Bird Diet / Carnivore Diet, various Central Nebraska® Bird of Prey Diet / Carnivore Diet, various

Appendix M: Buff-crested Bustard Chick Hand-rearing Protocol

Day	Brooder/Temp.	Number of feedings	Notes
0	• 37.2 °C (99 °F) when chick first placed in brooder and then lowered to 35.5 °C (96 °F) by the end of the first day.	0	• Let chick rest until first feeding which should occur 24 hours post hatch.
1	• 37.2 °C (99 °F when chick first placed in brooder and then lowered to 35.5 °C (96 °F) and adjust as dictated by the chick's reaction (panting, shivering). • Brooder: 60 cm x 30 cm x 30 cm (24 in. x 12 in. x 12 in.) floor is carpeted (or covered with non-slip material) and a feather duster made out of bustard feathers is hung in a corner. • Single chicks are given a mirror to help curtail imprinting.	5	• Chicks should be fed every 3 hours for the first 24 hours post hatch. Night feedings may be necessary if the time elapsed since hatch is 24– 36 hours. • Refer to Table 45 for amounts. • Water is provided via food items dipped in water or offered via an eye dropper or 1ml syringe at each feeding. • Offer diet via a blunt end tweezer • Crickets must be maintained on a high calcium insect diet for 72 hours prior to being fed. • Healthy chicks will attempt to grab offered food. Birds that do not show interest in food may be dehydrated. Puffy legs are a good sign of hydration, if skin on the legs appears tight, hydration is poor and chicks must be given SQ fluids. • Chicks will often have poor aim when attempting to eat, but this improves on day 3. • For the first 2 weeks, cut all items into small pieces and as the chick grows, increase the size of the pieces in proportion to the size of the chick. • Breeder vitamin mix (50% Rep-Cal® without vitamin D: 50% Vionate®, by weight) should be added at a rate of 1/8 tsp. for every 10 g (0.04 oz.) of food offered. The vitamins should be sprinkled over the entire amount of food offered for the day • The surface of the brooder should have a non-slip surface such as cloth towels, shelf liner or indoor-outdoor carpeting with low nap
2	• 35 °C (95 °F) • Reduce the temperature one degree every day until 29 °C (85 °F) is reached, but adjust as dictated by the chick's reaction (panting, shivering, etc.)	5	• Chicks are fed at three-hour intervals from 0700h to 1900h. Refer to Table 44 for daily amounts of food to be fed. Chicks should be offered nutritionally complete food items first (pellets) followed by fruits, crickets, vegetables • Healthy chicks normally lose 10% of its weight on Day 1 and Day 2, but their weights stabilize on Day 2 and increase thereafter. • Chicks can stand and walk on Day 2. • Night feedings are not necessary provided the chick is gaining weight. • Limit waxworms to 2–3 per day • The surface of the brooder should have a non-slip surface such as cloth towels, shelf liner or indoor-outdoor carpeting with low nap

Day	Brooder/Temp.	Number of feedings	Notes
3	• 33.9 °C (93 °F) • Several feather dusters are suspended near the heat bulbs. • Single chicks are given a mirror. Mirrors can be used with multiple chicks, but this may cause some chicks to be agitated; the mirror should be removed if this is the case.	5	• Feed every 3 hours starting at 0700h and ending at 1900h. • Chicks must be encouraged to exercise in the pen following each feeding to avoid problems such as slipped tendon. • Chick should have a minimum of 20 minutes outdoors daily after day 10 if above 16 °C (60 ˚F) (watch for chick's response to the outdoor temperatures). More time is allowed if chick is tolerating it well and has an area to get out of full sun if he chooses. Dappled sunlight is best. If outdoors, be sure the bird is safe from predators and chick cannot eat substrate. • Limit waxworms to 2–3 per day • The surface of the brooder should have a non-slip surface such as cloth towels, shelf liner or indoor-outdoor carpeting with low nap
4–7	• 33.3 °C (92 °F)	5	• Feed every 3 hours starting at 0700h and ending at 1900h. • Limit waxworms to 2–3 per day
8–14	• At 2 weeks, move chick to larger area 122 cm x 81 cm x 30.5 cm (48 in. x 32 in. x 12 in.). The carpeted nursery area should be 1.8 m x 3.7 m (6 ft. x 12 ft.). • A heat bulb is suspended from the ceiling over one end of the brooder so that a separate section of floor is kept at 35 ºC (95 °F). The temperature of the area should not dip below 21 °C (70 °F).	4	• Feed every 4 hours starting at 0700h and ending at 1900h • Hand-reared chicks may develop slipped wings (i.e., an outward turning of the manus) anywhere from Day 7–11. Slipped wing is easily and permanently corrected if the primaries of the affected wing(s) are taped to the body in a natural position for 7–10 days at the first sign of the problem. • At day 10, a small dish with pebbles or large marbles in it can be left in the brooder • Limit waxworms to 2–3 per day • Dappled sunlight is best. If outdoors, be sure the bird is safe from predators and chick cannot eat substrate. • The surface of the brooder should have a non-slip surface such as cloth towels, shelf liner or indoor-outdoor carpeting with low nap
15–30	• The temperature of the area should not dip below 21 °C (70 °F).	3	• Feedings may be reduced to three per day (every 4 hours) between the hours of 0700h and 1900h. • Chicks are encouraged to pick up food pieces on their own from a dish. Food is left with chicks at all times to encourage self-feeding • Limit waxworms to 2–3 per day • By the end of the first month, the chick should be receiving diet items of the same size as that offered to the adults. • The surface of the brooder should have a non-slip surface such as cloth towels, shelf liner or indoor-outdoor carpeting with low nap
4–6 weeks	• The temperature of the area should not dip below 21 °C (70 °F).		

Day	Brooder/Temp.	Number of feedings	Notes
2 months			• Chicks may be moved outside into a temporary playpen covered in mats if the temperatures allow. • Chicks should be fully feathered at this point. • Rat predation is still a real threat so 1.3 cm (0.5 in.) small mesh wire should be used.

Table 45. Buff-crested bustard feeding amounts

Day	% Body WT.	# Feedings	Items
1–7	25%	5	Chopped fruit, pinkie, cricket bodies, Mazuri softbill pellets, mixed veggies, mealworms, waxworms (limit to 2–3 per day)*
8–14	25%	4	Chopped fruit, pinkie, cricket bodies, softbill pellets, mixed veggies, mealworms, waxworms (limit to 3–5 per day)*
15–21	25%	3	Chopped fruit, fuzzies, crickets, softbill pellets, mixed veggies, mealworms, waxworms,* Mazuri exotic gamebird maintenance
22–28	20%	3	Chopped fruit, fuzzies, crickets, softbill pellets, mixed veggies, mealworms, waxworms, Mazuri exotic gamebird maintenance, scratch grains, giant mealworms
20–60	15%	3	Chopped fruit, fuzzies, crickets, softbill pellets, mixed veggies, mealworms, waxworms, Mazuri exotic gamebird maintenance, scratch grains, giant mealworms

*The head of all small worms should be pinched prior to feeding.

Appendix N: Recommendations for Developing an Institutional Flight Restriction Policy

Developed by the AZA Avian Scientific Advisory Group, December, 2013

Recommendations:

Flight restriction is used by zoo/aquarium bird managers, primarily as a method to allow the display of birds in open spaces while precluding the birds from using flight to depart these spaces. Flight restriction can be accomplished using a variety of reversible or irreversible methods. It is important to note that each method may have benefits associated with it from both an animal welfare and institutional perspective. Therefore, the AZA Avian Scientific Advisory Committee (ASAG) recommends that:

1) Each AZA-accredited institution develops a written policy on if, when, and how flight restriction is employed. The AZA ASAG should be contacted if further information is needed.
2) Institutional flight restriction policies follow species-specific guidelines developed by the avian TAGs or SSPs.

The AZA ASAG encourages all AZA Avian TAGs and AZA institutions to collect data that could be relevant to the choice of flight restriction methodologies on individual animals. It recommends that appropriate scientific and veterinary reviews and investigations into the effects of flight restriction be conducted to best assess welfare considerations.

General Information on Flight Restriction Methods:

Reversible: There are several methods of flight restriction that are reversible. These methods vary in how quickly birds can regain flight when the method is removed:

- **Netted enclosure** – covered aviaries are a method of flight restriction and employed for a variety of avian species.
- **Tethering** – this is primarily used for raptors in educational shows and programs although other types of birds may also be affected. Tethering involves attaching a leash or tether to a bird's leg. The range of movement for the tethered bird depends on the design and size of the tethering device. This method should only be used for birds that have been conditioned to tolerate it.
- **Brailing** – a leather or plastic strap (brail) fitted around the primaries and patagium to bind the wing in a closed position. Generally used when temporary flight restriction is needed (such as during pairing introductions between adult birds). It is recommended that full flight ability be restored as soon as possible. Brailing should be done under the supervision of trained veterinary staff to ensure no permanent damage occurs to the brailed wing.
- **Vane trimming** – vanes of some of the primary and secondary feathers are cut to reduce lift and prevent flight. May be used for young birds until they are old enough to be feather clipped in the more general way (see point below).
- **Wing (feather) clipping** – cutting the distal portion of some or all of the primary and secondary feathers. Care should be taken to check for, and not to cut, developing feathers with a live blood supply. For some species feather clipping will need to be done only after the next full molt (annually or bi- annually). Species that molt sequentially may need to be clipped monthly or so and it may be more often in certain species.

Irreversible: There are several methods of flight restriction that are irreversible and will render birds flightless:

- **Pinioning** - the surgical removal of part of the metacarpal bone and the phalanges of one wing of a bird. This is commonly performed within the first days of life when the process is considered a minor veterinary medical procedure. Pinioning at this age may, or may not, be performed by trained veterinary staff. Pinioning after 7 days is a surgical procedure requiring anesthesia to be performed by a qualified veterinarian.
- **Tenotomy** – a surgical procedure requiring anesthesia to be performed by a qualified veterinarian involves severing the extensors of the wing. This procedure is generally performed on fully grown birds. Some tenotomized birds are still capable of limited flight. This procedure appears to be uncommon in AZA-accredited institutions at this time.
- **Tenectomy** – a surgical procedure requiring anesthesia to be performed by a qualified veterinarian which entails removal of a portion of the extensor tendons of the wing.
- **Patagiectomy** – a surgical procedure requiring anesthesia to be performed by a qualified veterinarian that entails removal of the patagial membrane and apposition of the radius and humerus. This is often done on fully grown birds as a less complicated method than pinioning. This procedure appears to be uncommon in AZA-accredited institutions.
- **Functional Ankylosis** – a surgical procedure requiring anesthesia to be performed by a qualified veterinarian that entails fixing the ulna, carpal and metacarpal bones with stainless steel wire. This procedure appears to be uncommon in AZA-accredited institutions.
- **Radical Amputation** – a surgical procedure requiring anesthesia to be performed by a qualified veterinarian that entails removal of the whole wing. This is generally an emergency procedure occurring as a result of significant wing trauma and not generally associated with flight restriction for other reasons.

Potential Considerations When Developing an Institutional Flight Restriction Policy

When developing an Institutional Flight Restriction Policy, several considerations should be taken into account depending on the methods selected to restrict flight. These include the increased potential:

- To provide larger, more naturalistic environments.
- To reduce aggression to/from new or existing enclosure mates.
- To mitigate injury from flying into objects within the habitat.
- If legally required, to meet federal and/or state requirements to ensure that the birds are not accidentally introduced to the wild population.
- For stress and/or injury from implementing the flight restriction method, including during handling and/or the procedures.
- For stress from inability to fly.
- For injury from enclosure mates or wild predators related to reduced mobility.
- For the reduction and/or loss of reproductive capacity and ability to perform courtship displays.

The AZA ASAG has experts and expert resources available to any institution as it develops its policies and as it makes determinations regarding flight restrictions for species and individuals.