ANDEAN CONDOR
(*Vultur gryphus*)
CARE MANUAL

Andean Condor (*Vultur gryphus*) Care Manual

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Authors and Significant Contributors:

Michael Mace, San Diego Zoo's Wild Animal Park, Andean Condor SSP Coordinator
John Azua, Denver Zoological Gardens, Andean Condor SSP Treasurer
David Oehler, Cincinnati Zoo and Botanical Garden
Dr. Bruce Rideout, San Diego Zoo, Andean Condor SSP Pathology Advisor
Dr. Nadine Lamberski, Andean Condor SSP Veterinary Advisor
Dr. Michael Schlegel, Andean Condor SSP Nutrition Advisor
Mike Taylor, White Oak Conservation Center
Darcy Henthorn, Oklahoma City Zoological Park
Susie Kasielke, Los Angeles Zoo and Botanical Gardens, Andean Condor Studbook Keeper
Chriss Kmiecik**,** Cleveland Metroparks Zoo, Andean Condor SSP Education Advisor
Kim Caldwell, San Diego Zoo's Wild Animal Park
Colleen Lynch, Lincoln Park Zoological Gardens, Andean Condor SSP Population Advisor
Courtney Kelly, BREC's Baton Rouge Zoo

Reviewers:

Michael Mace, San Diego Zoo's Wild Animal Park, Andean Condor SSP Coordinator
John Azua, Denver Zoological Gardens, Andean Condor SSP Treasurer
Barbara Henry, MS, Cincinnati Zoo and Botanical Gardens, Nutrition Advisory Group Chair
Candice Dorsey Ph.D., AZA, Director, Animal Conservation
Debborah Colbert Ph.D., AZA, Vice President, Animal Conservation
Paul Boyle Ph.D., AZA, Senior Vice President, Conservation and Education

Andean Condor Care Manual Project Consultant:

Joseph C.E. Barber, Ph.D.

Cover Photo Credits: San Diego Zoo Global

AZA Staff Editors:

Candice Dorsey, Ph.D., Director, Animal Conservation
Debborah Colbert, Ph.D. Vice President, Animal Conservation
Lacey Byrnes, B.S., ACM Intern

This nutrition chapter is an excerpt from the complete Animal Care Manual available at the

Association of Zoos and Aquariums (AZA)'s website:

http://www.aza.org/animal-care-manuals/

Further information about diets and the nutrition of this and other species can be found at the

AZA's Nutrition Advisory Group (NAG)'s website:

http://nagonline.net

Chapter 5. Nutrition

5.1 Nutritional Requirements

A formal nutrition program is recommended to meet the behavioral and nutritional needs of all animals (AZA Accreditation Standard 2.6.2). Diets should be developed using the recommendations of institutional veterinarians and nutritionists, the AZA Raptor TAG and Andean Condor SSP, and the AZA Nutrition Advisory Group (www.nagonline.net/feeding_guidelines.htm). 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)** A formal nutrition program is recommended to meet the behavioral and nutritional needs of all species and specimens within the collection.

Feeding Strategies: Andean condors are carrion consuming birds, and have been observed to feed on carcasses of domestic animals (e.g., cattle and horses) in Columbia (McGahan 1972). In Peru, condors have been observed feeding on feral burros, goats, and dead sea lions and sea birds that have washed up on the shore (McGahan 1972). Wiggins (1945) documented condors feeding on a calf in Ecuador. Gailey and Bolwig (1973) cite information provided by Lint (1959) and Koford (1953) that describes condors hunting and killing their own prey in some situations. In addition to killing birds and marmots, wild condors may also prey upon llamas, alpacas, and sheep (Gailey & Bolwig 1973).

Klasing (1998) provides an overview of the unique aspects of carnivorous bird digestive physiology and anatomy that are relevant to Andean condors. The highlights are listed below:

- A hooked tip of the beak to aid in holding and ripping prey.
- Minimal feathers on head, minimizing contamination during consumption of entrails.
- A very expandable esophagus and distinct crops.
- A larger proventriculus and gizzard to accommodate large meals high in protein.
- The gizzards of many carnivorous birds lack distinct pairs of thin and thick muscle, and the gizzard functions to massage and mix the contents rather than grind them.
- Relatively small pancreas.
- Short small intestine.
- Vultures have relatively longer intestinal length than other Falconiformes.
- Carnivorous birds have extensive lymphatic cells and nodes in the epithelium of the ceca and rectum suggesting active immunosurveillance.
- Autoenzymatic digestion and a slow rate of passage to digest food efficiently.
- Consumption and partial digestion of bone is important to provide adequate calcium.

Nutritional Requirements: There are no established nutrient requirements for raptors or vultures that are applicable to Andean condors. A combination of the requirements of a strict carnivore (e.g., domestic cat) and large poultry (e.g., turkey) could be used as models to develop target nutrient ranges for Andean condors (see Table 4 for cat and poultry target nutrient levels).

5.2 Diets

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

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

Sample Diet: Two sample diets for Andean condors are listed below in Table 3. While both diets utilize similar food items, one includes the provision of beef spleen. Spleen is high in minerals and some water-soluble vitamins. Indeed, most of the minerals and vitamins found in prey items are found in the organs, and organ meat is essential for proper nutrition. It is important when whole prey items are fed to Andean condors that the carcass is not eviscerated or deboned. Similarly, consumption of bone by Andean condors is needed for proper calcium nutrition. The storage of frozen prey for extended periods of time (>6 months) can result in a reduction in vitamin E. (Bernard & Allen 2002) To account for this decrease, prey items may be supplemented with vitamin E at a rate of 100 IU/kg fresh weight prior to feeding (Bernard & Allen 2002). When fish makes up more than 25% of the diet dry matter, the fish should also be supplemented with 25-30 mg of thiamin per kg fresh weight to replace the thiamin that was lost due to thiaminase activity in the fish (Bernard and Allen, 2002).

Vitamin E deficiencies have produced a decrease in fertility and egg hatchability, skeletal muscle and myocardial degeneration, and steatitis in mature birds (Calle et al. 1989). In embryos and chicks, vitamin E deficiency can produce encephalomalacia, exudative diathesis, and neuronal, cardiac and pipping muscle degeneration (Calle et al. 1989). If Andean condors do not consume a nutritionally balanced and formulated carnivore diet, additional mineral and vitamin supplementation may also be warranted using a commercial product that is designed to supplement slab meat without bones or whole prey. It is important to weigh the birds regularly and adjust the quantity of food as needed to prevent birds from becoming obese. Examples of diet items, weights, and feeding schedules for Andean condors are provided in Table 5A.

The diets provided in Table 3 can be used for maintenance and breeding, but when chicks are being parent reared, fast days should be eliminated so parents can feed chicks daily by adding rats and/or carnivore diet during the fast days. If females lay thin-shelled eggs, the diet can be supplemented with calcium carbonate to increase the total diet concentration to the upper recommended range.

Table 3: Two sample diets from AZA institutions for maintaining and breeding male and female Andean condors.

		Measure	Weight (g)	Schedule
Diet 1	Rabbit, large	1 whole	1300	Su
	Rabbit, large	½ whole	650	Tu
	Rat, adult	2 whole	630	Tu W
	Spleen, beef	¼ whole	980	W
	Trout, medium	1 whole	215	F
	Spleen, beef	½ whole	1360	F
	Carnivore Diet[1]	1 lb	454	F
Diet 2	Rabbit	1 whole	1000	Su
	Carnivore Diet[14]	3 lbs	1360	Tu
	Rat, adult	2 whole	630	W
	Trout, medium	4 whole	865	F

[1]Body weight ranges for females: mean 9.5 kg (8.0 – 11.4 kg) and for males: mean 10.9 (9.9 – 12.5).
[2]No additional supplements
[2]Fast days are provided on 3 days a week to simulate natural feeding ecology.
[1] Guaranteed analysis (as-fed): moisture 70%, protein 18%, fat 5%, crude fiber 2%, ash 3%, calcium 0.6%, phosphorus 0.3%.

The nutrient composition of the sample diets list in Table 3 is compared to Table 4, the known nutrient requirements of cats and turkeys, which are used as the model species for Andean condors until species-specific nutrient profiles are established for this species.

Table 4: Nutrient composition of Andean condor sample diets compared to target nutrient concentrations for turkey and cat nutrient requirement models

Nutrient Composition	Diet 1	Diet 2	Suggested range	Turkey[3]	Cat[4]	
				Laying hen	Maintenance	Growth
DM. %	27.61	30.4	---	---	---	---
CP, %	63.91	56.62	>20	15.56	20	22.5
Arginine, %	1.615	1.219	0.67-0.96	0.67	0.77	0.96
Lysine, %	1.99	1.39	0.67-0.85	0.67	0.34	0.85
Methionine, %	0.53	0.45	0.17-0.44	0.22	0.17	0.44
Meth & Cys, %	1.25	0.72	0.34 – 0.88	0.44	0.34	0.88
Fat, %	22.1	26.0	>10	1.2	9	9
Ca, %	1.82	2.27	0.8-2.5	2.50	0.29	0.80
P, %	1.62	1.59	0.39-0.72	0.39	0.26	0.72
Ca:P ratio	1.12	1.43	>1.0	6.41	1.12	1.11
K, %	1.22	1.01	0.4-0.67	0.67	0.52	0.4
Na, %	0.47	0.54	0.1-0.13	0.13	0.068	0.1
Mg, %	0.16	0.27	0.04-0.06	0.06	0.04	0.04
Fe, ppm	974	361	>80	67	80	80
Cu, ppm	25	23	>9	8.8	5	8.4
Mn, ppm	26	23	>67	67	4.8	4.8
Zn, ppm	165	156	>75	72	74	75
I, ppm	0.11	0.48	0.44	0.44	1.4	1.8
Se, ppm	0.86	0.80	0.3	0.22	0.3	0.3
Vit. A, IU/kg	39,946	34,381	5,000	5,000	3,333	3,333
Vit. D, IU/kg	153	638	224-1100	1100	281	224
Vit. E, IU/kg	70	170	25-30	25	38	38
Vit. C, ppm	678	458	---	---	---	---
Thiamin, ppm	2.0[5]	5.6[5]	>6	2.0	5.6	5.5
Riboflavin, ppm	6.8[5]	9.1[5]	>5	4.0	4.6	4.0
Metabolizable energy						
kcal/g	7.85	6.64	---	---	---	---
kcal/day	1826	1111	588-661 [6]	---	---	---

[1]NRC 1994
[2]NRC 2006
[3]The addition of 1.1 g and 4.2 g of calcium carbonate daily would increase the total dietary calcium to 2.0% and 2.5%, respectively
[4] Calle et al. (1989) suggest that a vitamin E concentration of 220 - 330 IU/kg DM improves hatchability and chick vigor can stimulates reproduction.
[5]Nutrient content for prey items are not known.
[6]Based on a body weight range of 9.4-11kg and using the equation 115 x $(BW,kg)^{0.729}$ to estimate the amount of metabolizable energy (kcal) required for maintenance daily (Robbins 1983).

AZA accreditation standard

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

Food Preparation: Food preparation must be performed in accordance with all relevant federal, state, or local regulations (AZA Accreditation Standard 2.6.1). Meat processed on site must be processed following all USDA standards.

The AZA Nutrition Advisory Group (NAG) recognizes the feeding of animal carcass and whole body prey as a practice desired by some AZA institutions to stimulate activity and normal feeding behavior. Carcass refers to the body of an animal other than that of rodents, rabbits, invertebrates, or day old poultry. All institutions responsible for feeding carnivores in zoos and aquariums are required to be aware of and follow the USDA policy #25. Even though policy #25 states that it is for large felids, the NAG recommends this policy be applied to all carnivores. The NAG urges institutions that choose to carcass feed to acquire the carcass from USDA inspected facilities. The NAG also recognizes that many institutions are involved in the feeding of whole body prey that differs in composition and quality from animal carcass as defined by USDA. The NAG cautions institutions that choose to feed carcasses and whole body prey about numerous hazards (pathogenic and parasitic) that exist for collection carnivores (Harrison et al. 2006). Precautions are necessary to ensure the carcass and whole body prey is wholesome. In addition to USDA policy #25, the NAG strongly recommends institutions that choose to feed carcasses and whole body prey exercise caution and employ wholesome feeding practices including the acquisition of fresh killed carcass and whole body prey, and appropriate handling to ensure rapid cool down and minimal bacterial contamination of the meat. If the carcass is not that of a neonate collected at birth, the removal of head, hide and entrails is recommended to avoid possible exposure of collection animals to pathogenic bacteria or prion diseases. Finally, and most importantly, unless the carcass or whole body prey is that of a neonate collected at birth and fed fresh or is from a USDA inspected facility, the institution is urged to freeze the carcasses and prey items solid and properly defrost them prior to offering to an animal to minimize potential parasite exposure for collection animals. The NAG only condones carcass and whole body prey feeding as part of a feeding program that ensures the diet of the animal is nutritionally balanced and wholesome.

AZA Accreditation Standard

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

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

5.3 Nutritional Evaluations

There are limited published data on blood or tissue mineral and vitamin concentrations for Andean condors. (Table 5). With respect to differences between free-ranging and zoo Andean condors and mineral nutrition, Toro et al. (1997) only evaluated calcium, phosphorus and magnesium with the free-ranging Andean condors having significantly lower magnesium than zoo condors. Serum mineral and plasma vitamin E concentrations of zoo-housed California condors (Table 6) can make a useful comparison to Andean Condor, especially when diets are very similar. The plasma vitamin E concentrations of zoo Andean and California condors are within the range of vitamin E concentrations documented by Mainka et al. (1992) and Calle et al. (1989) (see Table 7). It is interesting to note that the zoo-housed birds, in general, had greater plasma vitamin E concentrations than the free-ranging birds, which, in part may be due to the zoo birds receiving vitamin E supplementation.

Table 5: Serum mineral and plasma vitamin E concentrations form zoo-housed and free-ranging Andean Condors.

Nutrient	Zoo			Free-ranging			Reference
	N[a]	Mean ± SD[b]	Range	N	Mean ± SD[b]	Range	
Ca, mg/L	9	89 ± 4	83 – 94				Gee et al. 1981
	12	83 ± 16.8	57 - 110	19	83 ± 11.9	60 – 100	Toro et al. 1997
P, mg/L	9	22 ± 9	7 - 30				Gee et al. 1981
	12	41 ± 14.5	25 – 76	19	38 ± 11.1	21 – 56	Toro et al. 1997
Mg, mg/L	12	20 ± 4.7	13 – 27	19	15 ± 3.7	10 – 22	Toro et al. 1997
Na, meq/L	9	147 ± 3	140 – 151	-	-	-	Gee et al. 1981
K, meq/L	9	2.9 ± 0.4	2.4-3.8	-	-	-	Gee et al. 1981
Cl, meq/L	9	111 ± 2.0	108-114	-	-	-	Gee et al. 1981
Fe, mg/L	9	1.31 ± 0.36	0.68 – 1.71	-	-	-	Gee et al. 1981
Vitamin E, mg/L	2	38.1	38 – 38.2	-	-	-	Calle et al. 1989

[a]Number of individuals in sample size.
[b]Standard deviation.

Table 6: Serum mineral and plasma vitamin E concentrations from zoo-housed Californian condors as a comparison for Andean condors (unpublished data).

Nutrient	N[1]	Mean	SD	Range (low-high)
Calcium, ppm	106	86.32	2.09	0.33-11
Copper, ppm	99	0.36	0.21	0.11-10
Iron, ppm	106	1.96	0.42	0.19-28.7
Magnesium, ppm	106	22.17	1.96	16.9-50
Phosphorus, ppm	106	57.14	19.27	4.2-211
Potassium, meq/L	106	6.66	3.90	1.9-155
Sodium, meq/L	106	152.62	36.39	1.76-414
Zinc, ppm	106	1.35	0.11	0.68-2.15
Vitamin E, ppm	2	13.5	---	11.6-14.7

[1] Number of individuals with at least one serum/plasma sample analyzed.

Table 7: Serum vitamin E concentrations of zoo and free-ranging injured raptors in southern Alberta (adapted from Mainka et al. 1992).

Species	Zoo		Free-ranging	
	Vitamin E, ppm[1]	N	Vitamin E, ppm[1]	N
Swainson's Hawk (*Buteo swainsoni*)	36.5 ± 2.2	5	2.4	1
Snowy owl (*Nyctea scandiaca*)	17.9 ± 2.5	4	9.1	1
Red-tailed hawk (*Buteo jamaicensis*)	31.0, 35.5	2	---	---
Harris' hawk (*Parabuteo unicinctus*)	17.8	1	---	---
Rough-legged hawk (*Buteo lagopus*)	32.8	1	15.3 ± 1.7	3
Turkey vulture (*Cathartes aura*)	9.4	1	-	---
Great horned owl (*Bubo virginianus*)	---	---	8.7, 10.3	2
Bald eagle (*Haliaeetus leucocephalus*)	---	---	3.5	1

[1] Values with ± sign represent mean ± standard error of the mean

7.5 Assisted Rearing

Artificial Incubation: The AZA Andean Condor SSP Coordinator should always be contacted whenever Andean condor eggs are laid. Recommendations for incubating and pulling eggs may change based on seasonal population performance. Communication with the AZA Andean Condor SSP Coordinator will also facilitate foster placements of eggs or chicks when needed.

Although Andean condors are typically excellent parents given the right environment, artificial incubation of eggs and hand-rearing of chicks has been done extensively with this species. The Andean condor served as a model for developing both *in situ* and *ex situ* management techniques for application to the critically endangered California condor. More than 60 juveniles produced by the AZA SSP population have been released to the wild in South America.

Various facilities have successfully incubated Andean condor eggs at temperatures between 36.4˚C (97.5˚F) and 37.5˚C (99.5˚F) in forced air incubators, with 36.7˚C (98˚F) being most common and yielding optimum results. Eggs are typically started at 50-60% relative humidity, but this is adjusted during incubation to correct for optimum egg weight loss of 14% +/- 1% to pip. Eggs should be weighed daily and adjustments made early in incubation as the egg weight loss trend becomes increasingly difficult to change once established. Andean condor eggs may have difficulty losing sufficient weight even in a dry incubator and with the use of a room dehumidifier. Allowing eggs to remain under parental incubation for 10-14 days often helps establish a good egg weight loss trend. Eggs should be set on their sides so that they are turned around their long axes. Most incubators are set to turn eggs in opposite directions hourly, but turning every two hours works equally well. Many machines do not turn eggs through at least a 90˚ radius as is necessary for chorioallantoic membrane (CAM) completion, so it is important to also hand turn eggs 2-3 times daily through 180˚ in opposite directions regardless of the machine used.

As in any hatchery, a high level of sanitation should be maintained to prevent contamination of incubating eggs. Hands should be washed and exam gloves should be donned immediately prior to handling eggs. All water pans, wet bulbs and wicks should be replaced with freshly sterilized equipment and the hatchery cleaned twice weekly.

Candling and hatching: Andean condor eggs are some of the easiest to candle, much like white chicken eggs. Candling can be used to confirm fertility and to monitor the progress of the developing embryo and its extra-embryonic membranes. While it is not necessary to candle eggs daily once fertility is confirmed, it is useful to candle at least weekly and to mark the progression of the air cell. Once the hatching process has initiated, more frequent candling, usually every 2-4 hours, will help determine whether hatching is proceeding normally or whether and when intervention is indicated. Eggs that fail to show development on candling are best left in the incubator for 10-14 days to determine whether delayed development has occurred. Infertility cannot be determined by candling since very early dead embryos will not have blood development, so opening inviable eggs for necropsy is recommended to determine fertility and stage of mortality if applicable.

The hatching process begins about one week prior to hatching, with air cell draw down being the first observable change on candling. This results not from a true enlargement of the air cell but from a separation of the inner and outer shell membranes around the air cell edge, creating an irregular margin. The embryo's beak should be seen pushing under the air cell membrane 4-5 days prior to hatching and internal pip usually occurs about 4 days before hatch. This is usually confirmed by hearing and/or feeling vocalizations and/or by observing rhythmic respiration on candling. At this point, turning can be discontinued and the egg moved to the hatcher, which should be set ~0.3-0.5˚C (0.5-1˚F) lower than incubation temperature for best results. The embryo will externally pip approximately 3 days prior to hatch, but the pip-to-hatch interval can be quite variable. Normal self-hatching in artificially incubated California condor eggs has occurred from 45 to 96 hours after pip.

After external pip, the humidity should be increased to ≥80% relative humidity to prevent membrane drying. Placing the egg with the pip site downward in the hatcher also seems to help keep membranes moist. Monitoring of CAM vessel regression and embryonic vigor by candling will help determine if hatching is proceeding normally. After this lengthy rest, the embryo will break up the pip site, rotate and hatch by pushing the cap off the egg. It is advisable to allow umbilical vessels to dry before fully separating the chick from the shell to avoid tearing them too close to the body wall, which may result in hemorrhage or infection. The chick should be moved from the hatcher when it is maintaining sternal posture, is able to give a feeding response and preferably has defecated. The umbilical seal should be swabbed with a water-based antiseptic such as Betadine Solution®, Betadine Ointment® or dilute Nolvasan® 3-4 times daily for the first 3 days.

Eggs that do not follow the normal hatching process may require assistance. Radiographs are useful in determining whether an embryo is malpositioned. Further information on hatching assistance is provided in the guidelines below and in the book Hand-Rearing Birds (Kasielke 2007).

The brooder is typically set at 35.6-36.1˚C (96-97˚F) to start and the temperature lowered by ~0.5˚C (1˚F) daily until the chick can thermoregulate at room temperature at about 3 weeks of age. From 3-6 weeks, the chick may be housed in a rubber tub or half of a large air crate. The brooder and tub substrate is usually terrycloth towels that are rumpled into numerous folds to prevent leg splaying. At 6 weeks, the chick may be moved to an outdoor rearing chamber with supplemental radiant heat, such as an unused nestbox adjacent to adult birds. A sturdy wire mesh covering is place over the nestbox opening to allow the chick visual and auditory, but not physical, access to the adult(s) during the remainder of the rearing period (Dorrestein et al. 1980).

Rearing: Varied hand-rearing diets have been used in different facilities, with the majority based on mice and rats, beginning with pinks and graduating quickly to adult feeder animals (Kasielke 2007). Chicks may need to be fed by spoon for the first 2-3 days but should be able to self-feed from a shallow cup or bowl after that. A typical protocol includes finely minced mouse pinks mixed 2:1 with distilled water for the first 3 days, gradually transitioning to chopped fuzzy mice by 6 days, chopped peeled adult mice by 9 days and chopped whole adult mice by 12 days. Introducing fur and bones at this early age aids with proper casting. At 5 weeks, whole rats that have been slit open are added and mice are fed whole beginning at 6 weeks. By 11-12 weeks, adult diet items are being gradually introduced. Note that neither calcium nor vitamin/mineral supplements are normally used in rearing condors. This has not proved necessary due to the very slow growth rate in this species. Older hand-rearing protocols utilized enzymes, probiotics and/or regurgitant from adult birds to assist chicks with digestion but this has since been proved unnecessary (Mazza et al. 1982). Chicks are initially fed every 2.5 hours, 7-8 times daily but this schedule is gradually reduced to 2 daily feedings by 2-3 weeks of age (Zwart & Louwman 1980).

An important consideration in rearing Andean condors is the avoidance of malimprintation on human care givers. Condors should be hand-reared in strict isolation from seeing or hearing humans. Condors that are hand-reared without isolation remain tractable as juveniles but invariably become very aggressive to most handlers once they reach sexual maturity. For this reason, it is advisable to rear even condors intended for education programs in isolation until at or near fledging. This is achieved by keeping the chick in the brooder, tub or chamber in a lighted area while keepers work from behind a curtain in a darkened blind. A life-like condor hand puppet is used as a social focus rather than a feeding implement. The opposite hand can be covered with a closed sleeve of black fabric to disguise the hand but allow enough dexterity to care for the chick. Chicks can be covered with a dark fabric drape to facilitate weighing and brooder cleaning. They may be weighed daily for the first few weeks, then opportunistically. Further information on isolation rearing of condors can be found below and in the book Hand-Rearing Birds (Kasielke 2007).

****The following information on the handling and incubation of condor eggs and rearing of chicks is adapted from a current and effective protocol and is supported by the Andean Condor SSP.**

Sanitation: Prior to retrieving eggs from the condors, several steps should be taken to ensure a clean, disinfected environment for the eggs. The walls and floors of the incubation facilities should be washed with Roccal-D (1oz/gallon of water) and rinsed with water. The same surfaces should then be washed with a solution of one part bleach to 10 parts water, and then rinsed with water again. Incubators can be disinfected using the same steps, but distilled water should be used to rinse after the disinfectant. It is recommended that distilled water be purchased in one-gallon containers to ensure quick usage and avoid contamination.

In addition to the disinfectant procedures described above, an ultraviolet sterilizer can be used in the room for approximately 20 minutes prior to eggs being brought to the incubator room. This sterilizer should not be used when there are already eggs in the incubators. Once the room and incubators have been sterilized, a footbath should be placed outside the entrance door, using Vircon (1.3oz/gallon of water). After feet have been dipped and the trailer is entered, plastic slip-on booties should be worn over freshly dipped shoes to keep the floors of the incubation facility sanitary. Lab coats should always be worn upon entering the trailer, and hands should be clean (washed with soap before entering the trailer).

Exam gloves should always be worn during the handling of eggs. If booties are worn outside of trailers they should be discarded and replaced. Lab coats should be washed, and gloves and booties replaced, weekly.

Incubator preparation: Incubators should be sterilized (as described above) after they have been checked over for proper operation, and when egg laying is near. All of the working parts of the incubator should be checked before the season begins. It is recommended that the machines be run for several weeks prior to egg season. This will insure that the turners and thermostat controls are all functioning properly, and that fan belts and motors are in good working order, before the machines are sterilized and eggs put in them. Once it is determined that the machines are in good running order, they can be left running. Temperature and turning cycles should be continually monitored during this time. Temperatures should be checked 4-5 times daily and recorded on data sheets.

If there is a known standard wet-bulb/relative humidity parameter for the egg that is to go in a particular machine, it can be set a couple of days ahead of receiving the egg. Condor eggs vary in humidity requirements, and it is best to go with the history of eggs from the pair of origin and then adjust as necessary.

Collection of eggs: Prior to collecting eggs for the incubator, it is advised to have a container with fine seed (e.g., finch mix) heated to approx. 35 °C (95 °F) for transporting the eggs from the nest box to the incubator. A portable brooder can also be used if it is set up ahead of time to attain the aforementioned temperature. At least two individuals should be present to pull eggs from Andean condor pairs. Some pairs can be aggressive at nest sites, and one person will need to hold the birds off while the other removes the egg. This process can be facilitated by bringing food items into the enclosure, and enticing the birds to leave the nest box voluntarily. The second person can also assist with gates and doors as the egg is transported to the incubation facility. The eggs should only be handled with exam gloves, and not with bare hands.

Examining and preparing eggs for incubation: Once the eggs are in the incubation facility, they should be closely examined. Any excess dirt/debris or fecal material should be lightly brushed off with a scrub pad. After the debris is brushed off the surface of the egg, the egg should be checked for cracks, thin spots, and any possible abnormalities inside the egg. This is accomplished by looking closely at the surface of the egg with the naked eye, and by candling. Any cracks or serious thin spots should be repaired using white glue such as Elmer's. Paraffin can also be used. While candling, the air cell should be located and outlined with a pencil, if it is present. Eggs pulled fresh may not have an air cell.

Once the egg has been examined and repaired, it should also be measured with calipers and weighed. If the egg is non-incubated and is considered fresh, the first day of incubation should be considered day zero. If the egg has had incubation within the first 24 hours, it should be considered day one, etc. A weight-loss table should be set up, showing daily weights based on the initial weight taken and figuring in a 14% weight loss by pip. A chart based on a 12% weight loss can also be used to ensure that the egg is staying in a "safe" weight loss range between the two parameters. A computer program can be used to calculate daily weight loss.

Just before the egg is placed in the incubator, it should be clearly identified (e.g., egg number and dam's in-house I.D) and labeled with arrows and numbers to keep track of the egg's position for manual turning (e.g., 1↑, 2, 3, 4↓). All markings should be made with a #2 pencil. The point of the pencil should be oblique to the surface of the egg, and pressing hard on the shell should be avoided.

Eggs in the incubator: Eggs should be placed in the incubator horizontally, with the air cell pointing left. The egg should fit snugly in the tray so that it will not move during automatic turning. It is suggested that the eggs be placed on a softer buffer material such as nylon netting rather than directly on the metal trays. The automatic turners should be set to turn the eggs every hour. In addition, the eggs should also be turned one-quarter turn every 12 hours in one direction until a complete rotation has occurred. Then the process should be repeated in the opposite direction. Incubation temperature for California condor eggs is 36.4-36.6 °C (97.5-98 °F). The humidity readings (either as relative humidity or wet-bulb temperature) will vary with individual eggs. This will be determined with weight loss progression, as mentioned above.

In some cases, eggs will not lose enough weight even when the incubators are completely dry. If this is noticed in eggs from certain pairs, it is suggested that eggs be left with the parents for approximately 10 days before moving to the incubator. This will "set" the egg on a proper weight-loss course for the duration of incubation. The water pans that are used as the humidity source in the incubators should be changed at least twice a week, as should wet-bulb reservoirs, wicking, ear syringes, and any containers used to fill water pans. These items should be sterilized before being used again. A dishwasher/sterilizer can be used for this purpose. Staff should check incubators routinely several times a day to take temperature reading for dry and wet-bulb averages, and to ensure that the turner is working properly, as well as other incubation functions. Record sheets to record parameters, egg-tray positions, and any problems that might be noted, should be available near the machines.

Weighing and candling: Eggs should be weighed and candled on a regular basis. Candling normally can be done daily, at least through the first 14 days. Within the first four days of candling, fertility should be determined. If this is not the case, the egg should be left in the incubator for 10-14 days before it is removed to ensure there has not been delayed initial development (which has been observed in rare cases). During mid-incubation, it is not necessary to candle every day. By late term, the frequency of candling should increase to cover the critical time of the air cell drop and the chick's progression toward entering the air cell. If these activities seem to be falling behind relative to incubation, and it is noted that there seems to be a lack of activity during candling, it is suggested that the eggs be radiographed to determine if the chick is malpositioned. Based on the results of the radiographs, plans can be made to intervene in the hatching of the chick, if necessary.

Weighing the eggs can be performed on a more regular basis to keep eggs on track for attaining the desired weight loss by pip. Humidity can be adjusted accordingly to speed up or slow down weight loss to attain the aforementioned 14% weight loss.

Weighing and candling should be done with care. During candling, eggs should be held with the air cell against the light for short periods of time, and the egg rotated back and forth on its longitudinal axis with a steady, gentle motion. Weighing should be done on a buffered platform scale to ensure the egg will not roll off. As always, care should be taken to avoid contaminating the egg by using exam gloves and lab coats.

Pipping and hatching: Once it has been determined that the chick is pushing on the air cell, the egg should be checked around the clock on an hourly basis to determine the hour of pip. The egg should be turned a half turn every hour, and it can be tapped and vocalized to in order to stimulate the chick to continue the hatching process. Once the chick has pipped the shell, its pip weight should be taken, and it should be removed from the incubator and taken to a hatcher. The type of hatcher may vary. At an AZA Accredited Institution, a custom-made incubator box with wafer-controlled thermostats for incubation temperature and backup is used. The incubator has been modified to be a still-air hatcher by installing a switch to shut off the circulation fan. The control temperature is maintained between 36.1-36.6 °C (97-98 °F), and the wet-bulb temperature is between 31.1-32.2 °C (88-90 °F).

As still-air hatchers have uneven temperatures throughout, it is important to "map" the control temperature for the egg at a specific point in the machine, and place the egg in that specific spot. The temperature should be measured at the top of the egg. The egg should be "corralled" so that it cannot roll away from its mapped spot as the chick is moving inside the egg. During this time, animal care staff members should be available around the clock to monitor the egg and hatcher. At minimum, checks should be performed hourly. Beginning at one hour from pip, the egg can be stimulated every other hour by vulture noises played to the chick through a stereo player with a speaker that has been pre-installed in the hatcher.

Activity levels can be monitored and scored on a rating from 0 to 3, with 0 meaning no activity at all, and 3 meaning violent rocking or rotation. These scores can be compared to the activity of previous chicks to see how their activity levels compare at the same hour from pip. This subjective analysis can be used to get an idea of a chick's vitality compared to that of previous chicks during the pip-to-hatch interval.

Optimally, condor chicks will hatch without any assistance. Once rotation begins, it can be several hours before the chick is entirely out of the shell. The tapping of the egg and playing of vulture noises, as mentioned above, are very helpful to stimulate the chick to get out of the shell at this time. Careful monitoring of the chick's vitality should be maintained. If for some reason the chick continues to rotate without breaking shell, it is important to intervene as quickly as possible because the chick could rotate under the shell and suffocate (see below). Once the chick has capped the shell and its head is free, it can be moved from the hatcher. At this time, the chick should be examined thoroughly to check for abnormalities, etc. There may be a few tiny blood vessels going into the seal, which can be tied off and cut just above the entry into the seal. The seal area should have any waste material removed from it with a sterile swab. Any albumen that may be stuck to the chick's body can also be removed at this time. Routine cultures should be taken of the seal, cloaca, and eggshell membranes. It is important that no antiseptic be used on these surfaces until after the cultures are taken. Nolvasan Solution diluted 1:4 with distilled water can be used to clean the area around the seal and other areas. The chick can then be placed in a brooder for rearing. It is recommended that Nolvasan Solution or similar antiseptic be applied to the seal area three times a day for the first 72 hours.

- Assisted hatches: In some circumstances, it may be necessary to help a chick out at hatch. This may be due to several causes, including incubation parameters (irregular weight loss) or malpositions. The protocol for California condors is to intervene at 72 hours from pip, unless extreme circumstances dictate otherwise. The 72-hour time period was chosen because it appeared to be a point at which it was safe to proceed with a breakout, knowing that the yolk sac would be fully retracted and the blood vessels shut down. When it has been determined that a breakout needs to be done, every effort should be made to set up equipment ahead of time and to keep the area where the breakout occurs sterile. Smocks, lab coats, surgical gloves, caps, and masks should be worn. If possible, setting the breakout room temperature to at least 32.2°C (90°F) will prevent the chick from getting chilled during the procedure. Once the breakout team (which should include a veterinarian as well as keepers experienced in breakouts) is assembled, the tools, such as hemostats, tweezers, scissors etc., can be unpacked. The egg can be removed from the incubator, and the breakout can begin. Usually, the shell is broken starting from the pip site, and the hole around the area of the air cell is enlarged. As the hole is enlarged, warm saline solution can be sprayed on the membranes to enhance the visibility of blood vessels that may be active. Once the cap on the air cell has been removed, the breakout team can proceed to work down to the small end of the egg. It is best to try to look down inside the egg as far as possible to locate possible active vessels. As the breakout progresses further down the egg, the head should be freed last. At this point, it is possible to look down between the legs of the chick to view the status of the yolk sac. Hopefully, it has been fully retracted. If not, it becomes a veterinary procedure. After the head is free and the shell is removed until roughly halfway down the side, it is with little effort that the chick can be removed from the shell by gently tipping the egg downward while supporting the chick's head and upper body. The egg waste and blood vessels leading into the seal should be supported in order to not pull down on the seal. Once the chick is free, it should be examined to ensure that there are no deformities and that the seal is closed. If all appears normal, then the protocol for post-hatch, as mentioned above, should be followed.
- Malpositions: Malpositions should be treated as individual cases, depending on the circumstances. Eggs should be radiographed whenever it is suspected that they are not progressing as they should during late term. Some of the conditions for malpositions may be more difficult to deal with than others. In these cases, it is best to consult with other institutions that have dealt with malpositions for advice, as well as to work closely with veterinary staff.

Hand-rearing Protocols: After hatching, chicks may progress at different rates. The following table (Table 9) provides feeding guidelines based on records over several seasons with the SSP.

Table 9: Andean condor chick hand-rearing feeding guidelines

Age	Notes
Day 1-3	- Do not feed until chick has passed a stool - Offer minced pinkies. Supplement pinkies with calcium carbonate ($CaCO_3$) - Start to introduce puppet and get chick to eat on its own by Day 3
Day 3-4	- Graduate from minced pinkies to chopped pinkies (supplement with $CaCO_3$)
Day 6-7	- Start feeding whole, tenderized pinkies (supplement with $CaCO_3$)
Day 7-8	- Graduate to skinned fuzzy torsos
Day 13-15	- Offer small to medium mouse torsos
Day 19-21	- Chicks should be thermoregulating. Turn off heat but leave isolettes running for ventilation
Day 20-24	- Offer mouse torsos with back fur
Day 27-29	- Offer whole, peeled mice (tails removed)
Day 30	- Move from isolette to outdoor “nest area” and start on adult diet - Chicks are fed an adult diet of “bite-sized” melt/spleen, carnivore meat, and one trout (slit ventrally) - Two days a week, chicks are offered 15-16 whole mice
Day 40-45	- Add one rat (slit ventrally) to adult diet.
2-3 months	- Fast* chick one day per week (Monday).
3-4 months	- Fast* chick two days per week (Monday & Thursday).
After fledge	- Fast* chick three days per week (Monday, Thursday, & Saturday) and add one adult condor diet to adult food tubs.

* Fast days may be subject to change

7.6 Controlled Reproduction

Permanent sterilization is not recommended for Andean condors as a means to control reproduction. To prevent successful breeding between pairs of birds, males and females can be physically separated during the breeding season, or fertile eggs removed (and replaced with dummy eggs) if they are laid. See Appendix I for egg euthanasia training forms.

Egg Embryo Euthanasia

Potential Reasons for euthanasia

- Unplanned or inadvertent egg-laying outside of requests from AZA, foreign zoo associations, consortiums to not propagate certain species or offspring from specific condors.
- SSP collection management (reduce potential for overpopulation within the collection or species being phased out of collection plan).
- Eggs produced by parents with a history of genetic defects or mutations.
- No other facilities are willing/able to receive a chick/bird.
- Lack of suitable space or other resources to hold the specimen on a long-term basis without creating hardship for other collection specimens.
- Part of an approved research protocol (SSP & IACUC).
- In case of infectious disease outbreak (Avian Influenza, Exotic Newcastle’s disease for example).

Method(s) of euthanasia

Eggs from collection birds of unknown incubation stage or incubation stage 50% or greater (guideline – candling reveals circulation complete at air cell)

- Contact AZA SSP Coordinator and appropriate supervisor to discuss euthanasia and determine time. Leave eggs in nest or incubator until ready to euthanize.
- Eggs should be transported directly to the site of euthanasia. During transport handle eggs as if incubation were to proceed (handle gently; do not leave at ambient temperature for extended periods). Entered into your zoo’s Egg Log or the Keeper Egg Records.
- Euthanize by exposure to 90 – 100% CO2 for at least 20 minutes.
- Only trained staff members are allowed to euthanize eggs (see Appendix I for training documentation form).

- Eggs from collection birds less than 50% incubation (neural tube not closed; less chance of embryo feeling pain or stress; candling reveals circulation not complete at air cell):
    - Contact appropriate supervisor to discuss euthanasia and method. Document in your zoo's records.
    - Euthanize by cooling (4 hours at 4.4 °C/40 °F) or freezing (1 hour).
    - Alternative; euthanize by exposure to 90 – 100% CO_2 for at least 20 minutes.
- Research Project eggs (approved by AZA Andean Condor SSP)
    - Whenever possible, euthanasia of eggs used for teaching or research should follow the above recommendations.
    - Exceptions may be made on a case-by-case basis if approved by the IACUC. For example, in situations where use of CO_2 is not practicable (BL3 lab, field), other methods such as chilling or decapitation may be allowed. It is recommended that chilling of late-term or unknown-term eggs be followed by freezing or decapitation to ensure death.

Disposal of euthanized eggs

- Eggs potentially exposed to pathogens – dispose according to your zoo's medical waste protocol (red bags).
- Upon mutual agreement of the curator and pathologist, necropsy of euthanized eggs may be performed in special cases.

References

Amadon D. (1977). Notes on the taxonomy of vultures. Condor [Tempe], 79(4), 413-416.

Bercovitz A, and Sarver P. (1988). Comparative sex-related differences of excretory sex steroids from day-old Andean condors (*Vultur gryphus*) and peregrine falcons (*Falco peregrinus*): Non-Invasive Monitoring of Neonatal Endocrinology. *Zoo Biology*, 7: 147-153.

Bernard, J., and M.E. Allen. (2002). Feeding captive piscivorous animals: Nutritional aspects of fish as food. Nutrition Advisory Handbook Fact Sheet 005. available at www.nagonline.net/Technical%20Papers/NAGFS00597Fish-JONIFEB24,2002MODIFIED.pdf

Bitgood, S., Patterson D., Benefield A. (1986). Understanding your visitors: ten factors that influence visitor behavior. Annual Proceedings of the American Association of Zoological Parks and Aquariums, 726-743.

Bitgood, S., Patterson D., Benefield,A. (1988). Exhibit design and visitor behavior. Environment and Behavior 20(4): 474-491.

Bruning, D.F. (1981). Rearing young South American condors. American Association of Zoological Parks Aquariums Annual Conference Proceedings: 137-142.

Bruning, D.F. (1984). Breeding the Andean condor *Vultur gryphus* at the New York Zoological Park. *International Zoo Yearbook*, 23: 11-14.

Calle PP, Dierenfeld ES, Robert ME. 1989. Serum α-tocopherol in raptors fed vitamin E-supplemented diets. *Journal of Zoo and Wildlife Medicine*, 20:62-67.

Churchman, D. (1985). How and what do recreational visitors learn at zoos? Annual Proceedings of the American Association of Zoological Parks and Aquariums, 160-167.

Conway W. (1995). Wild and zoo animal interactive management and habitat conservation. *Biodiversity and Conservation*, 4: 573-594.

Del Hoya, Elliott, Sargatal. (1994). Handbook of the Birds of the World, Volume 2, Lynx Edicions, Barcelona.

Donazar, J.A., Feijoo J.E. (2002). Social Structure of Andean Condor Roosts: Influence of Sex, Age, and Season. *The Condor*, 104(4): 832-837.

Dorrestein, G.M., et al. (1980). Hand-rearing of vultures at Wassenaar Zoo, the Netherlands. In: Proceedings of the International Symposium on Diseases of Birds Of Prey, London (Cooper JE & Greenwood AG, Eds.), Chiron Publishers, Keighley, West Yorkshire: 51-52.

Gailey, J, Bolwig N. (1973). Observations on the behavior of the Andean condor (*Vultur gryphus*). *The Condor* 75: 60-68.

Gee, G.F., Carpenter J.W., Hensler, G.L. (1981). Species differences in hematological values of captive cranes, geese, raptors, and quail. *Journal of Wildlife Management.* 45:463-483.

Harrison, T.M., Harrison, S.H., Rimbeiha, W.K., Sikarskie, J., McClean, M.(2006). Surveillance for selected bacterial and toxicologic contaminants in donated carcass meat fed to carnivores. *Journal of Zoo and Wildlife Medicine,* 37(2): 102-107.

International Air Transport Association (IATA) Live Animal Regulations (2009), 36th Edition, Montreal and Geneva.

Johnston, R.J. (1998). Exogenous factors and visitor behavior: a regression analysis of exhibit viewing time. *Environment and Behavior*, 30(3): 322-347.

Kasielke S. (2007). Condors. In: Hand-Rearing Birds. (Gage, Laurie J. and Rebecca S. Duerr, editors) Blackwell Publishing, Ames, Iowa: 171-186.

Kasielke, S. (2007). Incubation of Eggs. In: Hand-Rearing Birds. (Gage, Laurie J. and Rebecca S. Duerr, editors) Blackwell Publishing, Ames, Iowa: 39-54.

Klasing, K.C. (1998). Comparative Avian Nutrition. CABI Publishing. New York, New York.

Koford, C.B. (1953). The California Condor. Research Report No.4 of the National Audubon Society. New York. pp.154.

Lint, K.C. (1959). San Diego's Andean Condors. *Zoonooz*, 32: 3-7.

Mace, M., Kasielke. S., Lynch, C. (2007). AZA Andean Condor Species Survival Plan.

MacMillen, O. (1994). Zoomobile effectiveness: sixth graders learning vertebrate classification. Annual Proceedings of the American Association of Zoological Parks and Aquariums, 181-183.

Mainka, S.A., Cooper, R.M., Black, S.R., Dierenfeld, E. (1992). Serum α-tocopherol in captive and free-ranging raptors. *Journal of Zoo and Wildlife Medicine,* 23: 72-76.

Mazza, R., Whelan, C., Bruning, D. (1982). The hand-rearing of the South American condor at the Bronx Zoo. American Association of Zoological Parks and Aquariums Regional Conference Proceedings: 174-185.
McGahan, J. (1972). Behavior and ecology of the Andean condor parts I-III. Unpublished Ph.D. thesis, University of Wisconsin.
Morgan, J.M., Hodgkinson, M. (1999). The motivation and social orientation of visitors attending a contemporary zoological park. *Environment and Behavior*, 31(2): 227-239.
National Research Council (2006). Nutrient Requirements of Dogs and Cats. National Academies Press. Washington DC.
National Research Council(1994). Nutrient Requirements of Poultry 9th revised edition. National Academies Press. Washington DC.
Ozier, J.C. (1986). Social behavior of Andean condors at Patuxent Wildlife Research Center. M.S. Thesis, University of Georgia: 135p.
Peters, J. (1979). Checklist of the Birds of the World, Volume 1, 2nd Edition. Cambridge Press.
Povey, K.D. (2002). Close encounters: the benefits of using education program animals. Annual Proceedings of the Association of Zoos and Aquariums.
Povey, K.D., Rios, J. (2002). Using interpretive animals to deliver affective messages in zoos. *Journal of Interpretation Research,* 7: 19-28.
Robbins, C.T. (1983). Wildlife Feeding and Nutrition, 2nd ed. Academic Press. San Diego, California.
Samour, H.J., Olney, P.J.S., Herbert, D., Smith, F., White, J., Wood, D. (1984). Breeding and hand-rearing the Andean condor *Vultur gryphus* at London Zoo*.* *International Zoo Yearbook*, 23: 7-11.
Sherwood, K.P., Rallis, S.F., Stone, J. (1989). Effects of live animals vs. preserved specimens on student learning. , 8: 99-104.
Toro, H., Pavez, E.F., Gough, R.E., Montes, G., Kaleta, E.F. (1997). Etude biochimique et recherche d'anticorps vis a vis de certains agents pathogenes a partir de serums de condors (*Vultur gryphus*) vivant en liberte ou en captivite au Chili. Chemische serumeigenschaften und antikoerperstatus gegen einige aviaere infektionserreger von frei lebenden und gefangenen kondoren (*Vultur gryphus*) aus Zentralchile. Quimica serica y niveles de anticuerpos de ciertos patogenos aviares en Condores (*Vultur gryphus*) cautivos y en libertad en Chile Central. [Serum chemistry and antibody status to some avian pathogens of free-living and captive condors (*Vultur gryphus*) of Central Chile.] *Avian Pathology*, 26(2): 339-345.
United States Fish & Wildlife Service (USFWS). (1995). Convention on International Trade in Endangered Species (CITES).
Whitson, M.A., Whitson, P.D. (1969). Breeding behavior of the Andean condor (*Vultur gryphus*). *The Condor*, 71(1): 73-75, 1 Fig.
Wiggins, I.L. (1945). Observation of the South American condor. *The Condor*, 47: 167-168.
Wilkinson, R., Manning, N., et al. (1988). Artificial incubation and hand-rearing of Andean condors (*Vultur gryphus*) at Chester Zoo. In: The Hand-Rearing of Wild Animals (Colley R, editor). Proceedings of the Symposium of the Association of British Wild Animal Keepers, 13: 15-21.
Wolf, R.L., Tymitz, B.L. (1981). Studying visitor perceptions of zoo environments: a naturalistic view. In: Olney PJS. (Ed.), *International Zoo Yearbook*. Dorchester: The Zoological Society of London. pp.49-53.
Yerke, R., Burns, A.. (1991). Measuring the impact of animal shows on visitor attitudes. Annual Proceedings of the American Association of Zoological Parks and Aquariums, 532-534.
Yerke, R., Burns, A. (1993). Evaluation of the educational effectiveness of an animal show outreach program for schools. Annual Proceedings of the American Association of Zoological Parks and Aquariums, 366-368.
Zwart, P., Louwman, J.W.W. (1980). Feeding a hand-reared Andean condor and king vulture *Vultur gryphus* and *Sarcoramphus papa* at Wassenaar Zoo. *International Zoo Yearbook*; 20, 276-277.