MICRONESIAN KINGFISHER SPECIES SURVIVAL PLAN

HUSBANDRY MANUAL

Halcyon cinnamomina cinnamomina

First Edition

1998

Edited by

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DEDICATION

"It was the heavy silence. A dawn in the tropics without bird sounds bordered on the surreal. The silence was so complete that it seemed to be audible, and so eerie that I felt like shuddering.
There were no more native forest birds in southern Guam. Their last stand was in the northern third of the island. Rachel Carson's
silent spring was already a year-round affair in southern Guam. Extinction was no longer some textbook abstraction here; it was a reality – a silent reality".

L. C. Shelton, 1986

The first edition of the Micronesian Kingfisher Husbandry Manual is dedicated to Larry C. Shelton, past Curator of Birds at both the Philadelphia and Houston Zoos, and leader in the establishment of the Guam Bird Rescue Project in 1983. What started out as one individual's willingness to provide technical assistance to biologists on Guam, has turned into one of the most unique conservation opportunities to challenge modern zoos. Larry's concern about the rapid decline of endemic species and subspecies of avifauna on Guam earned him the dubitable opportunity to experience extinction first hand. Although three species disappeared before the plan could be fully implemented, the successful translocation and captive reproduction of Guam rails, Micronesian kingfishers, and Mariana crows in mainland zoos stands as a testimony to Larry's quick response. His initiative has served as an inspiration to many and it is through the dedication of individuals and institutions involved in the effort to save these species, that Larry's spirit and commitment to avian conservation lives on.

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Acknowledgments

This document represents the first edition of the Micronesian Kingfisher Husbandry Manual and fulfills a priority action recommendation (Micronesian Kingfisher SSP Action Plan, 1996) to complete and distribute a husbandry manual for this species. It could not have been completed without the help of the Management Group: B. Bahner- Philadelphia Zoological Garden, A. Cramm- Lincoln Park Zoological Gardens, S. Derrickson- National Zoological Park/Conservation and Research Center, M. Mace- San Diego Wild Animal Park, D. Rimlinger- San Diego Zoo, P. McGill- Brookfield Zoo, C. Plasse and J. Barkowski- Houston Zoological Gardens, B. Seibels- Riverbanks Zoological Park and Botanical Garden, and K. Smith- Milwaukee County Zoological Gardens; SSP advisors: K. Brock- Guam DAWR, S. Crissey- Brookfield Zoo, S. Haig- FRESC/USGS and OSU, R. Junge- St. Louis Zoological Park, D. Nichols- National Zoological Park , L. Petrik- Brookfield Zoo, and B. Toddes- Philadelphia Zoological Garden; past Propagation Group members: K. Bell, B. Bohmke, S. Branch, E. Diebold, M. Healy, P. Shannon, and C. Sheppard; Contributing researchers: R. Gonser and K. Slifka- Brookfield Zoo, K. Lewis- Philadelphia Zoological Garden, S. Marshall- Miami University; as well as those institutions formerly participating in the Micronesian Kingfisher SSP: Audubon Park and Zoological Garden, Cincinnati Zoo and Botanical Garden, Denver Zoological Gardens, Discovery Island Zoological Park, Kansas City Zoological Gardens, Lowry Park Zoological Garden, Bronx Zoo/Wildlife Conservation Park, Phoenix Zoo, National Aviary in Pittsburgh, St. Louis Zoological Park, San Antonio Zoological Gardens and Aquarium, and Sea World of Florida.

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Introduction

The Micronesian kingfisher *Halcyon cinnamomina cinnamomina* was once found throughout the island of Guam in the Mariana Islands. Populations of most of Guam's native birds, including the Micronesian Kingfisher, declined dramatically in the 1970's and 1980's as a result of predation by the introduced brown tree snake, *Boiga irregularis*. In 1984, the Guam Bird Rescue Project was initiated resulting in the capture of 29 (15.14) Micronesian kingfishers which were transferred to US zoos for captive breeding. The current population is descended from 17 (10.7) of these wild-caught individuals (Bahner 1995). The last wild Micronesian kingfisher, a single male, was sighted on Guam in 1988 and this subspecies currently survives only in captivity.

When the rescue project was initiated, very little was known about the reproductive biology or general husbandry requirements of the Micronesian kingfisher (Shelton 1986). Despite this lack of knowledge the population increased to 65 birds by the end of 1991. Unfortunately, the population began to decline soon thereafter, and numbered only 48 birds by the end of January 1993. Since this time, the population has hovered around its current size (50 to 55 birds) with adult mortalities balanced by juvenile recruitment.

At the initial master planning session at Front Royal in 1989, the target for population growth was set at 160-200 birds by the year 1996; clearly this goal has not been met. When it became clear that attainment of this goal was not possible, an action planning meeting was held at the National Zoo in 1995. The resultant Action Plan (1996) recommended publication of the husbandry manual in an effort to standardize protocols, outline our current knowledge of the husbandry requirements of the Micronesian kingfisher and identify those areas which require additional research. This recommendation subsequently was identified as an Action Plan priority at the Micronesian Kingfisher SSP meeting held at the Phoenix Zoo in 1997.

This document summarizes our current knowledge of the captive requirements of the Micronesian kingfisher and identifies information gaps that will require further research. The manual represents over 10 years of information gathered by the Species Coordinator in collaboration with the curators, keepers and researchers at participating institutions. We hope that this effort assists us in establishing a self-sustaining captive population and successfully reintroducing this species to Guam.

CHAPTER 1
GUIDELINES FOR INSTITUTIONAL PARTICIPATION IN THE MICRONESIAN KINGFISHER SPECIES SURVIVAL PLAN (SSP)

1) Institutions wishing to participate in the Micronesian Kingfisher SSP should submit a written proposal to the Species Coordinator. This proposal should include information on the institution's expertise with similar species, a description of the proposed facilities (including dimensions), management plans, and any other information which they feel qualifies them for participation. Facilities must meet minimum dimensional requirements established in the Husbandry Manual and have staff experienced in artificial incubation and hand-rearing. Non-AZA accredited institutions wishing to participate will be considered in accordance with the protocol established for application by non-AZA members.

2) The proposal will be circulated to the Management Group for review and final approval. Approval will be determined by majority vote.

3) Once approved, a Memorandum of Participation (MOP) will be sent to the new institution. An institutional representative must be appointed and the MOP signed and returned to the Species Coordinator and AZA Conservation Center.

4) Birds will be assigned to new institutions as they become available. Facilities, expertise, and other criteria will be considered in prioritizing assignments.

5) The Management Group requires that participating institutions have the ability to hold at least two pairs of kingfishers in optimal breeding conditions and recommends the ability to hold at least 3 pairs. Capacity to hold offspring of that year until placement for breeding is also required.

6) Participants must comply with **all record keeping and reporting requirements** (see Chapter 3) and be prepared to collect and share data. All holding institutions are expected to take part in efforts to document and upgrade the management protocols for this species. Participating institutions should have a good comprehension of the unique and critical status of this population and be prepared to provide maximum attention to meeting the needs of birds in their care.

7) Management Group members will be elected from institutional representatives who have expressed a dedicated interest in serving and a willingness to respond to program needs. See Responsibilities of the Micronesian Kingfisher SSP Management Group.

CHAPTER 2

RESPONSIBILITIES OF THE MICRONESIAN KINGFISHER SSP MANAGEMENT GROUP

The purpose of the Micronesian Kingfisher SSP is to develop and implement a management and conservation program for Micronesian kingfishers in North American zoos. This is a significant responsibility given that the captive population also constitutes the world population for this species.

1) Individuals interested in participating in the Management Group for this SSP must be committed to active participation and should exemplify highest standards with regard to responsible documentation and reporting of data. (see Guidelines for Institutional Participation, Chapter 1)

2) Individuals elected to the Management Group must be prepared to respond to inquiries, proposals, directives, etc. from the Species Coordinator or other Group members within the time-frame provided. Major proposals referred to the Management Group, may be adopted by majority vote. Consistent lack of response by a Management Group member may result in that person being asked to resign from the Management Group.

3) Management Group members must maintain a familiarity with the captive population through the review of published studbooks, updates, and Masterplan recommendations, as well as through consultation with the Species Coordinator and SSP Scientific Advisors.

4) Management Group members are responsible for reviewing research proposals, contributing to and reviewing masterplans, ensuring the implementation of SSP policies by participant institutions, and serving as a liaison between the zoo community and the SSP.

5) The Husbandry Manual is a dynamic document and requires input from all participating institutions. Consequently, Management group members will be expected to participate in applied research projects aimed at improving our knowledge of this species' biology and captive requirements. Members will collaborate on changes, modifications, and additions to the Husbandry Manual.

6) Group members should have institutional support for attendance at meetings. Meetings are generally held in conjunction with the annual AZA conference but special meetings or masterplanning sessions may be called at other times.

CHAPTER 3

RECORD KEEPING & REPORTING FOR THE MICRONESIAN KINGFISHER SSP

General Records

Accurate and comprehensive records are essential for the long term management and propagation of the Micronesian kingfisher. All institutions participating in the SSP are expected to participate in ISIS and comply with requests for additional information to meet the needs of the Species Coordinator/Studbook Keeper.

Institutional records should include: institutional ID number, studbook number, parentage, date of arrival and source, medical history, weights, breeding history, and necropsy information. Reporting to the studbook keeper can be greatly simplified by incorporating all of this information (especially weights) into your ARKS/ISIS records. Studbook numbers for birds can be found in the published studbooks. Birds of the year will be assigned temporary numbers (T___) when they hatch, with permanent numbers assigned at year's end. Records personnel are advised not to incorporate temporary (T___) numbers into institutional records as it creates a confusion in the ISIS central database. Please wait for permanent numbers to be assigned before updating ARKS or ISIS. Thorough medical records should be maintained for all specimens in the collection. Medical information should be entered into MEDARKS.

Micronesian kingfishers should be entered into ISIS at the subspecific level, *Halcyon cinnamomina cinnamomina*. This may require the addition of this taxon to your system. All Micronesian kingfishers, wild-caught and captive-hatched, remain the property of the Guam Division of Aquatic & Wildlife Resources (GDAWR) and should be recorded as "loans, births on loan, or loan transfers" from "AGANA". Use UNK (unknown) as Agana's vendor ID. When you are the recipient of a "loan transfer" it is important to record the shipping institution and ID number in the Special Data section. There is currently (7/98) no formal loan agreement from Guam DAWR.

Clutch/Egg Records

In order to facilitate the collection of accurate information related to reproduction in this species, a Clutch/Egg Record form was created (see Appendix J). Participants in this program must use this form to report breeding activity to the Species Coordinator. Form copies should be made from the **most recent** version, as forms are periodically updated, and used to record data for every clutch of eggs laid by birds in the collection. When the form is as complete as it can be, i.e., eggs discarded, chick fledges, or embryo/chick dies, it should be copied to the Species Coordinator/Studbook Keeper. An ARKS specimen report should also be sent at this time. It is important to fill out the clutch/egg record in its entirety and to include all pertinent information regarding the method of incubation (parent, artificial from which day, etc.). Embryo deaths/stillbirths are included in the studbook even if your institution does not choose to assign an ID number (this is important for demographic analysis).

Births/Deaths/Transfers

Because small changes in the population can greatly affect management strategy, timely reporting of hatches and deaths is critical. Assigning this responsibility to a single keeper or records technician should be considered in order to avoid reporting delays. Deaths should be reported by FAX, phone or e-mail within one week of their occurrence, with copies of necropsy and specimen reports to follow as soon as practicable. Refer to the necropsy protocol for procedural and sampling specifications. Copies of reports and appropriate samples MUST be sent to the SSP Pathology Advisor. When possible, necropsies should be cosmetic with carcasses transferred to a reputable museum for preservation. Museum acquisition numbers should be reported to the Studbook Keeper and included in the institution's permanent record. Museum numbers for other preserved specimens, e.g., eggshells, should also be recorded. As with hatches and deaths, it is desirable that the Studbook Keeper be notified when moves take place. This can be accomplished by sending a copy of the updated Specimen Report by mail or FAX. Although formal requests for Taxon Reports and institutional updates will continue to occur biannually, the dynamics of this population make it preferable to record changes as they occur. Your assistance in facilitating this is greatly appreciated.

Pair Activity and Introductory Observation Reports

Participating institutions will submit an annual Pair Activity/Evaluation Report at the end of each breeding season. When new pairs are introduced, a Summary of Introduction Day Observation form should be filled out in conjunction with the requisite observations of the pair’s introduction (see Chap. 6).

The fact that we still have much to learn about this species and need to improve reproductive and husbandry techniques suggests that any and all data collected may be of significance to the management program. Keepers and other appropriate zoo staff should be alerted to this need and every effort made to carefully document the behavior and physiological mien of this species. In particular, detailed records should be maintained on reproduction and development of chicks as our expertise in this area will determine the survivorship of the species. In an effort to improve our developmental record, institutions are asked to photograph hand-reared chicks at various stages. All of this information will be incorporated into future versions of the Husbandry Manual.

CHAPTER 4

MICRONESIAN KINGFISHER MANAGEMENT

Individual Identification Methods

The fact that the Guam subspecies of the Micronesian kingfisher is dimorphic and is housed best as a single pair (see Chap. 6, Optimal Social Groupings section) simplifies the need for intricate identification methods. However, all birds should be banded. Currently, the majority of institutions use aluminum butt end bands. A few use colored cable ties in addition. The CBSG has identified the TROVAN system as the standard for transponder systems. Therefore, it is recommended that if transponders are utilized for identification, the TROVAN system be used. Although a standard implant site has not been identified for the species, CBSG recommendations for birds suggest subcutaneous implantation on the left pectoral muscle.

Determination of Sex

(Quoted from Jenkins, 1983) "*Halcyon c. cinnamomina* is sexually dimorphic, the male possessing a cinnamon-brown head, neck, upper back, and underparts. A narrow black line extends around the nape; the orbital ring is black. The lower back, lesser wing coverts, and scapulars are deep greenish-blue. The tail is blue. The feet and irides are dark brown, and the bill is black except for some white at the base of the lower mandible (Baker 1951). The weight of five adult males collected by DAWR staff averaged 58.7 g. (range 50.5-63.8).

The adult female resembles the male except that the upper breast is paler, as are the chin and the throat, with the rest of the underparts and underwing coverts white. Immature birds have the crown washed in greenish-blue, and whitish chin and throat. Underparts are buffy-white in the immature male, but may be paler in the female.

In adults, the sexes are easily distinguished in the field. It also is possible to identify immature birds in the field, by the greenish-blue sheen of the crown; they cannot be reliably sexed, however."

In general, sexing by plumage has been a reliable method for captive birds although there have been several notable exceptions:

1- Studbook #117, originally sexed as a female based on the white breast coloration, was found to be a male on necropsy at 13 months of age.

2- Studbook #171, originally sexed as a male based on the rufous coloration of the breast feathers, was found to be a female on necropsy at 15 months of age.

While it is normally possible to sex young birds on the basis of plumage at 30 to 45 days, in some instances sex could not be determined until birds were considerably older. To facilitate rapid and accurate determination of sex, chicks must be definitively sexed at the earliest possible date by sending blood samples to ZOOGEN, INC. Lab analysis takes 7-14 days. Samples should be sent to:

ZOOGEN, Inc.
1105 Kennedy Place, Suite 4
Davis, CA 95616
Phone: 916-756-8089
Fax: 916-756-5143

*Note- Several errors have occurred with Zoogen sexing. If a bird's plumage does not match its Zoogen determination (e.g., Zoogen identifies a bird as male but it clearly has white breast feathers) notify the Species Coordinator and make arrangements to submit a second sample to Zoogen for sexing.

Weights

Individual birds should be weighed twice a year unless there is a weight or suspected health problem which necessitates more frequent weighing. Birds should not be weighed (or handled) once reproductive activity has begun for the year, rather, routine (non-medical) weighing should be timed to coincide as closely as possible to the pre and post-breeding periods. Routine weights should be taken prior to the morning feeding. More frequent weighing is required for obese birds (>80 grams) or birds that are severely underweight (<50 grams; see Table 1 for male and female weight ranges). Sudden changes in weight (increases or decreases) could indicate nutritional, medical or behavioral problems (see Chap. 8 and 9). All institutions regularly weigh hand-reared birds until they are self-sufficient. Micronesian kingfishers should also be weighed opportunistically, i.e., whenever they are in hand for other reasons (e.g., beak trimming).

Table 1. Weight ranges of captive-bred and wild-caught Micronesian kingfishers.

Sex	Birds weighed	Weight range	Reference
Male	11 wild caught	56.0-62.0 grams	Baker (1951)
	5 wild caught	50.5-63.8 grams	Jenkins (1983)
	26 captive bred	53.4-84.8 grams	
Female	10 wild caught	58.0-76.0 grams	Baker (1951)
	19 captive bred	53.0-71.8 grams	

Several institutions have been exploring the possibility of installing automated scales that would eliminate the risk associated with having to catch the birds. This would provide needed information regarding annual fluctuations in weight and their potential relationship with the breeding cycle. Although it has been suggested that both males and females gain weight prior to breeding, we have yet to substantiate this with systematic data collection. More information on this technology will be provided in subsequent editions of the husbandry manual.

Pest Control

Nest disruption due to pest infestation has not been a major factor with this species, however, one bird was predated (presumably by a rat) and at least one chick has ironically been lost due to a snake invading the nest cavity and eating it. All measures must be taken to reduce the likelihood of infestation. In outdoor exhibits, precautions to be taken may include building burrow-proof foundations and using wire of small enough window size to exclude pests of particular concern (e.g., snakes, mice, rats). Properly mounted hot-wires may be used to exclude larger predators (e.g., raccoons, feral cats, etc.) from cage tops.

Roaches may be problematic due to the risk of parasites, e.g. *Geopetitia*, which use the roach as an intermediate host. Parasite infestations in previously unexposed birds could have fatal consequences. Severe mouse infestations may cause disruption of nesting attempts due to their high level of nocturnal activity. Rats pose a direct threat to the adult birds, eggs, and chicks. Rodent control programs should be carried out with extreme care such that the birds do not come in contact with poisons or traps. In addition, there is a risk of secondary poisoning from fumes with some insecticides and these should not be used in proximity to the birds. Captive Micronesian kingfishers are known to consume both roaches and feral mice and the death of at least one adult kingfisher is believed to have been associated with impaction following the ingestion of an extremely large feral mouse.

Recommended Methods of Capture, Handling, and Restraint

By all accounts this species is fairly hardy and tolerant of routine handling, however, extreme caution is recommended in light of its critical conservation status. All captures should be carefully planned in order to minimize the amount of time the bird is being pursued and thus the likelihood of injury or imposition of unnecessary stress. As virtually all of the Micronesian kingfishers in the current captive population are maintained in pairs or as singles in small to medium-sized exhibits, it seems unlikely that methods other than simple netting of an individual will need to be used. Likewise, all hands-on procedures should be carefully planned in order to minimize the amount of time the bird is in hand. During handling, covering the head with a light cloth may further reduce stress. Non-essential handling should cease once birds begin to show signs of nesting.

The above guidelines apply to routine capture, handling, and restraint procedures such as weighing, nail and beak trimming, banding, etc. See Chap. 8 for handling and restraint for medical procedures such as radiography, surgery, and other prolonged medical procedures.

Recommended Crating and Transport Procedures

Pre-shipment

All blood work required for pre-shipment physical examinations should be done at least one month prior to a move in order to reduce the amount of stress immediately prior to shipping. The final pre-shipment visual exam can be performed within one week of shipment in order to complete the required health certificate(s). Birds should be weighed in the crate immediately prior to shipping and this information forwarded to the receiving institution. All medical records should be forwarded, as well, to the receiving institution prior to the bird's arrival for review by curatorial and veterinary staff. Information on avian TB screening and/or exposure to any bird determined to be TB positive, should be **highlighted** in the records.

Whenever possible, direct, non-stop flights should be booked in order to reduce time in transit. If non-stop service is not a possibility, various express air services may be utilized to ensure that the birds receive special handling in order to make quick connections. Shipping should take place in only the most moderate of weather conditions. A complete pre-shipment checklist is provided in Appendix A.

Transport

Specifications for transport crates are found in Table 2; all plywood should be treated with a wood sealant to facilitate cleaning.

Table 2. Recommended guideline for transport crates.

Material	exterior	0.25 inch plywood
		screened mesh cage front
		exterior bumpers
		burlap shield over screened mesh
	interior ceiling	foam rubber or burlap stuffed with straw
Dimensions	external minimum size	9 in x 9 in (22.9 cm x 27.9 cm)
	external maximum size	15 in x 15 in (38 cm^2)
	internal height	minimum 10 in (24.5 cm) clearance between floor and ceiling padding
	perching	0.5 in diameter (1.3 cm)

Post-shipment

The receiving institution should weigh the bird in the crate on arrival and obtain regular weights (approximately 2-6) during the 30 day quarantine period to verify that the bird is adjusting well to its new surroundings. Along with this suggestion, consideration must be given to the fact that each time a bird is captured it experiences stress, and different individuals may respond differently to being restrained. Food consumption should be carefully monitored during quarantine and a variety of food items (including green anoles) should be offered. Post-shipment medical screening should be postponed as late as possible in the quarantine period.

The receiving institution should contact the shipping institution prior to receiving a bird in order to ascertain the type of enclosure the bird was kept in, diet, method of food delivery, etc., and try to simulate these conditions, as nearly as possible, for the newly arrived bird.

CHAPTER 5
ENCLOSURE PARAMETERS

Dimensions and Material

The recommended minimum enclosure size for breeding pairs of kingfishers is 10 ft x 8 ft (3 m x 2.4 m) with a height of 10 ft (3 m). Although pairs have bred in enclosures as small as 6 ft x 8 ft x 8 ft (1.8 m x2.4 m x 2.4 m), problems with pair compatibility may be exacerbated by these small enclosure sizes. Pairs should be provided with the opportunity to put some distance between themselves when not breeding. Enclosures with a floor area less than 75 ft^2 (22.8 m^2) are not recommended. Ceiling heights below 8 ft (2.4 m) may not allow sufficient height for hanging nest logs (see Chap. 6). Holding cages for non-breeding, single birds should be a minimum of 4 ft x 4 ft x 4 ft (1.2 m^3), although larger spaces are preferable.

Containment should be either solid material, wire mesh, or glass. For wire enclosures, mesh size should not exceed one inch (1/2 by 1 inch is preferred). Mesh size should be smaller in outside exhibits and holding situations in order to preclude infiltration by pests (see Chap. 4). Birds have escaped from enclosures that use piano wire barriers; therefore, this type of barrier should not be used with this species. There have been several cases in which kingfishers have attacked their images reflected in glass cage fronts. Birds housed in glass-fronted enclosures should be monitored carefully to assure that this does not occur and cause injury to the bird.

As a forest bird, the Micronesian kingfisher has a natural shyness and needs adequate cover. Marshall (1989) found that kingfisher nest cavities were found in areas with approximately 80% canopy cover. An effort should be made to provide privacy for display pairs through heavy frontal planting and provision of out-of-sight perching areas. Plants should also be provided in off-exhibit areas for cover and to provide areas of refuge.

Substrate

The most common substrate used in public displays is soil or soil covered with bark mulch. Institutions have also used pine straw or a mixture of solite rock, peat moss and pine bark. Alternatives to soil substrate are recommended because of the risk of ATB exposure (see Chap. 8). In off-exhibit areas, enclosures generally have floors made of concrete which allow for daily cleaning that may minimize the risk of disease transmission or pest infestation. Concrete floors are sometimes covered with wood chips, shavings, or bark mulch. Since kingfishers prefer to perch up high and spend little time if any on the floor, the most important consideration for selecting a substrate should be effective cleaning and disinfection. Disinfectants with tuberculocidal properties greatly reduce the risk of ATB infection (see Chap. 8).

Water Source

Kingfishers must be provided with water dishes that are large enough to permit bathing, a minimum dish size of 5 in (12.7 cm) deep and with a diameter of 18 in (45.7 cm) is recommended with a water depth of 2-3 in (5.1-7.6 cm). The water dish should be elevated approximately 1 foot off of the floor for easier access by the birds. Most exhibits on public display have concrete pools, or in some cases, concrete streams with pools. Most of these pools are reported to be shallow (3-7 in; 7.6-17.8 cm), but pools that are several feet deep have been used without problems. Fresh water must be provided daily.

Feeding

Food dishes should be shallow and open to allow birds to feed on the wing. It is preferable to place feed dishes above ground level in a central location to facilitate feeding. Pairs should be provided with more than one food dish in different locations, to minimize the potential for aggression over food. It is also recommended that birds be fed twice daily. Observations have shown that kingfishers prefer to feed early in the morning and in the evening. Food cups which attach to the side of the cage (as used for psittacines) should not be used. Complete diet recommendations are provided in Chap. 9.

Temperature and Humidity

The average daily temperature on Guam is 27^{o} C (80.6 ^{o} F) with a range of 21^{o} to 30^{o} C (70-86^{o} F) with little variation throughout the year. Average humidity ranges from 65 to 75% at night and 85 to 100% during the day with the rainy season beginning in July/August and ending in October/November (Jenkins 1983, Beck and Savidge 1990). For indoor enclosures where climate can be controlled, an attempt should be made to mimic these conditions as closely as possible. For outdoor facilities (all in southern latitude zoos), birds should be provided with shading, protected enclosures and heat lamps for inclement weather. NOTE: Toxic fumes produced by Teflon-coated heat lamps have caused mortality in Micronesian kingfishers and should not be used.

Lighting - Source and Cycle

Although the seasons on Guam are defined predominantly by rainfall, there is a noticeable change of photoperiod throughout the year (Jenkins 1983). Kingfishers have successfully bred under a variety of light regimes in captivity. The most common photoperiod for indoor enclosures has been a natural daylength schedule provided through a skylight or greenhouse roof combined with a 12L:12D artificial light source. Artificial lights are provided using fluorescent or incandescent light. Because of the variation in light regimes and reproductive success with a large range of photoperiods, it is difficult to determine if kingfishers are stimulated by changes in photoperiod. Based on the limited information we have regarding the reproductive cycle of kingfishers on Guam, they appear to initiate reproduction in late December and it is likely that they are stimulated partially by the increasing photoperiod although initiation of breeding also coincides with the end of the rainy season. At this time, where the birds have no access to natural light it is recommended that pairs have some variation in photoperiod to provide them with a defined winter and summer "season". Hormonal studies are currently underway which may provide additional information regarding the effects of photoperiod on reproductive cycling in the Micronesian kingfisher. Further studies are required to determine the best photoperiod for successful reproduction in captivity.

CHAPTER 6
BEHAVIOR AND SOCIAL ORGANIZATION

For descriptions of specific behaviors, refer to Appendix B; Micronesian Kingfisher Ethogram Draft.

Optimal Social Groupings and Species Composition

The Micronesian kingfisher has a reputation as an aggressive bird in the wild, both towards conspecifics and other species. Jenkins (1983) describes the threat display and a fight between two adult males. Micronesian kingfishers have been observed harassing flocks of Bridled white-eyes *Zosterops c. conspicillata* (Marshall 1949) and Micronesian starlings, *Aplonis opaca guami* (Kibler 1950). In the early 1900's, the Micronesian kingfisher earned its reputation as a chicken thief (Seale 1901). At the Philadelphia Zoo a Micronesian kingfisher killed a superb fruit dove (*Ptilinopus superbus*) and, over a two-year period, the Pittsburgh Aviary lost one silver-eared mesia (*Leothrix argentauris*), three shama thrush (*Copsychus malabaricus*), one dhyal thrush (*Copsychus s. saularis*), and two fairy bluebirds (*Irena puella*) to kingfisher aggression.

Several zoos, e.g., Philadelphia, National, Bronx, have successfully maintained Guam rails and kingfishers together, however, a Guam rail may have been responsible for the brain injury of a Micronesian kingfisher at the Pittsburgh Aviary and the death of a kingfisher chick that went to the ground after it fledged at the Audubon Zoo. The San Diego Zoo reported the trauma-related death of a male Micronesian kingfisher resulting from aggression by a yuhina (*Yuhina* sp.) and the Brookfield Zoo reports a Micronesian kingfisher showing aggression towards a neighboring toucanet whenever the toucanet vocalized. Given the current state of the captive population, it would be inappropriate to risk housing this species with any other potentially aggressive species, leaving single-species maintenance the best option for the Micronesian kingfisher. Birds should also be monitored to assure that neighboring species are not causing a disruption as well. Another concern about mixed species exhibits is the difficulty of monitoring and controlling food intake. Given the weight control problems, incidence of hemosiderosis and other nutritional concerns, kingfishers are best housed alone.

Because of their propensity for intra-specific aggression, adult Micronesian kingfishers should be held either in breeding pairs or as singletons. Young birds can often be housed together if monitored carefully for aggression. While adults have occasionally been held in same-sex groups without incident, this practice is not recommended. In addition, because several pairs have failed to breed until neighboring pairs or singletons were removed from sight, it is recommended that all breeding pairs be visually isolated from conspecifics.

Pair Introduction Protocol

Beginning in 1990, there have been an increasing number of reports of incompatibility between paired individuals. Many of these incidences have involved one of several "problem" birds that show a propensity for aggression. The following protocol for introducing birds to each other is recommended in an attempt to decrease the potential for aggression. In addition, because so little is known about the courtship and pair bonding process of this species, it is important to gather as much information as possible from each pair introduction.

Pre-Introduction

The recommended minimum duration of the pre-introduction period is 2 weeks. Whenever appropriate birds are available, as determined by the Species Coordinator, mate choice should be attempted. The recommended procedure for introducing pairs is described both for birds with mate choice opportunities and those without.

Mate Choice

It is important to arrange perching in the cages so that all birds have perches that are clearly near one of the other birds and also have perches that are "neutral" (Figure 1). The perches labeled with "B" in the diagram below are those which allow the birds to show a clear preference while those labeled "A" are neutral perches where a preference can not be determined. If there are no neutral perches it would be impossible to determine if the birds had a preference for another bird or if there were simply no other place to sit. Two separate perches connected to each adjacent cage are preferable to one long perch through the center of the cage since this forces the choosing bird to make a clear choice of one side or the other and eliminates the ambiguity of having the bird sit near the center of the perch. Because the Micronesian kingfisher is not a demonstrative bird, preferences have been difficult to determine when mate choice has been allowed in the past. The current recommendation is to monitor the birds to determine if there are any perch preferences by noting the location of each bird as often as possible throughout the day as well as other evidence of perch preferences such as the location of droppings on the floor. All instances of aggressive behavior directed at adjacent cages should be noted as well. In addition, compatible pairs have been noted to sit together and give soft warbling vocalizations (pair vocalizations), therefore any vocalizations should be noted as well. As more information is gathered, protocols for assessing mate preferences will be better defined.

Acclimation Without Mate Choice

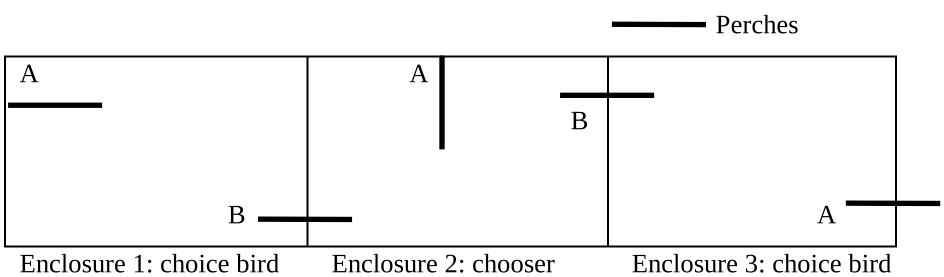
Prior to placing the birds in the same enclosure, the prospective pair should be placed in adjacent cages for acclimation prior to introduction. Placement of perches in the adjacent cages should use the same considerations as the mate choice procedure (see above) since we still want to determine all that we can about the preferences of the birds before putting them together.

During the 2 weeks that birds are housed in adjacent cages birds should be observed in order to address the following questions: Is there other evidence of preferred perches (e.g., location of droppings)? If any birds were housed in the same enclosure before acclimation, have any changed their preferred perches? For example: if a bird consistently preferred to perch in one location before other birds were placed in adjacent cages and then changed to a new preferred perch, this could indicate either a preference for or an avoidance of an adjacent bird. Have any aggressive behaviors been observed? Are there any behaviors which may indicate an interest by one of the birds? For example, prior to pairing for one reproductive pair, the male would fly and cling to the common wire divider whenever the female would move away from his cage.

Introduction Procedure

To minimize problems that could arise from territorial defense, when possible potential mates should be introduced into a neutral enclosure. All pairs should have nest logs available at the time of introduction. Several successful pairs have begun striking at the nest within 1 hour of introduction. This may be 1) important to initiate the pairing process and 2) a good early indicator of the potential for success. All pairs should be observed for at least 1 hour after introduction and a Summary Of Introduction Day Observation form (Appendix F) should be completed and submitted to the Species Coordinator.

Figure 1. Recommended perch location in cages used for mate choice. Perches labeled “A” are neutral and those labeled “B” are those where a bird is considered to have made a choice.



Mating and Reproductive Behavior

Because of their rapid decline on Guam the opportunity to conduct extensive field observations on wild pairs of Micronesian kingfishers was lost. As a result, much of the information reported in this section is compiled from a variety of sources, including (1) extensive time lapse video recordings of a single reproductive pair (male studbook #205, female studbook #61) during 2 breeding seasons at the Philadelphia Zoo; (2) data gathered from individuals who have observed Micronesian kingfishers in the wild or in captivity; (3) video recordings of breeding pairs at Lincoln Park and Milwaukee County Zoos; and (4) observations conducted during site visits to all 8 participating institutions in 1996/97.

Mating System and Territoriality

The Micronesian kingfisher is believed to be a monogamous species which forms a long-term pairbond and is highly territorial year-round. Although birds in captivity are managed with these assumptions, direct evidence to substantiate them is sparse. Wild pairs are known to nest in all months of the year except August through November (the height of the rainy season; Jenkins 1983). Based on his observations, Jenkins believed that pairs produce two clutches in a year and Sam Marshall (personal communication) observed that one pair which successfully fledged a chick, re-nested to produce another clutch of eggs soon after the chick fledged. In captivity pairs produce multiple clutches in a year and there have been females that lay eggs almost continuously throughout the year. Because of potential problems with eggshell thinning, more than 4 clutches per year are not recommended. For pairs that do not stop breeding on their own, removal of nest log(s) may be necessary. Removal should occur in the “non-breeding” season, approximately August through November.

John Groves (personal communication) observed on Guam that Micronesian kingfishers appeared to isolate themselves as much as possible from conspecifics and other species, lending support to the assumption that these birds are highly territorial in their behavior. It should be noted, however, that his observations occurred during the period of peak extinction on Guam and may not reflect their traditional spacing patterns.

Although mated pairs have been observed defending territories during the breeding season, it is not known if these pairs remain together for subsequent seasons or if these territories are maintained throughout the year. Most of the published observations of Micronesian kingfishers have been conducted during the months of highest breeding (March through July) and, with only one exception, birds have always been observed in pairs during these months (Kibler 1950, Baker 1951, and Jenkins 1983). This exception occurred during Marshall’s nest site survey when single males were common during the breeding season presumably because of the small number of females still alive on Guam in 1985. Stophlet (1946) reported observations during October and November, a time when Jenkins reports that no active nests have been found; he documented a total of 10 single males and 2 single females but only one pair. Therefore, it is not clear that Micronesian kingfishers do, in fact, remain paired throughout the year and this should be considered for future captive management.

Courtship and Pair Formation

The details of much of this process remain a mystery due to a lack of systematic observations of banded wild individuals prior to the extinction of the Micronesian kingfisher on Guam. There are still healthy populations of congeners on Palau (*H. c. pelewensis*) and Pohnpei (*H. c. reichenbachii*), however, and an effort is currently underway to initiate field studies on one of these closely related subspecies. The establishment of protocols for standard observations of newly paired birds in captivity (Appendix C) and an increase in our ability to allow individuals to choose their own mates (as a result of the consolidation of birds to 8 institutions as of January 1997) should provide the opportunity to answer some of the questions regarding the process of pair bonding in this species.

Several observers have suggested that the process of excavating a nest cavity plays a substantial role in pair formation or the maintenance of pair bonds (Jenkins 1983, Shelton 1986). Micronesian kingfishers excavate a nest cavity by "...alternating thrusts of the beak at the chosen site in flights initiated from a nearby perch. Not having the foot structure necessary for perching on the vertical trunk, a bird flaps its wings vigorously (and awkwardly) as it attempts to deliver more than one blow per flight. Usually a bird is restricted to only one or two blows and returns to the perch after each attempt. A call is given upon leaving the perch with each new attempt." (Jenkins 1983).

Marshall (1989) observed that 80% of all nest excavations that were initiated were incomplete. Captive pairs have been observed engaging in excavation behavior in between clutches within a breeding season but subsequently re-nested in the same cavity that was previously used. On the day of introduction, 2 pairs that subsequently reproduced began taking alternating strikes at the nest log within 1 hour after introduction (personal observation). In contrast, 2 other pairs that were observed for the same length of time following introduction, failed to show any interest in the nest log and also failed to reproduce. These observations support the contention that the process of excavating is important for both pair formation and the maintenance of the pairbond through the breeding season, although it is clear that more information is needed to document the importance of this behavior to successful pair formation and maintenance. It is also possible that the provision of nest logs which are not soft and, therefore, difficult to excavate, could interfere with this bonding process.

Although both sexes participate in the excavation process, the male and female may allocate their time differently. During the first breeding season for pair 205/61, the male spent significantly more time inside the nest than the female prior to egg laying once the cavity had been excavated (Wilcoxon signed ranks test, $N = 17$ days, $T = 0.0$, $P < 0.001$). This difference was not apparent during the second breeding season (Wilcoxon signed ranks test, $N = 26$ days, $T = 148.5$, $P > 0.05$). Although the level of effort was more equitable during the second breeding season, the female appeared to spend more time at the nest on each of her "visits" whereas the male made more frequent visits to the nest.

Although the Micronesian kingfisher does not appear to have an elaborate courtship display the male does courtship feed his mate. Courtship feedings occur coincident with successful pair copulations (see below) and have not been observed during the early stages of pair formation, incubation or care of the young. The male may offer the female a variety of food items but the female does not always accept what he offers. The female in one pair that has been extensively observed on video (male 205/ female 61) has never refused the offer of an anole, her preferred food. On the days immediately preceding egg laying the male has been observed feeding her all of the green anoles provided for this pair.

Nest Parameters

The following is summarized from Marshall (1989) based on his description and characterization of Micronesian kingfisher nest sites and nest cavities that he found on Guam. All nest cavities in the wild were located in areas with a high degree of canopy cover (over 80%).

Nests were located in a variety of tree species including: coconut palm (*Cocos nucifera*), umumu (*Pisonia grandis*), and breadfruit trees (*Artocarpus* sp.). The average diameter (dbh) of excavated trees was approximately 17±5 in (43±13 cm). Marshall (1989) characterized tree softness using thrusts of an icepick to determine “penetrability”. The average penetrability of trees with completed cavities was approximately 3±1 in (7.5±2 cm). This was significantly softer than live trees that did not have excavations in which the average penetrability was less than half an inch. Arboreal termitaria (nests of *Nasutitermes* sp. termites) were also used for nesting; these were even softer than nest trees and, consequently, each had fewer excavations but individual cavities were slightly deeper.

Nest entrance holes were found to be about 2 in (5 cm) in diameter and nest cavity depth was approximately 6.5±1 in (17±3 cm). All nest trees had multiple excavations but most (about 80%) of these were incomplete. Pairs were observed to work on up to 5 excavation sites at a time before focusing in on one site. Nests were rarely found below 12 ft (3.7 m). While it is difficult to place nest logs at this height in captivity, attempts should be made to hang nest logs as high as is reasonable given the height of the enclosure. National Zoo has had success using a pulley system to lower and raise the nest log in order to check for eggs/nestlings with minimal disturbance to the birds.

In captivity, a wide variety of tree species have been used with success including several species of palm, willow and pine. Logs have been placed in several orientations with success. The most common is vertical placement (simulating a tree trunk) but pairs have successfully excavated and used logs that were placed horizontally and those that have been placed with the core of the log exposed so that the pair had access to the soft, rotted center without excavating through the hard outer bark. There has also been one report of a pair excavating and laying eggs in a rotted staghorn fern.

Perching is generally provided 2 to 6 ft (0.6-1.8 m) from the log to allow the birds to fly at the log to start the cavity. Nest excavation may play an important role in courtship and reproduction and every effort should be made to facilitate this process. Multiple nest logs are highly desirable, giving the birds a choice of nest sites. Provision of a very soft, rotting log is important to successful excavation. When a soft enough log cannot be found, creating a cavity conforming to the above specifications and repacking the cavity with mulch has been successful. Nest logs should be a minimum of 2 ft (0.6 m) in length with a diameter of no less than 15 in (38 cm). Appendix D outlines the recommended guidelines for use and manipulation of nesting cavities.

Copulation

Most copulations and attempts occur within 3 weeks of egg laying and mounting frequency peaks during the week preceding oviposition (Figure 2). Mountings have been observed during brooding for the pair observed by Marshall on Guam (personal communication) and have been noted to occur in captivity as well. Most mountings for the one videotaped pair, however, did not result in cloacal contact. For this pair, mountings without cloacal contact outnumbered successful copulation by approximately 7:1. Mountings without cloacal contact are not uncommon among birds and may play a role in pair bond formation and maintenance (Birkhead and Moller 1992). At least one successful copulation did precede the laying of each egg produced by this pair (male 205/ female 61) by approximately 2 days. Male and female behavior during mounting and copulation is fairly stereotyped; descriptions of 2 successful copulations are given below.

1) Pair 205/61 at the Philadelphia Zoo-

The video sequence began with the female on the perch directly in front of and facing the nest log and the male about 6 inches to her left and facing the camera. The male turned to face the nest log (16:23:50) then faced and approached the female (16:24:52). After the female began to flatten her body and crouch low on the perch (16:25:11) the male moved closer and touched her bill as she leaned away slightly while facing him (16:25:14). She then turned to face the nest log as the male mounted while maintaining contact with her bill (16:25:15). As the female completely flattened herself on the perch, keeping her bill slightly open, the male tapped the top of her head twice while mounting. The male partially dismounted (16:25:21) but immediately adjusted his position and remounted while tapping the female's head. During this second mount, the male's head was to the left of the female's and his tail bent to the right, under hers. Cloacal contact appeared to occur twice (16:25:37 and 16:25:40). The male dismounted immediately after the second cloacal contact (16:25:45) and flew out of camera view. The female then followed (16:25:52). The time from the male's first approach until he dismounted was only 53 s but the actual mount was the longest one recorded for this pair at 30 s. There was 1 fertile and 1 infertile egg in this, their 4th clutch.

2) Pair 24/99 at the Brookfield Zoo-

Three copulations were observed on 3 occasions over 2 days; all appeared to involve successful cloacal contact. The first 2 mountings lasted just under 20 s and the third just over 20 s. In each case, the male appeared to tap or rub the female's head near the base of her bill. The male gave soft warbling vocalizations during his approach for all three copulations and throughout the copulation during the 3rd and longest mount. While it was not possible to determine if the female was also vocalizing because of the noise level in the building, male and female vocalizations were recorded during 2 copulations observed at the Houston Zoo (pair 88/92).

Incubation

Clutch size ranges from 1-3 eggs. Incubation begins with the laying of the first egg, and both sexes participate in incubation duties. Although it has been stated that the female incubates at night (Beck and Savidge 1990, Shelton 1986), observations of a pair on Guam (S. D. Marshall, personal communication) and video recording of pair 205/61 could not confirm that this was the case. Several observers have suggested that the male is responsible for more of the daytime incubation than the female. This pattern was observed in the video recording of pair 205/61, with the male and female incubating 55% and 39% of the time, respectively (Wilcoxon signed ranks test, $T=1$ $P<0.001$, $N= 22$ days, averaged over 4 clutches). Male and female incubation bouts lasted 20-40 minutes and 10-30 minutes, respectively. The nest was attended at almost all times by either the male or female, however, they spent significantly more time off of the nest during clutch # 7 than on the other 3 clutches analyzed (Wilcoxon 2-sample test, $U=344$, $P<0.001$, $n_1=19$ $n_2=22$) and this was the only clutch with a fertile egg that did not hatch. If the time spent off of the nest was the cause of embryo death (rather than the result) this suggests that Micronesian kingfisher eggs are very sensitive to temperature changes or changes in egg manipulation by the pair since the differences, although statistically significant, are not great (11% compared with 3 to 6% of the time that the nest was empty, on average, per day). It is unlikely that the time off the nest was the cause of embryo death, however, since the one pair monitored on Guam (Sam Marshall personal communication) spent more time off of the nest while incubating than this captive pair (on average 29% of the observation time in the morning and 18% in the afternoon).

Eggs should be candled at least 1 week after egg laying (also see Chapter 7). During the first week of incubation, eggs can be quite fragile and easily punctured or broken. Although it is possible to determine egg fertility earlier than 7 days, candling the eggs before this time is not recommended because of the potential risk to the developing chick.

Summary of Reproductive Parameters

Unless otherwise noted, the following is based on information collected on a total of 86 pairs between 1 January 1985 and 31 December 1995; 44 pairs produced eggs (not necessarily fertile) and 36 of those produced offspring.

Table 3. Age of egg production.

	minimum	minimum fertile	average	maximum (as of 6/97)
Females	0.67 years	0.75 years	2.12±1.03 years	10.83 years
Males	0.83 years	0.92 years	2.33±1.08 years	> 13 years*

*minimum estimated for wild-caught

Although captive-bred birds are capable of reproducing within their first year (Table 3), fecundity begins to rise after 2 years of age with peak fecundity between 3 and 5 years for both males and females.

Although pairs have produced eggs in all months of the year in captivity, there is a peak in reproduction from March through June (Figure 3). Pairs rarely initiate reproduction for the year (or for first time pairs) between August and November and this coincides with the non-breeding season on Guam.

Clutch size ranges from 1 to 3 eggs; 64% of clutches consist of 2 eggs, 32% are 1 egg clutches and only 4% are 3 egg clutches. Out of 631 eggs produced, 254 (40%) were fertile and 194 (76% of fertile eggs) hatched with 116 (60.56) viable offspring produced. Of the 116 viable offspring (i.e., lived > 30 days) 96 (47.49) lived to reproductive age (9 months, see above).

The length of incubation averages 22±1.65 days with a mode of 23 days (N=136 parent incubated eggs) with a range of 17 to 28 days. Fledging occurs, on average, 33±2.74 days after hatching (N=18 parent-reared birds) with a range of 29 to 39 days. The extreme maximum and minimum recorded values for incubation and fledging time probably result from the fact that these periods are often calculated from estimated rather than known dates.

The longevity of wild-caught and captive-bred birds may differ. The average estimated age of death for wild-caught birds is 8.5±3.1 years (N=27) and the known average age of death for captive bred birds is 3.3±2.5 years (N=76). Because wild-caught birds were adults (estimated to be 2 years of age) when they were captured, these differences could be exaggerated if there is high mortality of juvenile birds. To adjust for this, we can calculate the average age at death for captive-bred birds that lived more than 2 years (N=44) which is 4.7±1.9 years. Although the difference is not as large, wild-caught birds still appear to survive almost twice as long as captive-bred birds. There is no apparent sex difference in age at death for captive-bred birds but wild-caught males lived an average of 2 years longer than wild-caught females. As of July 1997, there are 2 wild-caught males still living, estimated to be at least 13 years of age and 40 living captive-bred birds (not including 1996 offspring) with an average age of 5.6±2.7 years. The longevity record for a captive-bred bird was a female who died at 11 years and 10 months of age. This female produced offspring in the year prior to her death and was beginning to nest at the time of her death.

Pairs that are close in age may be more productive than pairs with disparate male and female ages. Out of 46 pairs that were less than one year apart in age, 20 (43%) produced viable offspring while only 30% (12 out of 40) of pairs more than one year apart in age produced viable offspring. Of those pairs that did reproduce, pairs that are closer in age produced a slightly higher proportion of fertile eggs than those farther apart in age (0.43 vs. 0.31, respectively) but the difference was not statistically significant (t=1.22, n.s.).

Although wild-caught pairs produced more offspring than captive-bred pairs; an average of 4.5 vs. 3.2 offspring per pair, this may reflect the longer duration of pairbonds among wild-caught pairs. On average, pairs composed of wild-caught birds lasted over twice as long as pairs composed of two captive-bred birds. Overall, pairs that lasted longer were more likely to reproduce; the average reproductive pairs lasted almost 3 times longer than pairs that were unproductive. While the differences are small, wild-caught pairs have produced a higher percentage of fertile eggs than captive-bred pairs (42% and 36%, respectively) and a higher percentage of fertile eggs that hatched (84% and 68%, respectively). In general mixed pairs did not do as well as either captive-bred or wild-caught but there is a lot of variation so this is difficult to interpret. Overall, the proportion of fertile eggs has been low from the beginning. Pairs vary from producing only infertile eggs (N=7) to a few pairs who have produced only fertile eggs (N=4). On a yearly basis, the highest proportion of fertile eggs produced has been 0.65 in 1987, the second year of reproduction in captivity. Most years have seen production of fertile eggs at less than half of all eggs produced.

The cause of the observed differences in mortality and reproduction between captive-bred and wild-caught birds is not known. Rearing type is one possible explanation since (due to the high mortality of parent-reared chicks) the majority of captive-bred birds are hand-reared. The few parent-reared, captive-bred birds that survived to reproduce (4.1) did produce a higher proportion of fertile eggs than the average hand-reared bird. This was still within the range found for hand-reared birds, however, and because of the small sample size it is impossible to interpret these findings.

Figure 2. The average frequency of copulation in relation to egg laying for pair 205/61; N= 5 clutches, error bars represent 1 standard deviation from the mean.

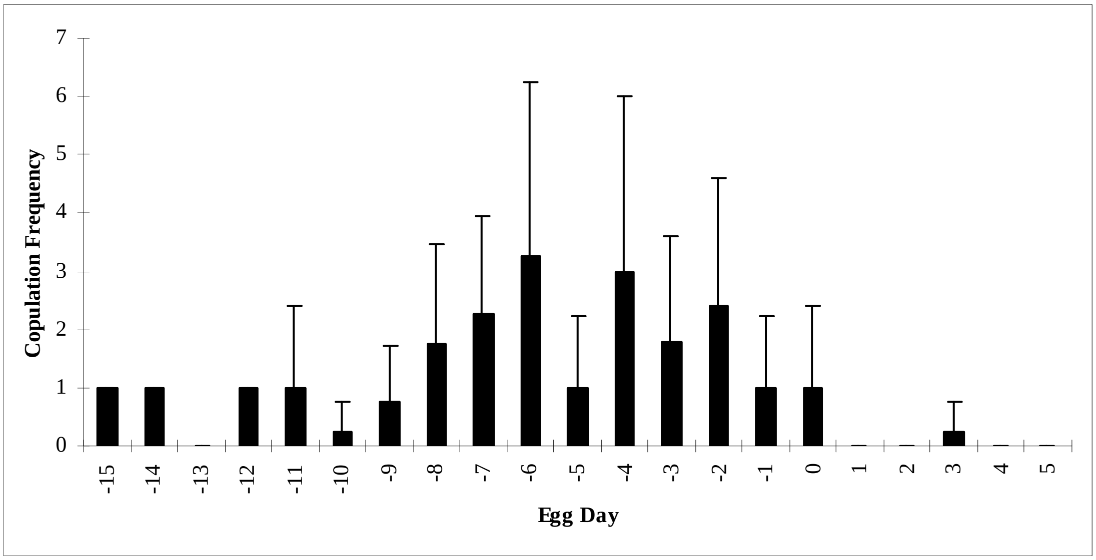
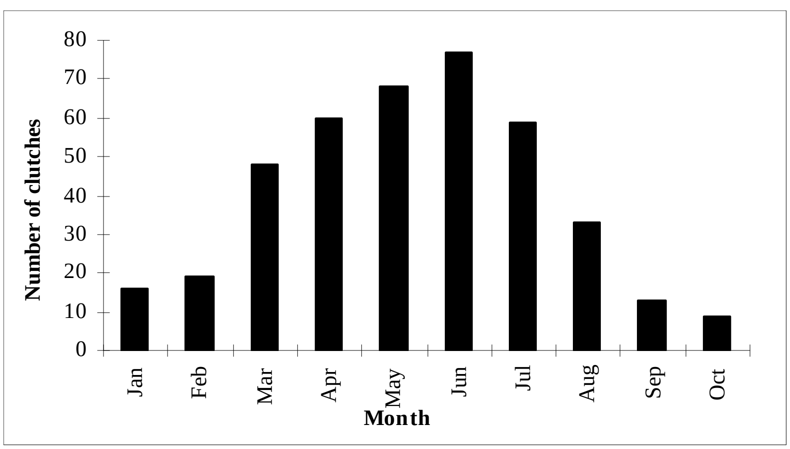


Figure 3. The distribution of egg laying throughout the year.



CHAPTER 7

CARE AND DEVELOPMENT OF OFFSPRING

Parental Care

In the wild, both sexes are known to brood and feed the young (Baker 1951, Jenkins 1983, S.D. Marshall personal communication) and adults have been observed to feed fledglings (Kibler 1950). In captivity, pairs have not been very successful at fledging offspring from the nest. As of 31 December 1995, 69% of parent-reared chicks have been lost in the nest. Of those chicks that are lost prior to fledging, 75% disappear from the nest and the cause of death is unknown although cannibalism has been suspected, 90% are lost within the first 10 days. Hand-rearing has been more successful with only 19% lost before 30 days of age; 76% of those are lost within the first 10 days. As a result, hand-rearing is recommended unless otherwise noted by the Species Coordinator.

The cause of this failure to rear their own offspring is not known. Pairs that have successfully reared their own offspring to fledging have done so only sporadically and there is no predictable pattern to their success. Parent-rearing success does not appear to be correlated with parent origin or experience. When parent-rearing is attempted, it is important to gather as much information on the process as possible. At this time, the benefit gained from the information collected far outweighs concerns about the potential costs of disturbing the pair, although eggs should not be candled within the first week after laying (see Chap. 6, Incubation section).

In preparation for parent-rearing, the parents should receive an increase in the amount and frequency of food beginning several days prior to expected hatch. It is important to provide small pieces of food that a parent can bring to the nest and provision of cut up pinkies and green anoles is recommended. Shelton (1986) suggested that parents regurgitate to their young in the first 5 days, but this has not been confirmed, although Marshall reports a male making “regurgitation movements” at the entrance to a nest containing a day old chick (personal communication). Careful measurement of food consumption by parents during the first critical week when most chicks are lost, may provide information necessary to determine what parents may be feeding their offspring at this time. Supplemental feeding of chicks in the nest has been used with varying degrees of success.

Age of Dispersal/Removal of Young

During the breeding season, pairs may re-nest quickly after fledging a chick (within 2 weeks) and the presence of fledglings may disrupt this process and aggression could arise. Several institutions have seen aggressive behavior from the parents toward a fledged chick when they were ready to re-nest. In one case, a male chick had to be removed from the breeding enclosure as a result of aggression by the male parent. In contrast, a female chick at the Audubon Zoo remained with its parents during renesting and did not receive any aggression from the pair; however, the pair did lose their nestling.

In light of these experiences, it is recommended that all parent-reared chicks be removed from the enclosure in which they are reared within two weeks of the time they fledge. In the interim period the chick(s) should be monitored closely for signs of parental aggression.

Artificial Incubation

Parent incubation has generally been successful. The recommended protocol for pulling eggs for hatching and hand-rearing is to remove the eggs approximately 20 days after laying. Artificial incubation techniques have not been consistently successful; 69% of all embryos that died in the shell were artificially incubated although only 32% of fertile eggs have been artificially incubated. Therefore, artificial incubation should only be used when necessary due to parental neglect, a tendency for a pair to discard fertile eggs or for other reasons as recommended by the Species Coordinator.

Table 4 lists the incubation parameters (dry bulb/wet bulb, degrees F) used by 5 facilities, typical weight loss (if available), and number of egg turnings per day. Each resulted in successful hatchings although each institution also experienced dead-in-shell embryos while using these artificial incubation techniques.

Table 4. Incubation parameters used at selected institutions.

Institution	Temp. (^{o}F)	% Weight Loss	# Egg Turnings/Day
Houston	100/84-86	14-16% loss	12 (auto)
San Diego	99.5/86.0	13% loss (ave.)	5 or 12 (auto)
St. Louis	99.5/85		5 or 12 (hand & auto)
Cincinnati	99.5/86		4 (auto)
Audubon	99.5/87	9-10% loss	4 (hand)

More information is needed to determine which of these incubation regimes is most successful. In order to gather this information it is important to provide complete information regarding the incubation parameters on the clutch/egg report for all artificially incubated eggs. Since parents have been successful at incubating their own eggs, artificial incubation should only be used when absolutely necessary.

Micronesian Kingfisher Hand-Rearing Protocol

The following summary is adapted from the protocols developed by Healy (in Bahner 1990), Sheppard (1986) and the San Diego Zoo (Appendix E). This latter protocol describes changes in diet for each developmental stage and provides a description of a diverse diet recommended for Micronesian kingfisher chicks. The San Diego Zoo uses a Micronesian kingfisher hand puppet for hand-rearing and chicks are isolated from all human contact once eye slits open. Not all institutions take steps to minimize contact with humans and it is not clear at this time what effect, if any, this has on chick development. Because we do not know the effect(s) of hand-rearing, it is important to carefully record, on the Clutch/Egg Report and Specimen Report, the method of hand-rearing and whether or not (to what extent) chicks are exposed to people. Because Micronesian kingfishers are cavity nesters, it may be beneficial to maintain dim or subdued lighting while hand-rearing chicks.

In accordance with the 1993 Masterplan, it is recommended that all kingfishers be hand-reared using the procedure outlined for pulling eggs just prior to pipping based on an incubation period of 22-23 days unless otherwise instructed by the Species Coordinator. The necessity of hand-rearing outlined in the 1989 Studbook is still relevant as the captive pairs have not improved their parent-rearing ability (see Parental Care section, this chap.).

Eggs should be candled to determine fertility at least 1 week after laying of the last egg and removed from the nest approximately 2 days prior to hatching, preferably after the chick has penetrated the air sac. The egg(s) should be placed in the hatcher set at a wet bulb temperature of 88-90^o F (31.1-32.2^o C) and dry bulb of 100^o F (37.8^o C) with a relative humidity of 64%. After hatching, follow the protocol outlined in Appendix E. High humidity appears to be very important to successful hatching. In cases where the egg is removed prior to penetration of the air sac, set the wet bulb at 86-88^o F (30-31.1^o C) for a humidity of 58% and adjust after air sac penetration. The egg should be turned approximately 3 times/day in the hatcher until the air sac is penetrated. Care should be taken to avoid removing the egg more than 4 days prior to hatching. Careful monitoring of a pair's behavior once nest activity begins should provide sufficient information to accurately estimate within 1-2 days of actual egg laying. Chicks generally hatch within 48 hours of pipping although a range of 12 to 71 hours has been reported.

Housing

Because the chicks are naked at hatching, they require a brooder/isolette temperature of 96 degrees F (35.5^o C). Temperature can gradually be decreased as the chick gets larger and the feathers begin to develop (Appendix E). Chicks are typically housed in a bowl (nest cup) lined with a non-slip substrate, such as toilet paper, paper towel or washcloth. These are quickly soiled and should be replaced regularly. Clutch-mates can be housed in the same brooder/isolette but should not be placed in the same nest cup since aggression has been observed between siblings. As the chicks get older and more mobile, birds that are housed within the same brooder should be monitored to assure that no aggressive interactions occur.

When chicks begin perching at approximately 21 days of age, they can be moved to larger quarters. At approximately 30 days of age, chicks will begin to attempt flying but are not yet completely independent and self-feeding. Fledging occurs for parent-reared chicks at 33$\pm$2.74 days after hatching (N=18 parent-reared birds).

Diet

Begin chicks on chopped pinkies (whole pinkies or eviscerated) and chopped anoles. To minimize problems with dehydration, pinkies are dipped in dextrose or Pedialyte. The typical signs of dehydration are dry, baggy skin and dry fecals. A well-hydrated chick is pink with the skin fitting well on the body. Calcium carbonate and vitamin supplements can also be added to the pinkie diet.

Additional diet items are added as the chicks grow, beginning as early as one week of age at some facilities (Appendix E). Additional food items can include cricket abdomens, chopped whole crickets, mealworms, and waxworms. As the chick grows, diet items can also increase in size and whole insects can be fed. Analysis of pellets cast by young chicks revealed that they do not digest many of the components of soft food (such as BOP; Sheppard 1986) Additional information on nutritional needs of growing chicks is found in the nutrition section.

Feeding

Chicks should be weighed in the morning before the first feeding. Normal hatch weight is in the range of 4.4 to 7.1 grams. Chicks should be fed 6-8 times per day initially, beginning first thing in the morning and continuing approximately every 1 1/2 to 2 hours throughout the day. After 7-10 days, additional feedings are dropped until the chick reaches independence (Appendix E). The frequency of feedings should be adjusted to assure that the chick follows the expected weight curve (Figure 4).

For young chicks, small food items are offered by touching them to the side of the chick's beak. They quickly learn to respond to the touch and gape or lunge for the food item. Blunt-ended toothpicks work well to feed young chicks until tweezers or forceps become practical.

Chicks begin to refuse food after reaching a weight of 60 to 70 grams (20-25 days). Force feeding is not necessary as long as the chicks remain on the typical weight curve (Figure 4). Hand feedings can be reduced to 3-4 per day and food items can be left with the chick between feedings. Chicks typically decrease in weight at this time down to as little as 50 to 55 grams. By day 26, the chick can often be encouraged to pick up food on its own by moving it with forceps. Birds are capable of self-feeding at 32-35 days and, generally, they can be weaned by 40-55 days of age. Chicks should be moved within sight and hearing of conspecifics as soon as possible after weaning. This may be particularly important for chicks that were reared in isolation from other Micronesian kingfishers. It should be noted that chicks may continue to accept food after they are capable of self-feeding which may result in hand-feeding long after it is necessary.

Chicks will generally defecate immediately after or between each feeding (note: they do not produce fecal sacs). If defecation seems to be a problem, the chick can be stimulated with a moist cotton swab. Chicks will often refuse food prior to casting a pellet. Pellets may or may not be produced depending on the type of diet but have been observed as early as Day 2.

Molt

The first molt typically occurs at approximately 120 days of age. In the wild, there was an apparent peak in molting during July, August and September for adults. Molting was noted to occur throughout the year at irregular intervals (Baker 1951).

Estimated Time Of Developmental Markers

Day	Developmental marker
1	Blind and naked
2	Casts have been produced as early as 2 days but, depends on the type of food offered
5	Flight feather tracts visible on wings
6	Bill mostly black
7	Feather tracts visible on back, sides and head
10	Eyes begin to open, bill all black
13	Feathers begin breaking through skin
19	Breast feathers breaking out of sheaths
20	Skin is completely covered by pin feathers
23	Back and wing feathers breaking out of sheaths; flight, head, and neck feathers still sheathed
27	All feathers out of sheaths except for tail and a few feathers around the eyes
29	Perching
27-30	Fully feathered
32-35	Fledging

Figure 4. A typical weight curve for hand-reared Micronesian kingfisher chicks.

CHAPTER 8
MEDICAL MANAGEMENT AND CARE OF MICRONESIAN KINGFISHERS

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While comparatively little is known regarding the incidence and significance of disease syndromes in Micronesian kingfishers, several specific disease entities have been identified in captive birds. The small number of birds and the relatively short time that they have been held in captivity have resulted in a paucity of medical events from which to draw conclusions and base recommendations. Because no wild population is available for comparison few opportunities exist to determine if diseases or medical conditions are natural or an artifact of captivity, or if conditions of captive management are contributing to or affecting the incidence of significance of syndromes.

The issues addressed here are either specific medical problems identified in the captive population, specific medical problems possibly present in the population, i.e., poorly documented, or general medical concerns common to similar avian species that are likely to be encountered. The information in this section will hopefully be expanded regularly as more information is accumulated, and as thorough medical care more completely defines current problems.

Infectious Diseases

Few specific infectious diseases are prevalent in the information available for Micronesian kingfishers. A brief survey of necropsy records attribute death to infectious disease in 37% of deaths, however most (1/3) were neonates in which septicemia and infection was probably secondary to or related to being otherwise compromised. Avian tuberculosis has been identified and has been a significant cause of mortality (9 of 40 adult deaths). Of equal or greater significance is the potential for spread and the impact this has on quarantine and movement of birds.

Avian Tuberculosis

Disease Characteristics

Avian tuberculosis (ATB) is a contagious disease caused by *Mycobacterium avium*. The organism is most commonly shed in the feces of infected birds, and may survive in the soil for years (over 4 years in one report). Birds are infected by ingesting the organism. Although it is possible that the organism is passed in eggs, chicks from these infected eggs die before hatching. In the unlikely event that a chick would hatch harboring the infection, it is presumed that the disease would progress rapidly in such a weak host, and the chick would die before maturity. It is not known precisely how long it takes for an infection to progress from exposure to death. Certainly not every bird exposed develops an active infection and there are known species differences. The size of the inoculating dose, the bird's health, and husbandry conditions all have an effect. Several estimates put the incubation period at several months, but little is known conclusively about the development of the disease. Experience with Micronesian kingfishers suggests incubation may be much longer and intervals from exposure to clinical disease may be up to 2 years. Clinical signs of illness usually do not occur until the infection has become well-established and is affecting organ function. Clinical signs include lethargy, significant weight loss, decreased egg production, anemia, and loss of feather condition. The mycobacterial organism enters the body through the intestinal tract, and most frequently becomes established in the liver, spleen, intestine, and bone. At death, gross findings often include markedly enlarged liver and spleen, due to the presence of sheets of bacteria-laden cells. Thickening may also be present in the intestinal tract, and nodules may be seen in the bone marrow as well. In some cases, there are no grossly apparent changes, and the diagnosis must be made on the basis of microscopic changes.

Diagnosis

Premortem diagnosis is difficult, due to the slowly progressive nature of the disease and the lack of consistent signs. At necropsy, characteristic staining properties of the organism, along with typical tissue reactions are used to make the diagnosis. As the disease progresses, hepatomegaly and splenomegaly (enlargement of the liver and spleen) is often present, and can be detected radio-graphically. Radiographs may also reveal involvement of bone. In advanced cases, the white blood cell count <u>may</u> be markedly elevated and can be detected with a complete blood count (CBC). Acid-fast staining of feces and fecal cultures have been suggested as screening tools, however they are of limited value, as affected birds typically shed the organism intermittently. Therefore, a negative test (not finding the organism) is not conclusive. Also, acid-fast staining organisms other than *M. avium* may be shed in bird feces. Even when present in feces, the organism is difficult to culture, and results may be falsely interpreted as negative. Specialized culture techniques may aid in diagnosis. Radiometric culture and polymerase chain reaction (PCR) identification is currently being done at the University of Wisconsin and shows promise of improving diagnostic capabilities. It should be noted, however, that some conflicting results have occurred. For test availability and submission procedures contact:

Johnes Testing Center
608-265-6463 or 608-263-6920

As the organism very often involves the liver and spleen, laparoscopic examination of internal organs is useful. Acid fast staining of liver biopsies is an excellent way to diagnose ATB.

The liver is usually involved in the disease, and may be involved early in the spread of the disease. Once the disease is established, it is likely to be diffuse enough in the liver that a biopsy sample will include some affected tissue.

Control

As with mammalian tuberculosis, the best control is to eliminate infected birds from the population. This can be done by selective euthanasia of suspected cases, however with a limited population strict quarantine protocols may salvage some birds. **There is no effective treatment for ATB in birds**. Control of spread is aided by the use of tuberculocidal disinfectants, e.g., 1-Stroke Environ[R] or bleach in footbaths and for washing instruments. Footbaths should be used at all entrances to bird enclosures to minimize the risk of spreading the disease. In known or highly suspicious cases, cages should be stripped of all organic material (dirt, bark, limbs, nest logs, etc.) that could harbor the organisms and thoroughly disinfected with a tuberculocidal agent. New, uncontaminated material should be used when the cage is reassembled.

For disease control measures, it is useful to group birds into three categories for considering TB recommendations.

<u>Group 1</u>. The first group consists of clinically healthy birds with no known exposure to avian tuberculosis. This means that within the last 12 months they have not been housed in a facility that had a confirmed case of avian tuberculosis during the time that the bird in question was housed there and extending 3 months after the bird left (to cover the possibility that the bird was exposed to early cases just before leaving). The holding facility should not have had ATB in kingfishers or in other species housed physically close to kingfishers. This should not be difficult to determine as the studbook keeper has records of the kingfisher ATB cases and the holding zoos should be aware of ATB in adjoining exhibits (hopefully institutions are isolating kingfishers from exposure to any known diseases in other bird species). A facility should be defined as a distinct building or group of cages in close proximity, sharing common keepers and services in which disease transmission may be possible. A zoo that displays birds in two physically separate exhibit areas would be considered as two facilities; the likelihood of transmission of disease organisms between the two areas can be effectively controlled by disinfection, use of separate tools, and separation of personnel.

<u>Group 2</u>. The second category, possible exposure, would be birds held in facilities (or recently coming from facilities) with confirmed cases of ATB in kingfishers or other species physically near kingfishers. The birds in this category would be held in these institutions, **but not sharing cages with** known ATB cases. These birds are known to have had the possibility of exposure, yet are not in intimate contact with them, and are therefore less likely to be infected. Birds in this group may be moved between institutions provided full disclosure of the medical status is made to the receiving

institution, and the receiving institution has the ability to provide strict quarantine for the remainder of the 6 month quarantine period.

Group 3. The third category are those of known exposure. These would be birds housed with kingfishers that die of confirmed ATB. This would include birds sharing enclosures with birds at the time of death, as well as birds that have been with a confirmed case in the last 6 months.

Methods of testing and quarantine would vary with the group that the bird has been placed in. Birds in group 1 can be moved without special consideration, following the institution's policy for quarantine procedures. (Both AAZV and AZA have quarantine recommendations available as guidelines. AZA accredited institutions should be using those recommendations as a minimum standard. These are briefly outlined under quarantine recommendations.)

Group 2 birds (possibility of exposure) should be tested immediately upon realization of that status. These birds should have whole body radiographs taken and blood collected for a complete blood count (CBC). As discussed above, these tests are likely to identify active clinical cases of ATB. Birds should remain in the holding institution and be re-tested in 6 months and 1 year. If initial and repeat testing is normal, the birds can be considered clear. During the holding period, these birds may remain with current exhibit mates, but should not be introduced to any new birds.

The final group (known exposure) constitutes the highest risk. These birds should be tested immediately upon recognition of their status. They should have whole body radiographs and CBC as Group 2. In addition, based on the high likelihood of exposure, they should be laparoscoped for examination of internal organs for suggestive changes, and should have liver biopsies collected for histopathology and acid-fast staining. Fecal samples should also be submitted for radiometric culture, if available. The birds should remain isolated for 1 year, then the testing regime repeated. If all tests are normal in both initial and repeat episodes, the bird can be considered free of evidence of active ATB. It is possible that these birds could develop active infections later, however due to the importance of each bird to the survival of the species, the minimal risk of a latent infection can be accepted.

The species coordinator should be contacted for recommendation regarding the disposition of birds that are confirmed to have tuberculosis via acid fast staining of liver biopsies. The rare exception to this would be in situations where eggs or newborn chicks could be pulled for hand-rearing. It is very unlikely that the disease can be transmitted through the egg, so that incubated eggs or chicks pulled soon after hatching should be TB free.

Obviously, when dealing with cases of ATB all species of birds exposed must be considered. In mixed species displays, decisions regarding treatment, quarantine, and disposition of all types of birds will have to be made. In outdoor aviaries, wild birds will also have to be considered as sources of infection, as well as possibly being exposed. The appropriate decisions will vary from species to species; testing protocols proposed here may be useful as general guidelines.

Chlamydiosis

Infection with the bacteria *Chlamydia psittaci* may result in a clinical disease known most properly as chlamydiosis. (When present in psittacine birds, the disease is known as psittacosis, but by strictest definition, this term should not be applied to non-psittacine birds. Chlamydiosis is the more correct, broader term.) The disease has been diagnosed at least twice in Micronesian kingfishers, one conclusively. In both cases, no premortem signs of illness were present, and the diagnosis was made at necropsy. In other species, clinical signs include respiratory and intestinal disease. Signs of dyspnea (labored breathing), coryza (nasal discharge), sinusitis, conjunctivitis, diarrhea, and polyuria (increased urination) have been reported, as well as more general signs of lethargy, anorexia (loss of appetite), and fluffed plumage. The disease has been identified in more than 100 species of birds and can be transmitted between them, as well as to mammals, including man.

Premortem diagnosis may be made by culture and serology. Culture techniques require that feces be collected for several days, then submitted to an appropriate lab. Serological techniques vary, and include compliment fixation (CF), hemagglutination inhibition (HI), enzyme-linked immunosorbent assay (ELISA), and latex agglutination. These tests have varying sensitivity and usefulness, depending on the species involved. As experimental infections and trials are not possible in this species, it is unknown which test is most accurate. The latex agglutination test has been determined to be one of the most accurate and reliable tests in other bird species, and is therefore the recommended test.

Postmortem diagnosis is made by examination of tissues with special stains by a qualified veterinary pathologist. Collecting and submitting necropsy materials in accordance with the Micronesian kingfisher SSP necropsy protocol will assure correct tissues are sampled. Lesions are variable, and may include air sacculitis, pneumonia, pericarditis, enteritis, and hepatitis. The observation of LCL bodies (Levinthal-Coles-Lillie) in the organs or on impression smears is pathognomonic for this disease.

Treatment for other bird species consists of long term tetracycline therapy. The efficacy or safety of this treatment in Micronesian kingfishers is not known. However in confirmed cases of chlamydiosis, attempts at treatment are recommended. Current literature should be consulted for treatment protocols (Ritchie, Harrison and Harrison 1994).

As this diagnosis is often made at postmortem examination, the significance of a confirmed case has more impact on the surviving birds exposed to the known case. In situations of known or presumed cases, other kingfishers and other exposed avian species should be tested by culture and serology (as described above) and decisions on treatment made on individual case basis.

Parasitology

No specific parasites are of special concern for Micronesian kingfishers, however they are likely to be susceptible to a wide variety of common avian parasites. Internal parasites of the gastrointestinal tract include a variety of nematodes (such as ascarids and spirurids) and protozoa (such as coccidia and *Giardia*). Internal parasites of the respiratory tract have been reported in other species, but not in Micronesian kingfishers (to my knowledge). Most nematodes can be effectively treated with oral or injectable ivermectin (0.2 mg/kg, one dose, repeat in 14 days) or oral pyrantel pamoate (4.5 mg/kg, one dose, repeat in 14 days). A variety of other anthelmentics are likely to be safe and effective, and treatment should be at the discretion of the attending veterinarian.

Hemoparasites (parasites present in the circulating blood) have not been reported yet in kingfishers, but occur in many other species. Many hemoparasites are not of clinical significance, however some cause significant disease in certain bird species. As with many other conditions in Micronesian kingfishers, the fact that the condition has not been documented may not indicate that it doesn't occur, simply that it has yet to be found.

External parasites are usually of little significance in captive birds. A variety of lice and mites may be present, and may be transmitted between species kept in close proximity. External parasites are easily eliminated by application of a carbaryl powder and cleaning the environment.

The issue of transmission of parasites from green anoles has been raised. The capture, presentation and consumption of small lizards was part of the normal behavior of wild kingfishers and may be important for courtship, breeding or training young (see Chap. 6). The green anoles commonly available to North American zoos are often wild-caught in the Southern United States. Although transmission of parasites from green anoles is a possibility, this has not been documented for kingfishers. As no problems with the procedure of feeding live/freshly killed green anoles have been encountered, there is currently no medical recommendation against feeding green anoles in this way. Another option that has been used successfully is the freezing of green anoles in water. This has two advantages; this procedure may kill intermediate parasite stages and solves availability and storage problems that may be encountered. It will be of critical importance that veterinarians working with kingfishers be aware of the potential parasite problems of feeding unfrozen green anoles and report any problems to the SSP coordinator or veterinary advisor as soon as possible.

Metabolic diseases

Hemosiderosis/hemochromatosis

Hemochromatosis is defined as the deposition of excessive iron in tissues, especially the liver, resulting in liver disease (cirrhosis) due to organ compromise. This condition must be carefully differentiated from hemosiderosis, which is the accumulation of iron in the liver, without accompanying pathological changes. Deposition of iron in organ tissue is not a problem in itself, unless it becomes excessive. Iron is deposited in tissues in several situations, including genetic disorders of iron metabolism, excessive dietary intake, and excessive red blood cell destruction (iron released from hemoglobin). The significance of iron accumulation (hemosiderosis) is as an indicator of an underlying problem. The accumulation of iron pigment becomes significant when it is excessive, exceeding the ability of the liver to store the surplus. At that stage, tissue damage and destruction begins, potentially resulting in the eventual failure of the organ (hemochromatosis).

It is difficult to determine a state of excessive iron intake in a specific individual. Iron levels in the circulating blood remain constant as long as the liver has the capacity to store the excess. The only conclusive way to determine if excessive iron is being accumulated in the liver is by biopsy and special staining techniques. Even when these procedures indicate iron deposition in the liver, it is difficult to assess the significance, as some degree of iron deposition may be normal.

Hemosiderosis is a relatively common postmortem finding in Micronesian kingfishers. To my knowledge, no cases of hemochromatosis (clinical disease) have been documented. The significance of the hemosiderosis lies in what it indicates. Although a variety of underlying causes have been documented in other species, in kingfishers it is likely to be due to dietary iron intake. Such a phenomenon has been seen in captive birds of paradise and mynahs. Theories have been proposed that the captive diets provide more iron than the wild diets that the birds have evolved with, or that the iron is in a more accessible form, resulting in increased dietary iron intake.

Analysis of the diet suggests that much of the iron in food items (bird of prey diet, pinkies, green anoles) is in the form of heme (in blood cells) and is readily available. Considering the natural food preferences of kingfishers (small prey items), it would be difficult to devise a diet that would be significantly lower in iron.

Obesity/Weight Fluctuations

Problems regarding weight control in kingfishers have been reported and tend to fall into one of two categories, obesity or dramatic weight loss. Specific medical causes have not been found for either condition, and are likely to be due to variation in food consumption. Obesity should be avoided for several reasons. In many species, obesity is associated with poor reproduction. In addition, excessive obesity may lead to hepatic lipidosis (fatty infiltration of the liver) and potential health problems related to liver compromise. Dramatic weight shifts are also potential medical problems. Rapid weight loss in many species may result in mobilization of body fat and protein for energy, and in extreme cases result in secondary health problems. Problems related to weight fluctuations are best controlled by frequent weighing of birds. A minimum schedule of monthly weight recording (though not recommended during the breeding season) will allow management staff to "fine tune" the diet. The addition or removal of dietary components, i.e., waxworms, that affect the overall caloric intake will help regulate the birds weight and avoid major fluctuations.

Routine Medical Procedures

Normal Values

Normal values for blood parameters are not well established for Micronesian kingfishers due in part to the small population size and lack of medical intervention. The values in the following table are extracted from the International Species Inventory System (ISIS) Normal Physiologic Values notebook, and represent the only compilation available. To improve the statistical significance of these values, it is vital that any clinical values collected for kingfishers be reported to ISIS for inclusion in future tabulations.

Blood Collection

Blood samples for diagnostic evaluation may be collected from the jugular vein or the ulnar vein. Either site may be accessed with the bird physically restrained, using a 23 to 25 gauge needle (personal preference for a 25 gauge needle and tuberculin syringe). Sufficient volume may be collected for routine procedures. A rough approximation is that 1% of a bird's weight in grams is the volume of blood that can be collected without undue risk. Based on this approximation, a healthy 65g. kingfisher may have 0.65 ml. of blood collected without undue risk. With special techniques now available at veterinary labs, this volume of blood can be divided in half to provide whole blood for complete blood cell counts and serum for biochemical profiles. This collection volume should be reduced in ill birds, but still can provide sufficient blood for diagnostic evaluation.

Table 5. Micronesian Kingfisher: Normal Physiological Data

Parameter	Units	ISIS mean	S.D.	Number
WBC	10^3/ul	4.657	3.846	7
RBC	10^6/ul	3.183	0.886	7
HGB	gm/dl	14.4	0.0	1
HCT	%	43.3	1.5	7
MACH	pg	28.0	0.0	1
MCHC	g/dl	33.5	0.0	1
MCV	fl*	142.7	27.6	7
Heterophil	10^3/ul	2.029	2.048	7
Lymphocyte	10^3/ul	1.426	0.777	7
Monocyte	10^3/ul	0.594	0.545	7
Eosinophil	10^3/ul	0.424	0.711	7
Basophil	10^3/ul	0.176	0.119	6

Restraint/Anesthesia

Micronesian kingfishers can be physically restrained for most brief procedures, such as physical examination, blood collection, or administration of medications. For prolonged or painful procedures, general anesthesia should be used. Gas anesthesia is the preferred method for anesthetizing small birds. Gas anesthesia is quick, safe, and can be manipulated readily to control depth of anesthesia. Isoflurane has proven to be safe and reliable for inhalation anesthesia, with rapid induction and recovery times. Halothane is also widely used and safe, however induction and recovery are slightly prolonged (compared to isoflurane) and control of anesthetic plane is not as precise. For short procedures, anesthesia may be maintained with a facemask while the bird is manually restrained. For prolonged procedures, the bird should be intubated and gas anesthesia provided via a nonrebreathing circuit. With any delivery system, the patient should be closely monitored to assure a safe procedure.

Injectable anesthetics, i.e., ketamine, should be avoided if gas anesthesia is available. If injectables are the only choice available, it is likely that the drugs will be safe and effective. There is much less precise control of depth of anesthesia than with gas agents, and induction and recovery are more likely to be prolonged and potentially rough.

Sex Determination

ZOOGEN, Inc. has established the technique for determining sex of Micronesian kingfishers via polymerase chain reaction techniques on blood samples (see Chap. 4). This method should be used in cases of undetermined sex. Laparoscopic examination is also a proven method of sex determination, but may present an unwarranted risk of anesthesia.

Shipment/Quarantine Protocols

Normal safety procedures and precautions should be taken when shipping Micronesian kingfishers. Before birds are shipped, they should receive a routine examination, a standard procedure necessary before health certificates are signed. No specific preshipment testing is required but a preshipment weight should be taken (see Chap. 4). In specific instances, tests may be requested by the receiving institution, and should be accommodated if possible. In the case of a zoo with a history of avian tuberculosis, special care must be taken to review the history and likelihood of exposure of the kingfisher to be shipped. Recommendations may be found in the section discussing avian tuberculosis.

Upon receipt of a Micronesian kingfisher, normal quarantine procedures should be followed. Both the American Zoo and Aquarium Association (AZA) and the American Association of Zoo Veterinarians (AAZV) have adopted quarantine recommendations for all animal types. These guidelines should be adhered to, and treated as the minimum standards for quarantine. For non-psittacine birds, the recommendations are as follows:

1. Quarantine of not less than 30 days
2. Three consecutive fecal flotations negative for parasites

In addition, <u>recommended</u> procedures include a complete blood cell count (CBC) and serum biochemical profile.

Reproductive Evaluation

Questions regarding lack of reproduction are often directed to veterinarians. While a variety of evaluations are established for mammals, little similar work has been done for birds. In cases where reproduction is poor (or nonexistent) and suspected to be the result of a medical/ physiological problem as opposed to a behavioral/social one, a complete medical evaluation should be conducted. This should include a physical examination, CBC, serum profile, and fecal flotation. In addition, when indicated, specific serological tests may be required, and radiographic evaluations may be warranted. If internal abnormalities are suspected, laparoscopic examination of the organ systems, especially the gonads, may be useful in identifying sources of abnormal health.

Miscellaneous Conditions

Several potential medical concerns have been noted in reviews of mortality and consultations of cases. These have been isolated or incompletely documented conditions, so the significance is difficult to assess. These conditions are reported here to alert those with kingfishers of their existence.

Several cases of inhalation pneumonia have been reported at one institution. In these cases, noxious fumes were present, resulting in acute death. The sensitivity of birds to toxic fumes is well known, and this indicates kingfishers have similar sensitivities.

Two cases of lymphomatosis (lymphoid leukemia) were diagnosed in 1990. In poultry, lymphomatosis is caused by a virus and is communicable. Unfortunately, no further information is available from these kingfisher cases for investigation.

A final syndrome of interest was myopathy or myocardial degeneration. This was listed as the cause of death in three cases between 1986 and 1991. Although there are many causes of myocardial degeneration, some cases may be caused by nutritional deficiencies (vitamin E and/or selenium). As no further cases have been reported, if this was a nutritional problem, improvements in the diet may have eliminated the problem.

CHAPTER 9

Diets for **MICRONESIAN KINGFISHER** ***Halcyon c. cinnamomina***

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Nutrition Advisors

Micronesian Kingfisher SSP

Due to the rapid decline of the Micronesian kingfisher on Guam, little information on their nutritional needs was gathered prior to the establishment of a captive population. Although it is essential to develop appropriate dietary guidelines which meet the nutritional needs of the Micronesian kingfisher, additional information on their natural feeding behavior can no longer be obtained now that the species is extinct in the wild. Therefore, the dietary guidelines presented here are based on a combination of sources: 1) published accounts of the feeding ecology of this species, 2) food preferences in captivity, 3) availability of food items in captivity and 4) known nutrient requirements of other avian species. Recommendations given here are subject to revision as we learn more about the nutritional needs and food preferences of these birds.

Part I: BACKGROUND

Feeding Ecology Data

Micronesian kingfishers have been observed to feed primarily on grasshoppers, skinks, annelids, insects, hermit crabs, other small crustaceans (Beck and Savidge 1990), and occasionally small mammals and young birds (Shelton 1986). During a 1985 nest survey, Sam Marshall observed that several species of skinks, green anoles and geckos made up the main part of the diet of one pair which was raising chicks, although they also ate a variety of insects (Marshall personal communication with A. Baltz). Wild prey items were not analyzed for nutrient content nor is there information on feeding frequency, feeding preferences or seasonality of food sources. Some information may become available on the feeding ecology of a related kingfisher species if proposed field studies are funded.

Food Preference And Diet Intake

In contrast to the variety of food available in the wild, the variety items offered to animals in captivity are much more limited. Because of this limitation, captive animals cannot be relied upon to make nutritionally optimal choices of food. Among mammals it is often the case that poor nutritional choices are made in captivity (Oftedal and Allen 1996). There is no evidence that birds will behave differently, in this respect, than mammals. Thus, it is important to offer foods, which compliment each other nutritionally and assure an appropriate nutrient intake. Surveys of North American zoos holding Micronesian kingfishers were conducted by the nutrition advisors to the SSP in 1993, 1994 and 1996 to determine the types of food offered, food preferences, food consumption and nutrient content of the diet.

The first two nutrition surveys ('93,'94) demonstrated that many different food items were offered to captive kingfishers (Table 6). The '94 and '96 surveys allowed an evaluation of the intake of each food item offered in the diet (Table 7). In addition, the '94 survey identified nutrients that were potentially inadequate in the kingfisher diet and the '96 survey allowed an accurate quantification of the diet's nutrient density (Appendix K, Table 8)

TABLE 6: Micronesian Kingfisher Nutrition Survey results 1993, 1994. Participation, birds represented and number of items offered.

Year	Number of Institutions Surveyed	Number of Institutions Responding	Number of Birds Represented	Total Population	Number of Different Items Offered
1993	19	17	40	56	16
1994	14	13	42	51	12
1996	6	6	18	52	10

TABLE 7: Micronesian kingfisher Nutrition Survey 1994, 1996 significant intake results

Items offered	1994		1996	
	Intake rating	Offered by (# inst)	Intake rating	Offered by (# inst)
Mice (pink or fuzzy)	Good (>80% of offered)	13	Good (76.7 – 100 % of offered)	5
Insects (mealworms, crickets, waxworms)	Good (>80% of offered)	13	Good (>80% of offered)	6
Green anoles	Excellent (100%)	2	Excellent (100% of offered)	4
Complete foods (Dog food, Bird of Prey)	Poor (<40% of offered)	5	Poor (<40% of offered)	4
Vitamin and Mineral supplements	Unquantifiable	NA	Not surveyed	NA

Birds housed singly consumed about 12 g of food per bird/per day while pairs consumed between 20 and 38 g per day. Of major importance is the frequency of dietary items consumed by the kingfishers between the 1994 and 1996 surveys, indicating that the birds have a strong preference for whole food items (e.g., anoles, mice, insects, etc.) compared to the prepared commercial diets that were offered (e.g., dry dog food, Bird of Prey). The nutrient density of whole prey is highly variable (Appendix K), therefore, the nutrition advisors are very concerned about the actual nutrient intake of the Micronesian kingfisher.

A computer analysis of the 1994 survey intake results, using common data base values, showed that some of the diet items were inadequate in nutrients. Diets appeared high in energy, fat, protein, vitamin A and iron but low in vitamin E with some potential problems with the calcium to phosphorous ratio.

In order to more accurately determine the nutrient intake of the kingfishers, a more in-depth survey and intake study was performed in 1996. Food item consumption data were collected from six institutions. Each institution provided detailed diet consumption data for the birds in the institution's collection. The data collected included food items offered, remaining and consumed for 10 days quantified by weight. Each institution also submitted an appropriate number of food samples for chemical evaluation. Chemical evaluation was performed to ensure valid data existed for dry matter, protein, fat, energy, vitamins A, D, and E, carotenoids and minerals. Results from the analyses were entered in a nutritional diet assessment database and used to compute the nutrient content of the diet consumed (Table 8).

All Micronesian kingfishers received either fuzzy or pink mice. These comprised the largest contribution of total quantity in the diet with insects, as a group, following. The green anoles (*Anolis carolinensis*) that were offered were consumed at nearly 100%, suggesting that if they were offered at higher levels in the diet, the contribution to the diet may have been higher as well (Table 7). As expected, nutrient content of these items proved somewhat variable (Appendix K).

TABLE 8: Average dry matter, protein and kcal density of the diet consumed by Micronesian kingfishers (1996 survey results, computer analysis N2 computing).

Nutrient	**Range**
% Dry Matter	24 – 33%
% Protein (DM)	55 - 56%
Kcal /g (DM)	5.5 – 6.1

Protein

The protein content of the foods offered to the kingfishers ranged from 30–68% DM (Appendix K). The actual level of protein in the insects, after correction for the nitrogen contained in the chitin of the exoskeleton, will fall by approximately 10% (Allen 1989). Even after the insect portion of the diet is corrected for chitin, the level of protein in the diet will significantly exceed the target level of protein for the diet (Appendix L). This is not unusual in the diets of captive carnivores when the majority or entire diet is composed of foods derived from animal sources. Although we do not have an amino acid profile on the foods used in the kingfisher diet (except cat food), proteins from animal sources with known profiles contain a wide array of essential amino acids. Because the level of protein in the Micronesian kingfisher diet exceeds the requirements of both cats and poultry (NRC 1978, 1994), we believe this diet will meet the kingfisher's protein requirement.

Fats and Fatty Acids

Fat is important in the diet for energy, as a carrier for the fat soluble vitamins (A, D, E and K) and to supply fatty acids. The fatty acids, linoleic acid (18:2, n-6) and α-linolenic acid (18:3, n-3), are essential in the diet of birds because they cannot be produced endogenously. Although deficiencies of these fatty acids are not common, a deficiency of linoleic acid can result in decreased resistance to disease, impaired sperm production, and problems with embryo development. One percent linoleic acid in the diet, however, will alleviate these problems in poultry (NRC 1994). The dietary items used for the kingfisher (Appendix K) appear to contain enough fat for energy to support growth, reproduction and maintenance. The analysis of pink mice shows that, in general, mice are excellent sources of these fatty acids (Crissey et al., submitted).

Vitamins/Minerals

While each nutrient is important to the health and well-being of the animal, in this section we consider only those which appear to be related to the problems observed in the captive Micronesian kingfisher population as reported by the SSP participants.

Vitamin A

Vitamin A deficiencies in domestic poultry may lead to decreased growth, loss of appetite, weakness, staggering and ruffled feathers. In addition, susceptibility to infections may increase while egg production and hatchability can be reduced. Abnormal eye exudate and drying of the eyes have also been found (Machlin, 1984). Maximum tolerances for vitamin A in poultry are 10 to 30 times the requirement (NRC, 1994). Since Micronesian kingfishers naturally consume animal matter which may contain high levels of vitamin A, their tolerances may be slightly higher than that of poultry, although actual tolerance levels are unknown. Vitamin A levels in pink mice and green anoles exceeded domestic poultry requirements for growth by 9-18 times and 6-11 times, respectively, whereas insects provide less than 73% of the vitamin A level required by poultry (see Appendix K).

Vitamin E

The effects of Vitamin E can be related to the levels of other nutrients in the diet. When a diet high in polyunsaturated fatty acids is deficient in vitamin E, encephalomalacia (hemorrhage and necrosis of the cerebellum) occurs in domestic poultry. When selenium and vitamin E are both deficient, exudative diathesis (capillary permeability resulting in subcutaneous edema) occurs. Degeneration of the testis epithelium has also been reported as a symptom of vitamin E deficiency in chickens (Machlin 1984).

Vitamin E tolerance is 100 times the requirement for domestic poultry (NRC 1994). It should be noted that high dietary concentrations of vitamin A have been shown to depress vitamin E status in a number of mammals as well as chickens (Frigg & Broz 1984) indicating that nutrient interactions may significantly influence nutritional status. The mechanisms, while unknown, may be related to the fact that the antioxidant properties of vitamin E protect vitamin A from oxidation. Extrapolating to species other than domestic poultry may be difficult since there are differences in the response to vitamin E deficiency even among domestic poultry species (Machlin 1984). Vitamin E appeared moderate to low in adult mice, pink mice and insects and almost undetectable to moderate in green anoles (Appendix K). Green anoles stored for >6 months by the standard method (Appendix M) may lead to some degradation of Vitamin E although this was not tested directly.

Carotenoids

Although it was once thought that carotenoids served only as a precursor to vitamin A, more recent studies suggest that carotenoids, particularly β-carotene, may influence immunity and reproduction (Chew 1987, Simpson and Chichester 1981). Carotenoids also play a well-known role in bird pigmentation (Brush 1981). In domestic poultry, lutein, zeaxanthin and some β-carotene contribute to the yellow color of skin and egg yolk (NRC 1994). In the laying hen, 50% of the body's zeaxanthin is found in the ovary (NRC 1994). In the diet of the Micronesian kingfisher, carotenoids including Lutein + zeaxanthin, β-cryptoxanthin, and β-carotene, were much greater in green anoles compared to the other food items (Appendix K). Carotenoids were undetectable in pink mice. It should be noted that carotenoids are naturally present in some crustaceans (Simpson et al. 1981) and possibly other lizards. Although carotenoid levels in diets consumed in the wild remain unknown, these types of prey were reportedly consumed by wild kingfishers (Beck and Savidge 1990). While the role of carotenoids in the diet of the Micronesian kingfisher is unknown at this time, the implications cannot be overlooked.

Vitamin D, Calcium and Phosphorus

Vitamin D, calcium (Ca) and phosphorus (P) are nutrients with inter-related functions (Machlin 1984). In general, the function of vitamin D is to elevate plasma calcium and phosphorus to a level that will support normal mineralization of bone as well as other body functions (McDowell 1989). The deficiency symptoms of Ca and vitamin D for domestic poultry include: thin-shelled eggs, lowered egg production, decreased hatchability, and bone deformities. It is believed that birds better utilize vitamin D in the form of vitamin D3 (cholecalciferol; NRC 1994) which primarily originates from animal sources. Maximum tolerance for vitamin D in poultry is 4-10 times the requirement (NRC 1994), with excessive levels of vitamin D and Ca can causing calcification of soft tissues at end stages.

Insects appeared low in calcium regardless of reported insect supplementation (Appendix K). The Ca content for vertebrates appeared adequate, however, an inverse ratio of Ca:P was found in most of the samples analyzed as part of the 1996 survey. These Ca data appeared similar to the published values for the calcium content of feeder rodents but the levels of P were not reported (Dierenfeld *et al.* 1995). The ratio of Ca:P ideally should be 1:1 for adult animals at maintenance. The mean inverse Ca:P ratios of vertebrates were unexpected. This highlights the fact that these food items are biological and fluctuation is common. It is possible that other samples would show ratios consistently nearer 1:1 or above. Regardless, these values point out the importance of analyzing nutrients in the diet and formulating diets based on real analyses. These data may indicate a relationship between the soft-shelled eggs reported in the population and dietary Ca. Large amounts of phosphorus in relation to calcium have been reported to cause bone loss in some animal studies (LaFamme *et al.* 1972, Krook 1968). While the actual Ca content of the vertebrates and manufactured diets was at least adequate compared to domestic poultry requirements (with one analysis for green anoles being very high) if the Ca:P ratio is not at least 1:1, Ca metabolism problems may occur. This would especially hold true for females with multiple clutches or thin shelled eggs.

Vitamin D analysis showed only trace levels (<1 IU/g dry matter basis) in all food items except large mealworms and green anoles which were considerably higher. However, the minimum detectable level for the analysis is higher than the target nutrient levels. Therefore, it is not known whether all food items are adequate in vitamin D. What is evident is that green anoles contain adequate and even high vitamin D. The levels reported for mighty mealworms are based on just one sample so, while high, this one data point is not strong. It must be remembered that the chemical analysis for vitamin D is difficult and results can be variable.

Iron

Iron deficiency causes anemia as well as reduced hatchability of eggs. Birds lose iron through egg and some feather production, therefore, adequate levels of iron are important to maintain in egg-laying birds. The maximum tolerable levels of iron for domestic poultry are 1000 mg/kg (NRC 1980). There have been reports of excessive iron storage in the livers of some Micronesian kingfishers (Chapter 8). The analyzed values for Micronesian kingfisher foods average 150 mg/kg DM (with some meat-based bird diet samples being higher), which should meet requirements without being excessive. Water is another potential source of iron and this should be quantified if iron accumulation continues to be a problem.

Part II: **DIET RECOMMENDATIONS**

Diet Formulation

It is possible to formulate appropriate diets for Micronesian kingfishers using the National Research Council (NRC) guidelines along with historical reports on natural Micronesian kingfisher feeding and nutrient content of food items available in zoos. A range of values for nutrient levels are provided based on NRC requirements for both domestic fowl (NRC 1994) and carnivorous mammals (NRC 1978, NRC 1982) given that these are carnivorous/ insectivorous birds (Appendix L).

Because of individual preferences, weight, exercise, physical condition, environment, and psychological/social considerations as well as food availability; some flexibility is needed when formulating diets for Micronesian kingfishers. Therefore, guidelines for nutrient content and food categories are provided that allow flexibility in diet formulation while assuring that an appropriate nutritious diet is consumed. If consumed in its entirety, the recommended diet should contain the nutrients presented in Appendix L. Levels are expressed in quantity per unit of diet, on a dry matter basis. These should be considered target nutrient levels until more specific nutrient levels are established (Table 9 provides appropriate quantities).

Schedule of Feeding

Care must be taken to schedule feeding to synchronize with the animal's habits, not the care-giver's preferred routine. The quantity fed at any feeding time should correspond to the feeding activity. For example, the morning (or activity period) feed should consist of more food than the afternoon (less active period) feed. The current recommendation is to feed birds twice per day (Chapter 5). It may not be appropriate to leave all food items out at night anticipating feeding in the early morning hours, because of spoilage and nutrient losses as well as pest infestation or competition. It would be preferable to provide fresh food in those early morning hours when the birds begin feeding. Water should be provided at all times (Chapter 5).

Recommended Diet

In zoos, the diet items offered are often restricted to items available commercially. The items most commonly offered and accepted are mice of different age/size categories, insects, green anoles and some commercially available manufactured products (Table 7). The specific diet recommendations are based on these items as well as the nutritional analysis of the prey items (Table 9 and Appendix K). These recommendations should support the target nutrient values with respect to protein, fat and energy (Appendix L).

In the past there have been supply problems with green anoles since they are wild-caught. For this reason green anoles are often purchased in bulk when available and there may be potential nutrient degradation problems resulting with long term storage. Concerns have also been raised regarding the potential for parasite transmission from wild caught green anoles. These concerns have been addressed with a storage protocol for green anoles obtained in bulk that will minimize the risk of parasite transferal. Anoles are frozen using a standard method (Appendix M) and to date, there have been no reports of parasite transmission.

Table 9 Recommended Diet for Micronesian Kingfishers

Food item	**% (as fed) by weight**	**range to feed (in grams) per bird**	**per pair***	**Expected consumption (single birds)****
Pink mice	25-50%	3-6	10-20	77-100%
Green anoles[1]	25-50%	3-6	10-20	100%
Insects[2]	20-30%	2.4-3.6	8-12	>80%
Dry cat food[3]	0-20%	0-2.4	0-8	na[5]
Supplements	see below[4]			
Total Diet		**12 g**	**40 g**	

*For pairs with chicks a portion of the offered pink mice and green anoles should be chopped and total diet amount should be increased by 25% per chick in nest. Care should be taken to keep the ratio of each dietary item offered consistent with the table.

**Actual consumption may differ depending on caloric density of the diet and energy needs of the individual. Foods selected can be based on availability and seasonality.

[1] During courtship, incubation and chick rearing feed green anoles at upper level (50%). The combined quantity of pink mice and green anoles should not exceed 75% of the diet.

[2]Insects (crickets, mealworms, mighty mealworms or waxworms) should be fed a commercial cricket diet which contains at least 8% Ca to improve the Ca:P ratio. Care should be taken to keep crickets at 80 ^{o} F in order to encourage adequate diet consumption (Ward and Crissey 1997). Insects should comprise at least 20% of the diet.

[3]Cat food should be offered, especially during chick rearing and fledging (see text) . It can be soaked or presented in such a way as to encourage consumption. The type of cat food should contain a nutritional content of 30% protein, 8% fat (see Appendix K).

[4]Calcium: For birds consuming 12 grams of a combination of mice, crickets and green anoles; supplement crickets with 0.36 to 0.6 grams of an insect supplement which contains 8% Ca.
Vitamin E: 0.6 mg of vitamin E per day.

[5]Consumption of manufactured diets must be monitored.

Prey Items (mice, lizards and insects)

The intake data collected on this species strongly suggests that these birds will readily accept almost any whole prey item offered. Since prey items are biological organisms, there will be fluctuations in nutrient content between individual items. It appears to be possible to alter the nutrient content of some prey items by manipulating what is fed (e.g., insects, Allen 1989). At this time we are not able to correct all nutrient imbalances of prey through their diets alone. For this reason and because reproductive problems as well as egg quality problems have been recorded for this species, we recommend the inclusion of a manufactured diet and nutrient supplementation of the basic whole prey diet (see below and Table 9).

Not addressed in detail in this chapter is the behavioral component of the diet. It has been suggested that courtship behavior may be stimulated by the presence of whole prey items (Chapter 6). If this proves to be true, seasonal variation in diet composition may be appropriate. Live prey items (e.g., crickets) may also stimulate natural feeding behaviors. Because the goal of captive breeding is the eventual reintroduction of this species to their natural habitat, efforts to stimulate natural feeding behavior are important to consider along with the nutritional aspects of the diet.

Manufactured Diets

A commercial diet which is readily accepted by adult kingfishers has not been identified. If birds are exposed to a manufactured diet at a very early age, however, they may be more inclined to consume these items. Offering items such as moistened dry cat food or a nutritionally consistent meat-based bird diet is recommended at a 20% level, as fed. Please note that the meat-based bird diet analyzed in the study was not nutritionally consistent, particularly for vitamin A (which can be toxic) as well as fat and energy. In fact, fat content was double in some samples compared with others of the same product. These fluctuations were even more dramatic in some cases than was found in the whole prey items. Thus the nutrient consistency of any meat-based bird diet should be monitored. Always compare the label of commercial diets being considered to the nutrient specifications recommended in Appendix L. Care should be taken to monitor consumption of all manufactured diet items and not to assume that the diet is nutritionally complete simply because an acceptable diet is offered.

As development of a more acceptable manufactured diet continues, this may play an increasingly important role in the total diet. In an attempt to develop an acceptable manufactured diet, a 15-week pilot study (18 February - 29 May 1997) was conducted using 1.1 Micronesian kingfishers at the Philadelphia Zoo. The goal of the study was to determine the acceptability of an artificial gelatin diet by the birds and the feasibility of feeding this diet in combination with whole prey items. The results of this study offer some promise that a manufactured diet can be successfully fed to Micronesian kingfishers and therefore allow better control over the supplementation of nutrients deficient or lacking in the whole prey items.

Supplements

Because of potential nutritional inadequacies in the whole prey diet and the marginal acceptance of nutritionally complete commercial diets, supplementation is advisable. The current recommendation is supplementation of Ca and Vitamin E.

Calcium

Calcium supplementation by feeding insects (both mealworms and crickets) a commercial insect diet that contains at least 8% Ca on a dry matter basis, is recommended. Several appropriate products are currently marketed; read the labels to ensure that the correct product is chosen. Feed the diet for 3 days while the insects are held at 80^{o}F and supply water as a source of moisture. Please note that insects kept on an 8% Ca diet will die after approximately 5 days due to accumulation of feed in the gastrointestinal tract, i.e., calcification of the insect. The insects can also be dusted with this same diet to increase Ca levels even more although the amount ingested by the bird will be highly variable with this method and dusting should not be relied upon as the only source of calcium supplementation. In addition to ensuring adequate Ca intake, supplementation will help to keep the Ca:P ratio of the total diet between the target of 1:1 and 2:1.

Use of the recommended diet (Table 9) will assure that proper nutrient levels and ratios are maintained. When there are deviations from the recommended diet items, care must be taken to ensure proper nutrient levels. Adequate Ca intake can be maintained by determining the Ca and P content of the prey items fed and calculating the quantity of insect diet needed to provide at least 0.7 % but not over 2.5% Ca dry matter in the diet. If needed, the other food items can be dusted using this same insect diet. Please note that the use of supplements must be controlled because too little or too much can cause problems. To determine how much supplemental insect diet to add to the whole diet, the Ca and P contents of all ingredients must be calculated; the nutrition advisors can be contacted for help with these calculations.

Vitamin E

Because vitamin E tolerance is high and the levels present in the recommended diet are low, supplemental vitamin E can be provided at the target levels without adverse effects. Vitamin E is available in both a dry powder form and a liquid oil suspension. The appropriate amount of supplement can either be dusted over the diet (with dry) or injected into a food item (oil suspension). When using the recommended diet (Table 9) a daily supplement of 0.6 mg of vitamin E per 12 gram diet will be sufficient. Since vitamin E is a fat-soluble vitamin which can be stored in the body, it can be supplemented periodically at a more practical level. For example the vitamin E supplementation can be given twice per week at about 2 mg. Care should be taken to determine the potency of the supplement administered to ensure dosage is correct.

Diet Reassessment and Adjustment

Because these diet recommendations are based upon preliminary research, continued reassessment of the adequacy and acceptance of the diet will be necessary. In addition, if one food item is substituted for another or if food intake changes, reassessment of the entire diet may also be necessary. Other factors potentially affecting the diet are individual food preferences and weight fluctuations. Weight problems are not uncommon among captive Micronesian kingfishers; particularly the tendency towards obesity (Chapters 4 and 8). Diets may also be adjusted seasonally; for example green anoles can be increased to 50% of the diet combined with 10% crickets and 40% pink mice, during the breeding season. When green anoles are scarce or it is not breeding season, the proportion of lizards in the diet can be reduced. It is advisable, however, to include green anoles in the diet at least weekly throughout the year because of the carotenoid and vitamin D content.

Any diet changes should begin slowly, starting with a 5% change in the amount of food offered. Increasing or decreasing the calorically dense food can aid in maintaining appropriate body weights. For example, substituting the same quantity by weight of crickets for mealworms will provide the animals with less energy (Appendix K). It is important to monitor weights when any diet alteration is made, although weighing frequency should not exceed once per month (Chapter 4). Please take care not to catch and weigh birds that are reproductively active. Additionally, weight records will provide an indication of possible seasonal weight fluctuations which would be ‘normal’ for a particular bird. Careful record keeping with respect to any diet alterations is crucial for determining the best captive diet for the Micronesian kingfisher.

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