Kiwi first aid and veterinary care

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Cover: Okarito brown kiwi (rowi) having a computer tomography scan to diagnose a fractured acetabulum.
Photo: B.D. Gartrell.

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ABSTRACT

This document provides information about the treatment of sick or injured kiwi (*Apteryx* spp.) for veterinarians, conservation field workers and wildlife park staff. It incorporates basic techniques to stabilise sick or injured kiwi. Specific diseases and common injuries that have been seen in kiwi are also addressed. Diagnostic and treatment techniques specific to each condition and, in some cases, specific to kiwi, are included. The information given is not intended to be a complete reference for the veterinary treatment of kiwi, and is subject to change as more information on diseases or injuries and their treatment becomes available.

Keywords: kiwi, *Apteryx*, first aid, veterinary care, injuries, disease, parasites, New Zealand

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1. Introduction

1.1 BIRDS—THE PRESERVATION REFLEX AND STRESS

In many circumstances, it is immediately obvious when a bird is sick or injured. However, in other instances it is less obvious and disease processes may be challenging to detect.

Birds go to great lengths to hide clinical signs of illness. In the wild, sick birds attract the attention of predators and, in flocking species, a sick bird will be shunned by flock mates (Hume 2000). This masking of signs of illness is known as the 'preservation reflex'. Birds generally do not look sick until they are in an advanced state of illness and near collapse (Cannon 1991).

Identifying that birds are sick when they are in the early stages of disease requires detection of subtle clinical signs. Initial warning signs may include a reduction in food intake and a drop in body weight. A bird with intermittent clinical signs of illness is definitely unwell, and a bird with constant signs is seriously sick.

Stress can play a significant role in the pathogenesis of avian disease. Healthy birds can accommodate some degree of stress, but sick birds are universally intolerant of all types of stress (Cannon 1991). Birds with subclinical problems (i.e. diseases without detectable manifestations) often become sick when stressed. In the wild, stressors include starvation, territorial aggression, and physiological stress such as breeding. In captivity, stress may be induced by malnutrition, poor hygiene and sanitation, poor handling, and overcrowding. Furthermore, it must be remembered that captivity removes the birds' natural escape mechanisms (Cannon 1991). In captive birds, malnutrition caused by an imbalanced diet is one of the most common forms of chronic stress, and this may contribute to disease (see section 7.6).

1.2 LEGAL ASPECTS OF TREATING PROTECTED WILDLIFE

The Department of Conservation (DOC) has a Captive Management Policy (DOC 2003). Veterinarians undertaking wildlife rehabilitation are required under this policy to obtain an authority (permit) from DOC to hold absolutely protected wildlife. Contact your local DOC office for more information. Contact details for DOC offices can be found on the DOC website (www.doc.govt.nz > Publications > About DOC > Role > Policies & plans > Captive Management Policy).

When a kiwi is brought in for treatment, there are two priorities. The first is to administer emergency treatment and pain relief and to assess the bird's injuries/disease status. The second is to inform DOC.

According to the Captive Management Policy, DOC will:

> 'Support the treatment of injured absolutely protected wildlife only in cases where it is likely that the animal will: make a good recovery and be suitable for release into the wild; become part of an approved Species Recovery Programme. If the animal's full recovery is unlikely and it cannot be placed in an authorised programme, treatment should be withheld and the animal euthanised.' (DOC 2003: section 2.2.10, p. 7.)

Consult with DOC regarding the prognosis of treatment and possible options for release of the bird or its use in the Kiwi Recovery Programme.

If further care is required after initial treatment has been completed, the kiwi may only be transferred (after consultation with DOC) to a facility which possesses an authority to hold absolutely protected wildlife. If/when the kiwi is suitable for release into the wild, DOC must be consulted to determine the appropriate location and process for release.

2. Assessment, first aid and stabilisation

2.1 INITIAL ASSESSMENT

Initial assessment of a sick kiwi involves taking a history and performing a distance examination prior to handling the bird. This may give the assessor an indication of the appropriateness of further handling for physical examination and initiation of therapeutics. For example, a bird in respiratory distress may substantially benefit from a period of pre-oxygenation and warmth prior to further handling.

2.1.1 History

Gaining a thorough history is an important part of assessment of the avian patient. The ability to obtain a complete history is highly dependent upon the bird's situation, which will vary from captivity, where birds are visually assessed by their keepers daily, to completely wild, with very little known history.

Important points to note

- What is the age and sex of the bird?
- Is it a wild or captive bird?

Captive birds

- What is the origin of this bird (e.g. captive-raised, wild-caught, transferred from another captive institution)?
- How many birds (in particular, other kiwi) are in contact with this bird?
- How long has the bird been in the particular kiwi house or other captive facility?
- Have any new birds been introduced recently?
- Is the bird in contact with people?
- Is the bird housed indoors or outdoors? Obtain a description of the enclosure, including substrate. Is there any potential for exposure to foreign objects (nails, screws, etc.)?
- Have there been any recent changes in substrate, sources of leaf litter, etc.?
- What is the bird's diet? Have there been any recent changes in diet? What is the water source for the bird?
- How many birds are affected?
- When was the bird noticed to be sick?
- What are the observed abnormalities?
- What is the breeding history and current status of the bird? Is there a possibility the bird is gravid (carrying an egg)?
- Are there any changes in the consistency, frequency or appearance of the bird's droppings?

- Are there any changes in respiration, e.g. open-mouth breathing, wheezing, sneezing, dyspnoea (respiratory exaggeration)?
- Has the bird had any treatment to date, such as fluids, medications (including, but not limited to, antibiotics, antifungals, analgesics, corticosteroids, antiparasitics)?
- Does the bird have any possible access to toxic materials such as rat bait, snail bait, herbicides, toxic plants (e.g. karaka (*Corynocarpus laevigatus*) berries)

Wild birds

- Where was the bird found?
- What are the observed abnormalities?
- When was it found? If there has been a delay between the bird being found and the current presentation, where has it been previously?
- Has it had any treatment (treatment may include fluids and medications including, but not limited to, antibiotics, antifungals, analgesics, corticosteroids, antiparasitics)?
- Has the bird been offered food? Has it eaten?
- Is the breeding history of this bird available?
- Has it passed any droppings? If so how did they look (volume, colour, consistency of faeces, urates and urine)?

2.1.2 The distance examination

A distance examination should ideally be undertaken prior to a bird's capture and restraint, i.e. prior to the induction of further stress. This will allow the handler to get some idea of the severity of the disease, and how well the bird may cope with the stress of further restraint at this stage. However, a distance examination will not always be possible. If the bird is presented in a transport box, observe the bird for a few moments in the box before picking it up.

A normal bird should be alert, standing straight and even on both feet, with barely visible respiratory efforts at rest (Hume 2000).

Points to note include:

Mental status

- Is the bird bright, alert and reactive to stimuli?
- Or is it quiet and dull?
- Or moribund?

Posture

- Is the bird standing?
- If so, is it standing evenly on both legs?
- Is it using its bill for balance?

Gait

- Is the bird ambulating normally?

Respiration

This should be barely visible. Abnormalities include:

- Open-mouth breathing
- Exaggerated respiratory effort
- Respiratory noise or click on inspiration/expiration
- Ocular or nasal discharge

Feathering

- Is there any obvious damaged or misshapen feathering?
- Are feathers fluffed up?

Wounds

- Are there any signs of injury (fresh or dry blood, cuts, open wounds, matted feathers, bald patches, etc.)?

Body symmetry

- Are there any obviously damaged or misshapen parts of the body?

2.1.3 Handling and restraint

Once these visual assessments have been completed, the decision whether to handle and restrain the bird for further diagnostics and supportive therapy should be made. A bird in respiratory distress will invariably benefit from supplemental oxygen, warmth and rest prior to further handling.

In many cases, there will probably be an experienced kiwi handler present. However, if not, the following description of handling kiwi by R. Jakob-Hoff may be useful:

> 'Grasp both legs above the hock joint between the thumb and middle finger of the right hand with the index finger between the two legs. The left hand supports the ventral and lateral body into a sitting position in the crook of the right elbow. Direct the bird's bill and eyes under cover of the left arm.
>
> 'An alternative hold is often used in the field with the holder in a sitting position. Grasp the two legs as previously described, hold the bird in an upside-down position with the dorsal body placed on the lap of holder and the kiwi's head under the left arm.' (Doneley 2006)

Taping the legs of kiwi together during handling should occur only if absolutely necessary, and then just for short periods. Kiwi should never be transported with legs taped. There is evidence that taping legs together causes severe muscle damage, as indicated by marked elevations in creatine kinase in great spotted kiwi (*Apteryx haastii*) that were transported with legs strapped together (unpubl. data). If taping is necessary for weighing or transmitter changes in the field, current recommendations are for periods no longer than 15–20 minutes. However, the impact of leg taping on kiwi physiology requires further study. (B.D. Gartrell, Massey University, pers. comm.).

2.1.4 Physical examination

A systematic and thorough physical examination is important to ensure that subtle abnormalities are not overshadowed by obvious injuries. Doneley (2006) gives the following summary as a guide to examination of ostriches.

A similar approach can be applied to examination of kiwi. The extent to which the described examination can be done depends upon expertise and equipment available.

Head examination

Examine the eyes, ears (caudo-lateral to the lateral canthus of the eye), bill and nostrils (at the tip of the bill), looking for defects and discharges.

Oral cavity

Examine the choana (slit on the roof of the mouth which communicates with the upper respiratory tract), glottis (opening into the trachea), tongue, and mucosa lining the oral cavity.

Body condition

As kiwi are flightless, it is not possible to determine the body condition score by palpation of the pectoral muscles. Palpation of the epaxial muscles (adjacent to the spine) and assessing the prominence of the ribs will give a subjective indication of kiwi body condition.

Heart and respiratory system

Auscultate (preferably with a paediatric stethoscope) for abnormal cardiac and respiratory sounds. The rigid avian lungs lie in the dorsal cranial body wall, and are best listened to over the dorsum, halfway between the vestigial wings. [Although less developed in kiwi than in flighted avian species (B.D. Gartrell, Massey University, pers. comm.), the air sac system extends throughout the entire coelomic cavity.]

Normal kiwi heart rate: 70–240 beats per minute

Normal kiwi respiratory rate:12–60 breathes per minute (Doneley 2006)

Palpate the abdomen

The firm gizzard lies in a caudoventral position, slightly to the left of the midline.

Legs and feet

Observe the legs and feet for gross abnormalities. Limbs should be palpated for obvious fractures and joint effusion (abnormal fluid). In chicks, tibiotarsal alignment should be evaluated for rotational defects (see section 7.7.1).

Skin and feathers

The skin and feathers should be evaluated for external parasites. Many kiwi carry ticks. These are often found in large numbers over the head region, where the bird is unable to preen them off. They are also common in the external ear canal (see section 7.8.3). Kiwi also carry lice, and it has been observed (in birds in general) that debilitated individuals tend to have increased numbers of lice.

The skin should also be evaluated for traumatic injuries and other lesions, including dermatitis, which may be indicative of a vitamin B deficiency (see section 7.6.2).

Vent

The vent should be examined for prolapse, trauma and vent soiling with faeces and urates.

Neurological examination

Compared with other avian species, kiwi show minimal reliance upon vision, as indicated by eye structure, visual field topography and reduced visual centres in the brain (Martin et al. 2007). Functionally blind kiwi have been found in the wild. These birds were surviving well, and for this reason blindness should not be grounds for non-release. Instead of eyesight, kiwi have an increased reliance upon olfactory and tactile information as an adaptation to their nocturnal behaviour (Martin et al. 2007). However, despite the fact that kiwi might not need good vision to survive, cranial nerve evaluation should include assessment of the pupillary light reflex, the menace reflex and the corneal reflex. In performing these examinations it is important to note some differences that are particular to birds. There is no consensual light reflex, and birds are extremely sensitive to movement of air across the surface of their eye, which may confuse results of the menace reflex. Birds also have some voluntary control over their pupil size because of skeletal muscles within the iris. Anisocoria (asymmetrical pupil size) may be indicative of cranial trauma. Other abnormalities may include strabismus (deviation of the eye) and nystagmus (involuntary movement of the eyeballs in unison). Other cranial nerve abnormalities may cause reductions in olfactory ability, torticollis (head tilt), dysphagia (difficulty in swallowing), and alterations in facial sensation.

Segmental reflexes in kiwi include the leg withdrawal and the vent reflex. These reflexes require intact peripheral nerves without the need for an intact spinal cord. Withdrawal of the leg when stimulated (e.g. a toe pinch) indicates an intact peripheral nerve supply to that limb. Perception of pain requires an intact spinal cord in addition to an intact peripheral nervous system. Indications that a bird can perceive a painful stimulus may include an attempt to escape, or the bird looking at the painful area in response to the stimulus.

The vent reflex can be elicited by pinching the skin adjacent to the vent. The vent should normally contract in response to this stimulus.

It should be noted that many reflexes and responses may be confounded by pain, and the mechanical inability to move severely fractured limbs. The neurological examination is most useful for initial assessment, and to evaluate changes in neurological function over time.

2.2 EMERGENCY STABILISATION OF A DEBILITATED KIWI

The priority in the initial 24 hours after presentation of a sick or injured kiwi is to stabilise the patient. Stabilisation involves attempting to regain physiological homeostasis in what are often hypovolaemic, hypothermic, malnourished and septic patients. The extent to which stabilisation can be achieved is dependent upon the available equipment and expertise.

It may be advisable for field staff working with kiwi to carry a first aid kit containing useful items for provision of at least warmth and fluid therapy (Appendix 1).

The following text details the achievable priorities for treating sick or injured kiwi in the field and in the veterinary hospital. It is recognised that there will be many occasions and situations where the procedures that are achievable will fall somewhere between these two extremes.

The goals of treatment in the first 24 hours include rehydration, provision of warmth, stabilisation of fractures, dressing of open wounds, pain relief, and placement of the kiwi in a warm, stress-free environment.

2.2.1 In the field

Field workers should be able to provide the debilitated kiwi with the following:

Fluid therapy (see section 2.2.5)

- Fluids may be administered orally or by subcutaneous injection.

Warmth (see section 2.2.4)

- In-the-field heat therapy may be provided with heat pads or hot water bottles.

Fracture and wound management (see section 2.3)

- Lower limb fractures should be dressed and bandaged in order to prevent further injury to the limb and contamination of wounds, as well as reducing pain associated with the injury.

Quiet, stress-free environment (see section 5.2)

- The bird should be placed in a warm, quiet, dark and stress-free environment if there is a delay in its transportation out of the field.

2.2.2 In the veterinary clinic

In the veterinary clinic, the first priority must be to address life-threatening dyspnoea (breathing difficulties) if present.

Provision of oxygen (see section 2.2.3)

- Dyspnoeic patients will benefit from the provision of supplemental oxygen, either via a mask or in an oxygen tent, prior to further handling.
- An upper airway obstruction requires immediate intervention by placement of an air sac cannula through which oxygen can be supplied.

Fluid therapy (see section 2.2.5)

- Intravenous (IV) access should be prepared primarily for fluid therapy, as well as IV administration of analgesics and anti-infective medication. Oral and subcutaneous fluid therapy may also be used, but these are less than optimal in the compromised patient.
- Intraosseous cannulation provides access for critical fluid and medical therapy in the event that intravenous access is not possible. The suggested site in kiwi is the tibiotarsus via the tibial crest.

Warmth (see section 2.2.4)

- Warming hypothermic kiwi is critical, and can be achieved by a variety of methods.

Analgesia (see section 2.2.7)

- Opioids (butorphanol) may be given for analgesia. Non-steroidal anti-inflammatory drugs (e.g. carprofen, meloxicam) should only be used in well-hydrated patients.

Anti-infective medications (see section 2.2.8)

- Antibiotics should be started immediately if the patient is suspected to be septic, or has injuries that are likely to become infected.

Bandaging and wound management (see section 2.3)

- The goal of initial bandaging and wound management is to prevent further desiccation of the wound, and to provide external stability for lower limb fractures. This is important in preventing further tissue damage and relieving pain associated with unstable fractures.

Initial diagnostics (see sections 4.1 and 4.2)

- Ideally, a blood sample should be taken in the initial stages of treatment to enable investigation of haematological and biochemical parameters before any treatment is given. At the very least, the packed cell volume (PCV) and total plasma protein (TPP) should be assessed.
- A faecal sample should also be examined to investigate the presence of parasites, including coccidia.

Quiet, dark, stress-free environment (see section 5.2.1)

- The bird should be placed in a warm, quiet, dark and stress-free environment, away from foot traffic and other animals.

N.B. Corticosteroids are contraindicated in the stressed or septic avian patient, as they may significantly suppress the hypothalamic pituitary axis, and may result in a severe and lasting immunosuppression.

2.2.3 Oxygen therapy

Stress resulting from handling and other procedures can be fatal in a severely debilitated bird. Clinical signs suggestive that handling for examination is contraindicated until the bird is stabilised include prolonged dyspnoea, and prolonged panting and gasping for air (Harrison et al. 2006). These patients benefit significantly from oxygen therapy prior to handling.

Prior to examination, oxygen may be administered in a chamber where the air comprises 40–50% oxygen. An intensive care unit or incubator can be used or, more simply, a cage covered with a plastic bag (ensure an outflow is present) (Harrison et al. 2006). A facemask fashioned for kiwi (see section 3.2.3) is effective for short-term treatment if an oxygen enclosure is unavailable, but the stress resulting from handling the bird to attach the facemask may outweigh any benefit from the extra oxygen.

Air sac cannulation is well described for other avian species. This procedure is indicated for upper respiratory tract obstructions at the level of the trachea or syrinx (Harrison et al. 2006), providing the bird with the ability to inspire and expire through the caudal air sacs, bypassing the trachea. Air sac cannulation is an emergency procedure, and involves placement of a cannula into the caudal thoracic or abdominal air sacs (Harrison et al. 2006).

However, adaptations for flightlessness have resulted in a reduction in air sac development in kiwi, making this a difficult procedure to perform in these species (B.D. Gartrell, Massey University, pers. comm.). Several attempts at using this procedure with kiwi have been made, with varying success, and its use in kiwi is still at an early stage. For further information about this procedure, contact the New Zealand Wildlife Health Centre, Massey University (see Appendix 2 for contact details).

2.2.4 Warmth

Debilitated birds are unable to thermoregulate as effectively as healthy birds, and are often hypothermic. Provision of warmth is indicated for most sick and injured kiwi. Options for providing heat vary depending on the situation and availability of equipment.

The gold standard for the provision of warmth is an intensive care unit specifically designed for small animal use. Alternatively, a human paediatric incubator works well. Both pieces of equipment allow thermostatic and humidity control of the air within them. Other options include hot water bottles, wheat sacs and heat pads, heat lamps or, simply, a heater in a small room. All approaches have advantages and disadvantages.

Hot water bottles and heat pads are readily accessible and cheap and are ideal for taking into the field. However, care must be taken to ensure that birds do not get burnt from direct contact with them, and it is recommended that heated objects such as hot water bottles be wrapped in a towel before use. A major disadvantage of hot water bottles, heating pads and wheat bags is that they cool down and need to be removed for reheating, which causes disturbance and further stress to the bird.

Heat lamps are a good source of overhead heat. They can be purchased from a pet store or lighting store. Bulbs are either ceramic (without glow) or infrared. Heat lamps should never be left unattended, and care must be taken to not burn the bird, especially with recumbent birds that cannot move away from the heat source.

A heater in a small room may work well also. Ideally, the heater should have a thermostat which can be set to regulate the temperature in the room.

The desirable ambient temperature for most sick birds is 29–30°C, with 70% humidity (Harrison et al. 2006). However, kiwi have inherent lower body temperatures than other avian species, and it may be more appropriate to house them at lower temperatures (e.g. 25–26°C). Birds should be monitored closely for signs of heat stress. Clinical signs of hyperthermia may include open-mouth breathing and the flattening of feathers to the body. Once the bird has reached its normal body temperature (c. 38°C), the ambient temperature should be reduced, as kiwi do not tolerate high ambient temperatures well. If the heat source is not under thermostat control, a maximum-minimum thermometer should be installed in the enclosure to monitor any fluctuations in temperature. If an ICU or incubator is unavailable, placing a small dish of water or a moist heated towel in the bottom of the enclosure will provide some humidity with the warm air (Harrison et al. 2006).

2.2.5 Fluid therapy

It should be assumed that debilitated birds will be hypovolaemic upon presentation. Hypovolaemia refers to a reduction in circulating blood flow, with a resultant reduction in hydration of body tissue. Hypovolaemia may be caused by a combination of blood loss, shock (including septic shock), and severe dehydration due to a prolonged lack of food and water intake or excess loss of body fluids.

Hypovolaemic animals require rigorous fluid therapy. Goals of the fluid therapy plan include:

- Replacement of lost fluids.
- Provision of maintenance fluids to cover normal daily losses, estimated at 50 mL/kg/day.
- Replacement of any ongoing abnormal losses (e.g. diarrhoea, continued bleeding).

Assessment of dehydration and hypovolaemia

The bird's history may assist determination of the likelihood of hypovolaemia. The likely route and duration of any fluid loss should be assessed (e.g. blood loss, diarrhoea), as well as any indications suggestive of septic shock. Fluid and food intake immediately prior to presentation should also be taken into account.

Assessment of hydration status in a bird is subjective and requires practice; however, in general, many workers assume a loss of 10% of total blood volume in a debilitated bird (Monks 1996).

Below is a guide to a more detailed subjective assessment of hypovolaemia in the avian patient (Redig 1984; Abiu-Madi & Kollias 1992; Monks 1996):

< 5% dehydrated	No detectable clinical signs.
5–6% dehydrated	Subtle clinical changes—subtle loss of skin elasticity.
6–8% dehydrated	Moderate dehydration—skin tenting is visible over the dorsal tarsometatarsus (i.e. when the skin is pinched, it takes longer than normal to return to its normal position). Dry mucous membranes (oral, cloacal, conjunctival). Decreased sliding of the skin over the sternum.
10–12% dehydrated	Severe dehydration—all of the above plus skin turgor, sunken eyes, thick and stringy mucous in caudal pharynx, central nervous system depression, lethargy, weakness.
12–15% dehydrated	SHOCK. Death is imminent unless therapy is rapidly established.

Objective assessment of hydration status can be achieved by evaluation of haematological and biochemical parameters. Dehydration may increase the packed cell volume (PCV) by 15–30%, and the total plasma protein (TPP) by 20–40% (Lumeij 1987; Martin & Kollias 1989).

It should be remembered, however, that TPP may be low in a starving bird regardless of hydration status (Kaufman 1992). Also, PCV may be low due to any chronic disease, and this may mask any rise due to dehydration (Martin & Kollias 1989; Hoefer 1992).

Initially, acute blood loss results in no change in PCV and TPP. As the time between haemorrhage and presentation increases, a reduction in PCV and TPP becomes more likely (Forsyth et al. 1999).

Routes of administration of fluids

Regardless of the route of administration, all fluids should be warmed to body temperature (38–39°C) (Quesenberry & Hillyer 1994). Using warm fluids is particularly important with neonates and with intravenous or intraosseous administration of fluids for hypothermia or shock (Abou-Madi & Kollias 1992).

Oral fluids

Oral fluids are indicated for treatment of mild dehydration, or for daily maintenance fluids only (Quesenberry & Hillyer 1994). They are inadequate in birds with sudden or excessive fluid losses. Oral fluids should not be given to birds that are seizuring, laterally recumbent, regurgitating, in shock or have gastrointestinal stasis (Quesenberry & Hillyer 1994). For effective rehydration, oral fluids need to be readministered within 60 to 90 minutes of the first treatment (Quesenberry & Hillyer 1994).

Types of oral fluids include:

1. Oral electrolytes

- Calf electrolytes, e.g. Revive™, Vytrate™, Dexolyte™.
- Human electrolytes, e.g. Gastrolyte™, Powerade™ (though not ideal because of high levels of artificial colours and flavour).
- Bird-specific electrolytes, e.g. Polyaid™ (includes nutritional support).

2. Intravenous solutions which may be given orally

- LRS, 0.9% NaCl, 2.5–5% dextrose.

The relatively small capacity of the kiwi gastrointestinal tract limits the volume of fluids that can be administered orally. The kiwi lacks a distinct crop and has only a small proventriculus (Fergus et al. 1995). This limits the capacity for storage of food and fluids. Administration of excessive fluids may result in regurgitation and possible aspiration into the trachea. The author recommends that no more than 20 mL of oral fluids be given to an adult kiwi at any administration, and no more than 3–4 mL to a kiwi chick. This should be given slowly. To account for individual variation, it may be safer to give a smaller amount than this initially, and work up to this larger volume over subsequent administrations.

Oral fluids are administered into the distal oesophagus using either a metal avian crop tube or a silicone rubber feeding tube attached to a syringe. With the head well restrained and the neck extended, the tube should be passed beside the tongue on either the left or right side of the oral cavity, avoiding the glottis (opening to the trachea) (Fig. 1). The tube is then passed down into the oesophagus which lies on the right side of the neck. The glottis is clearly visible as an opening at the base of the tongue, identifiable by movement of the glottic cartilages as the bird breathes. When the tube is in place, the glottis should be observed to ensure that the apparatus is clear of this structure. Fluid should then be slowly syringed through the tube while the back of the oral cavity is observed for reflux. If reflux occurs, the tube should be removed and the bird's head released to allow it to swallow and shake its head to clear the glottis.

Figure 1. Intubation of a kiwi with a Cole™ endotracheal tube. The stepped portion of the endotracheal tube will be advanced to sit against the glottis (arrowed) to form a seal. Uncuffed endotracheal tubes are used in birds because of the complete cartilaginous rings in their trachea. If cuffed tubes are used, the cuff should never be inflated, as this can cause pressure necrosis and subsequent stricture of the trachea.
Photo: K. Morgan.

Subcutaneous fluids

Subcutaneous fluids should be utilised for mild dehydration and for maintenance fluid therapy only (Quesenberry & Hillyer 1994). This route of administration is ineffective at treating hypovolaemia and moderate to severe dehydration, because associated peripheral vasoconstriction reduces the absorption of fluid in hypothermic or shocked patients (Forsyth et al. 1999).

Types of subcutaneous fluids include:

- Lactated Ringers solution (LRS)
- NaCl 0.9%

Fluids for subcutaneous fluid administration need to be isotonic (Forsyth et al. 1999). Subcutaneous administration of 5% dextrose should be avoided because equilibration of the extracellular fluid with a pool of electrolyte-free solution may result in an aggravation of an electrolyte imbalance (Forsyth et al. 1999). Sterile abscesses and local fluid accumulation may also occur at the site of subcutaneous dextrose administration (Forsyth et al. 1999). Fluids for subcutaneous administration must be sterile to avoid causing infection at the site of injection.

Sites for administration of subcutaneous fluids are the:

- Intrascapular region—this is the skin over the dorsum (back) of the bird, halfway between the vestigial wings.
- Inguinal fold—this is the fold of skin cranial to (in front of) the stifle (knee), between the stifle and the body wall.

Fluids administered at each site should not exceed 5–10 mL/kg bodyweight, with only one puncture hole per administration site (Quesenberry & Hillyer 1994). Use small needles (25–27 gauge); a butterfly catheter may assist administration.

Intraperitoneal fluids

Never administer intraperitoneal fluids to birds. This is likely to result in fluid getting into the air sacs, and there is a risk of organ laceration (Monks 1996).

Intravenous fluids

Intravenous fluid therapy is indicated in cases of severe debilitation or severe hypovolaemia (Harrison et al. 2006). The author recommends intravenous fluid administration in the majority of cases of debilitated kiwi, and for all surgical cases.

Intravenous access also allows the administration of antibiotic and analgesic medication with minimal stress on the bird.

To minimise stress, the author recommends placement of the IV catheter while the bird is under general anaesthesia. In kiwi, intravenous access points are limited to the bilateral medial metatarsal veins, and the right jugular vein as a last resort. The medial metatarsal vein is most accessible proximal to the first phalange (medial) and courses in a caudal direction (Fig. 2). In some cases, the right jugular vein may be useful, but it is difficult to maintain a catheter in this position.

A 20- or 22-gauge catheter is recommended for adult kiwi, and a 24-gauge catheter for chicks. The catheter should be securely fixed with tape, and covered in a self-adherent dressing. Extension sets are useful (Fig. 3).

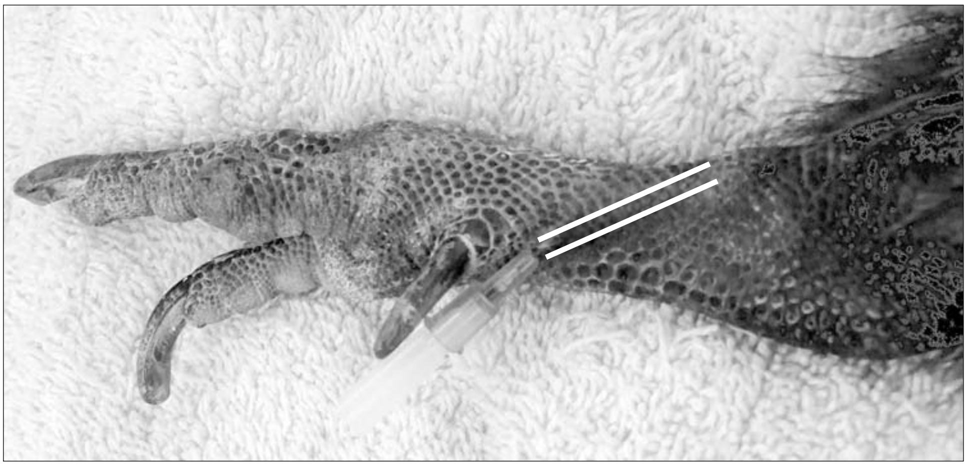

Figure 2. Medial distal limb of a kiwi (with a 22-gauge catheter in place). White lines show position of the medial metatarsal vein. *Photo: K. Morgan.*

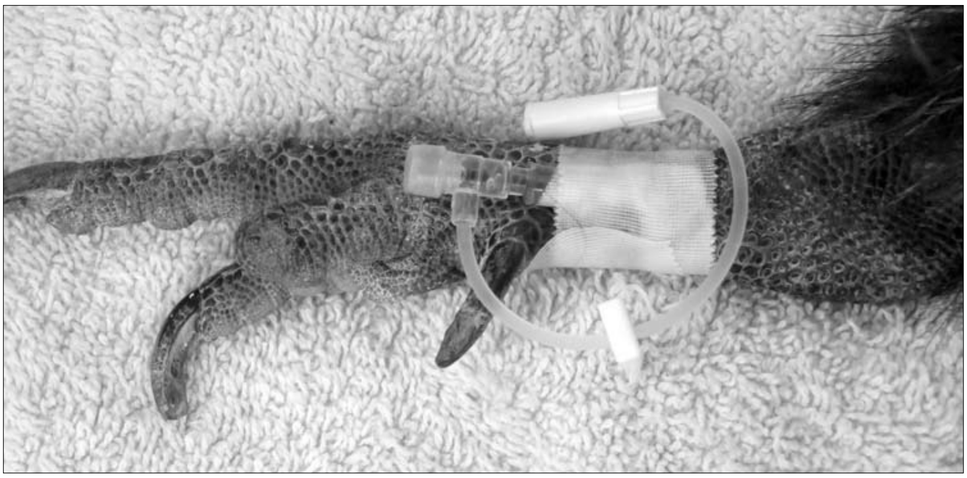

Figure 3. Extension set connected to a 22-gauge catheter in medial metatarsal vein of the right leg of a kiwi. *Photo: K. Morgan.*

Kiwi tolerate intravenous catheters, and they can be maintained for up to 7 days with an aseptic technique. Complications of catheterisation include thrombosis, cellulitis and loss of patency (Monks 1996).

Intravenous fluids can be given either as an intermittent bolus of fluid, or by constant infusion in an IV drip, ideally with a fluid pump.

If a fluid pump is unavailable, it is wise to administer fluids as a bolus to prevent accidental overhydration which may occur with a normal drip set-up. Bolus fluid administrations should be given slowly intravenously, at a rate of 10–25 mL/kg over 5–7 minutes. This should be repeated at 3–4 hourly intervals for the first 12 hours, then every 8 hours for the next 48 hours. Thereafter, IV boluses can be given twice daily for maintenance therapy (Harrison 1986).

Intraosseous fluids

Intraosseous fluid therapy is indicated in severely debilitated birds where it is not possible to gain intravenous access. It allows the parenteral administration of fluids (including blood), nutritional support and glucose, analgesia, antimicrobials and drugs for cardiovascular resuscitation. Hypertonic or alkaline solutions should be avoided, as they are painful on administration (Quesenberry & Hillyer 1994).

Intraosseous catheterisation is a painful procedure, and in most cases should be done under general anaesthesia. Exceptions to this include very moribund birds that are unlikely to survive an anaesthetic.

Because they lack significant wing bones, intraosseous catheter sites in kiwi are limited to the tibial crest (Fig. 4). In birds, the femur should be avoided for intraosseous cannulation as this bone is pneumatic, having communications with the air sac system (Quesenberry & Hillyer 1994).

Figure 4. Intraosseous catheterisation in the left tibial crest of the tibiotarsus of a kiwi, using a 20-g intravenous catheter and butterfly catheter. *Photo: J. Youl.*

In young birds, an 18- to 22-gauge, 1.5- to 2.5-inch spinal needle is ideal for cannulation (Quesenberry & Hillyer 1994). An 18- to 22-gauge hypodermic needle will also suffice. In adult kiwi, a kirschner wire and chuck may be used to drill a hole for cannulation. Aseptic preparation of the site is extremely important to avoid introducing infection to the area. The needle/kirschner wire should be advanced into the bone marrow via the tibial crest, passing distally. Pressure should be applied with a slight rotating motion. Once the cortex is penetrated, the needle/kirschner wire should slide easily, with little resistance. If predrilling the cannulation hole with a kirschner wire, the wire is removed and the needle inserted. Bone marrow occluding the needle lumen should be aspirated and flushed with a small amount of heparinised saline. The catheter should be secured with a light padded bandage. Initial fluids should be given slowly while checking for subcutaneous swelling, which indicates improper catheter placement (Quesenberry & Hillyer 1994).

Intraosseous fluid rates of 10 mL/h have been administered to pigeons using an infusion pump (Lamberski & Daniel 1992). Bolus administration rates are limited, so boluses need to be given frequently.

In pigeons, it has been demonstrated that 50% of fluids given into the ulna enters the systemic circulation within 30 seconds (Lamberski & Daniel 1992). Over a 2-hour period, flow into the systemic circulation approximates the administration rate (Lamberski & Daniel 1992).

Intraosseous therapy is most successful if it is used during the first 24–48 hours of treatment for initial rehydration and shock therapy (Quesenberry & Hillyer 1994). The catheter may be maintained for up to 72 hours without complications if it is placed aseptically and maintained with heparin flush every 6 hours (Otto & Crowe 1992). Clinically, after 2–3 days many birds develop a painful response when fluids are administered through an intraosseous catheter. This may be in response to local oedema or extravasation of fluids around the marrow cavity (Quesenberry & Hillyer 1994).

Rates of fluid administration for dehydration

Replacement fluids

10% dehydration may be assumed in all debilitated birds (Harrison 1986). Fluid deficits in avian species may be calculated using the following formula (Harrison 1986):

Fluid deficit (mL) = estimated dehydration (%) × body weight (g)

Half of the required fluid should be given over the initial 12–24 hours, with the remaining half divided over the following 48 hours.

Maintenance fluids

Maintenance fluids are estimated to be 50 mL/kg/day (Redig 1984). If the bird is not eating or drinking on its own, fluids need to be administered either parenterally or orally to ensure the bird is adequately hydrated. Paediatric birds require 2–3 times the maintenance volume on a weight basis (per kg) compared with adult birds (Bauck & Kupersmith 1991).

Ongoing losses

This includes fluids lost due to diarrhoea, polyuria, etc. If such losses are occurring, the fluid rate should be adjusted to compensate for them.

Example of a fluid therapy plan:

A 2.3-kg kiwi, estimated to be 10% dehydrated, with no ongoing fluid losses.

Replacement fluids: fluid deficit (mL) = 10% × 2300 g = 230 mL

Maintenance fluids: daily fluid (mL) = 50 mL × 2300 g = 115 mL/day

First 12 (–24) hours:

Fluid (mL) = replacement + maintenance fluids

= (½ × 230 mL) + 115 mL = 230 mL

Next 24 hours:

Fluid (mL) = (¼ × 230 mL) + 115 mL

= 57.5 + 115 mL = 172.5 mL

Next 24 hours:

Fluid (mL) = 172.5 mL

From here on, all of the replacement fluids have been given, and maintenance fluids of 115 mL per day need to be administered. If the bird is eating and drinking on its own, it may or may not require supplementation, depending on the individual case.

Selection of fluids for intravenous rehydration

Replacement fluids

Fluid and electrolyte deficits most commonly result from inadequate water intake, or from excessive loss of extracellular fluid due to haemorrhage or diarrhoea. Replacement fluids are required to contain the same electrolytes as those found in the extracellular fluid (Forsyth et al. 1999). Crystalloids are ideal as replacement fluids. They are capable of distributing to all body fluid compartments.

Lactated Ringer's solution (LRS) warmed to 38–39°C is recommended for fluid replacement and shock therapy (Quesenberry & Hillyer 1994). This most closely approximates the extra-cellular fluid (Forsyth et al. 1999), and is a good choice in most instances of fluid replacement. 0.9% NaCl (sodium chloride) may also be used for fluid replacement.

Total body potassium loss occurs through a lack of intake (anorexia) or through excessive loss (e.g. diarrhoea). Compared with other electrolytes, potassium losses are relatively greater, so supplementation with KCl (potassium chloride) should be considered (20 mEq/L) (Forsyth et al. 1999).

Maintenance fluids

Solutions used for replacement therapy are not suitable for long-term maintenance because of variations in electrolyte requirements (Quesenberry & Hillyer 1994). Fluid losses occurring through the gastrointestinal tract, urinary tract, lungs and skin contain approximately half of the sodium and chloride concentrations found in serum and LRS (Forsyth et al. 1999). Maintenance fluids should reflect these electrolyte concentrations by containing much less sodium (40–60 mEq/L) and more potassium (15–30 mEq/L) than replacement fluids (Forsyth et al. 1999; Lichtenberger 2004).

Options for intravenous maintenance fluids include:

1. Half-strength LRS plus 2.5% dextrose plus 20 mEq/L KCl
   e.g. 500 mL LRS plus 500 mL × 5% dextrose plus 20 mEq KCl
   This is an adequate maintenance solution (Forsyth et al. 1999).
2. Lactated Ringers solution (LRS)
   If used alone for maintenance fluid requirements, LRS can result in hypokalaemia (low potassium) and hypernatraemia (excessive sodium) (Forsyth et al. 1999).
3. 0.9% NaCl
   This is unsuitable for maintenance fluid requirements as it is not a balanced electrolyte solution (Forsyth et al. 1999).

Hypovolaemic shock

Rapid or extensive fluid loss causing hypovolaemic shock requires rapid replacement of blood volume. The most common cause of hypovolaemic shock in birds is haemorrhage (Lichtenberger 2004).

Boluses of warmed LRS may be given by slow intravenous or intraosseous administration at 10–30 mL/kg (Harrison 1986; Lichtenberger 2004). This amount should be administered over 5–7 minutes (Harrison 1986). There is usually an associated transient bradycardia (Harrison 1986). Fluid overloading rarely occurs, but may be indicated by tachypnoea (rapid breathing), cardiac dysrhythmias, agitation and collapse (Abou-Madi & Kollias 1992).

It is difficult to determine the exact fluid requirements of birds in shock (Quesenberry & Hillyer 1994). Thirty minutes after treatment with IV fluids, only 25% of isotonic crystalloid fluids remain in the vascular compartment, as the remainder of the fluid has redistributed to the interstitial fluid compartment (Haskens 1992). Therefore, improvement in circulation may be transient, requiring additional fluid therapy to prevent recurrence of hypotension and vasoconstriction (Quesenberry & Hillyer 1994).

Haemodilution is the primary limitation to crystalloid therapy, making the administration of colloids or blood necessary for effective shock therapy (Quesenberry & Hillyer 1994).

Hypertonic saline (7.5%) can be used as an intravenous bolus at a rate of 3–4 mL/kg, followed by intravenous crystalloid solutions (Firth 1995).

Colloids have not been used to any great extent in birds (Quesenberry & Hillyer 1994). Harrison (2006) recommends intravenous hetastarch at a dose rate of 10–15 mL/kg every 8 hours for one to four treatments for the treatment of hypoproteinaemia. Other reports suggest colloids should be given at a maximal dose rate of 20 mL/kg (Monks 1996). Colloids are large molecular weight substances that are unable to pass through capillary membranes, thus remaining in the intravascular space. They act by drawing fluid from the interstitium, and are thus more effective blood volume expanders than crystalloids (Abou-Madi & Kollias 1992; Haskins 1992). They are particularly useful at restoring circulating blood volume without aggravating hypoproteinaemia or causing pulmonary oedema in birds with low oncotic pressure and hypoproteinaemia (Quesenberry & Hillyer 1994).

Whole blood should be considered for severe blood loss.

Continue to next file: Part 2