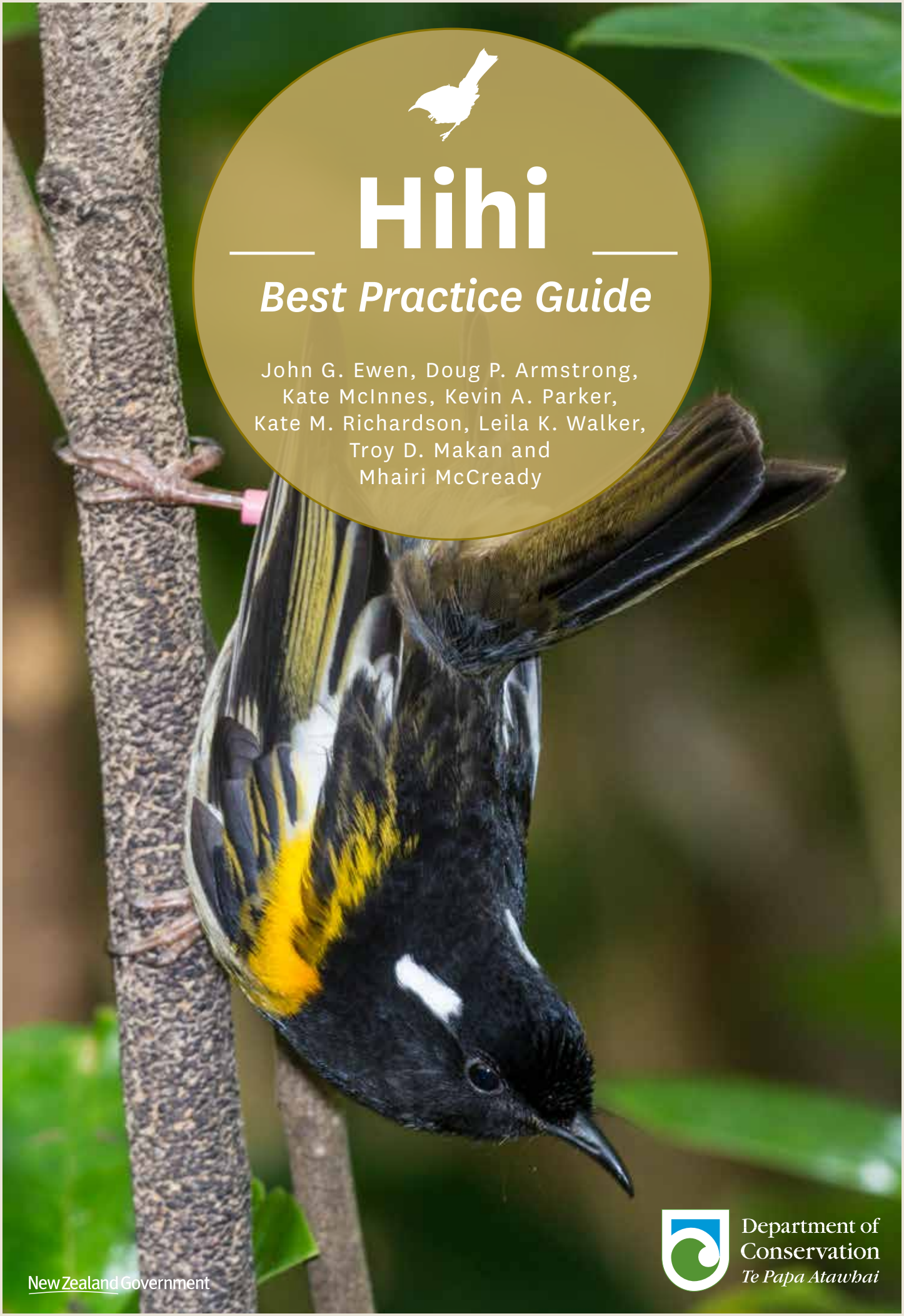

Hihi

Best Practice Guide

John G. Ewen, Doug P. Armstrong,
Kate McInnes, Kevin A. Parker,
Kate M. Richardson, Leila K. Walker,
Troy D. Makan and
Mhairi McCready

New Zealand Government

Department of Conservation
Te Papa Atawhai

Cover: Male hihi. *Photo: John Sibley.*

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Hihi Best Practice Guide

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1. Introduction to this hihi best practice guide

Hihi (*Notiomystis cincta*) have been actively managed since 1980 with a strong focus on protecting existing populations and using reintroduction to increase the species' range and global population size. There has been mixed success in establishing additional populations of hihi, with only 50% of translocation sites maintaining a population, and all of these reintroduced populations needing supportive management in the form of biosecurity, supplementary feeding and sometimes provision of nest boxes. In the early years of this recovery project the majority of work was undertaken by the New Zealand Wildlife Service and then the Department of Conservation (DOC). More recently, conservation has been increasingly undertaken by non-government organisations including community-run conservation groups and research institutions in collaboration with DOC. Representatives from across these organisations collectively make up the current Hihi Recovery Group (HRG). Given the diversity of groups working with hihi, and that hihi are spread over multiple populations, there is a need to share knowledge on past and present management. This and the following chapters therefore cover a range of current best practice protocols in hihi management. Throughout these chapters we have included:

- A series of box texts that provide more detailed background on points of interest
- A question and answer section on common queries and justification for why things are done as they are
- Recommended additional reading

This best practice guide will replace DOC's Hihi Standard Operating Procedure (SOP) to guide management and the ultimate recovery of hihi nationally.

1.1 Background on the Hihi Recovery Group (HRG)

The HRG is made up of a mix of DOC employees, scientists and community group representatives. The HRG works towards an agreed set of objectives and provides advice to requests for information, management guidance and recommendations. The group has never had any exclusive right to make or enforce management decisions. Management decisions will be made by relevant site managers and are ultimately governed by permit requirements set down by DOC. Right from the earliest translocations of hihi, there has been an involvement of research (e.g. Angehr 1984). This partnership has developed such that most forms of management are underpinned by research (e.g. Armstrong et al. 2007), and changes to management practice are monitored to determine how the change affects the population (e.g. Chauvenet et al. 2012).

1.2 Hihi Recovery Group objectives

What are objectives?

Objectives define the things that matter to us and should be expressed as concise statements including the thing that matters and (usually) a verb that describes the desired direction of change (Gregory et al. 2012). ***Objectives are not targets****. We should view targets, at best, as our agreed desired level of performance toward an objective (see more in the Q & A section below). It is also essential to separate out 'fundamental' from 'means' objectives, otherwise there is a risk of incorrectly evaluating management alternatives. 'Fundamental objectives' are the basic things we care about whereas 'means objectives' are more about the ways these fundamental objectives can be met. To identify what type of objective you have, you can ask yourself 'why is this important?' Once the answer is 'because it just is' then you have probably identified a fundamental objective.*

The HRG has identified four fundamental objectives with respect to hihi, each with a small number of criteria against which they can be measured (Table 1.1). Being explicit about how the HRG objectives will be measured allows for management alternatives to be assessed for their likely effect (consequence) on each objective. It therefore allows the HRG (or managers) to better choose between alternatives. Once a chosen management option is undertaken, the HRG (or managers) can then assess its success against these agreed measures. This allows explicit feedback on how management is, or is not, achieving what is considered important. A wider range of means objectives were expressed by the HRG and these are portrayed in a means-ends diagram (Fig. 1.1) to show how they can assist the group in achieving what is fundamentally important.

Table 1.1 Fundamental objectives of the Hihi Recovery Group, with suggested measures to judge our performance at achieving them.

FUNDAMENTAL OBJECTIVE	MEASURE	UNITS OF MEASURE
Maximise the number of hihi	Populations	Number
	Population size	Number
Maximise the degree to which the ecological setting of hihi is natural	Sugar water (minimise)	L/bird/year
	Nest boxes (minimise)	Number/bird/year
	% rare alleles maintained (maximise)	%
	Presence of non-native disease causing agents (minimise)	Number
	Presence of non-native predators (minimise)	Number
Minimise cost	Value of management effort	$/year
	Sugar water	L/year
	Field effort	Hours/year
Increase public appreciation	Visitors	People/year
	Hihi volunteers	People/year
	Public investment	$/year
	News/newsletter/magazine articles	Number & readership
	Bird of the year	Votes
	Scientific publications	Number
	Social media 'followers'	Number

1.3 How to use this guide

This best practice guide is made up of a series of standalone chapters, each dealing with a specific aspect of hihi management. Within each chapter we summarise the protocols used across populations and note any differences in approach to particular management issues. We acknowledge that there is often more than one solution to a management issue and that our best solutions will continually be improved through application and adjustment. This is how the current set of protocols have evolved. We therefore also include some of the trial approaches used and why they were, or were not, retained. By doing so we are hoping to provide a track record of solutions so that future adjustments build on previous work rather than 'reinventing the wheel'. As much as possible we also explain why things are done as they are, so that staff working with hihi have a greater understanding of why particular approaches are important and do not just 'follow a recipe'. Finally, and importantly, we are striving for a more unified and national approach to hihi conservation. This is made


Photo: Abi Low.

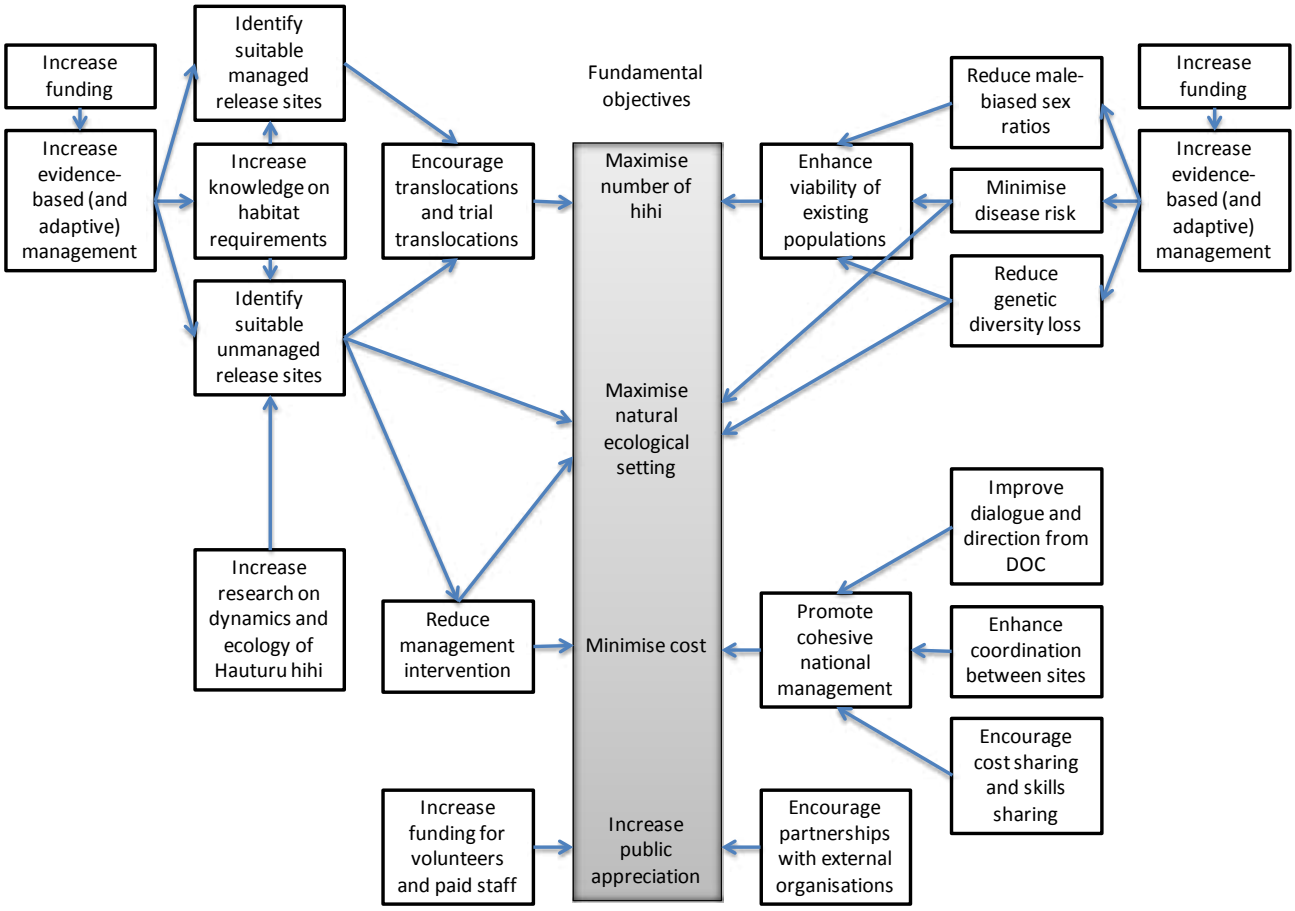


Figure 1.1. Means-ends diagram developed for hihi recovery. Many objectives were suggested by the Hihi Recovery Group and most of these represent means objectives. Whilst important, they need to be separated from fundamental objectives or they risk skewing weighting across our objectives and they can limit creative problem solving (Runge et al. 2011). Means objectives are important, but they are more so for helping identify what methods will help us get to what is fundamentally important.

easier by more standardised approaches to collecting and managing monitoring data. Monitoring is often a highly costly component of threatened species conservation and far too often provides data of limited value to the greater management programme of which it is a part. We therefore provide a set of suggested data recording sheets (one partially completed to use as an example and another without data that can be printed out and used).

1.4 Q & A: why do we do it this way?

Over many years our approach to hihi management has evolved, and frequent modification of management techniques has identified the best practice protocols just described. We recognise that these best practice approaches will continue to be modified and optimised as new needs are identified, new products become available and new insights are made. In order to be efficient in our continued refinement of management we need to keep track of approaches we have trialled but discarded, and log the key justifications for our current decisions. These are addressed in the following Q & A section for this chapter and in the Q & A sections in subsequent chapters.

Q. Why should we be careful about using targets?

A. Targets set minimum thresholds of achievement on particular issues. They can often act, therefore, as constraints on possible alternative actions, or constraints on trade-offs between different objectives, because they immediately eliminate those management alternatives that

cannot achieve the targets. This can be especially damaging if a best compromise between competing objectives is not considered because the compromise doesn't reach a pre-set target. For example, the hihi recovery group used to state a long-term goal of '...increasing the number of self-sustaining hihi populations to at least five'. Whilst this may be an exciting prospect it could risk removing important alternatives, such as the current focus on managed populations as being, at worst, a waste of time. This is, of course, not how the group works but our operation often then doesn't clearly help us achieve our previously stated long-term goal. This becomes more important when using a structured approach to hihi management. Furthermore, if we do manage to successfully establish a second 'self-sustaining' population we have still failed to achieve our goal (of five populations), despite achieving a feat that has eluded the recovery of hihi for more than three decades.

Q. Do we need to have specified measures and directions linked to our objectives?

A. Yes. Directions are important as they clearly show what we want to do and allow us to compare how different alternatives achieve each objective. We should avoid the word 'optimise' in stating our objectives because this is what we do across objectives, not within them! If you want to optimise something then it is likely that you are focussed on something other than a fundamental objective (or are focused on at least two of them). Measures are essential regardless of how hard they are to obtain. If it matters to choose among alternatives, then they need to be there. Some measures may rely entirely on expert opinion and this is valid for a decision, if treated correctly, and there is a large body of literature on doing this. If it seems hard to choose a good measure for an objective then be careful to recheck that the objective is clearly stated. It is important there is no ambiguity in the language of an objective. The hihi recovery group has previously discussed this issue in relation to terms such as 'self-sustaining' which can have variable definitions between different group members. Either avoid such terms or clearly define what you mean by them.

1.5 Suggested further reading

Gregory, R.; Failing, L.; Harstone, M.; Long, G.; McDaniels, T.; Ohlson, D. 2012: Structured decision making: a practical guide to environmental management choices. Wiley-Blackwell. 312 p.

This book provides a great introduction to structured decision making. It's easy to read and has loads of examples.

Ewen, J.G.; Walker, L.; Canessa, S.; Groombridge, J.J. 2014: Improving supplementary feeding in species conservation. *Conservation Biology* DOI: 10.1111/cobi.12410 (available online open access).

This paper has a worked example of a basic structured decision making application to a hihi management problem on Kapiti Island.

Converse, S.J.; Moore, C.T.; Folk, M.J.; Runge, M.C. 2013: A matter of tradeoffs: reintroduction as a multiple objective decision. *The Journal of Wildlife Management 77*: 1145–1156.

A great paper from Sarah Converse and team providing a worked example of multi-objective decision making in the Florida non-migratory whooping crane reintroduction effort.

1.6 Additional literature cited in text

Angehr, G.R. 1984: Ecology and behaviour of the stitchbird (*Notiomystis cincta*). Internal report to the New Zealand Wildlife Service. Wildlife Service, Wellington.

Armstrong, D.P.; Castro, I.; Griffiths, R. 2007: Using adaptive management to determine requirements of re-introduced populations: the case of the New Zealand hihi. *Journal of Applied Ecology 44*: 953–962.

Chauvenet, A.L.M.; Ewen, J.G.; Armstrong, D.P.; Coulson, T.; Blackburn, T.M.; Adams, L.; Walker, L.K.; Pettorelli, N. 2012: Does supplemental feeding affect the viability of translocated populations? The example of the hihi. *Animal Conservation 15*: 337–350.

Runge, M.C.; Cochrane, J.F.; Converse, S.J.; Szymanski, J.A.; Smith, D.R.; Lyons, J.E.; Eaton, M.J.; Matz, A.; Barrett, P.; Nichols, J.D.; Parkin, M.J. 2011: An overview of structured decision making. U.S. Fish and Wildlife Service, National Conservation Training Center, Shepherdstown, West Virginia, USA.

2. Hihi advocacy


Photo: Melissa Boardman.

One of the four fundamental objectives set by the Hihi Recovery Group (HRG) is to increase public appreciation of hihi. This objective was recognised because the HRG had a desire for the general public of New Zealand (and internationally) to love this bird with the same passion as we do kiwi and other iconic species. This chapter therefore sets out to share the reasons for loving hihi, rather than a more traditional approach to advocacy which is to provide support for restoration programmes. There are many alternative ways of achieving an increase in the public's appreciation of hihi, but all rely to some degree on sharing information about them, and related conservation efforts, with external audiences. In this chapter we develop some of the basic information and advice that may assist with advocacy work. The information we provide here is not exhaustive; rather, it provides some relevant background and a set of key messages that may be useful for providing insight into what hihi are and some unusual or unique 'marketable' hihi facts that may foster a love for them. This information is provided to assist the people whose job it is to provide information to relevant audiences and who may not be species experts themselves. In addition, we hope to facilitate a unified and national response to hihi advocacy, following a recommendation made by group members at the 2013 Recovery Group Meeting held at Bushy Park. We encourage messages to be supportive and collaborative across the wider community and the research and government groups involved in hihi management. Our key messages have been included in an information pamphlet that is available at sites where hihi are present and which is also freely available for download from the HRG website (www.hihiconservation.com).

2.1 Audiences

Regional community: All sites with hihi can educate local visitors about why hihi are no longer found there naturally and how the current conservation effort has allowed the species to be brought back to the area. Increasing this connection between local communities and hihi will increase the knowledge, appreciation and love of this and other birds found in their region. Furthermore, using hihi as a model can help encourage communities to do more in their region to protect birds and increase conservation projects. Some ways they could do this would be by donating financially, getting involved through volunteering, planting bird-friendly gardens, checking they are free of rodents – each site will have specific messages that are appropriate for their location.

National community: All hihi sites are encouraging visitors from around the country to learn more about conservation and its importance to New Zealand and therefore all play an essential role in raising awareness of hihi nationally. Advocacy targeted here can encourage visitors to support their local hihi programmes, if they exist, or to look at ways they could create safe places for hihi or other New Zealand natives.

International community: We are educating international visitors and volunteers about conservation issues in New Zealand, fostering a love of New Zealand biota and hoping to inspire them to not only contribute to conservation while in New Zealand, but to take back a love of conservation to their home country.

Audiences who do not visit hihi sites: There are a growing number of ways of providing an advocacy message beyond the borders of sites with hihi restoration projects and aviaries with hihi.

- The researchers on the recovery group play an important advocacy role in promoting the model system that has developed around hihi in relation to small population biology and conservation biology research. Scientific and popular science articles relating to hihi address wide-ranging topics in conservation, behavioural and evolutionary biology and wildlife health. Academic outputs often get picked up by local and international media and reach wide audiences.
- Social media and online presence: rapid exposure is possible from promotion through social media (e.g. Facebook, Twitter) and websites.
- Public speaking at conferences, events and to interested groups.

2.2 Key messages

Little ***ray of sunshine***. Can hihi, with their patches of bright yellow feathers, become a symbol of the challenges and hope for conservation in New Zealand?

We have identified **10 key messages** that could be used in hihi advocacy work. These messages may be selected to best suit particular opportunities and are not exhaustive. This list was developed through discussion with marketing experts at each community project where hihi have been reintroduced and through a brainstorming exercise at the 2014 hihi recovery group meeting held at Maungatautari. Our aim here is to provide a set of key messages that cover a breadth of interests and are trusted to be factually correct and up-to-date. An advocacy plan developed for hihi at Pukaha Mount Bruce is described in Box 2.1.

Box 2.1 Advocacy role for Pukaha Mount Bruce's captive hihi programme

A hihi advocacy plan has recently been developed by DOC for Pukaha Mount Bruce. This plan will be implemented in partnership with a non-DOC organisation working at Pukaha (The Pukaha Mount Bruce Board). The agreed advocacy roles are:

1. To increase public awareness of hihi as a species.
2. To help visitors understand the ecological niche of the hihi and how it fits into the forest ecosystem.
3. Explain to visitors that hihi are at high risk of extinction because:
   - They are extremely vulnerable to all predators and competitors.
   - They are believed to require complex forest habitats with a high diversity of invertebrates and nectar-producing plants – and there is very little of this type of forest left in New Zealand.
4. To raise the profile of the hihi recovery programme and show how communities are making a significant contribution to the conservation of the species through:
   - Building pest-proof fences, trapping programmes or leading restoration and recovery at pest-free sites.
   - Volunteering on hihi management projects such as monitoring, supplementary feeding and nest box maintenance.

1. **Hihi have a strong place in Maori tradition**
    - Hihi means '**ray of sunshine**'
    - **Into the fire**! In Maori folklore the hihi refused to fetch water for Maui after he had tamed the sun. Maui threw the hihi aside and it landed in the fire, burning its feathers. Thus the black and yellow feathers are its permanent reminder of the lesson learned.

Photo: Kim Westerkov.

Photo: Robert Okell

2. Hihi are unique

- Did you know that hihi are unique? They are the only representatives of an **endemic** (only found in New Zealand) bird **Family** (for hihi this is Notiomystidae) (Note: level of biological classification is species<genus<family<order). Being endemic at this higher level of classification emphasises just how unique the hihi is (not many other New Zealand birds can claim such evolutionary distinctiveness). Even the kōkākō shares its family (Callaeidae) with another genus (the saddleback (tīeke)).
- For many years hihi were considered to be in the honeyeater family (Meliphagidae) along with New Zealand's tūī and bellbird (korimako) until their new status was determined from genetic studies shown in the Phylogenetic tree illustated in Fig. 2.1 below. Their **closest living relatives** are the Callaeidae (kōkākō and tīeke).

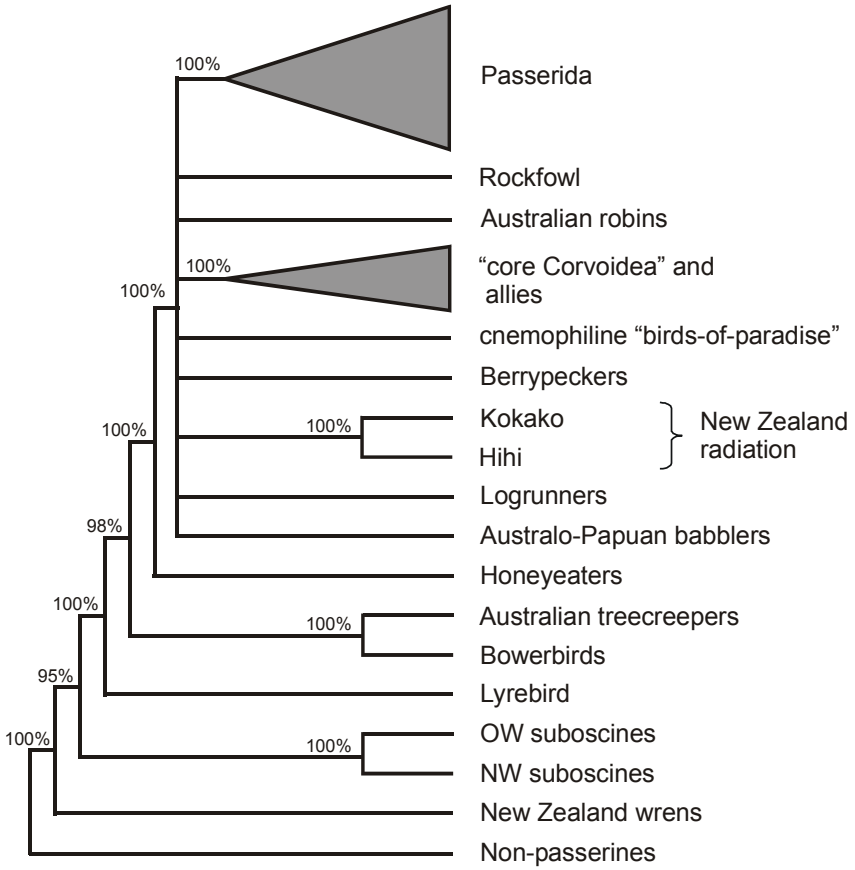


Figure 2.1. Phylogenetic tree showing the systematic position of the hihi (*Notiomystis cincta*). The tree was derived from 3486 bp obtained from 71 species representing all major groups of passerines. The DNA sequences derive from two nuclear, protein-coding genes (RAG-1 and c- mos). To enhance visibility, the tree has been simplified to only show clades that received posterior probabilities of 95% or higher in the Bayesian analysis. The tree indicates hihi and kokako group strongly together as a single clade within the newly described paraphyly of the oscine radiation. Figure reproduced with permission from Ewen et al. 2006.

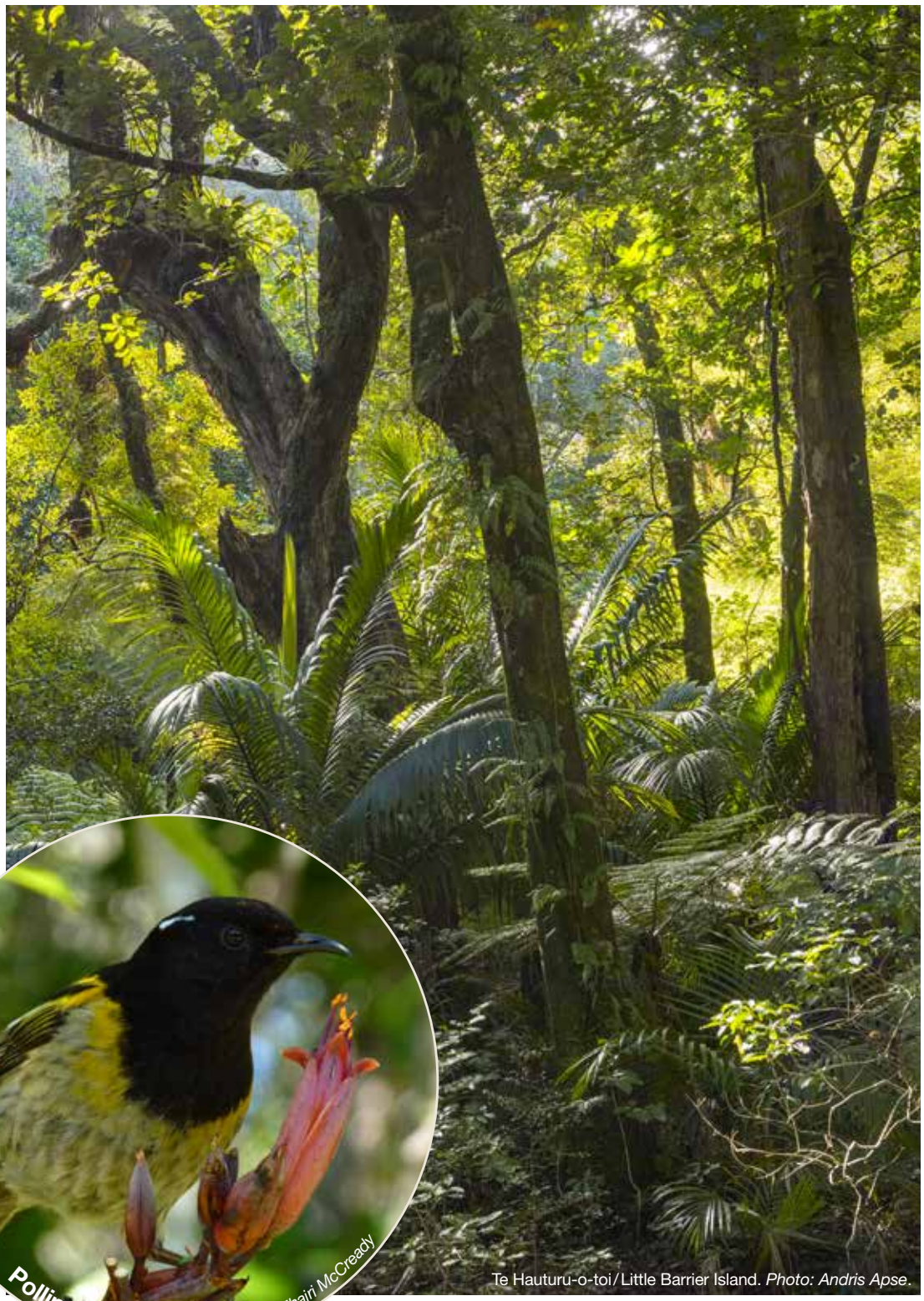
3. Hihi are rare

- Did you know that hihi are rarer than brown kiwi?
- Only a **few thousand hihi remain,** mostly on Te Hauturu-o-toi / Little Barrier Island.
- Hihi are classified as **Nationally Endangered** under the New Zealand Threat Classification System (Robertson et al. 2016) and vulnerable by the IUCN Red List.
- Hihi were once found throughout **northern New Zealand** (North Island and larger offshore islands) but after introduction of mammalian predators and clearance of forest they disappeared from these areas and, by about 1890, were restricted to just one offshore island population on Te Hauturu-o-toi / Little Barrier Island.
- **Reintroductions started in 1980** and there are currently six small reintroduced populations (each with less than 200 adult birds) and a captive (aviary) population (see Fig. 2.2 below).

Figure 2.2. Map showing the distribution of remnant hihi population (on Te Hauturu-o-toi / Little Barrier Island) and reintroduced populations.

4. Hihi are important in our forests

- Hihi provide **a litmus test for the state of New Zealand's North Island forests** because they require diverse and relatively intact forest ecosystems to support them.
- Hihi are **important pollinators** of forest plant species. Recent studies have shown loss of hihi has resulted in loss of **ecosystem services** of pollination, causing reduced seed production and plant density for some plant species.



Te Hauturu-o-toi / Little Barrier Island. *Photo: Andris Apse.*

Pollination in action! Photo: Mhairi McCready

5. Hihi have colourful sex lives!

- Hihi hold **world records** for levels of ***extra pair sexual activity*** and ***illegitimate young*** amongst song bird species! 68% of all young are the result of ***extra-pair copulation*** and ***illegitimate young*** are found in 89% of nests. Both males and females pursue extra-pair copulations.
- Hihi are the **only bird species known** to copulate in a **face-to-face** position. They also copulate in the more typical male-on-the-female's-back position. Face-to-face copulations are forced on females by males.
- **Size matters!** The testes of male hihi are huge during the breeding season, swelling to take up a large proportion of their abdominal cavity and weighing about 1.73 g (which equates to about 4% of their body mass). At this time the testes of a male are bigger than his brain!

Fighting males. *Photo: unknown.*

Photo: Geoff Beals.

Photo: Simon Fordham.

6. Hihi are colourful and curious

- The male hihi is one of the most **strikingly colourful** of New Zealand's bush birds, with a **jet black head**, **white ear tufts** (which can be raised and flared in display) and a broad band of **bright yellow** feathers on the wing, shoulders and breast.
- **What you wear counts**. In birds, bright and striking male plumage often acts as a **signal of reproductive ability**. The larger a male's area of yellow feathers is, the more likely he is to be a successful territory holder, but the lighter the yellow colour of these feathers, the greater the likelihood that he will be cuckolded (lose paternity to other males). Longer white-ear tufts and a lighter black head signal a greater ability to score with the girls!
- Hihi are **very curious and charismatic**. Look for their distinctive tail-tilted-up pose. Hear them constantly chattering away whilst moving through the forest or coming to check you out!

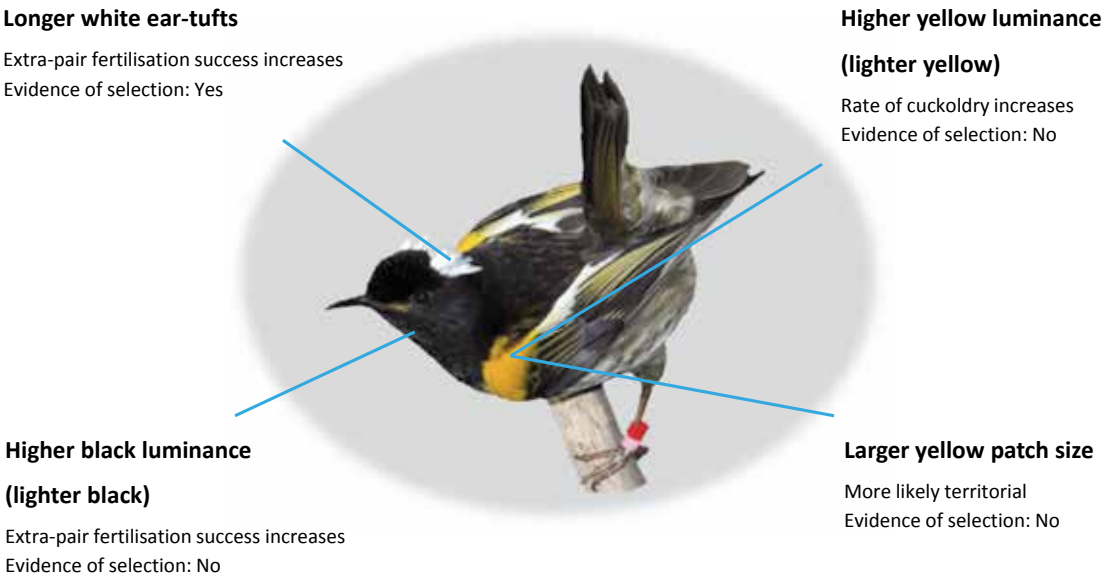


Figure 2.3. Signal content of hihi plumage. Reproduced with permission from Walker et al. 2014.

7. Hihi chicks are cute!

- Hihi can have up to five fluffy chicks in a nest.
- Hihi chicks start off naked except for a fluffy crown of head feathers!
- Hihi nestlings stay a long time in the nest – up to 30 days – which is about 2.5 times longer than other New Zealand bird species of the same size.
- Hihi nests are built in cavities, such as holes in trees, and they are BIG, a high-rise building of sticks with the nest cup right at the top, like a penthouse apartment.
- Where there are not enough mature trees with nesting cavities, hihi will happily use artificial nest boxes.
- Because they spend so long in the nest, hihi chicks leave behind a big mess when they depart, which means nest boxes must be given a big clean-out at the end of each breeding season!

Natural nest in tree, Maungatautari. *Photo: Lydia Doerr.*

8. Hihi are challenging to save

- Populations have established at only **50% of sites** where hihi have been reintroduced and all of these populations currently need supportive management.
- All reintroduced populations are fed with **sugar water** which is essential for their survival.
- Hihi are regarded as the **acid test** for restoration ecology in northern New Zealand due to their sensitivity to habitat quality (i.e. only high-quality habitat can support hihi).
- Hihi are extremely vulnerable to all predators and competitors.
- Hihi require complex habitats with high diversity of invertebrates and nectar-producing plants, and trees large enough to have nesting cavities, but very little of these sorts of habitats remain in New Zealand. Hihi therefore highlight the complexity of the conservation problems faced in New Zealand. There are currently no quick and easy fixes. No silver bullet has been identified to help them... yet.

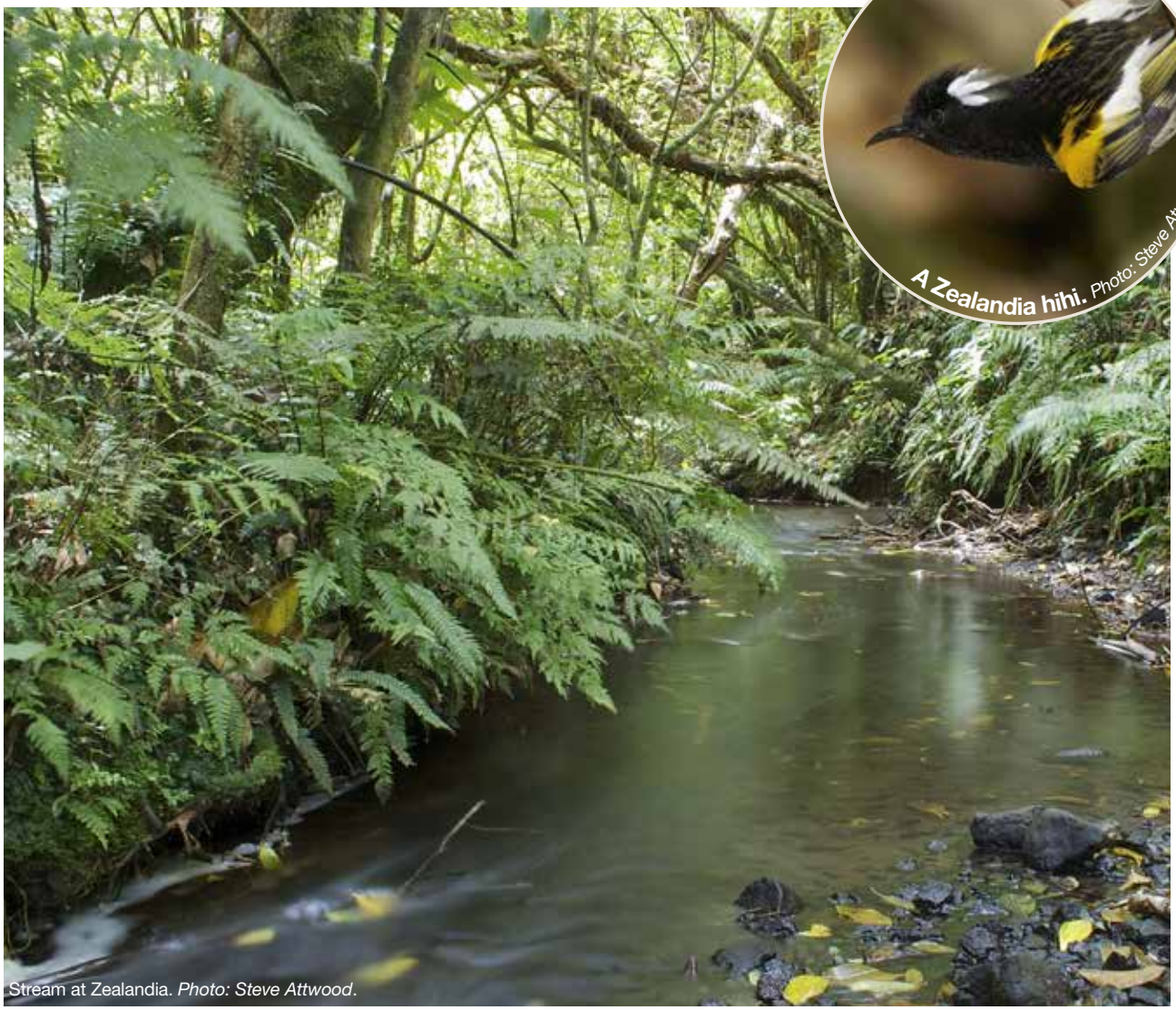
At present, hihi need high-tech help!

Photo: John Ewen

Sugar water feeders on Kapiti Island can readily be adapted to catch hihi for monitoring and translocation purposes.

9. Hihi conservation is a national team effort that needs your help

- Populations are currently managed at
  - Kapiti Island (DOC & local Hihi Heroes programme)
  - Tiritiri Matangi Island (DOC & Supporters of Tiritiri Matangi Inc.)
  - Zealandia (Karori Sanctuary Trust)
  - Maungatautari (Maungatautari Ecological Island Trust)
  - Bushy Park Sanctuary (Bushy Park Trust)
  - Rotokare Scenic Reserve (Rotokare Scenic Reserve Trust)
  - Pukaha Mt Bruce (captive – aviary) (DOC & Pukaha Mount Bruce Board)
- Volunteers are essential and there are many ways to become involved. Contact your local DOC office or community-based project to find out more.
- A good example of how hihi could drive community-based projects is the Polhill Restoration Project in Wellington. Local people are working together to improve the quality of Polhill Reserve adjacent to Zealandia, which is experiencing 'spillover' of rare and endangered birds from Zealandia. Tīeke (saddlebacks) in particular are now breeding in Polhill Reserve, 10 minutes from downtown Wellington. This is a very modern restoration project, being mainly organised via its Facebook page. For more information, go to https://www.facebook.com/groups/Polhill/

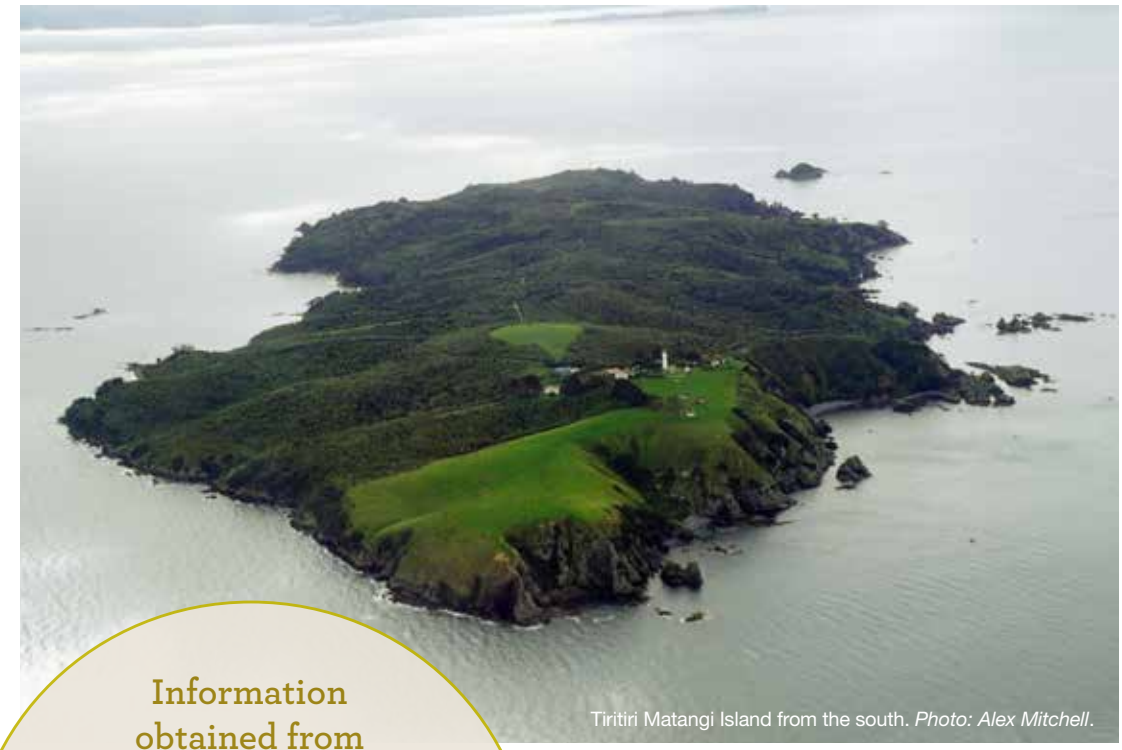


A Zealandia hihi. *Photo: Steve Attwood.*

Stream at Zealandia. *Photo: Steve Attwood.*

10. Hihi provide a model system in global conservation biology research

- Researchers from New Zealand and around the world study important issues in small population biology and conservation and the hihi is a **globally renowned** model study system in this area.
- Hihi conservation uses leading research principles such as an evidence-based approach with explicit theory, focused monitoring and adaptive management.
- To keep up-to-date with hihi conservation activities and research, got to: **www.hihiconservation.com/**



Tiritiri Matangi Island from the south. *Photo: Alex Mitchell*.

Information obtained from hihi management and research, including from the translocated population on Tiritiri Matangi Island, is contributing to the conservation of other endangered species.

2.3 Assessment of our advocacy achievements

We have identified a range of measurable attributes (Table 2.1) to determine how well we may be achieving our advocacy objective, or to compare possible alternative advocacy actions for helping us best achieve them. The list is a first brainstorming exercise but gives an idea of how we can estimate whether public awareness is increasing.

Table 2.1 Measurable attributes for determining achievement of hihi advocacy objectives.

FUNDAMENTAL OBJECTIVE	MEASURE	UNITS OF MEASURE
Increasing public appreciation of hihi	Visitors	People/year
	Hihi volunteers	People or hours/year
	Public investment	$/year
	News/newsletter/magazine articles	Number and readership
	Bird of the year	Votes
	Scientific publications	Number
	Social media 'followers' and/or website 'hits'	Number

2.4 Available resources

A wide range of resources are available that contain information on hihi. Field guides and other published bird books provide some detail and the main caution with respect to these references is checking that they are up to date on recent taxonomy and distribution. We are constantly learning more about hihi and their management, so information can become dated relatively quickly. Also, translocations are occurring frequently so hihi distribution is also changing.

We have a website dedicated to hihi which provides the most up-to-date summary of recent management and research. The website has a secure section (password protected) for recovery group members to access all population reports, recovery group minutes and other news, data and updates. Ideally, this site will also provide a nice logbook of print and online media coverage.

Website: www.hihiconservation.com

Twitter: @hihinews

In addition, each of the locations that have hihi will also have dedicated websites and other forms of social media that provide up-to-date information on what is happening at the local scale.

2.5 Key contacts

All people on the hihi recovery group know hihi and are therefore valid contacts for advocacy advice. If in doubt about locating a particular expert then please contact one of the recovery group chairs who can help link relevant contacts together. All details are available at: **www.hihiconservation.com**.

2.6 Suggested further reading

Ewen, J.G.; Flux, I.; Ericson, P.G.P. 2006: Systematic affinities of two enigmatic New Zealand passerines of high conservation priority, the hihi or stitchbird *Notiomystis cincta* and the kokako *Callaeas cinerea*. *Molecular Phylogenetics and Evolution 40*: 281–284.

Ewen, J.G.; Renwick, R.; Adams, L.; Armstrong, D.P.; Parker, K.A. 2013: 1980–2011: 31 years of reintroduction efforts of the hihi (stitchbird) *Notiomystis cincta* in New Zealand. In: Soorae, P.S. (Editor): *Global reintroduction perspectives: additional case studies from around the globe*. IUCN/SSC Re-introduction Specialist Group, Abu Dhabi, UAE.

This book chapter provides a recent and brief summary about hihi recovery and it is freely available through the IUCN's Species Survival Commissions Reintroduction Specialist Group website.

Robertson, H.A.; Baird, K.; Dowding, J.E.; Elliott, G.P.; Hitchmough, R.A.; Miskelly, C.M.; McArthur, N.; O'Donnell, C.F.J.; Sagar, P.M.; Scofield, R.P.; Taylor, G.A. 2016: Conservation status of New Zealand birds, 2016. *New Zealand Threat Classification Series 19*. 27 p.

The latest listing of threat classifications for New Zealand birds. Available online at:
http://www.doc.govt.nz/about-us/science-publications/series/new-zealand-threat-classification-series/

Walker, L.K.; Ewen, J.G.; Brekke, P.; Kilner, R. M. 2014: Sexually selected dichromatism in the hihi *Notiomystis cincta*: multiple colours for multiple receivers. *Journal of Evolutionary Biology 27*: 1522–1535.

Photo: Neil Davies

3. Hihi nest box design and maintenance

Hihi are cavity nesters. However, at some translocation sites the forest is not mature enough to provide sufficient nesting cavities for a breeding hihi population. Nest boxes are therefore provided. The hygienic maintenance of nest boxes between breeding attempts is very important. This chapter details all recommended procedures relating to the construction of nest boxes, their placement and their hygienic maintenance.

3.1 Nest box design

A variety of nest box designs have been used in hihi populations, and certain features are now recognised as being important. A good hihi nest box will:

1. Provide easy access for hihi, but exclude larger cavity-nesting species
2. Have an internal partition that acts as a roosting perch and provides an entrance 'corridor' beside the nest base
3. Have a weather-proof hinged roof to provide easy access for monitoring
4. Be made of a material that is breathable and does not allow moisture to build up inside the box
5. Have a main unit that can be removed from the backboard for ease of cleaning

A nest box design that includes all of the features listed above, and that has been successfully used on Tiritiri Matangi Island, is detailed here (and shown in plan version in Fig. 3.1). Each nest box requires:

- 18 mm tanalised pine plywood of at least CD grade
- 26 × CSK surefast stainless steel screws 8G×35
- 3 × pan head stainless steel screws 8G×20
- Ados F2 glue
- Butynol for hinge 255 mm × 120 mm

The nest boxes are most easily constructed using jigs (as pictured in Fig. 3.2). You should avoid plywood that has buckled due to poor storage. The D face of the ply (a lower-appearance grade with permitted defects) will have open knots on its face; you should avoid positioning these knots on the inside face of the sides near the front joint, as water will penetrate into the nest box. Eight sheets of plywood will produce between 40 and 43 nest boxes (depending on the quality of the D face and how flat the ply is). A person experienced in making these nest boxes can produce about 40 boxes in 45 hours.

When fixing the nest box to a tree, two 75-mm timber screws are required per nest box. Holes should be pre-drilled in the middle of each of the backboard cleats for receiving the screws, and a 5 mm internal hex drive bit is required to drill-in the screws.

3.2 Nest box location

Nest box placement should encourage hihi use. If possible, nest boxes should be positioned in locations where hihi have already been found. If this is not possible – for example, if nest boxes are being put up at a new site prior to translocation – then the boxes should be positioned in suitable hihi habitat. Recent work at Maungatautari Ecological Island has shown that hihi may prefer to establish breeding territories in habitat near streams (within about 150 m) (Richardson & Ewen 2016) and we believe hihi will prefer the most mature forest areas available within sites. It may be worth selecting this combination of habitat features when selecting nest box sites. The

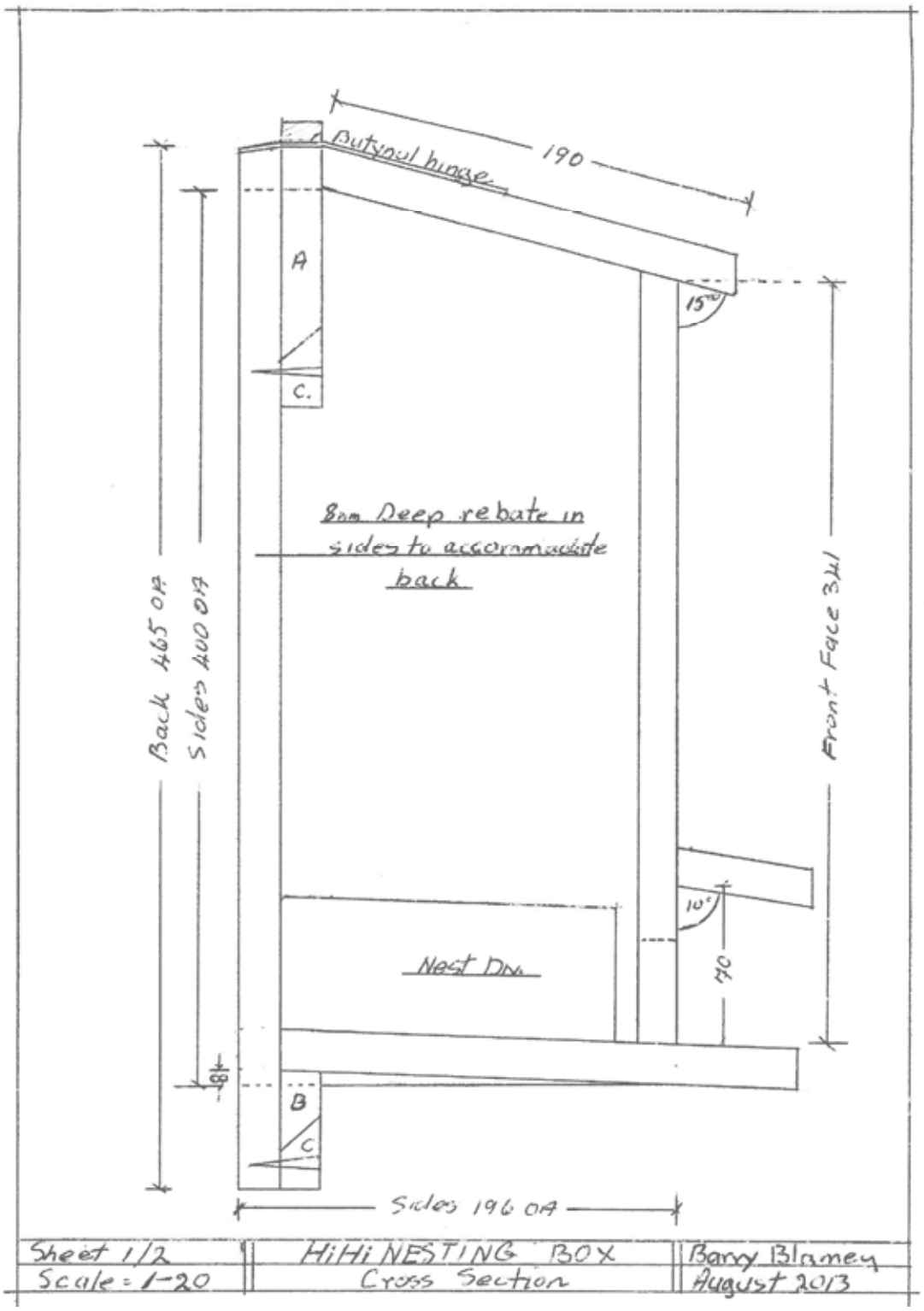


Figure 3.1. Tiritiri Matangi nest box design (a).

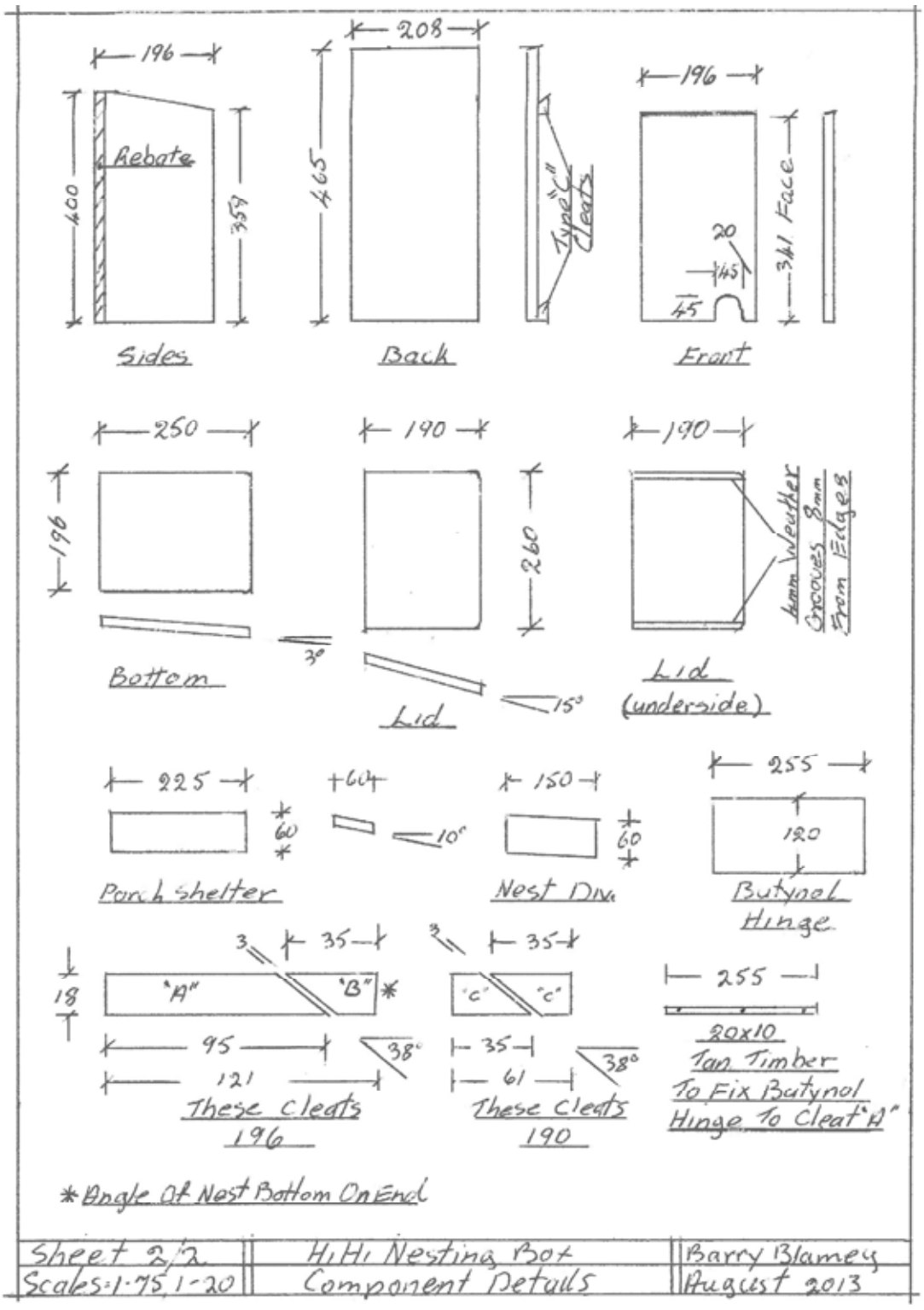


Figure 3.1 continued. Tiritiri Matangi nest box design (b).

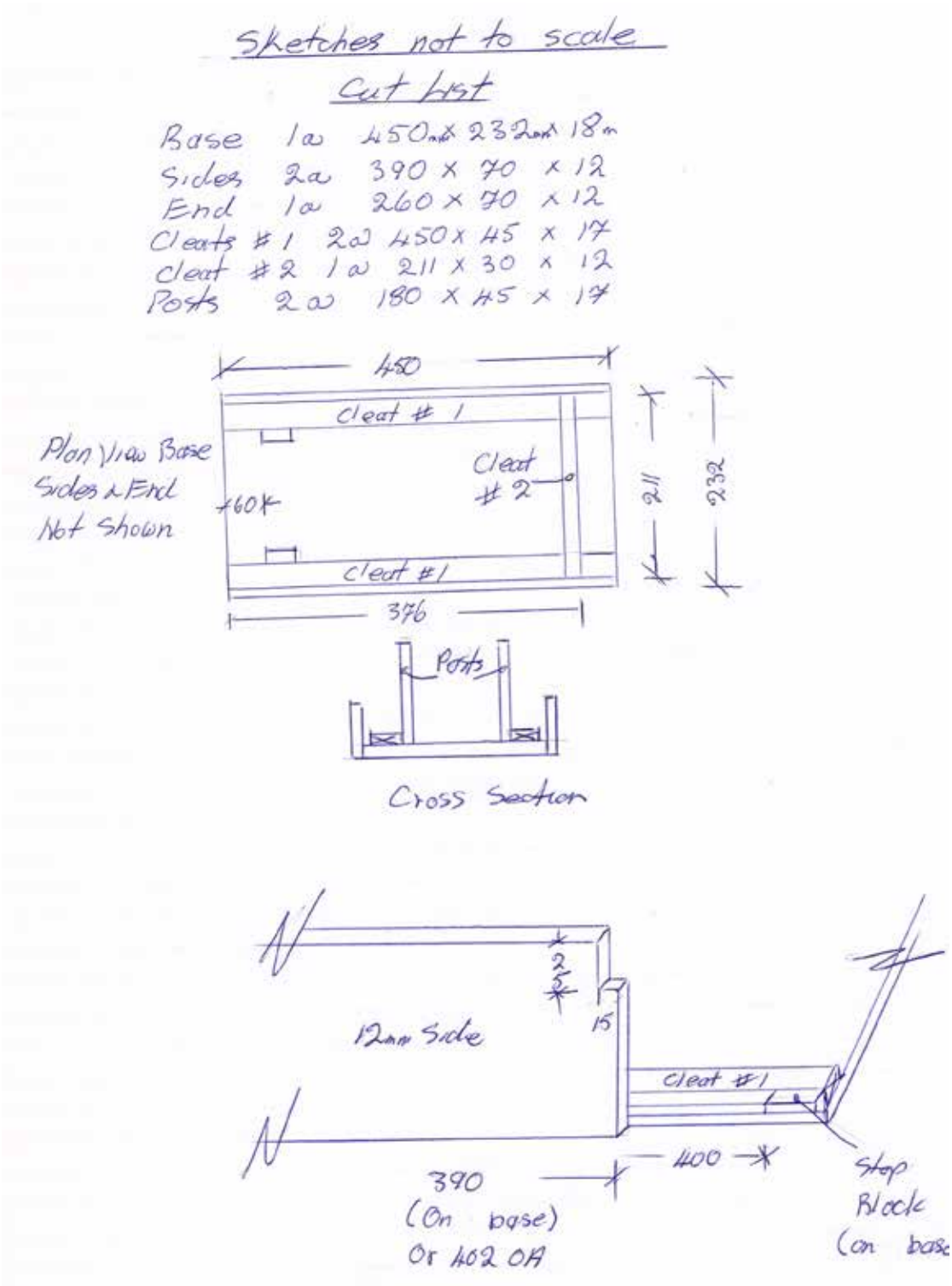


Figure 3.1 continued. Tiritiri Matangi nest box main jig design.

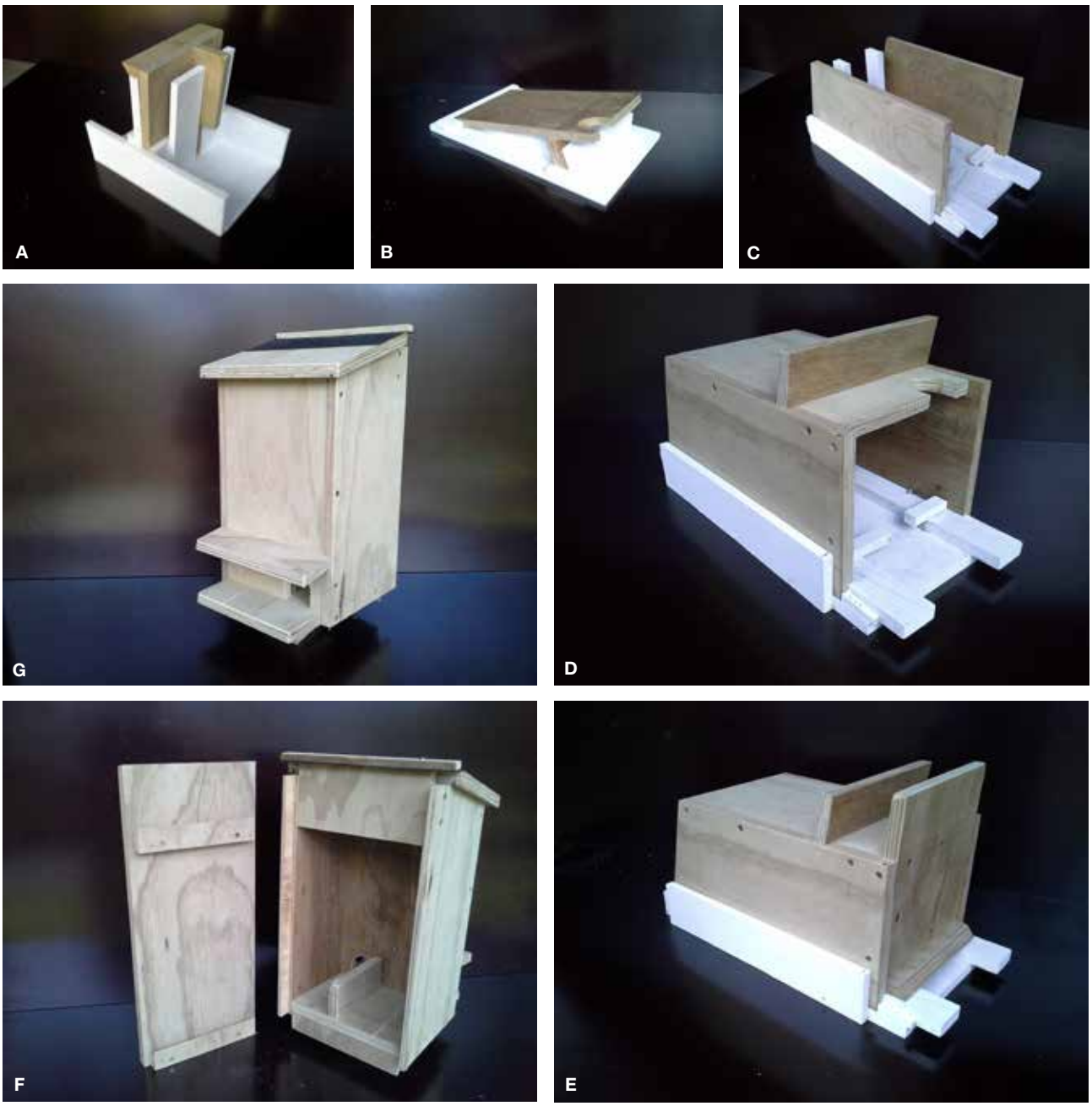


Figure 3.2. Constructing a hihi nest box using a jig (clockwise from top left): A. The base; B. The front panel; C. The side panels; D. Front and side panels; E. Front and side panels with base; F. Backboard and main unit; G. The finished product.

nest box location should be sheltered, semi-shaded and under the forest canopy. The nest box should not be in direct sunlight for long periods of time, and should not be positioned directly above streams or other forest features that may present a hazard to young birds during fledging. Ideally, the nest box should not be easily visible from public footpaths, and should not be within 30 m of communal hihi feeding stations. The tree to which the nest box is attached should be sufficiently stable to support a nest box, and the nest box should be attached at a height that allows safe checking of its contents. A single territory will usually have two nest boxes within 10 m of each other. This allows for some choice of nest site within a territory, and for quick turn-around between clutches. Nest boxes need to be cleaned regularly. The cleaning process is described in **Hihi Best Practice Sheet 1**.

Figure 3.3. Nest boxes in action: A. A nest box in location; B. Filming inside a nest box; C. A female surveying her territory from a nest box porch; D. A completed nest awaiting eggs; E. Young nestlings; F. Older nestlings in an artificial nest, complete with mess.
Photos: Leila Walker and Rose Thorogood.

3.3 Q & A: Why do we do it this way?

For a general description of the reason for this Q & A section, see Chapter 1. The following set of questions and answers capture some of the recent trials and changes relating to nest boxes:

Q. Do we always need to put out boxes in pairs before birds have established territories?

A. Once a territory has been established, it is relatively important that a nesting pair have more than one nest box available to them. This is because the turn-around between clutches is frequently very fast. Often the female will start building in the alternative nest box before the first clutch has fledged. Without an alternative nest box, the female's productivity could ultimately be compromised, but at the very least she may start building a new nest directly on top of an un-hygienic old nest. However, when selecting a new possible breeding territory it is reasonable to initially put up just one nest box to trial whether the location will be used. If it is used, a second nest box can be added to the territory in time for the second clutch. If there are no spare boxes to add to a single-nest-box-territory that is being used, you will need to be very fast at cleaning the nest box once the first clutch has fledged/failed, in time for subsequent nesting attempts.

Q. How close should boxes be spaced to encourage new territories?

A. The distance between territories, and therefore between pairs of nest boxes, is very much dependent on the density of hihi. When the population is small, and hihi density is low, territories are likely to be large and far apart. As the population grows and hihi density increases, territories will become smaller and closer together. Nest box placement will necessarily reflect this. As an example, the density of hihi on Tiritiri Matangi is relatively high, and the closest territories can be within 30 m of each other. In contrast, at Maungatautari Ecological Island the closest territories are within 100 m of each other (although here they do not use boxes).

Q. What is our current best guess on the most suitable nesting habitat for hihi?

A. We do not know with any certainty what the most suitable hihi nesting habitat is. Indeed, the question of habitat suitability is relevant to why previous hihi reintroductions have failed, and why extant populations require supplementary feeding. Te Hauturu-o-toi / Little Barrier Island provides some clues, as the home of the only remnant population of hihi. In terms of selecting suitable locations for nest box placement we currently recommend choosing a site that has mature forest with a closed canopy, or at least the most mature forest available within the release area. Hihi will use young regenerating forest, but as a general guide, the older the forest the better. It is also worth spreading the boxes widely throughout the release area to provide maximal choice by the birds for where they prefer to nest. As space becomes occupied by hihi, more boxes can be added to preferred areas. Trees that are chosen for nest box attachment need to be robust and, ideally, sheltered from direct exposure to sun or rain. In regenerating habitats this can be a primary criteria restricting nest box placement (see further details above).

Q. Which direction (side of tree) should we place boxes?

A. We have speculated on the possibility that hihi have some preference for the orientation of nest boxes. This could, for example, be related to compass orientation and the resulting prevailing weather, or topography and surrounding vegetation. However, there is currently no evidence to suggest that hihi prefer nest boxes to be on a particular side of a tree. We instead recommend that the placement of nest boxes on trees should maximise stability, reduce exposure to direct sunlight and rain, and reduce fledging risk. Research at Bushy Park suggests areas relatively clear of vegetation directly in front of nest entrances are also preferred.

Q. Hihi normally nest high up trees but we place the boxes about chest height of the average person. Does this matter?

A. We place nest boxes at chest height so that nests can be monitored quickly and safely, with the minimum of disturbance to the nest. Hihi appear to show no reluctance to using nest boxes

at this height and nests in these boxes typically have very high success rates in monitored populations such as at Tiritiri Matangi. Most,but not all natural nests are found high up trees. The reasons for this are not known with certainty, although it is likely that more cavities are available higher in trees. Whatever the reason for hihi nesting at height, there appears to be no substantial cost to providing nest boxes that are closer to the ground.

Q. Should we drill holes in the bottoms of the boxes to improve ventilation and improve hygiene?

A. We have experimented with drilling holes in the bottom of nest boxes with the idea of increasing ventilation and limiting moisture build up, and therefore limiting mite and hygiene problems within the nest. The holes were drilled beneath the nests themselves (not in the hallway). However, hihi avoided these modified nest boxes, negating any positive effect the holes may have had on nest box hygiene.

Q. Could we laminate the inside of boxes to make them easier to clean?

A. In one season on Tiritiri Matangi Island we trialled a nest box design where the inside of the nest box was covered in Formica laminate. The hope was that this would make the nest boxes easier to clean and prevent the accumulation of pathogens. However, we found that when there were chicks in the nest, a very large amount of moisture built up inside the laminated boxes. The level of moisture inside the boxes was so great that large drops would gather on the roof of the box and fall onto the nestlings below. We now recommend using wood without lamination, as this is breathable.

Q. How long do nest boxes last before they need replacing?

A. Well-built nest boxes may last many years before they are unusable. However, a primary consideration with hihi management is that the nest boxes provide a low-pathogen environment in which birds can nest. As wooden nest boxes age, they will crack and warp and present opportunities for bacteria, viruses and fungi to be harboured. Because of this, we recommend replacing nest boxes more frequently than their purely functional lifespan demands: on Tiritiri Matangi we are working to about once every 5 or 6 years.

3.4 Suggested further reading

Lindsay, K.; Craig, J.; Low, M. 2008: Tourism and conservation: the effects of track proximity on avian reproductive success and nest selection in an open sanctuary. Tourism Management 29: 730–739.

This study provides important evidence that the proximity of nest boxes to public walking tracks on Tiritiri Matangi Island does not impact on hihi reproductive success. This is an important finding for a site like Tiritiri Matangi that sees 20,000 visitors annually and performs an important advocacy role.

Richardson, K.M.; Ewen, J.G. 2016: Habitat selection in a reintroduced population: social effects differ between natal and post-release dispersal. Animal Conservation DOI: 10.1111/acv.12257.

This study investigates factors influencing post-release dispersal and breeding site selection of hihi at Maungatautari and differences between these released juveniles and those that were locally born. Importantly, it shows that hihi prefer to establish breeding territories in gully systems near streams.

Hihi Best Practice Sheet 1: Cleaning nest boxes

Nest boxes should be cleaned at the start of the breeding season, whenever a nesting attempt has finished (either because the chicks have fledged or it has failed), and whenever a mite infestation necessitates nest replacement (see **Hihi Best Practice Sheet 12** (Chapter 7) for nest replacement procedure due to mite infestation). At the start of the breeding season nest boxes can be cleaned in situ. At other times the nest boxes should be removed to the hihi service area for thorough cleaning. The recommended procedures for these two scenarios are detailed below, with **necessary equipment** highlighted in bold.

To clean nest boxes at the start of the breeding season:

1. Remove the nest box from its backboard.
2. Empty any contents of the nest box about 10 m away from the nest box location. Use a stick to scrape out most of the loose contents. Be careful that you are not removing a partially built new nest. The most likely content of boxes (if any is present) will be a few sticks and some faecal material from birds that have used the boxes as roosting sites during the non-breeding season. If a partially built nest is found it should be left and monitored to determine if the breeding pair has commenced breeding.
3. Using a **bottle** filled with **tap water**, douse the nest box and the backboard and remove any faecal material etc. with a **scrubbing brush**.
4. Return the nest box to its backboard.

To clean nest boxes when a nesting attempt has finished or when a mite infestation necessitates nest replacement:

1. Remove the nest box from its backboard.
2. Empty the contents of the nest box about 10 m away from the nest box location. Use a stick to scrape out most of the loose contents.
3. Using a **bottle** filled with **tap water** douse the backboard and remove any faecal material etc. with a **scrubbing brush**.
4. Spray the backboard using a **spray bottle** containing **SteriGENE®** solution at a concentration of 1 part SteriGENE® to 100 parts water. Leave for 10 minutes before rinsing with water.
5. Return the nest box to the hihi service area and rinse all surfaces of the nest box using a **high-pressure hose**.
6. Using standard **washing-up liquid**, fill a large sink with hot soapy water and scrub all surfaces of the nest box with a scrubbing brush.
7. Rinse off the soap suds in the sink, and then spray all surfaces of the nest box using a spray bottle containing SteriGENE® solution at a concentration of 1 part SteriGENE® to 100 parts water.
8. Leave SteriGENE® on the nest box for 10 minutes and then rinse using the high-pressure hose.
9. Leave to dry in the sunshine. Once dry, the nest box is ready to be returned to its location.

4. Supplementary feeding

Supplementary food is currently provided to all reintroduced hihi populations. Research has been conducted to identify whether supplementary food is needed, and what the most suitable supplement is (Box 4.1). Sugar water solution is currently the recommended supplement. The supplementary sugar water is provided at specially designed hihi feeding stations that require regular re-filling and maintenance. This chapter details all recommended procedures relating to the construction of feeding stations, the provision of food, and the hygienic maintenance of both feeding stations and feeding bottles. Health and disease risks associated with supplementary feeding are addressed in Box 4.2.

4.1 Feeding station design

Different feeding station designs have been in use at different sites, and each design has had its advantages and disadvantages. We are in the process of replacing these older designs with a new one (see Fig. 4.1). A good hihi feeding station will:

1. Allow hihi safe and easy access whilst excluding larger nectar-feeding species such as tūī and kākā
2. Allow ample internal space, multiple entrance/exits and multiple feeding points within each station, particularly when stations are shared with more-dominant bellbirds
3. Be made of a material that limits the accumulation and spread of pathogens
4. Be easy to clean and disinfect during regular cleaning and maintenance

Figure 4.1. A. Original wooden feeding station. B. Aluminium feeding stations on Kapiti Island. C. Latest design (stainless steel) of hihi bird feeder on Tiritiri Matangi Island. D. The set-up for collecting rainwater to make the sugar water solution on Kapiti Island. *Photos: A,B & D, John Ewen; C, Alex Knight.*

5. Allow observation of birds whilst they are inside the feeding station, to read band combinations and identify injury or illness
6. Allow the safe and easy capture of individual hihi using the feeding station
7. Permit the quick and easy change of feeding bottles kept within the feeding station

Our new feeding station design is produced by DOC and if you would like details on construction, or wish to place an order, then please contact the Hihi Recovery Group chairs.

4.2 Feeding station location

Multiple feeding stations should be available at any given time. As a guide, the Tiritiri Matangi population in 2012 had six feeding stations and an adult population of about 200 individuals. The Kapiti Island population in 2012 had ten feeding stations during the breeding season and six during the non-breeding season, with an adult population of about 100 birds. Ideally, feeding stations should be relatively evenly distributed throughout the site where hihi occur. However, at very large sites this may not be practical. For example, until 2017 at Maungatautari there were six feeding stations and these are all located within the Southern Enclosure. An important consideration with feeding station placement is the subsequent ease of servicing these with food and for cleaning. Placement near service tracks is advisable, if possible. Most feeding stations on Tiritiri Matangi, for example, are easily serviced with the use of a 4WD ATV and the six feeders spread over this 220 ha site can be serviced (cleaned and sugar water changed) in approximately 1.5 hours.

Feeding stations should be located in sheltered, semi-shaded positions under the forest canopy, and should not be exposed to direct sunlight for extended periods. They should not be within 30 m of a hihi nest site, as nesting birds may monopolise the food source, or females may face increased harassment by extra-pair males during breeding. At least some feeding stations should be located near to public footpaths, to create opportunities for members of the public to view hihi. There is some concern that feeding stations may congregate birds and, importantly, their pathogens. Where possible, it may be appropriate to rotate the use of feeding stations so that any given station is not in continuous use for extended periods of time. Closing feeding stations, by not putting sugar water out, will temporarily halt the flow of birds through them, and this may allow any pathogen build up at these stations to reduce to natural levels. The frequency of rotation to achieve this is unknown but if stations were paired (i.e. dual stations separated by short distances), then a fortnightly cycle may be appropriate.

4.3 Feeding bottle options

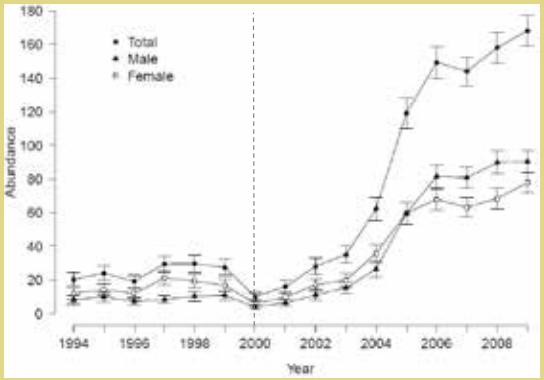
Inside the feeding stations, supplementary sugar water is provided in feeding bottles. Two varieties of feeding bottles are currently used. The first is a plastic 1.5 L drink bottle screwed into a hummingbird feeder base. The plastic drink bottles are simply recycled 1.5 L drink bottles, and the hummingbird feeder bases can be ordered from the Supporters of Tiritiri Matangi shop (www.tiritirimatangi.org.nz/shop). The second version of hihi feeding bottle is a chicken drinker, in either 3 L, 5 L or 14 L sizes, which can also be ordered from the Supporters of Tiritiri Matangi shop or from local pet stores or agricultural supply stores. Both options are similarly priced. The merits of these alternative feeding bottles are detailed in Fig. 4.2 below.

Box 4.1 An adaptive approach to food supplementation in hihi

The failure of early hihi reintroduction attempts gave rise to a hypothesis linking reintroduction failure to poor food availability in modified habitats (Armstrong & Perrott 2000). The provision of supplementary food in many reintroduced populations has been used to test this hypothesis. Experiments showed that provision of carbohydrate nutrients, in the form of sugar water solution, failed to enhance adult condition or survival in one population on Mokoia Island (Armstrong & Perrott 2000) but did in another on Tiritiri Matangi Island (Armstrong & Ewen 2001). Similarly, the provision of carbohydrates enhanced carrying capacity in a third population on Kapiti Island, with an initial strong and positive demographic response involving better survival and recruitment (Chauvenet et al 2012; Fig. 4.1a) and increased female reproductive success at Maungatautari (Doerr et al. 2017). Study of different nutrient supplements has shown a sex-specific cost of protein and fat supplements to male nestlings on Tiritiri Matangi (Walker et al 2013) and that carotenoids assist nestlings' ability to cope with common ectoparasites, again on Tiritiri Matangi (Ewen et al 2009). One cost of management success from ad libitum feeding has been an over-commitment of the manager's ability to provide food supplements, as has occurred on Kapiti Island (Chauvenet et al 2012), leading to closely monitored reductions in food provisioning and the exploration of alternative methods to optimise feeding (Ewen et al. 2015).

The choice of what to feed hihi on Mokoia Island involved application of adaptive management (Armstrong et al 2007). This formal process involved iterations of alternative supplements, and management of nestling ectoparasites, over a period of 8 years. Population demographic responses to (i) control of nestling ectoparasites, (ii) provision of carbohydrates, (iii) provision of protein, fats and carbohydrates (in the form of Wombaroo™ Lorikeet and Honeyeater Food) and (iv) a combination of either of these supplements with nestling ectoparasite control were monitored. The results indicated substantially improved population growth if some form of supplementation was used, but still some uncertainty in positive population growth, especially in the absence of additional ectoparasite control (Armstrong et al 2007; Fig. 4.1b). In addition, it was shown that there was very little added benefit from feeding Wombaroo™ supplements, a finding that was also supported by research on Tiritiri Matangi island (Walker et al. 2013). Given Wombaroo™ is more expensive than sugar and does not remain fresh as long in feeding stations, particularly in hot weather, we currently do not recommend its use. The uncertainty in population recovery of hihi on Mokoia Island, combined with the manager's low risk tolerance for a negative outcome, resulted in a decision to stop management and to remove the remaining birds to another population.

A.



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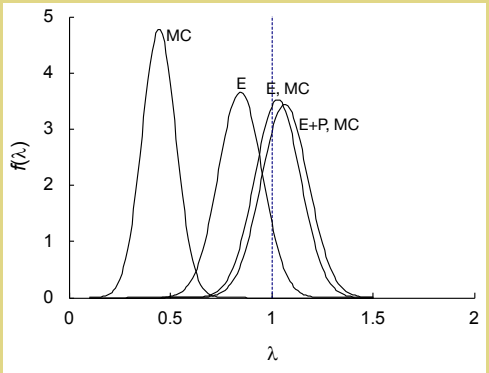

Figure 4.1. A. There are indications of a strong positive demographic response to providing carbohydrates on Kapiti Island, where hihi abundance rapidly increased following establishment of feeding stations in 2000 (shown by dotted vertical line; for details see Chauvenet et al. 2012). B. Using adaptive management, Armstrong et al. 1997 provided strong evidence that providing some form of supplementation would substantially increase population growth. However, regardless of the type of supplement fed, there remained high uncertainty in population growth leading to population recovery on Mokoia Island. MC = ectoparasite control, E = carbohydrate supplementation, P = protein, fat and carbohydrate supplementation (for details see Armstrong et al. 2007).

Box 4.2 Health and disease risks of supplementary feeding

There is considerable concern that providing wild birds with supplementary food exposes them to greater health and disease risks (Robb et al 2008). This is a concern that applies as much to endangered species in management programs as it does to garden birds provisioned with peanuts. For example, disease transmission at feeders has been implicated in the spread of mycoplasmal conjunctivitis in US house finches (*Carpodacus mexicanus*) (Altizer et al 2004), and in the spread of *Salmonella* and *Trichomonas* in UK finch species (Robinson et al 2010; Lawson et al 2010). In these cases, it is speculated that the use of feeding stations increases the contact rate between susceptible and infected hosts (and their contaminated food) and thus facilitates pathogen spread. Major causes of mortality in hihi populations include aspergillosis (Alley et al 1999; Perrott and Armstrong 2011), a fungal disease caused by *Aspergillus fumigatus*, and salmonellosis (Ewen et al 2007), a bacterial infection caused by *Salmonella* bacteria. We have so far found little evidence that feeding stations pose a significant disease risk to hihi populations. Nevertheless, it seems sensible to be aware of health and disease risks and act to minimise these, and this is the justification for our current best practice protocols.

4.4 Sugar and water

Raw sugar is used to make the sugar water solution. Chelsea Sugar kindly provides raw sugar free-of-charge to Tiritiri Matangi Island. At Bushy Park, Pam's raw sugar is kindly provided free-of-charge by Whanganui Pak'nSave.

It is important that the water used to make supplementary sugar water is as clean as possible. The water tanks supplying the water should ideally be fitted with filters to guarantee a clean supply. On Tiritiri Matangi, rainwater is filtered to 1 micron and UV treated. At Maungatautari the water, obtained from a mountain stream, is filtered twice, firstly using a 20 micron 'primary filter' and then a secondary filter of 1 micron. These filters work without electricity (hence no UV-filter operation), are available in many brands and work well if serviced regularly (at Maungatautari this means cleaning the primary filter and replacing the secondary filter at least every 6 months). Where filters are not possible, alternative options are used. For example, on Kapiti Island, 500 L water tanks are located near feeding stations and collect rainwater from platforms above the tanks. Unused water in these tanks is emptied once a month and the tanks are cleaned, unless drought conditions require more careful water management.

The sugar water solution at feeding stations should be provided ad libitum, unless there is an experiment or adaptive management process in place that controls how much or when it is made available. As a result, the amount of sugar water provided will often change on a day-to-day basis to meet the demand of the birds. Feeding stations should never be empty of sugar water (excluding any planned fortnightly closures; see feeder rotation above), and any refill of sugar water should never be left out for longer than 2–3 days. For example, on Tiritiri Matangi feeders are changed every second day and this requires a visit to feeding stations at least every other day to replace old sugar water with fresh solution. At periods of extremely high demand feeders can require changing daily.

Plastic bottle with hummingbird feeder base

Photo: Leila Walker.

Chicken drinker

Photo: John Ewen.

Positives

- Limited sugar water exposure to air, which may reduce evaporation in hot weather, and may limit access to wasps
- More robust than chicken drinker
- Maximises the volume of sugar water that can be put out
- Easier to clean than hummingbird feeder
- Provides easy access for all hihi, which may reduce the ability of some male hihi and dominant bellbirds to monopolise access

Negatives

- Volume of sugar water in container is less than in chicken drinker
- Harder to clean than the chicken drinker
- Greater sugar water exposure to the air, which may increase evaporation in hot weather, and allow easy access by wasps
- Hihi can and do poo directly into the sugar water unless a cover is fitted
- Less robust than the hummingbird feeder

Figure 4.2. Options for sugar water feeders.

4.5 Q & A: Why do we do it this way?

For a general description of the reason for this Q & A section, see Chapter 1. The following set of questions and answers capture some of the recent trials and changes relating to supplementary feeding:

Q. What can be done to prevent wasps taking over the feeding stations?

A. Wasps like sugar water, and occasionally take over feeding stations to the exclusion of hihi. In the past this has been a particular problem on Kapiti Island. Certain practices may be adopted to keep wasp activity around feeders to a minimum. For example, sugar water may be provided in feeding bottles that limit wasp access (see Feeding Bottle Options above), and care should be taken to limit excessive spillage of sugar water on and nearby feeding stations. Placement of a wasp trap (e.g. Victor wasp trap or similar) under feeding stations can reduce wasp numbers at individual stations. In the event that German or European wasps do start to dominate at feeding stations, it is advisable that the surrounding area be searched for their nests. If the nests of these

invasive wasp species are found they should be safely destroyed. Wasp management continues to evolve and other options such as Vespex® may work. Always consider what impacts controls may have on hihi and other native species at the site.

Q. Do hihi prefer fresh sugar water?

A. Some field workers report that hihi prefer fresh sugar water over that which has already been at the feeding station for a day. Others report no obvious preference. Without a targeted experiment, we cannot currently give a definitive answer to this question. However, we can say with some certainty that very old sugar water (left out for longer than three days) is unlikely to be relished by hihi. Sugar water left out for this long, particularly in hot weather, will ferment. In addition, the longer sugar water is left out at feeding stations, the greater the opportunity for exposure to potentially harmful pathogens.

Q. How long can sugar water be left out for?

A. Sugar water should never be left out long enough for it to ferment. We recommend that sugar water be left out at feeding stations for no longer than 3 days. The time to fermentation may be shorter than this in hot weather and, if necessary, sugar water should be changed more frequently than this. Ideally, sugar water should be put out at feeding stations as soon as it is made, and not be left sitting in a hot shed (or similar) for an extended period of time.

Q. What causes sugar water consumption to increase/decrease?

A. Knowing what drives fluctuations in hihi sugar water consumption would make it easier to provide appropriate quantities and pre-empt any surges in demand. Some field workers have reported that consumption increases immediately after heavy rain, although this is yet to be confirmed. It has also been suggested that certain times of the year (e.g. around the time of the post-breeding survey at some sites) are associated with reduced demand. Sugar water demand is probably influenced by a number of factors, including natural food availability, weather events and the time of the year (season).

Q. What is the best volume drinks bottle for providing sugar water in?

A. When using the plastic bottle and hummingbird feeder base set up, bottles any larger than 1.5 L tend not to be supported very well by the bases. 1.5 L therefore tends to be the optimal volume for maximising sugar water provision while not compromising feeder stability. If demand is particularly low, sugar water may be provided in smaller bottles or, alternatively, a lesser volume of sugar water may be provided in the 1.5 L bottles. The chicken drinkers are available in 3 L, 5 L and 14 L volumes, all of which are good for maximising sugar water provision.

Q. What have been bad features of previous feeding station designs to be avoided in the future?

A. These are some features of previous feeding station designs that have not been successful:

- An excess of solid, flat areas beneath the feeding bottles that collect faecal material and harbour pathogens.
- Operators not having a clear view of the entrance holes and landing perches while triggering catching cages. There is a risk of injuring hihi (and bellbirds) that are in the way of the closing door and it is difficult for operators to easily read bands and identify individuals.
- Hanging feeders in cages can tilt when birds land on them, causing sugar water to leak out.

Q. What is the best material for feeding stations to be made out of?

A. deally, feeding stations should be made of a material that is easy to clean, and that limits the accumulation and spread of pathogens. In general, metal and plastic will be more suitable than wood. Our new feeders have removed wood from the design. With time, wood will weather and provide cracks and crevices that are both perfect for harbouring pathogens and difficult to clean.

Q. Why do we tip sugar water away from feeders, should we, and is 10 m far enough?

A. When changing sugar water at feeding stations, it is inevitable that some will be spilt when the bases are removed. Because it is largely impractical to prevent this loss (e.g. by collecting the sugar water from bases into other containers, perhaps), we instead recommend tipping this minimal amount away from feeders. The risk of discarding this small volume of old sugar water has not been quantified, but there is a possibility that it increases the risk of pathogen exposure. We do not know if a 10 m distance is far enough away to reduce this risk, but we consider this action a good balance between precaution and manageability. Wherever possible we recommend bringing left over sugar water back to the cleaning area for disposal rather than tipping large amounts in the forest. If this is not practical then moving well away from feeding stations is advised.

Q. Why do we feed raw sugar and not other forms of sugar or honey?

A. This remains a good question. There has been a suggestion that raw sugar is more natural or less refined. However, raw sugar from Chelsea is actually granulated sugar that has a syrup coating on each crystal, giving it the golden colour. It's made by dissolving, filtering and recrystallising the raw sugar produced by sugar mills. It probably has less syrup than brown sugar. Whether the syrup is a good or bad thing for hihi remains to be tested, although there is no obvious sign from current use that it is bad for them. Concern has also been raised about thiamine deficiency in nectar-feeding specialists resulting from their only feeding on nectar (Paton et al. 1983). Again, however, there is little knowledge on thiamine content of sugar (raw, brown or white) nor whether use of feeders reduces thiamine access gained from alternative food types that hihi can obtain from foraging. While this is an interesting issue to explore, there is presently little evidence to support this risk in hihi. Honey is not recommended because it increases the risk that bees transfer diseases to one another; plus, it is expensive.

4.6 Suggested further reading

Armstrong, D.P.; Perrott, J.K. 2000: An experiment testing whether condition and survival are limited by food supply in a reintroduced hihi population. *Conservation Biology 14*: 1171–1181

Armstrong, D. P., and J.G. Ewen. 2001. Testing for food limitation in reintroduced hihi populations: contrasting results for two islands. *Pacific Conservation Biology. 7*: 87–92.

These two articles highlight the fact that food availability may not always be the main or only factor constraining hihi survival. The differing consequences of supplementary feeding for survival on Tiritiri Matangi and Mokoia Islands emphasise that management strategies will often be site-specific.

Armstrong, D.P.; Castro, I.; Griffiths, R. 2007: Using adaptive management to determine requirements of re-introduced populations: the case of the New Zealand hihi. *Journal of Applied Ecology 44*: 953–962.

This paper sets out a fantastic example of how to implement adaptive management to help evaluate the merits of alternative management strategies on population growth. Details of the study are covered in this chapter in Box 4.1.

Chauvenet, A.L.M.; Ewen, J.G.; Armstrong, D.P.; Coulson, T.; Blackburn, T.M.; Adams, L.; Walker, L.K.; Pettorelli. N. 2012: Does supplemental feeding affect the viability of translocated populations? The example of the hihi. *Animal Conservation 15:* 337–350.

This paper uses a long-term dataset on Kapiti Island to evaluate the importance of supplementary feeding. On Kapiti, the volume of sugar water provided has increased dramatically since hihi were first introduced, and this paper provides important justification for supplementary feeding.

Doerr, L.R.; Richardson, K.M.; Ewen, J.G.; Armstrong, D.P. 2017: Effect of supplementary feeding on reproductive success of hihi (stitchbird, *Notiomystis cincta*) at a mature forest reintroduction site. *New Zealand Journal of Ecology*, in press.

This paper tested our belief that sugar water would provide reduced benefits to breeding hihi in more mature forest and we found that it was still clearly able to influence reproductive success.

Ewen, J.G.; Walker, L.; Groombridge, J.J. 2015: A recipe for success: improving supplementary feeding in species conservation. *Conservation Biology 29*: 341–349 (available online open access)

This essay provides a short overview of the use of supplementary feeding in conservation, including documented benefits and risks. It also offers a way of improving how we go about using this management tool and ensuring it is providing the benefits we would like.

Paton, D.C.; Dorward, D.F.; Fell, P. 1983: Thiamine deficiency and winter mortality in red wattlebirds, *Anthochaera carunculata* (Aves: Meliphagidae) in suburban Melbourne. Australian *Journal of Zoology 31*: 147–154.

Robb, G.N.; McDonald, R.A.; Chamberlain, D.E.; Bearhop, S. 2008: Food for thought: supplementary feeding as a driver of ecological change in avian populations. *Frontiers in Ecology and the Environment 6:* 476–484.

By focusing on the popular pastime of feeding garden birds, this review provides a broad overview of the kinds of issues that are also relevant for supplementary feeding programs. It is particularly valuable for highlighting the potential negative impacts of supplementary feeding, from increasing the risk of disease transmission to increasing predation risk.

Walker, L.K.; Armstrong, D.P.; Brekke, P.; Chauvenet, A.L.M.; Kilner, R.M.; Ewen, J.G. 2013: Giving hihi a helping hand: assessment of alternative rearing diets in food supplemented populations of an endangered bird. *Animal Conservation 16*: 538–545.

This paper takes a slightly different approach to the supplementary feeding question, by asking what the most appropriate supplement might be for developing hihi nestlings. Despite not finding any long-term effect of nestling supplementation, this study does identify important sex-specific short-term responses to alternative supplements

Photo: Claudine Laugrost.

Hihi Best Practice Sheet 2: Providing supplementary sugar water (from Tiritiri Matangi Island)

The recommended procedure for providing supplementary sugar water at feeding stations is detailed below, with **necessary equipment** highlighted in bold.

1. Determine how many **feeding bottles** are required given current demand. You will have a reasonable idea based on how much has been provided in the preceding days.
2. The sugar water solution is made up using **raw sugar** and **water**. In a large **mixing jug** (about 5 L volume), mix up sugar and water at a concentration of 250 mL (one metric cup) of sugar per 1000 mL of water. This roughly equates to a 20% by mass solution. Use a **stirring stick** and a small amount of warm water (e.g. 1 L warm water for 5 L of solution) to aid dissolving. Once dissolved, make up to 5 L with cold water. No lumps of sugar should be present in the solution and the final sugar water solution should not be warm when placed out.
3. Decant the sugar water solution into clean feeding bottles using a **funnel**. Screw **bottle caps** onto the bottles for transportation.
4. Mix additional sugar water solution as required and decant into feeding bottles.
5. Place filled plastic bottles into a **clean container** for transportation out to feeding stations. Also place the required number of **hummingbird feeder bases** or **chicken drinkers** into a container. Take a **second container** for bringing back dirty bottles, bases or chicken drinkers.
6. Transport filled plastic bottles, feeder bases or chicken drinkers to feeding stations.
7. At feeding stations, remove old plastic bottles or chicken drinkers from feeding stations. Place those that are empty directly into the container for dirty bottles. If any bottles still have sugar water remaining in them, estimate the volume that remains. Then, about 10 m away from the feeder, tip the bottle upside down to empty the contents of the feeder base, remove the feeder base, and screw on a bottle cap. Place bottle in container, so that remaining sugar water can be disposed of in sink.
8. With the old feeding bottles removed, the feeding station can now be cleaned according to **Hihi Best Practice Sheet 3**.
9. Once the feeding station has been cleaned, screw feeder bases onto new bottles. Alternatively, fill chicken drinkers from the bottles. With a quick, smooth motion (to avoid spillage) turn the new bottles or chicken feeders upright, place the required number in the feeding station, and tap each bottle to remove air bubbles.
10. Transport all dirty bottles and bases back to the hihi service area for cleaning (**see Hihi Best Practice Sheet 4**).
11. Record the volume of sugar water that is put out and remaining at each feeding station (see **Hihi Template Record Sheet 1**).

Hihi Best Practice Sheet 3: Cleaning feeding stations

It is extremely important that a hygienic supplementary feeding regime is maintained. The recommended procedure for cleaning the feeding stations is detailed below, with **necessary equipment** highlighted in bold.

1. Feeding stations should be cleaned regularly. Choice of how often to clean needs to be managed based on common sense and balancing time available with the level of feeder use and associated build-up of faecal material (i.e. once weekly at heavily used feeders on Tiritiri Matangi is encouraged). Frequency of cleaning may therefore vary between feeders and over time. We would, however, encourage cleaning be done once per month regardless of use. If a responsive cleaning regime is used then it is recommended that a diary is kept to record when cleaning is done. If an infectious disease emergence is occurring, with parasite transmission perhaps being facilitated by birds using feeders, then the recommended response is daily cleaning.
2. Remove all old feeding bottles from the feeding station as instructed in **Best Practice Sheet 2**.
3. Use **bottles** filled with **tap water** to douse all surfaces of the feeding station.
4. Spray all surfaces of the feeding station using a **spray bottle** containing **SteriGENE®** solution at a concentration of 1 part SteriGENE® to 100 parts water. You should expect to use about 0.5 L of SteriGENE® per feeding station.
5. Leave SteriGENE® on the feeding station surfaces for 10 minutes.
6. Scrub all surfaces of the feeding station using a **scrubbing brush**. Pay attention to all surfaces in order to remove sugar water residue and faecal material. Ideally, each station should have its own dedicated scrubbing brush.
7. Rinse off SteriGENE® and faecal material using bottles filled with tap water. You should expect to use about 1 L of water per feeding station
8. Return fresh feeding bottles to feeding station as instructed in Hihi **Best Practice Sheet 2**.

Hihi Best Practice Sheet 4: Cleaning feeding bottles and bases

It is extremely important that a hygienic supplementary feeding regime is maintained. The recommended procedure for cleaning the feeding bottles/chicken drinkers and feeder bases is detailed below, with **necessary equipment** highlighted in bold.

1. Once dirty feeding bottles/chicken feeders and feeder bases have been collected from feeding stations, they should be cleaned as soon as practically possible (preferably immediately).
2. Empty any sugar water that remains in dirty bottles into sink.
3. Rinse all feeding bottles/chicken drinkers and feeder bases under running water.
4. Using standard **washing-up liquid**, fill the sink with hot soapy water.
5. Use **bottlebrushes** to thoroughly clean feeding bottles/chicken drinkers and feeder bases. Pay particular attention to the screw of the bottle-neck, the inside of bottles, and the inside of feeder bases (they can be separated into two parts to achieve this). Leave to drain.
6. Empty sink and rinse any remaining soap suds from feeding bottles/chicken drinkers and feeder bases using running water.
7. There are two alternative options for the next step:

   Option 1: completely submerge all feeding bottles/chicken drinkers and feeder bases into a **large tub** of water containing **Milton** (at a concentration of 1 tablet per 5 L). Leave to soak for a minimum of 2 hours, but overnight if this is more convenient. Equipment sterilised in Milton solution does not require rinsing and is ready to be used immediately.

   Option 2: use a **spray bottle** to treat all feeding bottles/chicken drinkers and feeder bases with **SteriGENE®** solution (at a concentration of 1 part SteriGENE® to 100 parts water). After 5 minutes application time, rinse with water and leave to dry.
8. Finally, clean out the containers that were used to transport clean and dirty bottles. Rinse out with running water and scrub with hot soapy water before rinsing again and leaving to dry.
9. If soaking bottles in Milton: the large tub of Milton solution remains active for only 24 hours. This means that either the water should be changed for each feeder change or, where water restrictions are a concern, fresh tablets are added to the same tub of water used in consecutive feeder changes. In the latter we would still recommend changing this water once a week. Empty the old water down the sink.

Hihi Template Record Sheet 1: Sugar water consumption

This is an example of a record sheet used for recording sugar water consumption at supplementary feeding stations, and includes example data to illustrate how it should be used. A blank version for photocopying is included at the end of this handbook.

Site: Tiritiri Matangi			Year: 2013					
Feeding Station: Wattle Valley			Feeding Station: B1 (top)			Feeding Station: B1 (bottom)		
Date	Volume remaining (L)	Volume put out (L)	Date	Volume remaining (L)	Volume put out (L)	Date	Volume remaining (L)	Volume put out (L)
01/10	1	3	01/10	1.5	3	01/10	0.5	1.5
03/10	0.5	3	03/10	0.75	3	03/10	1	1.5
05/10	none	4.5	05/10	0.5	3	05/10	1	1
07/10	0.5	4.5	07/10	0.5	3	07/10	0.5	1
09/10	0.5	4.5	09/10	0.2	3	09/10	0.5	1
11/10	1	3	11/10	none	4.5	11/10	none	1.5
13/10	0.5	3	13/10	0.5	4.5	13/10	0.5	1.5
15/10	0.5	3	15/10	0.5	4.5	15/10	0.5	1.5
17/10	1.5	1.5	17/10	1.5	3	17/10	1	1.5
19/10	0.2	1.5	19/10	0.2	3	19/10	1	1
21/10	0.2	1.5	21/10	0.2	3	21/10	0.5	1
23/10	none	3	23/10	1.5	1.5	23/10	0.75	0.5
25/10	none	4.5	25/10	0.5	1.5	25/10	0.1	0.5
27/10	1	4.5	27/10	0.5	1.5	27/10	0.2	0.5
29/10	0.5	4.5	29/10	none	3	29/10	0.2	0.5
31/10	0.5	4.5	31/10	0.5	3	31/10	0.1	0.5
02/11	1.5	3	02/11	0.2	3	02/11	none	1.5
04/11	0.2	3	04/11	0.2	3	04/11	0.5	1.5
06/11	0.1	3	06/11	0.1	3	06/11	0.2	1.5
07/11	0.5	3	07/11	0.5	3	07/11	none	3
09/11	1.5	1.5	09/11	1.5	1.5	09/11	0.5	3
11/11	0.5	1.5	11/11	0.1	1.5	11/11	0.2	3
13/11	0.5	1.5	13/11	0.1	1.5	13/11	none	4.5
15/11	0.3	1.5	15/11	none	3	15/11	0.4	4.5
17/11	0.2	1.5	17/11	0.5	3	17/11	1.5	3
19/11	None	3	19/11	0.3	3	19/11	0.5	3
21/11	0.5	3	21/11	0.5	3	21/11	0.2	3

Instructions:

- This sheet should be filled in every time sugar water is changed at a feeding station.
- In the 'volume remaining' column, record the volume of sugar water that was removed from the feeding station (i.e. not consumed by birds).
- In the 'volume put out' column, record the volume of fresh sugar water that was put out (i.e. available to be consumed by birds). If any sugar water is being left in the feeding station from the previous day, this should be included (i.e. added) in this volume.

5. Catching and handling hihi

5.1 Catching hihi

There are a number of reasons why you might need to catch hihi. Initially, birds will need to be caught for banding. In some cases this will simply be a case of removing nestlings from their nest, but in other cases this will involve catching juveniles and/or adults. Occasionally, sick or injured birds will need to be caught in order to treat them or fix band injuries. Birds will also need to be caught during planned translocations. There are two techniques that can be used to catch hihi: mist netting and capture at supplementary feeding stations.

IMPORTANT: Catching birds in mist nets and in catching cages requires training and appropriate permits and should not be undertaken by untrained or unlicensed individuals.

Mist nets are made of a fine nylon mesh and are suspended between vertical poles held in place by guy ropes. When unfurled, mist nets are virtually invisible to birds, which fly into them, fall into the fine mesh pockets and become entangled. Trained mist-netters can then safely and relatively quickly extract entangled birds. The success of catching with mist nets is very much dependent on their placement. Firstly, the less visible the mist net is, the greater the likelihood that a bird will fly into it. For example, a mist net that passes through alternately sunny and shady patches will be more visible to birds than a net that is entirely in shade. Secondly, and fairly obviously, you are more likely to catch hihi if you place the nets where hihi are likely to fly. This may, for example, be a favoured feeding spot (e.g. a fruiting coprosma dripping with ripe berries), an often-frequented stream (particularly during dry weather), or simply the territory of an individual bird you are targeting.

Removing a hihi from a mist net.

The two older feeding station designs can both be modified for catching hihi. **Hihi Best Practice Sheet 5** details how to use the wooden and aluminium feeding stations as catching cages. Figure 5.1 illustrates different aspects of catching hihi. Our new feeding station design has a very similar catching method to that of the old wooden feeding stations with some modification and the **Hihi Best Practice Sheet 6** should be used.

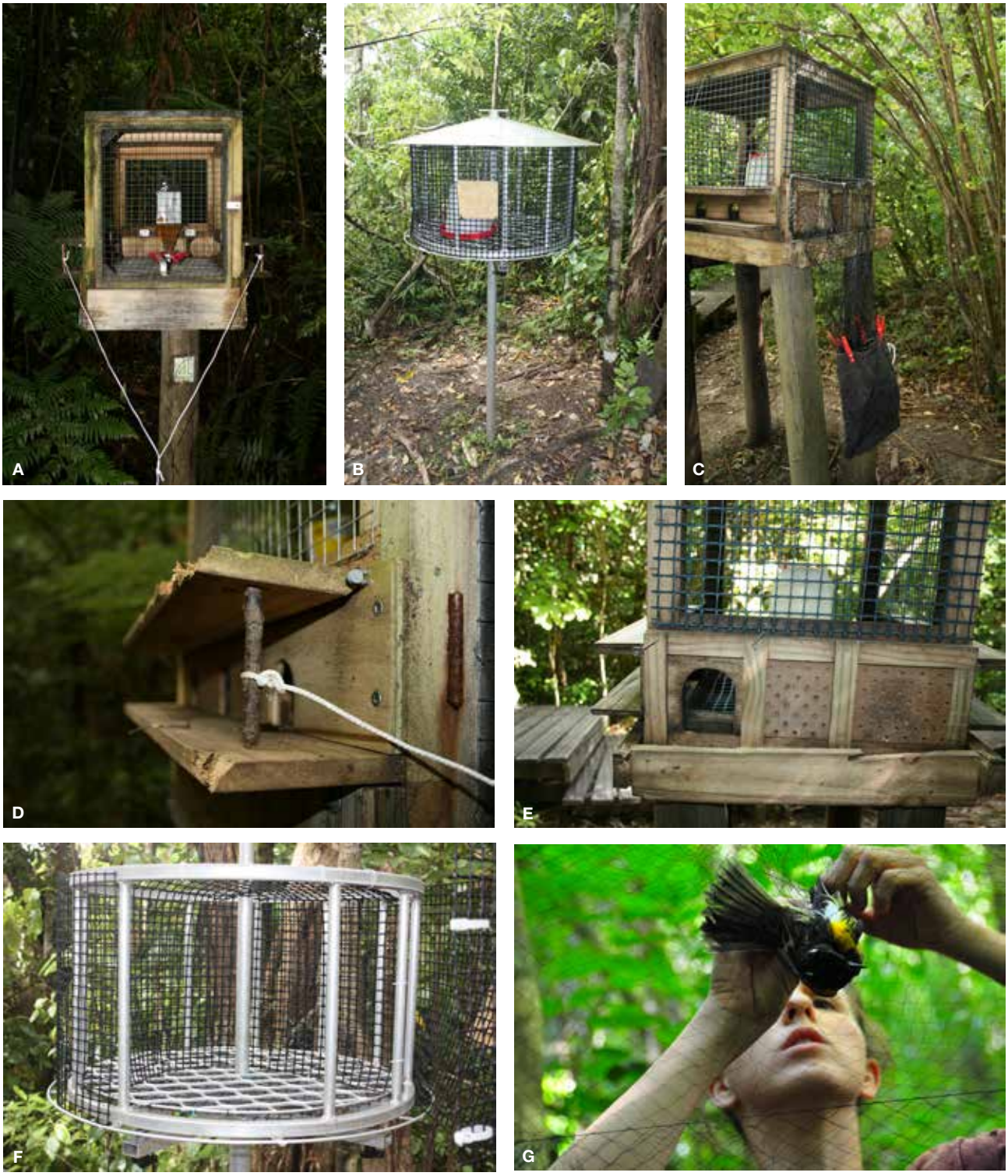

Figure 5.1. Catching hihi: A. Wooden feeding station modified to catch hihi, with sticks propping open the side-flaps and attached to a pull-string; B. Aluminium feeding station modified to catch hihi; C. A catching net with bird bag attached to the rear entrance/exit hole of a wooden feeding station; D. Propping open a side-flap on a wooden feeding station; E. The rear entrance/exit hole of a wooden feeding station, with removable door; F. An aluminium feeding station with mesh insert to modify for catching; G. A hihi being extracted from a mist net.
Photos: Leila Walker.

5.2 Handling hihi

IMPORTANT: Handling birds, including performing all of the techniques described below, requires training and appropriate permits, and should not be undertaken by untrained or unlicensed individuals.

The reasons why birds need to be caught vary, and all recommended procedures are detailed below.

Banding

An important aim of managed hihi populations is to have all birds banded with a unique combination of one metal and three coloured plastic leg bands. Where practical, this is achieved by finding all nests and banding all nestlings. Where this is not achievable, any unbanded individuals should be caught for banding as soon after fledging as possible.

Hihi should be banded with C-size split metal bands. These should only be obtained through orders to the DOC banding office (bandingoffice@doc.govt.nz) by a L3 bander. There are two metal band options: 1) C bands produced by Mekaniska which are 5.0 mm high and have an internal diameter of 3.5 mm and a wall thickness of 0.5 mm; 2) CP bands produced by Porzana which are 6.0 mm high and have an internal diameter of 3.3 mm and a wall thickness of 0.3 mm. Each ring has a unique reference number stamped on it, which becomes the bird's individual ID number. Small pliers (with 5 holes, suitable for closing bands from 2.0 to 5.5 mm in internal diameter) made by Porzana Ltd should be used to close Porzana Ltd metal bands. Non-Porzana pliers make closing Porzana bands difficult and it is important that metal bands are closed completely. If the gap is wider than the width of a fingernail, sticks and other vegetation can become caught inside the band, injuring the bird. It is important to check that the band is not wide enough to slip down over the first toe joints of the leg but is still loose enough to be rotated without scraping the leg.

On rare occasions, it may be necessary to remove a metal band that has become damaged or overlapped by accident. Extreme care needs to be taken in this process, as mistakes can cause broken legs. The bird's leg should be immobilised using the ring finger and thumb using the banders grip. The tips of the band removal pliers slip between the leg and band, just underneath where the band opens. By gently squeezing the band removal pliers the band should slowly open, with the leg feeling no pressure. Only experienced banders should do this.

In addition to one metal band, each bird should be banded with a unique combination of three double wrap-around or split-type plastic colour bands. Various plastic colour bands have been used over many years in hihi and each have good and bad aspects (see Q & A section at the end of this chapter). Double wrap-around plastic bands with an internal diameter of 3.5 mm are made by DOC and have been recommended; however, they are made in small quantities and currently orange is not available as a colour option. When ordering these bands from the banding office, you should request **double wrap C bands with an internal diameter of 3.5 mm for hihi**. Alternatively, C-size plastic split bands, with an internal diameter of 4 mm are also available through the DOC banding office. Split style plastic bands come in a wider variety of colour options and are commercially produced. Split style bands currently come in three plastic types (Darvic, Acetal and Celluloid) and each has a different capacity for staying tightly closed on a bird's leg as well as being resistant to fading. Recently, birds banded with split bands have suffered high rates of band loss. Plastic bands made from Darvic or Acetal are slightly stiffer and maybe less likely to fall off (although evidence is limited that this is the case). Darvic has the added advantage of not fading in sunlight. We recommend the use of the smaller diameter Darvic double wrap bands for hihi banding unless your population is sufficiently large that a larger range of colours are required (Tiritiri Matangi is currently the only population requiring this). The same type of band should always be used on an individual as mixing and matching band types has been shown to cause more issues with band injury and/or loss.

We are currently testing a new hihi-specific band. This will come with a recommendation to melt or 'weld' the bands closed using a pen-like battery-operated soldering iron for plastic split bands once they are applied to the birds so that the split edges are permanently joined. Early indications are that welding works well for of the new plastic and we are hopeful this will reduce the frequency of band loss.

All hihi should be banded with two colour bands on one tarsus, and one colour band above one metal band on the other tarsus. We do not recommend banding a bird with fewer than three colour bands (see Q & A section at the end of this chapter). The band combination is read in the following order: top left leg to bottom left leg to top right leg to bottom right leg. For example, a bird with dark green above metal on the left leg, and yellow above dark blue on the right leg would be read as dark green-metal-yellow-dark blue (i.e. DG MT–YE DB). The abbreviations for metal and colour bands are detailed in Table 5.1. We recommend two-letter abbreviations that will reduce mis-identification or ambiguity of colour (except for the three-letter codes used for the special PIT tags). For example, the abbreviation B could stand for blue or black and should therefore be completely avoided in place of BK for black and DB for dark blue and LB for light blue. This is especially the case when there is a high turnover in field staff and/or when volunteers are used to record combinations.

Table 5.1. Example of colour abbreviations used for bird bands on Tiritiri Matangi island.

BAND	ABBREVIATION
Metal	MT
Black	BK
Dark blue	DB
Dual (red + white)	RW
Dark green	DG
Pink	PI
Light green	LG
Orange	OR
Red	RD
Light blue	LB
Purple	PP
White	WH
Yellow	YE
Dual (green + orange)	GO
Black PIT tag	BKP
Grey PIT tag	GYP

When drawing up a list of band combinations to use in a given season, care should be taken not to re-use the combinations of birds that are currently alive. If combinations are being re-used, a clear record of this should be kept (e.g. by suffixing the combination with its issue number, such as DG MT–YE DB (2) for the second use of the combination DG MT–YE DB). In some situations it may be sensible to randomise the list of band combinations. For example, on Tiritiri Matangi, siblings from the same nest had previously been banded with colour combinations that differed only subtly (e.g. only one colour band different). This caused problems subsequently, in the event of band loss, when siblings became parents. In some cases, siblings with lost bands ended up having identical band combinations, and the resulting identity issues could not be resolved genetically, given that they also shared very similar genotypes. On Tiritiri Matangi, where the maintenance of a genetic pedigree is important for on-going research, we therefore recommend randomising the list of band combinations. We recommend, to avoid any confusion, that any data recording of colour combinations also includes the metal c-band associated with that colour band combination as this is the true permanent unique identifier for the bird.

Taking biometric measurements

There are a number of standard biometric measurements that can be taken for hihi:

- **Weight** – measure a bird's weight using either digital scales (to the nearest 0.01 g) or a pesola and bird bag (to the nearest 0.5 g). If using digital scales, an inverted cone placed inside a plastic container can be used to hold the bird in place (Fig. 5.2A).
- **Tarsus length** – there are two techniques for measuring a bird's tarsus length: Hold the leg with the tarsus at a right-angle to the tibia, and the foot at a right-angle to the tarsus. Use calipers to measure the length (to the nearest 0.1 mm) from the top of the foot to either i) the 'notch' of the intertarsal joint ('to-the-notch' tarsus length), or ii) the distal point of the intertarsal joint (full tarsus length; Fig. 5.2D).
- **Head-to-bill length (HB)** – use calipers to measure the length (to the nearest 0.1 mm) from the base of the bird's skull to the bill tip (Fig. 5.3).

Figure 5.2. Handling hih: A. Cone for weighing hihi using digital scales; B. Taking a blood sample from a newly banded male; C. A juvenile male complete with transmitter; D. Measuring tarsus length. *Photos: A, B, D – Luis Supangco Lachica; C – John Ewen.*

Figure 5.3. Measuring head-to-bill length of a male hihi, Te Hauturu-o-toi / Little Barrier Island. *Photo: John Ewen.*

These measurements should be taken as standard when banding nestlings (see 'processing nestlings' below). When individuals are banded as juveniles/adults (see 'processing juveniles/adults' below), it is not necessary to take these measurements unless they are being collected for a specific purpose/research project (e.g. during translocation). If time permits, then it adds little extra time to processing a bird and may eventually provide useful data for population comparisons.

Bleeding

Blood samples are usually taken from hihi as part of defined research projects (e.g. the maintenance of a genetic pedigree on Tiritiri Matangi), or for health screening purposes (e.g. during translocations). The recommended technique for bleeding hihi is brachial venipuncture (Fig. 5.2B). This allows the safe collection of a sufficient volume of blood for most purposes. If storing blood for later DNA extraction we prefer to preserve the sample in >80% ethanol with a ratio of at least 9× ethanol to 1× blood (then mix well).

Determining the age of hihi

If you are catching unbanded hihi, being able to determine their age as juvenile (hatched this breeding season, before their first moult), hatch-year (hatched in the previous breeding season, being no longer defined as juveniles after their first moult is finished around April (between roughly 5 and 12 months of age)) or adult birds (hatched at least two breeding seasons ago) can be very useful. Understanding moult is an essential part of being able to age birds. In hihi, moult data has been collected on Tiritiri Matangi. After growing all body and flight feathers in the nest, juvenile hihi undergo a partial moult (of body feathers and wing coverts only) starting in about early February. This juvenile partial moult appears to finish sometime in April. They do not undergo another moult until after their first breeding season, although a few juvenile hihi appear to moult a few primaries at some point during their first year. This moulting of some but not all of their wing coverts (and occasionally primaries or secondaries) creates what are called moult limits. Moult limits are two ages of feather on a bird after moulting has finished, often being obvious because of differences in colour or wear. Adults undergo a complete moult (of both body and flight feathers) once a year, starting in January and usually finishing sometime in March, causing them to not have moult limits because every feather is replaced. The time at which birds of different ages moult will have consequences for the degree of feather wear and this in addition to moult limits can be used, at most times of the year, to differentiate between juvenile, hatch-year and adult birds:

0 No wear. Feather edges perfect with entire edge light coloured, including tip.

1 Slight wear. Feather edges slightly worn with no fraying or nicks.

2 Light wear. Feathers definitely worn, but with little fraying or nicks.

3 Moderate wear. Considerable wear with definite fraying. Nicks and chips obvious.

4 Heavy wear. Feathers very heavily worn and frayed. Tips often worn completely away 1–2 mm.

5 Excessive wear. Feathers extremely ragged and worn. Shafts usually exposed beyond vane with all tips usually worn completely away.

Using the above descriptive scale of feather wear, the following tips can be used to age and sex hihi. Note that it will take some practice before you are able to confidently age birds using this technique. When catching birds of known ages it is worth taking time to familiarise yourself with these differences:

- It seems possible to accurately sex nestlings in the nest box when their primary coverts have grown enough to make their bases visible. Many can be sexed when they are banded at age 21 days, but smaller individuals may need another week before their coverts are grown enough. Birds with white at the base of their primary coverts are likely males. Those with no white on the base of their primary coverts are likely females (see Fig. 5.4). This is especially helpful for translocations in March, when juvenile males still look like females and may have not started to moult in black head feathers yet.
- Between mid-April and mid-December it is possible to differentiate between hatch-year and adults.
- Hatch-year males will show a moult limit either in their carpal coverts, alula, or sometimes among their primary and greater coverts. These are quite obvious in males as the juvenile feathers are brown and the hatch-year feathers are black (like adults). Occasionally, inner primaries (those closest to the secondaries) are also moulted and appear black next to the other brown feathers. Adults will not show these moult limits. Male tail wear is also very useful for aging and follows the same patterns as females (see Fig. 5.5).
- In females it is much harder to find moult limits, and they are more easily aged by wear of feathers. Juvenile female tail feathers in mid-April show a wear of 3, whereas adult female tail feathers at the same time show a wear of 1–2. Adult tail feathers have been grown more recently and are stronger/higher quality due to being grown one at a time

Figure 5.4. A. Female hihi wing, showing brown bases of primary coverts. Compare with B. Male wing which has white bases to primary coverts and darker feathers. *Photos: XXX*.

Figure 5.5. Aging hihi: A. Hatch-year male (in August) showing a moult limit between two black inner primary coverts and outer brown ones. The largest alula is also brown compared with the smaller two which are black; B. Adult male (in January) showing moderate wear in tail feathers; C. Adult female (in January) showing moderate wear in tail feathers; D. Hatch-year female (in January) showing excessive wear in tail feathers. *Photos: Chris Smith*.

and usually with good nutrition. This contrasts with juvenile tail feathers that are grown simultaneously in the nest under a time of stressful body growth. By September, the now hatch-year (previously juvenile) females have tail feathers showing a wear of 5, whereas the tail feathers of adults at the same time show a wear of 3 or (rarely) 4. The wear of the outer primaries in hatch-year and adult females follows a similar pattern, with hatch-year females often showing >4 mm broken off the tips of their outer primaries in November, whereas

adult females show wear of 4, with few short tips broken. It is important to note that the feather will look freshest in April and will gradually become more worn into January when the new moult starts. Birds of both ages will have much nicer feathers in August than in November (with hatch year's feathers more worn relative to those of adults).

- From January until February–March, any bird undergoing symmetric moult in their primaries and secondaries is likely an adult. Adults start moulting the primaries from the inside towards the outside of their wing. Newly moulted feathers will appear shorter than other wing feathers while growing, are fresher and darker, and should be moulting at similar feathers on both wings (symmetric). Moult can be seen even when feathers are in pin by blowing on the underside of the wing thus revealing the bases of the primaries.
- Hatch-year birds start their first adult moult (all wing feathers) in January (about a year after they are born) and after completing this moult are impossible to tell from older adults.

Attaching transmitters

IMPORTANT: Attaching transmitters to hihi requires training and appropriate permits, and should not be undertaken by untrained or unlicensed individuals.

Transmitters should only be attached to hihi for specific research or management projects, where the information gained from transmitter use cannot be obtained using other methods, and where it provides a clear and direct benefit to hihi conservation. Transmitters are most frequently used post-translocation to obtain data on survival, dispersal (particularly at mainland sites) and causes of mortality (particularly at sites where low levels of mammalian predators may be present).

To date, three different attachment methods have been used with hihi: back-mounted, tail-mounted, and harness. All methods come with advantages and disadvantages. Back-mounted transmitters will detach from the bird once new feathers grow through and push the transmitter off, and tail-mounted transmitters will detach from the birds when rectrices are moulted. Harnesses have a weak link built into their design such that they should eventually break and fall off the birds at some point after the battery has failed. Back-mounted transmitters require trimming of body feathers on the bird's back, which may have an effect on thermoregulation, whereas tail-mounted transmitters can cause premature loss of the rectrices they are attached to, which may incur an energetic cost to the bird as these feathers are replaced and/or affect flight performance. **Hihi Best Practice Sheet 7** outlines the general procedure regardless of attachment method, with specific detail for attachment using the tail-mount method.

Transmitters can be obtained from Holohil Systems Ltd (info@holohil.com). The model used (BD2) comes in a variety of weights. Previously, transmitters attached to hihi have weighed between 0.62 g (battery life approx. 21 days) and 1.2 g (battery life approx. 56 days). It is recommended that transmitters do not weigh more than 3% of a bird's body weight, and preferably less. Most recently, the 0.62 g model has been used for lighter female hihi, and the 0.75 g model (approx. 28 day battery life) used for male hihi. The attachment method will need to be specified at the time of ordering – if transmitters are to be tail-mounted, request hollowed tubing on units to allow units to be tied.

Frequencies used range from 160.120 to 161.110 MHz (TR4 channels 00–99). The use of frequencies between 160.600 and 161.110 MHz (TR4 channels 48–99) is being phased out and their use currently requires a temporary license from DOC (contact the Banding Office, bandingoffice@doc.govt.nz). Care should be taken to ensure frequencies selected for use do not overlap with frequencies used for monitoring other species at or close to the release site.

Transmitters should be ordered well in advance of the date required; but equally, care should be taken to ensure units are not stored for too long, as battery life will decrease. Transmitters should be checked upon initial receipt (by removing magnets) to ensure they are in working order, and

magnets should then be replaced and left attached until transmitters are ready for attachment. Transmitters can be stored in refrigerators to minimise battery life decrease during this time.

Dealing with injuries

Occasionally birds will suffer from band-related injuries. Injuries sustained from metal bands can be minimised by ensuring that bands are properly closed and that there is no gap between the butting ends. Injuries are more often caused by the plastic colour bands, and both wrap-around and split bands have been known to cause injuries in the past. If a bird is observed with a banding-related injury, it should be captured immediately and the band(s) causing the injury removed. Depending on the extent of the injury, it may be possible to replace the faulty band with a new one (so that the bird keeps the same colour combination). If the extent of injury means that the band cannot be replaced, the bird's new band combination should be recorded, including metal band number.

Dealing with sick birds

If an obviously sick bird is observed there are typically only limited options available. Please refer to chapter six for more detail and options. Box 5.1 addresses two health problems commonly observed in hihi

Box 5.1 'Tongue birds' and feather loss

There are two commonly observed health problems in hihi that we currently do not treat. Firstly, sublingual oral fistulas are lesions which occur on the floors of birds' oral cavities. A lesion becomes most noticeable when it causes a hole through which the bird's tongue protrudes (which is the reason why birds with the condition are sometimes called 'tongue birds' (image A below). Protruding tongues can become extremely long, curling round beneath the birds' mandibles (Castro & Taylor 2001; Low et al. 2007a). Research on Tiritiri Matangi has found that about 10% of adult hihi have some form of sublingual lesions, but that these were not present in nestlings (Low et al. 2007a). Having a lesion doesn't seem to have a negative impact on the birds except for sometimes changing foraging behavior when the birds need to extend their tongues. Supplementary feeders may assist these birds. The cause of the lesions is currently unknown. The second condition is a progressive feather-losing dermatitis (image B below) likely caused by ovoid burrowing mites identified as *Knemidocoptes* spp. (Low et al. 2007b). Interestingly, this condition is normally seen during the breeding season and is more common in males. Again, there is no noticeable impact on survival or condition from this parasite (Low et al. 2007b).

Two common health problems in hihi: A. Sublingual oral fistulas often become noticeable when birds' tongues protrude through them. B. A feather-losing dermatitis caused by burrowing mites. *Photos: Leila Walker.*

5.3 Processing nestlings

If nests are accessible, nestlings should be banded and have biometric measurements (weight, to-the-notch tarsus length, full tarsus length, head-to-bill length) taken when they are as close to 21 days since hatching as possible. At this age nestlings are developed enough to be handled and banded, but they are not so old that your presence at the nest is likely to prompt premature fledging. Standardising the day at which measurements are taken also allows comparison of body size and condition within and between years and, perhaps, across sites. For some projects, a blood sample will also be taken at this age. See **Hihi Template Record Sheet 2** for an example of how to record data collected when processing nestlings.

5.4 Processing juveniles/adults

If nests are not accessible, fledglings should ideally be caught for banding as soon after fledging as possible (dependent on monitoring needs). If possible, it is also helpful to collect biometric measurements (weight, to-the-notch tarsus length, full tarsus length, head-to-bill length) at this time.

5.5 Q & A: Why do we do it this way?

For a general description of the reason for this Q & A section, see Chapter 1. The following set of questions and answers capture some of the issues relating to catching and handling hihi:

Q. Does each bird have to be banded with three colour bands? Why can't we use just one or two colour bands?

A. We recommend using a complete set of colours to avoid any confusion if a bird subsequently drops a band. Dropped bands can lead to mis-identification of a bird in surveys if it then matches another bird banded with less than a full set of colours. By always banding with three colours it becomes quickly obvious if a band drop has occurred and the bird can be targeted for capture and band replacement.

Q. Why should biometric measurements be taken as standard in nestlings?

A. Given that nestlings can be banded at a standardised age and with relative ease we encourage taking the opportunity to record some basic biometrics. This data has proved useful for many projects on populations where the data has been recorded. Equally, we would also encourage some basic biometrics be taken whenever birds are being caught and handled, no matter what the age, but this has to be balanced by logistical constraints and other needs linked to the sampling event.

Q. Why do we take two repeated tarsus measurements?

A. Taking two measurements allows some assessment and control over the accuracy of these measurements. If a biometric measurement is difficult to measure or a particular field person has a very different technique for measuring (or is simply not well-practiced at taking measurements), then the data and their interpretation can become erroneous. Repeat measurements mean that measurement error can be included in the analysis so that differences between individuals can be more accurately quantified.

5.6 Suggested further reading

Melville, D.S. 2011: New Zealand National Bird Banding Scheme bird bander's manual. Department of Conservation, Wellington.

An obvious and essential aid for bird banders in New Zealand.

Armstrong, D.P.; Castro, I.; Alley, J.C.; Feenstra, B; Perrott, J.K. 1999: Mortality and behaviour of hihi, an endangered New Zealand honeyeater, in the establishment phase following translocation. *Biological Conservation 89*: 329–339.

This study describes an unusual problem caused by split type colour bands on hihi released onto Mokoia Island. This was corrected by changing the type of bands used. Although it was unclear why these split bands were problematic it does highlight that care needs to be taken when deciding to use bands and observation is required to ensure there are no major problems with whichever method you choose.

Splittgerber, K.; Clarke, M.F. 2006: Band-related leg injuries in an Australian passerine and their possible causes. *Journal of Field Ornithology 77*: 195–206.

Another cautionary tale of injuries resulting from colour banding which calls for attention to be paid to the relative size of a band compared with the diameter of a bird's tarsus.

Pyle, P. 1997. Identification guide to North American Birds Part 1. Slate Creek Press, USA.

Chris Smith (Tiritiri Matangi and Bushy Park hihi technician and moult expert) recommends the introduction text from this book as a very helpful guide to aging and scoring moult in birds. He notes that 'all North American banders read this text'.

Lessells, C.M.; Boag, P.T. 1987: Unrepeatable repeatabilities: a common mistake. *Auk 104*: 116–121.

How do we know that differences we see across years or sites in hihi morphometrics are true differences, or simply an artefact of our ability to measure them? This paper helps explains why we should test how repeatable we are in taking measurements. It shows the value of repeat measurements of hihi and how to account for variability within and between observers. This is important when using morphological data across individuals and sites for any form of comparison.

5.7 Additional literature cited in text

Castro, I; Taylor, J. 2001: Survival and reproductive success of stitchbird (hihi, *Notiomystis cincta*) suffering from a bill abnormality (oral fistula). *Notornis 48*: 241–244.

Low, M.; Alley, M.R.; Minot, E. 2007a: Sub-lingual oral fistulas in free-living stitchbirds (*Notiomystis cincta*). *Avian Pathology 36:* 101–107.

Low, M.; Alley, M.R.; Scott, I. 2007b: Pruritic facial dermatitis in a population of free-living stitchbirds. *Journal of Wildlife Diseases 43*: 262–268.


Photo: Martin Sanders

Hihi Best Practice Sheet 5: Catching hihi at wooden and aluminium feeding stations

Wooden

Wooden feeding stations can be modified to act as catching cages. The following procedure should be followed when catching at wooden feeding stations, with additional **necessary equipment** highlighted in bold.

1. Prop open the side flaps above the entrance holes using appropriate-length **sticks** tied to 5–8 m of **string**. Ideally, the flaps on both sides of the feeding station should be propped open, and only one length of string should lead away from the feeding station. You should aim to set it up so that when you pull the string, both flaps close simultaneously.
2. All wooden feeding stations have an additional entrance/exit hole at the back that is usually covered with a small section of wood. In some designs the section of wood can be slid sideways or upwards to reveal the entrance/exit hole. In other designs it is screwed in, and you will require a **screwdriver** to remove it. Do not remove this wooden cover yet, but you may need to loosen the screws (if present) in preparation for catching.
3. Have ready at hand some **mesh netting** attached to a **wire frame**, and attach a **bird bag** to the bottom of the mesh netting using **clothing pegs**. This frame can be hooked onto nails above the extra entrance/exit hole once a bird has been caught. Alternatively, it can be screwed into the wooden backing of the feeding station over the extra hole, but not before a bird has been caught, as its presence may make birds less inclined to go inside the feeding station.
4. Once you have propped open the side flaps, prepared your netting-bag device, and loosened the screws on the wooden panel (if necessary), you are ready to catch.
5. Sit about 5–8 m from the feeding station, with clear view of the entrance holes and landing perches on both sides of the feeder, holding the string.
6. When a target bird goes into the feeding station, give the string a single, hard pull to remove the sticks and therefore close the flaps and block all exit routes out of the feeding station. Ensure that no other birds are perched beneath the flaps before pulling the string.
7. Once your target bird is trapped in the feeding station, quickly and calmly position the netting-bag device over the extra entrance/exit hole and remove the wooden panel.
8. If your target bird is the only bird trapped inside the feeding station, you just need to wait until it finds the exit hole and tumbles down into the bird bag. If it takes more than a couple of minutes to do this, you can encourage it down that end of the feeding station by standing at the opposite end. If it gets caught in the netting on the way down to the bird bag, you may need to gently shake the netting-bag device so that the bird falls down. One tip for birds that take a long time going into the net is to place a dark jacket over as much of the station as possible, opposite the net hole, leaving the hole with the net over it as the only lighted area. When you do this, birds will often immediately head towards the lighted opening, and exit the hole into the net.
9. If there are a number of birds trapped in the feeding station at once, you will need to release non-target birds without releasing your target bird(s). To do this, you can wait for each bird to go into the bird bag in turn, releasing any birds you don't need and keeping your target bird(s). When unhooking the netting-bag device to remove birds from the bag, be careful to cover the exit hole so that birds still in the feeding station do not escape (unless they are birds you don't want to keep). If you are feeling more confident, you may try to release unwanted birds from the feeding station by lifting the side flaps. This of course risks releasing your target bird(s), and must be carefully timed.
10. Once your target bird is safely inside its bird bag, you should open the side flaps so that the feeding station can continue to be used while you process the captured bird.
11. Each bird should be caught and held in a clean bird bag, so ensure you have sufficient bird bags for your catching session.

Continued on next page

Best Practice Sheet 5 continued

12. When using the feeding station as a catching cage, try to avoid any actions that might dissuade hihi from entering the feeding station. This includes sitting too close to the feeding station, hanging the netting-bag device before birds are caught, and only propping open one of the side flaps. In general, you should try to ensure that the feeding station looks as similar as possible to when it is just being used as a feeding station.

Aluminium

As with wooden feeding stations, aluminium feeding stations can also be modified to act as catching cages. The following procedure should be followed when catching hihi at aluminium feeding stations, with additional **necessary equipment** highlighted in bold.

1. Insert **mesh inserts** into the feeding station. These act as a temporary internal partition that reduces the catching area within the feeding station to a manageable size.
2. Position one sugar water bottle within the section of the feeding station in which you will be trying to catch a bird.
3. Position a **wooden door** above the entrance hole to the relevant section of the feeding station. A **metal pin** threaded through the mesh should be used to hold the wooden door open above the entrance hole, and the pin should be attached to a length of **fishing wire** that leads away from the feeding station.
4. Sit about 5–8 m away from the feeding station, holding the fishing wire.
5. When a bird you want to catch goes into the feeding station, pull on the fishing wire to release the pin and drop the wooden door over the entrance hole.
6. With the bird trapped within a section of the feeding station, use the larger, removable door to put your arm inside the feeding station and catch the bird by hand.
7. Once caught, place the bird into a clean **bird bag** for processing.

Hihi Best Practice Sheet 6: Catching hihi at stainless steel feeding stations

Feeding stations can be modified to act as catching cages. The following procedure should be followed when catching at feeding stations, with additional necessary equipment highlighted in bold.

1. Prop open the side flaps above the entrance holes by replacing the paperclip with metal rods tied to **5–8 m of string**. Ideally, the flaps on both sides of the feeding station should be open, and only one length of string should lead away from the feeding station. You should aim to set it up so that when you pull the string, both flaps close simultaneously.
2. Feeding stations have an additional exit hole at the back that is usually covered with a sliding door, this door should be left closed at this stage. The **catch net** (netting attached to a wire frame) should be fixed to the back of the cage, it hooks over the frame above the exit hole.
3. Once you have propped open the side flaps, prepared your catch net and have a **bird bag** to hand you are ready to catch.
4. Sit about 5–8 m from the feeding station, with clear view of the entrance holes and landing perches on both sides of the feeder, holding the string.
5. When a target bird goes into the feeding station you must wait until it hops off the perch and is down in the cage at floor height. Then, give the string a single, hard pull to remove the metal rods and therefore close the flaps and block all exit routes out of the feeding station. Ensure that no other birds are perched beneath the flaps before pulling the string.
6. Once your target bird is trapped in the feeding station, quickly and calmly slide open the exit door, checking that the catch net is still in the correct position.
7. If your target bird is the only bird trapped inside the feeding station, you just need to wait until it finds the exit hole and jumps into the catch net. If it takes more than a couple of minutes to do this, you can encourage it out by covering the cage with **towels**, making the exit hole the only light source. When you do this, birds will often immediately head towards the lighted opening, and exit the hole into the net.
8. If there are a number of birds trapped in the feeding station at once, you will need to release non-target birds without releasing your target bird(s). To do this, you can wait for each bird to go into the catch net in turn, releasing any birds you don't need and keeping your target bird(s). When unhooking the catch net to remove birds from it remember to close the sliding door to cover the exit hole so your target bird does not escape.
9. Once your target bird is caught in the net, remove it as you would from a bird bag then place into a clean bird bag. Once the bird is safely inside its bag, you should open the side flaps and fix with paper clips so that the feeding station can continue to be used while you process the captured bird.
10. Each bird should be caught and held in a clean bird bag, so ensure you have sufficient bird bags for your catching session.
11. When using the feeding station as a catching cage, try to avoid any actions that might dissuade hihi from entering the feeding station. This includes sitting too close to the feeding station and only propping open one of the side flaps. In general, you should try to ensure that the feeding station looks as similar as possible to when it is just being used as a feeding station.

Hihi Best Practice Sheet 7: Attaching transmitters to hihi

Procedures in *bold italics* should be followed regardless of attachment method. Otherwise, procedure detailed is for attaching tail-mounted transmitters **only**:

1. ***Transmitter attachment requires two operators: one to attach the transmitter and an assistant with experience handling hihi to hold the bird.***
2. ***Prepare all materials and the working space prior to capturing the bird.***
3. ***Have on hand a pre-prepared data sheet with the following columns: date, start time, finish time, band number/combination, transmitter serial number, TR4 channel (fine tuning), fitted by, additional comments.***
4. ***Check the transmitter aerial is straight and that the signal is functioning properly. Ensure there are no sharp edges to the transmitter casing (smooth these off if necessary) and that the unit has no structural defects that may influence attachment.***
5. ***Obtain and record the fine tune frequency for the transmitter.***
6. Sand back (flat surface) of transmitter lightly using **sandpaper**.
7. Thread two 25–30 cm pieces of **dental floss** through both the top and bottom tubes of the unit, and then thread a **curved needle** through each end of the top piece of floss (Fig. 5.6).
8. Remove the bird from the bag. The person attaching the transmitter should be seated on one side of a desk (or flat and stable surface), with an assistant seated on the opposite side. The assistant should hold the bird with legs between the second and third fingers (photographer's grip), with their hand flat on the desk, and the tail of the bird facing the person attaching the transmitter and the bag covering the rest of the bird to keep it in darkness (Fig. 5.6).
9. Check the number of rectrices (there should be 12), and separate out the inner four (Fig. 5.6). It is important to check no feathers are missing or re-growing to ensure the transmitter is fitted evenly and centrally. If any of the inner four rectrices are missing/re-growing, transmitter attachment may not be possible, depending on which feathers are missing and the stage of regrowth.
10. Use a **crochet hook** to gently push down feathers away so the inner four rectrices are clearly visible down to the shaft. Slide a piece of **cardboard (similar size to a business card)** underneath and use a **hair clip** to pin cardboard to the remaining outer rectrices.
11. Place the unit of the transmitter over these four rectrices, so the top of the unit is almost at the base of the feathers, but not quite. Use the curved needles to thread the floss through the rectrices, alternately one side and then the other, and tie. It is particularly important to carry out this part of the process correctly, and this should be taught by an experienced operator (Fig. 5.6).
12. Remove the curved needles from the top floss, and thread through bottom floss. Loosely thread the bottom floss through the rectrices, but do not tie yet. Check that the transmitter is sitting evenly and centrally along the tail (Fig. 5.6).
13. Place superglue on shaft of rectrices where unit will sit, then press unit down. Tie the bottom floss.
14. Cut floss close to knot, and remove cardboard.
15. In some cases, the transmitter may be attached a number of days prior to monitoring starting (e.g. when there is a quarantine period between initial capture and release for translocation). After the transmitter has been attached it is possible to replace the magnet onto the transmitter (using the same tape it arrived with). A small number may subsequently become dislodged, but in previous attempts the majority have stayed attached and been removed during catch-up on translocation day, saving valuable battery life. To date this has only been carried out with tail-mounted transmitters.
16. ***After the transmitter has been attached, offer the bird a drink of sugar water or jam/honey mix prior to release and ensure all data has been recorded.***

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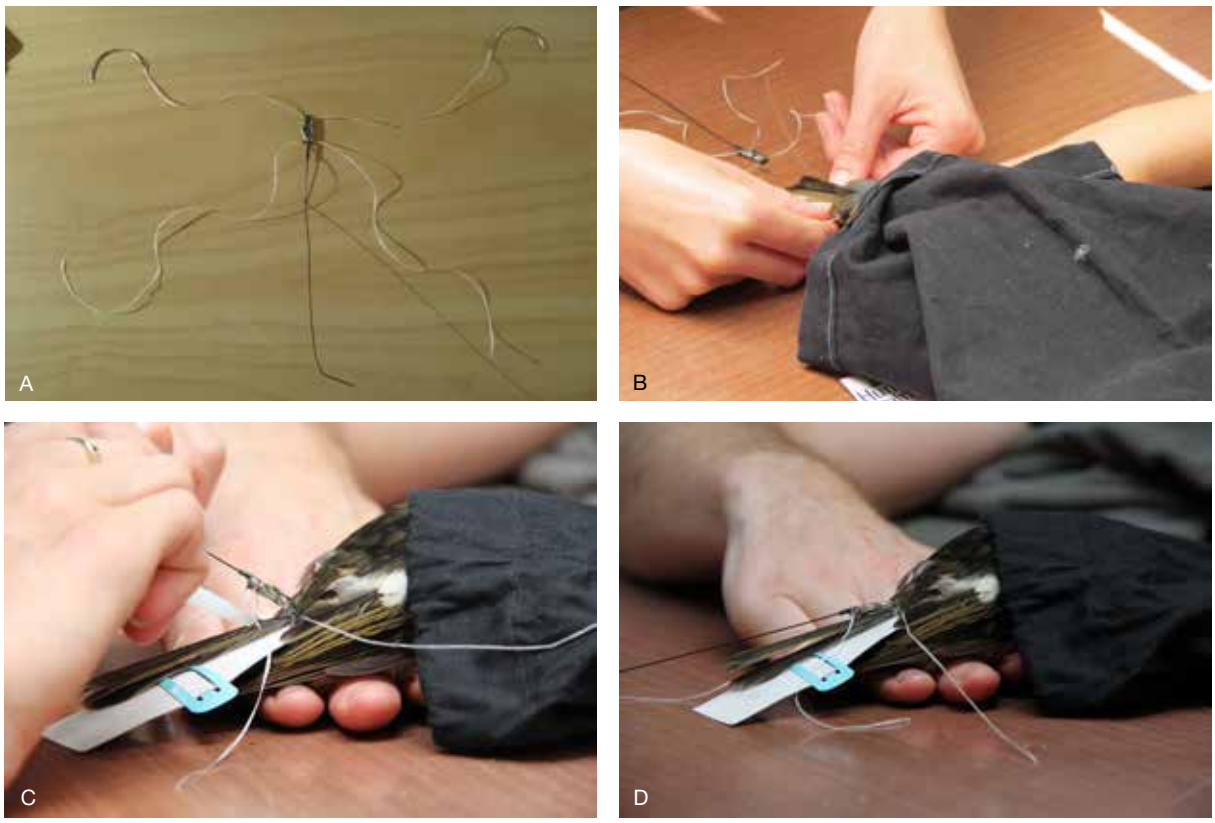

Figure 5.6. Attaching tail-mount transmitters to hihi: A. Preparing transmitter prior to removing bird from bag (steps 4–7). B. Counting rectrices and separating out the inner four (step 9); C. Checking the lie of the transmitter after tying top piece of floss, prior to threading bottom floss and supergluing unit (step 12). D. Transmitter attachment almost complete aside from cutting of floss and removal of cardboard (step 14). *Photos: unknown*.

Hihi Template Record Sheet 2: Nestling banding and measurement sheet

This is an example record sheet for recording the banding, measurement and bleeding of nestling hihi. This version includes example data, to illustrate how it should be used. Blank versions for photocopying are included at the end of this best practice guide.

Combo	C-band	Nest	Date	Age	Weight (g)	Tarsus (notch)		Tarsus (full)		Head-to-bill		Blood number	Slide
						1	2	1	2	1	2		
BKMT-YEDG	89101	1/28	30-Nov	21	34.50	26.6	26.5	31.0	30.9	38.0	37.9	1	Y
DGMT-DBPP	89102	21/4	02-Dec	21	37.42	27.2	27.3	31.1	31.0	38.5	38.1	2	Y
WHYE-DBMT	89103	21/4	02-Dec	21	45.59	27.2	27.2	32.2	32.2	40.4	40.5	3	Y
DGMT-DGBK	89104	22/24	02-Dec	21	42.49	28.9	28.8	33.0	33.1	39.8	39.9	4	Y
WHMT-PPPP	89106	22/24	02-Dec	21	41.87	26.7	26.7	32.4	32.4	39.6	39.5	5	Y
DBMT-PPBK	89107	22/24	02-Dec	21	42.41	23.0	23.0	32.3	32.4	39.7	39.8	6	Y
RDMT-DBDG	89108	22/8	03-Dec	21	38.45	27.6	27.7	30.9	31.0	38.4	38.3	7	Y
RDMT-ORYE	89109	22/8	03-Dec	21	43.58	28.2	28.2	32.9	32.9	40.4	40.3	8	Y
RDMT-PPPP	89110	22/8	03-Dec	21	39.56	29.1	29.1	33.9	33.8	39.4	39.3	9	Y
DBMT-RDYE	89111	22/8	03-Dec	21	42.39	28.8	28.7	33.0	32.9	39.9	39.8	10	Y
DGDG-DGMT	89112	22/26	04-Dec	21	46.99	28.7	28.7	31.9	31.9	38.8	38.8	11	Y
YEMT-YEDG	89113	22/26	04-Dec	21	39.13	26.4	26.3	30.6	30.6	38.0	38.0	12	Y
RDDG-RDMT	89114	22/26	04-Dec	21	47.28	27.7	27.7	32.4	32.4	39.8	39.8	13	Y
RDWH-YEMT	89115	22/26	04-Dec	21	28.79	24.7	24.7	28.9	28.9	35.3	35.3	14	Y
RDMT-WHRD	89116	WR/4	05-Dec	21	36.66	25.8	25.8	31.1	31.1	37.2	37.2	15	Y
DBOR-DGMT	89117	WR/4	05-Dec	21	39.65	27.8	27.8	31.7	31.7	37.4	37.4	16	Y
DBMT-WHRD	89118	WR/4	05-Dec	21	36.39	26.7	26.7	31.0	31.0	39.4	39.4	17	Y
RDMT-WHWH	89119	1/3	05-Dec	21	40.31	28.2	28.2	33.1	33.1	38.6	38.6	18	Y
DBMT-WHDG	89120	1/3	05-Dec	21	29.74	27.0	27.0	31.0	31.0	37.0	37.0	19	Y
DGMT-PIBK	89121	1/3	05-Dec	21	30.45	26.9	26.9	31.6	31.6	36.4	36.4	20	Y
PIRD-RDMT	89122	1/3	05-Dec	21	34.23	25.7	25.7	29.8	29.8	36.0	36.0	21	Y
PPMT-PIRD	89123	LHV/2	06-Dec	21	34.26	26.5	26.5	31.7	31.7	38.7	38.7	22	Y
DGMT-DGDG	89124	LHV/2	06-Dec	21	24.58	26.5	26.5	29.6	29.6	35.5	35.5	23	Y
DBYE-DGMT	89125	LHV/2	06-Dec	21	29.95	26.9	26.9	31.1	31.1	38.7	38.7	24	Y

Instructions:

- The colour combinations to be used should already have been generated at the start of the breeding season, preferably with the order randomised. Double-check that the bands have been put on in the correct order.
- Once a bird has been banded with its metal band, record the metal band number under the C-band column. Ensure the correct number is recorded.
- Record the nest id, the date of banding, and the age of the nestlings when they are banded.
- Record weight to nearest 0.01 grams if using digital scales, or to nearest 0.1 grams if using a pesola.
- Take two measurements within 0.1 mm of tarsus length (both methods) and head-to-bill length.
- If blood samples are being taken, record the blood sample number, and whether or not a slide has been made.

6. Dealing with sick hihi

Dealing with sick hihi requires a tricky balance between ensuring the welfare of individual birds, protecting the wild population, satisfying legal requirements around holding wildlife and managing the expectations of people regarding assistance for sick wildlife.

Valuable information can be derived from veterinary examination and diagnostic testing of sick birds or necropsy of dead birds (see boxes 6.1 and 6.2). This information can have significant implications for future management of the species in general or at a specific location, and obtaining it should be given a high priority.

Some options are discussed below. Consider carefully the requirements and outcomes of each option when you are deciding what to do with a sick hihi.

What is the best option may vary, so be prepared to assess each event as an individual occurrence.

6.1 Important considerations for holding sick birds

Disease risk management

- Threats to biosecurity are minimised if birds can be managed on location. For example, to prevent introduction of disease, your site may follow a 'no returns policy', meaning that if a bird leaves the site, it will not be returned regardless of the success of its treatment. In this case euthanasia is likely the only option if the treatment cannot be carried out onsite.
- Veterinary clinics contain other species and therefore may represent a hub for disease transmission; however, they also undertake good hygiene and disinfection of facilities to manage this risk. Additionally, clinics may be able to set up a quarantine room for hihi to isolate them from other animals and thereby minimise disease risks. Risks around contracting disease can be managed if required and site policy and veterinary quarantine practices should both be discussed during the decision-making process.

Diagnosis and learning

- Veterinarians can undertake detailed examinations and diagnoses in an attempt to determine the cause of illness and the best treatment to save the bird. They have skilled staff and appropriate equipment to provide the treatments, and to provide for the welfare of the animal while being held.
- In the event that a bird dies, veterinarians are able to rapidly forward the fresh dead body for diagnosis from necropsy examination (see processing dead birds and eggs later in this chapter).
- The results of this work may provide important information for managing current or future disease issues for the species in general and at the site it originated from, Understanding why a bird is sick should be a high priority for hihi.

Legalities of holding birds

- People holding birds for rehabilitation must meet obligations under the Animal Welfare Act 1999 and a DOC Authority is required (check your current permit conditions) under the Wildlife Act 1953. Only authorised personnel may undertake captive care.
- Captive care (beyond holding birds overnight for observation) is a serious undertaking and requires the correct equipment, food, holding facilities and training.
- Veterinarians are able to hold birds for treatment purposes and currently are not required to hold a DOC Authority for this.

Public perception

- The public generally have an expectation that conservation staff will intervene and provide care to a sick or injured animal. If the choice is made not to intervene then good messaging must accompany this decision. There may well be good reason not to intervene (e.g. biosecurity, human health risk, logistic constraint) and with appropriate information the public will have a better appreciation of why particular decisions are made.

6.2 What to do if you find sick birds

CAUTION: Sick birds may carry zoonoses (diseases and infections that can be transmitted between vertebrate animals and humans). Members of the public who find sick birds should be encouraged to report them but not pick them up. DOC staff / volunteers should handle sick birds with appropriate caution.

Three or more sick birds, or a combination of sick and dead birds (all hihi or a mix of hihi and other species):

- This represents a disease outbreak and may indicate a serious risk to the population.
- The Ministry for Primary Industries may undertake an incursion investigation. Alert them by phoning 0800 80 99 66.
- Advice from the HRG and the DOC veterinarian regarding response options should also be sought (see also the section on dead hihi in this chapter).

One to two sick birds:

- This could be the start of an outbreak, or just one or two sick birds.
- Undertake increased monitoring for sick or dead birds in the area, including species other than hihi, to see if there are any other affected birds.
- Undertake one of the options below based on what is most appropriate for your current situation, noting that the extra effort to obtain a diagnosis may be justified on the basis of the knowledge it may bring or by saving the individual bird if it is a valuable specimen.

  Option 1: Leave in situ – note location and recheck later.

  - *Advantages*: minimal effort is required, action meets the no returns policy.
  - *Disadvantages*: bird may be more likely to die, it may move away with the result that its fate will be unknown, there will be less opportunity to send a fresh dead body for diagnostic necropsy, careful justification to the public may be required to explain why this particular choice was made.

  Option 2: Uplift and hold on site overnight for observation.

  - *Advantages*: the bird's life may be saved, it can be monitored more readily, the bird's fate will be known, it is easy to send a fresh dead body for autopsy, you will be seen as doing the right thing, minimal equipment is required, the action meets the no returns policy
  - *Disadvantages*: you will have obligations under the Animal Welfare Act 1999, what will you do if the bird is still sick the next day?

  Option 3: Uplift the bird, observe it overnight then hold it for rehabilitation (if the bird is still sick after overnight care).

  - *Advantages*: the bird's life may be saved, it can be monitored easily, diagnostic samples can be taken on advice from a veterinarian, the bird's outcome will be known, it is easy to send a fresh dead body for autopsy, action meets the no returns policy.

– *Disadvantages*: a rehabilitation permit is required, only authorised staff can look after the bird, staff will have obligations under the Animal Welfare Act 1999, care requires equipment, skills and time, care may become protracted, staff need to know how to obtain veterinary advice for correct diagnoses and treatment, what do you do if the bird cannot be released but staff have become emotionally attached to it?

Option 4: Uplift and take to a veterinarian

– Advantages: time and resources managed by external party, fewer Animal Welfare Act obligations (transport obligations only), bird will receive best practice for diagnosis and treatment.
– Disadvantages: biosecurity risks, transport required, what do you do if the bird cannot be released but staff have become emotionally attached to it?

6.3 Holding birds temporarily on site

CAUTION: Sick birds should be held temporarily in isolated holding cages or boxes. Do not use aviaries or boxes used in translocations. Sick birds may carry infectious disease-causing agents that can contaminate these structures causing a serious biosecurity risk for their future use.

Legal considerations:

Holding birds requires:

- A DOC authority
- An authorised person skilled in managing animals in temporary captive care

Equipment:

The equipment required to hold birds temporarily includes:

- A holding cage/box (this can be a disposable pet box if the bird is only being held overnight; a more-sophisticated cage/crate/aviary is required if it is being held for longer)
- A suitable floor or substrate (non-slip, cleanable (e.g. towels))
- Perches, food and water containers, artificial food (e.g. Wombaroo™ mix for initial feeding, more balanced diet required if bird is to be held for >24 hours
- Disinfectant (e.g. SteriGENE®, F10, Chlorhexidine)
- Crop tube
- Syringes

Location

A small room isolated from humans and other animals where you can provide:

- Warmth (25°C plus)
- Humidity
- Quiet
- Dark

Injured birds:

Consider your actions and their possible outcomes carefully before you intervene and capture an injured bird. Injured or unhealthy birds are also natural prey for native predators in the environment and injuries are natural events. Bear in mind that there may be restrictions or

difficulty in providing care to the birds once they are caught (see above). In cases where the injury is related to our management (e.g. band injuries), then intervention is deemed to be a responsible and appropriate action. When the decision is made to intervene, then the bird will require a physical examination to determine the type and severity of the injury, treatment requirements and the likely outcome.

The need for pain relief and anaesthesia for diagnosis or treatment and the likelihood of successful release once treatment is completed must be considered. For band injuries, simply removing the band can be a solution. In closely monitored populations these individuals can be subsequently monitored and recaptured if their injury is not healing.

If your site has a no return policy, then anything beyond a very simple injury will require veterinary advice regarding on-site treatment v. euthanasia. For example, a broken bone requires veterinary care or euthanasia, and in the case of a no return policy the only option is euthanasia. Euthanasia should be undertaken by somebody authorised and qualified, such as a veterinarian or experienced DOC staff. In all cases the body should be sent for necropsy (see below) along with documentation justifying why euthanasia was performed.

6.4 Processing dead birds and eggs

Inevitably, monitoring and managing a wild bird population will involve dealing with a certain amount of death. There are several important reasons why hihi death should be closely monitored. Firstly, necropsies performed on adult birds can reveal the cause of death, and provide a bank of samples that may be analysed retrospectively if future disease outbreaks occur. Information from such samples may allow measures to be taken, if necessary and possible, to prevent or limit future deaths from the same cause. The growing database of pathogen risks to hihi has informed disease risk analyses done prior to translocation. The Institute of Veterinary, Animal and Biomedical Sciences (IVABS) at Massey University maintains a database (the Huia database) of necropsy reports from all wildlife, including hihi, submitted to them for necropsy. Secondly, recording the death of eggs and/or chicks will identify patterns of survival during the nesting stage. This information is important for identifying the point of failure during nesting, its cause and, again, allows decisions to be made about the need for management to reduce this.

Tissue samples from dead eggs and/or chicks can be used for sexing and genotyping (see **Hihi Best Practice Sheets 8 & 9** for sample processing methods). In combination with equivalent data on surviving nestlings, this will provide data to monitor any genetic causes of variation in reproductive success in hihi populations (see Box 6.1). The collection of samples for genotyping from both living and dead hihi is beyond standard management and requires additional high-impact research and collection permits from DOC. Genotyping is also not cheap and requires dedicated effort and funding from research partners. This means, even where sampling is possible and permitted, it is not a given that genotyping will or can be done. Any plans for collecting additional information and data should be approached with clear discussion with the people likely to be involved so that expectations, data needs and analyses are clearly defined. It is also worth noting that collected samples can be stored indefinitely for future genetic analysis and systematic collection, where possible and easy, is encouraged.

6.5 Health and safety

CAUTION: Dead birds may carry zoonoses (diseases and infections that can be transmitted between vertebrate animals and humans). Members of the public who find dead birds should be encouraged to report them and not pick them up. DOC staff / volunteers should handle and store dead birds with appropriate caution

Box 6.1 Putting hihi embryo and nestling samples to good use

If the necessary permissions and funding have been obtained, the collection of embryo and nestling samples can generate management-relevant answers to fascinating research questions. The establishment of new populations via reintroduction usually involves a small number of founding individuals and, consequently, such populations are susceptible to inbreeding. Any loss of fitness resulting from inbreeding is termed inbreeding depression, and its extent can vary depending on the stage of development and the sex of the individual. By sampling unhatched hihi embryos and both dead and surviving nestlings, extracting DNA from these samples, and genotyping and sexing all individuals, Brekke et al. (2010) were able to estimate levels of inbreeding at these two key stages of development. They found that inbreeding causes reduced survival at both embryonic and nestling stages of development, and that this effect is most pronounced in males. These findings suggest that inbreeding is an important contributor to the high hatching failure reported in hihi populations (approx. 35%). Despite this high hatching failure, however, hihi populations can grow quickly following reintroduction. In most cases this rapid growth is due to supportive management that addresses other limiting processes on these populations. Managing genetic effects, such as inbreeding, is generally considered a longer-term management issue. Longer-term management is no less important to consider and the evidence so far accumulated by Brekke and colleagues suggests this is very true for hihi.

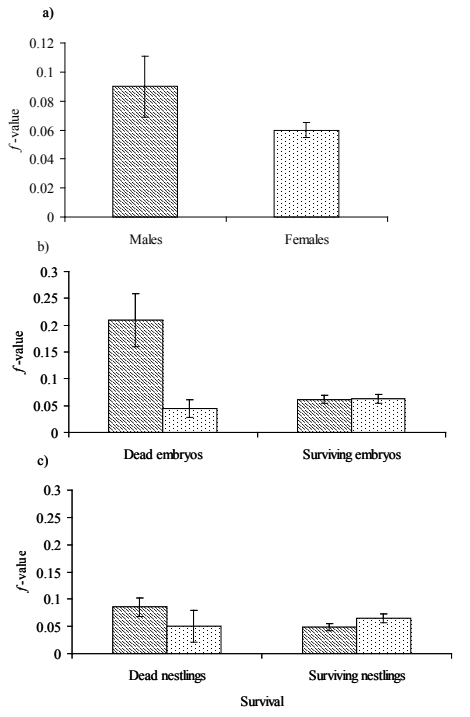

Figure 6.1. Left: Mean inbreeding values for (a) all hihi males and females and (b, c) dead and surviving hihi of either sex at each stage of development, with vertical bars showing standard error distribution. Sample sizes are (a) males at all stages of development, *M* = 133; females at all stages of development, *F* = 94; (b) dead embryos: *M* = 28; *F* = 9; surviving embryos: *M* = 105, *F* = 85; (c) dead nestlings: *M* = 37, *F* = 15; surviving nestlings: *M* = 69, *F* = 70, from the 2006–07 cohort.(b, c) hatched column, male; dotted column, female. Figure reproduced from Brekke et al. 2010.

Many disease agents that may be found in birds are capable of infecting humans (zoonotic disease). All persons performing field preparation of dead birds (including adults, nestlings and eggs) should follow these recommendations:

- Wear a mask and gloves.
- Perform field preparations in areas with good ventilation (or outside) and use surfaces that are easily cleanable (e.g. metal benches, or dedicated surfaces such as plastic chopping boards that can be moved and cleaned). Porous surfaces (e.g. wooden boards) are not appropriate, as they are difficult to disinfect properly.
- Avoid handling sampling pots and writing equipment/data sheets after touching a dead bird. Ideally, a second person can be used to record notes, open and label pots, and take photos. Handling pots with contaminated hands presents a zoonotic risk to the lab personnel receiving the samples.
- Clean and disinfect the instruments and area thoroughly after the procedure. Correct procedure for this is to first wash off any tissue from instruments or the working surface

using a detergent, then disinfect them using an appropriate agent such as SteriGENE® (1:125 dilution), ensuring it remains in contact with the surfaces/instruments for preferably 30 minutes (minimum 5 minutes).

6.6 Dead adults

When a dead adult is found, a necropsy should be performed as soon after death as possible in order to identify the cause of death. There are three broad necropsy options available:

- Send fresh specimens to Wildbase Pathology at IVABS at Massey University in Palmerston North.
- Send fresh specimens to The New Zealand Centre for Conservation Medicine (NZCCM) at Auckland Zoo.
- Fix whole specimens in formalin or perform a basic post-mortem and prepare tissues for diagnostic analyses by Wildbase Pathology at IVABS.

Which of these options should be used will depend on your location, the degree of specimen decomposition and timing. Important information relating to each of the above options is detailed below:

Option 1: Wildbase Pathology at IVABS, Palmerston North

- **Freshly dead birds should be refrigerated and not frozen. Freshly dead** birds that **will arrive at IVABS in reasonable time** should be sent to IVABS by courier. 'Freshly dead' normally means within hours of death and certainly before any evidence of small maggots (a good field tip). 'Reasonable time' normally means that it can reach IVABS by courier within 24 hrs of death. Time from death to arrival at IVABS can be longer (up to 48 hrs) if the bird is refrigerated immediately after death (for example, if it died while being handled, or was caught when sick and then died). For places like Tiritiri Matangi this means sending a dead bird off the island the day it is found and shipping that night by overnight courier to reach IVABS the following morning.
- DOC has a contract with IVABS and will pay for any necropsies that IVABS perform on hihi (regardless of where they were found).
- DOC has a contract with NZ Couriers (tel: 0800 800 841) and Express Couriers (tel: 0800 COURIER) and will pay for specimens to be couriered to IVABS (regardless of where they were found). DOC staff at hihi sites hold the charge code for this service. If you are not at a DOC site, contact DOC for details.
- Do not send fresh specimens to IVABS if they will arrive on the weekend. They will not be processed until Monday, which will add an unreasonable time delay. Use one of the other options instead (see below).
- Detailed instructions of how to submit specimens to IVABS can be found on the IVABS website:
  (http://www.massey.ac.nz/massey/learning/departments/centres-research/wildbase/wildbase-pathology/how-to-submit-a-specimen.cfm)
  and are copied at the end of this chapter (IVABS instructions).
- Two copies of the Wildlife Submission Form (provided at the end of this chapter) should be completed upon submission; one to accompany the specimen and one to be retained for on site records.
- The address for courier delivery is:
  Wildlife Post Mortem Service
  Fifth Floor
  Vet Tower
  IVABS
  Massey University

Fitzherbert Road
Palmerston North

- Wildbase contact details:
  E-mail: wildbase@massey.ac.nz
  Phone: 06 350 4525
  Fax: 06 350 5636

Option 2: NZCCM at Auckland Zoo

- **Freshly dead** birds that **will not reach IVABS in reasonable time (see above)** can be sent to NZCCM.
- DOC does not hold a contract with NZCCM, so the cost of the NZCCM necropsy service must be covered by other funds.
- NZCCM will process and send appropriate samples to IVABS, so that a necropsy report can be generated and submitted to the Huia database, a final report of which should be provided to the person who originally submitted the dead bird.
- All courier costs from site to NZCCM will be covered by DOC contracts, as detailed above. Before sending any dead birds, check with appropriate managers and ensure there are funds to do this. Ideally, check before dead birds are recovered (e.g. at the start of any seasonal monitoring) so that clear protocols are in place before any urgent responses are required.
- Fresh specimens should be prepared for submission as detailed in the IVABS instructions copied at the end of this chapter and using the same submission forms.
- Two copies of the Wildlife Submission Form (provided at the end of this chapter) should be completed upon submission; one to accompany the specimen and one to be retained for records.
- The address for courier delivery is:
  New Zealand Centre for Conservation Medicine
  Auckland Zoo
  Gate 2 Motions Rd
  Western Springs 1022
- Staff at Auckland Zoo should be contacted before submitting dead birds to NZCCM (please cc all names below to ensure duty veterinarian receives email. Also note that staff change and it is worth calling the NZCCM to confirm – 09 353 0753 or 0274061943. If no one answers the phones then please send a text explaining situation and NZCCM will respond):
  - Mikaylie Wilson, Clinical Co-ordinator
    Mikaylie.wilson@aucklandcouncil.govt.nz
  - James Chatterton, Senior Clinical Veterinarian
    James.chatterton@aucklandcouncil.govt.nz
  - An Pas, Associate Veterinarian
    An.pas@aucklandcouncil.govt.nz
  - Sarah Alexander, Resident Veterinarian
    Sarah.alexander@aucklandcouncil.govt.nz

Please phone Mikaylie Wilson on 0272772562 to discuss logistics of getting birds to NZCCM

Packaging tip: Ideally, samples should be packaged in polystyrene foam chilly bins with freezer blocks, both of which are available from veterinary clinics and laboratories. If these are not available, a cardboard box with ample newspaper and bubble wrap will suffice. Freezer blocks can be improvised using 500 mL plastic PET drink-bottles – don't quite fill them and put them in your freezer. Alternatively, partially fill snaplock bags with water and put them in your freezer. It is recommended that supplies for sending dead birds be kept on hand at all times.

Option 3: Field preparation of specimen for later diagnostic analysis by IVABS

CAUTION: Skin contact and inhalation of formalin is hazardous. Handle with extreme care, and wear gloves when using formalin.

- Birds that are **not freshly dead** or **cannot reach either IVABS or NZCCM in reasonable time** (see above options) should be processed for later diagnostic analysis and samples sent to IVABS by courier.
- Whole specimens can be fixed or, alternatively, tissues can be fixed following field dissection (the latter preferred, if possible). See Table 6.1 for details of alternative field dissection options.
- Courier costs and IVABS necropsy charges are covered by DOC contracts, as detailed above.
- Two copies of the Wildlife Submission Form (provided at the end of this chapter) should be completed upon submission; one to accompany the specimen and one to be retained for records.

6.7 Museum collections

There is currently a shortage of complete and recently collected hihi specimens in New Zealand museums. Necropsies can usually be modified so that remains are still suitable for use as museum specimens. If you would like a specimen to be sent on to a museum after the necropsy has been performed, you should do the following:

- Ensure that iwi considerations have been met. In some areas iwi will have their own protocols for disposal of bodies or restrictions on where they can go. In addition, iwi may also have wishes for use of bird feathers (for example) and in some cases these wishes are reflected in translocation agreements. So check with and talk to iwi as part of this process.
- When contacting IVABS and/or NZCCM to inform them that you are sending a specimen (see above), indicate that you would like the specimen to go to a particular museum.
- When completing the Wildlife Submission Form (see above), you should indicate in the 'History' section which museum you want the specimen to go to and the contact details of the museum.

The following museums will be interested in receiving hihi specimens:

Auckland War Memorial Museum

Postal address: Private Bag 92018, Auckland
Courier address: Domain Drive, The Domain, Parnell, Auckland
Ph 09-3090443
Fax 09-3067065
Staff to contact: Matt Rayner, 3067063, mrayner@aucklandmuseum.com
Ramola Prasad, 3067063, rprasad@aucklandmuseum.com

Museum of New Zealand / Te Papa

Postal address: PO Box 467, Wellington
Courier address: 169 Tory Street, Wellington
Ph 04-3817000
Fax 04-3817310
Staff to contact: Gillian Stone, 3817304, gillians@tepapa.govt.nz
Alan Tennyson, 3817315, alant@tepapa.govt.nz

Te Papa has an account with Tranzlink Refrigerated; they will pick up and deliver door to door. Contact Gillian or Alan for the account number.

Canterbury Museum

Postal address: Rolleston Ave, Christchurch 8001
Courier address: Rolleston Ave, Christchurch
Ph 03-3665000
Fax 03-3665622

Staff to contact: Paul Scofield, 3669429 x 853, pscofield@canterburymuseum.com

The positive and negative aspects of the various options for post mortems and preservation of dead hihi are examined in Table 6.1.

Table 6.1. Options for post mortem examination and preservation of specimens from dead birds.

OPTION	DESCRIPTION	POSITIVES	NEGATIVES
Place entire body in formalin	Slit abdomen to expose contents to formalin, place entire bird in a pot that is large enough to contain a 10:1 ratio of formalin:bird	Quick, simple, cost effective and samples can be sent to IVABS when convenient.	If an infectious process is identified at histopathology (e.g. suspected virus, bacteria, fungus) it will not be possible to pursue further diagnostics on formalin fixed tissue (e.g. tissue culture). PCR may be possible on formalin fixed tissue, but is not ideal.
Basic post mortem	Open abdomen, collect small pieces of key organs into formalin, freeze and store remains of bird and organs. Key organs: heart, brain, lung, kidneys, spleen, liver, gastrointestinal tract.	Time efficient, can be performed in the field with limited training required, frozen tissues may be used for further work up/diagnostics if indicated by histopathology findings.	More time consuming than first option, requires storage space and curation of frozen specimens on island (although can be sent in formalin to IVABS at later date), some training required to collect right organs/amounts.
Full post mortem	Collection of all key organs, description of lesions if noted, collection of appropriate specimens for culture/PCR etc as indicated by findings at post mortem.	Most likely to get full diagnosis of cause of death.	Time consuming, requires expertise or understanding of normal/abnormal gross anatomy and pathology of birds. May require storage and appropriate curation of specimens on island.

6.8 IVABS instructions – copied unmodified from Wildbase Pathology Website

How to submit specimens

The three important points to consider when submitting an animal for post mortem are:

- Preservation
- Documentation
- Packaging

Preservation

To be of most benefit, post mortem examination should be performed as close to the time of death as possible. If this isn't possible, place the animal in a refrigerator (approx. 4°C) as soon after death as possible, and then send as soon as possible (please don't send over the weekend). Freezing the body interferes with results and should be a last resort. Fixing a body whole in formalin or 70–80% alcohol, or field dissection and submission of fixed tissues for histopathology, are alternatives that can be used in some circumstances.

If you are collecting material into fixatives, remember skin contact and inhalation of formalin is hazardous. The volume of fixative needed is 10 times the amount of tissue you are fixing; for example, 100 g of tissue needs 1 L of formalin. The smaller the piece of tissue the better the

fixation; ideally, pieces of tissue should be no thicker than 1 cm to allow for rapid fixation. It is surprising how much information can be gleaned, even from fairly decomposed specimens, so do not let a rotten carcass discourage you from submitting it for post mortem.

Documentation

Proper documentation is essential to get the most benefit from the post mortem. The Wildlife Submission Form (PDF file) should be sent with the body or faxed to 06 350 5636. If this isn't possible, please include the following information:

- Animal, tissue or specimen identification (including species, individual's ID)
- Geographical location where animal was found, time of collection (who, what, where, when?).
- Any history you think relevant; for example, previous signs of ill-health, use of toxins/baits in the area. The more history you provide the better.
- Any other special requests; our routine practice is to try to establish a cause of death and other intercurrent diseases when a whole body is submitted. You may want to know something else instead of or in addition to these things.
- Let us know what you would like us to do with the remains of the body; would you like it returned or disposed of? Let us know if you would like the animal returned for taxidermy purposes as we will need to modify the post-mortem technique.

Packaging

To prevent contamination of people and equipment with potentially infectious or hazardous substances, a suitably sized polystyrene foam chilly bin is best. Alternatives can include a cardboard box with newspaper and bubble-wrap protecting the well-wrapped and bagged body. Freezer blocks can be improvised using 500 mL plastic PET drink-bottles – don't quite fill them and put them in your freezer. To contain the body and prevent any leakage, use multiple tear- and puncture-resistant sealed plastic bags, or plastic containers with firmly screwed down tight-fitting lids; don't use glass. Place the submission form in a separate plastic bag.

Send to:

Wildlife Post Mortem Service
Fifth floor
Vet Tower
IVABS
Massey University
Fitzherbert Road
Palmerston North

- Mark the package: Urgent, Perishable or Keep Cool, Do Not Freeze.
- Inform us by email, phone or fax so we know to expect a parcel
  - Email: wildbase@massey.ac.nz
  - Phone 06 350 4525
  - Fax 06 350 5636
- The Huia database submission form (PDF file) can be included with the animal's body and/or emailed/faxed to the above contacts (a copy of this form is reproduced on the next page).
- The following courier companies are recommended:
  - NZ Couriers
  - Tranzlink

In summary:

- Chill and despatch ASAP
- Identify and specify what you want in the documentation
- Contain, preserve and protect in transit by appropriate packaging

WILDLIFE SUBMISSION FORM

Forwarding Instructions

This animal is the property of the Department of Conservation. Please send a copy of test results to: Wildlife Mortality Database Manager, c/- Pathobiology, IVABS, Massey University, Private Bag 11-222, PALMERSTON NORTH

Submitter Details

Surname: _____
First name: _____
Organisation: _____
Address/Box: _____
Suburb: _____
City/Town: _____
Phone (bus.): _____
Phone (home): _____
Mobile: _____
Fax: _____
Email: _____

Submission Details

Date submitted: ____/____/____ Submitter ref: _____
Date found: ____/____/____ Number dead: _____
Number at risk: _____ Number sick: _____
(In-contacts)

Mortality

Date animal died: ____/____/____

Death circumstances:

- ☐ Found dead
- ☐ Found alive and died
- ☐ Treated and died
- ☐ Capture or release
- ☐ Infertile
- ☐ Euthanased
- ☐ By-catch

Specimen Details

Animal Details

(Please use separate page for additional animals)

Species/common name: _____
Animal ID: _____
Identification type: _____
(Leg band, microchip implant, rig tag, attoo toe clip etc.) _____
Individual name: _____

Sex: ☐ Male ☐ Female ☐ Unknown
Age Classification: ☐ Adult ☐ Subadult ☐ Juvenile
☐ Neonate ☐ Foetus ☐ Embryo ☐ Egg

Date of birth/mating: ____/____/____

Age/incubation/gestation period / period: _____ Years _____ Months _____ Weeks _____ Days

Where born/hatched: ☐ Wild ☐ Captivity

Weight: _____gm/kg

Location Type

Wild
- ☐ Mainland National Park
- ☐ Mainland Reserve
- ☐ Mainland Private Land
- ☐ Maritime Park
- ☐ Island
- ☐ Coastline
- ☐ Sea
- ☐ River
- ☐ Other: _____

Captive
- ☐ DoC Facility
- ☐ Private Breeding Facility
- ☐ Rehabilitation Facility
- ☐ Zoological/Wildlife Park
- ☐ Other: _____

Location name: _____
Conservancy: _____
Description: _____

☐ Poisons are being used in the area. Please include details of the toxin.

_____

☐ Special requirements for disposal of body parts, e.g. return to submitter for iwi requirements, genetics, or forward to Te Papa etc.

Please state details of which body parts required and invoice submitter for carrier costs.

_____

History

Include any information which you think may be relevant to this case.

Previous health history:

Clinical signs; external examination; individual treaments; abnormal behaviours (feeding, reproductive, agnostic); breeding history; diet with any changes; exposure to toxins; translocation details; previous clinical pathology (attach relevant reports).

_____
_____
_____

Environmental Conditions (including climate):

Enclosure substrate/size/type; group treatments; in-contacts; clutch details if relevant - sire ID/name, dam ID/name, number of eggs, egg lay interval, season number, season clutch number, incubation temperature and humidity.

_____
_____
_____

Coptinue over leaf

Invoice Instructions

Invoice: Submitter ☐ National Wildlife Surveillance Fund ☐

(Refer to 'Guidelines for the use of the National Wildlife Surveillance Fund' for eligibility on the WILDLIFE HEALTH PAGE - WGNCR-37176)

Results of post mortem studies of hihi samples at IVABS

Table 6.2 provides details of post mortem disease diagnoses of hihi recorded from 1991–2008 at IVABS.

Table 6.2. Post-mortem disease diagnosis categories for dead hihi (excluding neonates <3 weeks old) submitted to the Institute of Veterinary and Biomedical Sciences (IVABS) between 1991 and 2008. Hihi were submitted from the single captive breeding population and from dead individuals recovered across all monitored wild populations. Data from 1991 to 2000 contain few hihi recovered from wild populations. We present results for the major categories of mortality (contributing ≥5% of total records) and report numbers of diagnoses from wild hihi and totals for both wild and captive cases in parentheses. There are often multiple disease diagnoses for an individual, in which case the pathologists made a decision on which seemed most important. Reproduced from Ewen et al. 2012.

	1991–2000	2001	2002	2003	2004	2005	2006	2007	2008	Total
Aspergillosis	20	2 (9)	2 (4)	0 (2)	0	1 (2)	8 (12)	3 (6)	5 (6)	21 (61)
Coccidiosis	11	0 (3)	2 (3)	0 (2)	0 (1)	0	2 (2)	0 (2)	1 (2)	5 (26)
Trauma (excluding predation)	3	0 (1)	2 (3)	0 (1)	0 (1)	3 (5)	2 (3)	5 (6)	2 (3)	14 (26)
Hepatic haemosiderosis	10	0	0 (1)	1 (4)	0 (1)	0 (1)	2 (5)	0	1 (3)	4 (25)
Bacterial infection	8	0 (1)	2 (3)	1 (1)	0	2 (2)	2 (2)	2 (2)	1 (2)	7 (21)
Bacterial infection: salmonella*	0	0	0	0	0	0	6 (6)	0	0	6 (6)
Myocardial/skeletal myonecrosis	7	0 (1)	0 (2)	0	0 (1)	0	0 (1)	1 (2)	0	1 (14)
Nematodes	2	0 (2)	1 (1)	0	0	1 (1)	2 (2)	2 (2)	1 (2)	7 (12)
Other	20	1 (5)	2 (6)	1 (3)	1 (3)	0 (1)	8 (10)	7 (16)	1 (4)	21 (68)

* Denotes a special case of bacterial infection from an outbreak of a novel Salmonella strain in hihi on Tiritiri Matangi Island in 2006 that is treated separately from the general bacterial infection category.

6.9 Further information

The best practice and template record sheets found at the end of this chapter are related to supplementary data collection protocols. They have been included so that a standard practice can be followed, should a site choose to investigate egg and nestling mortality. These standard practices have been refined on Tiritiri Matangi Island. We stress that this work is non-essential. However, if time, resources and site logistics mean these protocols are relatively easy to perform, it is worth bearing in mind that the data collected can be very informative.

6.10 Q & A: why do we do it this way?

For a general description of the reason for this Q & A section, see Chapter 1. The following set of questions and answers capture some of the issues relating to dealing with sick hihi:

Q. How long does it take to transport a bird from a remote site to IVABS? What is the best way of achieving this quickly?

A. It is imperative that a freshly dead bird reaches IVABS as quickly as possible (within reasonable financial constraints!). The aim should be to have the specimen reach IVABS within 24 hours of it being found. This may be harder to achieve at island sites, where birds can only leave by scheduled ferry services. On Tiritiri Matangi, for example, the ferry does not run on Mondays or Tuesdays, and is occasionally cancelled in bad weather. On these days, the specimen is unlikely to reach IVABS in reasonable time, and we recommend field preparation of the specimen for later diagnostic analysis. If the fresh specimen can leave the island on the day it was found, then ideally it should be couriered overnight. In the case of Tiritiri Matangi, a courier can then pick it up from the Ferry Terminal office (360 Discovery Cruises, 139 Quay Street, Pier 4,

Downtown Auckland) or from a helpful volunteer's home. If it cannot be couriered overnight, then the specimen should be kept in a fridge overnight (note there is no fridge at the Ferry Terminal office) and picked up first thing the following morning.

Q. Why should dead birds not be frozen?

A. Freezing a freshly dead bird is not advised, since this will interfere with the results of the post-mortem examination.

Q. Do we need to respond to the pathology reports received following a hihi necropsy?

A. Whether or not a response is required will depend on the results of the report. In all instances, a copy of the report should be kept for reference and forwarded to any interested parties. If the cause of death is diagnosed as salmonellosis, then the following action should be taken immediately:

- Inform the site manager, the head of the hihi recovery group, DOC veterinarian Kate McInnes (e-mail: kmcinnes@doc.govt.nz, phone: 04 495 8604) and NZCCM veterinarians (particularly if samples were handled by them). There will certainly be discussion on a recommended course of action.
- Keep feeding stations open, unless advised otherwise.
- Increase frequency of cleaning at feeding stations to daily (according to protocols in **Hihi Best Practice Sheet 3** from Chapter 4.
- Do not provide antibiotic treatment via feeders. Antibiotic treatment can promote carrier status and resistant strains.
- Actively search the areas around the feeding stations for dead birds (of any species), and perform watches at feeders to identify any sick birds.
- Increase site-wide vigilance for dead birds (of any species) and advise all staff/volunteers/visitors to report any dead birds found. Remember that salmonella can also infect humans, and any dead birds should therefore be handled with necessary caution (see health and safety section in this chapter).

Q. Why don't we also send dead nestlings for necropsy?

A. This is largely because of the volume of samples that would be sent to IVABS and their ability to cope with the added work. Doing so may well provide important knowledge. Previous examinations of causes of nestling death have been particularly interesting (see Rippon et al. 2013).

6.11 Suggested further reading

Information gained from collecting samples from eggs and dead nestlings:

Thorogood, R.; Ewen, J.G. 2006: Rare occurrence of embryonic twins in an endemic passerine of New Zealand, hihi (stitchbird, *Notiomystis cincta*). *Ibis 148*: 828–829.

A short note describing a surprise find when examining the contents of unhatched eggs.

Brekke, P.; Bennett, P.M.; Wang, J.; Pettorelli, N.; Ewen, J.G. 2010: Sensitive males: inbreeding depression in an endangered bird. *Proceedings of the Royal Society of London B 277*: 3677–3684.

First evidence of inbreeding depression in hihi. DNA samples were analysed from unhatched embryos and dead nestlings and compared to those of surviving offspring to reveal lowered viability in more inbred individuals. Interestingly there was a male bias in this pattern. See details in Box 6.1.

Hemmings, N.; West, M.; Birkhead, T.R. 2012: Causes of hatching failure in endangered birds. *Biology Letters 8*: 964–967.

We assume that unhatched eggs that we can separate nicely into yolk and albumen are infertile but this paper shows that many of these eggs are fertile with sperm present.

Parasites and health in hihi

Ewen, J.G.; Thorogood, R.; Nicol, C.; Armstrong, D.P.; Alley, M. 2007: *Salmonella typhimurium* in hihi, New Zealand. *Journal of Emerging Infectious Diseases 13*: 788–790.

A brief but detailed analysis of the 2006 salmonella outbreak on Tiritiri Matangi island providing rarely available population-level impacts from a disease emergence.

Ewen, J.G.; Armstrong, D.P.; Empson, R.; Jack, S.; Makan, T.; McInnes, K.; Parker, K.A.; Richardson, K.; Alley, M. 2012: Parasite management in translocations: lessons from an endangered New Zealand bird. *Oryx 46*: 446–456.

A review of how disease risk management has been implemented and refined over time in hihi. It also provides a summary of health problems and causes of death in adult hihi from across all populations.

Schoener, E.R.; Alley, M.R.; Twentyman, C.M.; Howe, L.; Barta, J.R.; Charleston, W.A.G.; Castro, I. 2013: Coccidiosis in hihi/stitchbirds (*Notiomystis cincta)* due to coccidian of the Eimeriidae. *New Zealand Veterinary Journal 61*: 68–76.

A detailed summary of the coccidian parasites that infect hihi. Coccidiosis is a problem mostly in captive hihi, but coccidian parasites are common in wild birds and are the focus of disease risk management in translocations.

Rippon, R.J.; Alley, M.R.; Castro, I. 2013: Traumatic ventriculitis following consumption of introduced insect prey (Hymenoptera) in nestling hihi (*Notiomystis cincta*). *Journal of Wildlife Diseases 49*: 80–90.

An interesting study showing the risks that wasps pose to hihi. The population used in this study is at Zealandia, in Wellington.

For more reading on common health issues in hihi (such as feather loss and protruding tongues) please see Box 5.1 in Chapter 5

Hihi Best Practice Sheet 8: Processing unhatched eggs

When incubated eggs fail to hatch, they can be opened to determine whether the failure was due to infertility or embryo death. Samples can also be taken for subsequent genetic analyses and to identify sperm presence. The method of storage will depend on the purpose for which samples are being kept. The recommended procedure for processing un-hatched eggs for these purposes is detailed below, with the **necessary equipment** highlighted in bold.

1. After removing unhatched eggs from the nest according to the procedure in **Hihi Best Practice Sheet 10** from Chapter 7 (Monitoring breeding), store them in the fridge until analysis.
2. Always analyse unhatched eggs as soon as possible after they have been collected. The longer they are left the more decomposed they will be, and the harder it will be to determine their contents.
3. Open the egg over a clean and dry **petri dish**. Hold the egg with the air space (usually at the rounded end) up and, using a clean **disposable pipette tip** (or similar sharp instrument), tap through the shell in a circle around the edge of the air space.
4. After taking the top off from the egg, you should see a thin membrane separating the egg contents from the shell and air space. Pierce this membrane with the pipette tip, and peel some away. This will now expose the egg contents (Fig. 6.1).
5. Tip the egg contents gently into the petri dish. You may need to enlarge the hole in the egg in order to do this more easily. Depending on when the egg failed, the contents will appear quite different. How to identify the different stages of embryonic development, and how to process samples appropriately, are detailed below.

Figure 6.2. Egg with shell removed. Note membrane over yolk.

6. INFERTILE / NOT INCUBATED:
   - The egg contents will separate perfectly into yolk and albumen (Fig. 6.2).
   - Record as ‘separated’ in data sheet (see **Hihi Template Record Sheet 3** for example data sheet).
   - Tip entire contents (yolk and albumen) into **an approximately 25 mL container** and fill remainder of tube completely with **10% formalin**.
   - Using a pencil, label the side of the container with sample number, nest ID, clutch number, the date the egg was collected from the nest, and ‘separated’. Cover label with **cellotape**. Label the lid with the sample number.
   - Store the sample upright in the **fridge**.
   - Samples processed in this way can be used to identify sperm presence.

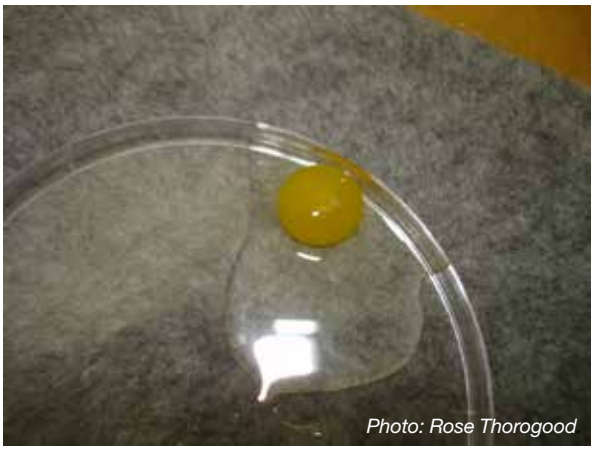

Figure 6.3. An egg which is either infertile or has not been incubated.

7. ‘GUNK’:
   - Yolk broken up and albumen discoloured.
   - This will indicate either that the embryo died very early in development, or that it is an infertile egg that has gone rotten.

Continued on next page

- A small white ring of white on the yolk, a small white lump, or a small spot of blood all indicate that the egg was fertile and has developed.
- Record as 'gunk' in data sheet and include comments on any signs of development (see **Hihi Template Record Sheet 3**).
- Tip entire contents into an appropriately sized container and use a pipette to cover with 5% formalin.
- Using a permanent marker pen, label the side of the container with sample number, nest ID, clutch number, the date the egg was collected from the nest, and 'gunk'. Cover label with cellotape. Label the lid with the sample number.
- Store the sample upright in the fridge.
- Samples processed in this way can be used to identify sperm presence.

8. MID DEVELOPMENT EMBRYO (MDE):
   - Mushy yolk and discoloured albumen with a visible embryo (Fig. 6.3).
   - The embryo will have an eye spot, limb buds, and may or may not have a tail depending on its age. You may be able to make out the brain lobes on the back of the head, and a small mouth.
   - Record as 'MDE' in data sheet and make notes on embryo appearance (see **Hihi Template Record Sheet 3**).
   - A tissue sample from this embryo can be kept for genetic analysis. To sample the embryo, place it in an approximately **25 mL container** and use a pipette to fill the remainder of the container with **>80% ethanol**. You may need to use two pipette tips to remove the membrane and yolk from the embryo, whilst it is still in the petri dish.
   - Using a **pencil**, label the embryo sample, on the side of the container, with sample number, nest ID, clutch number, the date the egg was collected from the nest, and 'MDE'. Cover label with cellotape. Label the lid with the sample number.

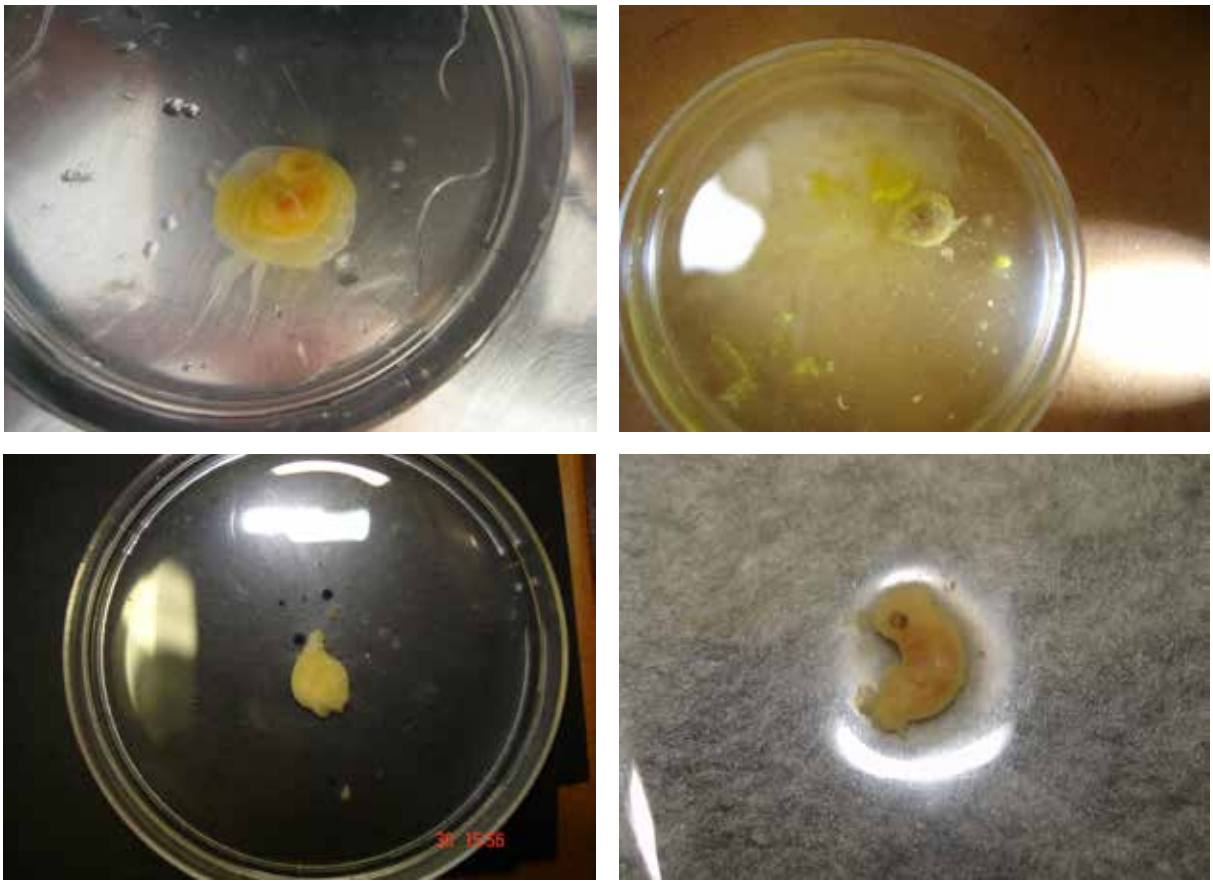

Figure 6.4. A range of hihi mid-development embryos (MDE) of different ages and condition. *Photos: Rose Thorogood.*

Continued on next page

9. LATE DEVELOPMENT EMBRYO (LDE):
   - The embryo looks like a chick, but is not developed enough to hatch (Fig. 6.4). It will have fully formed head, wings and legs, and may have down starting to appear on its head and spine. The yolk will not be internalised and will still be quite large. There will still be albumen present. Depending on how rotten the egg is, the albumen may no longer be separate and the liquids inside the egg may be discoloured and sloppy, or even quite thick.
   - Record as 'LDE' in data sheet and make notes on embryo appearance (see **Hihi Template Record Sheet 3**)
   - Remove both legs from the embryo and store in ethanol exactly as detailed above for an MDE egg.

Photo: Rose Thorogood

Figure 6.4. An LDE. Note how the body looks 'soft'.

10. FULLY FORMED CHICK (FFC):
    - A fully formed chick is one which is ready to hatch. It will have a lot of down on the head and back, and the body will no longer look 'soft'. The yolk may not be completely internalised, but there will not be a lot of it. There will be very little liquid within the egg.
    - Record as 'FFC' in data sheet and make notes on embryo appearance (see **Hihi Template Record Sheet 3**).
    - Sample the embryo as detailed above for LDE eggs.
11. After processing eggs, rinse out petri dishes with hot water and spray with a strong bleach solution. Rinse again, and leave to dry. Seal all pipette tips and eggshells in a zip-lock bag and dispose of in the general rubbish.

Hihi Best Practice Sheet 9: Processing dead chicks for genetic analysis

The recommended procedure for processing dead chicks, in order to provide tissue samples suitable for genetic analysis, is detailed below. The **necessary equipment** is highlighted in bold.

1. Store dead chicks in **labelled zip-lock bags** in the **fridge** until processing, or process immediately in the field. Dead chicks should be processed as soon as possible, ideally on the day that they are found.
2. On a clean work surface, or at the nest if done immediately, use a **scalpel blade** to remove about 1 cm of leg.
3. Transfer the tissue sample into an approximately **25 mL container**. Dispose of the scalpel blade into a **sharps bin**.
4. Use a **pipette** to completely fill the container tube with **>80% ethanol**.
5. In **pencil**, label the outside of the tube with the sample number, the nest ID, the clutch number and the date. Record this same information on a data sheet (see **Hihi Template Record Sheet 3** for example data sheet).
6. Cover the label with **cellotape**, to prevent any leaking ethanol from erasing the label. Also label the lid of the eppendorf tube with the sample number.
7. Store the eppendorf tube upright (so that sample remains submerged) in a **freezer** at –20°C. If no freezer or fridge is available on site, then samples can be stored at room temperature once preserved in ethanol. Ideally, these should be removed to a freezer at some point and regularly checked until this occurs to ensure ethanol has not evaporated from the tube. If ethanol levels are low then top up with more.
8. The samples should remain stored in this way until the planned genetic analyses can be conducted.

Hihi Template Record Sheet 3: Unhatched eggs and dead chicks

This is an example record sheet for recording un-hatched eggs and dead chicks. This version includes example data, to illustrate how it should be used. Blank versions for photocopying are included at the end of this best practice guide.

Sample Number	Nest ID	Date of death	Date collected	Date of sample	Type	Embryo stage (if egg)	Comments
1	1/28	n/a	13-Nov	15-Nov	e	LDE	Small amount of down on head, quite a lot of yolk not internalised.
2	1/28	n/a	13-Nov	15-Nov	e	LDE	Small amount of down on head, lots of yolk not internalised.
3	21/4	n/a	15-Nov	15-Nov	e	gunk	Yolk broken up, albumen discoloured, white lump ~6 mm diameter, no obvious embryo.
4	22/24	n/a	15-Nov	15-Nov	e	MDE	Obvious eyespot, 4 limb buds, embryo ~1.5 cm in length.
5	1/28	14-Nov	14-Nov	15-Nov	ch	n/a	Found dead at entrance of nest box.
6	3/3	17-Nov	17-Nov	17-Nov	ch	n/a	Found dead ~10 m from box.
7	22/8	n/a	16-Nov	17-Nov	e	separated	
8	3/3	n/a	17-Nov	17-Nov	e	gunk	Yolk beginning to break up, albumen slightly discoloured, small white lump.
9	22/26	n/a	17-Nov	17-Nov	e	MDE	Very small embryo, pinkish, can just make out eyespot.
10	22/31	18-Nov	18-Nov	18-Nov	ch	n/a	Found dead in stick cup.
11	22/29b	n/a	18-Nov	18-Nov	e	FFC	Head sample taken, no other egg contents, small indentation in shell from outside.

Instructions:

- Date of death: date that chick was known to have died. If exact date not known put the range of dates over which it would have died. N/A if egg.
- Date collected: date on which dead chick or egg was removed from the nest.
- Date of sample: date on which chick or egg was processed and sample was taken.
- Embryo stage (egg only): one of separated, gunk, MDE, LDE, FFC (see **Hihi Best Practice Sheet 8** for details).
- Comments: additional important and/or useful information. For example, size of embryo, presence of limb buds and eye spots, state of yolk etc. (in case of egg), or where chick was found, whether emaciated, presence of mites etc. (in case of chick).

7. Monitoring hihi breeding

7.1 Hihi breeding season and site differences

The length of the hihi breeding season varies from site to site, but typically begins in September/October and finishes in February/March. Monitoring procedures will vary depending on objectives and whether the nests are natural or in nest boxes (Fig. 7.1). At some sites nesting occurs almost exclusively in nest boxes, with only occasional natural nests, whilst at other sites all nests are natural. A recommended goal of monitoring breeding, regardless of nest type, is to band all the fledglings produced each year (Fig. 7.1). This ensures that all individuals in the population are individually identifiable and of known age and, if banding is done in a standardised way, it can provide estimates of productivity to compare between years. More intensive monitoring of breeding can establish individual pairs' breeding success by recording laying dates, hatching dates and fledging dates, and the genetic parentage of offspring. At some sites nest boxes become infested with mites (Box 7.1), and they should therefore be monitored

Figure 7.1. Monitoring breeding (clockwise from top left): A. The site of a natural nest at Maungatautari (the cavity visible on the left hand side); B. Young nestlings in a nest box nest; C. Getting weighed; D. A nestling with its newly acquired bands; E. Banding a nestling; F. Measuring the head-bill length of a nestling. *Photos: Leila Walker.*

Box 7.1 Increased productivity through mite control

In some hihi populations, adults and nestlings are parasitised by the blood-sucking mite *Ornithonyssus bursa*. This species of mite belongs to the Dermanyssidae family and is known to parasitise a number of bird species worldwide, including North Island saddleback (*Philesturnus rufusater*) (Stamp et al. 2002) and North Island robin (*Petroica longipes*) (Berggren 2005) in New Zealand. The life-cycle of *O. bursa* includes five stages (Powlesland 1977). After consuming a blood meal, a mature female will lay her eggs either directly on the host or in the nest material of the host. The eggs hatch into non-feeding colourless larva, which then moult into a feeding protonymph stage, followed by a non-feeding deutonymph stage, before finally moulting to the adult stage. In optimal, humid conditions the whole life cycle can take place within 7 days (Powlesland 1977). Mites are primarily transmitted to nests and nestlings by adult hosts as they enter and leave nests (Powlesland 1978).

Hihi populations in more northerly latitudes appear to be the worst affected by mite infestations. For example, on Tiritiri Matangi Island (36.6°S) around 74% of broods in a season will have mites, and around 18% will require nest replacement (Ewen et al. 2009). The now extinct Mokoia Island (38.1°S) hihi population was also badly affected by mites (Armstrong et al 2007). In contrast, at Zealandia (41.3°S), mites seem to be present at low prevalence and cause no problem (Rippon 2011). We do not know with certainty the reason for this latitudinal trend in mite presence, but it is possibly related to differences in ambient temperature and humidity.

Research has been conducted on the Mokoia and Tiritiri Matangi hihi populations to identify the individual and population-level consequences of mite infestations. Monitoring on Mokoia suggested that mite infestations could kill entire broods (Armstrong et al. 2007). Armstrong et al. (2007) estimated that 29% of broods would have been lost without mite control. Furthermore, population projections based on this data suggested that the Mokoia population would decline in the absence of mite control in combination with supplementary feeding. However, these results were based on the assumption that all nests with mites would fail, which may not always be the case. This highlighted a need for further quantification of the effects of *O. bursa* on nestling growth, condition, and survival, and this was provided by a study on Tiritiri Matangi Island. During the 2005–06 breeding season, Ewen et al. (2009) provided half of all nests with mite treatment (using Frontline Spray and nest replacement) and left the remaining half of nests to develop mite infestations. They found that nestlings from *O. bursa* removal nests were 8.1 % heavier (39.9 g v. 36.9 g; Fig. 7.2) and 1.4× more likely to fledge than nestlings from untreated nests. These results provide strong evidence of the detrimental effect of *O. bursa* infestations on hihi nestlings, and the beneficial effects of mite control on hihi productivity. **Hihi Best Practice Sheet 12** provides details for detecting and treating mites.

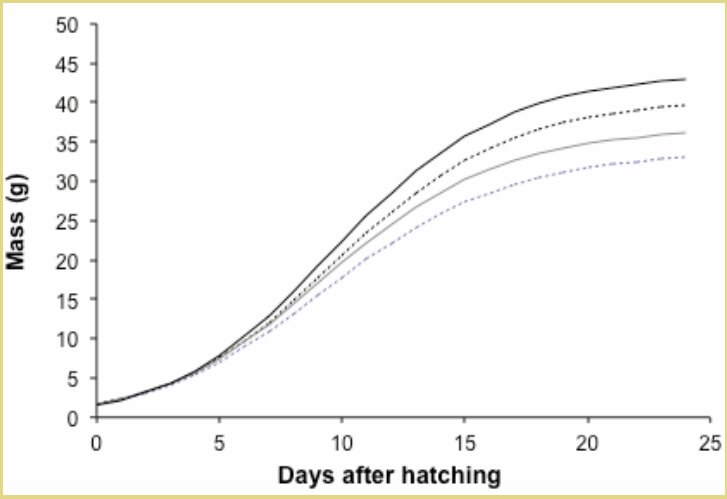

Figure 7.2. Mean growth of male (upper two lines in black) and female (lower two lines in grey) hihi nestlings from two treatment groups. *O. bursa* removal are represented by the solid lines and *O. bursa* controls (mites left on nestlings) are represented by the dashed lines. This figure is modified from that presented in Ewen et al. 2009 and details of how growth curves were calculated are available within that publication.

for mites and treated accordingly. This chapter details all recommended procedures relating to the monitoring of natural and nest-box nests (see **Hihi Best Practice Sheets 10** and **11**) and estimating the age of hihi chicks (see **Hihi Best Practice Sheet 13**).

7.2 Q & A: Why do we do it this way?

For a general description of the reason for this Q & A section, see Chapter 1. The following set of questions and answers address some of the recent trials and changes relating to monitoring breeding:

Q. What is the minimum age at which chicks can be handled and/or treated with Frontline®?

A. We recommend that chicks should not be handled if they are younger than 6 days old. From 6 days onwards they should be robust enough to withstand careful handling. Chicks younger than this rarely have bad mite infestations, although it can happen. If mites are detected on chicks younger than 6 days (by placing a hand in the nest, rather than handling them), the disturbance associated with treating them with Frontline® should be weighed against the severity of the infestation.

Q. How long should unhatched eggs be left in the nest before they can be removed?

A. The longer eggs are left in the nest, the more decomposed they will be when it comes to processing them. Even if eggs are not being processed, the longer they are in the nest the greater the likelihood they will get broken and make a (potentially unhygienic) mess. We therefore recommend removing eggs as soon as possible after the point when they might feasibly still hatch. Hihi sometimes show hatching asynchrony, but all nestlings within a clutch will have hatched within 3 days of each other. We therefore recommend removing any eggs that are still in the nest 4 days after the first chick hatched.

Q. Is it okay to look inside nest boxes if adults are inside? Should we wait for adults to the leave the nest box before checking the nest?

A. We recommend waiting for adults to leave before checking a nest box. This will involve observing a box until an adult leaves (usually about 10 minutes). Following this procedure will ensure that breeding hihi pairs are disturbed as little as possible. Only at sites where hihi are habituated to people due to intensive hihi management (e.g. Tiritiri Matangi Island) can nest boxes be checked when adults are still present. If this approach is to be used then careful observation should be made to ensure no nests are deserted or other negative outcomes occur. Nest desertion associated with this practice has not occurred on Tiritiri Matangi.

Q. Does frequent checking of nests cause parents and/or their young any harm?

A. There is no evidence that the level of disturbance caused by management checks on hihi nests causes any harm to either parents or their young. Rather, management procedures such as mite treatment and nest box cleaning are in place precisely to maximise the reproductive success of hihi. See Box 7.1 for the proven benefits afforded by mite treatment.

Q. What is the best material to make an artificial nest with, when replacing mite-infested nests?

A. The most important considerations when making an artificial nest for hihi are that it is accepted by the parents and their young, and that it provides the nestlings with a safe and hygienic environment. It doesn't necessarily have to look like the real deal, so long as the birds accept it as such. A number of different materials have been used in the past to make artificial nests. We currently recommend using a plastic sieve lined with polar fleece. The sieve has similar dimensions to the stick cup, and the polar fleece performs the soft and insulating properties of

the stick cup lining. However, other items that perform similar functions may work just as well including a malleable wire mesh. Old natural nests (removed from nest boxes) were used in the past but these are a pathogen risk and should be avoided.

Q. Why do we use Frontline® and not other forms of miticide such as powder?

A. Frontline® Spray is a product designed for the treatment of fleas and ticks in domestic cats and dogs. It is available from most pet shops, see www.frontlineplus.co.nz for more information. The active ingredient is fipronil. We currently use Frontline® Spray on hihi nestlings because it is safe and effective at treating mites (see Box 7.1). In the past a miticide powder has been used, but it required larger volumes than Frontline®, was messy in the nest and appeared to be more irritating to the birds. However, Frontline® spray is not appropriate for treating nest boxes, and a miticide powder may be more suitable in circumstances where mites cannot easily be cleaned from the boxes.

7.3 Suggested further reading

Armstrong, D.P.; Castro, I.; Griffiths, R. 2007: Using adaptive management to determine requirements of re-introduced populations: the case of the New Zealand hihi. *Journal of Applied Ecology 44*: 953–962.

This article provides a fantastic example of adaptive management in hihi and includes focus on mite management of the hihi population that was on Mokoia Island. It gives one (good) example of how targeted monitoring can be so informative for management decisions.

Ewen, J.G.; Thorogood, R.; Brekke, P.; Cassey, P.; F. Karadas, F.; Armstrong, D.P. 2009: Maternally invested carotenoids compensate costly ectoparasitism in the hihi. *Proceedings of the National Academy of Sciences (USA) 106*: 12798–12802.

This article provides the first quantitative test of how detrimental mites can be to reproduction in hihi. Whilst it is packaged in a novel way to also look at how nutrition can help offset parasitism, the basic information is in here to judge the benefit of the hard work of managing mites. That said, it also provides some of the ingredients to model the population response to discontinuing mite management. A future modelling study on the cards... ?

Stamp, R.; Brunton, D.; Walter, B. 2002: Artificial nest box use by the North Island saddleback: effects of nest box design and mite infestations on nest site selection and reproductive success. *New Zealand Journal of Zoology 29*: 285–292.

and

Berggren, A. 2005: Effect of the blood-sucking mite *Ornithonyssus bursa* on chick growth and fledging age in the North Island robin. *New Zealand Journal of Ecology 29*: 243–250.

These two papers provide examples of mite infestation in other New Zealand bird species on Tiritiri Matangi island and also illustrate that there is not always an obvious cost to being parasitised with these mites.

7.4 Additional literature cited in text

Powlesland, R.G. 1977: Effects of the haematophagous mite *Ornithonyssus bursa* on nestling starlings in New Zealand. *New Zealand Journal of Zoology 4*: 85–94.

Powlesland, R.G. 1978: Behaviour of the haematophagous mite *Ornithonyssus bursa* in starling nest boxes in New Zealand. *New Zealand Journal of Zoology 5*: 395–399.

Rippon, R.J.; Alley, M.R.; Castro, I. 2011: Causes of mortality in a nestling population of free-living hihi (stitchbird, *Notiomystis cincta*). *New Zealand Journal of Zoology 38*: 207–222.

Hihi Best Practice Sheet 10: monitoring nests in nest boxes

A great deal of information can be gathered from hihi pairs that nest in nest boxes. As with all hihi management work, high standards of hygiene must be maintained when handling nests, eggs and chicks. The recommended procedure for monitoring nest-box nests, from detecting nesting behaviour through to the completion of the nesting attempt, is detailed below, with the necessary equipment highlighted in bold. Note that some procedures are optional extras that are detailed in other Best Practice Sheets.

1. All data collected during nest monitoring should be recorded (see **Template Record Sheet 4** for example of how to do this).
2. Always wash your hands with an **antibacterial hand-gel** after you have finished checking a nest box, and before you go to check the next nest box (at all stages of the nesting cycle).
3. At the start of the season, visit nest boxes approximately once a week to determine whether nest building has begun (see Fig. 7.3). When checking nest boxes, gently open the lid, do not obstruct the entrance, and avoid any sudden movements that will disturb the box and any birds inside.
4. When checking nest boxes for nest building, use **binoculars** to sight the bands of the resident female and the territorial male. Record the band combinations in a **waterproof notebook**. There may be additional males visiting the territory, but the territorial male will be calling and chasing off other males.
5. As nests reach a more advanced stage, check nest boxes more regularly. Once nests are completely built (with a fully lined nest cup atop a stick base), nest boxes should be checked daily for egg laying. At this point it is worth re-checking the identity of the breeding female and male, as they may have changed since the last check. Getting the correct identity of breeding pairs is one of the most difficult things in our experience of monitoring hihi breeding on Tiritiri Matangi. Frequent checking is always a recommended strategy!
6. The female will usually lay her eggs in the morning, so check for egg laying in the afternoon. Be aware that eggs are sometimes covered with loose nest-lining material, so move this to one side when checking for eggs.
7. Once the first egg has been laid, continue to check the nest daily (in the afternoon) to determine how many eggs are laid and when incubation begins.
8. Normally a female will lay between 3 and 5 eggs in a single clutch. Incubation usually begins on the day the last egg is laid, although be aware that females sometimes mock-incubate before the completion of the clutch. Also, females will sometimes miss a day of laying so that a laying gap will appear in the sequence.
9. To confirm that a female is incubating, watch the nest for an extended period. An incubating female will be in the nest box for about 15–30 minutes at a time with 5 minute breaks for foraging.
10. Do not check the nest box once incubation has begun. Incubation normally lasts 14–15 days.
11. On day 13 after the start of incubation, start checking the nest daily for hatching. When checking for hatching, record the number of unhatched eggs and the number of chicks in the nest. Hihi chicks within a clutch will occasionally hatch over a 2–3-day period.
12. Any eggs that remain unhatched 4 days after the last chick hatched should be carefully removed from the nest. They are less likely to break if you hold them at the ends. Sometimes parents remove semi-hatched (dead) eggs from the nest, so if any are missing, search the surrounding area.
13. OPTIONAL EXTRA: unhatched eggs can be analysed to determine whether hatching failure was due to infertility or embryo death. See **Hihi Best Practice Sheet 8** in Chapter 6 for this procedure. If you will be doing this, use a pencil to label the rounded end (rather than the pointed end) of the egg with the date and the nest box ID.
14. Use a **small pot** padded with **cotton wool** to transport eggs away from the nest, for storage in the fridge. If unhatched eggs are not being kept for analysis, they can be disposed of well away form the nest.
15. Continue checking nests daily once chicks have hatched, until they are 10 days. When chicks are older than 10 days, check nests every other day. The purpose of these checks is to monitor nestling survival and condition and to check for nest mites. Protocols for approaching and checking nests vary between sites. On Tiritiri Matangi, for example, nests are typically checked immediately by carefully opening the nest box lid and allowing the female to exit if she is on the nest. This is done because there are often too many nests to check within a single day and the work would not be possible if we waited to ensure no adult

Continued on next page

is present in the box before checking. No instances of nest desertion have been recorded on Tiritiri Matangi and the breeding adults quickly return to the nest after disturbance. However, at other sites the protocols are different. At Zealandia, for example, nest checkers must observe the box for at least 10 minutes to confirm no adults are inside before approaching and opening the box. We recommend the latter, if at all possible, as a best strategy and would only encourage the former if the birds are well habituated to people and intensive hihi management (such as the case on Tiritiri Matangi).

16. If a chick is found dead, remove it from the nest.
17. OPTIONAL EXTRA: if sexing and genotyping data are required, dead chicks can be processed according to the protocol in **Hihi Best Practice Sheet 9** in Chapter 6. If you will be doing this, place the dead chick in a **zip-lock bag** and use a **waterproof marker pen** to label with the date and the nest ID or process the chick as detailed in the above best practice sheet. If a chick is missing from the nest, search for it in the surrounding area. Dead chicks may be found between the nest and the nest box wall, on the floor of the nest box, on the ground beneath the nest box, or on the ground some distance from the nest box (within about a 10 m radius of the box). Dead chicks can often be found by following the flies, or checking beneath perches that parents frequently use (indicated by collection of egg shell fragments). Store dead chicks in zip-lock bags in the fridge until processing.
18. If dead chicks are not being processed, they can be disposed of well away from the nest.
19. When a nest containing chicks is checked, the nest and chicks should be inspected for mites. The protocol for this procedure, and what to do if mites are detected, is detailed in **Hihi Best Practice Sheet 12**.
20. When chicks are 21 days they should be banded and have measurements taken and recorded (see **Hihi Template Record Sheet 4** for an example record). The protocol for this procedure is detailed in Chapter 5.
21. Once chicks have been banded, nests should not be checked until chicks are very close to fledging. Hihi chicks usually fledge at around 30 days, so nest boxes should be checked from 29 days onwards to confirm fledging. Care should be taken so that checking does not cause chicks to fledge prematurely. Only lift the lid of the nest box high enough to confirm that chicks are still inside, and cover the entrance to the nest box with your hand whilst you do so. Often you can hear the chicks inside on approaching the nest and this may be enough to confirm at least one chick remains in the nest. The detail you require will directly reflect the monitoring needs.
22. When all chicks have fledged (or the nesting attempt has failed), remove the nest box from the tree and clean according to the protocols in **Hihi Best Practice Sheet 1** in Chapter 3.

Figure 7.3. Monitoring hihi nest building: A. Box with ¼ stick base. B. Box with ½ stick base. C. & D. Two nests with nest cups in the process of being lined, although cups havn't yet been formed. E. A near complete nest with lining including feathers. F. The finished product with eggs. Note the nest with eggs has a short stick base, which highlights that it is worth regularly checking nest building, as some females build shorter nests and can complete the task quickly if they choose to. *Photos: Leila Walker.*

Hihi Best Practice Sheet 11: Finding and monitoring natural nests

Once found, natural nests that are easily accessible can be monitored using the same procedures as for nest-box nests (**Hihi Best Practice Sheet 10**). However, most, if not all natural nests are not easily accessible and will require different treatment. The recommended procedure for monitoring inaccessible natural nests, including how to find natural nests in the first place, is detailed below, with the **necessary equipment** highlighted in bold.

1. At the start of the breeding season (September–November, depending on the site), locate territories by walking the site to determine the position of calling males.
2. The nest location can be narrowed down initially by focusing on the location of the male calling, ideally prior to the start of incubation. Male calling will peak during the nest building stage, and males generally call most frequently from, or very close to, the nest tree itself. Once females start incubating, male calling rates decline, making finding the nest location more time-consuming. The presence of floater or extra-pair males, or a fertile female, can alter male calling patterns and pair behaviour. Attempting to find nests during these times can be more difficult and should be avoided, where possible.
3. Once the general nest area has been narrowed down, confirm the presence of a female. Some males will defend a territory even if they are unpaired (this can be particularly common in newly established or male-biased populations), so care should be taken to ensure a female is present before investing time in nest-searching. Sighting the female is easiest prior to or during the nest building phase, or when chicks have recently fledged. Female behaviour can be quite cryptic during the breeding season, especially in male-biased populations where levels of male harassment are high. Following the male, listening for contact calls, and plenty of patience, should enable confirmation of female presence. Additionally, the presence of a higher than usual number of males at a territory can be a good indicator that a fertile female is in the area.
4. Nests can be located in a wide range of tree species, with the most commonly used species varying between sites. On Te Hauturu-o-Toi / Little Barrier Island, the most commonly used species have been pūriri, pōhutukawa, rātā and taraire; on Kapiti Island, pukatea, hīnau, kāmahi and rimu; at 'Ark in the Park' in Auckland's Waitakere Ranges, all recorded nests were in kauri; and at Maungatautari, nests have been predominantly found in pukatea but also tawa, māhoe and rimu. Nests are usually several metres high (even up to 30 m), but can occasionally be lower or even at ground level. Hihi favour large cavities with small entrances, which are either covered by overhanging vegetation or a bark lip. Cavities will sometimes be visible from the ground, but can sometimes be obscured by vegetation in denser forest (e.g. rātā vine or epiphytes can reduce visibility). On occasion it will not be possible to identify the exact cavity without climbing the tree.
5. Locating the nest cavity can be done at any time in the breeding cycle, but is easiest prior to the start of incubation. Sighting birds carrying sticks/nest material can easily lead to the nest site. If the female is incubating, she will usually give a single 'stitch' call upon leaving the nest – listening for this call and narrowing in on its location can lead to the nest site. During chick feeding, the female and male (to varying degrees) will come and go from the nest, and when chicks are close to fledging it is often possible to hear chicks in the nest from ground level.
6. Once chicks are fledged, locating the nest site is generally not possible, but it does provide an opportunity to confirm breeding has occurred, the number successfully fledged, identify the female and social male, and provide an approximate location of the territory.
7. Once a nest cavity is found, use the activity around the nest to determine the stage of nesting. At natural nest sites egg laying starts an average of 9 days after the female has completed her nest. The calling rate of male hihi will increase dramatically when the resident female is close to laying, and there will be an increase in the number of other males present around the nest site.
8. To confirm that a female is incubating, watch the nest for an extended period. An incubating female will typically be on the nest for about 15–30 minutes with 5 minute breaks for foraging. Different incubation patterns may occur and you need to be careful that your presence is not disturbing the female's behaviour.

Continued on next page

Hihi Best Practice Sheet 11 continued

9. The pattern of female visitation to the nest will change when the chicks have hatched. Females will spend less time on the nest and more time foraging as the chicks grow older. Eggshell fragments below the nest may also indicate that chicks have hatched.
10. If adult bird visitation to the nest stops completely when the chicks are estimated to be around 30 days, then the chicks have likely fledged. Careful observation at this time can be used to estimate how many nestlings have fledged by locating them and observing adults feeding them outside of the nest. If activity at the nest stops much before this, the nest has likely failed.
11. Mark the location of the natural nest with **flagging tape** and use a **waterproof marker pen** to label the flagging tape with the nest ID. Also mark the location on a **GPS**.
12. Record band combinations of the adults (using **binoculars**), or note as un-banded (if this is the case), in a **waterproof notebook**.
13. The level of monitoring required for natural nests will be determined by requirements set by HRG or permit conditions, as well as logistical constraints. Tree climbing should only be attempted by certified tree climbers, and banding attempts of chicks in these nests should only occur under consultation with HRG and by an approved banding operator. Banding of chicks in more accessible natural nests should be undertaken with care and only where the operator is confident this can be carried out without damaging the nest site or placing the chicks at risk of harm.
14. Where possible and if meeting monitoring needs (see above point), when nestlings have fledged from inaccessible nests they will need to be caught for banding and measurement. The protocol for this procedure is detailed in **Chapter 5**.
15. All data collected during nest monitoring should be recorded (see **Hihi Template Record Sheet 5** for an example of how to do this).

Hihi Best Practice Sheet 12: Detecting and treating mites

Some hihi populations suffer from mite infestations in nest boxes, and these can cause chick death. Mites can be detected in the nest, on the nest box and on the chicks themselves. The recommended procedure for detecting and treating mites is detailed below, with the **necessary equipment** highlighted in bold.

1. Be aware that you may be less likely to detect mites if your hands are very cold. In cold weather, consider wearing gloves to keep your hands warm and therefore improve your chances of detecting mites. Being handled by cold hands will probably be unpleasant for the nestlings as well.
2. During nest checks, once chicks have hatched, put your fingers inside the nest cup and gently disturb it. After about 20 seconds remove your hand from the nest cup and check for mites crawling on your hand. Hihi mites are about 2–3 mm long and can be dark brown to red (after a blood meal) or completely transparent (just hatched).
3. Inspect the box for mites. Pay particular attention to the lid and any cracks or crevices. Avoid resting the lid on your head, as any mites present may transfer to you.
4. If the chicks are older than 6 days, inspect the chicks for mites. This is a relatively quick procedure that can be done whilst standing at the nest box. Remove each nestling in turn and inspect the exposed skin and the head feathers for mites. Mites are often found around the nestling's eyes and under the wings.
5. Also pay attention to the behaviour of the parents. They will scratch vigorously after leaving a nest infested with mites.
6. If mites are detected, the chicks should be treated with **Frontline® Spray**. Whilst standing at the nest box, spray a ball of cotton wool with Frontline®, remove each nestling in turn, and apply the Frontline®-soaked cotton wool to the nestling's exposed skin. Do not spray Frontline® directly onto the chicks as this risks spraying into their eyes and mouth. If nestlings are very wet after applying Frontline®, they can be cupped in your hands for a short while to dry off, before being returned to the nest. Make sure the nestlings have settled back down into the nest cup before leaving. They can be irritated by the application of Frontline® and there is a risk they could fall out of the nest.
7. If a very large number of mites (>100) are detected, the nest box should be replaced with a clean nest box and the nest itself replaced with an artificial nest. This should be done in addition to treating the chicks with Frontline®, as detailed above.
8. The replacement nest box should have been cleaned according to **Hihi Best Practice Sheet 1** in Chapter 3. To make the artificial nest, take an 8–10 cm diameter **plastic sieve** with the handle removed and line it with **polar fleece**. Place this inside the replacement nest box, jamming it securely in the base of the box.
9. Remove each nestling from the mite-infested nest and place all nestlings together into a clean **bird bag**. Temporarily hang the bird bag in a safe place (never place a bird bag with birds in it directly on the ground!).
10. Remove the mite-infested nest box from its backboard and empty contents onto forest floor about 10 m away (having already removed nestlings to a bird bag).
11. Using a **bottle** filled with **tap water** douse the backboard and remove any faecal material etc. with a **scrubbing brush**. Pat dry with **paper towels**.
12. Attach the replacement nest box containing the artificial nest to the backboard, and return the nestlings to the artificial nest. Ensure they are settled into the new nest before closing the lid. From a distance of 10–15 m, observe the nest box to confirm that the female returns and has accepted the new nest.
13. The mite-infested nest box should be taken away and cleaned according to **Hihi Best Practice Sheet 1** in Chapter 3.
14. Any mite treatment should be recorded (see **Hihi Template Record Sheet 4** for an example of how to do this).

Hihi Best Practice Sheet 13: Estimating the age of hihi nestlings

Day 0 – hatch day

- Red / dark pink skin
- Eyes closed
- Grey fluff on head and back
- Might be wet

Day 1

- Pink skin
- Eyes closed
- Grey fluff on head and back

Day 2

- Pale flesh-coloured skin
- Eyes closed
- Grey fluff on head and back

Day 3

- Flesh-coloured skin
- Eyes closed
- Grey fluff on head
- Dark strip on back and wings

Day 4

- Flesh-coloured skin
- Eyes closed
- Grey fluff on head
- Dark strip on back and and wings more prominent than on day 3

Day 5

- Flesh-coloured skin
- Small eye slits
- Grey fluff on head
- Dark strip on back and wings
- Wing pins
- Grey tail and strips on side of body

Day 6

- Flesh-coloured skin
- Larger eye slits
- Grey fluff on head
- Dark strip on back
- Wing pins
- Grey tail and strips on side of body

Day 7

- Flesh-coloured skin
- Wide eye slits
- Grey fluff on head
- Back pins
- Longer, darker wing pins
- Grey tail and strips on side of body

Day 8

- Flesh-coloured skin
- Circular eye opening
- Grey fluff on head
- Back pins
- Wing pins
- Tail and body pins

Day 9

- Flesh-coloured skin
- Eyes open
- Small head pins
- Some feathers broken on back
- Wing pins
- Tail and body pins

Day 10

- Flesh-coloured skin
- Eyes open
- Small head pins
- Back feathers
- Wing feathers broken
- Tail and body pins

Day 12

- Flesh-coloured skin
- Eyes open
- Head pins
- Back feathers
- Wing feathers
- Tail feathers broken

Day 14

- Eyes open
- Short head feathers
- Long back feathers
- Wing feathers (10–20 mm)
- Tail feathers (10–20 mm)
- Body feathers

Day 16–19

- Eyes open
- Mostly feathered
- Some pins still visible on tail and wings

Can band at this age

Day 21

- Fully feathered
- No fluff on head
- Look like an adult female with a yellow gape

Need to be banding at this age

Hihi Template Record Sheet 4: Nest monitoring (nest boxes)

This is an example record sheet from Tiritiri Matangi Island, used for monitoring nesting attempts at nest boxes. This version includes example data to illustrate how it should be used. Blank versions for photocopying are included at the end of this handbook.

Season (year): 2013/14	Site: Tiritiri Matangi
Box number: B22/25	Female parent: RD OR/PI MT
Clutch: 1	Male parent (social): BK DB/OR MT

Date	Day-by-day summary: egg laying, nestling death, mite presence and treatment
16/10	1st egg
20/10	5th egg laid, female starts incubating
03/11	3 eggs hatch (2 eggs remain)
04/11	1 chick hatches (1 egg remains)
08/11	1 unhatched egg removed from nest
11/11	1 chick found dead on ground approx. 2 m from nest box, no food in crop
15/11	Mites detected on all 3 nestlings, treated with Frontline
19/11	Very large number of mites on nestlings and box – nest box replaced, and nest replaced with artificial nest
23/11	3 nestlings banded
01/12	3 nestlings still in nest
02/12	All nestlings gone – fledged!

Nest summary information

Clutch details		*Unhatched egg sample details*	
No. eggs laid:	5	Unhatched egg 1: separated/gunk/MDE/LDE/FFC tissue sample id	1
Date 1st egg laid:	16/10	Unhatched egg 2: separated/gunk/MDE/LDE/FFC tissue sample id	
No. eggs hatched:	4	Unhatched egg 3: separated/gunk/MDE/LDE/FFC tissue sample id	
No. chicks fledged:	3	Unhatched egg 4: separated/gunk/MDE/LDE/FFC tissue sample id	
		Unhatched egg 5: separated/gunk/MDE/LDE/FFC tissue sample id	

Fledged chicks summary information

Chick details	Chick 1	Chick 2	Chick 3	Chick 4	Chick 5
Chick band:	BK WH–LB MT	RD RD–LB MT	PK DB–LB MT	–	–
C-band:	88891	88892	88893		
Tarsus (notch) (mm):	26.6 / 26.5	27.2 / 27.3	27.2 / 27.2	/	/
Tarsus (full) (mm):	31.0 / 30.9	31.1 / 31.0	32.2 / 32.2	/	/
Weight (g):	34.50	37.42	45.59		
HB (mm):	38.0 / 37.9	38.2 / 38.1	40.4 / 40.5	/	/
Blood sample (✓)	✓	✓	✓		

Dead nestling 1 age (days)	9	Tissue sample id	2	Dead nestling 2 age (days)		Tissue sample id	
Dead nestling 3 age (days)		Tissue sample id		Dead nestling 4 age (days)		Tissue sample id	
Dead nestling 5 age (days)		Tissue sample id					

Instructions:

1. Complete one nest sheet per nesting attempt.
2. When recording clutch number, specify whether it is a replacement of a previous unsuccessful attempt (e.g. replacement-1 would be a replacement first clutch).
3. Double and triple check that parent band combinations are correct.

Continued on next page

Hihi Template Record Sheet 4 continued

4. Day summaries only need to be recorded when something happens (e.g. eggs are laid, chicks hatch, chicks die, mites are found and treated or other interesting notes).
5. If unhatched eggs are being processed, circle egg content category, and record the sample number.
6. In event of nestling death, record the age of death and, if dead chicks are being processed, the sample number.
7. At banding, record the colour band combination, the C-band number, and measurements. We take repeat measurements of tarsus and head bill to assess our repeatability in these data (see Q & A in Chapter 5). We also measure two different options for tarsus because of researcher preferences and longer-term data collection consistency. If blood samples are being taken, confirm with tick. Ensure blood sample is clearly labelled with the bird's ID and the date the sample was taken. Remember that alcohol leaking onto labels will destroy them unless they are pencil written on paper – this is a strongly recommended backup method of labelling and should be added into every tube.

Hihi Template Record Sheet 5: Nest monitoring (natural nests)

This is an example of a record sheet used to monitor natural nests, and includes example data to illustrate how it should be used. Blank versions for photocopying are included at the end of this handbook.

Season (year): 2013/14	Site: Maungatautari
Nest ID: HIB 12	Female parent: Unbanded
Clutch: 1	Male parent (social): YE YE-YE MT
Nest location (description): In pukatea cavity approx. 15 m NE of tracking tunnel 12	
GPS coordinates: E1825884/N5790290	

Date	Day-by-day summary: egg laying, incubation, fledging
25/09	Nest site located - female visiting with nesting material @ 15:00 hrs
27/09	Nest observed for 1 hour - female still taking nesting material to nest
02/10	Nest observed for 1 hour - no nest building activity, male only present intermittently
05/10	Territorial male calling a lot, 3 other males near nest site (RD BK/OR MT, 1 unbanded and 1 un-identified) during 30 min observation, observed 2 chases of female
08/10	Territorial male still calling lots, 2 other males (both unbanded) observed near nest site during 45 min observation
11/10	Nest observed for 1 hour (10:00-11:00) - female observed leaving nest cavity twice at 10:22 (for 5 mins) and 10:45 (for 5 mins), little male activity around cavity
20/10	Nest observed for 1 hour - female left 3 times, for approx. 5 mins each time
30/10	Female leaving nest more frequently then previously
05/11	Nest still active, female leaving for 5-10 mins every 20 mins
10/11	Nest still active
15/11	Nest still active - can hear nestlings begging during feeding visits
20/11	Nest still active
25/11	No activity at nest, female observed with 2 fledglings approx. 30 m from nest cavity

Nest summary information

Estimated date(s) of egg laying: 5-10 Oct	Tree species: Pukatea
Estimated incubation start date: 9-11 Oct	Nest height: 15 m
Estimated date of fledging: 25 Oct	Nest accessible? (Yes) / No
Estimated number chicks fledged: >2	

Fledged chicks summary information

Chick details	Chick 1	Chick 2	Chick 3	Chick 4	Chick 5
Chick band:	/	/	/	/	/
C-band:					
Tarsus (notch) (mm):	/	/	/	/	/
Tarsus (full) (mm):	/	/	/	/	/
Weight (g):					
HB (mm):	/	/	/	/	/
Blood sample (					

Instructions:

1. Complete one nest sheet per nesting attempt.
2. When recording clutch number, specify whether it is a replacement of a previous unsuccessful attempt (e.g. replacement-1 would be a replacement first clutch).
3. Double and triple check that parent band combinations are correct. If they are unbanded, be sure to record this fact.

Continued on next page

Hihi Template Record Sheet 5 continued

4. Day summaries can be used to record frequency of parents' visits to the nest, to determine stage of nesting.
5. Egg laying, incubating and fledging dates should be estimated as accurately as possible, using the behavioural observations in the day summary.
6. Record the tree species and the height of the natural nest, and indicate whether the nest is accessible.
7. If the cavity is accessible and nestlings can be banded at approximately 21 days, there is space to record this information.

8. Monitoring hihi survival

Monitoring the survival of individuals is a crucial part of monitoring the health of a population. Yearly survival estimates can be used to estimate population size and, in combination with estimating reproduction, can be used to measure population growth, predict future population trajectories and estimate sustainable harvesting limits. The most suitable method for monitoring survival will depend on the site and the monitoring objectives. Site area and terrain, and population size and density should be considered when determining the most appropriate technique for monitoring hihi survival. Here we detail how hihi are currently monitored. Two alternative methods are currently being used in hihi populations: population surveys by re-sighting marked individuals and distance sampling of predominantly unmarked individuals. The suitability of the two survey methods in different situations, and how they should be applied, is detailed below.

8.1 Population surveys

We view this method of monitoring as the gold standard approach in hihi. Population surveys are ideal in populations (such as hihi) where:

1. Birds are individually colour-banded.
2. The population can feasibly be surveyed in a relatively short and focussed effort.
3. Surveys can be regularly repeated using standard methods.

Population surveys are currently conducted at Tiritiri Matangi Island (Box 8.1), Kapiti Island (Box 8.2), Zealandia, Maungatautari, Rotokare and Bushy Park. Surveys should ideally be conducted at least once a year, at the start of the breeding season. If resources and time are available, then additional surveys can be done. For example, a second population survey is done at the end of the breeding season on Tiritiri Matangi (Box 8.1). The basic protocol when conducting a population survey is to record the identity of as many banded individuals as possible during a standard number of hours and a standard area of searching. The standardised time and area searched ensures numbers are comparable between years. At some sites population surveys are conducted entirely at feeding stations (Box 8.2); whilst elsewhere, feeder observations are combined with walks of the site (Box 8.1). Surveying should not be conducted during prolonged periods of rain or strong winds, as there is a greatly reduced probability of detecting birds.

Surveying at feeding stations involves positioning yourself at a 5–10 m distance from the feeder, and using binoculars to read the colour band combinations of all hihi you see. How band combinations are read, and the standard abbreviations for the band colours used at different sites, are detailed in Chapter 5. Survey walks should cover locations that hihi are known to occupy, and potential areas that they may be moving in to. The sex and colour band combination of all hihi detected should be recorded in a waterproof notebook, and later entered into an electronic datasheet (see **Hihi Template Record Sheet 6** for an example datasheet). Surveying involves detecting hihi calls as well as seeing hihi, and often you will have heard a hihi long before you can track it down and read its band combination. It is important that you persevere with finding a hihi that you may initially only hear.

It is surprisingly easy to make mistakes when reading a bird's band combination and when recording the combination in your notebook. It is therefore worthwhile to double- and triple-check both your reading and your records. Sometimes certain colours can have a different appearance in direct sunlight or in shadow, and in these cases it is worth spending extra time to ensure you are confident in your observation. With experience you will come to know which colours require extra attention. You should also be aware that colour bands occasionally slip

Box 8.1 Monitoring survival on Tiritiri Matangi Island

On Tiritiri Matangi Island, pre- and post-breeding surveys have been conducted in late September and late February, respectively, since the population was established in 1995. The population surveys are done over 40 hours, spread over roughly one week, and combine feeder observations with walks of the site. Normally a team of about 3–5 people are involved in the Tiritiri Matangi surveys. This helps easily achieve the required hours and also provides opportunity for people interested in hihi conservation to get involved.

Some key findings:

Regular population surveys on Tiritiri Matangi Island have generated a number of key findings. For example, Ewen et al. (2011) used 12 years of intensive monitoring data to assess the demographic effects of fluctuations in sex ratio. Increased male harassment of females in male-biased populations may be costly for females in terms of injury and/or altered breeding behaviour, and this may have population-level consequences. However, Ewen et al. (2011) found that a changing adult sex ratio had little or no effect on adult female survival (Fig. 8.1) or on reproduction. This long-term survival dataset in combination with monitoring of reproduction has also been used to guide harvesting of the population for reintroduction to new sites (Armstrong & Ewen 2013). The high-quality data available has made it possible to make accurate pre-harvest projections of the impact on this hihi population (Fig. 8.2). The current harvesting strategy is designed to maintain the population near 70 females, because this gave good numbers for harvests and prevented ongoing management on Tiritiri Matangi from becoming overwhelming.

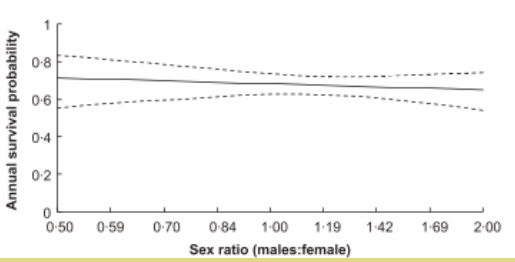

Figure 8.1. Estimated relationship between annual survival of adult females and population sex ratio, using re-sighting data from September 1995 to February 2008. Figure modified from Ewen et al. 2011.

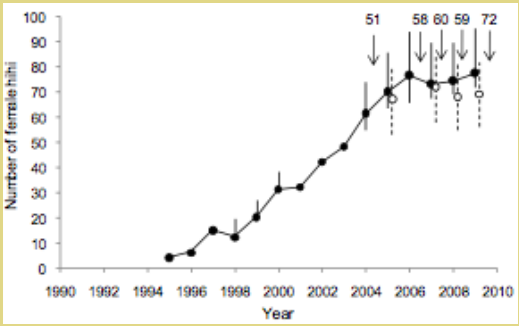

Figure 8.2. Application of population models for guiding harvesting of the hihi population on Tiritiri Matangi. Lines and filled circles show the estimated numbers of females at the start of each breeding season, arrows show numbers harvested, and open symbols show median pre-harvest projections with error bars showing 95% confidence intervals. Figure modified from Armstrong & Ewen 2013.

over or under each other, causing one colour to be partially obscured. Less frequently, colour bands can move above the intertarsal joint. A banding protocol has recently been introduced on Tiritiri Matangi whereby all birds are banded with 3 colours (i.e. not between 1 and 3 colours, as previously), so that an observer is immediately aware when a colour band is missing/obscured. We recommend that this protocol be adopted at all sites.

8.2 Distance sampling

Distance sampling is a viable option at sites where:

1. Hihi are not all colour-banded
2. The site is a large and access is difficult

Box 8.2 Monitoring survival on Kapiti Island

On Kapiti Island, October pre-breeding surveys have been conducted since 1993. The Kapiti population surveys are carried out over 36 hours, and observations are spread evenly between feeding stations and over an entire month.

Key findings:

These regular surveys have been used to assess the importance of supplementary feeding for hihi on Kapiti Island. Chauvenet et al. (2012) made use of 18 years of survey data to evaluate the merits of an ad libitum feeding regime over a regime where limited food was provided. They found that the provision of supplementary sugar water ad libitum positively affected the survival and abundance of adult hihi (Fig. 8.3), but that recruitment, despite increasing initially after the ad libitum regime begun, has more recently started to decline.

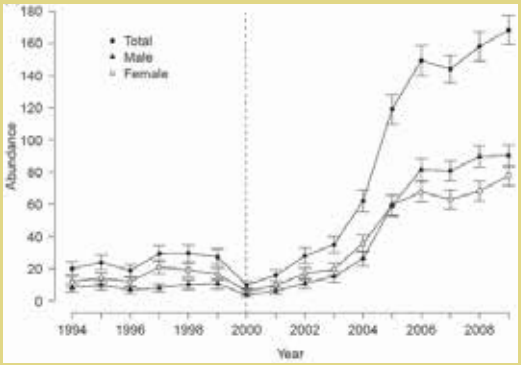

Figure 8.3. Estimates of male, female and total abundance between 1993 and 2000 (when little-to-no supplementary food was provided) and post-2000 (when ad libitum food was provided). Figure modified from Chauvenet et al. (2012).

Distance sampling has recently been used on Te Hauturu-o-toi / Little Barrier Island, and trialled at Maungatautari. On Hauturu, distance sampling was carried out each year over 3 weeks in late September and early October. Distance sampling was trialled at Maungatautari during one week in October 2012 when the size of the population was known quite accurately, with the aim of truthing the method and establishing its suitability for future use at this site (see Box 8.3 for further detail).

Distance sampling involves estimating the distance a detected individual is from a transect line or point. A model that captures how detectability decreases with increasing distance can then be used to estimate the number of individuals that have been missed and, consequently, to estimate population size. Line transects usually produce more data than point transects, and are less sensitive to inaccuracies in distance measurements and to immigration into the sampling area. For these reasons, point transects are normally only used where line transects are impractical due to dense vegetation and/or steep terrain. On Te Hauturu-o-toi / Little Barrier Island, a network of 148 observation points covering 600 ha has been used in the SW sector of the island, and a network of 58 observation points covering 230 ha is used in the NE sector. At Maungatautari, 53 × 500 m transect lines were used in 2012.

There are three important assumptions for distance sampling: 1) all birds at the observation point, or on the transect line, are detected; 2) all birds are detected at their initial location; and 3) all distances are measured accurately. These assumptions must be met for estimates of population size to be accurate. Furthermore, as a general rule of thumb, at least 60–80 observations and a minimum of 40 transects need to be observed for distance-based estimates of population size to be precise. Even so, it can be difficult to obtain a precise estimate.

It is recommended that the following methodology be adopted for distance sampling with hihi:

1. The observer should move through the bush very quietly, to limit birds fleeing from them or being attracted to them.
2. Care should be taken to measure distance to the location where the bird is first detected. This distance is perpendicular to the transect line rather than at the angle where the bird may have first been observed (unless this angle is measured to calculate the perpendicular distance from the line).

Box 8.3 Monitoring hihi survival at Maungatautari

At approx. 3400 ha in area, Maungatautari is the largest of the current hihi sites, closely followed by Te Hauturu-o-toi / Little Barrier Island. As with Te Hauturu-o-toi / Little Barrier Island, the large size of Maungatautari has made designing cost-effective and accurate monitoring techniques challenging. In contrast to Te Hauturu-o-toi / Little Barrier Island, in the early years post-release the Maungatautari population has been at low-density, with the birds' ability to disperse widely across the site presenting an additional challenge. A range of methods have been used to monitor hihi survival at the site, and the merits of each are outlined below. The advantages and challenges of hihi monitoring at Maungatautari are summarised in Table 8.1.

Feeder monitoring (2009–ongoing)

Feeder monitoring was initially carried out by volunteers, and also by researchers and contractors. The results provided a minimum estimate of initial post-release survival of hihi after translocation, and enabled the banding of some site-bred birds.

Mark-recapture surveys (2010, 2014–ongoing)

Mark-recapture surveys have been carried out in 2010, and every year since 2014, with intensive banding efforts carried out prior to commencements of all surveys since 2014. All surveys have been carried out by contractors with previous hihi experience. In some years, repeat surveys were carried out in areas of higher hihi density to enable sufficient detections to generate detection probabilities.

Territory mapping (2010–14)

Territory mapping across the site was carried out from 2010 through until 2012/13 as part of a research project, and by a contractor in 2013/14. This meant that hihi population size was closely monitored, with almost all adults banded during this time period. Field staff and volunteers involved in pest eradication and surveillance were (and still are) encouraged to report sightings.

In 2013 and 2014, one-week volunteer surveys were undertaken to assist in territory mapping. In 2014, the volunteer survey preceded the contractor mark-recapture survey, and these two methods together provided a minimum known number of birds (from territory mapping) and population estimate with condidence intervals (from mark-recaprure survey). Pairing volunteers with prior experience with local volunteers aimed both to foster local interest in hihi and develop skills among the volunteer base that will aid continuity of hihi monitoring at the site.

Distance sampling (2012)

Distance sampling using line transects (53 × 500 m lines) across the site was carried out in 2012, at a time when approximate population size was known from territory mapping. Population estimates generated (estimate = 431, lower 95% CI = 218, upper 95% CI = 864) were considerably higher than known population size (70–80), and this method has not been used again for this site. Reasons for this discrepancy are the focus of on-going analysis and may be explained by using sightings and distances based on calling but not seen birds. The recovery group continues to develop and assess methods for monitoring these difficult populations.

Some lessons learnt

1. It is both important and difficult to identify long-term monitoring methods suited to individual site and population needs.
2. There is great value in long-term collaborations between community conservation projects and external institutions (universities, DOC, recovery groups).
3. Monitoring is a valuable way of meeting another of the HRG fundamental objectives to 'increase public appreciation' of hihi (see Chapter 1). This is done by encouraging

Continued on next page

Box 8.3 continued

involvement of volunteers and on-the-ground staff. In doing so we can ensure others others understand why the information they can provide is useful. The importance of providing a positive experience to those participating in surveys should not be underestimated, particularly where volunteer input is crucial.

4. There is value in integrating species-specialist knowledge with that of 'on-the-ground' people, providing training to key staff and volunteers and ensuring continuity.

Table 8.1. Advantages and challenges of monitoring hihi at Maungatautari Ecological Island.

ADVANTAGES	CHALLENGES
Pest monitoring lines are spaced no more than 200 m apart across the 3400 ha site.	Size/terrain/vegetation, maintenance of pest monitoring lines.
ATV access across site and around fenceline.	Resourcing ATVs/vehicles, certification needed.
Farm access to site in majority of places.	Farm access to site restricted in some areas.
Male hihi are vocal and territorial during the breeding season.	Female hihi quiet and behaviourally cryptic during breeding season, especially in early years of population.
Staff and volunteers are regularly present on-site to monitor pests.	Hihi are a species less familiar to public, especially in early years post-reintroduction.

3. Do not record birds that fly through the sampling area unless you can identify the exact location that they flew from.
4. Generally, distance should only be measured to a bird that has been seen, as it is very difficult to accurately estimate a bird's location from its call.
5. Distances should be measured using a range-finder or a tape measure, rather than being estimated. Range-finders may give inaccurate readings in dense vegetation or mist. Point transect data is particularly sensitive to errors in measuring distance (because the analysis involves calculating area from distance, and the error is consequently squared).
6. If multiple birds are detected together: record the birds as a cluster, note the number of birds in the group, and measure distance to the centre of the cluster.
7. Point transects assume no immigration into the area during the observation period. The observation period must therefore be sufficiently short enough that this assumption holds, but sufficiently long enough that all individuals close to the observer are detected.
8. Line transects should be walked at a speed that is sufficiently slow to enable all birds on the line and most nearby to be detected, but sufficiently fast that animals ahead of the observer do not flee and mobile birds are not counted twice.

8.3 Q & A: Why do we do it this way?

For a general description of the reason for this Q & A section, see Chapter 1. The following set of questions and answers address some of the recent trials and changes relating to monitoring hihi survival:

Q. If two people are working together during a population survey, should the survey time account for this? E.g. does two people surveying together for one hour count as one hour survey time, or two hours survey time?

A. In terms of observation hours, we would normally count two people conducting a survey in the same location as one person. So, two people working together in the same place for one

hour will equate to one hour of survey time. Often it can be helpful to have two people working together with, for example, one person calling out band combinations and the other recording band combinations. However, if two people are working independently (i.e. in separate locations) for one hour each, that will equate to two hours survey time. The most important thing is that a consistent approach to tallying hours is taken year-on-year.

Q. Should the number of survey hours increase as the population grows, so that the additional birds have more chance of being detected?

A. No. The effort, in terms of survey hours, should remain consistent year-on-year. It is therefore important that the number of survey hours determined at the start of a monitoring program (when population size is relatively small) is able to absorb an increase in population size.

Q. Does it matter if population surveys are only conducted at feeding stations?

A. The location at which population surveys are conducted represents a compromise between maximising the likelihood of detection, and being logistically practical. Therefore, population surveys may be conducted at feeding stations only for practical purposes (e.g. the terrain elsewhere is unsuitable for dedicated surveying). Likewise, reliance on feeding stations during population surveys may depend on their popularity during the survey period. If hihi simply aren't using the feeding stations, it may be sensible to look elsewhere. Again, the important thing is that the approach is as consistent as possible year-on-year.

Q. Does it matter if population surveys are conducted over a protracted period of time (e.g. one month) rather than, say, one week?

A. Surveys should be relatively short compared with inter-survey periods to provide meaningful survival estimates. On Tiritiri Matangi island we aim for one week's survey time twice per year. However, it is not always practical to conduct a population survey over such a short time, particularly if only one person is involved. Where only one or two surveys are done per year it is perfectly reasonable to conduct a population survey over a longer time frame, such as one month, as occurs on Kapiti Island. Conducting a population survey over a short time period reduces the likelihood that birds that are seen do not also subsequently die during the course of the survey. Again, the important thing is that the approach taken is consistent year-on-year.

8.4 Suggested further reading

Why should we monitor populations and how do we ensure monitoring is useful?

Armstrong, D.P.; Reynolds, M.H. 2012: Modelling reintroduced populations: the state of the art and future directions. Chapter 6 in Ewen, J.G.; Armstrong, D.P.; Parker, K.A.; Seddon, P.J. (Eds): Reintroduction Biology: integrating science and management. Wiley Blackwell, United Kingdom.

Nichols, J.D.; Armstrong, D.P. 2012: Monitoring for reintroductions. Chapter 7 in Ewen, J.G.; Armstrong, D.P.; Parker, K.A.; Seddon, P.J. (Eds): Reintroduction Biology: integrating science and management. Wiley Blackwell, United Kingdom.

Parker, K.A.; Ewen, J.G.; Seddon, P.J.; Armstrong, D.P. 2013: Post-release monitoring of bird translocations: why is it important and how do we do it? *Notornis 60*: 85–92.

Overviews of population modelling in hihi:

Ewen, J.G.; Armstrong, D.P. 2007: Strategic monitoring of reintroductions in ecological restoration programmes. *Ecoscience 14*: 401–409.

Armstrong, D.P.; Ewen, J.G. 2013: Consistency, continuity and creativity: long-term studies of population dynamics on Tiritiri Matangi Island. New Zealand Journal of Ecology 37: 288–297

Population modelling to inform hihi management:

Armstrong, D.P., Castro, I.; Griffiths, R. 2007: Using adaptive management to determine requirements of re-introduced populations: the case of the New Zealand hihi. *Journal of Applied Ecology 44*: 953–962.

Chauvenet, A.L.M.; Ewen, J.G.; Armstrong, D.P.; Coulson, T.; Blackburn, T.M.; Adams, L.; Walker, L.K.; Pettorelli, N.: 2012: Does supplemental feeding affect the viability of translocated populations? The example of the hihi. *Animal Conservation 15*: 337–350.

Chauvenet, A.L.M.; Ewen, J.G.; Armstrong, D.P.; Pettorelli, N. 2013. Saving the hihi under climate change: a case for assisted colonization. *Journal of Applied Ecology 50*: 1330–1340.

Population modelling to inform hihi ecology and life history:

Low, M.; Pärt, T.; Forslund, P. 2007: Age-specific variation in reproduction is largely explained by the timing of territory establishment in the New Zealand stitchbird *Notiomystis cincta*. *Journal of Animal Ecology 76*: 459–470.

Ewen, J.G.; Thorogood, R.; Armstrong, D.P. 2011: Demographic consequences of adult sex ratio in a reintroduced hihi population. *Journal of Animal Ecology 80*: 448–455.

Photo: Derek Tearne.

Hihi Template Record Sheet 6: Population survey

This is an example record sheet that can be used during population surveys. This version includes example data, to illustrate how it should be used. Blank versions for photocopying are included at the end of this handbook. Note it is also possible now to enter data into the online hihi database, which can be used in place of, or in combination with, this datasheet.

Cohort	Band number	Band combination	Sex	Date							Total
				20/9	21/9	22/9	23/9	24/9	25/9	26/9	
J04/05	71079	BK BK-DB MT	M	1			1	1			1
J04/05	71097	YE MT-YE BK	M		1					1	1
J04/05	71146	OR BK-YE MT	M								0
J04/05	71159	WH BK-YE MT	F				1	1	1		1
J04/05	71169	BK MT-YE BK	F	1			1		1	1	1
J06/07	77627	RD YE-LG MT	M		1			1			1
J06/07	77628	RD DG-LG MT	F			1				1	1
J06/07	79246	LG MT-RD BK	F	1					1		1
J06/07	79274	LG MT-BK RD	M		1			1			1
J06/07	79279	LG MT-BK DG	M								0
J07/08	83255	RD OR-PI MT	M	1		1					1
J07/08	83261	DB RD-PI MT	M		1				1		1
J07/08	83262	DB DB-PI MT	F				1			1	1
J07/08	83266	DB DG-PI MT	F					1			1
J07/08	83267	DB WH-PI MT	F								0
J08/09	86568	PP MT-PP OR	M								0
J08/09	86576	RD DB-PP MT	M				1	1			1
J09/10	75680	WH LG-PP MT	M	1	1						1
J09/10	75681	YE PI-LG MT	F			1	1				1

Instructions:

- The most efficient approach is to record this information directly onto an electronic spreadsheet, from the observations noted in your field notebook.
- Create a spreadsheet with the above headings (changing dates as appropriate) and include all birds that have been seen in the two most recent surveys, plus all banded juveniles from the most recent breeding season. Include sex where it is known; usually sex is known for all but first year birds.
- Under each survey day, record a '1' if an individual was seen. Check that the sex recorded during the survey matches the sex from your records. If there is a discrepancy, add a note and ideally confirm this sighting.
- If a bird seen during the survey is not in your spreadsheet, check the band combination against older records. Depending on how recently a bird with this combination was last seen, you will need to make a judgement as to whether the sighting is incorrect or the bird simply hasn't been seen for consecutive surveys.
- When all survey days have been completed, record a '1' in the total column if a bird has been sighted on at least one day, or a '0' otherwise. This is the standard way of recording whether a bird has been seen or not during the course of a survey. You may create an additional column that calculates the number of separate survey days an individual was sighted. This may be helpful for judging whether an observation of a questionable combination is reliable.

9. Translocating hihi

Conservation translocations – the movement of organisms from one place to another (IUCN 2013) – have been an essential tool for many decades in New Zealand conservation management (Parker 2013). Translocation is critical for meeting the first fundamental objective of the Hihi Recovery Group (see Chapter One), that of maximising the number of hihi and the number of hihi populations. So far, hihi translocations have met with limited success. However, this is likely related to conditions at the release sites rather than the actual process of catching, handling, holding and releasing hihi during translocations. Hihi translocation protocols have been developed over several decades and hihi survival up to the point of release is typically 100%. In the unlikely event that deaths occur at any stage during a translocation, then advice on protocols to follow are detailed in Chapter 6, including contact details for a range of wildlife health specialists. It should also be noted that a wildlife specialist should be contacted and made aware that a hihi translocation is taking place and should be available to make recommendations following receipt of health screening results (see below).

IMPORTANT: Translocations can only be carried out following consultation with, and permits from, the Department of Conservation, iwi at both the source and receiving sites and all other interested parties (such as community groups and landowners).

9.1 Planning hihi translocations

Ideally, translocation planning should start 12 months before the translocation is due to take place. This allows time to work through the DOC approval process, to assemble a suitably skilled team (see below), book transport (e.g. ferries, charter boats, helicopter or commercial flights), order invertebrates, order health screening supplies and screening via a suitable laboratory and book accommodation (e.g. the Tiritiri Matangi or Te Hauturu-o-Toi / Little Barrier Island Bunkhouses).

9.2 The translocation team

Translocation teams should be led by one person who will take responsibility for assembling the team, organising all necessary equipment, supplies, accommodation, transport and logistics. While the team leader might delegate some of the required tasks it is essential that one person keeps track of the entire process.

A typical hihi translocation team will usually consist of 12 people. This includes one expert processor, who receives all captured birds for weighing, measuring, banding, health checks and aviary assignation, one captive husbandry expert whose sole responsibility is maintaining captive birds, three catching teams of three people each (at least one and, ideally, two of whom must be experts in catching and handling small passerines) and, finally, one cook whose primary responsibility will be keeping the team well fed. If additional procedures are carried out on hihi during translocation, for example applying transmitters, then extra experienced people will be required to fulfil these tasks.

IMPORTANT: Trained and experienced people are essential for successful translocations. Translocations should not be undertaken by under-skilled or poorly organised teams.

9.3 Aviary preparation

The first task following the translocation team's arrival at the hihi source site is preparation of holding aviaries. Holding aviaries should always be rectangular in shape as they are better for bird welfare and easier to work with. On Tiritiri Matangi Island, the aviary consists of three adjacent flights, each approximately 5 m × 3 m. On Te Hauturu-o-Toi / Little Barrier Island there is a single flighted aviary of a similar size. At times, small portable aviaries (approximately 2.5 m × 1.5 m) have also been used. However, hihi seem more at ease in larger aviaries and, where possible, they should be used. Tent aviaries have been used for some species in New Zealand but they are not recommended for hihi, as the configuration, light and movement inherent in the design of many tents renders them unsuitable.

All aviaries should have been cleaned prior to use for hihi by removing all vegetation and leaf litter, scrubbing all faecal matter and food waste from the aviary structure and spraying all surfaces with an appropriate disinfectant (e.g. SteriGENE®). This is normally carried out immediately after any preceding translocation and should not be necessary at the start of a hihi translocation.

Hihi holding aviaries should be lined on their interior with shade cloth. This shields the birds from disturbance outside the aviary and minimises collision injuries during the holding period. Prior to adding vegetation to the aviary the floor, sides and roof of the aviary should be very carefully checked. The primary need is to ensure there are no small holes in the shade cloth, or on the ground, that hihi might get caught in or be able to force their way through. Check also for sharp surfaces, such as protruding nails or staples, which might injure hihi. Finally, doors should be checked to ensure they can be easily operated when birds are in the aviary.

Following these checks the aviary floors can be covered with a dense layer of leaf litter (approximately 200 mm thick – it will compress over the holding period). Take care in collecting leaf litter to ensure that it has not been previously used in an aviary and that it is not taken from directly under obvious bird roosts (for example, starling roosts on Tiritiri Matangi). Ideally, it should also be damp. If it is very dry it should be lightly sprayed with water. This is to settle any dust which might carry fungal spores such as those of *Aspergillus fumigatus*. The aviaries can then be furnished with vegetation. The objective in furnishing the aviary is to provide dense cover from floor to ceiling along each wall thereby providing multiple hiding, roosting and foraging sites for the captive birds (Fig. 9 1), but allowing some space in the middle of the aviary for limited flight. A range of vegetation is used, including large structural branches such as 2–3 m lengths of kānuka (*Kunzea ericoides*), māhoe (*Melicytus ramiflorus*), karo (*Pittosporum crassifolium*) and coprosma (*Coprosma* spp.) along with smaller branches and fern fronds to fill the gaps. Ideally, the vegetation will also have fruit or flowers on it. Vegetation can be easily secured by attaching hooks and eyelets to the aviary frame and using lengths of rope tied with slip knots to hold it in place. If the aviary frame is not robust enough to attach hooks and eyelets to, light posts or waratahs (taking care to cover any sharp edges) can be driven into the ground and used as attachment points. Ideally, vegetation on the side of a 5 m-long aviary would be secured with four 2.5 m lengths of rope, two approximately 0.5 m above the ground and two at 1.5 m above the ground, each secured with a slip knot at the end closest to the aviary door. When the vegetation is removed from the aviary it is a simple matter of releasing the slip knots and removing the vegetation in two quick stages.

Figure 9.1. A furnished hihi aviary on Tiritiri Matangi. Note the thick vegetation, the two feeding stations, sugar water feeder with adjacent perches, leaf litter and two water baths. *Photo: Luis Lachica.*

When the aviaries are fully furnished, two feeding stations should be established, one at each end of the aviary, not too close to exposed walls where wind or rain might tip or fill dishes. The feeding stations usually consist of a timber platform approximately 800 mm × 100 mm × 20 mm, and a slightly smaller removable cafeteria platform that can be placed on top of this (see below). Light rope is attached to four fencing staples, one on each corner of the main feeding platform (larger piece of wood). The platform is then hung from the roof of the aviary along with stabilising ropes to ensure that it is solid (i.e. it should not swing or move when a bird lands on it). It should not be immediately underneath any obvious perches but it should be adjacent to vegetation so that birds can easily approach it. Consideration needs to be given to the captive husbandry person when establishing the feed station, i.e. the stabilising ropes and vegetation should not hinder their approach and it should not be so high that changing food will be difficult. The separate cafeteria platform, approximately 600 mm × 100 mm × 10 mm, with nails driven into it to hold food dishes in place, will be placed atop the first platform. This secondary platform will be removed and replaced with a clean cafeteria at each feed out (see below). On Tiritiri Matangi, one feeding station will usually be established perpendicular to the aviary walls at one end of the aviary while the station nearest to the aviary door will usually be parallel with one wall. Finally, at least two and up to four water dishes should be placed on the aviary floor, with consideration again being given to placement (e.g. not directly under the feeding stations). Hihi are extremely enthusiastic bathers and the dishes should be at least 400 mm wide and up to 50 mm deep. Large plastic planter pot bases with the ability to hold water up to 40 mm deep are ideal. If the dishes are quite smooth, one or two large or several small clean rocks can be added to provide stable landing places for the birds.

IMPORTANT: Aviaries must be fully furnished with all vegetation, food and water in place before hihi catching commences.

9.4 Catching and processing hihi

Before hihi capture commences, a processing station should be established. This will be a central location, usually adjacent to the holding aviaries, where all hihi will be taken for weighing, measuring, banding, health checks and aviary assignation. The primary goal for this area is that it should be dry, free from draughts and have good light (a head lamp can be very useful for the processor if light is limited). There should be a secure area for hanging birds in bags, such as a line of hooks, and there should be room for the processor, and an assistant if required, to comfortably process the birds. Individual preferences vary, with some processers preferring to sit on a bench while others prefer to stand at a table, but all recording, measuring, banding and sampling equipment should be within easy reach so that birds can be processed in an efficient and effective manner. Obviously, all team members must be aware of where the processing station is, the best approach to take when arriving with birds to minimise disturbance to holding aviaries, and that they must be very quiet when in the area (Fig. 9.1).

The different methods used for catching hihi are described in Chapter 5. Each team will be given a supply of nets, poles, ropes and black cotton holding bags, although a good supply of catch bags should always be maintained at the processing station, as catch success varies and some bags inevitably get wet, dirty or lost. Catching typically starts at first light, although care must be taken not to start too early to avoid catching bats at sites such as Te Hauturu-o-Toi / Little Barrier Island. Catching ceases each day at a time that allows the processer to finish all birds and get them into the aviaries with at least 30 minutes and, ideally, an hour of daylight left so that they can drink, feed and find a place to roost for the evening.

Each of the catch teams will generally go to a separate area from the others and will need to have a good idea of each team's movements throughout the day so that they do not attempt

to catch birds in areas that have already been worked. The person most experienced in hihi capture in each team will be the team leader while the other two people will act as assistants and runners with captured hihi. When hihi are caught they are placed in a black cotton bag that must be securely tied with a knot which must then be easily able to be undone by the processer. A cardboard tag will have been stapled to each bag on which capture time, location, sex, age and band details can be written, thereby allowing processing to be prioritised (ensure that the staple does not protrude into the bag in a way that could harm captured birds). Hihi should be taken to the processing station as soon as possible following capture (<30 min). Ideally, the team runner will then carry them (walking rather than running!) to the processing station, taking care to be quiet and to keep them cool. Vehicles can be used for transport but quad bikes and other all-terrain vehicles should be avoided as they are very noisy and have hard suspension.

Upon reaching the processing station the birds should be weighed, measured, banded, undergo health checks and any other required procedures as described in Chapter 5. All details should be recorded on a data sheet (see **Hihi Template Record Sheet 7**), including the capture time, location and which aviary the bird goes into. Where multiple flights are being used it is useful to have a sheet of paper stapled to each aviary door upon which each bird in the aviary is recorded. Male and female hihi can be held in the same aviary but, as much as possible, a relatively even sex ratio should be maintained in each flight. Up to 20 hihi have been successfully held in each flight on Tiritiri Matangi. The flight furthest from the entrance to the aviary alcove is filled first, followed by the middle flight and then the flight closest to the alcove door. Several bird species, including hihi, sometimes take longer to settle in the flight closest to the alcove door. It is not clear why this is so but this aviary should be always be used last and if ≤40 hihi are being translocated this flight should not be used.

Tiritiri Matangi hihi settle into aviaries quite quickly, likely because of a combination of familiarity with humans and that juvenile birds are typically targeted for translocation. In contrast, hihi from Te Hauturu-o-Toi / Little Barrier Island behave in a very different manner. They are much more timid and will typically hide in dense vegetation whenever the aviary is approached. They also have a tendency to force themselves into holes and crevices when stressed, something not seen on Tiritiri Matangi. Therefore, everybody working on Te Hauturu-o-Toi / Little Barrier Island must be aware of these behavioural differences and be extra careful with foot placement when in aviaries and all other potential disturbance factors. Minimising stress on hihi during translocations is addressed in Box 9.1.

9.5 Captive husbandry

Hihi eat a variety of foods while in captivity. Many of the foods require preparation prior to feeding out and are prone to spoilage, especially in hot weather. Therefore, careful food storage, cleaning of food dishes, general hygiene and regular replacement of foods are critical during the captive husbandry stage. Ideally, the captive husbandry person will have a dedicated area with running water and refrigeration for preparation and storage of food and for cleaning and sterilising all preparation tools and food dishes. The room adjacent to the hihi kitchen on Tiritiri Matangi can be used for this purpose, as can the old kākāpō room on Te Hauturu-o-Toi / Little Barrier Island (Fig. 9.2).

Figure 9.2. The Kākāpō shed on Te Hauturu-o-Toi / Little Barrier Island set up as a translocation kitchen. P*hoto: Scott Morrison*.

The required foods, recipes and quantities required for each flight are described in **Hihi Best Practice Sheets 14** and **15**. Many of the

Box 9.1 Minimising stress through translocation routine and predictability

Translocation is an inherently stressful process for hihi. They are exposed to many unfamiliar stimuli (noise, vibration, light), confined in a small space with a large number of conspecifics, given unfamiliar foods and may undergo invasive procedures such as blood sampling, cloacal swabbing and transmitter attachment. When faced with these novel situations hihi undergo an **acute stress response** as a means to enhance immediate survival. This is characterised by a fast-acting cardiovascular response (fight or flight) followed by a slower release of glucocorticoid hormones (corticosterone in hihi) (Dickens et al. 2009; Parker et al. 2012). The cardiovascular response increases heart rate, vigilance and energy mobilisation while the glucocorticoid response inhibits reproduction, increases foraging behaviour and suppresses the immune system (Parker et al. 2012). If captive hihi are exposed to multiple, consecutive and/or continuous stressors they may mount multiple or sustained acute stress responses and move to a state of **chronic stress**. Chronic stress can suppress the immune and reproductive systems, alter metabolism, increase susceptibility to predation by decreasing the fight or flight response and increase dispersal behaviour (Parker et al. 2012). All of these factors can decrease individual survival and overall translocation success (Dickens et al. 2009, 2010; Parker et al. 2012). Therefore, the number of stressors must be carefully managed throughout the translocation. The primary means of doing this is by having a highly experienced team. The second is by maximising the perception of control captive hihi have over their immediate environment, minimising unpredictability in captivity and minimising novelty (Dickens et al. 2010; Parker et al. 2012). A sense of control is provided by furnishing aviaries with lots of natural cover and providing a wide variety of ad lib food and water. Unpredictability is minimised by feeding birds on a regular predictable schedule, ideally by the same person. This predictability can be further enhanced by wearing the same clothing for each feed out. Novelty can be reduced by very carefully examining all aspects of the translocation process and removing any unnecessary handling, conducting all processes in one step (e.g. banding, bleeding, swabbing, transmitter attachment), minimising enclosure changes and carefully managing the transportation phase.

foods can be prepared ahead of time and either frozen or refrigerated until required. Two feed outs a day are generally recommended but extra checks should be made on the first day or two to ensure that sufficient quantities are being provided, especially in hot weather when liquid foods might spoil. Food is presented on a cafeteria board that has nails driven into it to prevent food dishes being tipped over. The boards are replaced and cleaned between feeds. A variety of food dishes have been used, including small plastic dishes and fish tins. Fish tins (185 g or larger) are ideal as they are stable, easily cleaned and hold a sufficient quantity of food. However, they must be properly cleaned and checked prior to use to ensure there are no sharp edges. Food dishes are replaced at each feed out so a large quantity is required. For example, if all three flights are in use on Tiritiri Matangi a minimum of 60 dishes are required for the aviaries so that one set can be cleaned while the other is in use.

When purchasing fruit for hihi try to obtain small fruit that is on the verge of ripeness. It is a difficult balance to achieve because while the fruit needs to last the duration of the translocation (6–10 days) it also needs to be soft and ripe, especially pears and apples. If pears and apples are hard and unripe they can be briefly boiled in a light sugar syrup to soften them up prior to feeding out. Oranges and ripe pears are consistently preferred over apples. Other fruits have been offered at times including bananas, grapes and kiwifruit. These fruits are often expensive, difficult to store and spoil quite quickly. However, as long as they are properly cleaned they can be used but should be considered additional to oranges, pears and apples. Additional natural vegetation, leaf litter and natural fruit might also be provided throughout the holding phase,

particularly during warm weather when cut vegetation quickly wilts and dries. Waxmoth larvae (*Galleria mellonella*) are a very important source of easy energy for captive hihi – and they love them! They are expensive, but well worth the cost. Ensure that supplies are ordered well before the translocation (3–4 months) and plan on 10–20 larvae per bird per day of captivity (order waxmoth larvae from www.biosuppliers.nz).

9.6 Transfer day

There should be at least one full day between capture of the last hihi and transfer to the release site. This is because the last birds for translocation are typically caught late in the day. If they are then recaptured for transfer the following morning it is unlikely that they will have had much time to feed in the aviary and their energy reserves might be very low during transfer and release. On Tiritiri Matangi the translocation team will normally arrive on Sunday, set up the aviary and then start catching on either Sunday afternoon or Monday morning. The full quota of birds is usually captured by Wednesday morning, at which time any health screening samples are sent to the laboratory for analysis. Initial results are usually received late on Friday afternoon or early Saturday morning, with transfer to the release site taking place on Saturday morning. Translocations from Te Hauturu-o-Toi / Little Barrier Island require a larger time window to allow for weather and greater variation in catching rates and a more flexible approach is required for final transfer, while still allowing for at least one full day between capturing the final bird and transfer.

Transfer day is especially stressful for hihi and the translocation team. Therefore, it should be very carefully planned and organised. The last feed out is conducted on the afternoon prior to transfer day, with extra food provided if necessary. On the morning of transfer the captive husbandry person will do a quick check of the aviaries but will only feed out waxmoth larvae and top up any dishes if necessary. This allows the maximum feeding time for the birds prior to aviary capture with birds given at least 2 hours of uninterrupted daylight for feeding before being captured in the aviaries.

Wooden transfer boxes with one wall covered with mesh and tight shade cloth, two perches and two doors, one small one for placing birds in the box and one large one for releasing the birds, are recommended for hihi translocations. Transfer boxes are prepared on the morning of transfer. They are lined with newspaper or corrugated cardboard with two staples at the door end of the box to hold it in place. Two or three soft branches of vegetation, such as kānuka (not mānuka (*Leptospermum scoparium*)) are placed under the two perches with the soft tips pointing to the ceiling of the box at the rear. This provides cover in the box for any particularly timid bird. Small branches with natural fruit, such as māhoe or māpou (*Myrsine australis*), can also be added to transfer boxes, as long as they are consistent with biosecurity protocols at the release site. Two to four orange halves are wired into the box to provide hydration and energy throughout the transfer phase. These are attached adjacent to each perch, with particular care taken to ensure that the wire ends cannot harm birds or people carrying the box (Fig. 9.3). Finally, a piece of masking tape is attached to the top of the box so that band combinations can be recorded on the transfer box as birds are processed.

Figure 9.3 Hihi translocation box showing wired-in oranges and food tray with liquid foods and water in the base of the tray. Slightly more vegetation can be added than is apparent in this photograph. *Photo: Jo McCarthy.*

Additional food and water can be provided in the boxes, particularly if there is a delay between the aviary catch and transport, in response to weather (e.g. late summer translocations when it is quite hot), for long trips (>4 hours) or if birds are being held

overnight. If food is placed in the box prior to aviary capture you must ensure that dishes do not block the door but are close enough to it to be easily removed. Regardless of whether it is actually provided, prepared hihi food, dishes and water should be carried during all transfers in case of unexpected delays in transit. **However, all liquid foods should be removed from the boxes during transit** to prevent them tipping and soiling birds. When food is provided it is easiest to use small round pot plant trays approximately 150 mm in diameter and 25 mm deep. These securely hold three 185 g fish tins in which liquid foods can be provided with additional water placed around the tins in the tray itself. The tray can then be put in or removed from the transfer box in a single movement. Hihi will readily use water dishes (e.g. the same trays used to hold the food dishes) for bathing and drinking during transit but these should be provided separately to the food trays as the hihi will throw water all through the transfer box. Water dishes should only be provided if it is quite warm or hot with particular care taken to avoid cold draughts post bathing. This is to prevent birds from chilling after bathing. A secure location, i.e. a room where birds could be easily caught if they escape, must be available whenever transfer boxes are opened as it is relatively easy for hihi to escape.

Prior to capturing birds in the aviary, a set of gear is prepared for each flight. This consists of black bird bags, scales for weighing the birds, a data sheet listing all of the birds in the aviary (including capture weight, health screening results and bands), a marker pen for recording band combinations on transfer boxes and a Philips screwdriver for securing each transfer box as it is filled. The capture team is then assembled and roles throughout the process are assigned so that everybody knows beforehand exactly what they will be doing. When people are in the aviary, particular emphasis must be placed on team safety when removing vegetation (i.e. being careful not to scratch or impale anybody) and bird safety (i.e. careful movements, foot placement and being as quiet as possible).

Two people then enter the first flight, remove food and water dishes and then begin removing vegetation, working their way down the aviary. A pile of vegetation is first made at the aviary door. The door is then opened, with one person pushing the vegetation out into the aviary alcove to people outside the aviary. The second person in the aviary watches the birds and guards the door as vegetation is removed. The door is then secured before removing vegetation from the alcove. This process is continued until most of the vegetation is removed, except for one or two branches left in each corner of the aviary for the birds to sit in. At this point additional assistants enter the aviary. The branches are laid down on the ground and two people capture birds with hand nets, pass them back to 2–3 people waiting with open bird bags, who then pass them on to another 2–3 other people who will secure the bags and hang them on pre-prepared hooks. This process happens very quickly, with vegetation clearance and bird capture each taking 5–10 minutes, so it is essential that all participants know exactly what their role is. When all birds are captured, the number of bird bags is carefully counted to make sure it equals the number of birds held in the aviary. Three to four people will then process the birds by weighing each one, checking its band number and colour combination, checking its weight against initial capture weight and then placing it in a transfer box and recording its band combination on the masking tape on the top of the transfer box. When the box is full (normally 5 birds per transfer box), it will be secured with a screw driver and placed in a shady quiet spot in the aviary with the mesh side of the box facing away from all activity. If there are multiple flights holding birds it is faster if one team is left processing birds while the other moves to the next aviary. The most skilled handlers will usually capture birds in the aviary and process the last flight, as they are often faster than other handlers.

9.7 Transportation

The primary objective when transporting birds is to minimise noise, vibrations and fluctuating temperatures. This can be achieved by padding boxes with closed cell foam or packing blankets, ensuring adequate ventilation between boxes, timing transport outside of the hottest part of the day, or when traffic congestion is anticipated, and planning smooth transitions from one form of transport to another (e.g. from ferry to car to airport) to minimise delays. For big moves, i.e. those taking >4 hours, we recommend catching and boxing birds late in the day and then moving them overnight. Vehicles used for overnight moves should be well insulated, have air conditioning and careful drivers. All transfer boxes should be carefully placed to minimise noise and vibration and windows should be covered to prevent outside light disturbing the birds. By carefully moving birds this way they do not miss any feeding opportunities and are generally calm and quiet with most sleeping throughout the trip, including during loading and unloading. It is critical that transfer plans are discussed with all transportation providers prior to the transfer day. This is especially important if commercial flights are used where birds might be loaded into a pressurised and temperature-controlled hold or the main cabin.

IMPORTANT: The transfer phase is when birds are at their most vulnerable. Therefore, they **must** be accompanied by an experienced person through to their final release and **must not** be left unattended at any time. Transfer boxes heat up very quickly if in direct sun, cool very quickly if in direct draughts and should not be exposed to unnecessary noise and vibrations. Commercial airline flights require extra planning. Airline staff can be very accommodating in allowing an experienced person to supervise loading, but they must be informed well ahead of time so that appropriate security protocols can be met.

9.8 Release

If hihi are being held overnight at the release site they should be held in a quiet, secure room that is neither hot nor cold and draughty. They should be fed and provided with water as soon as they arrive. If they have arrived after dark they should be unloaded in darkness and complete silence (ensure that any security lights have been turned off prior to unloading). Handlers can use red headlamps or torches so they can see what they are doing without disturbing the birds. If this process is done carefully the birds will not stir. They should then be left to awaken naturally at first light after which they should be given fresh food within 30 minutes of waking. Remember to remove food dishes prior to carrying the birds to the final release site.

The release site should have been selected prior to release day and must meet the needs of the birds over the desires of the release ceremony organisers – **ceremony is important, but not at the expense of bird welfare**. Therefore, the birds should be released as soon as possible with speakers and any other protocols working around their welfare. Ideally, birds should be released by 1.00 pm if released on the day of transfer and by 10.00 am if they have been held overnight, thus allowing them 1–2 hours to feed in the transfer boxes.

All spectators should be advised to keep as quiet as possible until release. If boxes are hand carried to the release site the people carrying the boxes need to be advised not to knock the boxes against their legs as they walk and to keep the mesh side of the box facing away from their legs. Birds should be released as a group, with all spectators and photographers either directly behind the transfer boxes or off to one side. This provides an obvious, unimpeded escape route for all birds, ideally directly into suitable vegetation or branches on which they might sit. Sugar water feeders should be set up either at or close to the release site so that the birds can immediately access a ready energy supply.

9.9 Q & A: Why do we do it this way?

For a general description of the reason for this Q & A section, see Chapter 1. The following set of questions and answers capture some of the issues related to translocating hihi:

Q. Why does each aviary have two feeding stations, especially as the birds always seem to favour one over the other?

A. Most of the captive hihi will use the same feeding station. However, within each group of hihi there are usually a few shy birds that may have trouble accessing busy feeding stations, especially if they have to compete with more dominant individuals. By having two stations the opportunities to feed are distributed more widely throughout the aviary, maximising the possibility that all captive birds get access to food.

Q. The birds seem to favour some foods, some are barely touched and they rarely eat all that is given. Why are amounts not reduced and a smaller variety supplied?

A. It is difficult to predict which foods will be favoured during each translocation and it often shifts between translocations. By providing a variety of foods a range of preferences can be catered to and energy uptake maximised. This is especially important for particularly fussy birds which might focus on one food type that other birds have rejected. All food dishes are filled to the brim so that the birds can easily see and access them. If there is small layer in the bottom of each dish they might be less-inclined to investigate the food. There should always be food left over between feeds (i.e. all hihi must have ad lib access throughout the holding period). If all of the food is gone, the amount was either just right or insufficient – and there is no way to tell which it was. A little bit of wastage is entirely acceptable for maintaining high welfare standards.

Q. Why is it important to have an experienced translocation supervisor and to accompany birds throughout the entire process from initial capture through to release?

A. As soon as we capture birds we take all of their options away from them, e.g. their ability to feed on what they want, when they want it, their ability to move to a warmer or colder location to maintain their own thermoregulation and their ability to choose whom to associate with. By having an experienced translocation supervisor all potential problems can be anticipated and planned for. The overview provided by this person has to be in place throughout the entire process, because things can go wrong very quickly. For example, if a transfer box is inadvertently left in direct sunlight it can heat up extremely quickly (< 20 minutes), potentially killing all of the occupants.

9.10 References and suggested further reading

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Hihi Best Practice Sheet 14: Captive diet recipes for translocations

Wombaroo™ lorikeet and honeyeater food:
Dissolve ½ a cup of Wombaroo™ in ½ a cup of warm water.
Mix until dissolved and add cold water to make 500 ml mix.

Jam and honey water:
Mix a ¼ cup of berry jam and a ¼ cup of honey with hot water until dissolved.
Add ½ a tablespoon of plain 'Pronutro' cereal and mix until dissolved.
Add cold water to make 500 ml of mix.

Pureed fruit and vegetable mix:
Blend the following ripe fruit and vegetable ingredients to make a runny puree:
½ a carrot, ⅓ of an apple, ⅓ of a pear, ¼ of an orange, ⅓ of a banana, 10 grapes, 500 g of corn kernels and 50 g of peas.
This mix can be divided into daily portions using ice cube trays and stored in the freezer until required. It should be very liquid when fed out so add sugar water or jam and honey water to the puree until it is a very sloppy constituency.

Sugar water:
Dissolve 1 cup of raw sugar in 1 cup of warm water. Add cold water to make up to 1 litre.

Fruit cake:
Cut the cake into small portions and pour jam and honey water or sugar water over the top of it before feeding out.

Fruit cake recipe (Lovegrove & Veitch 1994)
Cream 200 g of butter and 1 cup of sugar
Add 3 eggs and beat
Add 2½ cups of flour
Add 2½ teaspoons baking powder
Add 1 cup of pre-soaked sultanas
Stir well and spread into a greased baking dish or patty pans. Bake for c. 30 minutes in a moderate (180°C) oven. The fruit cake can be baked ahead of time and frozen for convenience.

Hihi Best Practice Sheet 15: Hihi translocation feeding protocol

1. Morning feed at approximately 0930.
2. Afternoon feed at approximately 1530.
3. Prepare all the food before each feed. All liquid foods can be mixed in large jugs and refrigerated until required (recipes in Hihi Best Practice Sheet 13). Fruit should not be cut until just prior to feed outs.
4. See the table below for the recommended food for each flight.
5. Sugar feeders can be replaced once a day (unless they are emptied, in which case they should be replenished at each feed out).
6. All other liquid food, fresh fruit and water baths should be replaced twice a day.
7. Fill the food dishes almost to the brim. It is sometimes easier to take a jug up to the aviaries and fill the dishes after they are secured on the feeding platforms.
8. Thoroughly wash and quarter all the fruit.
9. Place all food for feed outs into buckets and carry a water bottle for the water baths. Extra buckets might be required for the dirty food dishes and old fruit.
10. Keep time and people in the aviaries to an absolute minimum – each feed out should only require one person to enter and exit an aviary once.
11. Enter the aviary and place all of the dirty food dishes, food and fruit into the buckets.
12. Place fresh food into the cafeterias and distribute fresh fruit evenly throughout the aviaries by securing it onto vegetation by spiking it onto branches – do not leave fruit sitting on the feeding platforms as it will get dirty or be knocked to the ground.
13. Hang the sugar feeders. Ensure that the birds can access the sugar feeders (i.e. there are perches that they can use whilst feeding from the sugar feeders).
14. Arrange any fresh fruit or flowers into secure positions that are accessible to the birds.
15. Clean the water baths and replace the water.
16. Quickly scan the aviary floor and the birds themselves, checking for any birds that are fluffed up, immobile or have an unkempt appearance – please note the bands of these birds.
17. Scatter waxmoth larvae into the vegetation in the aviary just before exiting. Try to distribute the larvae as evenly as possible throughout the aviary to prevent dominant birds from monopolising them.
18. Clean all of the dirty dishes in hot soapy water and then submerge in a Milton or SteriGENE® solution as described in Chapter 4.
19. Do not enter aviaries between feed outs unless it is absolutely necessary and keep all other disturbance to a minimum. Many people will want to look at the hihi in the aviaries, but this disturbance is unnecessary and only unsettles the birds. If people are particularly interested in seeing the birds in the aviaries then get them to help you with the feed outs – and all the dishes afterwards.

Food requirements for each flight and feed out:

LOCATION/AVIARY	FOOD
Each flight	• Wombaroo x 2 (1 in each cafeteria) • Jam mix x 2 (1 in each cafeteria) • Vegetable puree x 2 (1 in each cafeteria) • Fruit cake with either jam mix or honey water poured over the top x 2 (1 in each cafeteria) • Sugar water x 2 (1 in each feeder) • Sugar feeders x 2 (replace once a day) • Water dishes for drinking and bathing x 2 • 1–2 apples (washed, quartered and spiked on branches) • 1–2 pears (washed, quartered and spiked on branches) • 1–2 oranges (washed, quartered and spiked on branches) • 10-20 waxmoth larvae per bird, removed from their cases, and cast throughout vegetation in the aviary so birds can glean them.

Hihi Template Record Sheet 7: Translocation data sheet

This is an example record sheet for recording the banding, measurement and bleeding of hihi during translocation. Blank versions for photocopying are included at the end of this handbook.

Date	Capture time	Process time	Into aviary	Location	Metal number	Left leg	Right leg	Bird & bag (g)	Bag (g)	Bird (g)	Sex	Blood	Swab	Aviary	Comments
27/3	741	801	807	WV	9239	LGLG	WHMT	62	30	32	F	Y	Y	1	Great condition
27/3	746	809	814	B22	9245	ORRD	PIMT	70	30.5	39.5	M	Y	Y	1	
27/3	757	823	829	B22	9200	YEPP	PPMT	61	31	31	F	Y	Y	1	Check band combination

Hihi Template Record Sheet 8: Aviary catch transfer day data sheet

This is an example record sheet for recording the weight and band combinations of captive hihi on transfer day. It includes health screening results (WBC: White Blood Cell Counts) and transmitter attachment (Tx). Blank versions for photocopying are included at the end of this handbook.

Metal	Left	Right	Sex	Catch weight (g)	Aviary weight (g)	Aviary number	Comments
9239	LGLG	WHMT	F	32	29	1	
9256	WHMT	RDLB	F	32	30	1	
9259	LBDB	WHMT	M	35	32.5	1	
9261	PILG	WHMT	M	40	41	1	
9289	LBLB	WHMT	M	37	36	1	
9300	WHMT	PPLB	F	31	30	1	Right leg actually PPLG
11805	YELB	WHMT	M	38	37	1	
11822	BKPP	WHMT	M	37	42	1	
11853	LBRD	PIMT	M	39	34	1	Lost 5 g – check
11869	PIMT	YEPP	F	29	28.5	1	
11882	PIMT	LBLB	F	28	27.5	1	
11883	RDPI	PPMT	M	34	35	1	
11884	PIMT	DBPP	F	27	25	1	
11886	PIMT	LBDG	F	35	34	1	

Processing Team

Catching and vegetation removal: Kevin, John
Bagging: Mhairi, Fiona, Chris
Boxing: Morag, Vix, Caitlin

Use the space below for calculating bird weights. Bags can be pre-weighed and labelled to save time during processing.

10. Blank forms

Hihi sugar water consumption record sheet								
Site:			Year:					
Feeding Station:			Feeding Station:			Feeding Station:		
Date	Volume remaining (L)	Volume put out (L)	Date	Volume remaining (L)	Volume put out (L)	Date	Volume remaining (L)	Volume put out (L)

Hihi nestling banding and measurement record sheet

Combo	C-band	Nest	Date	Age	Weight (g)	Tarsus (notch)		Tarsus (full)		Head-to-bill		Blood number	Slide
						1	2	1	2	1	2		

Hihi unhatched eggs and dead chicks record sheet

Sample Number	Nest ID	Date of death	Date collected	Date of sample	Type	Embryo stage (if egg)	Comments

Hihi nest box monitoring record sheet

Season (year):	Site:
Box number:	Female parent:
Clutch:	Male parent (social):

Date	Day-by-day summary: egg laying, nestling death, mite presence and treatment

Nest summary information

Clutch details		*Unhatched egg sample details*	
No. eggs laid:		Unhatched egg 1: separated/gunk/MDE/LDE/FFC tissue sample id	
Date 1st egg laid:		Unhatched egg 2: separated/gunk/MDE/LDE/FFC tissue sample id	
No. eggs hatched:		Unhatched egg 3: separated/gunk/MDE/LDE/FFC tissue sample id	
No. chicks fledged:		Unhatched egg 4: separated/gunk/MDE/LDE/FFC tissue sample id	
		Unhatched egg 5: separated/gunk/MDE/LDE/FFC tissue sample id	

Fledged chicks summary information

Chick details	Chick 1	Chick 2	Chick 3	Chick 4	Chick 5
Chick band:					
C-band:					
Tarsus (notch) (mm):	/	/	/	/	/
Tarsus (full) (mm):	/	/	/	/	/
Weight (g):					
HB (mm):	/	/		/	/
Blood sample (✓)					

Dead nestling 1 age (days)		Tissue sample id		Dead nestling 2 age (days)		Tissue sample id	
Dead nestling 3 age (days)		Tissue sample id		Dead nestling 4 age (days)		Tissue sample id	
Dead nestling 5 age (days)		Tissue sample id					

Hihi natural nest monitoring record sheet					
Season (year):		Site:			
Nest ID:		Female parent:			
Clutch:		Male parent (social):			
Nest location (description):					
GPS coordinates:					
Date	Day-by-day summary: egg laying, incubation, fledging				
Nest summary information					
Estimated date(s) of egg laying:			Tree species:		
Estimated incubation start date:			Nest height:		
Estimated date of fledging:			Nest accessible?	Yes/No	
Estimated number of chicks fledged:					
Fledged chicks summary information					
Chick details	Chick 1	Chick 2	Chick 3	Chick 4	Chick 5
Chick band:	–	–	–	–	–
C-band:					
Tarsus (notch) (mm):	/	/	/	/	/
Tarsus (full) (mm):	/	/	/	/	/
Weight (g):					
HB (mm):	/	/		/	/
Blood sample (✓)					

Hihi population survey record sheet

Cohort	Band number	Band combination	Sex	Date							Total

Hihi translocation record sheet

Year: **Capture location:** **Release site:**

Date	Capture time	Process time	Into aviary	Location	Metal number	Left leg	Right leg	Bird & bag (g)	Bag (g)	Bird (g)	Sex	Blood	Swab	Aviary	Comments

Hihi aviary catch record sheet			**Location**			**Year**	
Metal	Left	Right	Sex	Catch weight (g)	Aviary weight (g)	Aviary number	Comments

Processing Team

Catching and vegetation removal:

Bagging:

Boxing:

Use the space below for calculating bird weights. Bags can be pre-weighed and labelled to save time during processing.