Field guidelines for burrow-nesting petrel and shearwater translocations

A companion guide to the seabird translocation best practice documents

Helen Gummer, Graeme Taylor and Rose Collen

New Zealand Government

Department of Conservation
Te Papa Atawhai

Disclaimer: This report is instructional and intended to be read alongside the best practice techniques reports for the translocation of Chatham petrels (*Pterodroma axillaris*), Cook's petrels (*P. cookii*), Pycroft's petrels (*P. pycrofti*) and grey-faced petrels (*Pterodroma macroptera gouldi*). It is therefore written in simple, non-formal English.

Cover: Cook's petrel (*P. cookii*). *Photo: Dick Veitch.*

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Abstract

This companion guide provides detailed information relating to the translocation of burrow-nesting petrels and shearwaters. It considers the entire translocation process, from finding, selecting and collecting chicks at the source colony, through to installing artificial burrows, and feeding, measuring and monitoring chicks at the release site. It also includes equipment lists, information on how to clean the equipment and sample data sheets. This guide is written in a simple, instructional style and it is intended that it be read alongside the appropriate best practice document for the species.

Keywords: petrels, shearwaters, burrow-nesting, translocation techniques, New Zealand

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1. Introduction

This companion guide is intended to be read in conjunction with the following documents:

- Best practice techniques for the translocation of Chatham petrels (*Pterodroma axillaris*), Cook's petrels (*P. cookii*) and Pycroft's petrels (*P. pycrofti*) (Gummer et al. 2014a)
- Best practice techniques for the translocation of grey-faced petrels (*Pterodroma macroptera gouldi*) (Gummer et al. 2014b)

It contains additional information that is required for those planning and implementing translocations of burrow nesting petrels and shearwaters; however, it should be noted that this level of detail is not required in the translocation proposal document.

The detailed methods described here are based on techniques that have been tested and refined during many different translocation projects involving a range of species that have been carried out between 1996 and 2012. These methods have been developed to optimise project success, while minimising impacts on ecosystems and maintaining high standards relating to animal welfare. Therefore, they are recommended as current best practice techniques. It is expected that these methods will evolve over time as information and experience from further translocation projects become available.

The methods in each section are set out in such a way that they can be printed off and used as a step-by-step guide in the field by project personnel involved in the translocation. Alternatively, they can be used as a base document and modified to suit a particular location (source colony or release site). General methodologies that can be applied to all burrow-nesting petrel and shearwater species are presented, along with any specific techniques, measurements or protocols that must be used with a particular species or set of species.

2. Equipment lists for seabird translocations

The following equipment lists (with suggested quantities) are intended as a guide only, to assist with the planning of new translocations. These lists should be adapted to suit your particular project and extra columns can be added to make a checklist.

Note: The relevant DOC office may also hold up-to-date species-specific equipment lists for projects that are isolated or ongoing, e.g. Chatham petrel chick translocation guidelines.

2.1 Source colony recce and collection trips

Note: Personnel need to decide whether they will have a pool of tools that is located near to all workers in the team i.e. that will be used by all, or will carry individual tools for tasks such as burrow installation or repair. This will dictate the overall number of tools required. It is also a good idea to tie bright flagging tape to tools, so that they can be easily found in the forest.

2.1.1 General

ITEM	QUANTITY	COMMENTS
Maps (of source colony and burrow locations)	1 of each per person	Maps showing the location of the source colony and burrows in different parts of the source colony.
Mobile phones and/or hand-held radios	1 per person	For communication during chick gathering on the day of transfer and a safety requirement.
First aid kits	1 per person	Personal kits.
Disposable gloves	1 pack	Several pairs per person; useful for handling birds, eating lunch, etc.
Disinfectant hand wash or hygiene wipes	1 pack per team	To clean hands before eating.
Plastic bottles (1–2 L)	2	To hold water for hand washing—soft drink bottles are fine.

2.1.2 Repairing natural burrows or making study-holes

ITEM	QUANTITY	COMMENTS
Plywood boards 300 × 300 mm (> 10 mm thick)	Project and site dependent (150+)	To make study-hole or tunnel covers for petrel burrows; and to repair damaged burrows.
Plywood boards 500 × 500 mm (> 10 mm thick)	Project and site dependent (50+)	These larger boards may be required to repair larger damaged petrel burrows (more of this size would be required at grey-faced petrel source colonies) or even penguin burrows.
Plywood boards (other—200 × 200 mm) (> 10 mm thick)	Project and site dependent	These smaller boards are only useful as small study-hole covers (and must be weighed down with a rock).
Rocks (from the beach or nearby)	200+	To weigh down plywood covers; on some islands where the ground is not fragile and crumbling, large rocks are also sufficient for use as study-hole covers (in which case more would be required).
Plastic boxes (e.g. shoe-box size and larger) that will fit a range of chick sizes (with lids)	1 per person	To put a chick in to keep it warm while repairing its burrow, if required; needs air holes for ventilation and should be a dark colour to keep the chick calm.
Plastic boxes (< 2 L) that will fit a range of chick sizes (with lids)	1 per person	To put a chick in to keep it warm while repairing its burrow, if required; needs two air holes on each side and should be a dark colour to keep the chick calm.
Kitchen towels	Several rolls	Carry a good supply to replace the lining in the box that is used to hold chicks in.
Antibacterial wipes	A few small packets	To wipe out holding boxes if soiled by chicks.

2.1.3 Installing artificial burrows at source colony

Important note: This task should only be carried out if necessary (and approved) to prevent or repair damage to burrows of small to medium species. Specialist advice and approval from the relevant DOC office must be obtained prior to carrying this out.

ITEM	QUANTITY	COMMENTS
Artificial plastic burrows	Project and site dependent	Philproof™ nesting chambers (www.philproof.co.nz) are only suitable for use with small gadfly petrel species—not the large gadfly petrel species such as grey-faced petrels, etc. They have been used at the source colony for Chatham and Pycroft's petrels. Lids must be painted black on the outside, as the current red plastic lid design is translucent and lets light into the burrow, making it unsuitable. (Unfortunately, black lid production ceased prior to 2012, although it would be worth checking this with Philproof.) Any burrows that are seen to have become dug out or upturned on subsequent trips should be removed from the site or stored for future use, i.e. plastic non-degradable materials should not be left in or on the ground without a clear purpose.
Artificial wooden burrows (non-treated)	Project and site dependent	Could be used at sites with very friable soil, where there is a long-term benefit (for several years/translocations) and it is advantageous for boxes to eventually decompose. Any boxes that are seen to have become dug out or upturned on subsequent trips should be removed from the site.
110 mm diameter ridged PVC drainage pipe (for small species only)	1 m for 2–3 burrows	For tunnels, if needing to install an artificial box at a burrow site. These pipes need to be taken to the island pre-cut, and should include a mixture of lengths (min. 300 mm; max. 500 mm) and shapes (straight and curved; see below). Any dislodged pipes seen on subsequent trips must be removed from the site. If it is too difficult to install a 110-mm pipe, you may need to improvise with sections of ply or other wood.
Gas cylinder / stove	1	To gently heat straight sections of pipe over a low flame (turning all the time); the pipe can then be gently bent to the desired curve and plunged into cold water to set.
Fire extinguisher or buckets of water	1	Essential safety equipment if using a stove on the island.
Hacksaw and spare blades	1 or 2	To cut ridged PVC drainage pipe to fit individual burrows.
Durable wire	Several metres	Occasionally two sections of pipe need to be joined, in which case a joiner piece of pipe is wired over the assembly to permanently align the pipe ends.
Wire cutters / blunt-nosed pliers	1 or 2	See above.
Small, sharp spades	1 per person	To cut out holes for artificial burrows.
Narrow hand trowels	1 per person	To dig out trenches for artificial burrows.
Pruning saws	1 per person	To clear tree roots where necessary to install artificial burrows. Can also be used to cut plastic pipe and wood, if required, and to aid with digging into rocky ground.
Secateurs	1 per person	To clear tree roots to access chicks or to install artificial burrows.
Pocket knife	1	May be required to make a second hole in a plastic burrow, if there are known to be two entrances.
Plastic boxes (e.g. shoe-box size and larger) that will fit a range of chick sizes (with lids)	1 per person	To put a chick in to keep it warm while installing an artificial burrow. Needs air holes on each side for ventilation and should be a dark colour to keep the chick calm.
Kitchen towels	Several rolls	Carry a good supply to replace the lining in the box that is used to hold chicks in.
Antibacterial wipes	A few small packets	To wipe out holding boxes if soiled by chicks.
Old tea towels or cloths	5–10	Useful to cover the nest floor of a burrow if needing to install a box. Any soil that falls on it can then be lifted out with the cloth, maintaining the nest bowl and scent beneath.

2.1.4 Marking burrows

ITEM	QUANTITY	COMMENTS
White plastic wands or lengths of No. 8 wire, or other materials for the method advised by DOC	Up to 200	To mark burrows. Need to check with the relevant DOC Office regarding the best marking methods that will last for the duration of the translocation project and whether markers need to be removed at the end of each year and/or the end of the project. Markers must not conflict with other project markers at the source colony.
Plastic triangles or cattle ear tags	Up to 200	Need to check with the relevant DOC Office regarding where to source these from, what colour to use, etc. These can be nailed to trees or cable tied to wands.

ITEM	QUANTITY	COMMENTS
Hammer and nails	1 per person	Required if nailing markers to trees (only if approved by the relevant DOC Office).
Fence cutters	1 per person	To cut No. 8 wire if using this to mark posts.
Permanent tagging pens	1 per person + spares	To write numbers on triangle markers.
Flagging tape (four contrasting colours)	4 rolls of each colour	To mark burrow sites. Can colour code for chicks to transfer, marginal chicks, etc. (supplier: Geosystems; www.geosystems.co.nz).
Permanent ink marker pens	1 per person + spares	To mark flagging tape (fine-tipped).
GPS data logger	1 per team	To record burrow group locations (supplier: Geosystems; www.geosystems.co.nz).
Waterproof paper and pencils	Plenty	To hand-draw individual burrow locations in a burrow area or subgroup.

2.1.5 Processing chicks

ITEM	QUANTITY	COMMENTS
Bum bags	1 per person	To hold chick processing gear and notebooks.
Waterproof notebooks	1 per person	To record data.
Pencils	2+ per person	To record data.
Pencil sharpener	1 per person	To record data.
Rubber bands	Lots	For separating pages in notebooks.
Holding (weigh) bags These are unlikely to be used much on the recce trips as weights are not required; however, they are needed on the selection/collection/transfer trip)	6 per person	Refer to the appropriate best practice document for the species for suitable bag size. Dark-coloured, thick cotton pillowcases can work well (of the same brand/size so that all bags are the same weight) for medium to large species. However, custom-made bags that are all of equal weight and made from dark cotton material (not too thick as it takes longer to dry after washing) are ideal. No cord is required at the top.
Weigh boxes	1 per person or per pair	Dark boxes may be preferred over bags for weighing chicks of large species, e.g. grey-faced petrels, to reduce any risk of regurgitant soiling their plumage, which can be fatal.
Kitchen paper towels	Plenty	To soak up any regurgitant, especially from plumage, if required.
Fine sand	A few handfuls	To soak up any regurgitant, especially from plumage, if required (e.g. grey-faced petrels).
Sterile saline solution (10-mL sachets)	A few per person	Useful for flushing chick wounds or eyes that are damaged during extraction/ handling.
Betadine® antiseptic liquid	1 small bottle	First-aid treatment for open wounds (not recommended for prolonged use; seek veterinary advice).
Banding kit	1 per permitted bander	Should include banding pliers and circlip pliers. Chicks are generally not banded on the recce trip, but a kit should be available for banding adults or particularly large chicks if requested by the Banding Office.
Metal bands	300+	Obtained from the Banding Office. Mostly required on the collection trip.
Pesola scales	1 per person	Refer to the appropriate best practice document for the species for suitable scale weights. Probably not required on the recce trip, but some should be available. Scales are most important for the collection trip.
Bulldog clips	1 per scale set	Useful for attaching to scales (and re-calibrating) when weighing heavy chicks, to prevent bags from slipping out of the scales and falling.
Wing rules	1 per person	Needs a good solid stopped end (offset ‘stops’ can lead to unreliable readings).
Napisan®	500 g	To wash holding bags (note: regurgitant can be highly oily, so an additional washing agent might also be required, e.g. when working with grey-faced petrel chicks).
Bucket	<10 L	To soak/rinse holding bags.
Temporary washing line and clothes pegs	1 + 20 pegs	To dry holding bags.
Sticks (thin)	Hundreds	To erect at burrow entrances as required. Natural sticks can be obtained on site; avoid taking artificial materials into a nature reserve
Data recording sheets	Lots	Refer to section 10.1.

2.1.6 Collecting and transferring chicks

ITEM	QUANTITY	COMMENTS
Chick weighing and measuring equipment		Refer to section 2.1.5
White fluteboard* or cardboard pet boxes and diagonal pre-cut dividers (one per box)	Species dependent	See footnote below* regarding source, cost, specifications, etc. White boxes must have black linings to block out light; alternatively, you could try a neutral colour (so that there is no need to add a lining). Large species (e.g. grey-faced petrels) should be held individually in boxes. Medium-sized species (e.g. Cook's petrels) can be held two birds per box with one diagonal divider (or at most three birds per box for as short a period as possible with appropriate dividers—refer to the appropriate best practice document for the species). Small species (e.g. diving petrels) can be held four birds per box (with two interlocking diagonal dividers).
Newspaper	Lots	Enough to place a minimum of six layers in each box. Line transfer boxes with folded newspaper only for improved grip and to absorb excrement. Avoid using shredded paper as chicks may overheat.
Anti-slip matting	Sufficient to cover floor area of transfer boxes	Use black if available (to keep the inside of the box dark). This can be sourced at homeware stores (e.g. Briscoes) and is branded as matting for use on shelves or in kitchens to prevent items from slipping on surfaces. It is a perforated foamy plastic that is used as brooder matting in captive management. When taped securely over the top of newspaper, it prevents birds from slipping around, and excrement gets pushed through the perforations and is absorbed by the newspaper layers.
Duct tape	A few rolls	To secure anti-slip matting onto transfer box floors, if used. Must be applied in such a way that the tape cannot peel off and stick to the birds, e.g. by passing the tape under the flapped floor of the box. Also used to tape down the handles of transfer boxes if needed, so that they can be stacked more easily into a helicopter.
Plastic bin bags (large, drawstring)	One bag per box	To cover transfer boxes if boating or in case of heavy rain.
Tarpaulin (and rope and poles if required)	Large	Preferable method for keeping boxes shaded and dry, if required, as it still allows for ventilation.
Packing tape	One roll	To stick onto transfer boxes for writing on in marker pen (can be peeled off fluteboard after transfer so that the boxes are clean for the next year).
Permanent marker pens	4+	To clearly mark the natal burrow number (and fence status) at the source location on the transfer box.
Backpacks with long straps and/or string	One per person	One person can carry four transfer boxes (maximum) in one go if at least one box is attached to a backpack, although this is dependent on distance and terrain.

* Port Nicholson Packaging Ltd—General manager: Michael van Boheemen (michael@pnp.co.nz). PO Box 38133, 33 Fitzherbert St, Petone; email: sales@pnp.co.nz; ph: 04 568 5018; fax: 04 568 5538; website: www.pnp.co.nz. Their animal carry boxes (425 × 240 × 310 mm high) are a standard cut. Ventilation holes are a standard 20-mm diameter, but are only placed in the two long sides of the boxes; these are suitable for medium-sized and larger species (e.g. small gadfly petrels and larger), but you may need to make additional holes, if required. You can also request no ventilation holes, so that you can make smaller ventilation holes for smaller species such as diving petrels or fairy prions (*Pachyptila turtur*).

2.1.7 Data management

ITEM	QUANTITY	COMMENTS
Laptop	1	**Only if power source is taken**. Chick selection is easier if data can be entered onto a laptop and expected wing lengths calculated. Also, having some capacity to check the weather outlook is important, particularly if on the island for som time, during which the weather situation can change and there is unreliable or limited contact with mainland weather updates.
Generator, fuel and leads **or** Solar panel and inverter	1	For laptop.
CD-ROMs (and/or flash drives)	4	To back up data and to make copies of data sheets to send with chicks to the release site.
Data recording sheets—chick selection form	Lots	Refer to section 10.2. Paper data sheets should be taken as a backup for data in case of power failure, to enable chick selection.
Collection instructions and selection guidelines	1	Selection guidelines / colour coding for burrows to be copied into the back of notebooks.
Copy of approved translocation proposal and permits	1	To refer to as required.
Calculator	1	If laptop is not used or not working.

2.2 Release site chick housing, feeding and monitoring

2.2.1 Making sloping-ground/cliff artificial burrows

Note: This list can also be adapted for making flat-ground burrows, as the required materials are largely similar.

ITEM	QUANTITY	COMMENTS
H4 treated rough-sawn pine, 500 mm thick	Length as appropriate	For burrow chamber roof and inspection lid. Refer to the appropriate best practice document for the species for measurements.
H4 treated rough-sawn pine, 25 mm thick	Length as appropriate	For burrow walls. Refer to the appropriate best practice document for the species for measurements.
Measuring tape	1	To measure wood lengths.
Drop saw (preferably) or skill saw, and 114- or 165-mm-hole saw	1	To cut wood to length and to cut entry hole. The diameter of the entry hole should be 114 mm for smaller species and 165 mm for large species (e.g. grey-faced petrels).
Power drill with 4-mm bit and screwdriver bit	1	To pre-drill holes for screws and to drive screws.
Stanley knife or similar	1	To cut rubber sheet.
Pre-cut/pre-drilled parts for each burrow packaged together	1 roof; 1 lid; 2 side walls; 1 end wall	Timber can be sawn and drilled on the mainland and then transported to the release site for assembly. The lid and roof pieces need to be numbered to ensure a good fit.
Jig	1	To hold pieces in the correct position while putting screws in.
75-mm screws for roof	4+ per burrow	There has been some discussion about the use of screws versus nails to hold burrow boxes together, and about stainless steel versus galvanised steel. Stainless steel screws are very weather resistant but expensive. Zinc-coated screws have been used instead but are likely to rust and may not last as long as the wood (although silicone can be used to give the top screws some waterproofing). Therefore, stainless steel nails may be a more durable and affordable option for burrows.
50-mm screws for walls	4+ per burrow	
Butyl rubber hinges	1 hinge per burrow	Butyl strips are used as hinges for the burrow lid and run across the entire width of the burrow (i.e. strip size is dependent on burrow width).
Silicone sealant and applicator	1 tube	Silicone is applied under the rubber to make the hinge watertight.
Wooden (treated) beading	2 strips per burrow	To screw on over both edges of the rubber hinge to keep it in place. Size is dependent on burrow width.
Screws for hinge beading, at least 25 mm	6–8 per burrow (3–4 per strip of beading)	To screw on hinge beading.

2.2.2 Installing sloping-ground/cliff artificial burrows

Note: This list can also be adapted for making flat-ground burrows, as the required tools are largely similar.

ITEM	QUANTITY	COMMENTS
Template burrows to use for digging	At least 4	Leave at least four boxes without roofs (i.e. three walls, perhaps with a temporary narrow brace across the top at the back to keep its shape in the absence of a roof). These can be used as templates for the installation process.
Ridged PVC drainage pipe	1 per burrow	To create a tunnel to the artificial breeding chamber. Refer to the appropriate best practice document for the species for tunnel length and pipe diameter.
Hacksaw (plus spare blades)	1	To cut ridged PVC drainage pipe; leaves a tidier edge than a larger saw.
Stanley knife	1	To trim cut ridged PVC drainage pipe.
Measuring tape	1	To measure the distance between pairs of burrows.
Flat, sharp spades	4	To remove turf and dig holes.

ITEM	QUANTITY	COMMENTS
Hand trowels / grubbers	4	To dig out narrow trenches.
Ramming sticks or hammers	2	To compact soil around burrows. Sticks need to be c. 20–25 mm by ≤50 mm wide and c. 450 mm long.
Slasher / hedge clippers	1	Quite useful for shaving off turf around entrances.
Spirit levels	2	To check the angle of the burrow and pipe. Needs to be short (230–250 mm) to fit easily into holes. Ideally both levels should be the same make and model to ensure that burrows are installed at a standard angle.
Sand or fine beach gravel	10–20 L per burrow	At least a 10 L bucket load of this should be placed in each burrow for small to medium sized species (such as small gadfly petrels) and 20 L for large burrows (grey-faced petrels).
Shovel	1	To mix/move sand.
Old 10 L buckets	Several	To carry sand or beach gravel to burrows. Also useful for putting soil in for removal from the site—otherwise it becomes a mud bath in the rain. It is best not to smother the grass so that the site is well grassed over by the time of transfer. Several needed (c. 2 per person) to move/store spoil for repacking burrows, move sand/soil mixes, etc.
Old 20-L buckets	Several	As above. Note: 20-L buckets can be heavy for volunteers to move when full of soil/sand.
Paint (white or bright colour) and paint brush	0.5-L	To number burrows. Appropriate clean-up gear is also needed.
Burrow markers	100	To number burrows using a numbering system, which will make them easy to find each season, especially in forest.
Gardening gloves	One pair per person	To protect hands.
Rocks or mesh gates	Two per burrow	To blockade the pipe entrance. Two blockades are required for most species (chamber end and entrance end). Refer to the appropriate best practice document for the species for suitable materials. Birds must not be able to push past these, but you need to allow ventilation air flow.

2.2.3 Food / oral fluids

The following items are based on the Brunswick™ sardines in soya oil diet.

ITEM	QUANTITY	COMMENTS
Brunswick™ sardines in soya oil (106-g tins)	Refer to appropriate best practice document	Ring-pull tins.
Mazuri® Vita-zu® Bird Tablets— Small 5M25	1 tablet per 3 tins (106 g) sardines	Sourced from Napier City Council (contact Regan Beckett). Always check with the supplier on the tablet type (product number) being supplied and the recommended dosage for each batch. Where possible, request a copy of the composition of the tablet supplied and an expiry date. Note that Mazuri® recommend a dosage rate for the 5M25 tablet of 1 tablet per 225 g of fish, but that nationally, a rate of 1/3 tablet per tin (106 g) of sardines is currently preferred.
Supplementary oil	Species dependent	**Large gadfly petrels only**: refer to the grey-faced petrel best practice document (Gummer et al. 2014b).
Lactated Ringer's™ solution; Hartmann's™ solution; or Vy'trate™	2–3 L	Isotonic fluids for rapid oral hydration on transfer day. These products are similar, with different brands recommended by different vets. 3 L is needed for larger species that are given up to 30 mL of fluids on transfer day.

2.2.4 Preparing food and feeding chicks

ITEM	QUANTITY	COMMENTS
Large kettles	2+	With the ability to boil water for 3 minutes (precaution to avoid contamination).
Blenders with separate bases and jugs (800 W)	2	The two models should be the same, so that the bases can be rotated to save straining the motor of one with continuous use during food preparation. Sharpen blades before storage.
Portable generator	1	To run the blenders if no mains power.
Extension cable	1	To connect the generator to the blender.
Multiboard or two-way adaptor	1	So that both blending bases can be plugged in at the same time (although only one will run at a time).
Small kitchen knife	1	To extract fish from tins.
Standard dining knife	1	To open tins.
Small pestle and mortar	1	To grind vitamin tablets to a fine powder.
Small plastic spatula	1	To scrape blended fish from the blender.
Small measuring jug (e.g. 250 mL)	1	To measure water for the food mix. Must be able to measure to 10 mL (for water).
Plastic pottles	Species dependent	To store blended food (must be able to fit in the hot-water bath; therefore, pottle size dependent on the type of food-warming bath). Size and number depends on species: 300–500-mL pottles for small species; larger pottles (e.g. 1 L) for larger species. A few kg of puree might be prepared for smaller species, while 10 kg of food might be prepared for the large species (e.g. grey-faced petrels).
Plastic tubs	4	To use as rinse baths.
Plastic boxes (with lids)	1–2	To transport assembled syringes / feeding tubes.
Plastic bottles (3 L)	2–3	To store boiled (>3 minutes) water overnight and to carry fresh/clean water to the feeding site.
Plastic pump bottle (1 L)	1	To store boiled (>3 minutes) water for rinsing off antibacterial solution from syringes, crop tubes and pottles.
Small plastic funnel	1	To fill water bottles.
30 mL or 50 mL Plexi-vet syringes **or** disposable syringes	4–8	Refer to the appropriate best practice document for the species for suitable size and quantity (dependent on meal sizes). Species that are fed ≤50 mL require one syringe/tube per bird; while those fed >50 mL require two syringes/tubes per bird. Long-lasting plexi-vet syringes can be obtained from Shoof International Ltd farm products (www.shoof.co.nz).
Custom-made Teflon™ crop tubes **or**	4–8 + spares	Refer to the appropriate best practice document for the species for suitable size and quantity (see above). Teflon tubes are prepared by creating a screw-thread at one end and rounding off the other end. The tubes are then screwed directly into the syringe barrel (hand-tight only) once the metal Luer-lock fitting has been removed. These can be disinfected in the field and used for multiple birds (refer to section 8.7—'Food hygiene and temperature control').
Catheter tubing	One per bird (or 2 per bird if 2 food syringes)	Separate catheter tubes are needed for each bird to prevent cross infection between birds and contamination of food. The catheter tubes are more difficult to disinfect in the field.
Castor oil	1 small bottle	To lubricate syringes.
Spoons or plastic spatulas	10+	To stir food when warming (clean one for each new food pottle). Type and number are dependent on species (size and number of food pottles). Handle length is important so that the food warming bath lids close effectively while the spoon is left in to stir the food regularly.
Metal thermos flasks (min. 1.8 L)	2–6	To carry boiling water to the site for use in the hot-water bath (two 2-L flasks). Amount needed is dependent on species size (i.e. size of the water bath). More would be required for a grey-faced petrel transfer if feeding all chicks on one day.
Food thermos flask (smaller species) **or** tall yoghurt maker (larger species)—warm-water bath	4 (+ spare)	To warm food for up to one hour per batch prior to feeding. Pottles must be able to rest inside but not fall right in. For each feeding team, one is in use while a second is gently warming the next batch in the meantime.
Small chilly bins (smallest size, e.g. for a six-pack)	2+	An alternative warm-water bath, especially for larger species (bigger food volumes).
Soft tissues	20+ boxes	To wipe chicks and crop tubes during and after feeding.

ITEM	QUANTITY	COMMENTS
Kitchen paper towels	10+ rolls	To dry hands at the feeding site and during food preparation, to wipe down benches, etc.
Chilly bins (medium size)	1–2	To carry food to the feeding site each day. Pots of food need to be kept cool (with ice packs) for use later in the day. Size and/or number are species and site dependent. Two or more heavy-duty chilly bins would be required for feeding 80 birds of a large species (e.g. grey-faced petrels), as up to 10 kg of puree may need to be stored in pottles over the day (100 mL meals).
Ice-packs (chilly slicks)	10+	See above. Also used to pack with a dead chick in transit. Number dependent on species (food quantity prepared).
Freezer	1	To freeze ice packs.
Old hand towels	8+	To rest a chick on the surface when feeding. Note: Washed towels need drying time!
20 L buckets (clean)	2–4	To carry gear to the colony site and to use as lined rubbish bins in the feeding shed.
Fish bins (clean)	2	To store gear at the feeding shed.
20 L water containers (with tap)	2–4	To store fresh (non-boiled) water (for hand washing, etc.) at the feeding site. Number dependent on team size.
Benches in feeding shed	To allow two feeding teams	To spread out feeding equipment and rest chicks on for feeding.

2.2.5 Hygiene

ITEM	QUANTITY	COMMENTS
Dettol® antibacterial soap	2 + refills	To wash hands (one for food preparation area and one for feeding site).
Antibacterial wipes	1 box/tub	To clean hands quickly at feeding site.
Chlorhexidine, 5% concentration (runny pink liquid; not soapy hand wash)	500 mL (comes in 500 mL bottles)	For short-term disinfection of feeding equipment (dilute 5 mL with water to make up 100 mL). Available from veterinary suppliers. Amount dependent on species—e.g. up to 500 mL for most small and medium-sized species; up to 1 L may be needed for large species such as grey-faced petrels.
Bottle with lid	1 of e.g. 1 L	To hold freshly made chlorhexidine solution.
Small measuring jug (e.g. < 50 mL)	1	To measure chlorhexidine. Must be able to measure 5 mL.
Jars (100 mL glass caper jars)	2–4	To stand crop tubes in for disinfecting, while minimising chlorhexidine waste (which is quite expensive). (Gregg's® plastic spice jars with labels soaked off also work well.) Taller jars may be required for longer crop tubes (e.g. for grey-faced petrels). Four jars needed for species that take > 50 mL per meal.
Ice cream tubs (2 L) with two holes cut out in base	1–2	Upturned, these can be used as stands to hold the jars of crop tube disinfectant. Two 'stands' needed for species that take > 50 mL per meal.
Milton tablets (1 tablet to 2 L of water)	10–20 boxes	To disinfect equipment overnight. 30 tablets per box. Available from supermarkets. Amount dependent on the quantity of equipment needing to be sterilised.
Dishwashing liquid	5 L	To wash equipment daily—use plenty and rinse thoroughly!
Rubber gloves	5 pairs	One pair for washing-up, one for Trigene® and one for Napisan®—plus spares.
Dishwashing brushes	2	New one to clean petrel dishes, old one to clean sardine tins.
Pipe cleaners	1 packet (c. 20)	To clean crop tubes. Available from Spotlight stores (craft item)—called 'chenille sticks'; 6 × 300 mm.
Thick dishwashing sponges	2	To clean inside syringes (rolled up). Need to be correct thickness to be effective.
Bottle brush	1	To clean sterilising solution jars.
Dishwashing tubs	2	One for dirty and one for rinsed equipment. Allows economical use of very hot, soapy water.
Drainage racks or trays	2	To allow disinfected equipment to air dry.
15–20 L buckets	2	One for soaking equipment in sterilising solution; one for soaking towels and weigh bags in Napisan®.

ITEM	QUANTITY	COMMENTS
Napisan®	1 kg tub	To soak holding bags and feeding cloths. A sterilising type may be available.
Disposable kitchen-tidy bags	30	For the daily load of fishy clean-up tissues (alternatively, supermarket shopping bags can be used).
Large black rubbish bags	10	For rubbish, to store washed sardine cans, etc.
Packing tape	1 roll	To seal rubbish bags and label boxes.
Trigene®	Small bottle	As required, to clean transfer boxes. Recommended alternative to Virkon®, as used by DOC. Check with DOC for amount required (it is quite concentrated). Probably need to be able to make up to around 10–20 L in total.

2.2.6 Weighing and measuring chicks

ITEM	QUANTITY	COMMENTS
Plastic tool boxes	2–4	To carry birds between their burrows and the shed, particularly in bad weather. (Can be sourced from hardware stores.) Some operators prefer to use transfer boxes for this as, although tool boxes are darker, feathers can become trapped so great care must be taken. Size dependent on species.
Newspaper	Lots	To line carry boxes.
Coloured markers (e.g. pegs)	8+	To place on the burrow when a bird is removed and put into the corresponding colour-marked carry box, to ensure that chicks are returned to the correct burrow.
Holding (weigh) bags	10–20	Refer to the appropriate best practice document for the species for suitable bag size. Number dependent on species and frequency of feeding. Dark-coloured, thick cotton pillowcases can work well (of the same brand/size so that all bags are the same weight) for medium to large species. However, custom-made bags that are all of equal weight and made from dark cotton material (not too thick as it takes longer to dry after washing) are ideal. No cord is required at the top.
Bulldog clips	1 per scale set	Useful for attaching to scales (and recalibrating) when weighing heavy chicks, to prevent bags from slipping out of the scales and dropping.
Pesola scales **or** electronic digital tabletop scales (c. 1 kg)	2	If using electronic scales, spare batteries are needed; Pesola scales would be required as a back-up.
Box for weighing chicks using tabletop scales	2	If using electronic scales, the chick can be placed in a deep rectangular box of an appropriate size for the species.
Wing rule	1+	300 mm is ideal for small/medium species, but longer rules are required for larger species (e.g. grey-faced petrels). For larger species, it is better to custom-make a long rule (e.g. fix a metal rule onto a wider piece of plywood with a stopped end) to obtain consistently accurate measurements, rather than use a tape measure.
Banding kit	1	To remove a band if any injury occurs at the release site, or to band any new immigrants. Should contain bands, banding pliers and circlip pliers.
Washing line and clothes pegs	1 + lots of pegs	To hang up soiled holding bags and towels if needed.

2.2.7 Chick health / first aid

ITEM	QUANTITY	COMMENTS
List of relevant vet contacts	1	Refer to the appropriate best practice document for the species.
Sterile saline solution (10 mL plastic vials)	10	Useful for flushing eyes (if damaged during transit) or wounds.
Betadine® gel	1 small	To treat open wounds.
Bandage—flexible and self-adhering, e.g. 3M Vetrap™; 'vet direct cohesive bandage'; 'Co-plus'; or 'Easifix'	1 roll	Self-adhering bandages are recommended for bandaging birds (e.g. for a sprained wing) as they do not stick to the feathers.
Small sharp scissors	1	To cut bandages.
Spray bottle	1	To spray plumage if needing to stimulate preening.
1 mL disposable syringes	As required	Easiest way to administer drugs on an individual basis.

ITEM	QUANTITY	COMMENTS
Lactated Ringer's™ solution; Hartmann's™ solution; or Vy'trate™	1 L	Isotonic fluids for rapid oral hydration, if required (e.g. directed by a vet). These products are similar, with different brands recommended by different vets.
Needles (sharps) and suitable-sized disposable syringes (e.g. 35 mL for grey-faced petrel sized chick)	Lots	To draw electrolyte fluid from the bag via the port, allowing the remaining fluid in the bag to stay sterile. The sharp is removed from the syringe and the fluid administered orally using the crop tube that is usually used for feeding. For single use only.

2.2.8 Handling dead chicks

ITEM	QUANTITY	COMMENTS
Plastic zip-lock bags (A4 size)	20+	For sending dead chicks, samples, etc.
Plastic disposable gloves	20+	For handling dead birds, faeces, etc.
Polystyrene chilly bins (c. 300 × 200 mm)	6	For sending dead chicks with an ice pack to Massey University.
Ice packs (chilly slicks)	10 for dead birds	For sending dead chicks to Massey University (2 per box as boxes are quite large).
Wildlife health submission forms	Several	To include with dead chicks that are sent for post mortem. www.massey.ac.nz/massey/fms/NZ%20Wildlife%20Health%20Centre/huia_submission_form.pdf (viewed 1 May 2014)
Printed address labels to Massey University	Several	To quickly label chilly bins containing cadavers. Address can be found at www.massey.ac.nz/massey/learning/departments/centres-research/wildbase/wildbase-pathology/how-to-submit-a-specimen.cfm (viewed 1 May 2014)

2.2.9 Keeping records

ITEM	QUANTITY	COMMENTS
Data recording sheets	1 per chick	Refer to section 10.3. Hard copies should be kept in a folder in the shed; data can then be entered onto a laptop daily.
Clipboards or folders	2	One per team for data sheets in the shed.
Waterproof notebooks	2–3	For chick roll-calls; also used by handlers as required.
Pencils	10+	To record information.
Band/burrow lists (printed after transfer)	2	To quickly locate the home burrows of wandering birds that are found away from their own burrows. Should be listed in band order.
Laptop computer (and USB memory stick)	1	For data entry and backup.
Copy of approved translocation proposal and permits	1	To refer to if required.
Best practice documents for translocations—for the relevant species; and this document	1 printed copy of each	See www.doc.govt.nz/publications/science-and-technical
Chick feeding calendar	1	Refer to section 10.4.
Calculator	1	For backup, if the computer fails.

3. Guide to inspecting natural burrows at the source colony (recce trip)

3.1 Equipment preparation

1. Ensure that the following equipment is taken to each search area at the source colony:
   - One bird kit per person (each containing a trowel, notebook, pencils, wing rule, holding box for chick, kitchen paper towels, disposable gloves).
   - Plenty of plywood boards and rocks (to use as study-hole covers and to repair damaged burrows).
   - Spare artificial burrows and pipes (to repair damaged, occupied burrows—only if approved by the relevant DOC office).
   - A full set of tools for burrow installation (only if approved).
   - All equipment required to mark burrows.
   - Hand cleaning products (to use before eating and to remove sun-screens and sanitizers before handling birds to avoid spoiling plumage) or disposable latex gloves (for eating or for handling birds).

   Note: Some teams may prefer to separate the tasks, so that some people are searching and excavating, while others (with cleaner hands) are handling chicks as they are found and recording data.

3.2 Searching for burrows

2. Before commencing any searches, ensure that all personnel have received training in how to:
   - Investigate a burrow—approaching every burrow cautiously, as if it contains a small chick or even a late incubating adult (less likely).
   - Make a study-hole in a natural burrow and properly cover it after inspection.
   - Repair a collapsed burrow, usually with boards (or by installing a box and pipe where approved by the relevant DOC office and where considered necessary).
   - Extract, handle and measure a chick (including anticipating the regurgitation response and dealing with regurgitation incidences).
   - Record the data.
   - Mark the burrow using a method that has been approved by the relevant DOC office. (This usually requires discussion with Biodiversity staff, who will be aware of all other monitoring programmes using markers that are being carried out at the source colony location.)
3. Search each burrow area as a group, systematically covering sections within each colony area on the island. Mark inspected burrows either with tape (if they contain occupants) or with a stick placed at the entrance of the burrows (if empty).
4. Searchers need to consider how they approach burrow groups to minimise damage. It would be beneficial to temporarily mark commonly used trails to burrow groups if they are not already obvious (e.g. with occasional plywood boards where needed and flagging tape or wands).

   Note: If it is not considered necessary to wear petrel boards on the feet, searchers may still need to carry some plywood boards with them to use as required, i.e. to place on the ground to manoeuvre over or kneel on particularly fragile areas.

5. Knowledge of previous burrow marking systems will be useful when searching for burrows.

   Note: It can sometimes be difficult to connect markers with the burrows, especially where tags have been nailed to trees that have several burrows nearby, or where tags have been attached to branches or trees that have since fallen.

3.3 Inspecting burrows

6. Inspect the burrow via its entrance and:
   - If a bird or empty nest chamber is felt, record the burrow as a 'reach-in' (i.e. chamber accessible via entrance).
   - If no bird is felt, insert a short, soft-ended stick into the burrow until all chamber walls can be felt. The burrow can then be declared empty or occupied. If considered occupied, a study-hole may be required (see below) to safely access and extract the chick, and to prevent any further burrow damage, particularly around the entrance.
   - **For larger species, e.g. grey-faced petrels:** If no chamber walls are felt using a short stick, insert a longer soft-ended stick (e.g. flax flower stalk) to determine whether the burrow is occupied. If a bird is then felt, there are two options:
     - In most cases, a study-hole will need to be made (see below) to safely access and extract the chick. Ideally, searchers should only invest time in creating study-holes at burrows that they suspect are occupied, rather than at burrows of unknown occupancy; this also helps to minimise general burrow damage at the colony.
     - Alternatively, there is a chance that the chick can be gently encouraged up the tunnel to the entrance by allowing it to nibble or grip on the end of the flax stalk (refer to section 3.5—'Extracting and processing chicks at burrows').

   Note: If the occupant feels like an adult, there is no need to know whether it is on an egg or a small chick, as the chick will not be of use for transfer. Similarly, if it is unclear whether the occupant could be a large, well-feathered chick rather than an adult, there is also little point in disturbing it further because such an advanced chick would be too old to transfer.

7. Create a study-hole to the side of a suspected chamber location in a natural burrow as follows:
   - Determine the likely location of the chamber by laying your arm (with stick extension) on the surface above the burrow in the same position as it lay in when inside the burrow.
   - Mark a spot on the ground that is estimated to be the furthest point that can be reached inside the tunnel. Imagine this to be the point that you want to arrive at (underground) after digging an **angled** hole. Start digging to the side of this (as far as possible from the burrow entrance) and aim towards the marker as you dig down. Ideally, final access into the chamber should be through the side wall as opposed to the chamber roof directly above any potential occupants for the following reasons:
     - To prevent soil from caving in on top of the occupants.
     - Because the chick can be more safely extracted up a gentle slope rather than through a vertical hole; and if it regurgitates as it is being brought out of the study-hole, the vomit should end up on the study-hole floor rather than all over its plumage (a potentially fatal scenario that can occur if a chick is brought up through a vertical study-hole).

     Note: Study-holes that are positioned too close to the entrance (i.e. less than 300 mm away) can cause weakening and possible collapse of the tunnel roof.
   - Dig away the soil (preferably by hand or with a small, narrow trowel) and continually remove all of the loose soil.
   - Ensure that the study-hole is no wider than required to allow the bird to be comfortably extracted and is well-covered with a rock (only if very firm ground) or boards (weighed down with a rock) to keep the burrow light- and waterproof.

- A second study-hole may need to be made if the tunnel is long and the chamber is still not accessible from the first study-hole. Second study-holes should be made as far away as possible from the first to avoid weakening the entire tunnel roof.

Note: Some burrows (e.g. Cook's petrels) can be very long (i.e. 3+ m), and so several study-holes often need to be dug to determine the location of the chamber.

8. When covering up a study-hole, ensure that it is safe, lightproof and waterproof by checking that:
   - Any rocks that have been used to block the study-hole are not so heavy that they will collapse the burrow. Rocks must fit snugly, without gaps.
   - All four edges of any plywood boards that have been used to cover the study-hole lie flush on the surrounding ground and are weighed down effectively with a rock. Replace the soil over the board, as this will provide thermal advantages to the burrow, as well as preventing erosion around the sides and exposing the nest chamber.

   Note: It can be useful to indicate the position of the chamber and the burrow entrance (if they are not obvious) by drawing arrow(s) with permanent marker pen on the plywood board once it is in place, for quick future reference.

3.4 Anticipating chick regurgitation

9. Ensure that all handlers know exactly how to deal with chicks that are about to regurgitate, or have already regurgitated. **It is critical that a chick's plumage is not soiled by its own regurgitant** as this will greatly compromise its plumage condition with respect to insulation and waterproofing—such chicks are unlikely to survive at sea, even if they are still many weeks from fledging.
10. Always handle a chick in anticipation that it is going to regurgitate, aiming to keep its head area clear so that it can project the vomit away from its body. This will help to keep the plumage clean and considerably reduce the risk of the chick aspirating the vomit.

    Note: If a chick is going to regurgitate, it is likely to do so during or immediately after extraction from the burrow, and usually on the very first handling event. Bear in mind, however, that a chick may still vomit during further handling; if this happens, the chick must be **immediately** removed from the area of projection to prevent soiling of its plumage, which can be fatal.
11. In the event that a chick's plumage is soiled, attempt to clean it as best as possible using kitchen towels and/or fine dry sand, or dry dirt and dust.

    Note: It has been found that running fine dry sand, or dry dirt and dust over the feathers effectively soaks up the oily vomit and helps it to drop off the chick. Wet sand/dirt does not absorb the oil.
12. Avoid including a chick with badly soiled plumage in a transfer, because it will have a considerably reduced chance of surviving after fledging unless it can undergo prolonged waterproofing therapy before fledging, and may also have dropped below the transfer criteria.

3.5 Extracting and processing chicks at burrows

13. Treat young, less-mobile chicks differently from adults when extracting them from a burrow. Aim to support the entire chick's body during extraction. When extracting small chicks, it is important to avoid:
    - Dragging very young chicks and/or resistant chicks out of burrows using the bill. This technique is often used to bring an adult or more mobile older chick forwards out of a narrow tunnel or study-hole, but can be damaging to a younger chick with a weaker head/neck area and undeveloped mobility, especially if it shows resistance. In addition, chicks can be prone to regurgitating oily meals during handling, and if the bill is held there is a high chance that the regurgitant will flow back over the downy chick, which can prove fatal through spoiling of insulation and/or asphyxiation.
    - Extracting chicks backwards (e.g. out of narrow tunnels or study-holes) as, unless absolutely sure that all wings and legs are contained in the hold, there is a high chance of limb damage.
14. **For larger species, e.g. grey-faced petrels**—If using the method of gently encouraging a chick up the tunnel to the entrance by allowing it to nibble or grip on the end of a flax stalk, operators must be aware that young chicks may be less mobile and more vulnerable to neck strain if they are resistant; wings and legs can become stuck behind roots and damaged; chicks may be dragged through their own regurgitant; and eyes can be easily damaged (either with the stick or from brushing past tunnel walls). Therefore, to help avoid injuries, use the stick to get a feel for the terrain inside the tunnel, and do not attempt to lead a chick up a tunnel if it is narrow in places or contains obstacles such as rocks or tree roots. Also, be aware of how resistant the chick is—if it appears to have locked up or is not moving, do not force it. If the chick's beak gets snagged on the stick, there is also a higher risk of needing to force it and causing potential injury.
15. Avoid putting chicks in bags unless it is absolutely necessary. **Note that weights are not required during the recce trip**. If the chick does need to be placed in a bag, allow at least 30 seconds to pass before putting it in, to allow it to regurgitate beforehand. Watch the chick carefully when placing it in the bag and be ready to quickly withdraw it if needed. Avoid pressing a chick's head up in one side of the bag—aim to give the chick some space around the head area inside the bag for the potential projection of vomit.

    Note: Chicks that regurgitate inside a bag have a high chance of becoming soiled. Once settled and held reasonably firmly (i.e. not allowed to jump about inside the bag), the chances of regurgitation occurring are reduced. It may be a better option to put large, very recently fed chicks, e.g. grey-faced petrels, into a dark box of known weight (e.g. transfer box), and weigh the box and the chick with a large set of Pesola scales, to avoid needing to put the chick in a bag.
16. Inspect the chick's right wing and proceed as follows:
    - If there are feathers present and the tip of the feathers can be clearly seen (i.e. there are no wisps of down), measure to the tip of the longest primary (not to the tip of any down that may be stuck to the end of the primary).
    - If wing feathers are absent (i.e. only down is present), no measurement needs to be taken. Record that the wing is downy.
    - If pin feathers are developing but are only very short and do not extend beyond the down, no measurement needs to be taken. Record that pins are emerging.

    Note: It is important to ensure that all handlers are measuring and recording consistently.

3.6 Repairing or preserving burrows

17. If any work needs to be carried out at the burrow, place the chick in a dark, ventilated box. The chick should not need to be placed in a holding bag (unless it is too large for a box), as weights are not required on a recce trip, which is when this kind of burrow repair/preservation is most likely to be carried out.

    Note: Although holding bags will be carried, they probably will not used much on a recce trip.

18. Install artificial burrows at damaged and/or extremely fragile sites if required and approved (refer to the appropriate best practice document for the species and section 4.1—' "Flat-ground" burrow design at source colony').

3.7 Returning chicks to burrows

19. Always return a chick directly to its chamber by placing it with its head facing the back wall, to prevent the chick from moving down the tunnel as soon as it is released.

    Note: Forcing a chick to walk through the tunnel before it would naturally do so during its first emergence is not considered best practice, as the chick may not always end up back in the safety of the nest chamber. In addition, this may make the chick more restless and cause it to emerge a little earlier than perhaps it might have had it never ventured up the tunnel before.

20. Mark the position of study-holes and artificial chambers by tying coloured flagging tape to rocks or burrow lids. Label the tape with the burrow number, date and burrow content (for future reference during recce and collection trips)—this would be considered temporary marking to last the current year. More permanent markers also need to be added (refer to section 5.3—'Marking burrows').

    Note: All markers should be removed at the end of projects to avoid littering the location.

21. Record the findings at each burrow; see section 10.1 for examples of the types of data that should be recorded at each burrow.

4. Guide to installing artificial burrows

4.1 'Flat-ground' burrow design at source colony

The following methods are for installing flat-ground artificial burrows at **source colony burrows** that are located in fragile soil under forest and contain chicks. (Note: This should only be carried out if approved by the relevant DOC office for damage control purposes.) The artificial burrows must be carefully positioned around an existing burrow. To date, this has been undertaken for small gadfly petrels such as Chatham and Pycroft's petrels (using wooden or plastic boxes and 110-mm-diameter PVC drainage pipe), as well as diving petrels (using wooden boxes and wooden tunnels).

It is vital that the burrows are installed correctly and checked by experienced personnel or those under the instruction of experienced personnel—an incorrectly installed burrow can result in death of the chick (i.e. if one or both adults cannot enter the burrow as they normally would, the chick can be abandoned) and burrow desertion.

Note: The chick-rearing phase is the best time to install boxes at occupied natural burrow sites, as adults are strongly fixed to the burrow and will accept the tunnel changes in order to reach the chick, which they will be able to smell and hear in the chamber.

1. Check the burrow thoroughly to ensure that no adults are present. By this stage, the burrow will be damaged, so checks are likely to be made through broken holes in the chamber wall or roof. (Note: An adult that is present with a young chick during the day may be skulking at the back of the chamber.) If an adult is present, it would be better to temporarily preserve the burrow using plywood boards and return on a subsequent day to fit a plastic burrow.
2. Remove the chick via a study-hole and quickly place it in a lined holding box to prevent exposure (particularly if a cool, damp day).
3. Place the top layer of warm nest material beneath the chick in the holding box, and remove and carefully set aside the bulk of the remaining nest material onto a plastic bag.
4. Thoroughly check all walls of the chamber and tunnel to be absolutely sure of the location of the tunnel and entrance that leads to the chamber. Some burrows can be served by more than one entrance, and in some cases both are used. It is critical to ensure that the entrance that is used by one or both adults is not blocked off.
5. **If there is any doubt at all about which entrance the adults might be using**, either:
   - Cut an additional hole in the box (using a hacksaw and knife) and fit two tunnels; or
   - Preserve the burrow using plywood boards and even floorless sections of 160 mm, pipe if necessary (see below).
6. **The entrance is the key point that needs to remain in the same place**. Therefore, using the entrance hole as the main reference point, determine the length and shape of pipe required, aiming to use one section of pipe only where possible. Bear in mind that the actual chamber location can be shifted very slightly to the left or right or closer to the entrance, but the entrance (and any surrounding features) needs to remain in exactly the same place, and the tunnel needs to be similar to the natural shape and length of the original tunnel.

   Note: It is important not to extend the tunnel length (by shifting the chamber further back from the entrance), as this will mean that the adults have further than usual to walk up the pipe, which they may be apprehensive about doing.
7. In some instances the entrance hole may need to be set slightly deeper, to avoid installing a pipe that slopes down too steeply towards the box. The tunnel needs to be near to horizontal but can have a slight downwards slope. A steep tunnel is not at all desirable as it can channel water and debris down into the burrow, and can be difficult for the chick to exit.

8. Preserve the shape and location of the nest bowl by covering it with a plastic sheet or old tea towel.

   Note: If the burrow is quite shallow, it will not be possible to preserve the nest bowl as it will need to be dug up to allow for the box to be installed and buried underground. However, if the burrow is particularly shallow and it is not possible to install a deeper burrow (e.g. due to tree roots or rocks), then the burrow may have to be slightly exposed above ground, in which case it should be surrounded by logs (to retain soil) and well covered with additional logs and soil.

9. Lift out the chamber roof and excavate a box-shaped hole.

10. Fit ridged PVC drainage pipe to the original tunnel route using one length of pipe only wherever possible. Avoid angling the pipe steeply down to the chamber; it is better to sink the entrance deeper than the original so that the pipe is more horizontal leading into the box. Steep, sloping pipes can channel water and debris down into the burrow, and can be difficult for chicks to exit.

    Note: It is best to avoid joining sections of pipe, as this can easily lead to pipe ends misaligning underground, which can result in blockages in tunnels and, ultimately, burrow desertion.

11. If a joint must be incorporated between two pieces of pipe, the joint should be well sealed with a joiner (a third short section of pipe that is 'opened' by cutting down its length with a hacksaw), which is clamped and wired over the join to prevent soil from falling through and the pipes slipping out of alignment once buried. It is important not to fit a long pipe, as the entire length of pipe must be accessible to remove blockages.

    Note: When joining sections, use a hacksaw to cut the pipe at its widest diameter to prevent any birds that are moving through the pipe from being exposed to sharp edges. If a joiner is not used, the sections of pipe will become misaligned underground over time in such an unstable environment, or soil will fall through the join, both of which will result in a blockage and, ultimately, burrow desertion.

12. Where possible, preserve and retain the natural burrow entrance by setting the end of the pipe a little way back from the entrance of the tunnel. Ensure that the chamber end of the pipe is inserted several centimetres into the box, and pack soil underneath it to fill the gap between the chamber floor and pipe.

13. Backfill/ bury the sides of the box and tunnel with compacted soil, and remove the plastic sheet/towel and debris. Ensure that the plastic boxes are completely buried underground, with only the neck/lid above ground.

14. Scatter thin a thin layer of soil throughout the length of the pipe to simulate a more natural tunnel floor.

15. Return and arrange the preserved nesting material in the chamber, and then return the chick.

16. If erecting a stick fence at the burrow entrance, it is essential not to make this a barricade. The adults must not be deterred in any way from entering the burrow.

17. Camouflage the entrance and surrounding exposed soil with a scattering of leaves.

4.2 'Flat-ground' burrow design at release site

The following methods are for installing flat-ground artificial burrows at **release sites.** It is important that the burrows are installed correctly and checked by experienced personnel, or those under the instruction of experienced personnel—an incorrectly installed burrow can result in death of the chick (e.g. if drainage is not effective or tunnels are set too steep).

4.2.1 Phase 1: Positioning the burrows

1. Select an appropriate location for the burrows, with the following considerations in mind:
   - Burrows and access tunnels need to be installed in excavations, with the entry tunnels and all but the lifting lids of the burrows below ground level (see Fig. 1 for a diagram of the position of burrows in relation to the surrounding soil).
   - Sites must be sufficiently clear of trees and shrubs to avoid their roots.
   - Burrows need to be installed singly, and burrow entrances should be ≥ 0.5 m apart for small species and > 1 m apart for large species.
   - It is a good idea to lay out all the burrows, but to work on one line at a time along the slope, so that the next line (either uphill or downhill) can be set at the correct distance from the first.

4.2.2 Phase 2: Excavating a burrow and access tunnel

2. Excavate a box-shaped hole with a trench (for the tunnel) leading from it.

   Note: If there is a very gentle slope, the tunnel should not lead downhill or uphill, but rather should travel along the slope.

3. Fit ridged PVC drainage pipe to the tunnel route using only one length of pipe wherever possible. Avoid angling the pipe steeply down to the chamber; it is better to sink the entrance so that the pipe is horizontal leading into the box. Steep, sloping pipes can channel water and debris down into the burrow, and can be difficult for chicks to exit.

   Note: Use one section of pipe only. Joining sections of pipe should be avoided (and is not necessary at release site burrows), as it can easily lead to pipe ends misaligning underground, which can result in blockages in tunnels and, ultimately, burrow desertion.

4.2.3 Phase 3: Installing the burrow and plastic access pipe

4. Fill the burrow with at least 100 mm depth of sand or very fine beach gravel (up to the level of the lower edge of the wooden wall), and also fill the trench for the pipe with a similar depth of sand. (The purpose of this sand is to promote drainage of any water that enters the burrow or access pipe.) Tamp down the sand in both the burrow and trench.

   Note: The addition of drainage material to the chamber floor and beneath the pipe is highly recommended. Only occasional sites will have soil with excellent natural drainage properties.

5. Position the box and pipe in the hole. Ensure that the chamber end of the pipe is inserted several centimetres into the plastic box, and pack the soil underneath the pipe and box walls to help position both.

   Note: Ensure that boxes with sloping roofs (to allow water run-off) are positioned in the ground correctly, so that the roof remains sloping rather than sitting level with the ground.

6. Backfill/bury the sides of the box and tunnel with compacted soil, and remove any debris from within the box. Ensure that plastic boxes are completely buried underground, with only the neck/lid above ground.

7. **Where appropriate**, scatter a thin layer of sand throughout the length of the plastic pipe to simulate a more natural tunnel floor, and return and arrange all nest material in the chamber.

   Note: For grey-faced petrels, which tend to be found sitting in the tunnel more often than other species, sand or fine gravel can be placed in the pipe so that the chicks stay clean (i.e. their plumage is not soiled by waste matter accumulating in the pipe). For small diving petrels, sand or soil can be used to partially fill the pipes, to give tunnels a smaller diameter and potentially make them more attractive (less conspicuous entrances) to returning adults.

4.2.4 Final preparation before transfer

8. Number the completed burrows in a sequence that will logically follow a walking route from the feeding station, and paint the numbers on the lids.
9. **For forest-nesting species only**—If possible, incorporate a large piece of wood (log, branch) above the entrance of artificial burrows to provide a visual marker for their burrow. The log must be completely stable and preferably partially buried in the ground. It must not be able to roll or fall in front of the burrow entrance. Many petrel species naturally dig their burrows into the base or sides of fallen trees, tree stumps and rotting logs, so this can also make the site more attractive for prospecting birds when they return.
10. Shortly before the transfer of chicks takes place, create 'nests' of dried grass or dry leaves/fronds in the chambers.
11. If it appears necessary to keep the chicks cool, place sandbags of the woven plastic variety on top of the exposed wooden lids of each burrow. These can be numbered using a spray can.

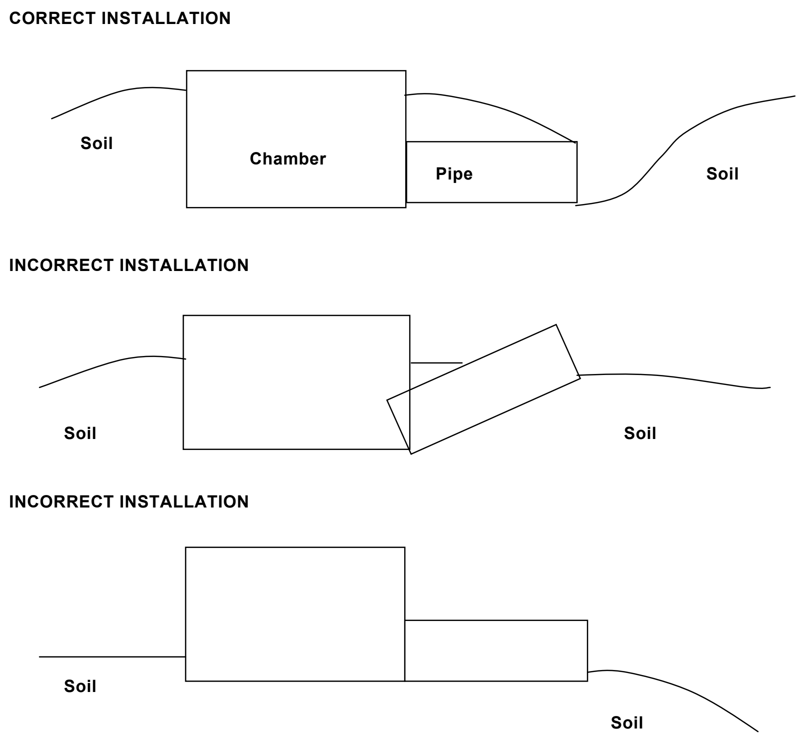

Figure 1. Position of flat-ground burrow in relation to the surrounding soil levels.

4.3 'Sloping-ground/cliff' burrow design at release site

The following methods are for installing artificial burrows on sloping ground at **release sites.** It is important that the burrows are installed correctly and checked by experienced personnel, or those under the instruction of experienced personnel—an incorrectly installed burrow can result in death of the chick (e.g. if drainage is not effective or tunnels are set too steep).

4.3.1 Phase 1: Positioning the burrows

1. Select an appropriate location for the burrows, with the following considerations in mind:
    - Burrows and access tunnels need to be installed in excavations, with the entry tunnels and all but the lifting lids of the burrows below ground level.
    - Sites must be sufficiently clear of trees and shrubs to avoid their roots.
    - Burrows can be installed singly or in pairs. The burrows of a pair should be sited on the same contour with a gap of c. 1 m between the boxes. Burrow entrances should be ≥ 0.5 m apart for small species and > 1 m apart for large species.
    - A pair of burrows should be accessed via a common access track, which starts downslope of the burrows. At its upper end, this track should split in two to form a Y-junction. The arms of the Y-junction continue as trenches and then enter plastic drainage pipe tunnels, which lead into holes of the same diameter in the sides of the burrows. Thus, for each pair of burrows, one has a left-hand entry and the other a right-hand entry (see Fig. 2).
    - It is a good idea to lay out all the burrows, but to work on one line at a time along the slope, so that the next line (either uphill or downhill) can be set at the correct distance from the first line.

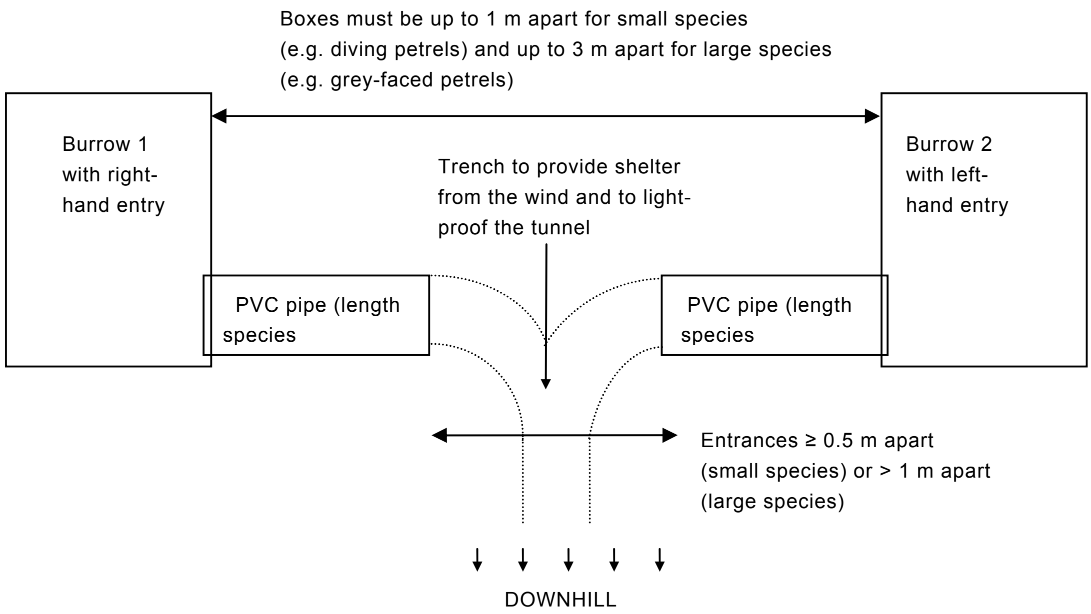

Figure 2. Positioning a pair of burrows.

4.3.2 Phase 2: Excavating a burrow, access tunnel and access trench

2. The burrow, access tunnel and open access trench need to be installed in such a way that any water within them or on the burrow lid will drain downhill. Therefore, the burrow and tunnel pipe need to be installed in the ground just off horizontal (sloping towards the entrance). The gradient of the open access trench is less critical. Gradients should be set with the aid of a spirit level; however, since different spirit levels have different-shaped glass tubes, it is necessary to check where the bubble sits in relation to the lines on the glass when the spirit level is on a slope of 2.5°. (At a slope of 2.5°, the lower end of the burrow will be c. 20 mm lower than the upper edge. To establish the position of the bubble at this gradient, place a piece of dressed 19-mm timber on a horizontal surface such as a table, place a burrow or testing frame on the horizontal surface, with one end supported by the dressed timber, and note the position of the bubble.)
3. Clear long grass from an area that is large enough to accommodate a pair of burrows along with the access tunnels and trenches to them, but leave the ground undisturbed.
4. The next stage is to dig the hole for the burrow. Start by placing a test frame comprising the front and sides of a burrow at the upper edge and to one side of the cleared area, with the long sides running downhill, and the closed end downhill and horizontal laterally. Make vertical cuts with a spade around the outside edge of the frame and across the open side, thus defining a rectangle of turf and the hole to be excavated beneath it. The amount of tolerance that should be provided for around the frame will differ according to the nature of the soil beneath the turf. No tolerance is provided for with soils such as damp clay, which cut cleanly and will hold a vertical cut from a spade, and so enable the frame (and ultimately the burrow) to snugly fit in the hole. However, where the soil is more friable, the outline needs to be c. 50 mm wider than the frame on each side and c. 50 mm longer on the downslope end (to allow sufficient space for the soil to be tightly packed around the burrow.)
5. Remove the frame and slice the rectangle of turf across its width about one-third of the way up from its lower end. Lift the resulting two pieces out as whole turfs and put them aside for later use (see step 19), taking particular care to preserve the larger turf in one piece if possible.
6. Excavate the hole to allow the frame to be fitted in such a way that its lower end is no more than 10 mm above ground level and its sides are angled at c. 4° downslope. Start by making the hole smaller than the final size required—the hole can be trimmed to its final size and shape more accurately after the bulk of the digging out has been done. Because of the slope of the ground, the upslope end of the frame will be well below ground level. The upslope end of the hole must be vertical.
7. Keep some of the more easily worked soil close to the excavation as it will be needed for backfilling (see step 14). Take the rest well away from the site, as it will get in the way and is likely to turn into mud.
8. Once the frame is fitting properly, with the open end against the uphill (back) wall of the hole, mark the bottom of the hole with spade cuts around the inside edge of the frame.
9. Remove the frame and excavate the area within the spade cuts a further 100+ mm (the deeper the better, as this area will be filled with free-draining material and a deeper layer will afford a drier burrow in heavy rains). This completes the excavation of the burrow.
10. Replace the frame in the hole. If the frame has a circular hole in one side only, this must be in the side that faces the direction the tunnel will take. The hole shows where the trench for the access pipe is to be located.
11. Excavate the trench roughly at right angles to the side of the frame. However, since the pipe is delivered in a coil, it may have a slight bend in it; this bend should curve downhill (as viewed from the top) when laid in the trench, and the line of the trench should follow this curve. The

gradient of the base of the trench should be 2.5°, but should grade down across the slope, away from the frame. The base of the trench will start at the level of the base of the hole for the burrow, which it joins, and grade down from there.

12. Continue the trench for the access pipe until it meets the corresponding trench from the other half of the pair of burrows, joining it as a tight Y-junction (not a T-junction with sharp right-hand turns). Continue the stem of the Y downslope until it runs out at natural ground level. This completes the excavation for the access tunnel and access trench.

4.3.3 Phase 3: Installing a wooden burrow and plastic access pipe

13. Place the wooden burrow in its hole, with the lifting flap downslope and the open back hard up against the vertical back wall of the hole—kick it towards the back wall if necessary to ensure a tight fit. Check that it is properly seated all the way round.
14. After a final check with a spirit level in both the across and downslope directions, backfill any space between the sides of the burrow and the sides of the hole with rammed soil. This includes carrying the rammed soil up to the top edge of the lower end wall of the burrow to promote good insulation and to discourage erosion at this point.
15. Fill the burrow with at least 100 mm depth of sand or fine beach gravel (up to the level of the lower edge of the wooden wall), and also fill the trench for the pipe with a similar depth of sand. (The purpose of this sand is to promote drainage of any water that enters the burrow or access pipe.) Tamp down the sand in both the burrow and trench.
16. Place the access pipe in the trench and fit it into the hole in the side of the burrow. The pipe should protrude c. 10 mm into the burrow (or more for larger pipes/burrows) to prevent it from becoming disconnected from the burrow if it is accidentally dislodged, e.g. by being trodden on.
17. Check the gradient of the pipe and then backfill the space around and above the pipe with well firmed in soil or turf.
18. Make a vertical spade cut the width of the burrow across the slope above the burrow, about one-third of the length of the burrow away. Make cuts at right angles to the first cut between the ends of the first cut and the upslope corners of the burrow. Undercut the rectangle so formed at the level of the lid of the burrow and remove the resulting turf.
19. Place the larger of the turfs that was removed when the hole for the burrow was begun (see step 5) over the space that is left by the removal of the turf upslope of the burrow, so that it covers both the exposed ground and the fixed part of the lid of the burrow about as far down as the edge of the hinge. Firm the turf into place to ensure that it binds with the uphill face and sides of the recent excavation. (The purpose of this is to prevent water that is running down the slope above the burrow from seeping down the back wall of the burrow.) It is important that the hinged lid can rest back on this turf when open without falling back into a closed position, i.e. there must be no risk that the lid will drop shut during a burrow inspection or chick handling procedure.
20. Firm up and ramp back the sides of the excavation on each side of the opening part of the lid to discourage loose soil from dropping down into the hinge and onto the top of the wood beneath the hinged lid, which would prevent the lid from fitting properly. At the same time, check all the lids to ensure that none have been fouled by soil. These operations may need to be repeated immediately before the transfers take place.
21. **For smaller species, such as diving petrels**—Make up a mixture of sand and soil, and insert it into the access pipes to form a floor at the same level as the sand floor of the burrow.

4.3.4 Final preparation before transfer

22 Number the completed burrows in a sequence that will logically follow a walking route from the feeding station, and paint the numbers on the lifting lids.

23 Shortly before the transfer of chicks takes place, create 'nests' of dried grass or dry leaves/fronds in the burrows under the fixed portion of the lids.

24 If it appears necessary to keep the chicks cool and prevent the lifting lids from warping, place sandbags of the woven plastic variety on top of the exposed wooden lids of each burrow. These can be numbered using a spray can.

5. Guide to selecting chicks on the selection/collection/transfer trip

5.1 Preparation: equipment, notebooks and data forms

1. Ensure that the following equipment is taken to each search area at the source colony:
   - One bird kit per person (each containing a trowel, notebook, pencils, permanent marker pen, flagging tape in a minimum of three colours, wing rule, Pesola scales, holding bags, holding box for a chick and disposable gloves).
   - A full banding kit for each qualified bander.
   - Plenty of plywood boards and rocks (for study-hole covers and to repair damaged burrows).
   - Spare artificial burrows and pipes (only if approved by the relevant DOC office for the repair of damaged, occupied burrows).
   - A full set of tools for burrow installation (only if approved).
   - All equipment required to mark burrows.
   - Hand cleaning products (to use before eating and to remove sun-screens and sanitizers before handling birds to avoid spoiling plumage) or disposable latex gloves (for eating or for handling birds).

   Note:

   - Personnel can expect to undertake the same tasks as on the recce trip in terms of burrow searching, chick extraction and damage control (refer to section 3—'Guide to inspecting natural burrows at the source colony (recce trip)'), including burrow installation if approved and necessary (refer to section 4.1—' "Flat-ground" burrow design at source colony'). Additional tasks include extra chick processing (banding, weighing and measuring).
   - Some teams may prefer to separate the tasks, so that some people are extracting chicks, while others are handling the chicks and recording data. It can be a good idea to work in pairs, with one person doing all the bird handling with clean hands.
2. Copy the chick wing length guidelines (from the respective species' best practice document) into notebook covers ready for the first chick processing day; the criteria will change with each search day, so there is a set range of wing lengths that searchers are looking for on each search day leading up to the transfer. The guidelines take into account the varying wing growth rates of a species, so that no chick is overlooked and all marginal chicks are reassessed on transfer day.

   Note: **All** grey-faced petrel chicks are reassessed on transfer day to ensure only chicks with a base weight that meets minimum criterion are taken.
3. Take plenty of paper data forms for filling out in the evening—although a laptop is preferable. The chick selection form in section 10.2 is based on one that was used for Chatham petrels. Alternatively, data fields can be put into an Excel spreadsheet and formulae entered to calculate maximum and minimum wing lengths on transfer day.

5.2 Banding, processing and assessing chicks–first measurements for chick selection

Important note: Banding cannot be undertaken at the site unless at least one banding permit holder (for the relevant species) is present. All banders must have undertaken adequate bird banding training before the collection trip.

4. It is recommended that operators work in teams of two people, as follows:
    - One person bands (while the other holds the bird, if required) and weighs the chick, and measures the right wing (for consistency), thus keeping their hands relatively clean, which prevents unnecessary spoiling of the plumage.
    - The second person checks the entire pipe length (if artificial) to ensure that there are no obstructions and there is no down around the entrance (so that chick emergence is more obvious if it occurs before transfer); makes any necessary repairs; and marks the burrow with flagging tape or wands featuring the burrow number and chick band. They then set a stick fence (at least two thin sticks).

    Note: Although competent banders can operate alone, it is more efficient to work together (particular if the weather conditions are not favourable). This also means that not everybody needs to have a banding permit, and that chicks are not handled with muddy hands (as one person processes the chick while the other repairs the burrow and erects a stick fence).

5. Weigh and measure (right wing) all chicks found, being aware that some chicks may regurgitate a highly oily meal during handling. Refer to section 3.4—**'Anticipating chick regurgitation' for essential information on anticipating and dealing with chick regurgitation incidences.** It is also useful to note the percent chick down coverage (e.g. on upper body surface).

    Note: For most species, the emphasis is on wing length at this stage, as weights can change over the next few days; however, it is also useful to obtain weights, as some exceptionally lightweight chicks can be ruled out for transfer, as they are unlikely to gain enough weight by transfer day, even after a parental meal. Most chicks will need to be weighed on a second occasion, e.g. transfer day. For grey-faced petrels, weights must be obtained on two occasions to obtain the chick's base weight (i.e. body weight without parental meal). The first weight is taken at first handling and the second no sooner than 2–3 days later (and often on transfer day itself).

6. Band each chick as follows:
    - If time permits, band all chicks found (if advised to do so by the Banding Office). Use one band number series for all the chicks that may be transferred, i.e. any chicks that fall within any of the suitable or marginal categories listed in the notebook (even though some of these chicks will not actually be transferred). This has been requested by the Banding Office manager, as it makes it much easier when it comes to reporting to the Banding Office. Use a different series to band chicks that are highly likely to remain at the source colony because they fall well outside the transfer wing length criteria.
    - If there are time constraints, band only those chicks that are likely to be transferred. Banding is the only way to be completely sure of a chick's identity and to avoid confusing data from different chicks—especially where burrows are in close proximity or nest chambers share the same burrow entrance.

    Note: It is also very useful to record the presence of adults in burrows during the day with chicks. A parent will stay in a burrow with its chick during the day if it has been unable to offload the meal it has collected for the chick—i.e. the chick is no longer accepting food because it needs to lose weight. This is very useful information for feeders at the release site. Adults can be identified and banded if time permits, and if banding has been requested by the Banding Office.

    - If there is no permitted bander present, chicks can be banded at the release site. In this scenario, it is critical that all chicks be measured and weighed on transfer day to ensure that they are the correct chicks to take (i.e. meet the transfer criteria).

7. Return the chick directly to its chamber (facing the back wall) and ensure that the following information is recorded:

- Burrow number/location
- Weight and wing length
- Down cover as a percentage (estimate) of upper body area
- Band number if banded
- Whether the chick is likely to be suitable, marginal or unsuitable for transfer

8. Erect a stick fence at the burrow entrance. This will primarily be useful for monitoring chick emergence—especially for the more advanced chicks; however, it can also be useful to know if a chick has been visited by a parent on the night before transfer, as this can assist with meal planning **(gadfly petrels only)**.

   Note: Fences that were erected at the start of the selection trip will most likely be knocked down on an unknown night before the transfer. However, if they are not knocked down, this will be very useful information for feeders at the release site, as the chick will be a priority for feeding. Fences of more advanced chicks would need to be put up again, preferably on the day before transfer, to be able to effectively monitor any potential emergence.

5.3 Marking burrows

9. Check the wing length of each chick against criteria set for the day, which will be written in notebooks, and tie the appropriately coloured flagging tape (depending on whether the chick is suitable, marginally small or marginally advanced) to the burrow lid, rock or marker as follows:
   - If the burrow has flagging tape, e.g. blue coloured, from the recce trip, add new tape of the appropriate colour. The date can be written on the new tape. Use colours as follows:
     - [Colour 1] tape—For chicks that are likely to be suitable for transfer
     - [Colour 2] tape—For chicks that are marginally small
     - [Colour 3] tape—For marginal chicks that may be too advanced for transfer
   - If the burrow has no tape, tie on tape of the appropriate colour, and write the new burrow number, current burrow content and date on the tape.

   Note:
     - The presence of tape will instantly show which burrows have been inspected
     - Any chicks that are calculated to fall just outside the wing length criteria should not be discarded—allow for some measurement error and treat these as marginal chicks that need to be remeasured closer to transfer day

10. If the burrow is empty or contains a chick that is not suitable for transfer, the burrow will need to be marked in a different way, such as by using another tape colour (e.g. a second [blue] tape or a fifth colour), or a sign (e.g. writing on the existing tape).

5.4 Confirming suitability—second measurements

11. Aim to remeasure and reweigh **all** chicks, especially marginal chicks, on a second occasion to ensure that they are suitable for transfer and that there was no error during initial processing. This can be done either:
    - During the last 1–2 days before transfer day (less common)—This can be preferable for marginal chicks that were first processed on arrival on the island, as a second wing measurement can give a clearer idea of whether a chick will meet the transfer criteria on transfer day (and if not, it can be eliminated); and it can also be useful if it is known that

there will be little time to measure all chicks on transfer day itself. However, this extra handling event is not ideal; or

- On transfer day itself (most common)—If there is plenty of time on transfer day, ideally **all** chicks should be weighed and measured before transfer. If there are time constraints, then all chicks should be at least weighed, and marginal chicks remeasured.

Note:

- If a chick has already been handled twice before transfer day, it is unlikely to need to be remeasured on transfer day unless it is in the marginal category or unbanded; however, it should still be weighed.
- The grey-faced petrel transfer criterion is based on a chick's base (pre-feed) weight, so all chicks must be reweighed on transfer day to ensure that the base weight has not fallen below the criterion.

5.5 Checking fences at burrow entrances

12. If time permits, it can be useful to visit the burrows of all chicks that are potentially suitable for transfer, especially the more advanced chicks (e.g. those that were colour flagged as marginal—advanced), on the day before transfer (as well as the day of transfer) to:

- Establish whether any chicks have emerged (in which case they can be removed from the list for transfer).

  Note: Time may not allow for these extra fence checks at the source colony. However, they can be very useful for determining what is happening at the burrows of the more advanced chicks before transfer day, so that it is clear whether any birds have emerged. Determining whether a chick has emerged requires careful inspection of both the burrow entrance and chick, and careful consideration—the less of this kind of decision-making that is required on transfer day the better, especially if there is time pressure.

- **Gadfly petrels only**—Record the fence status (of all burrows if time permits), to determine whether there have been any parental visits at night, which can assist with artificial meal planning. Ensure that these notes are provided to the feeders at the release site on transfer day, as this information is very useful in helping feeders to decide which chicks are overdue a meal and need feeding first, and which can wait several days (helping to avoid setting back the development of hungry chicks and causing oily regurgitations in already full chicks).

  Note:

  - Fence status can only be recorded for a given day if the fences were erected the day before inspection, i.e. it is known that the fence was knocked down on the previous night.
  - The use of fences can be valuable on grey-faced petrel projects; if a fence has not been knocked down between first handling and transfer day, and the chick only just met the weight criterion at first handling, then it is unlikely that the chick will be transferable because its base weight will be even lower (i.e. it has not received any parental meals since it was last handled). Thus, a second handling is not necessary.

5.6 Transcribing data

13. Ideally, data should be entered into an Excel spreadsheet, or at least onto paper forms (if no laptop is available) at the end of each search day. Aside from acting as a backup, this will allow the likely number of suitable chicks to be determined, which, in turn, will indicate how many new chicks might need to be found.

14. Data can be entered into an Excel spreadsheet that is similar to the chick selection form (section 10.2), and formulae can be used to calculate the minimum and maximum wing length of the chick on transfer day by adding the minimum and maximum growth (in mm) per day. It is important that the correct number of days from the day of measurement to transfer day is calculated, e.g. a chick that is measured on 6 March for transfer on 10 March has been processed four days before the transfer.

5.7 Searching for additional chicks

15. If more chicks need to be found to make up transfer numbers, ensure that old ground that was searched during the recce trip is not covered again. Since empty burrows that were found on the recce trip will not have been marked with flagging tape, it is preferable to move to a completely new area to commence further searches.

6. Guide to collecting chicks on the selection/collection/transfer trip

6.1 Preparations on the day before transfer

1. Compile a master list of chicks available for transfer (both suitable and marginal) and assign one person as coordinator—they will check chicks against a master list as they are collected, keep a running total on transfer day and ensure that boxes are kept in the shade at all times.
2. Divide the selected chicks amongst the collectors. It is easiest if people collect chicks from the areas that they previously checked (usually a team of people will have previously worked through one colony area together, checking their own blocks of burrows within that area before moving on to the next colony area).

   Note: Some teams may prefer to separate the tasks, so that some people handle the chicks, record data, make decisions and box up the chicks, while others collect full boxes and carry them to the transport pick-up area.
3. Prepare notebooks with a list of the chicks each person needs to relocate within each colony area. Notebooks should list whether chick wings are expected to be suitable in length or marginal (to help prioritise burrow searching on transfer day), burrow location, burrow type and any notes recorded about access (to help relocate chicks). Add in columns for weight, wing length and the transfer day decision (to transfer or leave).
4. Erect fences at all burrows containing chicks that are advanced, i.e. all chicks in the 'marginal—advanced' category, and even at burrows of some of the chicks with longer wings in the 'suitable' category, to help with establishing whether or not such a chick emerges during the night before transfer. (Refer to section 6.2—'Collecting chicks on transfer day').

   Note: For some species, the transfer criteria have been developed to take into account the fact that fences may not be an effective method of determining emergence, i.e. the criteria ensure that advanced chicks (with longer wings) are very heavy, with a low chance of emerging at the source colony. For example, fluttering shearwaters (*Puffinus gavia*) may be fed as often as every night right up to fledging, so it is impossible to determine whether a fence was knocked down by a parent or an emerging chick.
5. **Gadfly petrels only**—If time permits, erect fences at all other burrows containing suitable chicks, to assist with meal planning at the release site.

   Note: Visiting burrows on a second occasion prior to transfer day can be beneficial to some teams that might otherwise find it difficult to relocate all of the marked burrows on transfer day, e.g. if the burrows are in a forest habitat that can be difficult to orientate within and where areas of terrain look similar.
6. Prepare the transfer boxes (refer to the appropriate best practice document for the species). If transfer day timeframes are tight, these boxes can be positioned at accessible points around the colony on the day before transfer day, although they may need to be protected from the weather in some way.

   Note: Pre-assembling the boxes at the colony may not be appropriate if release sites have high biosecurity requirements, as insects may climb into the boxes during the night and be transported to the release site.

6.2 Collecting chicks on transfer day

Note: Ensure hands are clean and free of hand-cleaning products, sanitizers and sun-screen before handling any birds (to avoid spoiling plumage).

7. Relocate, weigh and measure each chick on the day of transfer, and double-check its suitability for transfer. Weights are extremely important and chicks must also fit the criterion for their wing length grouping to be suitable. Particular attention must be paid to the following chicks:
   - **All** marginal chicks (i.e. those that were predicted as being marginal—small or marginal—advanced when first processed).
   - Any chicks that have only been weighed and measured on a single occasion several days prior to the transfer, where it needs to be reconfirmed that they meet the transfer criteria.
   - Any chicks that are known or predicted to be close to the minimum weight criteria on transfer day. Chicks should not be included in the transfer cohort if they weigh less than the specified minimum weight for the wing length grouping on the morning of transfer day.
8. Carefully inspect burrow entrance for any signs of chick emergence, especially if the fence is down. A knocked down fence can indicate chick emergence and/or a parental visit; hence a careful inspection for down is required to determine whether the chick visited the surface. By contrast, an intact fence clearly shows that the chick did not emerge.

   Note:
   - Fences need to have been erected the day before transfer at all burrows containing chicks that are advanced, i.e. all chicks in the 'marginal—advanced' category, and even at the burrows of some of the chicks with longer wings in the 'suitable' category (refer to section 6.1—'Preparations on the day before transfer').
   - For some species, the transfer criteria have been developed to take into account the fact that fences may not be an effective method of determining emergences, i.e. the criteria ensure that advanced chicks (with longer wings) are very heavy with a low chance of emerging at the source colony. For example, fluttering shearwaters may be fed as often as every night right up to fledging, so it is impossible to determine whether a fence was knocked down by a parent or an emerging chick.
9. Chicks can be taken for transfer if one or both parents are present in the burrow on the morning of the transfer. However, if the chick does not have any down, make sure that the correct bird is taken by double-checking the band! Record the presence of adult(s) on the transfer box, as this can be useful for meal planning at the release site.

   Note: Parents are thought to stay over during the day if their chicks are refusing food; the adults stay because they need to offload or digest the food before going back to sea.
10. Carefully check each chick for any abnormalities or obvious signs of poor health. This includes checking both legs/feet, both wings and both eyes as a minimum. **If in any doubt about a chick's health or condition, do not transfer it.**
11. Avoid transferring a chick with badly soiled plumage (i.e. from regurgitation) because it will have a considerably reduced chance of post-fledging survival unless it can undergo prolonged water-proofing therapy before fledging, and may also have dropped below the transfer criteria.
12. **Gadfly petrels only**—Record the fence status on the morning of transfer—but **only** if fences were erected on the day before transfer day. Fence status can be written directly on the transfer box lid (above the relevant chick) using a permanent ink marker pen—FI (fence intact) or FD (fence down). This will then help the feeders at the release site to determine which chicks need to be fed on the first feeding day. Also record the natal burrow number on the box.

13. Ensure that the following data have been recorded:
    - On transfer boxes— The source colony burrow number; fence status; presence of any adults; and if the chick has regurgitated before being placed in the box (in which case, chicks would need to be reweighed to ensure that they still meet the minimum weight criterion for transfer).
    - In notebooks— The source colony burrow number; band number; chick weight and wing length, if taken; and whether transferred or returned to the burrow.
14. Leave all burrow markers in place at the source colony until after the chicks have been transferred to the release site. This is essential, in case a chick needs to be returned to its burrow for any reason (e.g. transport delays).
15. At all times, keep boxes well ventilated and as cool as possible by placing them in the shade and by leaving gaps between the boxes when placed together on the ground, to prevent chicks from overheating. Keep moving boxes as necessary as the position of the sun changes.

7. Guide to preparing chick food and feeding equipment

7.1 Tasks in the food preparation area on chick feeding day

7.1.1 Food preparation equipment

- Blender
- Knife (×2)
- Spatula
- Cooled boiled (> 3 minutes) water
- Small measuring jug (to 10 mL)
- Tinned sardines
- Small pestle and mortar (for crushing seabird vitamin-mineral tablets)
- Food containers

7.1.2 Food ingredients

- One 106 g tin of Brunswick™ sardines in soya oil (include oil contents).

  Note: sardine cans usually contain sardines (89%), soya oil (10%) and salt (< 1%)—always check the content proportions before purchase and seek specialist advice if the proportions have changed. PAMS™ brand (identical content) has also been used successfully on a single transfer operation to date.
- One-third of a Mazuri® Vita-zu bird tablet (vitamin/mineral supplement: Small 5M25)—always confirm the product number, dosage and expiry (best before) date with the supplier for each batch.
- 50 mL cooled boiled (> 3 minutes) water—note that this is the standard amount of water added, but the dilution varies with species, so the appropriate best practice document for the species must be consulted.
- **Large gadfly petrels only—**Supplementary oils (refer to the grey-faced petrel best practice document).

7.1.3 Packing sterilised equipment

1. Wash hands (with antibacterial soap).
2. Pack the dry, sterilised equipment required for day in a bucket (rinse bath containers, jars for chlorhexidine, lids of food-warming baths (if relevant), box of syringes / crop tubes and stirring spoons or spatulas), along with clean, dry holding bags and towels.

7.1.4 Making antibacterial solution

3. Make up **fresh** antibacterial solution (e.g. Milton) in a square bucket (1 tablet to 2 L cold water; 6 tablets to 12 L cold water). A minimum of 10 L is usually required (to cover an immersed blender jug).

7.1.5 Making food

4. Place required number of Mazuri® tablets (or portions of tablets) in mortar and crush with pestle to as fine a powder as possible.

   Note: Avoid getting the tablets moist this will make them difficult to crush effectively. The tablets do not dissolve, so crushing to a fine dust allows the vitamins to be evenly distributed in the mixture.

5. For three tins of fish—Place 150 mL cooled boiled (> 3 minutes) water in a blender with one tin of fish (chop fish up in the tin) and liquidise until runny (at least 30 seconds). Add the second tin of chopped fish and blend until runny. Add vitamin powder through the hole in lid while blender is running at low speed. Then add the third tin of chopped fish and blend until smooth.

   Note:

   - To prevent strain on the blender, a maximum of four tins of fish (with 200 mL water) can be processed in each batch. Three tins (with 150 mL water) is just enough mix to cover Sunbeam™ Multiblender Platinum blender blades.
   - Addition of the vitamin powder via the lid while the blender is slowly running ensures that all of the powder is incorporated and prevents it from being thrown up onto the lid of the blender when the blender starts up.

6. Pour the mix into containers immediately after the blender stops (so that the food does not settle). Distribute between several pottles to allow for any spillages at the feeding site.

   Note:

   - Four tins of food make up c. 700 mL of mix, which fills a 1 L pottle to approx. ¾ full. This amount of mix can feed approx. ten fluttering shearwaters or up to seven grey-faced petrel chicks. Two smaller 500-mL pottles can be filled approx. ¾ full, each of which would feed approx. ten small gadfly petrel chicks.
   - It pays to make up an extra tin of the mix each day to allow for spoiled food. Pottles should be small enough either to fit into a food thermos flask, which is used as a hot-water bath for smaller species, or to sit in a larger water bath (e.g. a chilly bin) without tipping over for larger species.

7. **Move the jug to the second blender motor for the next batch** (to prevent strain on the first motor).

8. Place coloured lids on the pottles of food and chill as follows:

   - White and/or blue lids, for example, on the first pottles of food made; if possible, put in fridge or briefly in freezer to cool down (up to 30 minutes only, then move to fridge if necessary). These pottles will be the last to be used at the colony, so need to be more chilled through the day.
   - Red lids, for example, on 1–2 pottles. Leave these at room temperature (for the first feeding session).

9. Turn the blenders off at the plug on wall.

10. Place food pottles in a chilly bin with ice packs. The food must be kept cool at the colony site (to prevent contamination) and then warmed just before use.

    Note: One or more of these ice packs may also be required to keep any dead chicks that may be found during the roll-call cool.

7.1.6 Clean-up

11. Remove blender blades and rinse out blender, etc. before doing a thorough wash (with the petrel dishwashing brush) in very hot, very soapy water to remove all oil. Rinse off the detergent before placing equipment in a bucket of antibacterial solution for the day (minimum soak period = 2 hours).

    Note: Some blender blades need to be placed upturned in a small pottle of sterilising solution so blades are immersed but the base is kept dry (i.e. where the base is prone to rust).

12. Put the dishwashing brush and cloth into a separate container of sterilising solution after thoroughly cleaning the brush with extra dishwashing liquid and clean hot water.

13. Wash out the sardine tins in hot, soapy water (use rubber gloves) for disposal/recycling.

Note: Avoid using the petrel brush as this will make it very greasy.

14. Wipe down blender bases and bench with a cloth (which has been soaked in antibacterial solution).

7.1.7 Rinsing weigh bags

15. Wash and rinse (several times) any weigh bags that have been soaking in Napisan® overnight, and hang out to dry.

7.1.8 Boiling water

16. Fill metal thermos flasks with boiled water (standard boil, for food-warming baths). Suggested amounts are:
    - Three 2 L flasks for species with meal sizes < 50 mL; or
    - Six or more flasks for species with larger meal sizes.
17. Boil enough water (for > 3 minutes) to fill the boiled water containers and to set some aside (in a clean/sterilised food container) for use in food preparation on the next feeding day.

    Note: Alternatively, this can be done at the end of the feeding day.

7.2 Checklist of equipment for chick feeding day

The following list specifies the equipment that is needed for one feeding team (i.e. the number of items per team).

7.2.1 Transported to the colony site each day

1. In the chilly bin place:
    - Food pottles—leave one or two pottles out at room temperature for the first feeding round
    - Several ice packs (chilly slicks) to keep food cool at the colony site (to prevent contamination)
2. In plastic buckets place:
    - A plastic box (with lid) containing:
        - Two 30 mL or 50 mL syringes (for species being fed meals up to 50 mL) or four 50 mL syringes (for larger species being fed 50–100 mL meals)
        - Two crop tubes (for smaller species) or four crop tubes (for larger species) (or one to two catheter tubes per chick if using these)
        - Clean teaspoons (small pottles) or spatulas (large pottles) for each pottle
        - Two 100 mL jars (to hold chlorhexidine solution for sterilising crop tubes)
    - Two rectangular rinse baths
    - One tray or lid to rest clean syringes on (can use the lids of rinse baths)
    - Lids to the food-warming baths (e.g. food flask cups for small species), or lid to a yoghurt maker (larger species)
    - The 'room' temperature food pottles (for first feeding round)
    - Containers of boiled (> 3 minutes) water (for rinsing)
    - Thermos flasks of hot water (for food-warming baths)
    - Waterproof notebooks and pencil (for roll-call / fence status rounds)
    - Clean weigh bags
    - Clean hand towels to rest chicks on

- Any other supplies for restocking, e.g. tissues, rubbish bags, paper towels, hand-washing water, newspaper for carry boxes

Note: Buckets can become rubbish bins and waste (slops) buckets during chick feeding.

3. Remember data folders.

7.2.2 Stored at the colony site (temporary shed)

4. Items that can be stored at the colony site and replenished as required include:
    - Hand-washing water and antibacterial soap
    - Antibacterial hand wipes and surface wipes
    - Food-warming baths (e.g. food flasks for smaller species, yoghurt makers for larger species)
    - Bottle of chlorhexidine concentrate plus measuring jug, and bottle for diluted mix
    - Device to stabilise sterilising solution jar(s)
    - Castor oil (to lubricate syringes)
    - Spare sterilised crop tubes
    - Fine-tipped permanent ink marker pens (to mark syringe barrels)
    - Food temperature thermometer(s), if required
    - Carry boxes (colour-coded) to carry chicks between burrows and shed
    - Newspaper for lining tool boxes
    - Coloured markers to mark burrows during collection of chicks
    - Pesola scales or digital tabletop scales (plus spare batteries)
    - Weighing boxes (for birds on scales)—a larger, high-sided box can also be useful for housing the scales in the wind
    - Wing rule
    - Spare weigh bags and hand towels
    - Spare tissues and paper towels
    - Spray bottles (for dampening plumage if needed)
    - Chick first-aid kit (with oral fluids, e.g. Hartmann's™ solution; refer to section 2.2—'Release site chick feeding, housing and monitoring')
    - List of chicks in band order
    - Pencils, erasers and pencil sharpeners

8. Guide to chick feeding, measuring and monitoring

8.1 Preparing syringes/crop tubes

The following methods are for the preparation of Plexi-vet™ syringes and custom-made Teflon crop tubes. Syringes / crop tubes can be prepared while other equipment is being set up and chick roll-calls are being undertaken (see below).

1. Wash hands (with antibacterial soap).
2. Assemble the syringes and crop tubes (hand-tight only).
3. Lubricate each plunger with a smear of castor oil (on clean tissue) and apply castor oil to screw thread of crop tube to make an airtight seal.

   Note: Use a clean tissue to hold the crop tubes to keep them clean and ready for use, i.e. avoid touching the tubes with your hands.
4. Accurately indicate a scale on the unmarked side of each syringe barrel using a fine-tipped permanent ink marker pen if required. Make clear marks around the barrel at the 5-mL line using the plunger as a guide (this will be useful during feeding for the sterilising process; see section 8.7—'Food hygiene and temperature control').
5. Lay the assembled syringes / crop tubes on clean (sterilised) trays ready for feeding.
6. Ensure the feeders double-check the tightness of tubes and numbering on the syringe barrels before they start feeding.

8.2 Setting up other equipment

7. Fill two rinse baths with boiled (> 3 minutes) water.

   Note: If there is a shortage of boiled water, the first rinse bath can contain tap water; however, the final rinse must be in sterilised water.
8. Set up a box of tissues, and spread a clean, dry hand towel on top of the feeding bench (between feeder and holder) for the birds to sit on.
9. Place the glass crop tube sterilising jars (see section 8.3—'Making the crop tube sterilising solution') in the stabiliser.
10. Weigh the holding bags to ensure that all bags used in one session are of the same weight and match the weight of the tare bag used to tare the tabletop electronic scales (with the bird weigh box on the scales), **or** check that all Pesola scales are calibrated.
11. Ensure carry boxes are lined with layers of newspaper.

8.3 Making the crop tube sterilising solution

12 Make up a sterilising solution of Microshields™ chlorhexidine (5%) (this is a dark pink runny liquid) with water (preferably boiled > 3 minutes) in a separate bottle:

   - 1 part chlorhexidine : 19 parts water; or
   - 10 mL chlorhexidine : 190 mL water

   Allow for c. 150 mL per syringe/tube setup; i.e. 100 mL for the jar that sterilises the outside of the crop tubes and up to 50 mL to draw up into the syringe barrel at disinfection breaks (see later). The solution is best made up on a daily basis, as chlorhexidine loses effectiveness if left more than a day or two in solution.

Note: 200 mL of solution is adequate for one feeding team for a smaller species (e.g. Cook's and Pycroft's petrels), whereas over 1 L may be needed for two teams feeding a large species (e.g. grey-faced petrels). For example, a total of 1300 mL of sterilising solution (65 mL chlorhexidine) was made on a fluttering shearwater chick feeding day whenever there were two disinfection intervals during the day and eight syringes in use.

13. Fill the crop tube sterilising jars with chlorhexidine solution (one jar for two syringes; two jars for four syringes; four jars for eight syringes) to the depth of the crop tube. Set aside the remaining pink solution to use for flushing syringes during the feeding interval—refer to section 8.8—'Disinfection interval(s)'.

8.4 Warming food

Food can be warmed c. 10 minutes before feeding is due to start, i.e. when the chick roll-call is close to completion.

14. Place the first food container in a food-warming bath containing hot water to warm up:
    - When using a food flask as a warming bath for small food volumes (e.g. small gadfly petrels), the pottle can be just resting in the water—but care must be taken to ensure that the flask water does not flow up, over and into the food.
    - When using yoghurt maker for larger food volumes (e.g. fluttering shearwaters), fill the yoghurt maker up to the top of the red 'shelf'. Water cannot overflow into the food pottles.
    - For larger species (e.g. grey-faced petrels), containers of food will need to be warmed in a small insulated bin or bag (e.g. chilly bin)
15. Remove the pottle lids, insert a clean stirring spoon or spatula, and replace the food flask or yoghurt maker lid.
16. Use a spoon or spatula to stir regularly (obtain an even temperature). When testing temperature on wrist, the mixture should be just warm, e.g. 33°C (a cold mix, e.g. < 30°C, may be rejected by the chick; while a hot mix, e.g. > 35°C, may damage the chick's internal tissues). If a thermometer is used, it must be appropriately immersed in the food for at least 10 seconds to allow for adjustment.
17. When the food is warm enough:
    - Food flask warming-bath system—Transfer the pottle to a second empty food flask and restore the flask cup on top. This flask, which does not contain hot water, will keep the food at a steady temperature until it is finished. However, if feeding is slow, the pottle may need to be returned to the flask with hot water when the food level is low (and cooler).
    - Yoghurt maker warming-bath system—Remove the pottle and place it on bench, remembering to restore yoghurt maker lid to keep water hot. The pottle will need to be returned to the yoghurt maker after several chicks have been fed to warm the food again, as it will cool on the bench.

8.5 Carrying out the chick roll call

The chick roll-call includes recording burrow entrance fence status (indicates emergence behaviour); checking for the presence of chicks and the wellbeing of all birds before commencing any feeding of the chicks; and checking vacant burrows and searching for missing chicks, if relevant.

Note: Ensure hands are clean and free of hand-cleaning products, sanitisers and sun-screen before handling any birds (to avoid spoiling plumage).

18. Complete rounds of all occupied burrows to record fence status (indicator of emergence behaviour) and check on the welfare of **all** birds before commencing feeds.
19. Visit each burrow in numerical order (to ensure all are checked) and:
    - Record the stick fence status (D = fence down; I = fence intact; PD = partially down) or the presence of a rock (R = rock).
    - Open burrow lid and check whether the chick is present. Also check the chick's welfare, i.e. that it is bright, alert and responsive (BAR).
    - Check for signs of regurgitation or abnormal excrement in the burrow, and for any signs of digging in blockaded burrows.
    - If burrow is empty, feel along the entire length of pipe from both ends (in case chick is in the tunnel).
    - Record presence or absence of the chick (✓ or ✗) in a waterproof **fences** book.

    Note: Do not worry about erecting stick fences at the burrow entrances at this stage. Fences are best restored at the end of the day after all chick processing has been completed.
20. Search pipes for any missing chicks (two chicks can be found in one burrow) by feeling along the entire inside length of every pipe for which the fence has been recorded as down.
21. If chicks are found away from their 'home' burrows, refer to the chick band list so that they can be returned to the correct burrow before feeding commences. Ensure that these burrow swaps are noted.
22. If time permits, check all burrows that do not normally contain chicks (even if the stick fences are intact) to ensure that other chicks have not been accidentally returned to the wrong burrows on the previous day. Such chicks might not otherwise be found and may be recorded as departed from their own burrows, yet still require feeding. For this reason, it is important not to install blockades at entrances of vacant (unoccupied) burrows to be sure any chicks accidentally put in wrong burrows can emerge/depart; spare, vacant burrows also provide additional places for wandering birds to occupy, reducing the incidence of birds disappearing and missing feeds.

    Note: Burrows that have housed chicks that have died should be left blockaded to prevent other chicks from using them.
23. Update chick feeding record sheets with the fence status found at each burrow.

    Note: If a folder system is used to manage data, data sheets of birds that have departed can be moved to the back of the folder so that they do not get filled out by mistake during the feeding process.

8.6 Processing chicks

Process chicks in the following order:

- Extract from burrow
- Check band if necessary
- Weigh (to obtain pre-feed or base weight)
- Measure wing length (right wing) if wing measuring day
- Carry out any other checks (e.g. physical examination, down coverage estimates)
- Feed (recording amount delivered in mL; no post-feed weight required)
- Return to burrow

8.6.1 Extracting chicks from burrows

Note: Ensure hands are clean and free of hand-cleaning products, sanitizers and sun-screen before handling any birds (to avoid spoiling plumage).

24. **Small gadfly petrels only**—Refer to the list of chicks (burrow numbers) that need to be fed that day. Write the collector's initials next to the chick to be collected.
25. Collect chicks in weigh bag placed inside a carry box, taking care not to bend or trap long tail and wing feathers when bringing chicks out through the top of the burrow and when closing the lid of the carry box.

    Note: The loss of tail or wing feathers can considerably compromise a chick during the emergence period and at fledging.
26. Replace burrow lid to keep chamber dry and cool. Mark the empty burrow when the chick has been removed (e.g. using a coloured marker matching the colour mark on the carry box) to ensure that the chick is returned to the same burrow.
27. If requested, check nest for signs of regurgitation and faeces present and normal (dark brown gritty faeces with white fluid urates, usually seen on the chamber walls).

8.6.2 Weighing and measuring chicks

The weights and wing lengths of chicks are primarily recorded to help with their management at the release site, i.e. to determine meal sizes and the dates of burrow blockade removal. The most recent weights and wing lengths are also used to assess whether chicks are likely to have fledged when they are no longer found in their burrow.

28. Weigh chicks on every feeding day (whether daily, alternate days or less frequently), **and** daily or on alternate days from the time that the chick is expected to depart until the chick has fledged (to obtain fledging weight).
29. Weigh birds as follows:
    - Tabletop scales— Place the weigh box and an empty weigh bag on scales and zero (tare) them. Remove weigh bag. Wrap bird up in its weigh bag and place in weigh box to obtain a nett weight. Note that this method relies on all weigh bags used on any one day weighing the same amount.
    - Pesola scales—Weigh the bird over a surface (to prevent injury if bag falls from scales).

    Note: Replace weigh bags as soon as they become soiled.
30. Wing measurements do not need to be recorded any more frequently than every third day for any species (usually every 3–5 days for younger chicks to assist with planning meals and blockade removal), **and** then daily or on alternate days when the chick is expected to depart (for fledging wing length). Wings can stop being measured once three equal measurements have been obtained (i.e. the wings have stopped growing).
31. Keep birds in bags during wing measurement (to keep them calm), removing only the right wing to measure—ensure it is straightened and flattened to record maximum wing chord.

8.6.3 Feeding chicks

Feeding methods include: loading syringes; holding chicks; introducing crop tubes, delivering food safely with appropriate breaks and withdrawing crop tubes; dealing with overflow and regurgitation; cleaning the chick after feeding; and recording the feeding outcome (meal size successfully delivered) and chick behaviour.

Meal sizes will be pre-planned and should be written on each chick's feeding record sheet prior to each feeding session, so that syringes can be loaded with the appropriate amount of food.

32. To feed a meal of 30 mL using a 30-mL syringe:

- Fully load the syringe to an excess of 30 mL (i.e. to 35 mL, with the plunger pulled out to its maximum), ensuring that all air bubbles are removed. (If air bubbles continue to enter while drawing up food, either the tube or the top of the syringe may need to be tightened.) The excess allows for 5 mL to be left in the bottom of the syringe after delivery of 30 mL to the chick, which is important for the sterilising process (see below).
- Wipe the crop tube with a clean tissue to remove any residue food before inserting it into the chick's throat.

Note: If feeding a 35-mL meal, **two** syringe loads (35 mL total in first syringe and 10 mL total in second syringe) are required. Remember that a used crop tube cannot be put back into the food pottle.

33. To feed a meal of > 50 mL using 50-mL syringes, use the following volumes in **two** syringes:
    - 50 mL = 2 × 30 mL
    - 60 mL = 2 × 35 mL
    - 70 mL = 1 × 45 mL and 1 × 35 mL
    - 80 mL = 2 × 45 mL

    Note: It is recommended that a consistent method is used to fill the syringes for each meal, to ensure that the correct total amount is loaded and delivered, and that both syringes are pre-loaded before commencing the feed.

34. During feeding:
    - The handler holds the chick firmly on a surface (with a towel) with a loose hand grip—the chick must not be tightly gripped or it will not feed properly; the crop area in particular needs to be unrestricted.
    - The feeder holds open the bill (mainly grasping the upper bill), stretching the head and neck out (at an angle of c. 30–40° from the horizontal).
    - With the other hand holding the syringe, the feeder inserts the crop tube to the back and side of the throat (to keep the airway clear).
    - It should take at least 30–60 seconds to deliver a syringe of food, with at least one rest approximately half way through the syringe load to check for any signs of meal rejection.
    - Food delivery stops at the pre-determined amount or earlier if there are any signs of food coming back up the throat.
    - The bill is **immediately** released as the crop tube is withdrawn, so that if there is any regurgitation the food can be projected clear of the plumage and there is a reduced risk of the bird aspirating the food.
    - There should be at least 5 mL of food remaining in the syringe, which is important for the sterilising process (see section 8.7—'Food hygiene and temperature control').

35. In the event that a chick's meal overflows:
    - Withdraw the crop tube immediately and **let go of the bill** (to allow the chick to project any regurgitant).
    - Wipe the bill clean.
    - Insert the crop tube a shorter distance down the throat (but still past the air pipe).
    - Reattempt delivery of a smaller volume of food.
    - If overflow occurs a second time, stop feeding, unless it is thought that the chick definitely needs the meal (in which case continue to deliver small volumes, withdrawing the crop tube after each, i.e. deliver food in short, fast bursts).

    Note: Regurgitation can be distinguished from 'overflow' (where a small proportion of the meal is lost towards the end of food delivery) as there will be a clear rejection of a substantial proportion of the meal; these rejections are usually made by chicks that are ready to accept smaller meal sizes.

36. After feeding, the chick may need to be cleaned with a soft tissue so that there is no food on the bill or plumage. Particular attention needs to be paid to the base of the bill, where food can build up and form a crust if not cleaned away.

37. In the event that a chick regurgitates all or part of its meal:
    - Wipe the bill clean.
    - Place the bird as quickly as possible in the dark carry box to reduce the risk of further regurgitation.
    - Estimate the amount of food rejected—this can be achieved by mopping up the regurgitant with tissues and weighing (subtracting weight of equivalent number of tissues).
    - Clearly record event in notebook as this will affect the planning of the subsequent meal size.

    Note: Avoid reweighing the chick as this can cause further regurgitation (and soiling of plumage) in the bag.

38. Record the following in the notebook:
    - The amount of food actually taken by chick—read the syringe scale, e.g. 17 mL (of a 20 mL feed). No post-feed weight is required as 10 mL of food is approximately equivalent to 10 g in weight.
    - Any details regarding food delivery, e.g. regurgitation, overflow, appears full, difficult feeder requiring plenty of breaks, resists food, good feed, etc.

    Note: These details are important and influence the planning of subsequent meal sizes.

8.6.4 Returning chicks to burrows

39. Carry chick back to the burrow inside the carry box—**not** in a weigh bag. Return chick directly to chamber, facing back wall, and restore lid. Remove marker.

40. Change newspaper in carry box if soiled, before collecting the next chick.

    Note: Antibacterial surface wipes can be used to clean carry boxes if they become soiled.

8.7 Food hygiene and temperature control

The following methods include monitoring and maintaining an appropriate food temperature and consistency throughout the feeding session; and disinfecting and rinsing the crop tubes between chicks.

41. After feeding each chick:
    - Wipe the crop tube thoroughly with a tissue and place it upright in a jar of sterilising solution for an absolute minimum of two minutes sterilising time—a longer sterilising time is preferable. There must be **no** organic residue on the outside of the tube for the disinfectant to be effective.
    - After sterilisation, remove syringe/tube and eject the food remaining in the syringe (≥ 5 mL) into the waste bucket—**this is important, as it removes any disinfecting solution that may have soaked into the food in the tube**.
    - Rinse the outside (entire length) of the tube using two rinse baths. The syringe/tube is now ready to draw up more fresh food (there should be no air bubbles present).

    Note: The delicate nature of the screw fitting of the tube into the syringe means that the tube cannot frequently be detached (or the fitting will be damaged); thus, the above sterilising process has been designed for this particular equipment.

42. Keep monitoring the food temperature regularly (aiming for c. 33°C if using a thermometer) and stir with a spoon before drawing up food (as the thick part of the mix can settle). Add

more hot water to the water bath if temperature drops, or return the pottle to a food-warming bath if it was earlier removed.

Note: When the current batch of food is starting to be fed to birds, the next pottle of food can be removed from the chilly bin and either left to warm up at ambient temperature or placed in another food-warming bath for a slow heat.

43. Use a clean (sterilised) teaspoon to stir the next pottle of food.

8.8 Disinfection interval(s)

On a full feeding day, the syringe barrels must be rinsed out and disinfected (filled with sterilising solution) several times during the day to coincide with feeding intervals / tea breaks, or between food pottles. This should be done **no later than two hours** into a feeding session, i.e. do not allow used syringes to be used for more than two hours without being fully disinfected.

44. Eject the 5 mL of food from the syringe into a waste bucket.
45. Rinse the used syringe out with hot water (e.g. from a food-warming bath)—this water can be squirted into used/empty food pottles to assist with cleaning them.
46. Draw up **fresh** chlorhexidine solution to the top of the barrel and stand in the existing jars of chlorhexidine solution for 30 minutes (minimum).
47. Empty rinsed pottles into waste bucket, and empty waste bucket away from the feeding area and burrow site. All of the 'used' water in the feeding shed from the rinse baths and food-warmers can be used to rinse out the waste bucket.
48. Clean down the benches and food-warmers using antibacterial surface wipes.
49. Remove new food pottles from the chilly bin to bring to room temperature (and then move them to the food-warming baths about 10 minutes before feeding).
50. Remove syringes from the jars of chlorhexidine and discard the old chlorhexidine. Rinse out the jars with water and place them back into the stabilisers.
51. Expel the freshest chlorhexidine solution from the syringes into the jars.
52. Thoroughly rinse syringes with clean water before using them again—at least three full flushes with tap water followed by two flushes with boiled water. Place the cleaned syringes on a tray ready for use.
53. Replace the rinse baths with tap and boiled water.
54. Stir the food and test its temperature ready for the next feeding session.

8.9 Clean-up after feeding

Clean-up methods include checking the records to ensure that all the right chicks have been fed before discarding food and packing up; rinsing and packing the feeding equipment for transport from the burrow site; and cleaning the feed station.

55. At end of the feeding session, check all of the data sheets to ensure that all birds have been fed, before discarding food and equipment.
56. Eject any remaining food from the syringes/tubes and flush them out with water (use hot water if any left in flasks as this is more effective at removing fish residue from inside the tubes). Place the syringes/tubes in the long plastic box for transport—they need to remain assembled for the washing-up process.

57. Soak up some left-over sterilising solution on to a paper towel and wipe out the food flasks and screw-top lids. These can stay at the feeding site, but the food-warming bath lids that were used to cover the food will need to be taken away for washing.
58. Stack all used items, soiled weigh bags and towels, waste food, etc. into chilly bin and buckets to take them down for washing.
59. Wipe down all work surfaces with kitchen towels and disinfectant spray (or left over sterilising solution or disinfectant surface wipes).
60. Throw out any remaining sterilising solution (this is made up daily and should be used within two days or it begins to crystallise).

8.10 Burrow entrance blockades and stick fences

61. After all feeding is complete, remove any outstanding blockades from the relevant burrows (if scheduled based on weight and wing length data, and down coverage) to allow chick emergence (refer to the appropriate best practice document for the species).

    Note: The removal of blockades **after** the feeding session may reduce the risk of the occasional chick wandering from its burrow during the daytime.
62. Check that **all** stick fences at burrow entrances without blockades are restored. Two to three thin, straight sticks are sufficient—lightly placed in the soil at the entrance so that they do not barricade the chicks in (or out!).
63. Check that **all** chamber lids are firmly in place (to ensure that the chambers are light-proof and waterproof).

8.11 Transcribing data

64. Ensure that data are transcribed onto computer spreadsheets on a daily basis (or at least on alternate days), in case field data are lost or damaged.
65. Prepare notebooks / data sheets for the following day, with planned meal sizes for all chicks.

9. Guide to cleaning equipment after feeding chicks

9.1 Waste food

1. Discard any surplus sardine mixture away from the burrow site, as it could be a source of contamination to the site, could interfere with the scent trails of chicks and spoil plumage if encountered by chicks exploring on the surface.

9.2 Hot water flasks and boiled water containers

2. Boil (> 3 minutes) enough water to fill the boiled water containers and to set some aside (in a clean/sterilised container) for use in food preparation on the next feeding day.

9.3 Weigh bags and towels

3. Shake out the weigh bags (pillowcases), turn them inside out (to allow faeces, etc. to soak off) and soak in Napisan® for at least 2 hours (using a half level lid of Napisan® to 5 L water), but preferably overnight. Bags should then be rinsed several times and hung out to dry the following morning.

9.4 Washing-up

4. Wash hands (with antibacterial soap).
5. Remove the food preparation equipment (blender jugs, etc.) that has been soaking during the day from the antibacterial solution and allow it to air dry in drainage trays or on a bench that has been wiped with a cloth soaked in sterilising solution.

   Note: The antibacterial chemicals should evaporate off the equipment so theoretically there is no need to rinse this off. However, if the items are still wet in the morning, it is probably best to rinse the antibacterial solution off the syringe barrels, crop tubes and food pottles using boiled (> 3 minutes) water to prevent it from tainting the food.
6. Retain the antibacterial solution for soaking the day's equipment after washing (as it is effective for up to 24 hours). Make up additional solution as required, e.g. an additional 15–20 L may be needed.
7. Empty chilly bin and return used ice packs to the freezer.
8. Wash the equipment as follows:
   - Rinse all equipment with hot (if possible) water to remove bulk of food before doing a thorough wash (using the petrel dishwashing brush) in **very hot, very soapy** water to remove all oil.
   - Vigorously plunge hot, soapy water through the crop tube and syringe several times while the syringe and tube are assembled, then remove tube and plunger for more thorough washing (put dishwashing liquid in the syringe barrel and use a rolled up sponge to remove the oil residue from all inside surfaces).
   - Use a pipe cleaner to clean the inside of the crop tubes.
   - Thoroughly brush all inside and outside surfaces of each food pottle, lid, glass sterilising solution jar and rinse bath. (Hold these up to the light to check that all oil residue has been removed.)

Note: Wear rubber gloves, as the water needs to be extremely hot to effectively clean the equipment. The bottle brush can only be used to clean the disinfecting jars as it tends to scratch all other equipment (e.g. syringe barrels, pottles).

9. Rinse off the detergent before placing the equipment in a bucket of antibacterial solution (minimum soak period two hours).
10. The dishwashing brush, bottle brush, sponges and cloth can be placed in sterilising solution in a separate container after thoroughly cleaning the brushes with extra dishwashing liquid and clean, hot water.
11. Wash out the chilly bins and equipment buckets, and then wipe them out using a cloth that has been soaked in antibacterial solution.
12. Once the equipment has been soaking in the sterilising solution for at least two hours, allow it to air dry overnight by removing it from the solution later in the evening, shaking off any excess liquid and placing the items on a clean bench (wiped over with a cloth that has been soaked in antibacterial solution).
13. Before discarding the antibacterial solution (as it is recommended that this be changed every 24 hours and so fresh antibacterial solution is made on the next feeding day), use some of the 'used' antibacterial solution to replace the solution in the tub for brushes and dishcloths.

10. Record-keeping / data forms

10.1 Marked burrows inspected on the recce trip for [species] chicks on [source colony location] in [month/year]

These data can also be entered into Excel worksheets if a laptop can be taken on a recce trip.

Key:	Adult only	Adult—no egg or chick present
	Adult/egg	Late incubating adult
	Adult/chick	Adult brooding chick
	Adult/unknown	Adult—presence of egg or chick not known
	Chick	Chick alone
	Dead chick	Chick corpse
	Egg	Abandoned egg
	Empty	No bird or egg

BURROW NUMBER	DATE	BURROW OCCUPANT (REFER TO KEY)	CHICK WING LENGTH (mm)	STATE OF WING IF NO FEATHERS (DOWN OR PINS)	BURROW TYPE (NATURAL OR BOX)	STUDY HOLE		CHAMBER ACCESS VIA ENTRANCE	COMMENTS
						PLYWOOD COVER	ROCK COVER		

10.2 [Species] chick selection form

This example of a chick selection form shows the sort of information that needs to be collected in the field to help team leaders to decide which chicks can be selected for transfer. Data collected by search teams can be presented in a form such as this, and also entered directly from notebooks into Excel worksheets if a laptop can be taken on the collection trip. If using Excel, formulae can be entered into the relevant columns to ensure that the calculations are correct.

CHATHAM PETREL CHICK SELECTION FORM

Planned transfer date: 21/04/2011

Date: 18/04/2011 Location within colony: Woolshed Bush

No. days to transfer (from first measure/weigh): 3

Burrow	Chick band (D-)	Weight (g) on given date			Wing (mm) on given date			First *minimum* (+2 mm/day) wing estimate for transfer day + 6 mm	First *maximum* (+4 mm/day) wing estimate for transfer day + 12 mm	Second wing estimate for transfer day + 3 mm	Suitability for transfer (√, X, ?)		Fence status on given date (I = intact; D = down)		Notes
		18/4	20/4	21/4	18/4	20/4	21/4				Prob	Def	20/4	21/4	
56	177628	290	307	300	184			190	196		√	√	D	I	Weigh on transfer day
213	177630	315		340	167		176	173	179		?	X	I	D	Measure " " "
194	177631	370		345	170		181	176	182		?	√	I	I	" " " "
193	177633	350			160			166	172		X		D	D	Wing too short
135a	177694	407	390		212	218		~~218~~	~~224~~	221	?	√	I	I	Marginal advanced
150	177696	300	280		219	225		225	231		?	X	I		Too advanced
257	177653		320	365		191		–	–	194	√	√	–	D	Weigh on transfer day
132	177655		310	295		197	200	–	–	200	?	X	–	I	" " " "

10.3 [Species] chick feeding and measurement record form

This data form can be adapted for printing. The data should also be entered into an Excel workbook, with one worksheet per chick/burrow. The sheet below is designed for use where digital tabletop scales are used (i.e. all weights entered are nett weights). If using Pesola scales, two additional columns need to be added: gross (pre-feed) weight; and bag weight.

[SPECIES] TRANSFER [LOCATION AND YEAR]						BAND				BURROW
Date	Meal size plan	Nett pre-feed weight (g)	Actual fed (mL)	Wing length (mm)	Down cover %	Good feed ✓/X	Over-flow (OF) or regurg-itant (R) (mL)	Full / resist-ing	Fence status D/ PD/I*	Notes
Notes										
10 Jan										
11 Jan										
12 Jan										
13 Jan										
14 Jan										
15 Jan										
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17 Jan										
18 Jan										
19 Jan										
20 Jan										
21 Jan										
22 Jan										
23 Jan										
24 Jan										
25 Jan										
Additional notes:										

* Fence status: D = down; PD = partially down; I = intact.

10.4 Calendar to schedule feeding of gadfly petrel chicks

Feeders can write approximate meal size (B= big meal / M= medium meal / S= small meal) in the boxes, or specify the volume that a chick needs to be fed. This helps to work out the total volume of food that needs to be made each feeding day.

BURROW	MARCH 2013																						APRIL 2013								
	10	11	12	13	14	15	16	17	18	19	20	21	22	23	24	25	26	27	28	29	30	31	1	2	3	4	5	6	7	8	9
1																															
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10.5 [Location] burrow monitoring form

These data are best entered directly from notebooks into Excel worksheets. For every date that the burrows are checked, there should be an entry to show that the burrow was inspected.

Examples of minimum information recorded for each burrow:

FI (fence intact)	Fresh nest material	Digging outside burrow
FD (fence down)	Old nest material	Digging inside burrow
Feathers at entrance	Excrement outside burrow	No sign
Feathers in chamber	Excrement inside burrow	Entrance blocked

	Observer initials:	PJ	PJ	PJ	
Burrow number	Date:	1 Oct. 2012	8 Oct. 2012	15 Oct. 2012	Etc.
1		FD—fresh nest material	FD—fresh nest material	FD—bird present on egg	
2		FI	FI	FD—feather at entrance	
3		FI	FI	FI	
4		FI	FI	FI	
5		FI	FD—no sign	FI	
6		FD—old nest and scrape	FI	FD—new nest material	
Etc.					

11. References

Gummer, H.; Taylor, G.; Collen, R. 2014a: Best practice techniques for the translocation of Chatham petrels (*Pterodroma axillaris*), Cook's petrels (*P. cookii*) and Pycroft's petrels (*P. pycrofti*). Department of Conservation, Wellington. 83 p .

Gummer, H.; Taylor, G.; Collen, R.; Ward-Smith, T.; Mitchell, C. 2014b: Best practice techniques for the translocation of grey-faced petrels (*Pterodroma macroptera gouldi*). Department of Conservation, Wellington. 94 p.

Appendix 1

Details of the contributers to and reviewers of this document

This document was compiled by experts who have developed methods during many burrow-nesting seabird translocations:

- Helen Gummer—Seabird translocation specialist employed by the Department of Conservation (DOC) and community groups
- Graeme Taylor—Seabird translocation specialist; Principal Science Advisor, Transformation and Innovation Unit, Science and Capability Group, DOC; and former Manager of the National Banding Office, DOC
- Rose Collen—Translocation specialist with experience in shorebird and seabird translocations, and contractor to DOC

This document was reviewed by experts with translocation experience, or with translocation process experience:

- Pam Cromarty—former Technical Advisor Systems Improvement, and coordinator of the translocation process, DOC
- Dr Robin Gardner-Gee—Former project manager Motuora Restoration Society for seabird translocations
- Dr Matt Rayner—Seabird specialist, particularly small gadfly petrels (*Pterodroma*); Centre of Biodiversity and Biosecurity, School of Biological Sciences, University of Auckland
- Cathy Mitchell—Veterinarian and independent contractor

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