# GUIDELINES ON

# RODENT SURVEILLANCE TECHNIQUES

## PURPOSE

- These Guidelines are to be used by Project Managers conducting rodent eradication projects based on the PII Resource Kit for Rodent and Cat Eradication.
- The Guidelines describe different techniques for surveillance of rodents. These techniques will be of use for the surveillance activities in the Biosecurity Plan to check for the presence of rodents and in the Monitoring Plan activities when assessing the success of the project.

## 1. TRAPS

### 1.1. WHICH TRAPS TO USE

- Select a trap that is specifically designed to catch rodents, taking care to note the difference between rat and mouse traps. Always use the right trap for the right species. Consider that big rats can get out of mouse traps or get a fright and not come back. Mice, because they are lighter and fast, may not set rat traps off.
- It is also important to consider the environment you will be trapping in and which traps will cope with the conditions. Look for one that is least likely to jam or rust. Talk to others who have done rodent trapping in similar environments to find out what traps work best. If you can't get information like this do some trials of your own away from the project site to see what traps work best.
- Victor Professional snap-traps are recommended as effective, highly portable traps. Victor Professional snap-traps are more portable and ok for ship rats or Pacific rats, but Norway rats have been observed to escape – take care to set then correctly to avoid mis-captures and trap-shy animals

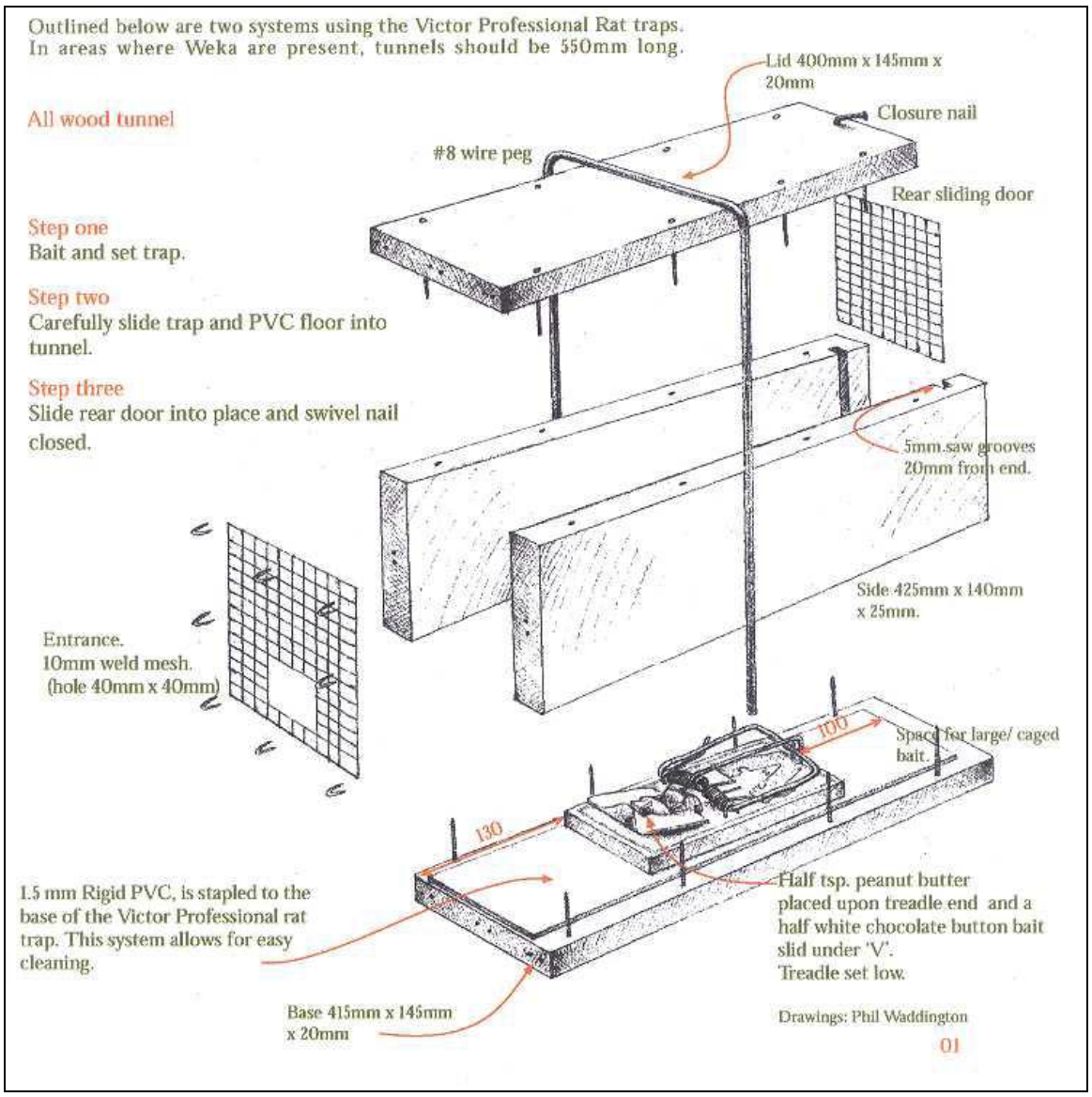



***Figure Wooden cover for use with Victor Professional Rat trap (or Victor Professional mouse trap). Increase length of tunnel if there are birds that may be inquisitive or attracted to bait (such as rails). Illustration provided by permission from the New Zealand Department of Conservation's Trapping Best Practice documents***

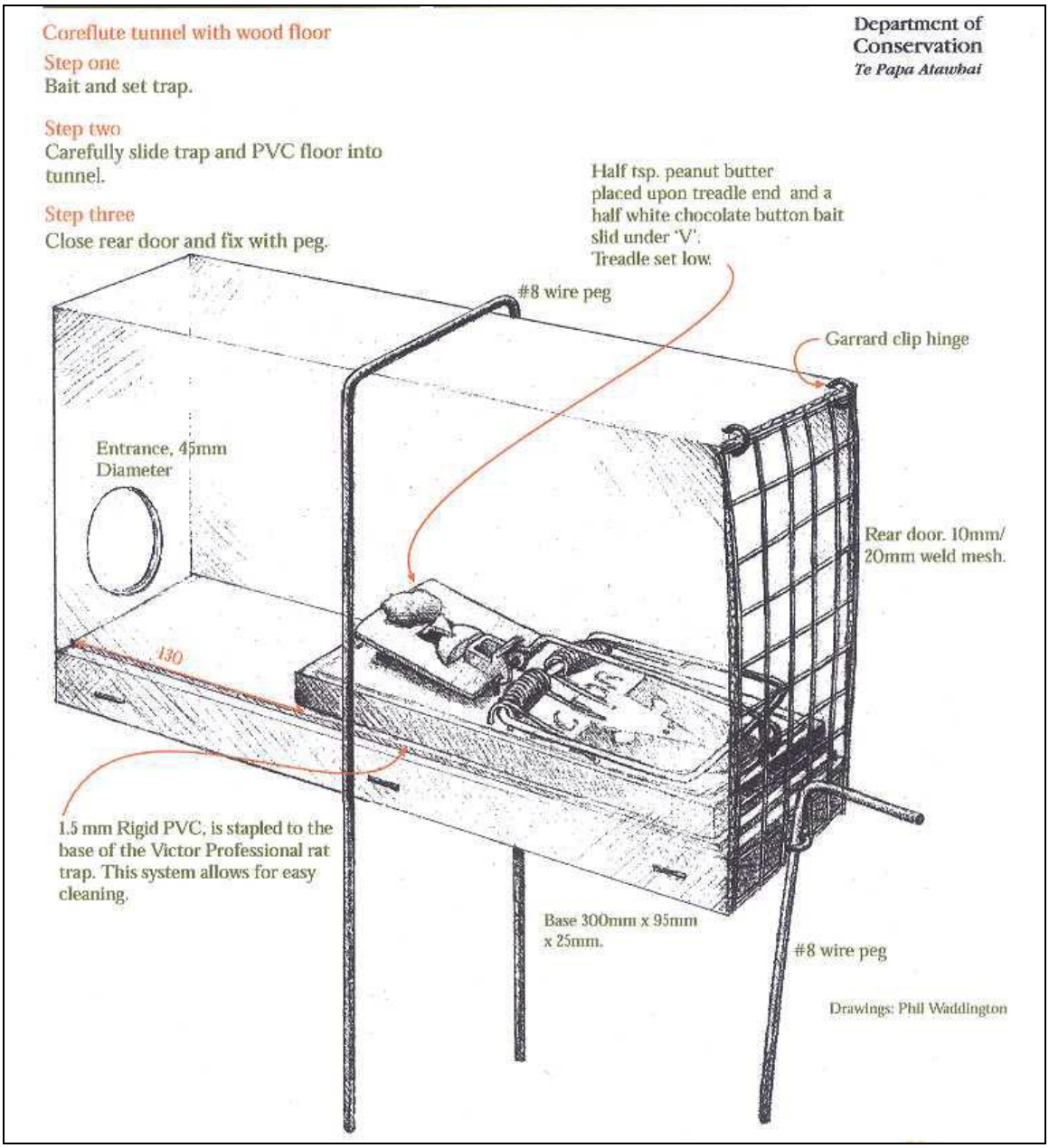



***Figure. Design for plastic, corrugated plastic (coreflute), or sheet metal cover for use with Victor Professional Rat trap (or Victor Professional mouse trap). Increase length of tunnel if there are birds that may be inquisitive or attracted to bait (such as rails). The PVC floor is not essential, the wooden base should be adequate. Illustration provided by permission from the New Zealand Department of Conservation's Trapping Best Practice documents***

- DOC 150's in purpose-built wooden tunnel but these are bulky and difficult to transport.

## 1.2. TRAP MANAGEMENT

- On new traps the end of the trigger arm often needs filing to make it smooth for maximum sensitivity
- Never leave traps out when not in use because the springs rust and weaken
- Treat all metal parts with fish oil (spray or paint on) or wax (use melted wax in boiling water as a dip into which the whole trap is dipped and then dried, or use melted wax to brush on) to reduce rusting before and after each period of use. Do not use anti-rust sprays (e.g. CRC) as the smell of these may deter rodents.
- Traps must be tied firmly with strong string or thin wire to vegetation or a firmly set peg, so that injured rats and large mice do not drag them away, or be themselves dragged away by scavenging predators

## 1.3. PLACING TRAPS

- Place traps where there is plenty of natural cover and where rodents are likely to be active (e.g. alongside large rocks, around the base of trees, under logs and overhanging vegetation). If rodent droppings, food remains or runways are visible, set traps nearby.
- The actual number of trap-sites will depend on the number of traps, the personnel available and the size of the habitat being sampled.
- Ensure traps are on a level surface and are stable (they don't move or 'rock' if pressure is put on any corner or side of the trap) as rodents can be easily frightened and may not return to a trap after having a bad experience. This is especially important when Norway rats are present
- Set two traps at each site. This increases the potential capture rate and doubles the number of trap-nights for little extra effort. If covers are not used, set traps within 1m of each other.
- When both rats and mice are present, or if you are unsure which rodents are present, set a mouse and a rat trap at each site. If only rats or only mice are present, set two traps of the appropriate type.

## 1.4. COVERING TRAPS

- Covers for traps should generally always be used (to make sure the rodent approaches the trap from the best side to ensure it gets caught, to limit trap disturbance by weather or other animals, and to help reduce the non-target animals likely to be caught).
- Covers can be made of whatever material is available and portable (e.g., wire mesh, clear plastic sheet, or plastic drain pipe). Traps should always have a cover (tunnel). These covers can be made of lightweight materials such as corrugated plastic, thin sheet metal or similar. The black plastic tunnels for the BlackTrakka tracking pads are also suitable for covering snap-traps

- Stones, sticks or wire pegs will hold covers in place. Bent wire hoops, or similar, should be placed across the entrances to exclude non-target animals.

- If covers are used, both traps may be set back to back under the one cover. A wire hoop or forked stick placed in the space between them helps prevent one setting off the other.

- Ensure the cover does not impede the action of the traps. To test this, set the selected trap off when it's within the selected cover. If the arm of the trap hits the cover at any point, the cover needs to be larger.

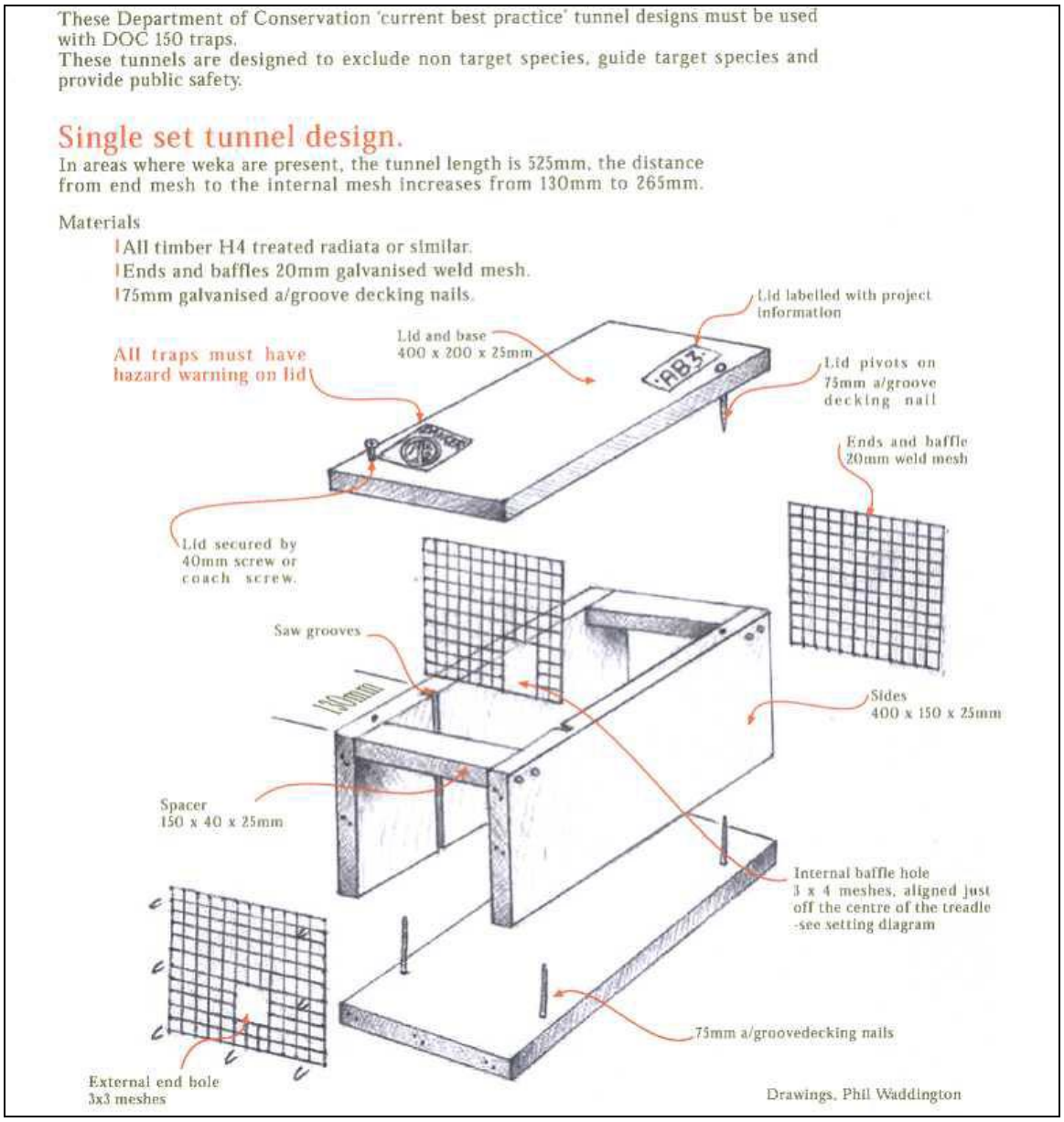



***Figure. Cover design for use with DOC150 traps. Increase length of tunnel if there are birds that may be inquisitive or attracted to bait (such as rails). Illustration provided by permission from the New Zealand Department of Conservation's Trapping Best Practice documents***

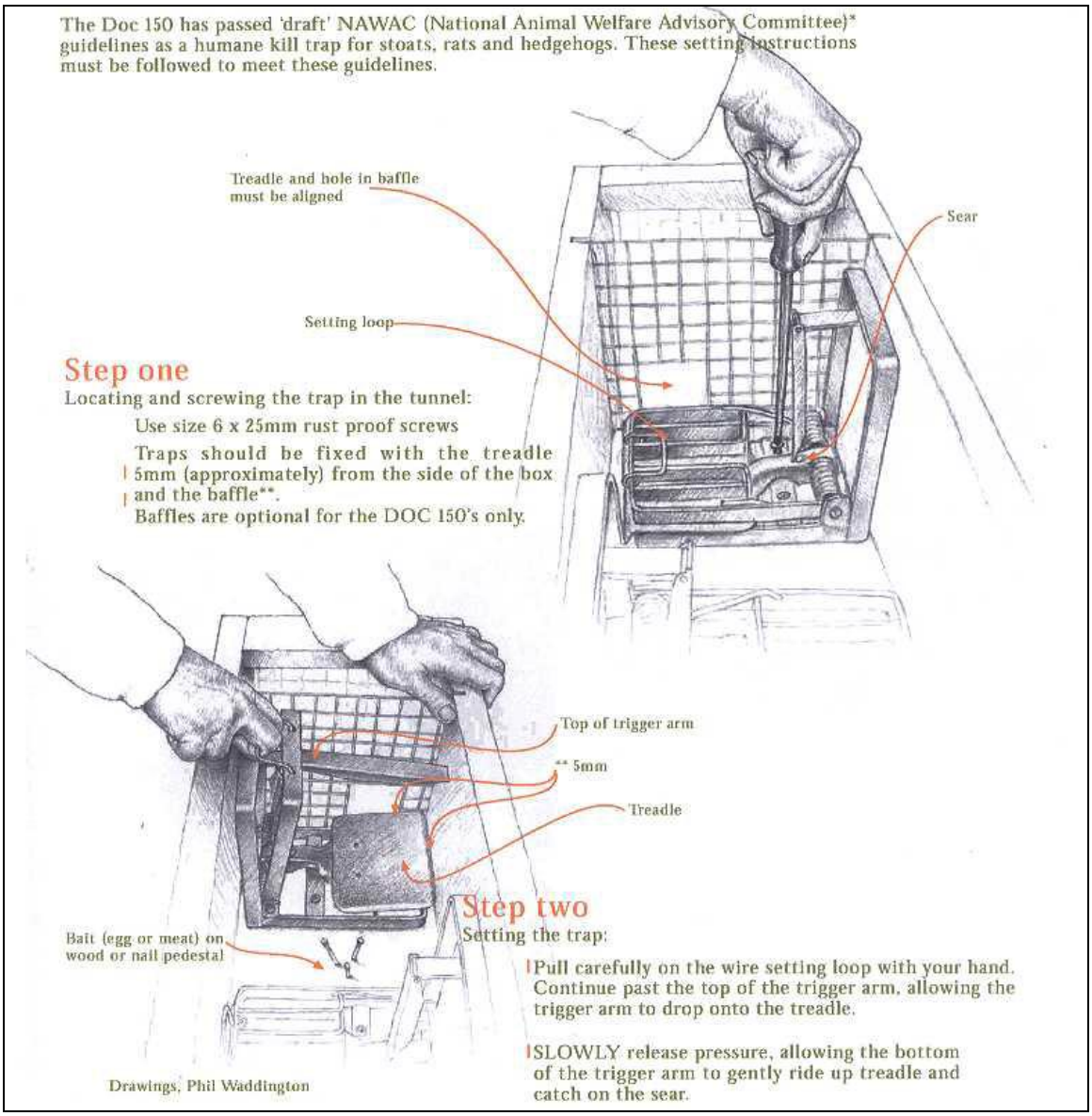



***Figure Setting the DOC150 trap. Illustration provided by permission from the New Zealand Department of Conservation's Trapping Best Practice documents***

## 1.5. BAITING OF TRAPS

- If time is available, experiment beforehand with several baits because species and individuals vary in their preferences. However, do NOT change bait types during the index trapping as this may give inconsistent or biased results. Always try to use the same bait for all index trapping, as this means results from all trapping in different areas or seasons is directly comparable.

- A stiff mixture of peanut butter and rolled oats is recommended as a reliable standard bait for index trapping and it lasts well.

- Other suitable baits include roasted coconut, cheese, nuts, chocolate, bacon, or leather soaked in fish oil.
- Be consistent and use the same bait in every trap
- Renew the bait whenever its attractiveness has been reduced by rain, hot weather, mould, or partial consumption by ants or other insects.

## 1.6. INDEX TRAPPING

- An index of abundance can be used to compare the density of populations on different islands, or in different regions or habitats. It can also provide useful information on changes in rodent abundance in the same location between seasons.
- The more trapping you do the more likely you are to trap more individuals. For example, take 2 locations (Site 1 and Site 2) with the same abundance of populations. If at Site 1 you deploy 50 traps and at Site 2 you deploy 100 traps, with all else constant you would expect to trap twice as many individuals at Site 2 than at Site 1. The Abundance Index is a measure of the number of individuals captured adjusted by the number of traps deployed

### 1.6.1 HOW TO USE INDEX TRAPPING

- Always use the same brand of trap for index trapping (otherwise differences in their effectiveness will bias your results, meaning you cannot compare them with other results)
- An index line should ideally consist of at least 25 sites evenly spaced apart, with two traps per site within the same cover.
- Plan index trapping so as to give a minimum of 100 corrected trap-nights in each habitat. Fifty traps for three nights gives a maximum of 150 trap-nights. (see Evaluating results, below).
- Index lines are usually run for three nights.
- The spacing between sites should be as large as possible within the range of 25-50m. Measure the intervals by pacing or using a tape measure. Permanent index lines should be accurately measured.
- To maintain consistency during index trapping, covers should either be present at every site or be absent from every site. Covers may influence trapping success so they should be left in place between trapping sessions to accustom the animals to their presence.

### 1.6.2 EVALUATING RESULTS

- Check the traps as early as possible each morning because tissue deteriorates quickly in warm weather and carcasses become fly-blown
- Make a brief note of the previous night's weather as this can affect the animals' behaviour and influence the trapping results

- Record whether each trap is sprung or unsprung and whether the bait has been removed, partly eaten, or left untouched
- Note any disturbance by other animals
- For example, your field notes may read:
    - *Line A. 23/11/2009, Banana Island. Weather: Wet, windy, warm night:*
    - *Site 1. OK/Sp.B.OK (i.e., 1 trap unsprung, bait OK/1 trap sprung, bait OK)*
    - *Site 2. 2 OK (i.e, both traps unsprung, bait OK)*
    - *Site 3. RAT/OKB.G. (i.e. 1 capture/1 trap unsprung, bait gone)*
- The next step is to calculate the corrected number of trap nights by making allowance for all those traps which had been set off
- Subtract half a night for each of the sprung traps (whether or not they had caught a rodent) on the assumption that each will have been sprung for an average of half the night. Do not make a correction for unsprung traps with the bait removed because they were still capable of catching a rodent
- The Abundance Index is calculated at the end of the trapping session from the total number of rodents caught and the total number of corrected trap-nights, and is expressed as the number of captures per 100 trap-nights. This index of abundance is the figure you can use to compare rodent abundance for example, between seasons on a single island; before and after control efforts at the same site; or to compare relative rodent densities on two or more different islands.

### 1.6.3 EXAMPLE OF ABUNDANCE INDEX FROM RINGGOLD ISLANDS, FIJI

- Example from Seniloli E. & Rasalato S. 2009

| Island | Species recorded in 2007 -2008 | 2007-2008 survey efforts & Findings | 2009 survey efforts & findings |
|---|---|---|---|
| Naqelelevu | Pacific rats Rattus exulans<br>Cat not present<br>1 Female Dog present | 2 trap nights<br>50×50 m grid<br>2 hours of night searches<br>Feeding signs & droppings<br>28 Pacific rats caught | 2 trap nights<br>6 transect lines (10stations)<br>night searches<br>Feeding signs & droppings<br>No rats caught |
| Nukusemanu | Pacific rats<br>Cat present | 2 trap nights<br>3 transect lines<br>30 cat traps set (not caught)<br>3 Pacific rats caught (index of abundance = 8) | 2 trap nights<br>3transect lines<br>1hour night search<br>No rat detected<br>Cat still present |
| Nukupureti | Pacific rats | 2trap nights<br>3 transect lines<br>Pacific rats observed<br>43 Pacific rats caught (index of abundance = 107) | 2trap nights<br>5 transect lines<br>2 hour night search<br>No rat activity signs<br>None caught/ detected |
| Nukubasaga | Pacific rats | 2trap nights<br>3 transect lines<br>2 hour night search<br>Feedings observations/signs<br>Evidence of droppings<br>24 Pacific rats caught (index of abundance = 33) | 2 trap nights<br>3 transect lines<br>No rat activity signs<br>No rat detected/caught |
| Vetauua | Pacific rats | 1 trap night<br>3 transect lines<br>1hour night search<br>Feedings observations<br>12 Pacific rats caught (index of abundance = 39) | 2 trap nights<br>4 transect lines<br>1hour night search<br>No rat activity signs<br>None caught |
| Tauraria | None | 1 trap night<br>1 transect line<br>Feedings observations<br>No Pacific rats caught | 2 trap nights<br>1transect line<br>Peanut butter & T/tunnels<br>No rats detected |
| Tainibeka | None | 1 trap night<br>1 transect line<br>Feedings observations<br>No Pacific rats caught | 2 trap nights<br>1transect line<br>Peanut butter & T/tunnels<br>No rats detected |

### 1.6.4 WORKED EXAMPLE CALCULATION OF THE ABUNDANCE INDEX

- In the following example, seven rats have been caught and 13 traps sprung without catching anything:

**50 traps set for 3 nights**: = 50x3

= 150 total trap nights

**Trap nights lost:** = ½ (captures + sprung, empty traps)

= 7 (Rats trapped) +13 (traps sprung with no catches)

2

= 10

**Therefore the corrected number of trap nights is**:

= total trap nights – trap nights lost

=150-10

= 140

**Index of abundance** **=** Captures x 100

Corrected trap nights

= 7x100

140

**= 5.0 captures/100 trap nights**

# 2. TRACKING TUNNELS

- Tracking tunnels are a long rectangular box with a piece of cardboard that has an inked section in the middle. Tasty bait such as coconut meat or fish is placed on the ink or suspended from the roof inside the tunnel (the latter is useful where crabs are an issue). Anything that goes through leaves footprints.
- Tracking tunnels are a simple and effective form of monitoring to record the prints of small animals. Footprints of many animal species are easily identified. More importantly the prints of key invasive species (e.g. rodents) are easily identified from those of native species.
- You can make your own tracking tunnels (modelled on the Black Trakka) or buy ready-made Black Trakka's from Gotcha Traps (www.gotchatraps.co.nz/html/contact_us.html) in New Zealand. The Gotcha traps website provides useful information on how to identify prints as well as set tunnels. This tool has been adapted from that information.
- We recommend buying readymade tunnels as it is difficult to replicate the ink cards with a product that weathers well and gives good footprints.

## 2.1. WHY USE TRACKING TUNNELS INSTEAD OF TRAPS?

- Tracking tunnels are a very effective detection device but obviously do not harm the rodent. They have advantages over traps or other control devices in that they can be placed out and left for many days if necessary without checking (traps generally need to be checked and rebaited every day). There is less apparent 'shyness' of tracking tunnels because there is no structure such as a metal trap for the rodent to be cautious of – it is more likely that a very low density of rodents (e.g. just one or two having arrived on an island) would be detected in tracking tunnels rather than in a similar network of traps. They are easier to carry, place out and maintain than traps and therefore a larger area can generally be covered by each single person. If there are very sensitive non-target species tracking tunnels will not harm them.
- The biggest disadvantage of tracking tunnels is that once you have detected unwanted rodents, then you must use a different method to capture or kill them!

## 2.2. HOW DOES IT WORK?

- The Black Trakka consists of a lightweight polypropylene tunnel (500mm long x 100mm high and wide), a pre-inked tracking card for small animals (up to large rat size but they are not suitable for cats) and two U-shaped pins to secure the tunnel. Lures are placed on the ink-free section in the centre of the card. When an animal eats the lure, it stands on the inked section and ink is transferred from their feet to either end of the card where the tracks of the animal are recorded on the absorbent area of the card. The tracks on this section of the card do not smudge allowing for easy identification using the sample prints provided.

## 2.3. LURES

- Choice of bait depends on what species you are targeting. Burnt coconut or peanut butter (crunchy peanut butter lasts longer than smooth) are good lures to attract rodents and are also likely to attract other animals such as insects, small birds and reptiles. You can use the tracking tunnels to monitor for lizards.

## 2.4. TUNNEL PLACEMENT

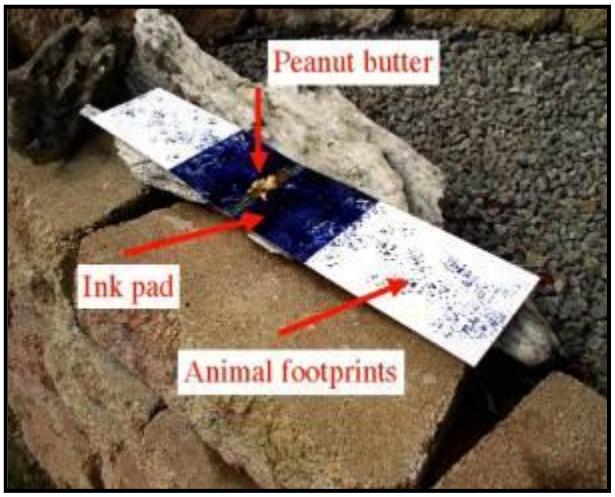



***Photo Source: [www.wildaboutnz.co.nz](http://www.wildaboutnz.co.nz)***

- Take care to be selective in your tunnel placement. Consider places the invasive species you are monitoring is most likely to be attracted too (e.g. where there is seed fall or fruit crops or shelter, on the edge of the vegetation/beach line, around buildings). Some species such as rodents do not always have a large range. To maximise your chances of tracking them either set tunnels at intervals of no more than 50m along a transect line or set them up in tandem with other monitoring devices

- If you are monitoring mice you may want to have the stations at closer spacings, i.e. 10-25m apart.

- Make sure you secure the tunnels to the ground. Large animals such as pigs and dogs may be attracted to the bait in the tunnels and disturb them. Black Trakka tunnels come with U-shaped pins which are placed over the top of each tunnel. You can make your own pins out of strong wire.

- On islands where land crabs or other non-target species create a nuisance by constantly removing the bait or smother the tracking card in their prints, consider placing some of the tunnels in flattish places above the ground (e.g. tops of fallen logs, on top of large boulders, on sloping tree trunks or on benches etc in buildings). Make sure the tunnel and card is secured well.

## 2.5. HOW LONG DO YOU RUN TUNNELS FOR?

- This depends on the information that you wish to obtain – it may be just to confirm that a species is present, which may not take long (1 or 2 nights), or it may be to work out how widespread it is over the island, which may take longer. Once you have gathered the information you require, the tracking can be stopped. If the species suspected as present is in low numbers it may take some time for an animal to find a tunnel, so take account of this in your planning when preparing your stay in the field to do this work. Try to cover as much of

the island as possible, or at least all the major habitat types. Consider placing more tunnels out, or shifting tunnel locations after several nights to ensure greater coverage if necessary.

### 2.6. HOW DO YOU IDENTIFY THE VARIOUS FOOTPRINTS?

- See Section 6.1 for a discussion on identifying footprints
- The Gotcha traps website has a good identification guide (http://www.gotchatraps.co.nz/)
- Build your own database of prints – keep cards with good prints, write on the card what species the prints belong too and where they came from

## 3. BAIT STATIONS

- Brodifacoum based bait blocks often with a chocolate lure can be used in bait stations to identify the presence of rodents by the way they eat the bait blocks.
- These can be used in places where you may have concerns about children or species like chickens having access to bait.
- These baits are often attacked by insects so it's important to be able to tell the difference between insects and rodent sign. Mice are generally messy eaters and leave lots of crumbs, rats tend to leave chunks as opposed to crumbs. Both will often leave dropping as shown in Figures below. Insects such as cockroaches and earwigs tend to drill lots of little holes so the baits resemble honeycombs.

***Figure : Mouse eaten bait***

***Figure: Cockroach eaten bait (photos Jo Ritchie)***

- Refresh as required by invertebrate or crab damage or weather degradation. Fresh bait is best, but old and even mouldy bait is still relatively palatable to rodents so depending on the conditions replace bait every few months (as an indication – local conditions will dictate this, but ensure you have a set schedule of checking and replacing baits). Do not wrap baits as this decreases palatability significantly.
- If possible, use wooden boxes (rat motels) for bait stations (these are preferred over plastic stations as rodents appear to enter them more readily). Covering station or tunnel entrances with insect mesh to reduce insect damage to bait is not recommended as this is likely to deter rodents from entering.

# 4. WAX BLOCKS

## 4.1. USING WAX BLOCKS

- A wax block is a wax-based, tasty lure with a nice smell e.g. coconut, peanut butter – rodents leave gnaw marks on them. Wax blocks can be placed on sticks at ground level or up trees (away from crabs). Plain wax tags do not appear to be sensitive to low numbers of rats so do not use these.
- Wax blocks can be used to collect teeth marks.
- See Section 6.3 for identification of the teeth marks

## 4.2. HOW TO MAKE CHOCOLATE FLAVOURED WAX BLOCKS

- This tool provides you with a simple, cost effective recipe for making flavoured wax blocks which can be placed in the field and are attractive to rodent species. They leave distinctive teeth marks in the blocks. This tool is for chocolate flavoured blocks but you may like to substitute this for more local flavours such as coconut.
- This recipe makes about 32 blocks

### 4.2.1 INGREDIENTS :

- 1 packet of 6 candles
- rounded teaspoons cocoa powder
- Paperclips or wire loops (one per cube)
- Vegetable oil for greasing ice cube trays

### 4.2.2 EQUIPMENT:

- Saucepan, preferably with pouring lid (or small jug or funnel if no pouring lid)
- Ice cube trays
- Wire cutters for making loops

### 4.2.3 METHOD:

- Grease the inside of the ice cube trays thoroughly, either using cooking spray or vegetable oil on a piece of kitchen roll or tissue. Put them somewhere flat on a couple of sheets of newspaper.
- Make wire loops (if using) or bend paperclips so that the two parts are at 90 degrees to each other. For the wire loops, cut pieces of thin wire into lengths approximately 4cm long. Bend into an 'omega' shape (straight bit, semi-circle, straight bit). It's worth preparing more loops/paperclips than you think you'll need in case the mixture makes more blocks than you're expecting.
- Melt candles slowly in saucepan. When melted, stir in the cocoa powder and mix well, as it tends to sink to the bottom. Fish out the wicks of the candles and discard.
- Pour the wax slowly into the ice cube trays, using the jug or funnel if necessary. If you do use a jug or funnel, make sure they are as warm as possible to stop the wax setting too soon. The ice cube holes don't need to be filled right to the top – about two thirds full is fine. It doesn't matter if they are not all the same size.
- Once the wax has partly set and a skin has formed on the surface, approx 5 mins) push a paperclip straight down into each cube so that around two thirds of the clip is embedded in the wax. For extra strength, bend the two parts of the paperclip out to a 90 degree angle before you do this, so that the part of the clip inside the cube runs parallel to the surface and the bit sticking out is at right angles to the surface.
- Allow the cubes to set and remove from the trays. If they don't come out easily, clear away as much wax as possible from around the cube and insert a thin knife blade down the side of the cube. Depending on how stuck they are, it might be necessary to do this on all four sides.
- Finally, use a sharp knife to trim away the excess wax from the top surface of the cube (where the metal loop or paperclip goes in. Try to leave the edges as flat as possible so that any potential rodent teeth marks will show up clearly on a flat surface.

## 5. VISUAL SIGNS

- Record visual signs by taking photographs. In the picture include a reference item, e.g. a hand, ruler, pencil that will help to indicate the size of the subject of the photograph.
- Also photograph around the scene to show the location. For example, when photographing broken eggs, take photos of the seabird nesting colony aswell (to determine later it its abandoned or presence of other predators).
- Record as much information about when and where the sighting was made. Include date, time of day, location (GPS co-ordinates if possible), weather conditions.
- Carefully label or identify each sample or photograph. You can take many samples in one trip and it is easy to mix them up later and not be able to trace the origin. This is particularly true for digital photographs.
- Use a field notebook to record all important information.
- Physically mark the spot, use flagging tape if available, so that you can return to the spot later.

## 5.1. FOOTPRINTS

- Footprints can be a great way of detecting sign in the field. You may use a formal monitoring system such as Tracking Tunnels (see Section 3) or just observe footprints in the field. Good places to find them are on wet sand, wet soil especially mud and at the edge of any soft soil or sand near water (e.g. puddles, pools).

### 5.1.1 RATS AND MICE

- Rats have four toes on the front foot and five toes on the hind. Ship rats have a clear split in the central pad of the hind foot. Unlike ship rats, Norway rats have a single central pad. Rat tracks will vary from foot width of 5mm through to 18 or 22mm in a large adult. You can identify individual animals by their foot widths. Mice tracks have the same pattern as rat tracks but are smaller and often there are a lot more of their prints on tracking cards.

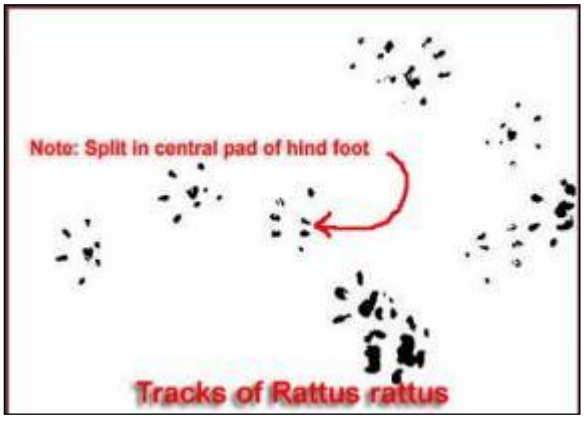



***Figure: Ship rat prints***

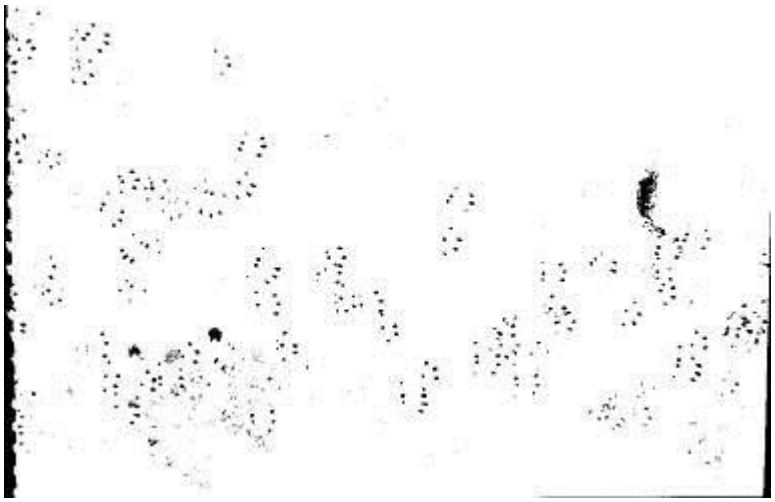


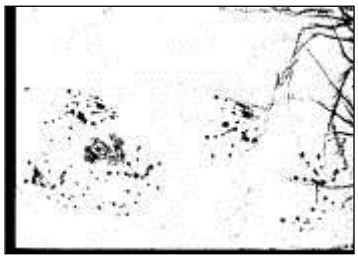
***Figure: Mouse prints Jo Ritchie***

### 5.1.2 CATS

***Photo: Gillies C, 2002***

- Cats only occasionally track in tunnels but the size of their print will make them readily distinguishable from other species

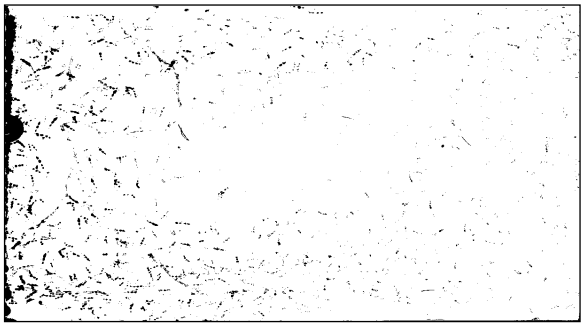
### 5.1.3 FROGS



**(Photo: Jo *Ritchie)***

- Frog prints are distinctive and reflect their webbed feet

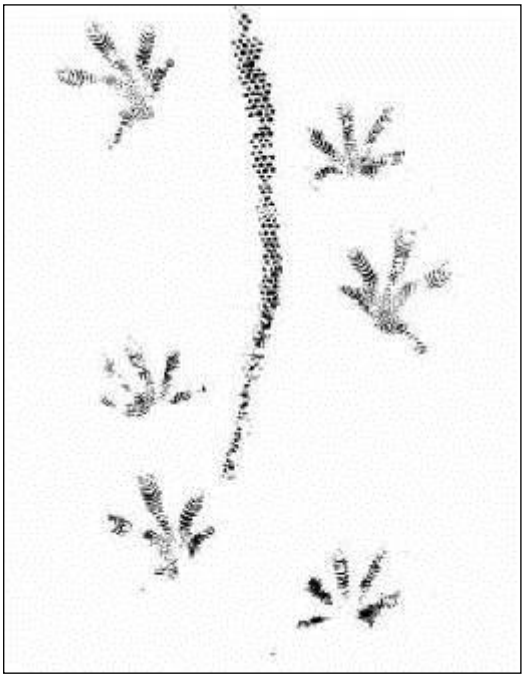
### 5.1.4 INSECTS



***(Photo: Jo Ritchie)***

- Insect prints are often made up of series of small dotted lines which reflect the jointed nature of their feet

### 5.1.5 LIZARDS



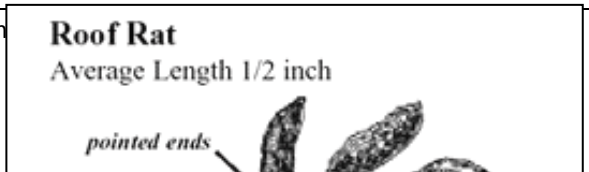
***(Photo: Jo Ritchie)***

- Lizard prints often have very well defined toe and foot prints and a long thin line in between them which is the tail or part of the body.

## 5.2. DROPPINGS

- Rat and mouse droppings are both a similar shape, but are quite different in size and can be easily distinguished. Both vary in colour from mid brown to black and in consistency from soft to very hard. Rat droppings are much bigger and thicker compared to that of mice. Rat droppings are generally about 12-156-





20mm long and 3-4mm in diameter. Mouse droppings are much smaller – only about 3.5-8mm long and 1-1.5mm in diameter

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***Figure: Ship rat droppings Source: Miller C. 2008***

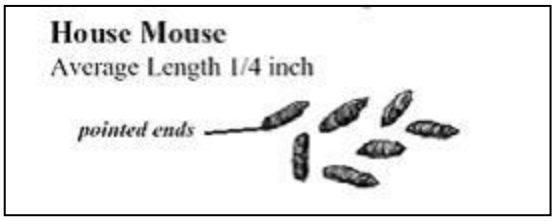


***Figure: Mouse droppings. Source: Miller C. 2008***

***Figure: Rat droppings about 2x size of that of mouse (photo on right). These rodents have eaten brodifacoum Photo source: Jo Ritchie***

***Figure : bird droppings are often thinner and longer. Seabird droppings are often white & may contain fish scales & bone &/or shell fragments. Insect droppings are generally much smaller than that of a mouse Photo: Jo Ritchie***

## 5.3. TEETH MARKS

- Teethmarks left by rats and mice are very similar in shape but, with a little practice, you can tell them apart by their size. You need to be able to do this if you are using wax blocks or tags and its also useful when looking at sign on food items such as fruit and berries.

- Rat teeth are larger and so they leave longer, wider marks than mouse teeth. Rat teeth marks are about 2mm wide (1mm p/tooth), while mouse teeth marks are about 1mm wide (0.5mm p/tooth). Both always occur in pairs – two parallel grooves with a slight ridge between them. Its useful to have a hand lens or small magnifying glass to investigate teeth marks more closely. Figures below illustrate the differences between the two.

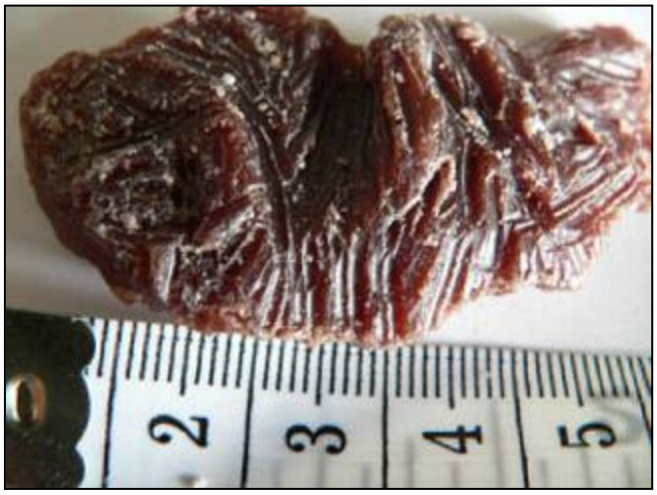


***Figure: Rat teethmarks***

***Figure : Mouse teethmarks Photo source:Miller C, 2008***

- Other animals are also likely to leave sign on bait blocks or monitoring blocks. Work out what these might be (e.g. crabs) and learn to identify them.

  *Figure : Bird beak marks – note the marks are single and deeper unlike the paired teeth of rodents. Photo:Miller C 2008 – Where is this photograph?*

### 5.4. SIGHTINGS

- While rodents are mostly reclusive and shy, the actual sighting of an individual is another method of detection.
- Rodents usually become more active after about 4pm.
- Search for them in abandoned buildings, occupied houses, food stores, plantations/crops, water supplies.
- If possible, catch specimens to confirm identity. If you catch none despite believing they are present, search at night using a strong headlamp or spotlight

### 5.5. SEA BIRD PREDATION

- Focus on colonies of small common seabirds (e.g. terns, noddies, shearwaters). Search the colony for abandoned or failed eggs - eggs eaten by rats have many jagged edges. Larger rats can prey on larger eggs while smaller eggs are often smashed into smaller pieces. On rat-free islands, failed/abandoned eggs are generally intact or broken open with no jagged edges – although some crab species can also smash up eggs. Note that species like frigate birds can defend their eggs against rats so may not be good indicators of rat sign.

## 6. PERMANENT RODENT MONITORING STATIONS

- Use of permanent monitoring stations for rodents is a common technique for monitoring the success of rodent eradications. These consist of a variety of devices designed to catch or detect any remaining or re-invading rodents, including snap traps, tracking tunnels and wax tags and are maintained at a number of sites for the period of the monitoring

### 6.1. EXAMPLE OF A PERMANENT MONITORING STATION

- The following example is from the Ringgold Islands (Seniloli E. & Rasalato S. August 2009):

- Permanent monitoring stations were established, consisting of 10 stations, 20m apart. Each station was marked with a number, on a white rectangular plastic strip nailed to a tree.

- **Each station contains:**

    - Two snap traps baited with roasted coconuts
    - A tracking tunnel baited with burnt coconut attached to each end of the tunnel roof
    - A peanut butter flavoured wax tag nailed to trees at random heights away from hermit crab reach

- **Monitoring tools:**

    - Peanut wax
    - Tracking tunnels, ink papers, burnt coconut
    - Snap traps and roasted coconut

- **Monitoring programme:**

    - Set for two nights
    - Checked daily for any signs of rodent activity

- **Number of transect lines per island:**

This is related to time available to undertake monitoring and island size. The following system was used on the Ringgold Islands:

| **Treated Islands** | **Nukusemanu** | **Nukubasaga** | **Nukupureti** | **Vetauua** | **Tauraria** | **Naqelelevu** |
|---|---|---|---|---|---|---|
| **Area (ha)** | 1.55 | 18.45 | 2.7 | 35 | | 145 |
| **Number of monitoring transects** | 3 | 5 | 3 | 4 | 1 | 6 |
| **Distance between transects in metres** | 20 | 20 | 20 | 20 | 20 | 20 |
| **Detection tools** | Peanut wax and tunnel | Peanut wax and tunnel, 2 snaps | Peanut wax and tunnel, 2 snaps | Peanut wax and tunnel, 2 snaps | Peanut wax and tunnel, 2 snaps | Peanut wax and tunnel, 2 snaps |

| | | | | | | |
|---|---|---|---|---|---|---|
| **Total number of stations** | 30 | 51 | 22 | 40 | 10 | 65 |


| | | | | | | |
|---|---|---|---|---|---|---|
| **Number of trap nights** | 2 | 2 | 2 | 2 | 2 | 2 |

***Figure: A permanent rodent monitoring station containing: rat trap, tracking tunnel and wax blocks***

## 7. DNA SAMPLING OF RATS

- This tool provides a useful technique to aid the identification of rats in the case of reinvasion after an eradication operation has been undertaken.
- DNA samples collected prior to an operation from both the target island and the most likely sources of reinvasion (e.g. an island within swimming distance), can be compared with samples collected from any animals that may be found after the operation. This will determine if they are survivors or re-invaders. You can then target your response to maximize the chances of removing those animals. If the rodents are from a re-invasion, it may also assist in determining where they came from and possibly therefore how they got there – very important information to help improve biosecurity measures.

### 7.1. WHY IS IT USEFUL?

- Different follow-up techniques may be required depending on where rats have come from. If they came from another island you need to consider doing prevention work on the source island as well as on the project island. If they are survivors you need to concentrate your efforts on the project island.
- If your eradication site is close to other islands where rats are present, DNA sampling prior to an eradication operation will give you information about whether rats are regularly moving between the islands (swimming and/or human assisted). If the answer is yes, you must either include those other islands as part of the eradication programme or have a biosecurity management programme in place before you start the eradication.

### 7.2. HOW DOES IT WORK?

- Every living cell contains DNA molecules with the information about that organism. No two organisms except clones and identical twins have the same DNA. Populations of rats that are isolated from one another with no movement between islands have different DNA. If island rat populations are linked by swimming their DNA may be very similar. This means you can potentially identify re-invasion sources and try to manage them.

### 7.3. SAMPLE SIZE

- It is recommended that samples from 20 rats are collected from the eradication island before the eradication takes place. This is because there is only one chance to sample that population: there is no possibility of increasing the samples after the eradication has taken place. Ten might be good enough but with no guarantees, 30 would be great, 50 would be fantastic. With 20, there is probably a good chance of getting a result, without requiring lots of fieldwork trapping them. (However this depends on how difficult rats are to catch in these places - sometimes 20 samples is asking a lot!) The samples need to be stored in >70% ethanol (preferably refrigerated, but not essential). The samples can be tested against any re-invaders.

- If a re-invasion occurs, you need to catch as many of the early re-invaders as you can: one is enough, but the important thing is that the re-invaders should be caught before they start breeding and creating a new population with its own genetic signature.

- Then collect samples from the possible source of invading animals. Ten rats from each source is recommended which should be sent to the lab and tested against the re-invader: if there is no clear result return to the potential source populations and get more samples.

## 7.4. COLLECTION REQUIREMENTS

- For each rat, snip off a 3-4cm length of tail and put it in the collecting vial. Try to immerse as much of the sample in ethanol as possible. Do NOT place too big a sample into a single bottle: it is vital that the ethanol is able to penetrate all the tissue and only a tiny amount of tissue is actually needed for DNA processing.

- ANY piece of rat flesh can be used if the tail is not intact. Note on the vial what part of the rat the sample is from (if possible).

- NEVER mix samples from two different rats in the same collecting vial: one rat per vial only. Make sure blood or tissue from one rat never contaminates the sample from a different rat, for example, from a dirty knife.

- The closer the sample is taken to the rat's death, the better, but up to 2-3 days should not make a difference to the quality of the sample. This might differ in a hotter climate.

- Collecting vials contain 70% - 95% ethanol. The higher the ethanol content, the better. After collection, it is best to keep the sample refrigerated. In New Zealand conditions, samples have been stored at room temperature in 70% ethanol for several months without negative consequences.

## 7.5. LABELLING

- Vials should have plain labels to record the information required for the study. Usually, sticky labels on the outside of the vial can be held on with extra plain tape. Write the notes on the label in pencil so that the labels are less likely to be harmed if any ethanol leaks from the vials.

- An alternative is to write in pencil and insert the label into the ethanol tube itself so that it can be clearly read from the outside (preferably without opening the bottle again). Labels both on the inside and outside is even better, to ensure the sample is not confused if one label gets damaged or lost.

- The advantage of using labels inside the bottles is that it avoids ink running due to leakage of ethanol, or by getting wet or labels falling off. The disadvantage is that it can be difficult to read the labels inside the bottles and can require unnecessary handling of the specimen inside, with the associated risk of contamination. Marker pens can be used to write directly onto the outside of the collection bottles: check for risk of smudging or fading.

- What information needs to be recorded for each sample?

    - Date – in a standard format, e.g. 23.11.2009

    - Species – e.g. kiore or ship rat; if there is doubt, indicate a best guess with a question mark. It should be possible confirm species using genetics, but it is useful to record a field guess to help decide lab processing order and to cross-check genetic results with field records.

    - Sex – male or female; only record this if you are confident, otherwise leave blank. (Samples are genetically sexed, but the procedure is not always reliable.)

    - Broad Location – the area where the rat was caught and the name of the Island, e.g. West landing, Banana Island.

    - Trap number – if the sample was caught on an established trapping line, record line and number to give exact spatial position. Separately create a record of GPS locations of every trap so that all samples are spatially referenced. Even if exact spatial location is not required for the study, it is very helpful to record trap number in case there is a query about the sample (e.g. genetic results) and you need to check against the field notes: *this happens a lot*!

    - Useful field notes - It is advisable to keep a separate field notebook duplicating the information on the vial label (broad location, date, trap number, species, sex) and adding any extra field notes.

    - Noting whether the sample was fresh will help to explain why genetic processing might have failed; if the specimen had been cannabalised it might explain a species misidentification; etc.

    - Record strange colourations, e.g. ship rats occur in three different colour morphs, and some have abnormalities such as white tail tips.

    - It is good scientific practice to also record morphological characteristics in the field notebook: Head and body length; Tail length; Mass. These characteristics can also clarify species ID. If sample collection is being done by volunteers (members of the public), do not ask for field notes; keep the collection process as straightforward as possible.

## 7.6. HEALTH AND SAFETY PROCEDURES

- Keep ethanol well away from flames or other sources of ignition

- Rats carry leptospirosis which is transmitted particularly through urine

- Scratches and cuts must be covered

- Gloves should be used
- Hands must be cleaned thoroughly with disinfectant before handling food
- Normal procedures for field safety apply

### 7.7. USEFUL CONTACTS

- The information on DNA Sampling has been adapted from a best practice developed by Dr Rachel Fewster, University of Auckland, New Zealand r.fewster@auckland.ac.nz

## 8. MEASURING AND SEXING RODENTS

- This tool provides a field guide to sexing and measuring key features of rodents.
- It is useful to confirm whether breeding is taking place after re-invasion and to confirm the species identification of individual rodents.
- This tool should be used in association with the Guidelines on Rodent Identification.
- Routine recording of body measurements also provides a good check on field identifications and may highlight any records that might be in error.
- **Tools for autopsy and/or measuring**

    1) a pair of vernier callipers
    2) a ruler or tape measure;
    3) accurate spring balances (e.g., Pesola brand) able to weigh up to 500g in 2g steps
    4) a pair of sharp scissors
    5) a pair of forceps;

    - This tool assumes that the rodent under study is dead. Ensure that everyone takes measurements in the same way.

### 8.1. MEASUREMENTS

#### 8.1.1 HEAD-BODY LENGTH (HBL).

- The combined length of the rodent's head and body is known as the head- body length.
- Place the animal flat on its back on a ruler or tape measure.
- Some careful flexing may be necessary with stiff, bent animals.

- Take the measurement in a straight line from the top of the nose to the end of the anus

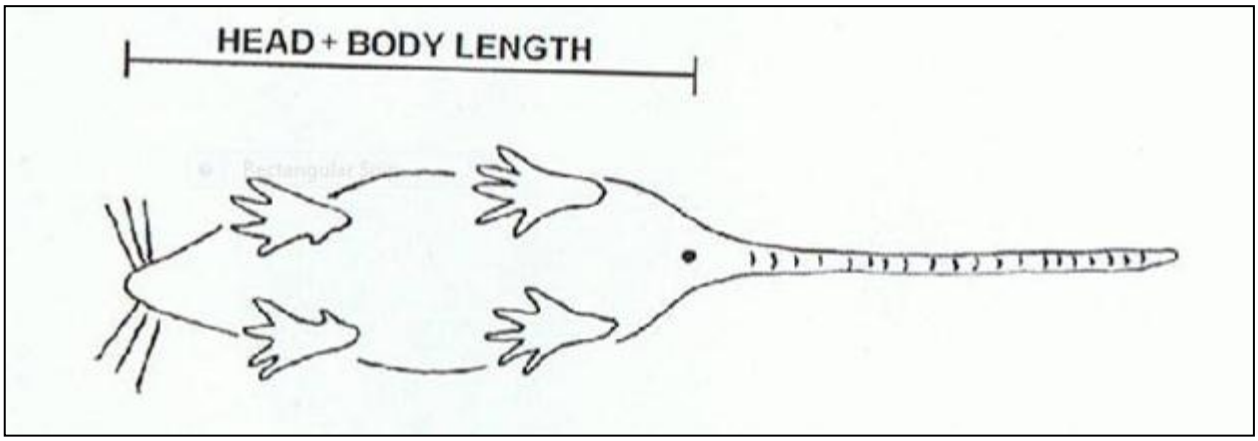


***Figure: Head-Body Length***

### 8.1.2 TAIL LENGTH.

- Measure the tail along a straight line from the middle of the anus to the tip of the tail
- Do not suspend the animal by its tail to take this measurement – the tail will stretch and distort the measurement
- Do not measure damaged tails (these often end with a short pale section that lacks hairs and scales). Make a note of any damage on your recording sheet

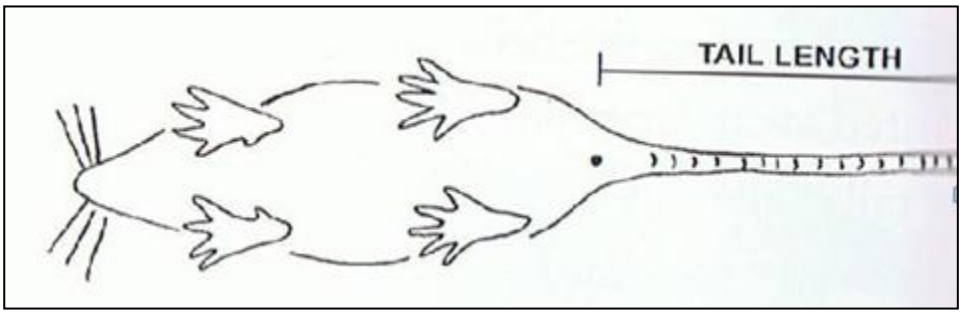


***Figure: Tail length***

### 8.1.3 HIND FOOT

- These are helpful measurements for separating species.
- Measure the pes (hind foot) from the heel to the tip of the central (longest) toe – exclude the claw

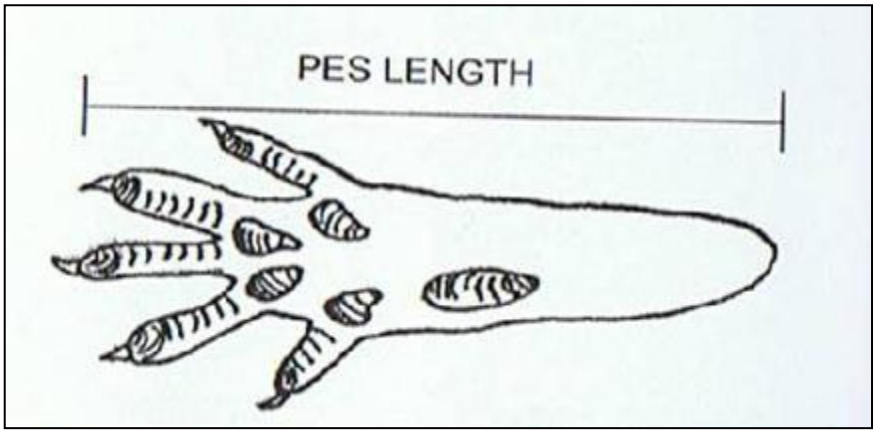


***Figure: Hind foot length***

### 8.1.4. EAR LENGTH

- Measure the ear from the bottom of the notch of the ear to the furtherest point along the rim
- Do not take the measurement if the margin of the ear is damaged. Make a note of any damage on your recording sheet

### 8.1.5 WEIGHT

- Rodents are usually weighed using a calibrated spring balance (such as a Pesola spring balance)
- Be sure to use the right sized balance to fit the rodent species you are weighing
- Check balance before use to make sure it is calibrated to zero (or to the correct mark if it has been adjusted for error)
- Always hold the balance by the swivel ring at the top
- Suspend dead animals by a foot or the tail
- If you are weighing live animals place them in a bag just a bit bigger than the animal
- Weigh the bag by itself as well as weighing the animal – the difference between the two measurements will be weight of the animal
- Note if it is wholly or partly wet as dampness affects the weight.

### 8.1.6 SEX

- Always record the sex of the animal

- Juvenile males and females are sometimes difficult to separate as external sexual features can be similar. However, the distance between the anus and the urethral opening is greater in males than females.
- The vagina in very young females is completely covered with a translucent layer of skin. This appears as a small bald patch immediately to the rear of the urethral opening. See Figure 1.

### 8.1.7 REPRODUCTIVE SYSTEMS

**Females**.

- Record if the vagina is open or closed.
- In very young females the closed condition is usually quite distinct.
- In older animals it is usually obviously open or can easily be opened with a probe.
- This latter condition is still regarded as "open" or "perforate".
- Nipples occur only on females and should be carefully counted when visible; this is especially important with ship rats which often have an extra nipple or two.
- If nipples are large and there is very little hair around each one, check for lactation by attempting to express milk and by examining the development of mammary tissue underneath.

**Males**.

- Note whether or not the testes are "scrotal" (in the scrotum).
- When captured, mature males sometimes retract the testes, but the presence of a dark bald patch on the scrotum is usually a good sign that the testes are normally scrotal.

### 8.1.8 GENERAL

- Always note the colour morph of ship rats because the frequencies of the three morphs can vary. Recording the colour morph can help when there is doubt about identification. Make a brief note of fur condition and any injuries. If you have opened the gut cavity, note the amount of fat around the gut as "none, little, medium or heavy".
- Protect yourself – always wear gloves. When you have finished, WASH YOUR HANDS thoroughly. Some rats carry diseases such as leptospirosis.

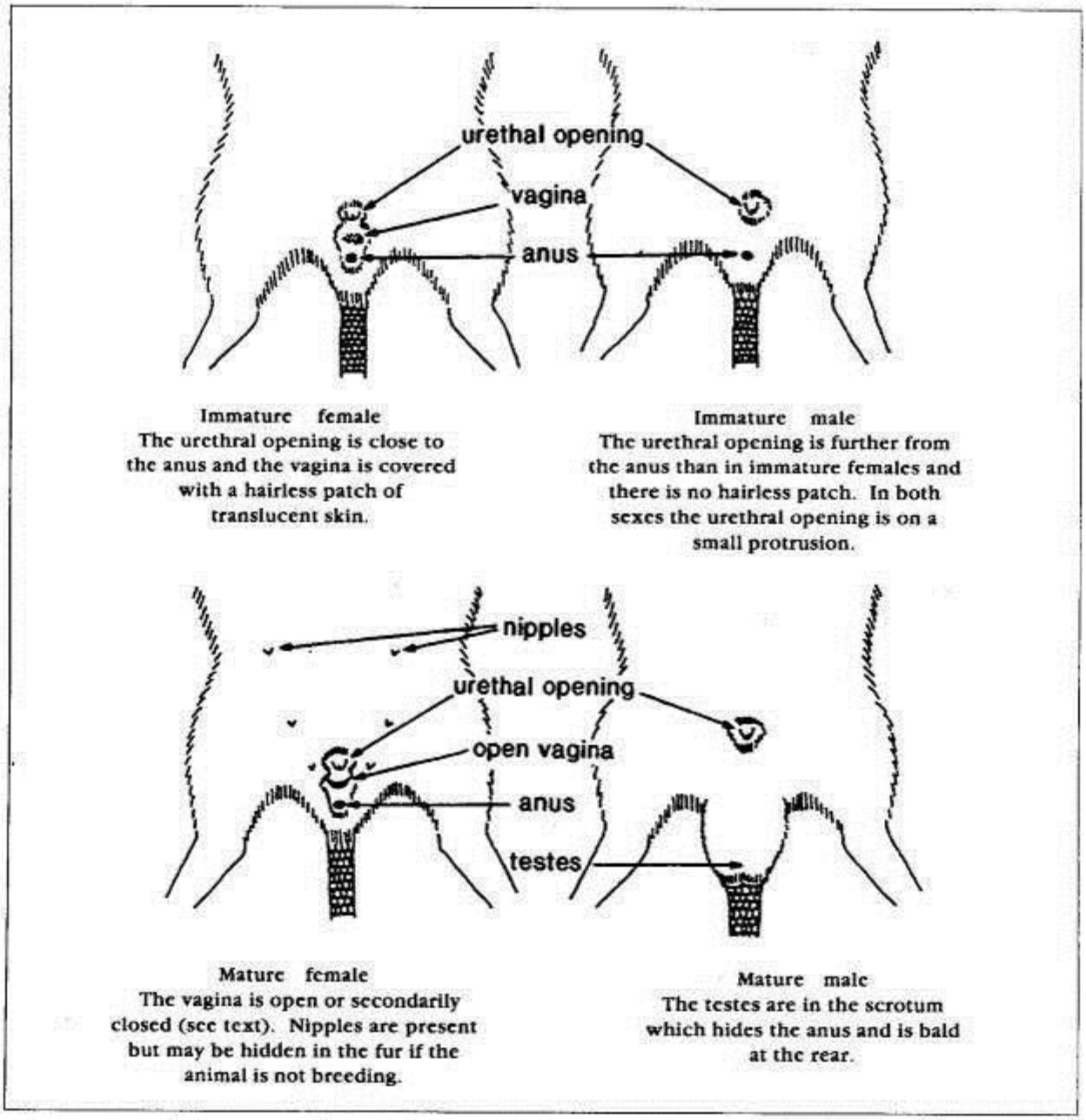


***Figure : Comparison of external sexual features of immature and mature rats.***

### 8.1.9 FURTHER INFORMATION:


- Aplin K.P. et al 2003. Field methods for rodent studies in Asia and the Indo Pacific. Produced for the Australian Centre for International Agricultural Research.
- Cunningham D.M. & Moors J.R. 3rd edition, June 1996. Guide to the identification and collection of New Zealand Rodents Department of Conservation, Wellington, New Zealand.

- PII Resource Kit: Guidelines on Rodent Identification

## 9 ACKNOWLEDGEMENTS


- The Trap Section is based on From: D.M. Cunningham and P.J. Moors, GUIDE TO THE IDENTIFICATION AND COLLECTION OF NEW ZEALAND RODENTS, 2nd Edition, 1993. New Zealand Department of Conservation
- Tracking tunnel photos were sourced from www.wildaboutnz.co.nz
- Tracking tunnel information was sourced from http://www.gotchatraps.co.nz/
- Photos of droppings from Miller, C, 2008
- The Measuring and Sexing section was adapted from Cunningham D.M. & Moors J.R. 3rd edition, June 1996. Guide to the identification and collection of New Zealand Rodents Department of Conservation, Wellington, NZ
- The recipe for chocolate flavoured wax blocks was adapted from Varnham, K. in C. Miller, 2008