Tuatara captive management plan and husbandry manual

THREATENED SPECIES OCCASIONAL PUBLICATION 21

Department of Conservation
Te Papa Atawhai

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Barbara Blanchard, Wellington Zoo, and the Tuatara Recovery Group within the Department of Conservation

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Tuatara captive management plan

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Tuatara captive management plan

ABSTRACT

The management of captive tuatara has, in recent years, played a crucial role in enabling the recovery of relict populations, aiding conservation-related research, and founding new wild populations as well as raising public awareness of these issues.

This document has been produced for the Department of Conservation, the Australasian Species Management Programme, the captive management coordinator, and the holders of captive tuatara. It is a guide to how these animals may best be managed to promote the conservation goals contained in the Tuatara Recovery Plan. It should be read in conjunction with the attached Tuatara Husbandry Manual.

1. Introduction

1.1 TAXONOMY

Cook Strait tuatara	*Sphenodon punctatus* (unnamed subspecies)
Northern tuatara	*S. p. punctatus*
Brothers Island tuatara	*S. guntheri*
FAMILY	Sphenodontidae
ORDER	Sphenodontia (Benton 1985)
CLASS	Reptilia

1.2 CONSERVATION STATUS CLASSIFICATION

IUCN Status	
Vulnerable	*S. guntheri*
Not listed	*S. p. punctatus*
Removed from Red List 1996	*S. punctatus*
CITES (Appendix I)	All tuatara.

Department of Conservation ranking

A: highest priority for conservation	*S. guntheri*
B: requiring work in the short term	*S. punctatus, S. p. punctatus*

1.3 HISTORY IN CAPTIVITY

1.3.1 Cook Strait tuatara

Cook Strait tuatara were first brought into captivity in the late nineteenth century, principally for their curiosity value but also for research. The evidence for this includes the granting of permits appearing in the National Archives and the references cited below. Thomas (1890) had a 'breeding group' in 1885, which apparently were all male. A second group of 12 pairs was added to Thomas's collection in Auckland in 1886.

A tuatara was at the Zoological Gardens in London in 1870 (Sclater 1870), which is probably the first record of the public display of tuatara. In 1890 one was sent to London Zoo. More recent history includes infrequent transfers of mainly pairs to institutions in New Zealand and overseas, so that by the mid-1980s almost 100 were in captivity.

A rapid increase in the number of captive Cook Strait tuatara occurred in 1987, when 133 hatchlings were produced from eggs collected in the wild for breeding ecology studies at Victoria University of Wellington (Thompson 1990; Thompson *et al.* 1991). A second major increase is occurring currently, with over 400 hatchlings (from eggs collected in the wild) being used for behavioural and sex determination studies — also at Victoria University. These animals are intended for eventual release to the wild.

By 2000 there were more than 600 Cook Strait tuatara in captivity.

1.3.2 Northern tuatara

In 1990, northern tuatara from Stanley, Cuvier and Red Mercury Islands were moved from the wild into zoos for captive breeding while rats on the islands were being eradicated.

In 1991 and 1992 eight adults, four of each sex, were captured on Little Barrier Island and kept in captivity for breeding on the island (Whitaker 1993).

1.3.3 Brothers Island tuatara

Tuatara from North Brother were held in captivity during the 1870s (Buller 1877; Newman 1878). The incubation and sex determination experiments with this species were done in three batches (1990, 1991 and 1992) at Victoria University and produced 170 hatchlings in total. These juvenile tuatara passed through a captive phase and were released as 5-year old juveniles to form new wild populations of 50 on Titi Island, in 1995, and approximately 34 on Matiu Island, in 1998. Two permanent populations have been retained in captivity: one at Southland Museum and Art Gallery (SMAG) and the other at San Diego Zoo, USA.

1.4 SUCCESS IN CAPTIVE HUSBANDRY AND BREEDING

Both husbandry and breeding in captivity have proved only moderately successful (Newman 1982). Before the breeding ecology studies of Mike Thompson, which began in 1985, tuatara were usually held in pairs (groups existed at Otorohanga National Kiwi Centre and SMAG). The work of Thompson and associated behavioural research (Gillingham & Miller 1991) suggested that tuatara were more likely to breed when more than one male was present. Cree and Daugherty (1990) reviewed breeding success in captivity from 1952 to 1988, concluding that mortality was high and recruitment poor. A number of possible contributing factors were noted, including high temperature, lack of physical cues for reproduction, lack of social interactions, improper nutrition, small enclosures/high densities and inappropriate conditions for egg incubation. They emphasised the need for a captive management programme to coordinate the activities of different institutions and ensure that they contributed to the conservation of wild stocks.

Since tuatara have a low metabolic rate compared with exotic reptiles, it is inappropriate to transfer such husbandry techniques without modification for the management of captive tuatara. Feeding rates are less and temperature requirements lower, for example, and failure to observe such differences can lead to stress (Goetz

& Thomas 1994) and a reduced lifespan. Holders outside New Zealand tended to keep their tuatara too warm (Newman *et al.* 1979).

Exact dietary and ultraviolet light needs are unknown (Thompson *et al.* 1988).

Although the Department of Conservation was a point of reference for holders, who provided an annual report on events through the year, there was little sharing of information. No reports were published, but some feedback was given to holders on matters of key interest such as the survival of juvenile tuatara in captivity. Since the formation of the Tuatara Recovery Group and the appointment of a captive coordinator in 1989, information has been more widely available.

Breeding has been regular only with certain pairs of Cook Strait tuatara and one group. A pair at each of Auckland (Tintinger 1987) and Wellington Zoos (Blanchard 1988), the National Wildlife Centre plus an outdoor group at the National Kiwi Centre have produced young over about 15 years (Fingland 1986). Similar success has been possible with Cook Strait tuatara at SMAG (Hazley 1993) where biennial laying occurred in 1985–90 and instances of annual laying have occurred since then. The captive population of Cook Strait tuatara is not yet self-sustaining.

At SMAG a group of seven juvenile Brothers Island tuatara is intended to form the only captive breeding population in New Zealand. They are now 9 years old.

There is no intention to hold Northern tuatara captive in the long term, but breeding success with those held temporarily from Cuvier, Red Mercury and Stanley Islands has increased the former two captive populations by over 100%. Tuatara from Stanley Island have produced only four offspring in 9 years, but they are now held in an environment suitable for breeding. Tuatara from Cuvier and Stanley Islands remain in captivity for the medium term for breeding to further augment the wild populations. All Red Mercury adults have been returned to the source island.

1.5 BIOLOGY OF CAPTIVE MANAGEMENT

1.5.1 Generation level

Second-generation captive-bred tuatara reproduced during 2001 at SMAG, Victoria University and Peacock Springs.

1.5.2 Reproductive rates

Tuatara on Takapourewa (Stephens Island) have been the most intensively studied and are used as the basis for management advice.

Tuatara on Takapourewa reach sexual maturity at 11–13 years and 170–180 mm snout–vent length (SVL), but on Lady Alice Island maturity occurs at 8–10 years old and the same SVL (Newman *et al.* 1994). In both instances it is probable that males do not have mating opportunities until they reach approximately 240 mm SVL (Gaze 2001).

On Takapourewa, vitellogenesis is prolonged (Cree *et al.* 1992), and females can reproduce only every 2–4 years. In captivity this can reduce to 1 year, as experienced at SMAG and on Little Barrier Island. The generation time is usually regarded as 30 years.

1.5.3 Mortality rates

As with most reptile species in captivity, death rates are relatively high early in life (Heatwole & Taylor 1987). Once 120 mm SVL and 4–5 years of age is attained, tuatara are regarded as more able to survive in the wild. The mean survival of adults in captivity in New Zealand from 1952 to 1995 was 7.41 years, based on a sample of 63 tuatara (Table 1). Overseas it was 7.6 years, but the mean was influenced by two long-lived males (27 and 35 years). Survival time in captivity has improved over the last decade with better husbandry techniques (Table 2). In 1996 the mean survival time for all adult Cook Strait tuatara in captivity had increased to 17.9 years.

Since there are more hatchings than deaths, the population can be said to be growing, although only a limited number of adults are breeding. This means that a few gene lines are well represented in comparison with many captive adults which have no descendants yet (i.e. are potential founders).

TABLE 1. SURVIVAL OF ADULT COOK STRAIT TUATARA (*S. PUNCTATUS*) IN CAPTIVITY 1952–95, IN YEARS.

	IN NZ	OVERSEAS
Females	11.6 (n=8)	5.7 (n=12)
Males	5.3 (n=6)	14.2 (n=9)
Unknown sex	5.3 (n=10)	2.9 (n=18)
All	7.4 (n=24)	7.6 (n=39)

(Data from International Zoo Yearbook, National Archives and tuatara holders, representing only those tuatara for which definite arrival and death dates were available.)

TABLE 2. SURVIVAL DATA OF ADULTS SINCE 1990, WHEN DATA COLLECTION FOR STUDBOOKS BEGAN.

SPECIES	SEX	NUMBER	DEATHS	TOTAL SURVIVING
S. punctatus[1]	Female	16	5	11
	Male	21	3	18
S. p. punctatus[2]	Female	22	1	21
	Male	17	2	15
S. guntheri	Female	1	0	1

[1] includes tuatara outside New Zealand
[2] not divided into island populations

1.5.4 Sex ratio of captive animals

The ratio of males to females was equal with captive Northern tuatara (before release to the wild), but is skewed in favour of the males with Cook Strait tuatara (Table 3). Tuatara of unknown sex are all juvenile (<13 years old).

TABLE 3. SEX RATIO AND AGE DISTRIBUTION.

SPECIES	> 13 YEARS OLD		< 13 YEARS OLD		UNKNOWN
	MALE	FEMALE	MALE	FEMALE	SEX
S. punctatus[1]	21	15	13	40	88
S. p. punctatus[2]	13	13	0	0	51
S. guntheri[1]	0	1	0	0	19

[1] includes tuatara outside New Zealand
[2] not divided into island populations and excluding Red Mercury adults, now back on source island

Not all captive tuatara of breeding size are breeding (Table 4).

TABLE 4. BREEDING ACTIVITY.

SPECIES	EGGS PRODUCED FROM FEMALES[1]	ABLE TO BREED NOW (ON SIZE, GROUPING)	
		MALE	FEMALE
S. punctatus	8	14[2]	11[2]
S. p. punctatus	8	13	13
S. guntheri	0	0	0

[1] none from females outside New Zealand
[2] includes tuatara outside New Zealand

1.6 ORIGINS OF EXISTING STOCK (WHERE KNOWN)

Sphenodon punctatus

All founders and potential founders originated from Takapourewa.

Sphenodon punctatus punctatus

Adults from Cuvier Island and Stanley Island remain in captivity, where they have produced young. Adults from Red Mercury have produced young and they have been returned to the island. Tuatara from Little Barrier are enclosed *in situ* and have produced young while in captivity.

Sphenodon guntheri

All are of North Brother Island origin. One adult female only is in captivity while the juveniles are from eggs taken from females which remain in the wild on the island.

In 2002, 17 holders in New Zealand and 7 holders overseas have over 800 tuatara. See Appendix 1, p. 23 for details.

2. Captive management within the tuatara recovery plan

The Tuatara Recovery Plan (Gaze 2001) establishes goals and objectives for the conservation of tuatara over the next 20 years; this section describes how captive tuatara will be managed to help meet those objectives.

The long-term goal of the Tuatara Recovery Plan is:

The genetic diversity of tuatara will be maintained by returning all existing populations to their natural levels and establishing new wild populations of tuatara throughout their pre-human range as components of healthy ecosystems for the benefit of all.

It is the three shorter-term objectives of the Tuatara Recovery Plan that are the focus of this management plan for captive tuatara. Each objective has an action (the original numbers are reproduced here) that must be supported through the management of animals in captivity.The role of captive animals in assisting identified research needs is also addressed.

Objective 1 The genetic diversity of all existing tuatara stock is preserved

Action 1.7 *Enhance the recovery of relict populations using captive breeding.*

This action is being met by the work of Auckland and Hamilton Zoos and Victoria University in holding and breeding animals from Stanley and Cuvier Islands, and Red Mercury Island (none in captivity in 2002) while introduced mammals were being eradicated.The intention was to return all *S. p. punctatus* to the wild once rats (and rabbits from Stanley Island) were removed (Gaze 2001). Auckland Zoo, however, requested that they keep the Cuvier Island adults to encourage further breeding to augment the most endangered population of the subspecies.The Recovery Group agreed (April 1999).

The Stanley Island animals have been slower to produce young than the other island groups and are being kept in captivity until in larger numbers before re-introduction.

Objective 2 Tuatara are reinstated as components of healthy ecosystems throughout their pre-human range

Action 2.5 *Establish sustainable captive populations of tuatara from existing stock. The technique of obtaining eggs from wild tuatara, artificial incubation of the eggs and captive raising of the young will be used as a tool in the establishment of new populations.*

To achieve this action it is necessary to retain the separate identity of island

populations, even if they are of the same subspecies or species. To retain maximum genetic diversity, inbreeding needs to be avoided by dispersing over-represented lines to make more places available for less common gene lines. At present, this concerns only those juvenile Cook Strait tuatara bred at Auckland Zoo and SMAG. The juvenile Cook Strait animals raised at Victoria University (Thompson 1990) is the largest group approaching breeding age and should be able to be grouped for that purpose, being of similar age and size.

If breeding can be increased, there should be no need to import more animals from the wild. Institutions with poor performance in breeding tuatara (e.g. Wellington Zoo) need to be identified and means to improve performance introduced. The following mechanisms will be used to achieve this:

- Tuatara compatibility should be greatest if the group comprises animals of the same size, to reduce dominance problems. There should be more than one male in a group, because male interaction probably has significance in breeding behaviour (Gaze 2001). Compatibility and breeding can also occur with a solitary pair, however.
- Soft tissue X-ray is used by Auckland and Hamilton Zoos and SMAG to look for shelled eggs each spring in the oviducts of adult females with no known deleterious effects (Newman & Watson 1985; Thompson *et al.* 1998).
- Hormonal induction of egg-laying (Thompson 1990; Thompson *et al.* 1991) provides a means of intercepting eggs to maximize hatchling production. Once a natural nest is made, it is very difficult to locate, and the eggs may already be dehydrating, or damaged by ground invertebrates, if the area is not optimal. Close observation to intercept eggs immediately after natural egg-laying (for artificial incubation) is the preferred option.
- Reptiles that have been raised in captivity have been found to have a better chance of surviving in the wild if they are older than hatchlings. Tuatara are not released until they are about 5 years old, and/or 120 mm SVL.

There may be advantages in ceasing additions to overseas subpopulations of *S. punctatus* for at least the next 5 years in order to retain as much diversity in New Zealand until the population has more descendants. This restriction on sending additional tuatara overseas will then be reviewed. It is probable that only Berlin and Chester Zoos will be self-sustaining once breeding commences (nine and eight tuatara, respectively), but the North American holdings are unlikely to be sustainable without exchange among the three: St Louis Zoo has three male and two female adults, and Dallas and Toledo Zoos have five and two animals hatched in 1987, of unknown sex respectively.

There are no adult tuatara in Australia, but there are three 14-year-old females at Taronga Zoo (Sydney) which hatched in New Zealand in 1987.

Although Otorohanga National Kiwi Centre has had a group of *S. punctatus* since the early 1970s, producing young since approximately 1984, the husbandry of juveniles should be improved. If conditions at this facility improve it would be appropriate to augment production and extend the bloodline beyond the parent group.

The *S. guntheri* juveniles at San Diego Zoo were too small for laparoscopy when they were exported, but sexed tentatively on the basis of incubation temperature. SMAG's animals were sexed by laparoscopy in 2001. The two permanent captive populations of *S. guntheri* (at SMAG and at San Diego Zoo) need to be self-sustaining, but San diego Zoo may require additional animals to do so if sex ratios are skewed.

Objective 3 Public awareness of tuatara and related conservation issues will be promoted through accessibility to captive animals and certain wild populations

Action 3.2 *Knowledge of tuatara and their conservation will be promoted through the appropriate use of captive tuatara.*

The purposes for which each institution are holding tuatara are listed in Appendix 1. Where animals are being held for public awareness purposes it is important that the holder is clear on just what the Department of Conservation's expectations are in this regard. Any such requirement should be a condition of the permit to hold the animals. The Department, for its part, has a role in providing good information, and where practical, advice on display and educational opportunities. Holders vary considerably in availability of resources (animals, displays, staff, funding), but it should be possible to develop programmes for advocacy and education within the capability of each institution, and/or increase its resources.

Welfare of tuatara used for these purposes is important, and guidelines have been produced as Appendix 2. These may need to be refined for each holder or programme.

2.1 RESEARCH PRIORITIES

2.1.1 To obtain new knowledge of tuatara through research

The recovery plan does not have an objective directly related to research, which is treated in a separate section (Gaze 2001, p. 25–26). There is much interest in scientific research on tuatara, which is expected and appropriate given the special taxonomic and biological position tuatara have within the reptile fauna. Research needs, specifically related to conservation, are listed in the recovery plan; however, there is also an international responsibility to foster less-applied research where it can be done without ill effects on conservation of tuatara. Opportunities for much of this research exist within the captive holdings.

3. Workplan

This section describes the tasks involved in achieving each action and how the progress in achieving this will be measured.

3.1 OBJECTIVE 1 TO PRESERVE THE GENETIC DIVERSITY OF ALL EXISTING STOCK

Action 1.7 To use captive breeding facilities and expertise to enhance the recovery of relict populations after habitat restoration.

Workplan

Returning all *S. p. punctatus* to the wild, except for most Cuvier Island adults, will require:

1. Transfer all juvenile tuatara from Red Mercury Island at the National Kiwi Centre (Otorohanga) and at Auckland Zoo back to a suitable site on Red Mercury Island. (This has already been achieved: there are no adults remaining in captivity.)

2. Retention of adult tuatara from Cuvier Island at Auckland Zoo, but allowing some exchange of captive adults with other wild adults to augment numbers and diversify the gene pool.

3. Return of juvenile tuatara to Cuvier Island when they are of a suitable age.

4. Retention and breeding of tuatara from Stanley Island at Auckland and Hamilton Zoos to increase the population. When there are sufficient animals of a suitable age, both adults and juveniles should be returned to Stanley Island. Any adults regarded as surplus before then should be returned to Stanley, and must include one female.

Timeframe

The actions which need to be taken for these islands should be clearer in 2004, when Cuvier production will be known and the tuatara from Stanley Island have had time to fit into their new group. This new group was formed by moving animals from Wellington to Auckland Zoo in May 1999, others are still held at Hamilton Zoo.

Criteria for assessment

In the short term (5 years) the following will be considered as indices of success:

- Number of hatchings increases overall and number of deaths decreases.
- All tuatara from Red Mercury are returned to the wild (already completed).
- Captive tuatara from Cuvier Island continue to increase in numbers.
- The number of tuatara from Stanley Island in captivity increases by at least 100%.

In the medium term (10–15 years) the following will be considered as indices of success:

- All tuatara from Stanley Island are returned to the wild.
- Sufficient tuatara from Cuvier Island exist in captivity to begin restocking the island, or creating a new wild population.

3.2 OBJECTIVE 2 TO RESTORE TUATARA AS PART OF HEALTHY ECOSYSTEMS

Action 2.5 To establish sustainable captive populations of tuatara from existing captive stock and to use captive breeding facilities and expertise to raise juvenile tuatara suitable for establishment of new wild populations.

Workplan

1. During 2002 the coordinator organised four new potential breeding groups, with two still to be completed, as Peacock Springs and Queenstown Kiwi and Birdlife Park have still to receive animals from Napier Aquarium and Otorohanga. Further moves are planned beyond 2002.

2. San Diego Zoo has eight of the original group of 10 *S. guntheri* alive, and SMAG has seven from 10. Both groups, without further loss, would be large enough for a permanent breeding population unless the sex ratio is skewed severely.

3. SMAG may have two males and five females (2.5) and San Diego Zoo may have 3.5.The San Diego Zoo animals have been housed individually but should be moved into a group situation. It may be necessary to supplement each group or exchange animals between them if sex ratios are badly skewed—although overseas exchanges should be avoided if possible.

4. Victoria University of Wellington should assist with artificial incubation as required, but limit its involvement to increasing the genetic diversity of captive stock of *S. punctatus* and the separate island populations of *S. p. punctatus.*

5. Other holders should incubate any eggs their animals produce, unless they are from an over-represented line, in which case they may be sent to Victoria University for freezing at –80°C for future research, or left in natural nests.The purpose of the latter is to give holders experience at finding natural nests or, by leaving the nest(s) undisturbed, to collect figures on hatch rate in captive situations (e.g. as at the National Wildlife Centre, spring 1999, with Auckland Zoo's breeding pair of Cook Strait tuatara).

6. Wellington Zoo is to be encouraged to allow grouping of Cook Strait tuatara outdoors for breeding.

7. Stocktaking of the Cook Strait tuatara in the outdoor enclosure (Rotary Park) at the National Kiwi Centre should be done during 2001/02, then decisions made on future management.

8. Consideration will be given to the X-raying of adult female tuatara in potential breeding situations in October of each year to enable preparation for safe incubation (Thompson *et al*. 1998).

9. Induction should be considered in captive tuatara from small island populations (Thompson *et al.* 1991), but with reference to the improved success possible in carefully managed wild-laid clutches.

10. Juveniles intended for release into the wild must be retained in captivity until they grow to a suitable size before release (head starting). Institutions suited to head

starting are Nga Manu, Auckland Zoo, National Kiwi Centre, Hamilton Zoo and Little Barrier Island. Other institutions should be able to raise their own hatchlings, and SMAG should continue to send head-started juveniles to Peacock Springs for growing on.

Timeframe

As detailed above. Those without specific time frames should be done on an as-needed basis until the next review of the this plan.

Criteria for assessment

In the short term (5 years) the following will be considered as indices of success:

- Number of hatchings increases overall and number of deaths decreases.
- Studbooks are upgraded yearly, using SPARKS software.
- Stable breeding groups are established and breeding behaviour observed, leading to the production of offspring equivalent in numbers to approx. 15% of the total captive population.
- *S. guntheri* are established in two stable groups.
- Captive-raised juveniles from Takapourewa are ready for release to form a new wild population.

In the medium term (10–15 years) the following will be considered as indices of success:

- SPARKS population analyses become possible as the population becomes less skewed.
- Sufficient *S. punctatus* juveniles are available for a new wild population.
- 25% of the female tuatara raised from eggs taken from Takapourewa in 1986–87 have produced eggs.
- No immigration is required to maintain populations of *S. guntheri*.

3.3 OBJECTIVE 3 TO PROMOTE PUBLIC KNOWLEDGE OF TUATARA AND TO ADVOCATE FOR THEIR CONSERVATION AND PROMOTE PUBLIC AWARENESS THROUGH ACCESSIBILITY

Action 3.2 To encourage the use of captive tuatara for promoting knowledge of the species and their conservation.

Workplan

1. Assess what advocacy material is currently held by institutions and what needs there are for updating this material.

2. Ensure that new and existing permits to hold tuatara for public awareness purposes adequately reflect the expectations of the Department and the holder and are consistent with standards in the Tuatara Husbandry Manual (Blanchard *et al.* 2002).

3. Standardise the advocacy information that is made available to holders.

4. Tailor holder obligations to their resources.

5. Encourage the use of captive animals for temporary displays but ensure that clear guidelines are followed for handling and care of the animals; such use must be supported by high-quality educational material.

Timeframe

1. Coordinator to request conservancy staff to visit local holdings by mid-2002 and to assess the supply of information, the quality of current displays and the need for better facilities.

2. Written resource material and access to photographs provided for all holders by late 2002 where this is considered necessary.

Criteria for assessment

In the short term (5 years) the following will be considered as indices of success:

- Holders to function satisfactorily as advocates/educators (assessed by the Department of Conservation).
- Public knowledge of tuatara to have a wider base with the provision of information at all conservancy offices and public facilities which keep tuatara.

In the medium term (10–15 years) the following will be considered as indices of success:

- Advocacy and educational materials to be revised, amended and distributed as required.

3.4 OBJECTIVE 4 TO OBTAIN NEW KNOWLEDGE OF TUATARA THROUGH RESEARCH

In facilitating the research identified in the Tuatara Recovery Plan (Gaze 2001, p. 25–26) and the increasing use of captive animals for this purpose, it is the role of the coordinator to:

- Ensure that the research will not conflict with the overall captive management regime.
- Ensure that disruption to holders is minimised through careful matching of research to the most suitable holding and through consultation with the relevant conservancy in obtaining the necessary authorisation.

4. Review of management plan for captive tuatara

This document will be reviewed annually in May by the coordinator and recovery group leader. Any recommended changes will be circulated to recovery group members for endorsement and then to the Department of Conservation Regional General Manager (Central) for approval. Substantive changes will be notified to licensed holders.

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Appendix 1

CURRENT HOLDING BY CAPTIVE INSTITUTIONS AND THEIR ROLE IN THE MANAGEMENT PLAN FOR CAPTIVE TUATARA

CURRENT HOLDINGS AND AGE OF TUATARA: MALE (M), FEMALE (F) AND UNKNOWN (U)

HOLDER	TYPE OF HOLDING	STOCK (M:F:U)	AGE >13YR	<13YR
Cook Strait tuatara *S. punctatus*				
Chester Zoo	Breeding group	3:5	–	3:5
St Louis Zoo	Breeding group	3:2	3:2	–
Berlin Zoo	Breeding group	4:5	–	4:5
Dallas Zoo	Breeding group	0:0:5 (1?:4)	–	0:0:5
Toledo Zoo	Advocacy/display	0:0:2	–	0:0:2
Taronga Zoo	Advocacy/display	0:3	–	0:3
Kevin Evans	Holding/head start interest	0:3	–	0:3
Auckland Zoo	Advocacy	0:0:4	–	0:0:4
Napier Aquarium	Breeding possible	1:4	1:4	–
Otorohanga Kiwi House	Breeding group outdoors, juveniles head-started indoors	4:4:9	3:2	1:2:9*
National Wildlife Centre, Mt Bruce	Advocacy/display/have bred (ex Auckland Zoo)	1:3	1:1	0:0:3
Wellington Zoo	Advocacy/display/have bred (Nocturnal House)	1:1	1:1	–
	Breeding/research (outdoor group 1) have bred	2:2	2:2	–
	Breeding /research (outdoor group 2)	1:3:1	1:0	0:3:1
Peacock Springs	Potential breeding group/divided/ outdoors	9:1:25	9:1	0:1:25
Reptile Research Centre (Goetz, Nelson)	Research	4:6	1:1	3:5
Nga Manu Sanctuary	Advocacy/display/ breeding potential	2:3	–	2:3
Victoria University	Advocacy/display/breeding/research	2:2:432	2:2	0:0:432
Hamilton Zoo	Advocacy/display	1:0	1:0	–
Orana Park Wildlife Trust	Advocacy/display	3:1	3:0	1:1
NZ Conservation Trust (Willowbank)	Advocacy/display	0:0:11	–	0:0:11
Pouakai Wildlife Trust	Advocacy/display	0:1	–	0:1
Rainbow Springs	Advocacy/display	2:3	–	2:3
Southland Museum and Art Gallery (SMAG)	Advocay/display/ Have bred	4:3:53	3:3	1:0:53*
	Totals	**c. 644**	**47**	**597**

* *Records incomplete*

HOLDER	TYPE OF HOLDING	STOCK (M:F:U)	AGE >13YR	<13YR
Northern tuatara *S. p. punctatus*				
Auckland Zoo	Breeding Cuvier I.	4:2:10	4:2	0:0:10
	Breeding Stanley I.	3:2	3:2	–
Otorohanga Kiwi House	Head starting Red Mercury I.	0:0:4	–	0:0:4
Hamilton Zoo	Breeding/Head starting (juveniles indoors) Stanley I.	5:2:4	5:2	0:0:4
	Head starting Red Mercury I.	0:0:10	–	0:0:10
Little Barrier I.	Breeding/Head starting (juveniles apart from adults)	4:4:80	4:4	0:0:80
	Totals	**134**	**26**	**108**
Brothers tuatara *S. guntheri*				
Southland Museum & Art Gallery	Breeding potential	0:0:7 (3?:4)	–	0:0:7
Victoria University	Advocacy/display	0:1:52	0:1	0:0:52
San Diego Zoo	Breeding potential	0:0:8	–	0:0:8
	Total	**68**	**1**	**67**
Total tuatara in captivity:			**c. 845**	**–**

** Records incomplete*

Appendix 2

PROTOCOL ON USING TUATARA FOR TEMPORARY DISPLAY

Live tuatara on display can be an excellent complement to any presentation on conservation, but should occur only where advantages to the effectiveness of the presentation are clear. It is important that the display is carefully planned to ensure that the security, welfare and dignity of the animal is maintained. The following protocol should be followed:

Selection criteria for tuatara for temporary display

If there is a choice, captive tuatara already on public display and accustomed to some handling are the most appropriate.

Daycare

Two experienced people need to be responsible for the well-being and handling of the tuatara. Staff involved should read information on tuatara care, behaviour and biology before participating in the project. (See *Tuatara husbandry manual*: Blanchard *et al. 2002*, and for a brief biology refer to the *Tuatara recovery plan*: Gaze 2001.)

Security

Any tuatara being taken from its approved captive holding must be kept in sight at all times by at least one of the two persons responsible for its care.

Siting of display

If possible, place the exhibit no lower than adult waist height and with a buffer zone between the cage and the public of at least 1 metre.

Escape cover and public viewing

Vegetative cover should be arranged so the animals can retreat enough to feel secure. If the animals choose to be completely out of sight, you may need to coax them out for viewing periodically, choosing a different animal each time. This should be done no more than four times each day, and for a maximum of 20 minutes each time. Breathing frequency is an indicator of stress. When a tuatara's breathing becomes more frequent than one inspiration and expiration approximately every 10 seconds, disturbance (i.e. display) should cease. If a tuatara, particularly an adult, panics and runs into the enclosure wall, nose damage could result. The animal should be placed under cover to calm down. To reduce stress, ask viewers to be quiet and to rely on natural light for photography.

The tuatara should be removed from the cage only when the audience is small and quiet. An animal should not be held for more than 10 minutes' duration. Most animals feel more secure if all their feet are stable on a non-slip surface when being held for display, and none should be tipped upside-down. It is possible to calm a tuatara by holding it vertically by the shoulders and stroking the undersurface with a finger firmly several times, from chest to vent.

Try to alternate the individual tuatara handled for display to avoid over-stressing any one animal.

Do not place adult tuatara with juvenile tuatara or any other smaller display animal.

Food

Food will not need to be provided during the day for tuatara, but fresh leaf litter with small live invertebrates may encourage them to be more active or visible.

Water

If conditions are dry, the interior of the cage(s) should be mist-sprayed at 8 a.m., 12 p.m. and 3 p.m., to the point where water is beginning to drip from the foliage and everything is damp (like a heavy dew). Spray once a day as a minimum.

Provide a shallow water dish (minimum depth 1 cm) large enough for a tuatara to rehydrate via the cloaca, if required, by sitting in the water.

The water should be changed daily. Tuatara quite often defaecate in water.

Heat

There should not be any direct sun on the cages, which should not be near a heater. A thermometer should be in the cage, and the temperature should not be allowed to rise above 25°C (fatal to tuatara) and should not drop below 5°C.

Appendix 3

PERMITTING CONSIDERATIONS IN THE MANAGEMENT OF CAPTIVE TUATARA

All holders of tuatara must be licensed under Section 53 of the Wildlife Act 1953. It is the responsibility of the Biodiversity Unit in the conservancy to ensure that this is done and that conditions are adhered to. These permits should be prepared in consultation with the Captive Management Plan Coordinator (CMP coordinator) and the leader of the Tuatara Recovery Group. It is in the interests of all parties that the permit addresses the following issues:

1. The purpose for which the animals are being held.

2. A description of the site to which the permit applies.

3. Expectations with regard to physical security of the animals.

4. The term of the permit, bearing in mind that the holder usually needs some indication of 'tenure' given the expense of preparing facilities and the service they provide to the Department of Conservation's management of tuatara.

5. The expectation, or otherwise, for public access.

6. The expectations of both the holder and the Department with regard to the quality and the preparation of interpretation material.

7. The responsibility of the holder to meet the standards set out in the Tuatara Husbandry Manual.

8. The responsibility of the holder to report to the CMP coordinator on the welfare of the animals.

9. An understanding that these animals are held as part of the wider programme for captive management of tuatara and that some shifting of animals between holders may be necessary to meet objectives.

10. A description of just what animals are held and their provenance.

11. A condition stating that no tuatara can be disposed of other than to an officer of the Department or to another authorised holder, and only if so advised by the CMP coordinator—and that this authority permits such transfers.

12. Whether or not animals may be removed for educational purposes and by whom.

13. Any requirement for individual marking of the animals.

14. Whether or not the animals may be available for research purposes.

Appendix 4

REPORTS FROM TUATARA HOLDERS TO CAPTIVE MANAGEMENT PLAN COORDINATOR

Interim reports are to be made as required to the CMP coordinator and recovery group leader.

Data returns (example of blank data sheet follows) may have questions appended.

The intention is to collect growth data before and after the growing season. In the Southern Hemisphere this is in spring (October) and autumn (April); Northern Hemisphere holders should adapt returns to their seasons.

Remit the most recent morphometric data each return, but indicate the date collected on the data sheet.

This information is entered into the DataPerfect database maintained on all captive tuatara. Life events go into SPARKS (software) and/or studbooks. With different identification systems used by holders, several computer programmes involved and with over 700 animals, it is important that the enclosed datasheet is used for returns, rather than ARKS (software) printouts or graphs for instance.

TO BE INSERTED

Appendix 5

ROLE OF THE CAPTIVE MANAGEMENT PLAN COORDINATOR

To maintain:
- studbooks
- SPARKS and DataPerfect databases
- records of holdings
- list of surplus animals.

To collect the following data:
- morphometric (weight, linear measurements)
- hatching.

To send:
- newsletters to holders and involved conservancies every 6 months to communicate new information
- data collection sheets to holders for returns in April and October each year
- to researchers collected data as requested.

To report:
- to the Recovery Group annually
- to the Conservation Management Group annually.

To assist with the review of:
- Husbandry manual
- Captive management plan
- Recovery plan.

To participate in:
- Recovery Group meetings
- Conservation Management Group meetings.

To advise on:
- medical problems
- who to contact about problems
- interpretation of husbandry maubnal to holders and conservancies.

Tuatara husbandry manual

THREATENED SPECIES OCCASIONAL PUBLICATION 21B

Barbara Blanchard, Wellington Zoo, and the Tuatara Recovery Group within the Department of Conservation

June 2002

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Wellington, New Zealand

1. Introduction

This husbandry manual has been prepared for all holders of tuatara within and outside New Zealand. It is a companion document to the Tuatara Recovery Plan 2001–2011 (Gaze 2001) and the Tuatara Captive Management Plan (Blanchard *et al*. 2002), and is designed as a reference standard for tuatara management in captivity.

As we learn more about tuatara, the information contained in this husbandry manual will require updating and therefore should be reviewed in 5 years.

1.1 TAXONOMY

Family	Sphenodontidae
Order	Sphenodontia
Class	Reptilia

Tuatara are represented by three taxa (Daugherty *et al.* 1990):

Northern tuatara	(*Sphenodon punctatus punctatus)*
Cook Strait tuatara	(yet undescribed subspecies of *S. punctatus*)
Brothers tuatara	(*Sphenodon guntheri)*

1.2 CONSERVATION STATUS

Conservation status as declared by the International Union for the Conservation of Nature (IUCN), Department of Conservation (Molloy & Davis 1994; Molloy *et al*. 2001; Hitchmough in press) and the Convention on International Trade in Endangered Species (CITES) of Wild Flora and Fauna are shown in Table 1.

TABLE 1. CONSERVATION STATUS OF THE THREE TUATARA TAXA.

SPECIES	2002 DOC PRIORITY (MOLLOY ET AL. 2001; HITCHMOUGH IN PRESS)	DOC PRIORITY (MOLLOY & DAVIS 1994)	IUCN	CITES
S. p. punctatus	Sparse, ST(stable), HI (human induced loss of range)	Category B	Not listed	Appendix I
S. punctatus 'Cook Strait'	Range restricted, ST, HI	Category B	Removed from Red List 1996	Appendix I
S. guntheri	Nationally endangered, ST	Category A	Vulnerable	Appendix I

1.3 CAPTIVE MANAGEMENT CO-ORDINATOR

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2. Identification method

2.1 INDIVIDUAL IDENTIFICATION

Adult animals may be identified by individual appearance, by toe clips or by electronic microchip (transponder) implants with unique numbers. The National Kiwi Centre at Otorohanga has successfully tagged adults with coloured beads on wire, fixed through the crest.

Implants

The preferred location for implants of transponders in juveniles is in the ventral tail muscles, on one side of the midline approximately 2 cm post-vent. Other sites tested on small animals in limited trials proved insecure (Green 1993). Auckland Zoo successfully inserts transponders subcutaneously in the loose skin of the inguinal area of adults. A group of Northern tuatara implanted in 1990 are still able to be scanned.

Toe clipping

Toe clipping is not favoured by iwi (Maori people) as it is considered too disfiguring and culturally insensitive. This technique is being replaced by the use of passive transponders wherever possible, however, there are still some problems with using transponders in juveniles. Microchipping is being tested for wider use. Until technology is available to overcome these problems, the use of toe clips may be necessary in a limited number of situations. A special case will need to be made and approved before toe clipping is undertaken. Alternative means of marking will be used wherever possible.

The Trovan system has been approved for world-wide use by the Captive Breeding Specialist Group (CBSG) of the IUCN and in New Zealand the equipment is available from the National Central Animal Registry. Contact initially the Science & Technical Centre at the Department of Conservation (Appendix 1). Allflex transponder equipment is held by the Tuatara Recovery Group.

Juveniles

Juvenile tuatara are best identified by unique toe clips. It is also possible to mark them individually with a small dab of nail polish/correction fluid until they are large enough to insert microchip implants, but care must be taken to ensure identities are not confused. The appropriate toe clip pattern for new hatchlings can be obtained from the Department of Conservation, making initial enquiries at Science, Technology & Information Services (contact: Herpetofauna Database: Benno Kappers, Otago Conservancy, Department of Conservation, Dunedin).

2.2 SEXING METHODS

Adult tuatara are sexually dimorphic. Males (up to about 1000 g) are approximately twice as heavy as females (300–500 g) with a more triangular head when compared to the more rectangular head of the female.The nuchal (neck) and spinal crests are larger in males.

Juvenile tuatara may be sexed laparoscopically at less than a year old (Nelson 2001).

Cree (1992) sexed 13 wild juveniles larger than about 115 mm snout–vent length correctly using width and length of the spines in the crest, snout length, throat colour, abdomen width and posture (males heads are held up) after sexing 22 juveniles by laparoscopy.

3. Natural history

3.1 DISTRIBUTION

Tuatara are currently found on 33 offshore islands although sub-fossil evidence indicates they were previously found throughout New Zealand. The three recognised tuatara taxa are the only extant representatives of the order Sphenodontia (Benton 1985). Tuatara populations in the wild were significantly reduced following the introduction of rat species (*Rattus exulans, R. rattus,* and *R. norvegicus*) to a number of islands and to the mainland.These rat species prey upon eggs and young and appear also to compete for resources.

Northern Tuatara are found on 26 islands, total estimated population c. 10 000. Island populations are managed individually (Gaze 2001). Islands range in size from 0.4 to 3830 ha with populations ranging from tens (where *Rattus exulans* occurs), to a several thousands on the largest rat-free island at 160 ha.

Cook Strait Tuatara are found on four islands, total estimated at c. 45 000 at a density in pasture on Stephens Island (150 ha) of 415-480 tuatara/ha to 1420–2015/ ha in forest.

Brothers/Gunther's Tuatara are found on three islands:

- North Brother, 4 ha, at a density of 132/ha in windswept scrub
- Titi Island in the Marlborough Sounds (31 ha) at a density of approximately 0.31/ha, set up in November 1995
- Matiu/Somes Island (24.8 ha) in Wellington Harbour,

3.2 ADULT WEIGHTS AND MEASURES

Stephens Island tuatara have been the most intensively studied and their data form the basis of physical characteristics and breeding physiology for the genus. Northern tuatara occur at a higher latitude (north of 38°S) which has a warmer climate than Cook Strait (40°S). The Brothers Islands are only slightly south of Stephens Island. Cook Strait tuatara data therefore can be used only as a guide for the two other tuatara types.

Average life expectancy in the wild is unknown. Castanet *et al*. (1988) and Saint Girons (1975) indicate 60–104 years. Recaptures in the late 1980s of tuatara clipped by W. H. Dawbin in the 1950s, with past and recent measurements, tend to support this range.

TABLE 2. ADULT WEIGHTS AND MEASURES OF STEPHENS ISLAND AND LADY ALICE ISLAND TUATARA AT MATURITY.

	SNOUT–VENT LENGTH (mm)	AGE (YEARS)
Stephens Island: mean air temp. 12.5°C		
Female	170–180	11–13
Male	180*	11–13
Lady Alice Island (north of 38°S): mean air temp. 16.3°C		
Both sexes	170–180	8–10

* *Probably do not have mating opportunities until 240 mm snout–vent or more (from Gaze 2001) The appropriate weight for tuatara of a given length can be determined by reference to Newman et al. (1979)*

3.3 HABITAT PREFERENCE

Tuatara prefer a relatively open understorey without extensive ground cover. Tuatara choose light forest cover where there are patches of sunlight for basking, but also screening from aerial predators. Preferred vegetation type is coastal forest or scrub, preferably with mahoe (*Melicytus ramiflorus*), pohutukawa (*Metrosideros excelsa*), ngaio (*Myoporum laetum*) or taupata (*Coprosma repens*).

Nesting areas tend to be sunny areas of open, north-facing soil, preferably deeper than 0.5 m and soil temperatures of 8–27°C. The soil should be friable and able to support burrows.

Ambient temperature should vary seasonally between 5 and 28°C. At 16–21°C growth was not suppressed in juveniles over winter, which may be deleterious in

the long term. Relative humidity should be generally high with heavy rain at least every few weeks. Standing water need not be present.

3.4 FEEDING BEHAVIOUR

Tuatara are 'sit and wait' predators. Movement of any animal able to be overpowered is the stimulus to feed. Feeding usually involves a head tilt (getting a line on the prey), a short 'run' of c.30 cm, and grasping the prey with the jaws. An initial firm grip is followed by slow crunching of the prey item, with some involvement of the tongue in manipulating the prey. Tuatara cannot chew, but do have some shearing ability.

3.5 PROTECTED SPECIES ROLE IN ECOSYSTEM

Tuatara are terrestrial predators at the top of their food chain. Their predators, apart from introduced mammals when sympatric, can range from kingfishers (*Halcyon sancta*) on hatchlings to the Australasian harrier (*Circus approximans*) on adults.

Tuatara are directly responsible for the failure of 25% of fairy prion eggs and nestlings (*Pachyptila turtur*) on Stephens Island (Walls 1981). Similar survival rates (mean 76%) on other islands where the tuatara population is smaller or absent suggest tuatara do not affect the overall prion production.

Food items include diverse invertebrate fauna, preferably including tree wetas (Stenopelmatidae) and darkling beetles (*Mimopeus* sp.), and small lizards. Small nesting seabirds (particularly fairy prions or diving petrels (*Pelicanoides* sp.) do contribute eggs and chicks to the diet, but nesting (burrowing) seabirds also cultivate and fertilise the soil, which allows a rich invertebrate fauna to exist. Burrows made by nesting seabirds are used by tuatara, but tuatara also dig their own. Any relationship between the species is not altruistic.

4. Facilities

4.1 CONSTRUCTION OF ENCLOSURES

4.1.1 Design and materials

Outdoor enclosures should be oriented towards the sun. Wild tuatara are vulnerable to aerial predators.

The enclosure should be located away from road traffic where possible. If the enclosure is for public display, there should be only one public viewing wall for privacy reasons. Victoria University's tuatara do not appear to be intimidated by many people crowding along the two viewing walls. The enclosure size will depend

on the intended social grouping, although at least 5 m^2 should be allowed for each tuatara, regardless of tuatara size (but see section 5 'Colony size and density'). Square-shaped enclosures are preferable over an elongate design, so that there is less chance of a dominant animal excluding others from water, burrows, or sunny areas.

Access to an enclosure should be provided by at least one door, large enough to move large cage furniture through. A 0.5 m-high wall should be placed across the inside of the doorway and a 'trap' built on the outside to increase security; this allows one door to be closed before the next is opened, when people enter the enclosure. Any gaps around the openings should be blocked.

In regions where summer temperatures exceed 30°C an overhead sprinkler system to cool the enclosure rapidly may be required. Indoor enclosures tend to become dry in summer and may need regular misting; care should be taken to prevent the ground becoming soggy.

The surface of the enclosure should be varied, with contouring and low mounds (approximately 0.3 m high), and with one or more flat areas onto which burrow exits open, preferably where sun reaches for part of the day. The burrow openings should not be onto low-lying flat ground where burrow systems may become flooded.

Water features should preferably be flowing and constructed to enable tuatara to climb out, with a gently sloping bank on at least one side. Pools should be large enough to allow tuatara to sit in the water with most of their ventral surface immersed.

The walls and roof of outdoor enclosures should be lined with mesh (e.g. 1 cm weld mesh), which provides security from predators, excludes mammalian pests such as adult mice, and provides natural sunlight for basking. A 6 mm mesh is required to exclude juvenile mice (Campbell-Hunt 2002)), but tuatara would eat juvenile mice. Double-layered mesh walls are recommended by the Australian Reptile Park in Gosford (Robert Porter pers. comm.) for outside cages housing juvenile reptiles, because 'any mice can get through 12 mm weldmesh'. The outer layer should be 12 mm weld mesh and the inner layer zinc or aluminium flyscreen mesh, preferably with a buffer zone in between.

Double glazing of windows may reduce tapping interference from the public at the viewing face. Darkened viewing areas reduce shadow intrusion, and elevation of the exhibit floor to approximately 1 m above the public viewing area floor reduces the 'look down' factor.

The outer walls should extend a minimum of 0.5 m below the surface to prevent exit/entry. The inner walls should be smooth to prevent animals damaging their snouts. A solid, opaque barrier 0.5 m high may deter tuatara from anticipating an escape route and hitting their snouts. Flat sheets of tin or plastic are suitable materials.

4.1.2 Enclosure floor

For floors of outdoor enclosures, well-drained, friable soil is preferable. Regions with clay sub-soil may need treatment to prevent flooding if the area is not sloping. Planting could include low ground covers in some areas to help prevent pooling, by plants aerating the soil and taking up water.

Floors of indoor enclosures can be covered with rubble graduated in size from 80 mm stones on the bottom, followed by coarse gravel, overlaid with weed mat to prevent drainage clogging, and topped with a porous, easily draining soil (e.g. as at Southland Museum and Art Gallery, Invercargill).

The floor can be overlaid with mulch in the form of leaves, leaf litter and/or peat and will improve moisture retention/dispersal and support invertebrate communities. Bark chip may also be used but is less invertebrate-friendly. This material should be gathered from areas where toxic sprays have not been used. Leaf litter in indoor enclosures should be renewed or seeded with fresh litter regularly (the periods will vary with the season, substrate moisture and the type of leaves used).

4.2 HOUSING

4.2.1 Design and materials

Artificial dens/burrows may be constructed for both adult and juvenile tuatara. For adults, artificial dens can be made of untreated wood, preferably hardwood, or with 30 cm diameter PVC pipe, placed on end (with screw-on cap for keeper access). Wood is preferable, however, as it allows a more natural humidity. Excessive condensation can be a problem in PVC dens (as experienced at Hamilton Zoo). Wooden dens should be 30 x 30 x 30 cm, and can be placed on top of the substrate, or partially sunken. Those on top of the substrate should be stabilised with a heavy weight (e.g. a concrete block) and enclosed except for the lid, in an earth mound, to assist insulation from noise and temperature extremes.

Each box should have two lids. The inner (wooden) lid should have a layer of expanded polystyrene foam approximately 3 cm thick, or similar material on its exterior upper surface. The outer lid (also wood) should cap the box rather than be flush with the top of its walls. If box floors are hard (e.g. wood, PVC), a layer of leaves provides cushioning for animals spending much of their time below ground.

Sedentary tuatara can develop pressure areas on their ventral chest and pelvis. Boxes constructed without floors can help prevent this, although tuatara may burrow horizontally and be less accessible. A layer of leaves on hard surfaces will also help maintain a more constant humidity.

Each box should be fitted with two tunnels to provide ventilation and escape routes, with the tunnel openings into the box on adjacent walls, far enough up the wall and sloped up towards the nest box to prevent water entering. The tunnels must be no shorter than 60 cm, curved to reduce light and draughts, and have an internal diameter of 10–12 cm (for adults). Tunnels may be constructed from 'Nova Flo', a corrugated black plastic drainage pipe with small holes in the corrugations to prevent water build-up (Goetz & Thomas 1994b). Longitudinally-split cardboard tubing may also be used for tunnels, although it is generally unsuitable for outdoor enclosures.

At least one tunnel exit should be in a sunny patch for part of the day and tunnels should be screened from neighbours by using mounds, planting, or other cage furniture. This enables each animal to feel secure and/or establish its own territory. Artificial dens should be located towards the perimeters of the enclosure, facing inwards.

Juvenile dens may be constructed from small unglazed terracotta plant pots (up to 15 cm diameter depending on juvenile size) cut lengthways and set with their long axis partly buried in the substrate. These dens can be adjusted to fit only juveniles. The porous terracotta will enable humidity within the shelter to equate with the surrounding substrate. These shelters may be covered with earth to improve enclosure appearance and increase insulation. The natural floor will allow burrowing (outside enclosures). The pot entrance should be screened from sight of other tuatara. Terracotta was used in the study by Goetz & Thomas (1994b).

Dens may also be constructed from wood, but juveniles seem to prefer closer confinement than that provided by a square box, particularly in cold weather.

4.2.2 Furnishing

A variety of rocks, logs and branches should be provided for both adult and juvenile tuatara. Such items have been recognized in other reptile species as necessary for normal social behaviour. Artificial burrows mounded on the surface of the ground, as well as rocks, can provide features suitable for male mating displays. In a newly-established enclosure, rocks may subside and squash juvenile tuatara, so precautions to prevent this should be taken. If rocks are to be included they can be supported, and the ground where rocks are to be located should be compacted during construction of the enclosure. Where juveniles and adults are housed in the same enclosure, refuges for the youngsters should be provided. These refuges should be well insulated and secure from adult interference, but allow access to sun, food and water.

Much of the enclosure should be planted with shrubs or small trees (e.g. single-stemmed shrubs) that provide an open rather than a cluttered ground level environment. Leaning trunks or branches for climbing can also be provided, but the floor below should be vegetated rather than covered with rocks, and have an escape route. Low ground covers in patches and small clumps (e.g. sedges) may also be useful. Planting should be arranged so watering will not wet the entire substrate, to give tuatara a choice of wet and dry areas.

An area of warm (north or west facing in Southern Hemisphere), uncompacted, open substrate (unplanted) should be reserved for nesting. Locations with heavy clay subsoil and/or base may need an area of loam, sand and compost created for this purpose.

4.3 CLEANING

Outdoors the removal of faeces is unnecessary. Tuatara do, however, defaecate in their water containers which need scrubbing out regularly: weekly in winter is sufficient unless activity does not reduce. Cleaning agents should not be necessary.

When cleaning indoors do not use chlorine, acids, detergents, or organic solvents. Scrubbing should be sufficient for cleaning water containers or watercourses. Faeces may need to be removed once they build up, but usually they will be passed in water and do need removing from there.

Prevention of the growth of mould is important: this can be done with good ventilation and regular replacement of leaf litter. Regularity of replacement will vary according to the season, the substrate moisture, and the type of leaves used.

Confine use of food containers to tuatara. If dishes are moved between tuatara enclosures, before re-use soak overnight in commercial bleach (e.g. Janola) diluted to give a 0.1% solution of hypochlorite (i.e. 3 mL Janola bleach plus 97 mL water) but rinsed thoroughly and dried before re-use (to prevent transfer of parasites/ disease). This is standard reptile husbandry practice.

4.4 ENCLOSURE CLIMATE

4.4.1 Lighting

Indoor enclosures must be supplemented with ultraviolet light, or the lights must provide a full natural light spectrum. Although the specific needs of tuatara for ultraviolet light have not been researched, reptiles generally require ultraviolet light to manufacture Vitamin D3, which assists in calcium metabolism. Failure to supply sufficient UV and calcium results in inadequate calcification and metabolic bone disease (MBD) in juveniles. This results in deformities, usually evidenced by 'rubber jaw', lack of robustness and ill-thrift. The first external evidence of rubber jaw is noticed as the lower jaw rolling down from the front, along with deformity of the brow ridges. Bones also tend to be soft. The condition is only partially reversible by calcium/ultraviolet therapy (Frye 1991).

Factors affecting full-spectrum light availability to captive tuatara include:

- Behavioural changes resulting in too much time spent under cover.
- Failure of keepers to distinguish full spectrum from normal white light sources.
- Screening by roof/wall/window construction materials (e.g. dense shade cloth, wooden baffles to conceal ceiling lighting from viewers, glass, Perspex, Duralite, plastic sheeting).
- Weathering and crazing of Perspex and similar products changes the quality of light transmitted.
- Diffusers with poor light transmission capability placed over light sources.
- Old lighting tubes (tube output decreases with age) and fittings (switching deteriorates with age, making the tube work harder and shortening tube life).
- Lights placed too high above animals, or too few fittings per area (light scatter increases with distance from source).
- Lights switched on for an insufficient time over a 24-hour period.

Full-spectrum lights used in tuatara enclosures include True-lite and Grolux. Grolux tubes, as used for tuatara by the National Aquarium Napier, were designed for plant growth but are often used over aquariums. Reptisun 5.0 tubes, which produce 5% Ultra Violet B light, and Desert light (7% UVB) are available from Australia (Robert Porter, pers. comm.).

Triton tubes are also available but these were developed for invertebrates and plants, and were not designed to reproduce sunlight. Graphs of spectra from the suppliers of Triton tubes show no near-UV or UV wavelengths.

UV-emitting incandescent lights are available in New Zealand but cost 66% more than True-lite tubes in 1999. Melbourne Zoo (pers. comm. Chris Banks, Curator of

Herpetofauna, Invertebrates & Education Animals) has had satisfactory results with the Osram 300 W bulbs ('Ultra-Vitalux'), which were developed for sunlamps for human use. The light is set 1.5 m above an exhibit but lizards can get closer by climbing on branches. There are no tuatara at Melbourne Zoo.

Duro-test True-lite is probably best, as it has the highest UV and near-UV output of those available, and is equivalent to natural outdoor light, except at the red end of the spectrum. Although expensive, True-lite has a long life before output declines. Note that Duro-test Vitalites are the same product as Duro-test True-lites, but are named differently for marketing reasons. A product called Trulite, made in the USA but not by Duro-test, is not a full-spectrum light. The maximum rated life is 20 000 hours (approximately 2 years continuous use) before output declines. This is with new fittings and no on/off switching.

Since the precise lighting needs of tuatara are unknown it would definitely be advisable to replace tubes earlier than 2 years. Wellington Zoo uses 3 True-lites over the animals and 2 cool white (ordinary light) fluorescent at the back in the tuatara enclosure in the Nocturnal House, an area of approximately 6.5 m^2, with the lights about 2 m above the animals. They are switched on/off twice every 24 hours, with use time about 8 in 24 hours. The True-lite tubes are renewed approximately every 12 months, without emission testing. The tuatara kept in there for 10 years remained healthy and laid eggs on several occasions.

Banks (1981) commented that 'Trulights' [sic] useful level of UV is greatly diminished after 6 months and we use them after this period for their 'general lighting capacity' (not UV). UV use of or need for by reptiles is a complex field and it seems the more we find out, the more we still need to understand'. This includes some 1998 findings at Melbourne Zoo 'that too much UV can produce just as harmful results as too little, and I don't mean high intensity exposure.' Banks doubts whether tuatara have a high UV requirement.

Burrowing reptiles should be able to control their exposure to UV light better than arboreal reptiles. If the tuatara pineal eye does have a part in regulation of such exposure, and if there are retreats in an enclosure, overexposure should not be a problem if other behaviour is normal.

Victoria University did use Black Light for their juvenile tuatara but still had Metabolic Bone Disease (MBD) develop. Since changing to True-lites there have been no further problems. Juvenile tuatara at Napier Aquarium, Rainbow Springs (Rotorua), and Capital Discovery Place (Wellington) developed MBD from a combination of inadequate lighting and lack of balancing supplements in their diet. The MBD of the tuatara from Capital Discovery Place was of short duration.

Diet quality and lighting affect reptile ability to reproduce and grow normally where access to natural conditions is prevented. Research on tuatara diet indicates that a supplement of the long-chain fatty acids found in marine organisms may be essential for optimum quality of life (Cartland-Shaw *et al.* 1998). Until the dietary problems are resolved it is not likely that the exact light needs can be determined.

If full-spectrum lighting other than True-lites is used, you need to know the spectrum produced, the amount of UV-B (most damaging UV wavelength), the distance from each size of tube where the maximum UV is produced, and the life of the tube before output declines. Red light/screens should be used where heat lamps are switched on outside normal photo-periods, to prevent possible disruption of animals' biorhythms.

4.4.1 Temperature and humidity

Temperature, humidity and photo-period should be varied seasonally if the enclosure is indoors, to enable tuatara to pass into a period of 'torpor' and to trigger specific stages of reproductive activity. Torpor in New Zealand involves a period of reduced activity rather than sleeping through the winter. Tuatara will bask on a sunny winter day. Enforced change to warmer temperatures during winter should be avoided.

Air temperature should be maintained approximately within the range of 4–15°C (39–59°F) in winter and 10–25°C (50–77°F) in summer. Diurnal fluctuations within ranges should be provided. Ventilation should be sufficient to keep the air moving in indoor enclosures to prevent abnormal fungal/mould growth and assist in circulation of respiration gases. Positive rather than passive ventilation is preferable.

Previously, 'long periods' (undefined) above 20°C have been regarded as possibly detrimental to tuatara health; but provided the fluctuations are similar to those found in wild habitats, tuatara should not be disadvantaged. Recommended temperature regimes are based on Stephens Island conditions, as most captive tuatara originated from there. Table 3 shows minimum, maximum and mean monthly temperatures at the Reptile Research Centre at Nelson, which is at a the same latitude, and has a similar coastal climate (from Goetz & Thomas 1994b). The enclosures at the Reptile Research Centre are outdoors and roofed, but not heated.

TABLE 3. MINIMUM, MAXIMUM AND MEAN MONTHLY TEMPERATURES (°C) RECORDED AT THE REPTILE RESEARCH CENTRE, NELSON DURING 1991.

1991	MIN.	MAX.	MEAN
Jan.	12	29	21.1
Feb.	16	31	23.5
Mar.	10	30	20.4
Apr.	5	26	16.9
May	6	21	13.4
Jun.	4	20	11.7
Jul.	4	20	12.1
Aug.	1	20	11.0
Sep.	5	21	13.8
Oct.	5	25	15.6
Nov.	9	26	16.8
Dec.	11	28	19.4

The sensitivity of tuatara to heat stress means that extreme care must be taken when providing hot spots (either by lighting from above for basking, or by heat sources beneath the soil). Artificially heated/cooled enclosures should therefore have suitable safety mechanisms included should a system fail, and an alternative area similar to the normal enclosure should be available as back-up accommodation. Most of the known failed attempts to keep tuatara in facilities outside New Zealand have been due to overheating.

Caution is advised in accepting previously published minimum and maximum heat tolerances. Juveniles in particular are suspected of having a narrower tolerance range. The maximum temperature should be below 30°C. In wild populations, the maximum temperature experienced would only be for a brief period during the hottest part of the day, during which tuatara would be able to go below ground.

5. Colony size and density

5.1 JUVENILES

Young should be reared in vivaria separate from the adults for several years to prevent predation.

5.2 ESTABLISHING COLONIES

All animals to be held in the enclosure should be introduced at the same time. The addition of extra animals at a later point should be avoided, particularly if the enclosure size or habitat provided is sub-optimal. Animals may need to be moved from the enclosure if they show signs of stress (e.g. staying in houses, not gaining or losing weight).

Colonies established with juveniles should have 50% more animals than ultimately required, to allow for deaths during development. If possible they should be sexed by laparoscopy (or a less invasive method if one becomes available) before introduction to the enclosure. This is especially important for groups outside New Zealand, given the difficulty and expense of such a project, as there will be few opportunities for modification at a later date.

Stable groups are necessary for compatibility so changes in the makeup of the group should be avoided. Tuatara become territorial at 6 months. Be aware of dominance problems and modify groupings to eliminate stress.

5.3 TERRITORIES AND DENSITY

The enclosure size and layout will determine the social environment. The density of tuatara and the space requirements of individuals within an enclosure will depend on the number of available territories, which in turn depend on the layout of the enclosure. A correlation between density to habitat quality and animal density is well documented in several reptile species, in both captive and wild situations. In captive reptile populations, an increase in available area can result in aggression and highly territorial behaviour, whereas a reduction in area reduces aggression and commonly leads to the establishing of hierarchies (B.R.G. Goetz pers. comm. 1995).

At the Reptile Research Centre in Nelson, tuatara (hatched 1987) are kept in mixed groups of six to eight animals at a density of 2.5 to 3.5 m^2 (per group) and a ratio of 1.5 to 2 per available territories, under experimental conditions. Tuatara remain healthy and continue to grow under this regime but it is too soon to report evidence of breeding.

Hatchlings can be kept in aquarium tanks in small groups until they are about 6 months old, when they become territorial. Wellington Zoo has kept 3 juveniles per 2 m^2 outdoors for 4 years without disadvantage.

The National Kiwi Centre at Otorohanga has maintained and bred a mixed group of tuatara in an outdoor enclosure of approximately 500 m^2 since 1980. The number of tuatara housed ranged between 13 and 18, with six of the original eight adults surviving (approximately 28 m^2 per individual). In contrast, Wellington Zoo has two male and two female adults in an outdoor rectangular area of 30 m^2. The sedentary nature of the females at Wellington Zoo suggests the enclosure is not optimal, though all tuatara maintain their weight and shed normally.

A primary reason for the failure of past captive breeding attempts may have been inadequate colony size. Research shows tuatara have many social interactions and these are probably important in stimulating reproductive activity. Although the minimum size and sex ratio of successful captive breeding colonies has yet to be established, a minimum of two adult males and five adult females may provide enough males for male interaction/ritual display and enough females to spread the attention of the males. It would also provide, on average, one potentially breeding female each year.

The density of adult tuatara in enclosures should be within the range of average densities for tuatara in the wild on Stephens Island (i.e. 500–2000 tuatara/ha, or 5–20 m^2 per animal).

6. Reproduction

6.1 REPRODUCTIVE CYCLE

6.1.1 Mating (Cree & Butler 1993)

Male tuatara have an annual reproductive cycle and can potentially mate each year. On Stephens Island, male pre-copulation breeding displays occur in mid to late summer (January–March), and mating occurs in late summer (late February–March). Males are territorial during the breeding season and control access to females resident within their territories. Aggressive encounters between males include displays with crest erection and throat inflation, chases, and fights. Biting during fights may inflict physical injury (e.g. tail loss); to date only one death from fighting has been recorded (a male at Auckland Zoo died from blood loss after an abdominal artery was severed by a bite).

Visual stimuli are probably very important during courtship. The display involves a 'head up' posture, often in a conspicuous location such as a low rock or mound.

Males court females by encircling them in an exaggerated stiff-legged walk displaying as described above. The male climbs over the female, forcing his tail under hers until their cloacas meet and the transfer of sperm becomes possible. Mating may take up to 2 hours, as observed at Wellington Zoo.

6.1.2 Oviposition

Oviposition occurs mid October to late December. Females lay only once every 4 years on average. The minimum known period between nestings in the wild is 2 years. Annual breeding has taken place in captivity at Southland Museum and Art Gallery (Invercargill), although this may compromise the health of the females (particularly indoors where light and dietary supplements may be inadequate).

The low frequency of nesting results from slow vitellogenesis and egg shelling. Yolking of a clutch takes several years. During the nesting year, a female mates in late summer (February–March), ovulates within 1–2 months, then carries the eggs in her oviducts until nesting the following spring. Shelling the eggs takes 6–8 months. Virtually no embryonic development takes place before nesting, as the embryo is 'only at gastrulation' at laying (Moffat 1985).

6.2 NESTING

At nesting, gravid females may travel up to 200 m to find preferred nesting areas (warm, north facing slopes), which are communal. Nesting females may spend several weeks travelling between the home burrow and the nesting area, each day. One or more holes may be dug, which may involve displacing eggs laid by another female. The final hole is usually 10–12 cm deep. Each female lays a clutch of soft-shelled eggs in one night. The hole is then filled with excavated material, and overlaid with grass and leaves. A female may return to her nest nightly over a week to increase the fill and protect the site from other females. Some may also hide nearby in grass during the day.

Clutch size

Table 4 shows average clutch sizes in the wild. Tyrrell *et al.* (2000) give clutch size data for five islands. The means and standard error vary from a low of 5.9 ± 0.2 eggs on Ruamahua-iti in the Aldermen Group to a high of 8.2 ± 0.7 eggs on Aorangi in the Poor Knights Islands. Values for Lady Alice and Coppermine Islands (in the Chickens Group) and Green (in the Mercury Group) are intermediate. There is no comparable captive study.

Egg size

Cree *et al.* (1996) measured eggs taken from the oviducts of wild females at 22.2 to 27.1 mm long and 16.8–18.7 mm wide. Weight at laying is from 4 to 6 g (Dendy 1899; Dawbin 1962). The eggs obtained by induction and laid naturally that were used by Thompson (1990, pers. comm.) ranged in weight from 3 to 6.1 g (mean 4.7 g).

TABLE 4. OVIPOSITION: CLUTCH SIZES IN THE WILD, ADAPTED FROM CREE (1994).

ISLAND	DEGREES LATITUDE	MAX. SVL (mm) FEMALE	MEAN CLUTCH SIZE	RANGE
Lady Alice Island	c. 36	257	7.9	5–13
Karewa Island	c. 37.5	—	7.75	4–12 *
Stephens Island (Takapourewa)	c. 41	245	9.4	1–18
North Brother	c. 41	213	6.5	4–8

** from Thomas 1890 and included for interest: n=4 only*

6.3 INCUBATION

6.3.1 Natural incubation

Around nesting time, the enclosure should be examined daily for signs of digging (the nests may be difficult to find). Alternatively, the weight of females due to lay can be monitored to detect sudden weight losses (e.g. 30+ grams). The approach taken will depend on the importance of the expected clutch and the nature of the animal(s) involved: any handling at nesting should be avoided.

The normal period of embryonic development is about 12–15 months, with eggs in warmer nests hatching earlier.

6.3.2 Artificial incubation

Eggs laid in enclosures should be removed for incubation under controlled conditions, to prevent loss from disturbance by tuatara or predation by beetle larvae (see Appendix 2 for procedure). Artificial incubation, with less fluctuation in temperature and humidity, produces hatchlings as early as 6 months.

Fungal infections of eggs, particularly in the latter half of incubation, may not be fatal. Death of the egg is indicated by rapid collapse of the shell. Infected eggs usually hatch earlier than uninfected eggs, the hatchling is smaller, and may not have absorbed all the yolk before hatching. Development of these hatchlings appears to be normal.

6.3.3 Artificial incubation and sex determination

Tuatara sex is temperature-dependent (Cree *et al.* 1995), therefore it is possible to produce hatchlings of the sex required for a named population. Sex ratios on Little Barrier Island are being investigated by Nicola Nelson, Victoria University.

6.3.4 'Head starting'

Raising tuatara in captivity to c. 80 g and c. 120 mm SVL before release in the wild is the only approved manipulation. The Reptile Research Centre (Nelson) and Berlin Zoo have forced growth and development to achieve adult size at a younger age, but that may not be beneficial for reproduction or longevity.

6.3.5 Controlling breeding

Separating sexes is the best way to prevent breeding. There is currently no fail-safe method for promoting breeding.

6.4 GENETIC DIVERSITY

Care should be taken to ensure representation of stock is as varied as possible. Until 2001 captive-bred F1 juveniles came mainly from compatible pairs, and while breeding in some new groups is occurring, parentage is known only in offspring from pairs.

Juveniles from the hatching experiments at Victoria University have clutch and individual identity with dams known, but sires must be regarded as random as they are unknown and remain in the wild.

Hay (1998) has shown there is little genetic variation in tuatara. The same genes are present, but frequency varies from island to island within an island group.

Allocations of animals should be made only after discussion with the co-ordinator. who is responsible for attempting to maintain as much of the genetic diversity of the wild population in captivity as possible.

Zoos normally make mating choices for managed populations on a mean kinship basis, by computer-generated selections using the International System of Species Identification (ISIS) software Genes, which is part of the Single Population Analysis and Records Keeping System (SPARKS).

The captive tuatara population does not have sufficient studbook data to run Genes, because the adults were brought into captivity from the wild as adults of unknown age and parentage. They are regarded as unrelated for the purposes of SPARKS.

The sires of juveniles hatched in captivity from eggs collected from wild females are unknown. They are selected for mating on the basis of their sibling relationships, as every wild female was given an identification number which was linked to the eggs she laid, and is regarded as unrelated to other wild tuatara.

Juveniles bred in captivity from a pair (as at Southland Museum and Art Gallery) have known parents; but in group breeding where eggs incubate naturally (as at the National Kiwi Centre at Otorohanga), parentage of the offspring is unknown. It is anticipated that more useful parentage data will arise from subsequent generations bred in captivity.

7. Medical issues

7.1 HEALTH ASSESSMENT

Good condition of tuatara:

- The skin should be elastic and glossy. There should be no patches of old, adhesive skin, brown scales, or reddened areas.
- Moulting should take about a month.
- Orifices should be free of discharges.
- Mouth tissues and gums should be pink in colour. The gumline should be clean, without brown stains.
- Skull and pelvic (hip) bones should not protrude obviously.
- There should be no pressure marks on the undersurface (ventrum). These are first seen in front of the vent and on the chest and indicate the animal is remaining immobile longer periods than normal.

Bad condition of tuatara:

- Refusal to eat, to emerge from the burrow or other cover, abnormal temperature regulation behaviour and/or prolonged inactivity indicate behavioural abnormalities, but physical evidence may take longer to appear.
- Urination may cease.
- Dehydration can be demonstrated if a pinched-up tent of skin remains tented on release. This should not be confused with normal wrinkled, flaccid skin around the neck and hind limbs.
- Abnormal skin is dry and old-looking, with a dull and/or indeterminate pattern. Moulting may be prolonged and patches of old skin adhere to layers beneath (dysecdysis).
- Posture is abnormal if a tuatara cannot hold its head up, the legs and/or feet are unresponsive to touch, muscle tone is poor and the tail flaccid. Paralysis as described usually progresses from the tail to the head.

7.2 MEDICAL PROCEDURES

7.2.1 Blood samples

Samples of blood for haemogram (which should include differential white blood cell count as well as the red cell and platelet picture) and biochemistry (sick animal profile, monogastric) plus uric acid should be taken from sick animals and copies of the results sent to the co-ordinator (see section 1.3, p. 34 for contact address and Appendix 3 for procedure).

Variations on the method used to process the blood sample depend on whether a centrifuge is available for separating cells from plasma/serum (the blood fraction required for biochemistry), the length of time samples will take to reach the laboratory, and the preference of the laboratory or pathologist for EDTA or lithium heparin anticoagulants for haematology and/or biochemistry. Check with the laboratory if in doubt. Smits' (1996) paper is useful, although it deals principally with birds. See Appendix 3 for other methods and blood film method.

Data of normal blood values, based on a small sample, is available (Appendix 4 shows haematology and biochemistry). It is generally cheaper to request a biochemistry sick animal panel (small animal) than individual tests, but not all normal values have been determined.

Any Ministry of Agriculture and Forestry animal health laboratory should be able to do the required tests in New Zealand, but uric acid may need to be done at a human pathology laboratory.

7.2.2 Infections and medication

Tuatara with obvious infections or discharges should be swabbed for microbiology culture and antibiotic sensitivities. Little is known of micro-organisms causing disease (pathogens) or of those normally present without harm (commensals), but knowing organism identity and sensitivities allows specific treatment and reduces the risk of developing antibiotic resistance.

Bacteria known from tuatara cultures include *Pseudomonas*, *Klebsiella*, *Morganella*, *Aerobacter* and *Proteus* species. Most of these are moderately difficult to eliminate. The low metabolic rate of tuatara could mean longer-than-normal courses of antibiotic are required (Wayne Boardman pers. comm.). Some drugs active against the above organisms are nephrotoxic (can damage the kidneys), so care is necessary.

Copies of the results should be sent to the co-ordinator who will forward a copy to the current National Wildlife Health database co-ordinator.

It is important to realise that tuatara do process medication more slowly than reptiles with a higher metabolic rate and a longer course may be needed. While the environmental temperature should be increased to increase metabolic rate, it should not exceed 20°C and should drop to 15–18°C over 24 hours; humidity should be maintained. Tuatara with respiratory compromise need a higher humidity than normal.

Since tuatara have a renal portal system, parenteral medication administered to the forequarters has best effect. Ointments and creams are generally not used on reptiles, nor is procaine penicillin.

7.2.3 Anaesthetising

Auckland and Hamilton Zoos have the most experience in anaesthetising tuatara, using Isoflurane, as it is removed via the lungs and is easily reversed with oxygen. Ketamine has been unsuccessful (Southland Museum and Art Gallery, Invercargill) as it cannot be reversed and must be broken down in the liver, which occurs slowly compared with other reptiles (Frye 1991; Mader 1996) .

7.3 SURGERY

Non-urgent surgery (e.g. laparoscopy) should be done in summer only because wounds heal more quickly than in winter. Close with stitches, which may have to be cut out rather than left to moult out. This is preferable over tissue glue which may wear off or be shed before healing and the wound can become infected.

7.4 MOULTING/ECDYSIS

Tuatara moult/shed their skin once a year, usually in summer. This should take about a month. Animals usually appreciate having water available to sit in to keep the shed soft, and there should be some branches, or other abrasive surfaces, for them to rub on if they need to (do not rely on concrete). Animals indoors may become dry. Juveniles, in particular, can have problems in two areas when shedding.

Disecdysis (abnormal/difficult shedding) of the third eyelid (nictitating membrane) can result in conjunctivitis which, if untreated, can lead to corneal ulceration. If the latter is untreated or associated with other ill-health, blindness can result. Treatment may be by application of Soframycin (framycetin sulphate), or other non-hormonal eye drops, two or three times a day. It is usually not necessary to assist the removal of old tissue unless it becomes stuck. Blunt forceps can be used, with care.

The toes should be checked for unshed skin which has shrunk and dried around the toes, cutting off the blood supply and leading to infected or dead or shed toes (avascular necrosis). Soaking the affected toes in tepid water and gently peeling the old skin off cures the problem. If infection has occurred veterinary advice is necessary. The bathing solution for infected toes (and other infections such as spider bites or other injuries) is:

1 mL Betadine antiseptic liquid (containing 1% available iodine)
1 teaspoon (5 g) magnesium sulphate (Epsom Salts)
1 teaspoon (5 g) plain sodium chloride (not iodised)
100 mL distilled water.

The bathing solution is more effective if the bathing begins as soon as possible in the course of the infection and the wound or infected area is open so the salts can draw out the infection (i.e. if the wound has closed over, treatment takes longer to be effective) (S. Keall, Victoria University, pers. comm.).

Any changes to toe clips must be recorded and reported to the co-ordinator.

7.5 ACUTE SPONTANEOUS PERIOVARIAN HAEMORRHAGE

A lethargic, almost-maturing female kept indoors with little variation in temperature or photo-period for long periods, may be experiencing acute spontaneous periovarian haemorrhage. In this case venepuncture is not advisable.

Victoria University has lost three 9-year-old female tuatara since December 1995 from a condition only able to be tentatively diagnosed when the third animal died. Haemorrhage was seen in the abdominal cavity of all three. The second and third females were carrying suspected abnormally high (unspecified) numbers of mature egg follicles.

The first female was found dead and autolysis obscured fine internal details. Lethargy and disinterest in food were the first symptoms noticed in the second female. She was moved to a cooler area as conditions were warm outside, but died. Similar symptoms in the third tuatara were accompanied by low blood pressure and a ballooning larynx due to the oxygen deficit. Treatment with Vitamin K (a factor assisting blood clotting in cases of haemorrhage) and a colloidal blood expander was not effective and death followed approximately 3 days after the illness began. Laboratory work on the latter two cases is unavailable.

The cause of the condition is indeterminate. Victoria University's animals differ from most in that they have been indoors since hatching and have no seasonal variation in temperature or photo-period, which means they have no 'hibernation' period in the winter. This could be regarded as 'forced growth'. They originated from Mike Thompson's incubation experiments, hatched in 1987. They weighed on average 295.9 g and measured 198.2 mm SVL when the first female died. This is in the middle range of the main group from 1987. The three dead tuatara were the heaviest of the group. The increased metabolic rate and activity from early summer (habitual for tuatara), probably is significant in prompting the condition.

Animals of the same age/size, and kept under the same conditions, and showing the same symptoms, should be treated as soon as possible. There is little information available beyond that by Frye (1991). The condition was tentatively diagnosed from Frye (Appendix 5) by Dr Bryan Gartrell.

7.6 DEATH AND DISPOSAL

Any dead tuatara is valuable for research purposes.

All dead captive tuatara must be reported to the local Department of Conservation Conservancy Office and the co-ordinator as soon as possible.

If the body is too decomposed for histology or microbiology, contact the Natural History Unit in Wellington at Te Papa Tongarewa Museum of New Zealand or Auckland War Memorial Museum, who should be willing to accept tuatara material not required elsewhere (contact details: Appendix 1).

Post mortem examinations should be done on all dead tuatara and copies of the results of laboratory tests, the necropsy report and the history surrounding the death, be sent to the co-ordinator, the Department of Conservation and the National Wildlife Health database co-ordinator (contact details: Appendix 1).

The remains of the animal should be frozen as soon after necropsy as possible in an airtight plastic container at –20 to –80°C and either forwarded to the Natural History Unit of the National Museum (Te Papa) or Auckland Museum or retained until further notice under instruction from the co-ordinator.

7.7 NECROPSIES/POST MORTEM EXAMINATIONS

A generalised necropsy is necessary, as soon after death as possible.

Tissue samples from all the main organ systems should be fixed in 10% neutral-buffered formalin. Swabs of gut lumen, mouth, lungs and any abnormal exudate or infected nodules, etc. should be taken and placed in transport medium.

Tissue submitted to the laboratory after necropsy by a veterinarian should be in 10% formalin. It should be sealed to prevent leakage before sending to the laboratory. As formalin is carcinogenic, handle with care and in well-ventilated areas at all times. Alcohol (70%) is suitable only as a preservative after tissues have been fixed in formalin for approximately 2 weeks. It should not be used on fresh tissue intended for histology as it makes tissue brittle and unsuitable for processing.

Costs will be the responsibility of the holder, but attempts can be made to get assistance from the National Wildlife Health co-ordinator (contact details: Appendix 1).

8. Handling

Avoid handling adults during breeding or egg-laying times.

Weigh and measure adults a minimum of once to twice annually; take weight (grams), length from snout to vent, total length and vent to tail tip (millimetres). The snout–vent and vent–tail measurements must be done using the same margin, front or rear, of the vent for both, as the vent margins may be separated by 1 to 2 mm.

Frequency of juvenile measuring depends on the experience of the holder. Juveniles should be weighed and measured a minimum of twice a year (c. autumn and spring). Mid winter should be avoided, particularly if animals are housed outdoors, as it is best not to disturb 'hibernating' reptiles. Ensure careful monitoring.

Holders with little experience should weigh and measure juveniles monthly, so any deterioration is more readily noticed.

9. Records and reporting

9.1 RECORD KEEPING

- Check enclosure visually daily.
- Record max/min temperature daily.
- Note which animals are seen, etc. until familiar with normal activity pattern.
- Note burrow/enclosure/ratio/group member shifts.
- Record what items are fed and eaten, how they are presented, the quantity fed and the tuataras' responses to food.
- Note moult signs.
- Presence of faecal pellets. Normal consistency: dark, some undigested matter visible, not foul-smelling (usually found in wet area/water) or on a prominent feature, such as the lid of a nestbox.
- Signs of fighting, e.g. marks on head, cuts on jawline, bites on feet or tail.
- Postures suggesting breeding behaviour.

9.2 REPORTS

Send information as in sections 'Handling' and 'Reports' to the co-ordinator in April and October every year, preferably by email on the datasheet available from the co-ordinator. A blank data sheet is also in Appendix 4 of the Captive Management Plan. Graphs are unsuitable. Include all measurements done between reports if more than just April and October data have been taken.

9.3 MOVES BETWEEN HOLDERS

- Contact co-ordinator before moving animals.
- The preferred time for movements between institutions is from late spring to early summer, and moves during winter should be avoided.
- Avoid moving juveniles less than 1 year old.
- Obtain permit from the local Conservancy of the Department of Conservation (and Ministry of Agriculture and Forestry if operating under a MAF licence in New Zealand).
- Provide individual travel bags/compartments, means of raising humidity (sponge, wet cloth/paper), grip for feet, cover if not in bags.
- Ensure proper security is observed when fastening travel boxes, especially if animals are unaccompanied.
- Consult section 11 for isolation and travel protocol if overseas moves or repatriations are involved.

10. Diet

Precise dietary requirements of tuatara are unknown. Cree *et al.* (1999) investigated marine versus terrestrial elements in wild and captive tuatara diets using stable isotope ratios as indicators.

Amounts and frequency of feeding are adapted to states of activity and seasonal changes; juveniles are fed more frequently than adults, depending on weight gain and health. Tuatara are usually fed less often and in smaller amounts in winter than in summer whether indoors or outdoors, but changes to boost female nutrition during egg production have not been investigated.

A vitamin and mineral supplement should be used, especially if the enclosure is indoors and little natural food is available. Vitamin D3 and calcium are essential if tuatara are held indoors. Supplements specifically for reptiles are preferable, (e.g. Rep-Cal + Herptivite, Repti-cal or Nekton-rep), but Calcipup is more easily obtained. Ground cuttlebone is a suitable calcium supplement but needs the addition of a vitamin supplement.

Avoid casually found food items known to carry parasites for which tuatara may have no resistance (e.g. marine organisms). Avoid sourcing food from areas where pest control operations have been carried out.

Use whole invertebrates where possible, and avoid feeding too many insect larvae as they contain more fat than adults. The ideal proportions of each are unknown, but probably no more than 25% of the total diet should be larvae. Vitamin E deficiency can ensue if too much fat is eaten (Boardman & Sibley 1991).

Use baby mice/rats only occasionally. They are well-balanced in calcium/phosphorus content and are particularly useful for boosting thin or sick tuatara. Obesity and associated high plasma lipid levels are commonly noted in captive tuatara (Cartland-Shaw *et al.* 1998). Use snout–vent length to weight relationships as in wild tuatara as a guide to appropriate mass for captive tuatara (Newman *et al.* 1979).

11. Tuatara quarantine procedures

Quarantine procedures are to be observed before the release of captive tuatara to the wild.

Only healthy animals should be released into the wild, or used as captive breeding stock to produce tuatara for release into the wild. No animal, however, can be guaranteed healthy (disease-free), despite taking precautions and undergoing pre-release health checks. Clearly it is essential that everything possible is done to minimise the risk of spreading disease. Strict compliance with the following protocols is essential:

11.1 ISOLATION

- Animals for release must be isolated from all tuatara for a period of not less than 30 days (ideally 90 days).
- Tuatara should be housed individually, so that it is easier to monitor condition, food intake and faecal output. A large enclosure may be divided by 0.75 m high opaque partitions to provide pens for single animals, or individuals may be kept in plastic 'fish bins' or water troughs (approx. 40 to 70 litre capacity). These should have paper flooring and suitable cover, such as a cardboard box. Mesh covers may be needed to prevent adults escaping.
- Animals being transferred together should enter and leave isolation at the same time.

11.2 HYGIENE

- Isolation enclosures or cages must be steam-cleaned, then sterilised with sodium hypochlorite e.g. standard commercial bleach, such as Janola diluted to 3% (3 mL Janola plus 97 mL water provides about a 0.1% solution of sodium hypochlorite). The solution should be rinsed off thoroughly with water to prevent burns, before the animals are introduced. Non-porous materials are

preferred when sodium hypochlorite is used as the residue is rinsed away more easily. These include smooth concrete, plastic, metal, wire mesh or wood painted with non-acrylic paint.

- Personnel servicing isolation enclosures must use barrier techniques at all times to prevent introduction of contaminants. That is, entry must be through 0.1% sodium hypochlorite foot baths, covered when not in use. Gloves, footwear, utensils and overalls should be sterile at each visit or initially, then retained for use only in the isolation area. Sterility of utensils and clothing is achieved by autoclaving, or soaking in 1% sodium hypochlorite (e.g. 30 mL Janola plus 70 mL water). Equipment and clothing must be sterilised before re-use, or should be disposed of at the end of the isolation period. Solutions of chlorine or volatile substances should not be left uncovered in the same room as tuatara.

11.3 LIGHTING

- Full-spectrum lighting should be available. True-lites 0.75 m above the animals for 6 hours a day help to maintain vitamin D3 levels for calcium metabolism.
- Tuatara should emerge from boxes during the day in undisturbed periods which should give access to beneficial light.

11.4 FEEDING

- While animals are in isolation, feed with cultured invertebrates (adults) only, e.g. two to four locusts twice a week. Ensure food items are dusted with a calcium supplement at every feed. Cease feeding 2 days before transport to the release site.
- Water should be available *ad libitum*. The dish should be large enough for animals to sit in which will assist hydration.
- For overseas transfers, encourage defaecation then rehydration of animals by removing the water dish one week (7 days) prior to transport. Replace the dish with loosely screwed-up damp paper towels; then 3 days before transport resupply the water. Alternatively, a veterinarian may perform colonic lavage to eliminate faeces.

11.5 TESTS AND TREATMENTS

On entry into isolation ensure correct identification of animals then weigh, and measure the snout–vent length; record these then re-weigh halfway through the isolation period. Check that there are no lesions, old injuries, external parasites, or unnatural discharges from orifices. The mouth membranes should be pink and the skin condition good (see section 7.1). Record defects, response to handling, and behaviour and feeding patterns throughout isolation. Animals should be checked visually at least twice every 24 hours.

It is recommended that laboratories with reptile experience are used. A list of vets and labs is attached (Appendix 1).

11.5.1 Faecal sample

Obtain before or as soon as possible after entry into isolation, and send to an animal health laboratory for parasite and *Cryptosporidium* screens plus an enteric screen (culture for bacteria). Any ova found are regarded as positive for endoparasites so treatment should follow. Ideally, parasites should be identified, but this would cost extra. Test results should be available in approximately 7 working days. Samples may be obtained by a veterinarian by colonic lavage with isotonic (normal/physiological/ 0.9%) saline (sodium chloride solution). Refrigerate samples immediately after collection, before sending to a laboratory, but send as soon as possible.

11.5.2 Treatment for parasites

Benzyl benzoate may be dabbed onto ticks, then the ticks removed when they detach. Forced removal may leave mouth parts embedded and cause infection. Ticks are usually found only on wild tuatara, since external parasites should be removed while a new animal is in quarantine before entering captivity. Visual checks should be made subsequently for more which may hatch or grow following treatment. Treat as before if more are seen. Mites (orange-red) may be scraped off body surfaces unless they occur close to the eye, where they may be suffocated by painting with vegetable (cooking) oil. Repeated treatments are necessary, to catch those that hatch after the adults have been killed.

Lorexane cream (Gamma hexachloride B.P. 1% w/v) has been used for red mite in geckos (Rowlands 1999), but should be limited to a minimum in case of toxic effects. This product is used for human head lice.

Until they became unavailable in New Zealand, Shell 'pest strips' (Dichlorvos-impregnated plastic strips designed for the removal of invertebrate pests in human habitation) were put into the enclosure of a mite-infested reptile. This had the benefit of removing mites in the enclosure as well as on the animal. No suitable substitute has been found.

Injectable ivermectin removes both internal and external parasites but its use is regarded as experimental yet in tuatara. It is recorded as being safe at 200 mg/kg body weight, for other reptiles, but in preference other methods should be used for tuatara.

Fenbendazole (Panacur suspension, 25 mg/mL) may be given orally at 50 mg/kg body weight for internal parasites, in two treatments, at 14-day intervals.

11.5.3 Cryptosporidium

There is no known treatment effective against this organism that does not endanger the animal. Tuatara testing positive should not be used for transfer. The incidence of *Cryptosporidium* has not been researched in reptiles in New Zealand but is found in poultry, pheasants and calves in New Zealand (Neil Christensen, Massey University, pers. comm.). It is found in reptiles elsewhere.

11.5.4 Enteric screen

Enteric screening in tuatara is experimental. Exotic reptiles, on entering a zoo collection, have faeces cultured to determine the presence of micro-organisms of

danger to other reptiles and humans. The principal genus of concern is *Salmonella*, several species of which cause infections in humans. For introduction into the wild the risk of introducing exotic bacteria to naive reptile populations should be minimised. It would therefore be advisable to culture faeces on selective media (media which exclude commensals, such as *Escherichia coli*, which can mask the presence in culture of other organisms) to detect any abnormal enteric flora which includes *Salmonella.*

If enteric cultures are found positive for non-commensals, contact the co-ordinator for procedure to follow.

11.5.5 Blood

A blood film should be made as soon as blood is obtained on entry to isolation (Appendix 3).

Blood films should be prepared by experienced personnel or veterinary staff. Intracellular parasites can be seen only in a thin film in which blood cells are intact, and tuatara blood cells degenerate rapidly after collection

Films should be dried (Appendix 2), correctly identified, stained (if possible) by the Giemsa method, then sent to Alpha Scientific Laboratories, Hamilton (Appendix 1). Results should be available within 10 days to 14 days.

11.5.6 Transfers outside New Zealand

Haematology and Biochemistry blood tests are necessary. Sufficient blood can be obtained from the ventral tail vein (Appendix 3).

Haematology

A haemogram comprises haemoglobin, packed cell volume, mean cell haemoglobin concentration, cell morphology, white blood cell count and differential white cell count); it requires a 1–3 mL blood sample plus 0.5 mL of EDTA (anticoagulant) or 0.5 mL lithium heparin (anticoagulant). Results should be available in 48 hours.

Biochemistry (sick animal panel)

Tests include those for liver enzymes, alkaline phosphatase (ALP), creatinine phosphokinase (CPK), bilirubin, cholesterol, urea, creatinine, total protein, albumin, globulin, albumin/globulin ratio, calcium and phosphorus. The serum from 2 mL of blood in a plain (no anticoagulant) container is required. The whole sample should be centrifuged after clotting, the serum removed to a separate new tube by pipette and sent to the laboratory. The clot is not required.

Blood and serum samples must arrive at the laboratory as soon as possible and should be kept at cool room temperature (see Appendix 3) until tested.

It is cheaper and more convenient for laboratories to use a standard test panel. Uric acid is also useful (Boardman & Sibley 1991), but is available only through human pathology laboratories in New Zealand. Copies of results of all tests, with records of any treatments done should be sent to the co-ordinator and the recipient of the tuatara.

11.6 TRANSPORTATION

11.6.1 Outside New Zealand

Animals must be housed individually for at least 96 hours prior to transportation. Follow instructions as above. Check animals visually twice a day.

Transport containers must meet International Air Transport Association (IATA) regulations. That is, they must be the size recommended for the size of the animal(s), robust, constructed of new wood or be of plastic or metal; must have a solid base, ventilation holes or mesh panels small enough to prevent interference and escape of the animal(s).

Labelling must include:

- Contacts for sender and recipient
- Instructions for care and for delay in transit: 'Do not refrigerate and keep out of direct heat or sun and strong draughts'
- Live cargo (green animal pictures) and 'this way up' (black arrows). International pictorial labels are applied to travel crates at air cargo offices.

Original copies of permits must be attached in a solid envelope to the outside of the crate, and copies posted to the recipient and retained by the sender.

Animals should be placed in individual new/sterile cotton cloth (e.g. calico) bags tied securely at the top. Bags should be about the length of the animal plus 50%. Bagged animals should be prevented from shock by placing on a bed of shredded paper with another looser layer above. To provide humidity include wet, but not dripping, new sponges in the packing in each corner of the box and in the centre.

Choose a flight

- With one plane and as few stops as possible
- Overnight, if it is necessary to pass through the tropics.

11.6.1 Within New Zealand

New cardboard pet carriers may be used if accompanied, but wooden or plastic crates locked or screwed closed are preferred if the animals are unaccompanied. If more than one animal is to use a box, they should not be on top of one another and should be bagged as above.

Juveniles may be transported in lengths of unused cardboard tubing, suitably ventilated (e.g. Post Shop 'Handitubes'). In warm weather keep out of sun, direct heat and enclosed vehicles. Do not refrigerate and keep out of strong draughts. If the journey takes more than 24 hours, box interiors should be lightly misted with water.

If more advice is required, contact the co-ordinator (section 1.3).

12. Research needs

Contact the co-ordinator (section 1.3, p. 34). This applies to New Zealand holders only.

12.1 TEMPERATURE-DEPENDENT SEX STUDIES

Dead animals are often useful for research projects e.g. juveniles of known incubation history that were unsexed at death, should be sexed at necropsy or histologically. Gonads, or whole animals, should be stored with

1. Date of death/freezing/fixing (in 10% formalin)
2. Identity of animal
3. Age

and the co-ordinator contacted (section 1.3).

12.1 MORTALITY

Every effort should be made to determine cause(s) of death, because little historic documentation exists; necropsy reports (including laboratory test results), plus recent behavioural and nutritional history must be sent to the co-ordinator (section 1.3).

13. Acknowledgements

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14. Recommended reading

Anatomy: Robb 1977.

Facilities: Newman *et al.* 1979; Heatwole 1982; Walls 1983; Goellner 1984; Townsend & Cole 1985; Gehrman 1987, 1994; Thompson *et al*. 1988.

Colony size and density: McIntyre 1988; Goetz & Thomas 1994a, 1994b.

Reproduction: Thomas 1890; Moffat 1985; Tintinger 1987; Thompson 1990, 1991; Gillingham & Miller 1991; Cree *et al.* 1992, 1995; Hazley 1993; Thompson *et al.* 1996; Tyrrell *et al.* 2000.

Medical and identification issues: Laird 1950; Si-Kwang Liu & King 1969; Robb 1977; Boardman & Sibley 1991; Frye 1991; Goold & Smits 1995; Clemance 1996; Smits 1996.

Diet: Walls 1981; Allen & Oftedal 1994; Cartland-Shaw *et al.* 1998; Cree *et al.* 1999; Blair *et al.* 2000.

Quarantine: Rowlands1999; Boardman & Sibley 1991; Jacobson 1994.

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Appendix 1

CONTACTS AND MATERIALS

A1.1. Laboratories and veterinarians in New Zealand with tuatara experience

Auckland

Auckland Animal Health Laboratory
131 Boundary Rd
Blockhouse Bay
P O Box 41
Ph. 09-626 2679
Fax 09-627 1942

Auckland Zoo
Motions Rd
Grey Lynn
Auckland
Ph. 09-360 3800
Fax 09-360 3818
(Dr Richard Jakob-Hoff, zoo veterinarian)

Waihi

Mark Vickers
18 Russell St
Waihi
Ph. 07-863 6877 or 07-8637435
(veterinarian, field work, blood work)

Hamilton

Ruakura Animal Health Laboratory
P O Box 14103
Hamilton
Ph. 07-834 1799
Fax 07-856 8797

Alpha Scientific Ltd
141 Ellis St
Hamilton
Ph. 07-846 2346
(blood tests) Fax 07-846 2266

Animal Health Centre
42 Whatawhata Rd
Dinsdale, Hamilton
Ph. 07-847 2669
Fax 07-846 6160
(Mike Goold veterinarian)

Palmerston North
Gribbles Animal Health Laboratory (MAF)
P O Box 536
Palmerston North
Ph. 06-351 7950
Fax 06-351 7909

Wellington
Wellington Zoo
Newtown Park
200 Daniell St
Newtown
Wellington
Ph. 04-381 6755
Fax 04-389 4577
(Dr Andrew Rissman, veterinarian)

Mosgiel
Invermay Animal Health Laboratory (MAF)
Private Bag
Mosgiel
Ph. 03-489 9160
Fax 03-489 7988

Dunedin
Port Chalmers Veterinary Clinic
27 George St
Dunedin
Ph. 03-472 7740

Invercargill
Flemming/Jensen Veterinary Clinic
533 Tay St
Invercargill
Ph. 03-217 7692

Australia
Dr Wayne Boardman (ex Auckland Zoo)
Taronga Zoo
PO Box 20 Mosman
NSW 2088
Australia

A1.2 Other relevant facilities

Natural History Unit of the Museum of New Zealand (Te Papa Tongarewa)
Cable St
PO Box 467
Wellington
Ph. 04-381 7000
Fax 04-381 7070
(Raymond Coory, ph. 04-385 9609)

Auckland War Memorial Museum
Auckland Domain
Auckland
Ph. 09 309 0443
(Brian Gill)

National Wildlife Health Database
C/- Massey University of Manawatu
Tennant Drive
Private Bag 11-222
Palmerston North
Ph. 06 356 0900
(Dr Brett Gartrell)
or
C/-Auckland Zoological Gardens
Motions Rd
Grey Lynn
Auckland
Ph. 09-360 3800
Fax. 09-360 3818
(Dr Richard Jakob-Hoff)

Science & Technical Centre
Department of Conservation
65 Victoria St
PO Box 10 420
Wellington

A1.3. Products mentioned in the text

CALCIPUP
Manufacturer: Villavet Products, 35 Havelock Rd, Havelock North.
Supplier: Ethical Agents Ltd, 19 Garfield St, Parnell, Auckland. Ph. 09-262 1388.

NEKTON-REP
Distributors: Nekton USA Inc., 14405 60th St. North, Clearwater, Florida 34620. Ph. (813) 530 3500, Fax (813) 539-0647.
Manufacturer: Nekton-Produkte, D-75177 Pforzheim, Germany.

REP-CAL
A phosphorus-free calcium supplement plus vitamin D3, usually used in conjunction with HERPTIVITE, a vitamin supplement without vitamin A but with beta carotene.

Available from Victorian Herpetological Society Inc., 16 Suspension St, Ardeer, Victoria 3022. Importation to New Zealand is difficult because the products are regarded as an animal remedy, not a dietary supplement.

REPTI-CAL (calcium and vitamin D3)

Available from Aristopet, 874 Kingsford-Smith Drive, Eagle Farm, Brisbane, Queensland, 4009, Australia. Ph. +61-7-36 3021 66, Fax +61-7-36 3021 77.

BIOSUPPLIERS

For cultured invertebrates: locusts and crickets are the largest insects available. 201 Eskdale Rd, Birkenhead, Auckland, ph./fax 09-418 2352.

LIGHT UNITS FOR FLUORESCENT TUBES

Available from aquarium suppliers.

TRUE-LITE

Importers: Illumination Distributors Ltd, P O Box 3095, Wakefield St, Napier,. Ph./Fax 06-843 5288.

LOREXANE

Available from pharmacies.

Appendix 2

PROCEDURE FOR ARTIFICIAL INCUBATION

1. Prepare a 2-litre plastic ice-cream container for the eggs; sterilise enough vermiculite to fill the container two-thirds; add distilled or rain water to give a water potential of -100 to -300 kPa (e.g. Victoria University's vermiculite needs 96 mL water per 120 g, but other stocks may differ).
2. Write a different number on the upper surface of each egg with a soft graphite pencil and keep the number uppermost during all subsequent handling.
3. Weigh each egg to the nearest gram.
4. Partially bury 6 to 9 eggs in the vermiculite to enable visual checks without disturbing the eggs.
5. Incubate eggs at 18°C (to increase the chance of hatching out females), or at 22-23°C (for males), but see section 6.3.3, p. 470.
6. Eggs should be weighed weekly to check for continued weight gain. Add the equivalent in grams of distilled water to replace any lost by evaporation or absorption. The vermiculite should also be mixed at each weighing to prevent uneven distribution of moist vermiculite. Tuatara eggs gain weight more rapidly, immediately prior to hatching. Total egg weight should be less than 18 g or the eggs are unlikely to hatch. Control of weight gain is possible by placing the egg(s) in drier conditions to allow water loss.

Appendix 3

BLOOD SAMPLING METHODS AND HANDLING

1 Aseptic sampling

Collect blood samples following recommended methods, e.g. venipuncture of caudal vein (Smits 1996). The site should be sterilised by swabbing with 70% alcohol first and after sampling should have light pressure applied with a clean gauze or cotton wool pad until bleeding stops.

2 Blood sample volume

No more than 1% of body weight should be taken. Most adults should be able to supply 3 mL.

Blood can be taken from the ventral tail vein, located at the midline, approximately seven scales post-vent with the needle angled at c. 45 degrees to the tail surface to access the vein past the spinous processes.

If there is a microchip in the ventral tail muscle take care to avoid it, as it is possible to break the glass around the chip.

Changing syringes must be done with minimal delay to avoid blood clotting in the needle.

Finding the vein can take several minutes. If you are using heparin as an anticoagulant, run some from a source other than the blood tube through the syringe and needle to prevent clotting; expel as much heparin as possible before the blood enters the syringe. Best remove the needle before the blood is transferred to tubes, to reduce the chance of red cell rupture.

Small samples (i.e. one packed cell volume (PCV) tube) can be obtained from juveniles or adults by clipping a toenail short. The foot may need warming or flicking with a finger to increase circulation before sampling.

Collect blood into lithium heparin and after mixing gently for 10–12 inversions, fill a glass capillary tube (PCV or microhaematocrit tube) as soon as possible. Use a drop from this to make two blood films on glass slides. Seal the capillary tube at both ends if transporting and use for PCV and protein, or ask laboratory for these tests.

With the remainder of the lithium heparin, spin the sample and remove the plasma with a fine (Pasteur) pipette from the cells, saving it in a fresh, plain blood tube, or transport it direct to the laboratory.

If you have the ability to collect sufficient blood into EDTA at the same time, also fill to the required level (250 mL into a microtainer is sufficient to run through a Coulter counter) and **still make 2 blood films**.

If you do not have lithium heparin or access to a centrifuge capable of 3000 r.p.m.: collect your second blood sample into a **plain tube** (no anticoagulant).

Ideal method

- Centrifuge available for separation of serum/plasma from samples.
- Access to laboratory within less than 4 hours of bleeding.

Practical method

- If there is no suitable centrifuge,
- but two tubes of blood have been taken from the animal,
- samples cannot be tested in under 4 hours and
- the film-making technique is unfamiliar:

Use smallest possible EDTA tube (microtainer is best). Fill to the recommended limit and mix gently. Make your films as soon as possible (for haematology).

Collect as much blood as you can in a plain tube for chemistry. Hold whole blood samples at cool room temperature (12–16ºC), but place serum in the refrigerator once it is separated from the clot.

3 Blood sampling equipment

(Compiled with reference to Smits 1996 and Clemance 1996)

- **1 mL syringes** exert less negative pressure than larger syringes so the vein is less likely to collapse.
- **20 or 22 gauge hypodermic needles** are less likely to rupture the red cells, which are large and are damaged easily.
- **EDTA microtainer** blood tube as used in human paediatrics. EDTA is the best anticoagulant for maintaining detail of blood cells, but plasma cannot be used for any chemistry tests. Take care to fill to the required mark for best results. The minimum useful volume is 250 mL.
- **microhaematocrit (PCV) tubes** are used to make blood films. If one end is sealed a PCV tube can be spun for packed cell volume reading and plasma protein determined using a refractometer. Seal both ends if the tube is to be sent to the laboratory.

PCV tubes are available as plain (blue) or heparinised (red). Use plain tubes for EDTA samples and heparinised for blood taken into heparin or plain, as from a toe clip. Lithium or sodium heparin blood tubes, 3 mL or less should be used only if sample(s) can reach the laboratory within 1–4 hours of bleeding, or clotting occurs. Lithium heparin plasma can be used for chemistry but its cells are useless for haematology unless they are fresh, so good blood films must be made at bleeding. Store heparin-rinsed blood samples at room temperature. Do not refrigerate before testing or the cells will lyse (rupture).

Sodium heparin can be used if lithium heparin is unavailable, as the amount of sodium occurring in the anticoagulant is minimal compared with the amount in extracellular fluid, and sodium values should be insignificantly affected. Also, sodium is not a usual diagnostic test.

Take care to fill to the required mark only for best results.

- **Plain blood tubes** (no anticoagulant) should be used for biochemistry if a delay of over 4 hours is expected. Do not refrigerate after bleeding, but leave at cool room temperature (12–15ºC) until tested/sent to the lab.
  Where 'plain tubes' are mentioned note that these are sterile, capped, glass tubes without anticoagulant e.g. 'Red top Vacutainers'.

- **Glass microscope slides** need polishing with a lint-free cloth before using for blood films, to remove any grease from the slide surface. Grease causes patchy films. Make at least 2 films per animal (see below).
- **Glass (not plastic) microscope cover slips** are used to spread the drop of blood. Use the coverslip edge which is the same width as the slide.

4 To make blood films

- Use only new slides, a minimum of two per tuatara.
- Polish slides with a lint-free cloth to remove surface grease.
- Approximately 1 cm from one end deposit a drop of blood 2–3 mm in diameter.
- Take a new cover slip, rest one edge of it beyond the drop of blood, hold it at an angle of 30–45 degrees, and draw it backwards onto the leading edge of the blood.
- Allow the blood to run along the edge of the cover slip to within 2–3 mm of each edge of the slide, then with a steady, continuous action, draw the blood along the slide until it is left behind as a convex-fronted, thin film. A single cell layer is required for cell examination.
- The ideal shape of the leading edge of a blood film is convex. Straight, ragged front edges with lines throughout are of limited value.
- Lay the films wet side up on a flat surface, out of sun or other direct heat to dry in air.

Label to identify, pack when dry so the film will not be scratched, then send to the laboratory.

Appendix 4

NORMAL BLOOD VALUES

Reptile blood values can vary with sex and time of year. Two sets are given for December (summer) and April (autumn).

HAEMATOLOGY OF CAPTIVE MALE AND FEMALE ADULTS, NORTHERN AND COOK STRAIT TUATARA (BLED DECEMBER=SUMMER), TESTED BY MAF BATCHELAR (NOW GRIBBLES) ANIMAL HEALTH LABORATORY, PALMERSTON NORTH.

TEST	POOLED (n=26 MEAN	POOLED (n=26 RANGE	MALE (n=16) MEAN	MALE (n=16) RANGE	FEMALE (n=10) MEAN	FEMALE (n=10) RANGE
White blood cell count, WBC–estimated (10^9/L)	7.5	1.2–21.0	7.6	3.5–21	6.6	1.2–11.7
Haemoglobin, Hb (g/L)	64.6	45–91	67.6	51–91	59.7	45–73
Haematocrit or packed cell volume, PCV (%)	34.2	22–53	34.7	23–53	32.7	22–49
Mean cell Hb concentration, MCHC (g/dL)	18.9	14.3–26.4	19.73	14.3–26.1	18.98	14.3–26.4

BIOCHEMISTRY, GROUP AS FOR HAEMATOLOGY

TEST	POOLED (n=25) MEAN	POOLED (n=25) RANGE	MALE (n=15) MEAN	MALE (n=15) RANGE	FEMALE (n=10) MEAN	FEMALE (n=10) RANGE
Calcium (mmol/L)	2.93	1.57–5.79	2.70	1.57–3.47	2.18	2.18–5.79
Phosphorus (mmol/L)	1.9	0.96–3.36	1.91	0.96–3.36	1.88	1.1–2.32
Aspartate aminotransferase, AST (mmol/L)	26.96	4–112	28.6	4–112	24.5	5–111
Alanine amiotransferase, ALT (mmol/L)	1.84	0–9	1.53	0–6	2.1	0–9
Glucose (mmol/L)	6.69	4.6–13.9	6.79	4.6–8.4	6.54	4.8–13.9
Creatine (mmol/L)	40.8	30–63	44.5	32-63	35.3	30-45
Total protein (g/L)	41.1 (n=24)	18.7–58.4	39.5 (n=14)	18.7–58.4	41.6	20.1–57.3
Urea (mmol/L)	2.82 (n=23)	1.3–7.2	2.34	1.3–5.9	3.49 (n=8)	1.5–7.2
Uric acid (mmol/L)	0.166	0.013–0.5	0.165 (n=14)	0.013–0.5	0.17	0.06–0.33

HAEMATOLOGY. NORMAL BLOOD VALUES n=3, 1 MALE, 2 FEMALE CAPTIVE ADULT NORTHERN TUATARA (BLED APRIL–AUTUMN), TESTED BY ALPHA SCIENTIFIC LAB.

TEST	POOLED (n=3) MEAN	POOLED (n=3) RANGE	MALE (n=1) VALUE	FEMALE (n=2) MEAN	FEMALE (n=2) RANGE
White blood cell count, WBC (10^9/L)	12.73	8.4–19.6	10.2	14	8.4–19.6
Differential white blood cell count, WBC)					
heterophils (%)	48.42	39.8–51.02	55.45	44.91	39.8–50.01
lymphocytes (%)	24.28	19.8–32.14	19.8	26.53	20.92–32.14
monocytes (%)	15.55	3.57–34.18	8.91	18.88	3.57–34.18
eosinophils (%)	9.88	4.08–14.85	14.85	7.4	4.08–14.85
basophils (%)	1.86	0.99–3.57	0.99	2.3	1.02–3.57
Haemoglobin, Hb (g/L)	75.33	70–80	70	78	76–80
Haematocrit or packed cell volume, PCV (%)	33.66	31–36	31	35	17–36
Mean cell Hb concentration, MCHC (g/dL)	22.44	20.59–24.52	20.59	23.37	20.59–24.52

BIOCHEMISTRY, GROUP AS FOR HAEMATOLOGY

TEST	POOLED (n=3) MEAN	POOLED (n=3) RANGE	MALE (n=1) VALUE	FEMALE (n=2) MEAN	FEMALE (n=2) RANGE
Calcium (mmol/L)	4.69	2.93–6.15	2.93	5.58	5–6.15
Phosphorus (mmol/L)	2.6	2.4–2.9	2.4	2.7	2.5–2.9
Aspartate aminotransferase, AST (µmol/L)	14	4–23	23	9.5	4–15
Alanine aminotransferase, ALT (µmol/L)	1197.7	1009–1319	1319	1147	1265–1029
Glucose (mmol/L)	5.96	5.4–6.99	5.5	6.2	5.4–6.99
Creatine (mmol/L)	26.3	22–30	27	26	22–30
Total protein (g/L)	47.6	42–52	42	50.5	49–52
Uric acid (mmol/L)	0.126	0.08–0.16	0.16	0.11	0.08–0.14
Total bilirubin (µmol/L)	0.968	0.922–0.992	0.992	0.957	0.922–0.992
Lactate dehydrogenase, LDH (µmol/L)	416	266–526	456	396	266–526
Creatinine phosphokinase, CPK (iu/L)	1098.3	99–1653	1653	821	99–1543

The above examples are from captive animals. Some published data are also available for plasma concentrations of hormones, lipids and glucose in wild animals (e.g. Cree *et al.* 1992; Cartland *et al.* 1994; Cartland-Shaw *et al.* 1998; Blair *et al.* 2000; Tyrrell *et al.* 2000). Brown *et al.*(1991) have values for calcium, total protein, inorganic phosphate, cholesterol and vitellogenin, and Brown *et al.* (1994) have plasma concentrations of sex steroids and vitellogenin in female *Sphenodon p. punctatus.*

Appendix 5

ACUTE SPONTANEOUS PERIOVARIAN HAEMORRHAGE

The following quote about this condition is from Frye (1991).

'An unusual condition observed in captive reptiles is the sudden and, as yet, unexplained, rupture of one or more ovarian arteries and subsequent haemorrhage into the ovary and periovarian tissues. Because of the suddenness of this disorder, there are few, if any, specific signs that would suggest its nature. The affected animals have included lizards, a tegu, and an iguana; another case was a king snake that exhibited acute distress, mucosal pallor, lethargy and death from hypovolaemic shock that resulted from massive blood loss that originated in one of its ovaries and was passed through the proctodeum and exited through the cloaca. Each of these cases was diagnosed at necropsy.

'The aetiology of this condition is unclear and appears to be unrelated to external trauma. The most likely cause would be spontaneous vascular rupture at the time of ovulation, although ovarian arterial haemorrhage can also occur spontaneously, especially if an arterial aneurysm is present. Hypovitaminosis-C-induced vascular weakness and/or gross obesity have been suggested but are, as yet, unproven in two of the cases examined grossly and microscopically.'

Note: Other spontaneous bleeding noted in reptiles involves bleeding from the gums, seen in vitamin C and K deficiency, but this has not been recorded in tuatara.