What effect does diet have on body condition of unowned cats in the Southern Downs Region, Queensland?

A thesis submitted by

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Abstract

Cats (*Felis catus*) have inhabited Australia since at least European colonisation and their negative impacts have been well documented in the literature.

Since free-roaming, unowned populations established across the continent and on many territorial islands, cats have been responsible for native small mammal declines and extinctions. In the temperate climate zone of south-east Queensland, currently no data exists on unowned cat diet. In the Southern Downs Region, unowned cats cost the local council in excess of $80,000 per year to manage. Furthermore, their impacts on small native animals are unknown.

Understanding unowned cat diet in the region is integral to determining the impacts they are having in addition to informing local cat management strategies. Body condition in unowned cats, which may be a function of cat diet, can also provide information on the overall health of the population.

The general aim of this project was to determine the effect diet has on body condition in unowned cats in the Southern Downs Region. It was hypothesised that cats that predominately consumed small mammals would be in better or ideal body condition. Further aims of this study were to determine the overall diet composition and body condition of unowned cats in the Southern Downs Region, Queensland. Unpublished data on unowned cats from Davenport Downs Station in an arid area of western Queensland was used to compare and contrast the results from the Southern Downs Region.

The study was conducted during autumn and winter in 2021. Stomach and faecal samples were used to conduct dietary analyses. Morphometric versus weight regressions, body condition score, and kidney fat index were used to assess body condition.

Commercial cat food and mammals, likely to be carrion, comprised the majority of the diet in Southern Downs Region cats. Mammals dominated the diet of cats at Davenport Downs Station. The majority of cats from both study sites were in ideal body condition, and there was no strong relationship between body condition and diet of unowned cats.

Future studies would benefit from including a larger sample size of adult unowned cats and movement data of those sampled as these were identified as limitations during this study.

Certification of dissertation

I certify that the ideas, experimental work, results, analyses, software and conclusions reported in this dissertation are entirely my own effort, except where otherwise acknowledged. I also certify that the work is original and has not been previously submitted for any other award, except where otherwise acknowledged.

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Signature of Candidate Date

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Signature of Supervisor Date

Key words

Australia

Body condition

Cat

Cat management

Diet

Felis catus

Feral cat

Kidney fat index

Stray cat

Unowned cat

Abbreviations and definitions

BC: body condition

BCI: body condition index

BCS: body condition score

FOO: frequency of occurrence

Modified environment: areas in which human activity and modification of the landscape has occurred (e.g. intensive agricultural land, urban areas).

Natural environment: areas in which human activity and modification of the landscape are predominantly absent.

Peri/semi-urban: fringes of suburban areas and those between suburban and rural areas.

SDR: Southern Downs Region

SDRC: Southern Downs Regional Council

Unowned cat: cat that is not owned and has either no or partial reliance on humans for feeding requirements.

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Acknowledgements

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Chapter 1: Introduction

Free-roaming, unowned cats (*Felis catus*) have become a significant nuisance and threat to wildlife worldwide. They are known for predating on threatened species and have been implicated in extinctions of many birds, reptiles and mammals (Gibson et al. 1994; Kutt 2011; Shionosaki et al. 2015; Doherty et al. 2016b; Cove et al. 2017; Woinarski et al. 2017; Woinarski et al. 2018a; Greenwell et al. 2019; Murphy et al. 2019; Trouwborst et al. 2020; Woinarski et al. 2020; Woolley et al. 2020). Worldwide, cats have a greater negative impact on native wildlife than other invasive predators such as wild dogs, *Canis familiaris*, red foxes, *Vulpes vulpes*, wild pigs, *Sus scrofa*, stoats, *Mustela erminea*, and small Indian mongooses, *Herpestes auropunctatus* (Doherty et al. 2016b).

In Australia, many of the native mammalian species fall within the preferred prey size range of cats (Burbidge & McKenzie 1989; Johnson & Isaac 2009), making them more vulnerable to predation than animals in other parts of the world (Doherty et al. 2016b). Unowned cats also cause a nuisance to humans in both modified and natural environments through their behaviours including fighting, spraying, defecating, and mating (Toukhsati et al. 2012; Sparkes et al. 2013).

Cats inhabiting peri-urban fringes are particularly problematic given ample resources from anthropogenic sources such as rubbish tips and feeders supplementing these cats' feeding (Denny 2005; Cove et al. 2017; Crawford et al.

2020). Additionally, once breeding occurs, cats will naturally disperse to inhabit new home ranges (Devillard et al. 2003), thereby moving into rural areas, and threatening wildlife. In the Southern Downs Region (SDR) of south-east Queensland, there are currently 39 threatened native vertebrate species (Department of Environment and Science 2013b), the majority of which fall within the prey size range preferred by cats (Burbidge & McKenzie 1989; Johnson & Isaac 2009). As a result, a great deal of effort and resources are put towards cat management.

Each state has different legislation relating to cats. In Queensland, no sole entity is responsible for the control of unowned cats, rather everyone has a general responsibility to minimise their effects under the *Biosecurity Act 2014*. At present, the SDR has no dedicated cat management program in place (Warren, M 2021, pers. comm., 31 August). Instead, the Southern Downs Regional Council (SDRC) conduct one-off cat management programs when nuisance complaints are repeatedly received and also offer cage traps for the public to hire and use (Warren, M 2021, pers. comm., 31 August). These management costs for the local council have been estimated at $85,000 per year in the SDR (Magnussen 2020).

There is often ambiguity in the use of the term ‘feral cat’ (Gosling et al. 2013), but they are defined as those that live in a wild state with their nutritional needs met through hunting and scavenging and do not need human intervention to survive (Moodie 1995; Department of the Environment 2015a). They are typically associated with rural areas or natural environments (Fleming et al. 2021). No behavioural traits are used to define feral cats in Australia (Moodie 1995; Dickman 1996; Department of the Environment 2015a), however, Gosling et al. (2013) states feral cats avoid humans when faced with them. In comparison, stray cats hunt prey and/or partially rely on humans either through scavenging anthropogenic waste or people who actively feed them (Moodie 1995; Dickman 2009). Stray cats are also defined by their presence in urban, suburban, or peri-urban environments (Fleming et al. 2021). Finally, pet or owned cats rely solely on humans for food and all other requirements (Moodie 1995). For the purpose of this thesis, feral and stray cats will be referred to as unowned cats hereafter, unless otherwise specified.

In 2020, the SDRC obtained funding from the federal Government to aid the development of management strategies for unowned cats in the region. The University of Southern Queensland was commissioned to undertake associated diet and movement studies on unowned cats in the region to assist the SDRC with their cat management planning, and this thesis reports on the dietary aspects of this work. Chapter 2 reviews literature on unowned cat ecology, diet, and dietary related factors and how these influence cat body condition (BC) and management. Chapter 3 describes and explains the methods used to establish diet and BC results in this thesis. Chapter 4 presents the results of this study, and Chapter 5 discusses these findings and compares them to relevant literature. Finally, Chapter 6 briefly summarises the findings of this research and provides recommendations for future work.

Chapter 2: Literature review

2.1 Introduction

Cats have been in Australia since 1788 (Denny & Dickman 2010), and potentially even longer (Burbidge et al. 1988). After colonisation, cats were kept as pets and used as pest control for rodents and European rabbits, *Oryctolagus cuniculus* (Campbell 1906; Denny & Dickman 2010). Cats quickly established unowned, free-roaming populations and now inhabit almost all of the continent of Australia and 98 of its islands, including the state of Tasmania (Legge et al. 2017). Cat populations are classified by the environment type they live in, such as natural and highly modified (Moodie 1995; Legge et al. 2017). Natural environments include areas where vegetation dominates the landscape (Legge et al. 2017). In comparison, highly modified environments are those where anthropogenic activity exceeds natural vegetation in the landscape (Legge et al. 2017). Examples include urban areas and intensive agricultural areas (Legge et al. 2017). Peri-urban areas are those that lie between suburban and rural areas and often have aspects of both modified and natural environments (Crawford et al. 2020). Unowned cat populations in Australia have been estimated between 1.4 and 5.6 million in natural environments and 2.1 and 6.3 million in highly modified environments (Legge et al. 2017).

Cats are listed on the International Union for Conservation of Nature’s 100 of the World’s Worst Invasive Alien Species list (Invasive Species Specialist Group 2021) and worldwide have been responsible for 63 vertebrate extinctions (Doherty et al. 2016b). Despite signs of predation on native wildlife by cats in the early 1900s (Campbell 1906), cats were not seen as significant predators of many Australian native wildlife species until the mid to late 20th century (Burbidge & McKenzie 1989;

Gibson et al. 1994). In 1999, feral cats were listed as a key threatening process under the *Environmental Protection and Biosecurity Conservation Act 1999* and in 2015 a threat abatement plan was introduced (Department of the Environment 2015b). Understanding the link between diet and unowned cat health is an important component in cat management (Algar & Burrows 2004; Christensen et al. 2013; Sparkes et al. 2013; Doherty et al. 2016a). Research of this kind can inform local governments, landholders and conservation staff on what management strategies need to be implemented.

This review will discuss the ecology of unowned cats in Australia, focusing on unowned cat diet and BC. Studies pertaining to measurements only useable on live cats (i.e. bloods, organ function, hormone levels) have been excluded from this review. Diet studies from Australia and its territorial islands have been included and some overseas studies have been used for comparison. Pet cat hunting behaviours are said to be similar to those of unowned cats (Pearre Jr & Maass 1998), hence their inclusion as well. Pet cat diet (i.e. commercially available foods) is outside the scope of this review. BC studies on relevant species other than cats have been utilised due to the limited BC literature specific to unowned cats.

2.2 Cat ecology

Cats are known to inhabit a wide range of habitats for example, in natural environments, cats inhabit woodlands, tree line fringes and pastoral lands (Bengsen et al. 2012). However, they are more effective at hunting in areas with low shrub density (Davies et al. 2017; Davies et al. 2020). In modified environments, cats are more frequently found in areas where there is excess anthropogenic waste, such as waste disposal sites and areas frequented by humans (Denny 2005; Campos et al. 2007; Cove et al. 2017; Crawford et al. 2020; Piontek et al. 2020).

2.2.1 Home range

Unowned cat home ranges vary significantly (Liberg et al. 2000; Bengsen et al. 2016). For example, Bengsen et al. (2016) found female home ranges ranged between 1.2–23.2 km^2. Although, majority of cat home ranges fall between 4–6 km^2 (Bengsen et al. 2012; Bengsen et al. 2016). The size of the home range is determined by resource availability (Burt 1943; Bengsen et al. 2016), therefore, it is possible that cats with larger home ranges have sparse resources and may need to roam further from their home range for them. This may also be why cat home ranges overlap (Page et al. 1992; Bengsen et al. 2012) in spite of cats being solitary animals (Cecchetti et al. 2021). In contrast, cats with small home ranges are often subsidised by anthropogenic feeding or waste and therefore have ample resources (Page et al. 1992). This may explain why cats living in modified environments have smaller home ranges than those that live in natural environments (Page et al. 1992).

2.2.2 Activity

Cats have been known to be active at all times of the day and night (Domm & Messersmith 1990; Fancourt et al. 2015) therefore, activity overlap with native carnivores is inevitable (Glen & Dickman 2008; Cowan et al. 2020). Season can have an effect on cat activity which can also lead to activity overlaps with native carnivores such as quolls, *Dasyurus* spp. (Fancourt et al. 2015). The presence of other carnivores such as the Tasmanian devil, *Sarcophilus harrisii*, can cause cats to change their activity from nocturnal to diurnal (Fancourt et al. 2015). Yet, cats are not significantly affected by the presence of naturalised and introduced apex predators such as dingoes and other wild dogs (Allen et al. 2015; Wysong 2016; Stobo-Wilson et al. 2020).

2.2.3 Life span

Unowned cat life span is dependent on several factors including, but not limited to, resource availability (Levy et al. 2003) and disease (Hartmann 1998; Murray et al. 2008; Finn et al. 2010). The lifespan of unowned cats has been estimated as two years (Jessup 2004), but can range up to nine years (Hayde 1992). The findings of Jessup (2004) are in line with research by Fleming et al. (2021) who found the majority of culled, unowned cats were between 1-3 years of age. More often than not, studies involving unowned cats assess age categorically (i.e. juvenile, adult, geriatric) rather than numerically (Jones 1977; Jones & Coman 1981; Brothers et al. 1985; Denny 2005; Yip et al. 2014; Gunther et al. 2018; Crawford et al. 2020). This is likely due to the need for accurate age data being outweighed by time constraints and lack of resource availability. As a result, unowned cat ages are difficult to compare between studies.

2.2.4 General impacts

Australia has one of the highest number of impacted mammal species in the world due to invasive predators (Doherty et al. 2016b) and unowned cats have played a significant role (Gibson et al. 1994; Davies et al. 2017). For example, a recent study found that across Australia, unowned cat presence has a positive correlation with mammalian declines (Legge et al. 2017). Furthermore, research has shown that after rodents, cats threaten the most bird species (Doherty et al. 2016b) and their presence has been linked to nest abandonment in seabirds (Greenwell et al. 2019).

Unowned cats also contribute to the failure of reintroduction programs (Gibson et al. 1994; Christensen & Burrows 1995; Frank et al. 2014). Reintroduction programs are used for native species that once inhabited an area but no longer do (Gibson et al. 1994). A captive population is bred from wild stock animals and then released into an area to re-establish the population (Gibson et al. 1994; Moseby & O'Donnell 2003). One example occurred between 1989 and 1991, when rufous hare-wallabies, *Lagorchestes hirsutus*, were reintroduced to two sites in the Tanami Desert in the Northern Territory (Gibson et al. 1994). During the program, cats killed close to half of the reintroduced population and subsequently, the population became extinct.

Cats also pose a disease risk to livestock, particularly sheep, causing various problems for sheep farmers including birthing and pregnancy problems and death (Dubey 2009; Taggart 2019). Diseases include those resulting from the parasites *Toxoplasma gondii*, *Sarcocystis gigantea*, and *Sarcocystis medusiformis* (Dubey 2009; Taggart 2019; Taggart et al. 2020).

2.3 Diet

In Australia, unowned cats kill approximately 894 million mammals (Murphy et al. 2019), 769 million invertebrates (Woolley et al. 2020), 466 million reptiles (Woinarski et al. 2018a), 316 million birds (Woinarski et al. 2017), and 92 million amphibians each year (Woinarski et al. 2020). Though these figures wane in comparison to other countries (Woinarski et al. 2017; Murphy et al. 2019), the growing body of evidence surrounding unowned cat impacts on native species in

Australia is enough to cause concern (Gibson et al. 1994; Doherty et al. 2016a; Doherty et al. 2016b; Woinarski et al. 2017; Woinarski et al. 2018a; Greenwell et al. 2019; Murphy et al. 2019; Woolley et al. 2020).

Cats have been described as opportunistic and generalist carnivores by some authors (Pearre Jr & Maass 1998; Campos et al. 2007; Yip et al. 2014; Doherty et al. 2015; Yip et al. 2015; Fleming et al. 2020), which means their dietary diversity includes most small prey types. Cats are also obligate carnivores (Bradshaw et al. 1996; Cecchetti et al. 2021), meaning that they must consume meat for certain nutrients that they are unable to synthesise (Knopf et al. 1978; Morris & Rogers 1978; Cecchetti et al. 2021). Further to this, cats have a higher protein requirement than other invasive predators such as wild/domestic dogs (Legrand-Defretin 1994; Zoran 2002). With these factors in mind, it is unsurprising cats have a strong prey drive.

2.3.1 Predator competition

Competition between cats and native species has been alluded to (Medina et al. 2013) however, evidence suggests cats present few competitive effects (Glen & Dickman 2008; Fancourt et al. 2015; Wysong 2016). For example, in Tasmania, quolls are not negatively affected by the presence of cats (Fancourt et al. 2015). A study by Glen and Dickman (2008) found house mice, *Mus musculus*, and European rabbits (staple cat prey items (Dickman 1996)), were not found in high frequencies in quoll diet. This is supported by the findings of Dickman (1992) who found mice had a stronger aversion to cats than quolls, suggesting quolls do not readily consume mice. These findings provide evidence that there is minimal competition between cats and native carnivores. However, dietary overlap between cats and native species may be under-reported due to cats burying their scats (Glen & Dickman 2008).

2.3.2 Prey selectivity

Prey selectivity is defined as the predation of specific individuals due to their size, species, or location, by a predator despite other prey being readily available (Childs 1986; Kutt 2012; Spencer et al. 2014). Prey selectivity is also a result of prey availability fluctuations (Spencer et al. 2017). Prey selectivity is measured using Jacob's Index which compares the proportion of a prey type against its abundance in a specified area (Kutt 2012).

Unowned cats in modified and natural environments exhibit prey selectivity (Childs 1986; Dickman 1996; Molsher et al. 1999; Dickman & Newsome 2015). Dickman and Newsome (2015) found more than 76% of pet cats showed prey selectivity. The authors stated small native birds were the most common prey type that pet cats showed selectivity towards. Similarly, in the same paper, unowned cats were observed at two sites, showing selectivity towards rabbits. Rabbit frequency of occurrence (FOO) in unowned cat diet during a high abundance period was between 42 and 57%. In comparison, during the low rabbit abundance period, rabbit FOO was still as high as 49%, which was above the lower range during the high rabbit abundance period. The authors found despite other native prey being accessible, cats still preferred rabbits. These findings are similar to those of Molsher et al. (1999) who found rabbit predation pressure was high even as rabbit numbers declined. In Dickman and Newsome's (2015) study, pet cat prey selectivity observations were made by owners and cats were located in Sydney, New South Wales, a highly modified environment. In comparison, unowned cat prey selectivity was determined using scats in peri-urban and natural environments. These factors could be the reason for different prey preferences (i.e. native versus introduced) found by the authors. The efficiency in catching preferred prey may be the reason for prey selectivity. Dickman and Newsome (2015) found efficiency in catching non-preferred prey was lower than that for preferred prey. For example, when cats hunted rodents or rabbits, cat hunting success rates ranged from 83–100%. In contrast, their success rate fell to 50% for non-preferred prey. Incidentally, three of the cats in the hunting efficiency study by Dickman and Newsome (2015) were observed in the United Kingdom (UK). The remaining 12 cats were observed in Australia. Of all 15 cats, two of the UK cats and three Australian cats showed significant differences in hunting efficiency. However, the authors did not calculate p-values for cats observed less than 10 times. Additionally, no comparison between countries is made. It is possible cats in each country showed different hunting success rates for the same species but, it is unclear as the data is not supplied.

Cats also show preference for ground dwelling and feeding animals. Tidemann et al. (1994) found that although black-eared flying foxes, *Pteropus melanotus*, are arboreal/flying mammals, they are likely to become prey to cats when feeding on the ground. Similarly, ground feeding pigeons fell prey to cats and dominated the prey FOO when staples such as rabbits were not present. Yet, these ground feeding animals did not regularly fall prey to cats when in trees, further confirming cat preference for ground prey.

2.3.3 Prey size

Cats are known to prey on animals that fall within what is known as the critical weight range (CWR) (Burbidge & McKenzie 1989; Johnson & Isaac 2009). The CWR is defined as animals with mean body weights between 35 g and 5,500 g (Burbidge & McKenzie 1989). Other factors including cat age, sex, environment type and body mass or weight influence what size prey is taken (Pearre Jr & Maass 1998; Denny 2005; Kutt 2012; Fleming et al. 2020).

Previous literature suggests that as cat size increases, so does prey availability and size (Denny 2005; Kutt 2012). This suggests larger cats, such as sexually mature male cats (tom cats), will consume larger prey and have a wider range of prey available to them. For example, tom cats have been known to prey on larger animals such as black-footed rock-wallabies, *Petrogale lateralis* (Read et al. 2019; Fleming et al. 2020). Tom cats that consumed the rock-wallabies, were of a similar weight range to the rock-wallabies (Department of Biodiversity Conservation and Attractions 2017; Read et al. 2019), further suggesting prey size is related to cat size. This is supported by previous research by Moseby et al. (2015) who found male cats were more likely to consume larger prey items than females.

Prey size is also dependent on the age of the cat. In research by Fleming et al. (2020), it was found older cats killed and consumed larger prey than younger cats. However, the authors found no difference between male and female cat diet composition, despite males being 20% larger than females. This suggests age is a stronger predicator of prey size than sex. The authors concluded that the experience of older cats in hunting prey was why they were able to take larger prey than younger cats. These findings are in contrast to Pearre Jr and Maass (1998), who stated there is no relationship between prey size and cat size. It is possible that the contrasting findings are not related to sex, age or size, but rather prey selectivity. However, none of the studies assessed this.

2.3.4 Bite size and force

The size of a cat's bite can be a predictor of the prey consumed by cats (Sicuro & Oliveira 2011; Fleming et al. 2020). For cats, head width is measured and used to estimate bite size (Sicuro & Oliveira 2011; Yip et al. 2014). Previous research by Yip et al. (2014), established the relationship between bite size and prey availability. It was found that as bite size increases, so does prey availability.

Bite force has been used as a predictor of prey size in felids (Sicuro & Oliveira 2011). For example, cats with greater bite force are able to consume larger prey (Fleming et al. 2020). This implies that cats with greater bite force have more prey available to them, thereby are a threat to more species. Bite force is determined by the body mass of the cat (Fleming et al. 2020). As a result, male cats and older cats typically have greater bite force than females and young cats (Fleming et al. 2020). From these findings, it is clear bite size, bite force and cat size are interrelated and have an impact on prey availability.

2.3.5 Diet composition

There are other factors that affect diet of unowned cats such as geographical location, season, and climate zone, which are interrelated (Barratt 1997; Doherty et al. 2015; Woinarski et al. 2018a; Parsons et al. 2019).

In Australia, native mammals make up over 50% of overall mammal prey in unowned cat diet (Murphy et al. 2019). Mammal FOO in unowned cat diet is highest in temperate and tropical climate zones (Tidemann et al. 1994; Barratt 1997; Molsher et al. 1999; Denny 2005; Crawford et al. 2020). For example, at Lake Burrendong in New South Wales, located within a temperate climate zone, mammal FOO in unowned cat diet was 100% (Molsher et al. 1999).

Though mammals are still consumed in arid and semi-arid climate zones, reptile consumption is higher in these zones (Doherty et al. 2015; Woinarski et al. 2018a). In fact Woinarski et al. (2018a) has suggested reptile consumption in Australia by unowned cats, is higher than continents such as North America and Europe. Doherty et al. (2015) found the presence of reptiles in unowned cat diet was highest in arid climate zones compared to tropical and temperate climate zones. Reptile presence in unowned cat diet is also related to temperature with more reptiles consumed in regions that experience high temperatures (Doherty et al. 2015; Woinarski et al. 2018a). Precipitation also plays a role in what prey is consumed by unowned cats. Across Australia, reptile consumption increases as rainfall decreases (Woinarski et al. 2018a). This is in line with findings from the United States where reptile consumption is highest and mammal consumption lowest during and post drought years (Parsons et al. 2019). However, these findings are in contrast to Molsher et al. (1999) who found that reptile FOO in unowned cat diet did not change between seasons or as a result of dry conditions. Additionally, Barratt (1997) also found reptile FOO did not differ between seasons. The studies by Barratt (1997) and Molsher et al. (1999) were conducted in temperate climate zones. The study by Woinarski et al. (2018a) used research from all climate zones in Australia however, arid, and semi-arid research collated by the authors dominated their study. Different climate zones and their different prey assemblages could be the reason for differing results between the studies.

Cats are responsible for 40 bird species extinctions globally, and of all prey types predated on by cats, bird species are most at risk of becoming extinct (Taylor 1979; Doherty et al. 2016b). In Australia, bird FOO in unowned cat diet ranges between 4.2 and 92.8% with the mean FOO being 31.6% (Woinarski et al. 2017). Cats tend to prey on ground dwelling or feeding birds (Tidemann et al. 1994; Fleming et al. 2020; Gaiotto et al. 2020). For example, Greenwell et al. (2019), found that a single cat was responsible for the nest failure of a colony of Australian fairy terns, *Sternula nereis nereis*, though the authors noted predation by native species on the fairy terns as well as other disturbances. In another colony surveyed by the authors, cat predation did not occur, and the colony had a small number of fledgelings survive. In contrast, the colony with high fairy tern mortality attributed to the cat, had no fledgelings survive.

Amphibians are consumed to a lesser extent by unowned cats than other vertebrates. Though millions are killed by cats each year (Woinarski et al. 2020), they do not form a major component of their diet.

Invertebrates in some cases have a higher FOO than mammals in unowned cat diet, but species level identification of invertebrate prey is often not reported in cat diet studies. Instead, prey is usually categorized by taxonomic order. In Australia, Orthoptera, comprising grasshoppers and crickets, is the most commonly consumed order by unowned cats (Woolley et al. 2020). Invertebrates are likely to be underreported due to the these animals being soft-bodied, thereby being easily digestible (Woolley et al. 2020). Additionally, invertebrates are often not counted in diet studies, therefore can be underrepresented (Pearre Jr & Maass 1998; Woolley et al. 2020). The impacts from cats on invertebrates are less frequently reported (Burbidge & McKenzie 1989; Doherty et al. 2016a) and with many invertebrate species yet to be described (Preece et al. 2014), the impact of cats is difficult to predict.

The presence of anthropogenic items in unowned cat diet is dependent on location or availability (Denny 2005; Crawford et al. 2020; Fleming et al. 2020). For example, unowned cats located at rubbish tips or near farmland have a high FOO of anthropogenic items in comparison to cats living in natural environments (Denny 2005). This has been reflected in a study by Fleming et al. (2020) who found urban cats consumed more anthropogenic waste than their rural counterparts. Deliberate feeding by members of the public also contributes to anthropogenic items in unowned cat diet (Lohr & Lepczyk 2014; Gunther et al. 2016; Swarbrick & Rand 2018). Nevertheless, even when anthropogenic items are readily available, cats will still prey on mammals (Denny 2005; Crawford et al. 2020).

2.3.6 Limitations of diet studies

Majority of diet composition studies for unowned cats use FOO. However, FOO has limitations when used in diet studies. One limitation with using FOO for the purpose of researching BC, is it does not indicate nutritional value (Pinkas 1971). Another limitation of FOO is that invertebrate FOO in unowned cat diet can be higher than mammal FOO (Woolley et al. 2020). Despite this, invertebrates contribute a far smaller biomass than mammals do to unowned cat diet (Woolley et al. 2020). Therefore, many studies incorporate prey biomass in their results to account for these biases (Campos et al. 2007; Medina et al. 2008; Klare et al. 2011; Shionosaki et al. 2015; Parsons et al. 2019).

Klare et al. (2011) has suggested biomass measurements from captive animal feeding trials are the most accurate measurement of diet contents in carnivores. The authors admitted captive feeding trial animals would not have the same variation in diet as those in the wild, nor would they experience the same conditions. Hence, this method would not be feasible for unowned cats in Australia, as multiple areas across Australia would need to be fenced to gain a relatively accurate measurement using this method. Furthermore, biomass is not always an accurate measure of diet. Hart et al. (2002) has suggested large prey items that are not consumed regularly can cause bias not only from the volume of the prey but also due to slower digestion. The authors state that slower digestion would cause an individual animal consumed by a cat to appear in multiple scats. In addition to this, research on methods used to analyse scats found certain biomass measurements can return negative values, leading to calculation errors (Klare et al. 2011).

As a result, ecologists are increasingly using the index of relative importance (IRI) to reduce biases arising from prey FOO and biomass (Risbey et al. 1999; Hart et al. 2002; Medina et al. 2008; Yip et al. 2014). The IRI reduces bias by combining FOO, biomass measurements and numerical data for prey (Pinkas 1971). However, like FOO, the IRI still has a shortcoming in that it does not measure the nutritional value of food items (Pinkas 1971). Despite this, the IRI is the most accurate and simple method used to assess diet composition (Hart et al. 2002).

All calculations used to measure diet have a bias in that not all prey killed by unowned cats are consumed, which is known as surplus killing (Greenwell et al. 2019). Additionally, scat and stomach content analysis only provide a snapshot of an animal's diet (Peachey et al. 2000; Nielsen et al. 2017). Therefore, scats and stomach content samples cannot be interpreted as an animal's entire diet.

2.4 Body condition (BC)

BC is a measure of nutrient profile, feeding regime, and body fat content (Burkholder 2000; Labocha et al. 2014). Optimal BC is dependent on the performance goal (Burkholder 2000; Labocha et al. 2014). For example, an animal bred for human consumption would have a different optimal BC to that of a racing animal. In companion animals (i.e. cats and dogs) BC is important to determine if an animal is obese and at risk of obesity related diseases (Scarlett & Donoghue 1998; German et al. 2006). Likewise, it can determine if an animal is emaciated and experiencing nutritional stress (Gunther et al. 2018). Reproductive status and contraceptive status also have an impact on BC in cats (Russell et al. 2000; Gunther et al. 2018). BC in pregnant animals is particularly important with both extremely low and extremely high BCs being related to pregnancy and birthing problems and poor milk yield (Verstegen et al. 2008; Fontaine 2012; Koyuncu & Altinçekiç 2013).

Some authors have indicated that BC is a poor measurement of an animal's fitness (Labocha et al. 2014; Wilder et al. 2016). However, the studies referred to by

Labocha et al. (2014) and Wilder et al. (2016) involved small mammals with fast metabolisms, which is not the case for cats (Kleiber 1947).

It is thought that if cats are in ideal condition, they are more likely to survive and reproduce (Jones & Coman 1982). Correspondingly, cats in poor condition are less likely to survive and breed (Jones 1977). If we can understand what prey are causing higher BC in cats, managers can potentially look at managing their prey as well, if they are non-natives. Additionally, when cats experience nutritional stress, or there is a lower abundance of their preferred prey, they are more likely to be susceptible to management tools that implement food baits, such as trapping and poison baiting (Short et al. 1997; Moseby & Hill 2011; Christensen et al. 2013; Comer et al. 2020). In contrast, if cats are in good condition, these control measures may not be as effective due to the cats already getting their nutritional needs from their prey.

There are numerous methods used to measure BC in different mammals. Due to the many different body condition indices (BCIs), only those that have been used or have the ability to be used on cats have been reviewed here.

2.4.1 Morphometric indices

Morphometric indices utilise the relationship between weight and a morphometric measurement to determine BC (Krebs & Singleton 1993; Denny 2005; Labocha et al. 2014). Morphometric measurements used can be linear or non-standard (Labocha et al. 2014). Denny (2005) states several linear measurements including head length, head width, head/body length, tail length and femur length can be used as a BCI in cats. Other linear body measurements include pes length which measures the length of the paws (Bakker & Main 1980; Fleming et al. 2020), and shoulder height which has been used in goats (Natsir et al. 2010). Non-standard measurements include heart-chest girth, which is measured around the thorax, on the ventral side of the forearms (Natsir et al. 2010) and pelvic girth (Labocha et al. 2014). Therefore, any morphometric measurement can theoretically be used as a BCI however, not all are guaranteed to give a reliable estimate of BC (Denny 2005).

In Denny's (2005) study, linear regression was used to assess the relationship between cat weight and linear measurements as per the method used by Krebs and Singleton (1993). For female cats, the strongest linear relationship with weight was head length (Denny 2005). In contrast, the strongest linear relationship with weight in males was femur length. Denny (2005) states femur length is the most accessible index to use in the field hence, their study provided linear regression models using femur length and body weight. Yip et al. (2014) also successfully used femur length to measure BC of unowned cats in semi-arid Queensland. Therefore, measuring femur length and assessing its relationship to body weight is a reliable tool in assessing BC in cats.

2.4.2 Body condition score (BCS)

BC scoring is used in many mammals including ungulates (Ferguson et al. 1994; Mattiello et al. 2009), rodents (Ullman-Culleré & Foltz 1999), lagomorphs (Cardinali et al. 2008), canids (Behrendorff et al. 2016), and felids (German et al. 2006; Junginger et al. 2015). In the livestock industry, BCSs are used to determine nutritional status and market value of the animal (Burkholder 2000). In animals such as cats and dogs, BCSs are used to determine if an animal is obese, ideal, emaciated, or in between, depending on the scale used (German et al. 2006). More succinctly, Burkholder (2000, p. 650) describes BCS as ‘a subjective, semi-quantitative method of evaluating body fat and muscle’.

For cats, BCS is more often than not, done on a 5-point or 9-point scale (Scott et al. 2002; German et al. 2006; Corbee 2014; Peterson et al. 2016; Gunther et al. 2018; Zito et al. 2019). In the case of unowned cats, BCSs vary, but typically juveniles have lower BCSs than adults (Gunther et al. 2018). Unowned cats are also regularly scored as either in ideal or just below ideal BC (Scott et al. 2002; Crawford et al. 2020). Due to its non-invasive nature, BC scoring can quickly and easily be implemented into studies.

As BC scoring is done subjectively by an observer, this can lead to bias if the observer is not skilled (German et al. 2006). Although, Ferguson et al. (1994) previously found even skilled observers can have variation in the values they give for BCS. Ferguson et al. (1994) found skilled observers disagreed on BCS 21–34% of the time and agreed 58–67%. This suggests comparing BCSs between studies is not reliable. Another bias with BCS has been observed between age classes. Mattiello et al. (2009) found hunters that scored the BC of juvenile red deer, regularly underscored them as being in poor condition. The authors believe this bias was due to hunters perceiving adults being in better condition due to their larger size. Similarly, many authors have found pet owners often incorrectly score their pet’s BC (Russell et al. 2000; Courcier et al. 2011; Eastland-Jones et al. 2014; Gerstner & Liesegang 2017). These findings make it clear that errors in measuring BCS are common.

2.4.3 Kidney fat index (KFI)

Kidney fat has been used as an index of total body fat in rabbits (Rioux & Diouf 2006), deer (Suzuki et al. 2004; Serrano et al. 2008; Mattiello et al. 2009), grey wolves, *Canis lupus* (Lajeunesse & Peterson 1993), dingoes (Behrendorff et al. 2016), and cats (Hayde 1992; Short et al. 1997; Yip et al. 2014; Yip et al. 2015). To calculate the KFI, the kidney of the animal is removed with surrounding fat but no other tissue (Serrano et al. 2008). Fat perpendicular to the kidney is removed and then the kidney is weighed with the remaining fat attached (Rioux & Diouf 2006). The remaining fat is then cut off the kidney and weighed alone (Rioux & Diouf 2006). Rioux and Diouf (2006) found a positive correlation between KFI and total body fat in rabbits suggesting KFI is an appropriate alternative to measuring total body fat. Furthermore, KFI is suitable to observe BC changes between seasons, in deer populations. Mattiello et al. (2009) found red deer, *Cervus elaphus*, stag KFI changed seasonally. However, KFI can vary between species (Serrano et al. 2008). For example, roe deer, *Capreolus capreolus*, and fallow deer, *Dama dama*, have differing kidney sizes which KFI does not account for (Serrano et al. 2008). Therefore, if a deer's kidney is large, then the KFI will be high and vice versa. Additionally, Mattiello et al. (2009) reported KFI is not suitable to be used between all age classes and sexes as it varies significantly. For example, comparisons of BC cannot be made between juveniles and adults (Mattiello et al. 2009). This suggests a KFI range would need to be calculated for each age class and sex, and new data compared to these ranges.

KFI has been used less often to assess BC in carnivores. Currently only four studies have used kidney fat to measure BC in cats, all of which were conducted in Australia

(Hayde 1992; Short et al. 1997; Yip et al. 2014; Yip et al. 2015). In the studies by Hayde (1992) and Yip et al. (2014), it was found KFI correlated with other BC measurements. However, the use of KFI in cats should be viewed cautiously as they do not follow the principles used to measure KFI as stated above. Instead, they measure fat coverage on the kidney subjectively and use this as a KFI (Hayde 1992; Yip et al. 2014). Though these studies state kidney fat has been used as an index of BC, one could argue that measuring fat coverage is more like the BCI of using visceral fat. Additionally, subjectively assessing fat cover may not be accurate. In contrast, Behrendorff et al. (2016) included thickness, measuring to the nearest millimetre, when assessing fat cover on kidneys in dingoes. Measuring the kidney fat thickness means results can be easily compared between studies, unlike subjective measures (Burkholder 2000).

KFI has also been evaluated for its accuracy in determining BC in grey wolves, and Lajeunesse and Peterson (1993) found KFI in grey wolves was correlated with subcutaneous, visceral and marrow fat. Lajeunesse and Peterson (1993) also found kidney fat was the last fat store to be depleted. This suggests if an animal has depleted kidney fat stores, it is in significantly poor condition.

With minimal research on the use of KFI in cats, researchers can use other BC measurements and compare them to KFI to determine its validity. With accurate KFI results in other carnivore species, it is plausible KFI is a valid method for measuring BC in cats.

2.4.4 Mesentery, visceral and subcutaneous fat scores

Mesentery fat can be assessed visually by a trained assessor, categorising into a point scale, incrementing up from no fat present to excessive amounts present (Yip et al. 2014; Behrendorff et al. 2016). Another way to measure mesentery fat is to dissect it out of the body cavity and weigh it (Hayde 1992).

Visceral fat is the fat that covers internal organs. Although kidney fat is a form of visceral fat, it has already been discussed above and is not mentioned again in this section. Measurements of visceral fat are conducted subjectively by assessing how much fat is covering the organ and then measuring the thickness (Behrendorff et al. 2016). Subcutaneous fat is measured by thickness and typically the measurement is taken from the abdomen (Hayde 1992; Junginger et al. 2015; Behrendorff et al. 2016).

2.4.5 Imaging

BC can be measured using imaging and is useful when non-invasive methods are needed (Michel et al. 2011; Hall et al. 2018) however, animals do need to be anaesthetised for the procedure (Castro et al. 2015).

Dual X-ray absorptiometry (DEXA) has previously been used to measure the body fat in cats (Launten et al. 2000; Scott et al. 2002; German et al. 2006). The studies by Scott et al. (2002) and German et al. (2006), found the DEXA results were correlated with BCSs given by assessors, suggesting DEXA is a viable BCI in cats.

Magnetic resonance imaging (MRI) has previously been used to determine body fat in dogs (Castro et al. 2015). The method described by Castro et al. (2015), involves imaging of subcutaneous and visceral fat. Surprisingly, no evidence of the use of

MRI to measure BC in cats has been reported in the literature. However, accuracy of the MRI in assessing canine body fat (Castro et al. 2015) is a promising start for its use in other carnivores.

Though out of the scope of this review, the monetary expense to utilise DEXA, and MRIs is likely to exceed the benefits gained from using these methods on unowned cats. Additionally, equipment such as these are not always readily available (Scott et al. 2002).

2.5 BC-diet relationship

Few studies have been conducted on unowned cat BC and its relationship to diet. Literature does, however, exist for pet cats, hence its inclusion.

Caloric restriction is related to weight loss in pet cats (Hoelmkjaer & Bjornvad 2014; Spofford et al. 2014). For example, pet cats fed distinct meals have lower BC than those with continuous access to food (Russell et al. 2000). Diet also has a direct influence on BC through the gross amounts eaten (Hayde 1992; Crawford et al. 2020). For instance, during periods of lower prey abundance (e.g. winter), unowned cat BC will decrease (Hayde 1992; Crawford et al. 2020). However, loss of BC may also be attributed to energy expenditure due to thermoregulation (Serisier et al. 2014). From these results, it can be confidently assumed that unowned cats that consume lower volumes of prey are likely to be in poorer condition.

A series of studies have found that pet cat BC is dependent on diet (Alexander et al. 2014; Coradini et al. 2014; Hoelmkjaer & Bjornvad 2014; Spofford et al. 2014; Allaway et al. 2018). For example, previous literature on pet cats found high protein diets resulted in an increase in BC (Coradini et al. 2011; Coradini et al. 2014; Allaway et al. 2018). This may explain why unowned cats that consume prey high in protein are often in ideal BC (Plantinga et al. 2011; Yip et al. 2014). Conversely, a study by Crawford et al. (2020) found BC of unowned male cats had no relationship to diet. Yet the authors suggested diet was a factor in female cat BC. Likewise, Yip et al. (2015) found no difference in BC between unowned cats sampled during periods of high prey abundance and periods of low prey abundance. Studies that have found a relationship between diet and BC were conducted on the same sample of pet/owned cats, over a period of time. Dissimilarly, the studies that found no relationship between diet and BC were conducted on different individual unowned cats as they were culled. This implies that using different samples from a population rather than observing the same samples, can give inaccurate results. Evidence clearly exists on the effects of diet on BC in cats however, more research needs to be conducted to ensure findings for unowned cats are similar to those of pet cats.

2.6 Cat management

In Australia, several management strategies are used for unowned cats. Though an in-depth review of the management strategies used is out of the scope of this review, it is important to understand the relationship between diet-BC and cat management.

Baiting is a management method that involves deploying toxic meat baits (Algar et al. 2011; Johnston et al. 2020). Baiting is most effective when resources are low (Algar & Burrows 2004; Christensen et al. 2013). Therefore, cats that have their nutritional needs met through prey or other food sources are less likely to consume baits (Christensen et al. 2013; Fancourt et al. 2021). For example, one study found unusually warm winter temperatures and high rainfall resulted in lower bait uptake by unowned cats (Fancourt et al. 2021). It is likely prey availability was higher during this winter study period compared to other winters.

The method behind trapping is to use a non-toxic bait to lure a cat into a trap (Molsher 2001). Like baiting, if there is no resource shortage, cats may be less likely to approach and go into traps (Algar & Burrows 2004; Christensen et al. 2013).

These findings indicate diet is a major factor in determining which cat management strategy to use. Furthermore, the BC of unowned cats can indicate resource availability, therefore can be used to determine the most effective cat management strategy.

2.7 Gaps in knowledge

The purpose of this review was to identify what is known about unowned cat diet in Australia and how BC relates to diet in unowned cats. There is extensive literature on unowned cat diet in Australia, however, the majority are based in arid or semi-arid environments (Figure 1). Some studies have been conducted in temperate zones, however none have been conducted in south-east Queensland. Additionally, BC studies for unowned cats are limited.

Finally, the relationship between BC and diet of unowned cats is seldom explored. Hence, after reviewing the literature, it is evident there is a lack of knowledge on the relationship between diet and BC in Australian unowned cats, and this thesis attempts to address this gap.

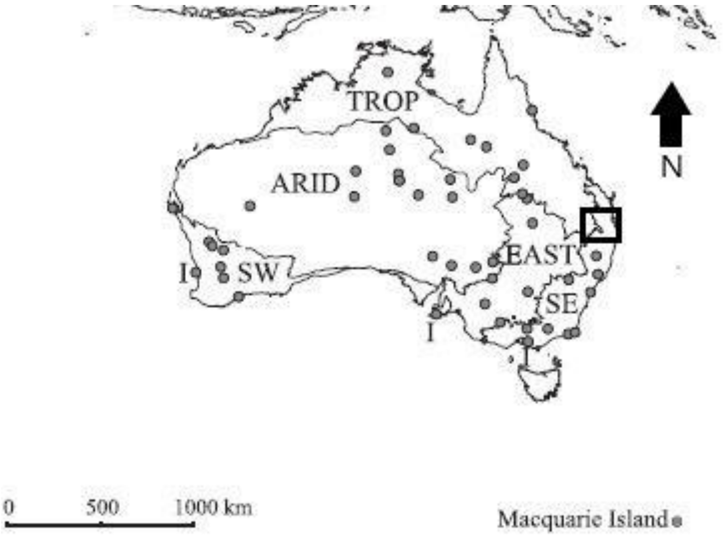

Figure 1. Locations of diet studies conducted in Australian climate zones (adapted from Doherty et al. 2015). South-east Queensland, outlined in black square.

2.8 Project aims

Unowned cat numbers in the SDR in Queensland are thought to be increasing (Magnussen 2020). However, the impacts of unowned cats on native wildlife in the region are not known. Therefore, it is necessary that diet studies are conducted to determine if native wildlife are at risk in the region. This in turn, informs local government on what management of unowned cats needs to take place to ensure native species survival. Additionally, the relationship between BC and diet in unowned cats has not been explored extensively in the literature. Determining the effects of diet on unowned cat BC, also determines management strategies for unowned cats.

This thesis investigates the relationship between BC and diet in unowned cats in the SDR, south-east Queensland and Davenport Downs Station (DDS), western Queensland. Various morphometric measurements, BCS, and KFI are used as indices of BC. FOO and IRI will determine the major prey type composition in unowned cat diets. The aims of this project are as follows:

1. Determine the overall diet and dietary diversity of unowned cats in the SDR.
2. Determine the overall BC of unowned cats in the SDR.
3. Assess effectiveness of KFI as an indicator of BC in unowned cats.
4. Determine the relationship between diet and BC of unowned cats in the SDR.
5. Investigate the relationship between diet and BC of unowned cats of DDS using unpublished data from Pettigrew (1993), and compare these to the SDR dataset.

The overall hypotheses are as follows:

1. It is hypothesised that SDR unowned cat diet will consist mostly of small mammals in both IRI and FOO, with their FOO being approximately 70%. This figure comes from the average small mammal consumption by unowned cats across Australia (Murphy et al. 2019).
2. It is hypothesised BC of SDR unowned cats will be in the ideal range. Previous research has found majority of unowned cats are in ideal condition (Domm & Messersmith 1990; Yip et al. 2014; Crawford et al. 2020).
3. It is hypothesised KFI outputs will correlate with other BC measurements.
4. It is hypothesised cats that consume small mammals will be in ideal condition.

5. It is hypothesised cats from DDS will consume a greater number of reptiles (arid climate zone) and their BCs will be similar to SDR cats.

Chapter 3: Methods

3.1 Study area

The primary study area was located in the SDR, Queensland (Figure 2). The two main towns from which cats were collected were Warwick (28° 12' 7.2"S, 152° 0' 21.6"E) and Stanthorpe (28° 39' 39.6"S, 151° 55' 51.6"E). The region covers 7,120 km^2 (Southern Downs Regional Council 2020a). Rural areas encompass majority of the region, with these areas used predominantly for agricultural purposes such as sheep and cattle grazing, viticulture, vegetable and grain crops, fruit orchards, and forestry (Southern Downs Regional Council 2020b).

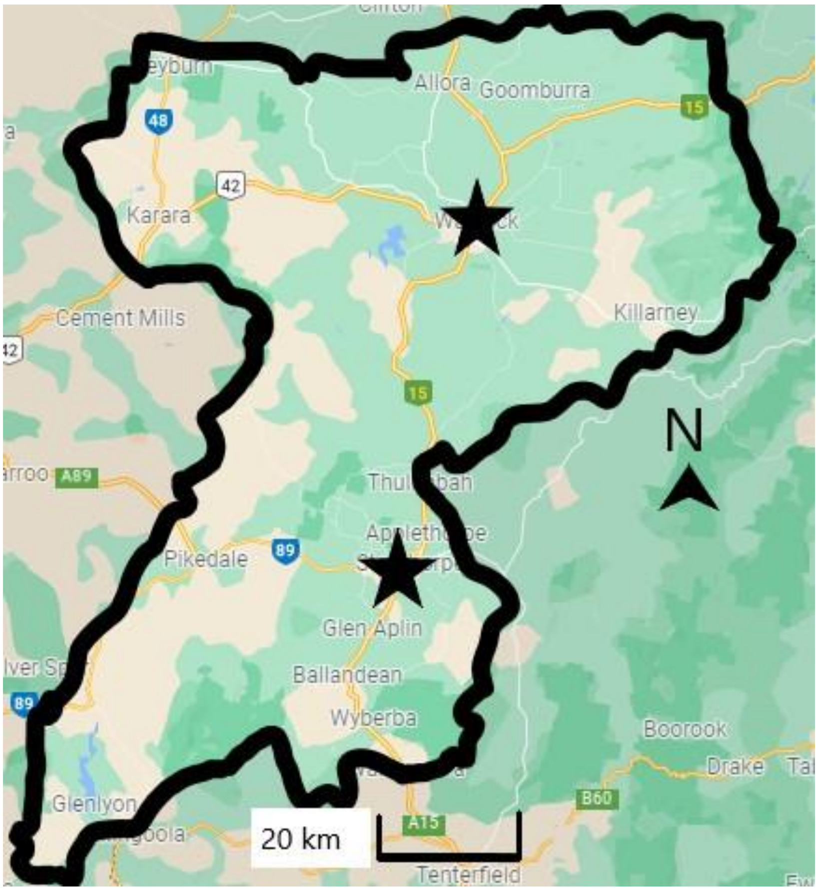

Figure 2. Map of study region, outlined in black. Black stars indicate main town centres of Warwick and Stanthorpe (adapted from Google Maps 2021).

The region is located within a temperate climate zone and experiences warm summers and cold winters (Bureau of Meteorology 2016). In Warwick, the highest mean temperature of 30.5°C occurs in January, while the lowest mean temperature of 3.0°C occurs in July (Bureau of Meteorology 2021b). In summer, Stanthorpe reaches its maximum mean temperature of 27.5°C in January, while in winter, the minimum mean temperature of 1.0°C occurs in July (Bureau of Meteorology 2021a). Warwick and Stanthorpe receive annual rainfall of 55.53 mm and 63.48 mm respectively (Bureau of Meteorology 2021a, 2021b).

In September 2019, several large bushfires occurred in the region (Southern Downs Regional Council 2020b). Additionally, as of May 2021, the region is drought declared (The Long Paddock 2021).

3.2 Veterinary clinic recruitment

Seven veterinary clinics were located within the region (three in Warwick, two in Stanthorpe, one in Allora, and one in Killarney). All clinics were contacted via phone and email for recruitment. Clinics were asked to retain any euthanased cats deemed feral or stray that would normally be discarded. In the SDR, unowned cats that come into pounds or vets undergo visual, behavioural and biosecurity assessments before they are either rehomed or euthanased (Appendix A). Two clinics were regularly visited for collection of euthanased cats (one in Warwick, and one in Stanthorpe). Other vets participated intermittently.

3.3 SDR species list

As of 2013, there are 629 identified vertebrate species that inhabit the SDR (Department of Environment and Science 2013b). Of these, 23 are introduced from overseas and one is introduced from interstate. Of the small mammals falling within the CWR, eight species are listed as vulnerable under the *Nature Conservation Act 1992* and 10 are listed as either endangered or vulnerable under the *Environmental Protection and Biodiversity Conservation Act 1999*. A full list of the vertebrate species present in the SDR is shown in Appendix B.

3.4 Animal ethics

Due to the nature of the project, ethics approval was not required, and an exemption was issued by the University of Southern Queensland, Animal Ethics Committee (AEC exemption number: 21EXE001).

3.5 Cat necropsy and data collection

Cats were collected between March and July 2021. Cats were stored frozen at -18^{o}C by vets and pounds until collection. Cats were then transported to a wet lab at the Department of Agriculture and Fisheries, Toowoomba, and kept frozen until necropsies could be conducted. Prior to necropsies, cats were thawed for 24 hours at room temperature.

Photos were taken of each cat in a lateral recumbent position pre-necropsy. Cat coat colours and whether it was a shorthair, medium-hair or longhair were noted. Cat coats were searched for the presence of parasites. The body was searched for any signs of injury and noted, and any post-euthanasia damage acquired during handling was excluded. Teeth were checked for rotting, broken teeth, plaque, complete loss, and wear. Internal parasites were noted when collecting stomach contents and faecal samples. Signs of recent pregnancy were evidenced by the presence of scars in the fallopian tubes and/or lactation.

3.5.1 Age

Cats were assigned an age category based on a visual 3-point scale (1 = juvenile, 2 = adult, 3 = geriatric). Age was determined by the physical size of the cats, tooth eruption and wear (broken, worn, rotting, plaque, tartar), and coat condition (patching). Due to time constraints, more precise aging methods such as tooth morphometrics (Fleming et al. 2021) or bone fusions (Scott et al. 2002) were not used as age determinants.

3.5.2 Sex

Cats were sexed by physical examination of genitalia by assessing distance between genitals and anus and the presence of testes. Reproductive organs were also collected as samples for an unrelated study.

3.5.3 Morphometrics

The weight of cats was measured to the nearest 50 g and collected using a pesola spring scale with a maximum of 10 kg. Nose-tail length was measured to the nearest 0.5 cm using a measuring tape. Forearm length, femur length, and head width were measured to the nearest mm using vernier callipers. For the femur, the femoral head and medial condyle were exposed on the left femur to allow accurate measurement of the femur bone (Bakker & Main 1980). The right femur was used in a small number of cases when the left femur had been broken or damaged. The ends of the forearm were determined externally by palpation of the olecranon and the carpus of the left forearm. Head width was measured at the widest point.

3.5.4 Stomach and faecal content analysis

Digestive tracts were removed from the body cavity and stomachs were dissected away at the gastro-oesophageal junction and the pylorus. The colon was dissected away from the digestive tract at the ileocaecal junction and as close to the anus as possible. Stomach and colon contents were removed, placed onto drying trays, and dried at 50°C for at least 48 hours. Contents were then placed in zip lock bags and sent to a specialist service provider for analysis (G. Story, Scats About, Majors Creek, NSW).

Prey items were identified to the lowest taxonomic group possible. Food items were categorised into mammals, reptiles, birds, amphibians, invertebrates, vegetation, cat food, and other anthropogenic items. For certain analyses, mammals were further categorised as small (mean body weight ≤ 5,500 g) and large (mean body weight > 5,500 g) mammals.

Diet composition was assessed using FOO%, volume (VOL%), body mass (BM) and IRI. Prey volume was estimated visually and given as a percentage of the total stomach or faecal sample. Prey BM was estimated by using resources available on each species (Olsen et al. 2006; Pizzey & Knight 2012; Queensland Museum 2020, 2021). IRI was calculated using the formula:

$$IRI = (BM\% + N\%)FOO\%$$

Where:

IRI = index of relative importance

N% = numerical percentage

BM% = biomass percentage

FOO% = frequency of occurrence percentage

N% is the total number of an individual prey item given as a percentage of the total number of prey items. BM% is calculated by multiplying the FOO of an individual prey item by its weight to get the BM. The BM is then divided by the total prey BM and given as a percentage.

Faecal and stomach content analysis was conducted both separately for comparison and together and noted as gastrointestinal tract (GIT) contents. When assessing GIT contents, any food type/item that appeared in both the stomach and faecal sample of an individual cat were recorded as a single occurrence.

3.5.5 BCI

For this project, three BCIs were used. BCS was the first BCI measured during necropsy. Cats were subjectively measured by one assessor on a 5-point scale (1 = emaciated, 2 = poor, 3 = ideal, 4 = overweight, 5 = obese) by visual assessment and palpation of spine, ribs, and pelvis (Lund et al. 1999; Brooks et al. 2014; Animal Welfare Victoria 2021). A diagram of how BCS evaluations were conducted is shown in Appendix C.

The relationship between morphometric measurements and body weight were assessed using residual values obtained from linear regression analyses, described below in Section 3.7.

KFI was the final BCI measurement collected. The method used to assess KFI was similar to that used by Rioux and Diouf (2006). The right kidney and attached kidney fat were removed from the body cavity of the cat. The fat perpendicular to the kidney was removed using either surgical scissors or a scalpel. The kidney with the attached fat was weighed to the nearest mg. The fat was then cut away from the kidney using a scalpel and weighed alone. The below equation was used to determine KFI:

$$KFI = \frac{KF}{KF + K} \times 100$$

Where:

KFI = kidney fat index

KF = kidney fat

K = kidney

KFI values were analysed using a linear regression against weight. The method used is described in Section 3.7.

3.6 BC-diet relationship

All three BCIs were used to compare BC and diet. Morphometric residuals were compared with diet using chi-squared tests. Due to the low number of samples within each standardised residual BC value, emaciated and poor cats were grouped together as were overweight and obese cats. The relationship between BC and diet was also assessed using ANOVAs and comparing the FOO of food items between BCSs. The BC-diet relationship was also assessed using student’s T-tests in which cat KFI values were compared between those that consumed mammals and those that did not.

3.7 Statistical analysis

Raw data was collated in Microsoft Excel and statistical analyses were conducted using SPSS version 28.0 for Windows (IBM Corp 2021).

Datasets were first assessed for normality using the Shapiro-Wilk test and visual inspection of histograms, box plots, and Q-Q plots. If outliers were identified, these were removed, and the normality tests were re-run. If data still failed the normality test, non-parametric tests were used.

To compare dietary diversity between cats from Stanthorpe and Warwick, Brillouin's Index was used (Brillouin 1956). The equation is:

$$H = (lnN! - \sum lnn_i!)/N$$

Where:

H = species diversity

N = total number of individual prey items

n_i = number of individual prey items for a particular species

Values close or equal to zero indicate low species diversity. Values close to the top range of 4.5 indicate high species diversity.

Pianka's Index was used to compare the similarity between cats sampled at each location using the equation (Pianka 1974):

$$O_{jk} = (\sum p_{ij}p_{ik})/(\sum {p_{ij}}^2 \sum {p_{ik}}^2)^{0.5}$$

Where:

O = the similarity

j = samples from first location

k = samples from second location

p_i = FOO of ith prey item or type.

A value of zero indicates no similarity in diversity while a value of one indicates identical diversity between samples.

Due to their rapid changes in morphometrics, juveniles were separated for the majority of statistical analyses to avoid skewing the data (Denny 2005). When analysing BCIs, males and females were assessed separately due to dimorphism.

The relationship between body weight and morphometric measurements were assessed using Pearson correlations and regression analyses. This method was also used to determine BC from KFI values. The residual values were standardised to allow comparison between these, BCSs and KFIs. Residual values were rounded to the nearest whole number. Residuals were then related to BC as follows, -2 = emaciated, -1 = poor, 0 = ideal, 1 = overweight, and 2 = obese. To determine whether there was a relationship between BCSs and residuals, a Spearman rank test was performed.

ANOVA and post-hoc tests were used to check for significant differences between BCS and morphometric residual BC values.

3.7 DDS data collection

DDS data was obtained from unpublished data from Pettigrew (1993). The data was collected in 1992, incidental to other research being undertaken in the area. The study area was located in southwest Queensland, an arid climate zone. In July 1992, 189 cats were shot. Of these, 59 were randomly sampled with age, sex, weight, BC, coat colour, and stomach contents recorded. A list of vertebrate species present in the area, as of 2013 is listed in Appendix D.

Chapter 4: Results

4.1 SDR dataset

4.1.1 Cat necropsy

A total of 68 cats were obtained and necropsied for this project. Fifty-four of these were collected from Stanthorpe, 13 were collected from Warwick, and one from Allora. The majority of cats were domestic shorthairs with only one domestic longhaired cat. Coat colours varied, with the predominate colours being tabby (46%), ginger/cinnamon (19%), black (15%), black and white (13%) and tortoiseshell (7%). Eight cats had external parasites (e.g. fleas) evident in their coats, however only two cats had evidence of internal parasites in their stomach contents.

Five cats had evidence of external injuries, including two cats with partially missing tails. An additional cat appeared to have suffered internal bleeding. Nine cats had either missing, rotten, or worn teeth, one of which was a geriatric female.

Of the adult females, 37% had evidence of a previous pregnancy. Two adult females were pregnant. One adult male was noted as having small testes.

4.1.2 Age

Ages recorded for the 68 cats were 30 juveniles, 37 adults, and one geriatric.

4.1.3 Sex

Of the 68 cats, 35 were female and 33 were male. Of these, 16 were juvenile females and 14 were juvenile males, with 19 adult males and females each. Three of the 68 cats were neutered, of which, two were adult males and one was an adult female.

4.1.4 Weight

A Shapiro-Wilk test and visual inspection of graphical data showed all weights were normally distributed (Appendix E). The mean weight for all cats was 2,727 g, and for females and males respectively was 2,328 g and 3,149 g. The mean weight for adult cats, inclusive of the single geriatric cat was 3,919 g. The adult mean weight for males and females was 4,642 g and 3,196 g respectively. The mean weight for all juvenile cats was 1,217 g. The mean weight for male and female juveniles was 1,124 g and 1,298 g respectively. There was a significant difference between mean adult weights of males and females (t = 2.028, df = 36, p = < 0.001) but not between juvenile male and females (t = 2.048, df = 28, p = 0.319).

4.1.5 Nose-tail

Adult male and female mean nose-tail length measurements were 82 mm and 77 mm respectively. Mean nose-tail length for juvenile males and females was 46 mm and 49 mm respectively. There was no significant difference in mean nose-tail length measurements between male and female juvenile cats (t = 2.048, df = 28, p = 0.436). In comparison, there was a significant difference (t = 2.030, df = 35, p = 0.024) between adult male and female nose-tail length measurements.

4.1.6 Forearm

Adult forearm lengths ranged from 47–132 mm. Mean forearm length for adult males and females was 120 mm and 107 mm respectively. Mean juvenile forearm length for males and females was 62 mm and 67 mm respectively. A significant difference between adult male and female forearm measurements was found (U =

40.500, p = < 0.001). There was no significant different in forearm measurements between juvenile male and females (t = 2.048, df = 28, p = 0.357).

4.1.7 Femur

Mean femur length for adult males and females was 113 mm and 85 mm respectively. There was a significant difference between adult male and female femur lengths (t = 2.028, df = 36, p = < 0.001). Mean juvenile femur length for males and females was 58 mm and 64 mm respectively. Femur length did not differ significantly between the sexes in juveniles (U = 85.000, p = 0.275).

4.1.8 Head width

Adult male head width ranged between 61 and 95 mm with a mean of 75 mm. Adult female head width ranged between 56 and 71 mm with a mean of 64 mm. Male and female adult head widths differed significantly (t = 2.028, df = 36, p = < 0.001). In contrast, there was no significant difference between sexes for head width in juvenile cats (t = 2.048, df = 28, p = 0.437).

4.2 Diet composition

Of the 68 cats sampled for this project, 63 faecal samples and 46 stomach samples were obtained and analysed for diet contents, which was due to five cats having no faecal contents and 22 cats having no stomach contents. A total of 58 food items were present in cat stomachs and a total of 99 food items were present in cat faecal samples. A total of 13 species of mammal were consumed by cats (Appendix G). Of these, six were introduced species. House mice were the most common mammalian prey item in cat diets. The most common native mammal in cat diet was eastern grey kangaroo, *Macropus giganteus*. None of the mammalian species detected in our sample from SDR were of conservation concern.

Vegetation was present in a total of five cat stomachs and nine cat faecal samples. Non-food anthropogenic waste was also not included as a food item but was noted in 10 GITs. Anthropogenic food items such as food scraps were included in food type analyses.

When all food items were analysed individually, including individual species, commercial cat food had the highest FOO% in both stomach and faecal samples with 52.17% and 36.51% respectively (Table 1). GIT contents had a commercial cat food FOO% of 60.61%. When categorised into prey types, mammals had the highest FOO% (88.89%) (Table 2). The FOO% for small mammals in stomach samples of SDR cats was 36.96% and for faecal samples was 57.14%. GIT FOO% for small mammals was 59.09%.

Table 1. FOO% of various food items in the stomach, faecal and GIT samples of unowned SDR cats.

Food item	**Common name**	**Stomach samples**	**FOO%**	**Faecal samples**	**FOO%**	**GIT samples**	**FOO%**
Bird		0	0.000	2	3.175	2	3.030
Coleoptera	Beetles	2	4.350	7	11.111	10	15.152
Commercial cat food		24	52.170	23	36.508	40	60.606
Dragon		1	2.170	1	1.587	1	1.515
Felis catus	Cat	0	0.000	1	1.587	1	1.515
Food scraps		4	8.700	10	15.873	5	7.576
Macropod sp.		1	2.170	5	7.937	6	9.091
Macropus giganteus	Eastern grey kangaroo	3	6.520	11	17.460	12	18.182
Mus musculus	House mouse	11	23.910	16	25.397	18	27.273
Notamacropus parryi	Whiptail wallaby	1	2.170	0	0.000	2	3.030
Notamacropus rufogriseus	Red-necked wallaby	1	2.170	0	0.000	1	1.515
Orthoptera	Grasshoppers	3	6.520	4	6.349	6	9.091
Oryctolagus cuniculus	European rabbit	3	6.520	5	7.937	5	7.576
Osphranter robustus	Common wallaroo	0	0.000	2	3.175	2	3.030
Petaurus breviceps	Sugar glider	2	4.350	2	3.175	3	4.545
Rattus rattus	Black rat	1	2.170	2	3.175	2	3.030
Sus scrofa	Pig	0	0.000	1	1.587	1	1.515
Trichosurus vulpecula	Common brushtail possum	1	2.170	1	1.587	1	1.515
Vegetation		5	10.870	9	14.286	12	18.182
Vulpes vulpes	Red fox	1	2.170	9	14.286	9	13.636
Wallabia bicolor	Swamp wallaby	0	0.000	1	1.587	1	1.515

Table 2. FOO% of prey types found in faecal and stomach samples of SDR cats.

Faeces n = 63		**Stomach n = 46**	
Type	**FOO%**	**Type**	**FOO%**
Mammal (all)	88.889	Mammal (all)	50.000
Small mammal	36.957	Small mammal	57.143
Large mammal	13.043	Large mammal	31.746
Bird	3.175	Bird	0.000
Reptile	1.587	Reptile	2.174
Invertebrate	17.460	Invertebrate	10.870
Vegetation	14.286	Vegetation	10.870
Commercial cat food	36.508	Commercial cat food	52.174
Anthropogenic	15.873	Anthropogenic	8.696

For stomach samples, the mean VOL% for each food type in descending order was commercial cat food (47.326%), mammals (37.093%), anthropogenic (6.163%), vegetation (4.884%), invertebrates (1.744%), reptiles (0.465%), and birds (0.000%). For faecal samples, the mean VOL% for each food type in descending order was mammals (47.379%), commercial cat food (31.690%), anthropogenic (10.638%), vegetation (6.017%), invertebrates, (3.328%), birds (0.603%), and reptiles (0.345%).

IRI was calculated for stomach, faecal, and GIT (Tables 3–5) samples. In the stomach samples, the highest ranked prey items in descending order were house mice and eastern grey kangaroos. For faecal samples, the highest ranked prey items in descending order were eastern grey kangaroos, house mice, red foxes, unidentified macropod species, and beetles. Similarly, the highest ranked prey items in descending order for GIT were eastern grey kangaroos, house mice, red foxes, beetles, and unidentified macropod species.

Table 3. Importance of prey items found in the stomach contents of unowned cats in the SDR, based on 46 stomach samples.

Prey	**Common name**	**FOO**	**N %**	**Weight (kg)**	**BM**	**BM %**	**FOO %**	**IRI**
Mus musculus	House mouse	9	31.034	0.015	0.135	0.062	19.565	608.416
Macropus giganteus	Eastern grey kangaroo	3	10.345	50.000	150.000	69.265	6.522	519.194
Oryctolagus cuniculus	European rabbit	3	10.345	1.500	4.500	2.078	6.522	81.018
Orthoptera	Grasshoppers	3	10.345	0.002	0.006	0.003	6.522	67.484
Petaurus breviceps	Sugar glider	2	6.897	0.130	0.260	0.120	4.348	30.507
Coleoptera	Beetles	2	6.897	0.002	0.004	0.002	4.348	29.993
Macropod sp.		1	3.448	19.000	19.000	8.774	2.174	26.569
Notamacropus parryi	Whiptail wallaby	1	3.448	16.000	16.000	7.388	2.174	23.558
Notamacropus rufogriseus	Red-necked wallaby	1	3.448	15.000	15.000	6.926	2.174	22.554
Vulpes vulpes	Red fox	1	3.448	7.000	7.000	3.232	2.174	14.523
Trichosurus vulpecula	Common brushtail possum	1	3.448	4.000	4.000	1.847	2.174	11.512
Dragon		1	3.448	0.375	0.375	0.173	2.174	7.873
Rattus rattus	Black rat	1	3.448	0.280	0.280	0.129	2.174	7.777
Osphranter robustus	Common wallaroo	0	0.000	28.000	0.000	0.000	0.000	0.000
Wallabia bicolor	Swamp wallaby	0	0.000	15.000	0.000	0.000	0.000	0.000
Sus scrofa	Pig	0	0.000	120.000	0.000	0.000	0.000	0.000
Felis catus	Cat	0	0.000	4.000	0.000	0.000	0.000	0.000
Bird		0	0.000	0.200	0.000	0.000	0.000	0.000

Table 4. Importance of prey items found in the faecal contents of unowned cats in the SDR, based on 63 faecal samples.

Prey	**Common name**	**FOO**	**N %**	**Weight (kg)**	**BM**	**BM%**	**FOO %**	**IRI**
Macropus giganteus	Eastern grey kangaroo	11	15.714	50.000	550.000	60.020	17.460	1322.349
Mus musculus	House mouse	16	22.857	0.015	0.240	0.026	25.397	581.164
Vulpes vulpes	Red fox	9	12.857	7.000	63.000	6.875	14.286	281.888
Macropod sp.		5	7.143	19.000	95.000	10.367	7.937	138.968
Coleoptera	Beetles	7	10.000	0.002	0.014	0.002	11.111	111.128
Oryctolagus cuniculus	European rabbit	5	7.143	1.500	7.500	0.818	7.937	63.185
Orthoptera	Grasshoppers	4	5.714	0.002	0.008	0.001	6.349	36.287
Osphranter robustus	Common wallaroo	2	2.857	28.000	56.000	6.111	3.175	28.471
Sus scrofa	Pig	1	1.429	120.000	120.000	13.095	1.587	23.054
Rattus rattus	Black rat	2	2.857	0.280	0.560	0.061	3.175	9.264
Bird		2	2.857	0.200	0.400	0.044	3.175	9.209
Petaurus breviceps	Sugar glider	2	2.857	0.130	0.260	0.028	3.175	9.160
Wallabia bicolor	Swamp	1	1.429	15.000	15.000	1.637	1.587	4.866
Trichosurus vulpecula	Common brushtail possum	1	1.429	4.000	4.000	0.437	1.587	2.960
Felis catus	Cat	1	1.429	4.000	4.000	0.437	1.587	2.960
Dragon		1	1.429	0.375	0.375	0.041	1.587	2.333
Notamacropus parryi	Whiptail wallaby	0	0.000	16.000	0.000	0.000	0.000	0.000
Notamacropus rufogriseus	Red-necked wallaby	0	0.000	15.000	0.000	0.000	0.000	0.000

Table 5. Importance of prey items found in the GIT of unowned cats in the SDR, based on 66 GIT samples.

Prey	**Common name**	**FOO**	**N %**	**Weight (kg)**	**BM**	**BM %**	**FOO %**	**IRI**
Macropus giganteus	Eastern grey kangaroo	12	14.458	50.000	600.000	58.110	18.182	1280.606
Mus musculus	House mouse	18	21.687	0.015	0.270	0.026	27.273	574.753
Vulpes vulpes	Red fox	9	10.843	7.000	63.000	6.102	13.636	224.271
Coleoptera	Beetles	10	12.048	0.002	0.020	0.002	15.152	177.208
Macropod sp.		6	7.229	19.000	114.000	11.041	9.091	161.204
Orthoptera	Grasshoppers	6	7.229	0.002	0.012	0.001	9.091	63.795
Oryctolagus cuniculus	European rabbit	5	6.024	1.500	7.500	0.726	7.576	49.636
Osphranter robustus	Common wallaroo	2	2.410	28.000	56.000	5.424	3.030	23.039
Sus scrofa	Pig	1	1.205	120.000	120.000	11.622	1.515	18.863
Notamacropus parryi	Whiptail wallaby	2	2.410	16.000	32.000	3.099	3.030	16.202
Petaurus breviceps	Sugar glider	3	3.614	0.130	0.390	0.038	4.545	16.113
Rattus rattus	Black rat	2	2.410	0.280	0.560	0.054	3.030	7.247
Bird		2	2.410	0.200	0.400	0.039	3.030	7.201
Notamacropus rufogriseus	Red-necked wallaby	1	1.205	15.000	15.000	1.453	1.515	3.908
Wallabia bicolor	Swamp wallaby	1	1.205	15.000	15.000	1.453	1.515	3.908
Trichosurus vulpecula	Common brushtail possum	1	1.205	4.000	4.000	0.387	1.515	2.341
Felis catus	Cat	1	1.205	4.000	4.000	0.387	1.515	2.341
Dragon		1	1.205	0.375	0.375	0.036	1.515	1.825

The Brillouin’s Index values for Stanthorpe and Warwick/Allora were 2.148 and 1.510 respectively. The Pianka’s Index similarity value for Stanthorpe versus Warwick/Allora was 0.945.

4.3 BCI

4.3.1 Morphometric indices

Although all morphometric measurements were significantly correlated with weight in adult males, the strongest relationship was between head width and weight (Table 6). Only nose-tail length and head width were significantly correlated with weight in adult females, with nose-tail length having the strongest correlation (r = 0.665, p = 0.003). All morphometric measurements were correlated with weight in juvenile males and females. The strongest correlations for juvenile males and females were forearm (r = 0.872, p = < 0.001) and head width (r = 0.877, p = < 0.001) respectively. The strongest regression analyses for the respective ages and sexes are shown in Figure 3. The results from an ANOVA and post hoc test revealed no significant difference between groups for any of the morphometric measurement residuals (Table 7).

Table 6. Pearson correlation between morphometric measurements and weight for adult and juvenile males and females.

	Morphometric measurement	**r**	**p-value**
Adult male	Nose-tail	0.644	0.003
	Forearm	0.745	<0.001
	Femur	0.664	0.002
	Head width	0.834	<0.001
Adult female	Nose-tail	0.665	0.003
	Forearm[1]	0.140	0.568
	Femur	0.366	0.123
	Head width	0.505	0.027
Juvenile male	Nose-tail	0.821	<0.001
	Forearm	0.872	<0.001
	Femur	0.846	<0.001
	Head width	0.846	<0.001
Juvenile female	Nose-tail	0.847	<0.001
	Forearm	0.844	<0.001
	Femur[1]	0.735	<0.001
	Head width	0.877	<0.001

1. Indicates non-normal data. Spearman rank test was used to assess correlation in these cases.

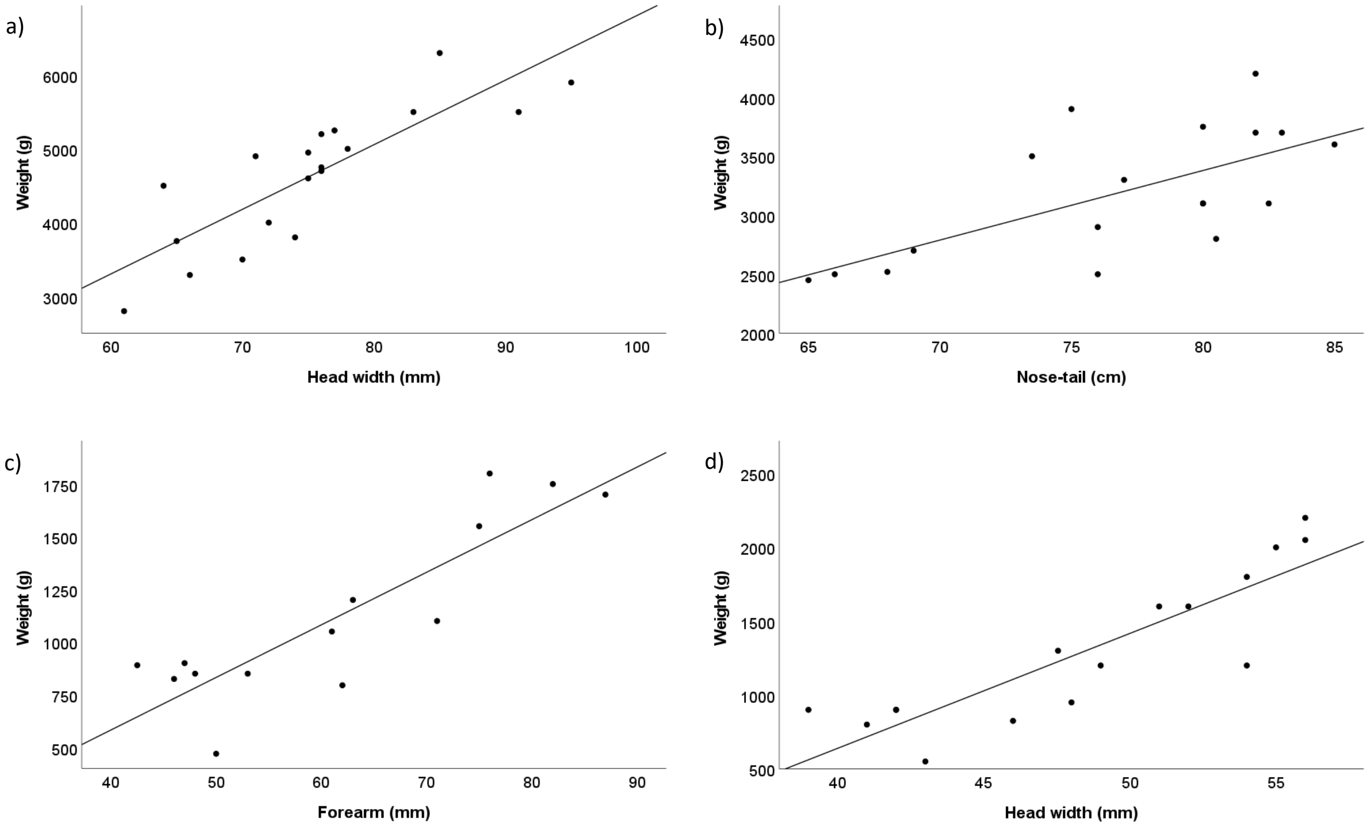

Figure 3. Relationship between body weight and head width for adult male cats. Regression line is described as y = -1950 + 87.62x (a).

Relationship between body weight and nose-tail length for adult female cats. Regression line is described as y = -1340 + 58.95x (b).

Relationship between body weight and forearm length for juvenile male cats. Regression line is described as y = -415 + 24.95x (c).

Relationship between body weight and head width for juvenile female cats. Regression line is described as y = -2470 + 77.81x (d).

Table 7. Results from ANOVA conducted between groups for various morphometric residuals and KFI residuals.

	f	df	p-value
Nose-tail residuals	0.274	3	0.844
Forearm residuals	0.089	3	0.966
Femur residuals	0.012	3	0.998
Head width residuals	0.037	3	0.990
KFI residuals	2.485	3	0.069

4.3.2 BCS

Overall, the majority of cats were given an ideal BCS (Figure 4). Juvenile male and female cats both had a mean BCS of 2 with a range of 1–3. Of the juveniles, only one male and one female had a BCS of 1 (emaciated). Adult male cats had a mean BCS of 3 with a range of 2–4. Adult female cats had a mean BCS of 3 with a range of 1–4. The single geriatric female cat had a BCS of 2. No significant difference was found between adult males and females (t = 2.028, df = 36, p = 0.057). Likewise, there was no significant difference in BCS between juvenile males and females (t = 2.048, df = 28, p = 0.867). A significant difference in BCS was found between adult and juvenile males (t = 2.040, df = 31, p = 0.001), but not between adult and juvenile females (t = 2.035, df = 33, p = 0.375). There was also a significant difference between adult cats and juvenile cats (t = 1.998, df = 64, p = 0.002).

Of the two pregnant females, both had an ideal BCS, and a large amount of fat was observed visually in the abdomen during necropsy. Two females were noted as currently lactating and one had recently finished lactating. Of these, the cat that had finished lactating had an emaciated BCS and the two lactating cats had ideal

BCSs. Of the cats with parasites, 70% were given an ideal or overweight BCS. Two of the neutered cats had poor BCSs.

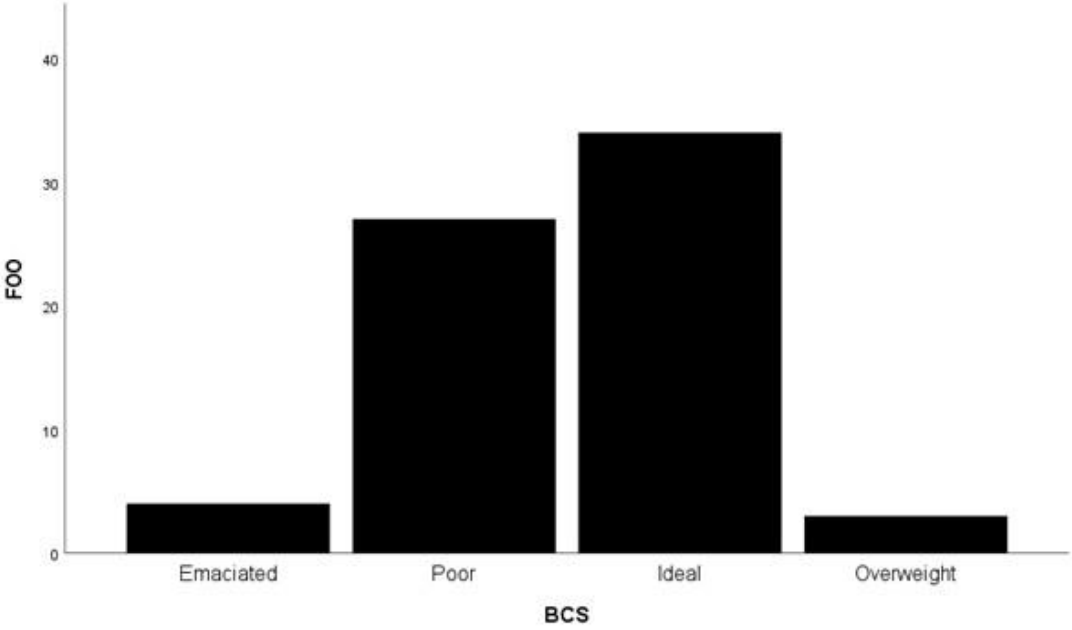

Figure 4. Total FOO of BCSs of all SDR cats necropsied.

BCSs and residuals obtained from regression analyses for nose-tail length versus weight in adult males were correlated with BCS (r = 0.552, p = 0.014) but not for adult females or either sex in juveniles (Table 8). Residual values for forearm length were not correlated with BCSs for any age or sex. Femur length residual values were correlated with BCS (r = 0.499, p = 0.049) for juvenile females only. Head width residuals were correlated with BCS for adult females (r = 0.509, p = 0.026) but not for males or either sex in juveniles.

Table 8. Spearman correlation between morphometric measurement residuals and BCS.

Group	Morphometric measurement	r	p-value
Adult male	Nose-tail	0.552	0.014
	Forearm	0.247	0.307
	Femur	0.367	0.134
	Head width	0.238	0.326
Adult female	Nose-tail	0.405	0.095
	Forearm	0.274	0.271
	Femur	0.352	0.140
	Head width	0.509	0.026
Juvenile male	Nose-tail	0.479	0.083
	Forearm	0.377	0.184
	Femur	0.479	0.083
	Head width	0.479	0.083
Juvenile female	Nose-tail	0.481	0.059
	Forearm	0.489	0.054
	Femur	0.499	0.049
	Head width	0.462	0.072

4.3.3 KFI

Two juveniles had no kidney fat on either kidneys. Both kidneys of one adult male were unable to be processed due to rupturing upon dissection from the body cavity. For all other cats, KFI ranged from 2.80–91.82 (Table 9). The cat with the highest KFI was an adult, pregnant female, weighing 3,900 g.

There was no significant difference between groups for KFI. Likewise, KFI residuals did not differ significantly between groups (Table 7). KFI was not significantly correlated with weight for any age group or sex (Table 10). Correlations between morphometric residuals and KFI residuals varied with adult females having the most significant correlations (Table 11).

Table 9. Descriptive statistics for KFI for each age group of cats.

Group	n	Range	Minimum	Maximum	Mean	Std. error	Std. deviation
Adult male	18	38.244	9.237	47.482	23.419	2.327	9.871
Adult female[1]	19	86.20	5.615	91.816	32.641	5.050	21.249
Juvenile male	14	42.651	0	42.651	19.081	3.552	13.289
Juvenile female	16	42.514	0	42.514	18.416	2.958	11.834

1. Includes single geriatric female (KFI% = 45.27).

Table 10. Pearson correlation between weight and KFI for each sex and age group of cats.

Group	r	p-value
Adult male	0.007	0.977
Adult female	0.331	0.166
Juvenile male	0.475	0.086
Juvenile female	0.474	0.063

Table 11. Spearman correlation between morphometric measurement residuals and KFI residuals of unowned cats.

Group	Morphometric measurement	r	p-value
Adult male	Nose-tail	0.777	<0.001
	Forearm	0.666	0.003
	Femur	0.802	<0.001
	Head width	0.395	0.105
Adult female	Nose-tail	0.535	0.022
	Forearm	0.973	<0.001
	Femur	0.860	<0.001
	Head width	0.723	<0.001
Juvenile male	Nose-tail	0.488	0.077
	Forearm	0.414	0.141
	Femur	0.480	0.083
	Head width	0.480	0.083
Juvenile female	Nose-tail	0.400	0.125
	Forearm	0.483	0.058
	Femur	0.500	0.049
	Head width	0.457	0.075

Table 12. Spearman correlation for KFI residuals and BCS of cats.

Group	r	p-value
Adult males	0.233	0.353
Adult females	0.140	0.568
Juvenile males	0.385	0.174
Juvenile females	0.401	0.124

4.4 BC-diet relationship

Nose-tail length and head width residuals had consistently strong correlations for all groups however, the tail is subject to injury which can affect nose-tail length measurements. Therefore, head-width residuals were used to determine the relationship with diet results. Chi-squared test results revealed no significant difference in food type between head width residuals (X^2 = 8.025, df = 10, p = 0.626). Likewise, no significant difference in the consumption of small and large mammals was found between head width residuals (X^2 = 1.318, df = 2, p = 0.517), however, it was noted that in most cases when small mammals were consumed, large mammals were not and vice versa.

Irrespective of BCS, commercial cat food was the most commonly consumed food item across all cats (Figures 5–8).

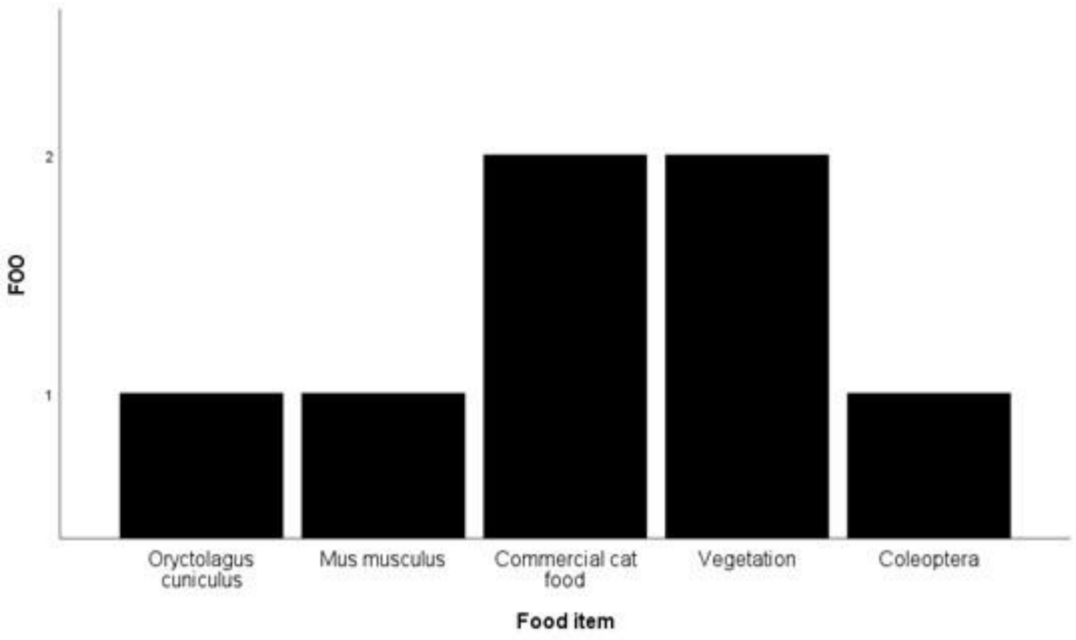

Figure 5. FOO of food items consumed by all cats with a BCS of 1. Food items with zero FOO not shown.

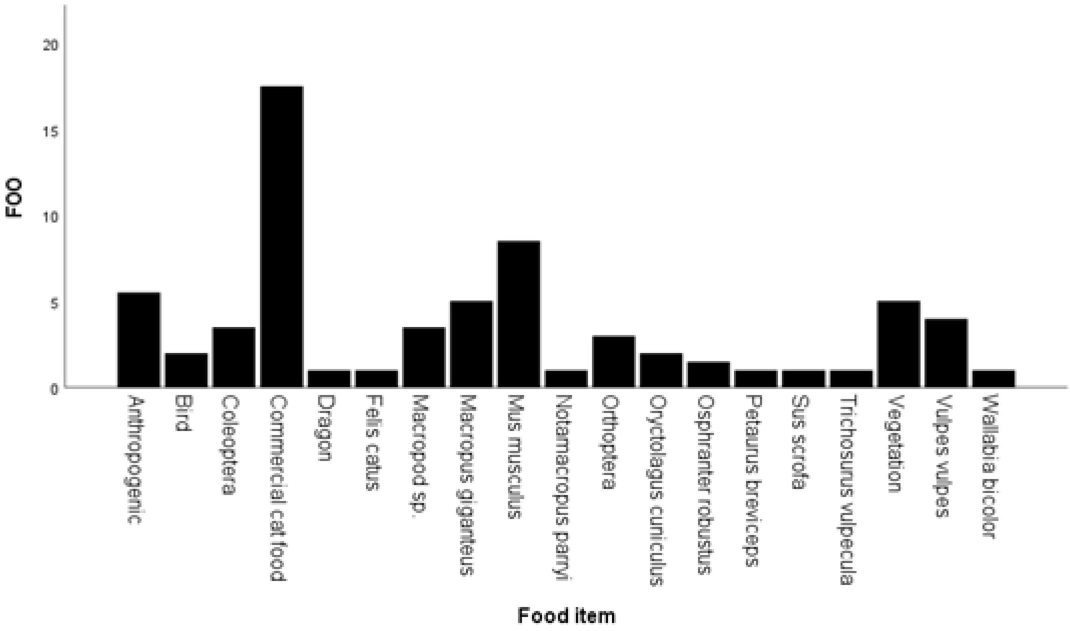

Figure 6. Frequency of food items consumed by all cats with a BCS of 2. Food items with zero FOO not shown.

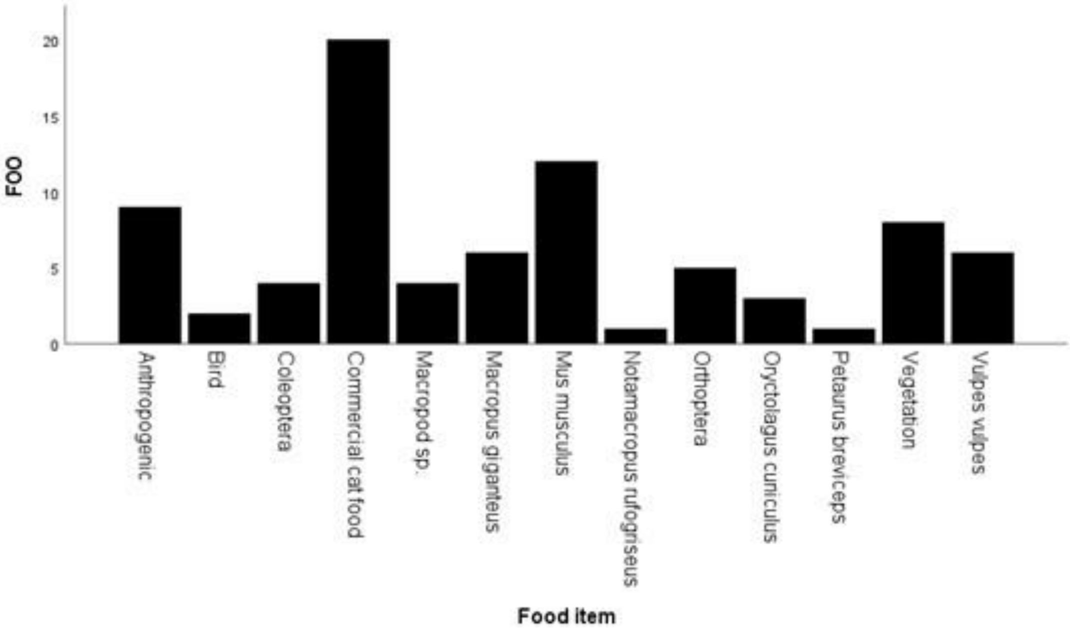

Figure 7. Frequency of food items consumed by all cats with a BCS of 3. Food items with zero FOO not shown.

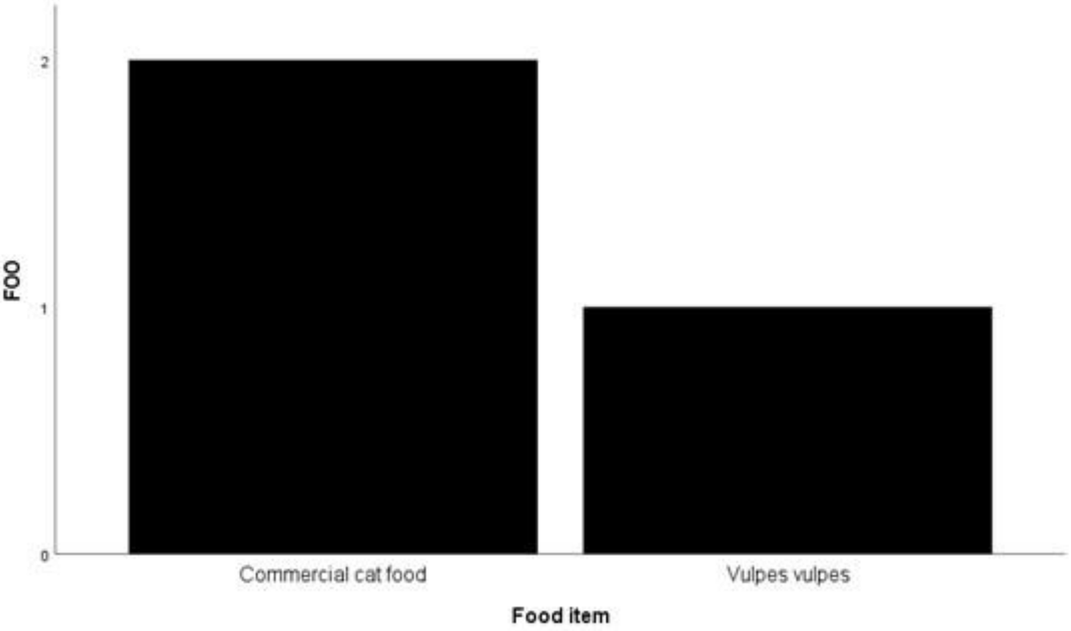

Figure 8. Food items consumed by all cats with BCS of 4. Food items with zero FOO not shown.

An ANOVA revealed no significant difference in the number of prey items consumed between BCSs (F = 2.545, df = 3, p = 0.085). Chi-squared test revealed no significant difference between BCS and food type (X^2 = 6.551, df = 15, p = 0.969).

There was a significant difference in KFI between cats that consumed mammals and those that did not (t = -2.463, df = 65, p = 0.016). No significant difference was found in KFI between cats that consumed cat food and those that did not (t = 0.547, df = 65, p = 0.587).

Irrespective of age or sex category, small mammals and commercial cat food were the most commonly occurring in diets and the mean BC for cats across all BCIs was ideal.

4.5 DDS dataset

Only data for adult cats were reported in the dataset. A total of 59 cats were randomly sampled from 189 cats shot during incidental cat monitoring in 1992. Coat colours were described as tabby (69.49%), ginger (22.03%), and black (8.47%).

Of the cats assessed, 15 were female, 41 were male, and three were not sexed. The mean weight for all cats was 4.5 kg, ranging between 2.0 and 6.1 kg. Male mean weight was 4.7 kg, ranging between 3.2 and 6.0 kg. Female mean weight was 3.7 kg, ranging between 2.5 and 5.0 kg.

Diet composition

No diet data was recorded for 42 cats of the 59 cats. It is unclear if these samples had no stomach contents or if they simply were not assessed. Of the 17 with diet data, the only prey items recorded were long-haired rat, *Rattus villosissimus*, bilby, *Macrotis lagotis*, lizard, and invertebrate (Figure 9). Long-haired rat had the highest FOO% (22.033%), followed by invertebrates (8.475%), bilbies (3.390%), and lizards (1.695%). IRI was calculated for the DDS dataset and is outlined in Table 13.

The Brillouin's Index value for the DDS dataset was 0.826. When compared with Warwick/Allora cats, the Pianka's Index similarity was 0.111. When compared to Stanthorpe, the value was 0.103.

BC and BC-diet relationship

All cats were reported to be in excellent condition. As all cats were reported to be in excellent condition, no further analyses between BC and diet could be made for cats from DDS.

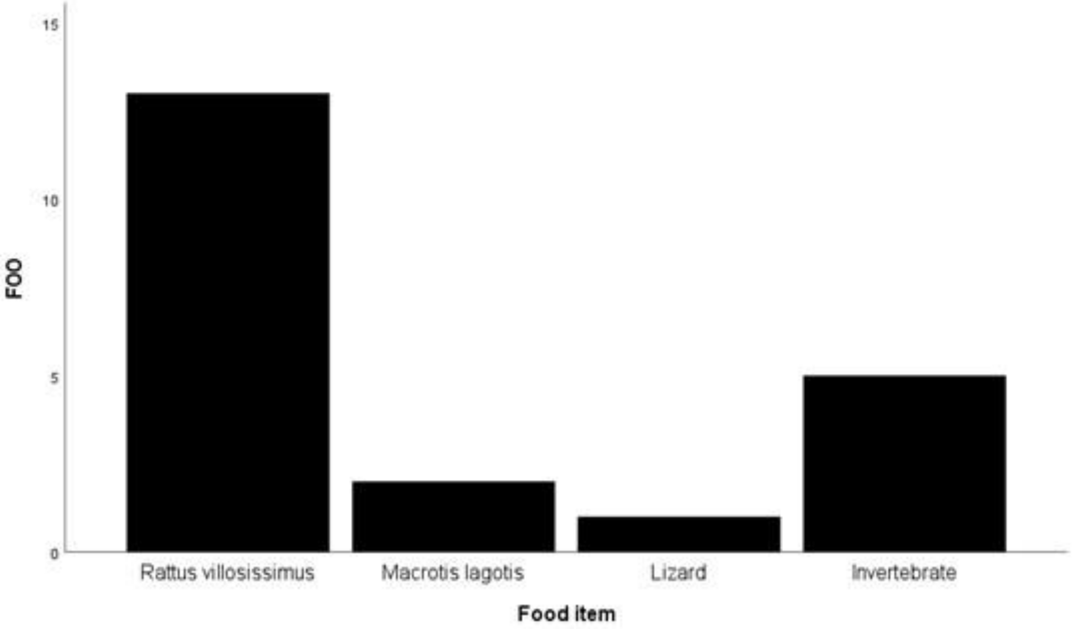

Figure 9. FOO of each prey item found in the diet of DDS cats in 1992.

Table 13. IRI for prey consumed by unowned cats at DDS.

Prey	FOO Stomach	Prey % (N)	Weight (kg)	Biomass	Biomass % (V)	Frequency % (F)	IRI
Rattus villosissimus	13	61.905	0.200	2.600	46.305	22.034	2384.273
Invertebrate	5	23.810	0.002	0.010	0.178	8.475	203.285
Macrotis lagotis	2	9.524	1.315	2.630	46.839	3.390	191.060
Lizard	1	4.762	0.375	0.375	6.679	1.695	19.391

Chapter 5: Discussion

The main aim of this research was to determine the diet composition, BC and their relationship in unowned cats from the SDR and DDS (Section 2.8). Overall, commercial cat food and mammals dominated the diet of unowned cats in the SDR (Tables 1–5). Mammals also dominated the diet of unowned cats at DDS (Table 13).

Morphometric measurements were not as strongly correlated with weight as predicted (Table 6). Additionally, KFI results did not correlate with BCSs as hypothesised (Table 12). The results from both datasets found majority of cats were in ideal BC with no significant differences in diet composition, suggesting that diet and BC are not strongly correlated.

5.1 SDR dataset

5.1.1 Diet composition

In Chapter 2, it was hypothesised that small mammals would make up approximately 70% FOO in unowned cat diet (Murphy et al. 2019; Crawford et al. 2020). In contrast to this, cats from the present study had a small mammal FOO below 60% (Table 2). Instead, unowned cats consumed large amounts of commercial cat food and carrion. This result is similar to other research where non-prey food items were consumed more often during cooler months (Cove et al. 2017; Crawford et al. 2020). When rabbit abundance is low, predation pressure on other animals increases (Catling 1988; Read & Bowen 2001; Doherty et al. 2015). Considering the low FOO of rabbits in the diet of unowned cats in this study, it may be that their abundance in the region was low. As such, cats utilised other food sources including carrion and commercial cat food.

It is also possible cats were lured into traps using commercial cat food as bait. This may have contributed to some of the cat food found in cat stomach and faecal samples. This is consistent with a previous study recording bait being the only food item in the stomach contents of trapped cats (Read & Bowen 2001). However, the amount of cat food in some cat’s GITs suggests they had been fed or readily had access to such food.

Prior to the study beginning, it was predicted that majority of the cats would come from agricultural and peri-urban areas based on the lack of urban areas within the region, and the land use in the region (Southern Downs Regional Council 2020b). Although their capture sites were unknown, it is likely many of the cats were from suburban or peri-urban environments, based on the amount of commercial cat food in their diets (Cove et al. 2017; Piontek et al. 2020). Unowned cats in agricultural areas are also known for having commercial cat food in their diet (Denny 2005), suggesting cats from this study may have come from such areas.

Another explanation for the amount of commercial cat food found in the GITs of unowned cats is due to humans feeding them. The SDRC are aware of several people who regularly feed unowned cats in the region (Lambert, R 2021, pers. comm., 15 June); which would explain why there was a high FOO of commercial cat food in cat GITs for this study. Deliberate feeding of unowned cats by members of the public is not uncommon (Lohr & Lepczyk 2014; Gunther et al. 2016; Swarbrick & Rand 2018), hence this is a plausible explanation.

Another potential explanation for the high amount of cat food in unowned cat diet is SDRC cat management practices. Trapped cats deemed feral are transported to vets as soon as practical. However, if a cat is acquired during the weekend, they are fed commercial cat food and housed until they can be taken to the vets for euthanasia (Lambert, R 2021, pers. comm., 15 June). Consequently, some of the cats from this study may have been fed by council staff and as such commercial cat food was recorded in their diet.

Finally, it is possible that cats were owned, and not unowned. When a cat is stressed, it can display behaviours that an observer would classify as feral or unowned cat behaviour (Dybdall et al. 2007; Gosling et al. 2013; Buckley 2019). Cats are not required to be desexed or registered under SDRC bylaws but owners are responsible for their pets not causing a nuisance (Southern Downs Regional Council 2020c). Consequently, it is possible that irresponsible cat owners allowed their cats to wander, and these cats inevitably ended up impounded and euthanased. This might also account for some of the cat food found in GITs during this study.

March and April – when this study was undertaken – are also the height of the breeding season for house mice in Australia (CSIRO 2021); and during the study period, a mouse plague was occurring in the region (Harden 2021; Paynter 2021). This may account for the high FOO of house mice seen in cat diets. Rodent consumption can also be higher in agricultural, suburban, and peri-urban areas, where such prey items can be common (Jones & Coman 1981; Campos et al. 2007; Piontek et al. 2020). Accordingly, the high FOO of small mammals in the diet of the cats from this study are unsurprising.

A total of seven native mammals were consumed by unowned cats (Table 1). Only two of these species were within the CWR (Tables 3–5). Additionally, these two mammals did not frequently occur in the diet of unowned cats (Table 1). Unowned cats in natural environments consume more native animals than those inhabiting modified environments (Jones & Coman 1981; Denny 2005). The low FOO and diversity of native animals in the diet of unowned cats in this study further suggests these cats were trapped in modified environments. Previous studies have indicated native mammals that fall within the CWR are at risk of population declines due to introduced predators (Burbidge & McKenzie 1989; Johnson & Isaac 2009; Murphy & Davies 2014; Doherty et al. 2015). The results from the present study tentatively suggest unowned cats inhabiting modified environments within the SDR are not causing significant impacts to extant CWR native mammals.

Due to biomass being part of the calculation for IRI, it is likely large prey items such as macropod species have had their IRIs affected due to their size. Macropod species, particularly the eastern grey kangaroo, have an average weight in excess of 15 kg (Queensland Museum 2020). Given these animals do not fall within the CWR (Burbidge & McKenzie 1989; Johnson & Isaac 2009), it is likely cats that consumed macropod species were scavenging on their carcasses. This is not uncommon with cats previously being shown to consume macropods as carrion (Paltridge et al. 1997; Molsher et al. 1999; Spencer et al. 2021). No maggots were found in any stomach or faecal samples for this study, to indicate scavenging (Catling 1988; Fleming et al. 2020), however caching (saving or storing of predated food) can also result in maggot presence (Gadbois et al. 2015; Ruiz-Villar et al. 2020). Cats are not known to cache prey (Short et al. 2002) therefore, it is more likely that large mammals were consumed as carrion. The results from the present study contradicts previous research which found only 10% of unowned cats that encountered carrion consumed it (Forsyth et al. 2014). During times of drought or low resources, carrion becomes a more important food source in unowned cat diet (Jones 1977; Catling 1988; Spencer et al. 2021). As the SDR is currently experiencing a drought (The Long Paddock 2021), it is plausible this is why carrion FOO in unowned cat diet was high in the region. An alternative hypothesis is that cats preyed on smaller macropods, such as joeys that were emerging from or that had permanently emerged from the pouch. This is not unheard of with unowned cats previously preying on rufous-hare wallabies and black-footed rock wallabies, macropods that fall within the CWR (Gibson et al. 1994; Read et al. 2019).

There was a distinct difference in the types of mammals consumed by unowned cats (Table 2). When small mammals were present in the diet of an individual cat, in most cases, large mammals were not. This suggests that cats in the region display two distinct feeding behaviours. One is that cats appear to prey on small mammals, and another is that cats appear to scavenge on carrion. It is possible the cats that consumed large mammals, were in areas of low small mammal abundance. Alternatively, cats may be displaying prey selectivity (Kutt 2012; Spencer et al. 2017), or in this case, food selectivity. Cats may have selected small prey despite carrion being readily available. Conversely, it is uncommon for cats to select carrion over live prey (Dickman 1996; Molsher et al. 2005; Forsyth et al. 2014; Read et al. 2019) further suggesting small mammals were limited in the areas where these cats were captured.

In the present study, SDR unowned cats had mid-low dietary diversity. In contrast, DDS unowned cats had considerably lower dietary diversity than SDR cats. These findings support previous research in which unowned cats living in temperate climate zones were found to have higher dietary diversity than those living in arid climate zones (Doherty et al. 2015; Murphy et al. 2019). This is due to temperate climate zones having suitable conditions for more mammalian species diversity than arid climate zones (Kutt 2012; Doherty et al. 2015). Arid climate zones experience higher temperatures and lower rainfall and as such, biodiversity decreases (Doherty et al. 2015; Murphy et al. 2019; Parsons et al. 2019).

The low number of native mammals present in the diet of the SDR cats may also be attributed to cover availability (i.e. vegetation, tree hollows, etc). The literature suggests, animals inhabiting open habitats are more at risk of predation than those inhabiting areas with adequate cover (Johnson & Isaac 2009; Leahy et al. 2015; Davies et al. 2017; Loggins et al. 2019). Being in a temperate climate zone, the SDR has more vegetation available for cover than the arid climate zone in which DDS was located. Due to this, more small native mammals were consumed at DDS than in the SDR.

5.1.2 BC

5.1.2.1 Morphometric residuals

Morphometric measurements versus weight regression analyses have previously been demonstrated as accurate measures of BC when compared to a cat’s expected weight (Denny 2005). In the present study, not all morphometric measurements had a strong relationship with weight across all groups.

All morphometric measurement residuals showed a correlation with weight in adult males. However, the strong correlation described by Denny (2005) between femur length and weight was not seen in this study. A moderate correlation was instead shown (Table 6). For the present study, head width showed the strongest correlation with weight in adult males (Table 6). The sample size for adult males (n = 19) in this study was considerably smaller than that of Denny (2005) (n = 65), which may account for the discrepancy in correlation between studies. Additionally, Denny (2005) included juveniles in their regression analyses while this study separated them.

In contrast to previous findings (Denny 2005), femur length was not strongly correlated with weight for adult females. Surprisingly, weight versus forearm length measurements did not have a strong relationship (Table 6). A previous study on various felid species found that those inhabiting densely vegetated natural environments had shorter and wider forearm bones than those inhabiting sparsely vegetated natural environments (Schellhorn & Sanmugaraja 2014). Though an interspecific example, the present study was undertaken in a temperate climate zone, and vegetated area. Hence, it is plausible to assume, unowned domestic cats are displaying the same results as other felid species and as such, forearm length is not strongly correlated with weight. However, as the locations of the cats used for the present study are unknown, no definitive conclusion can be made.

Nose-tail length versus weight was the strongest correlation in adult females (Table 6). The second strongest correlation in this study for adult females head width versus weight. A positive correlation has previously been seen between head width and weight in female adult cats (Denny 2005). The results from this study support those of Denny (2005) however, are clearly in contrast with regard to femur length versus weight correlations. Though head length residuals are a stronger BCI for female cats (Denny 2005), femur length is a more accessible method to use in the field for unowned cats (Denny 2005). As with adult males, the adult female sample size (n = 19) was smaller than that of Denny (2005) (n = 54) and juveniles were assessed separately for this study. It is likely the smaller sample size in this study attributed to the weaker correlations seen between morphometric measurements and weight in adult females.

In juvenile males, all morphometric measurements correlated with weight, but the strongest correlation was between forearm length and weight (Table 6). The juvenile male correlation results are similar to those observed by Denny (2005) for all males.

In juvenile females, head width had the strongest relationship with weight (Table 6). In fact, across all groups, head width had the strongest relationship with weight. Previous research found that bite force was a predictor of prey size in unowned cats (Fleming et al. 2020). This relates to the present study as bite force is related to head width (Sicuro & Oliveira 2011; Yip et al. 2014; Fleming et al. 2020). Fleming et al. (2020) state that age is the strongest predictor of prey size in unowned cats. In their study and the present study, head width, age, and cat body weight were interrelated. This may explain why head width was correlated with weight across all groups. With head width being the only measurement to be consistently correlated with weight, this is the best measure to use as a BCI. It is unclear why juvenile morphometric measurements had stronger relationships with weight than adult measurements though, it is likely the sample sizes influenced the results.

The cat that had recently finished lactating was in an emaciated condition. This supports the findings of Wichert et al. (2012) who found majority of queens lost considerable body fat at the end of the lactation period. The energy expenditure required for lactation increases as the kitten grows (Oftedal & Gittleman 1989); therefore, it is likely this cat was unable to match its nutritional needs to replenish its energy stores post lactating.

5.1.2.2 BCS

In line with the hypothesis that the majority of unowned cats in the SDR would have an ideal BCS, the results showed majority (54.4%) were either in ideal or above condition. This finding supports previous research on the BC of unowned cats in Australia with between 52–100% of unowned cats being in ideal or above condition (Domm & Messersmith 1990; Yip et al. 2014; Crawford et al. 2020). It is likely the number of mammals and commercial cat food consumed, contributed to the cats in the present study mostly being in ideal BC. High mammal FOO in diet in previous studies has resulted in high BC results (Yip et al. 2014). Alternatively, the high amount of anthropogenic items may account for the high BC as seen in a previous study (Crawford et al. 2020).

Juveniles had lower BCSs than adults for this study. Similar results have been found in Israel with juvenile cats being reported as being in poor condition more often than adult cats (Gunther et al. 2018). It may be that less adults are in poor BC because those cats that were have died. But for juveniles, more are still alive and contribute a higher frequency being in poor BC. This theory is supported by Gunther et al. (2018) who found disease prevalence and injury to be higher in juvenile cats than adult cats. The authors also suggest the kitten's higher energy expenditure and capability to compete with adults for resources may have contributed to their poor BC. This argument is further supported by Brothers et al. (1985) who found majority of unowned kittens do not survive to adulthood.

5.1.2.3 KFI

KFI did not have a significant correlation with weight for any age or sex. In Chapter 2, it was hypothesised that KFI would be an accurate measure of BC in cats. This result suggests KFI is not an accurate BCI. Additionally, contrary to the hypothesis that KFI would correlate with other BCIs, KFI results were not correlated with BCSs for all cat groups. Only adult and juvenile male BCSs correlated with KFIs. Likewise, the only correlation found between KFI and morphometric residuals was for all morphometric measurements in juvenile females.

Other studies have shown KFI does not account for differences in the amount of kidney fat or the size of the kidney in different species (Serrano et al. 2008). KFI is more often used in ungulates which have larger kidneys and large amounts of fat surrounding them (Serrano et al. 2008). In comparison, the kidneys from the cats used in this study, weighed < 50 g.

Standardised residual values were used to compare KFI and other BCIs in this study. This method may not have been the best method to compare KFI. It may have been more appropriate to transform all BCIs, including KFI values to logarithms (Labocha et al. 2014) then compare.

Although the KFI values did not correlate with other BCIs, the results were in accordance with findings previously reported by Short et al. (1997) where the mean

KFI for unowned cats were between 26 and 40 across several years. It is equally important to highlight that the KFI results were similar to body fat results observed in a sample of 89 research colony and pet cats, regularly fed commercial cat food (Launten et al. 2000). Unlike the cats in the study by Launten et al. (2000), SDR cats were unowned therefore, had limited access to commercial cat food. Despite this, commercial cat food was frequently observed in the diet of the unowned cats, which could be why their KFI is similar to the body fat results of the owned cats.

Previous research has shown that BCSs and morphometric measurement residuals are poor indicators of BC and can be subject to biases (Bakker & Main 1980; Krebs & Singleton 1993; Burkholder 2000; Michel et al. 2011; Labocha et al. 2014; Gerstner & Liesegang 2017). It may be the case that KFI was in fact the superior BCI however, with no total body fat results, it remains unclear to which degree KFI and BC are related in cats.

Given the conflicting results from measuring KFI in this study, further investigation into KFI as a BCI for cats should be conducted. Future studies could incorporate kidney fat thickness measurements as has been used been used in other carnivores (Behrendorff et al. 2016).

5.1.3 BC-diet relationship

In contrast to the hypothesis presented in Chapter 2, there was no significant relationship between the consumption of small mammals and ideal BC.

Dry commercial cat food has a lower moisture content than live prey (Plantinga et al. 2011; Alexander et al. 2014). Furthermore, high moisture content in diet results in lower food intake (Alexander et al. 2014). Therefore, it is likely cats that consumed commercial cat food had higher BC than cats with live prey only due to the lower moisture content of the dry food. In contrast, previous research found no significant difference in BC between pet cats known to hunt and those that did not hunt (Russell et al. 2000).

The cats used in this study were captured over a period of five months. Previous studies found variations in weight and BC between cats captured during different seasons (Brothers et al. 1985; Hayde 1992). It is possible that cats from the present study experienced weight declines as the study progressed into the winter months. However, this was not assessed as the dates of capture/euthanasia were not recorded for any cats.

Irrespective of morphometric residual or KFI value, the diet was consistent across all BCI values. This indicates that despite staple prey such as mice and rabbits not making up the majority of their diet, unowned cats are able to thrive on other available resources.

A final hypothesis is that cats were able to maintain their BC regardless of resource availability. Cats are able to regulate their food intake and maintain their BC when ample food is available (Bradshaw et al. 1996). If unowned cats in this study displayed this behaviour, those that had access to abundant resources such as carrion or commercial cat food would not have been overweight or obese.

5.2 DDS

Reptile consumption was low considering the study location was in an arid climate zone, with previous research reporting that cats in arid and semi-arid climate zones consume a higher number of reptiles (Doherty et al. 2015; Woinarski et al. 2018a).

The low FOO of reptiles in the diet of DDS cats can be explained by the study being conducted during winter and early spring months (Woinarski et al. 2018b), in addition to the sample size being small.

Furthermore, the study was conducted during a long-haired rat plague (Allen et al. 2014), hence a high FOO for this species was to be expected. Additionally, high FOO of native rodents in the diet of unowned cats inhabiting arid environments has been recorded by several authors (Paltridge et al. 1997; Hart et al. 2002; Yip et al. 2014; Murphy et al. 2019) suggesting these mammals form a significant part of these cats. It is possible some of the 42 cats that did not have their diet recorded, consumed reptiles, thereby giving a more accurate representation of the reptiles consumed at DDS.

Previous authors have suggested unowned cats would not persist in semi-arid climate zones if rabbit abundance is not high (Catling 1988). However, the data from this study appear to contradict this claim. Likewise, the results from the SDR dataset indicate cats will adapt and consume carrion when resource availability is low.

DDS cats had no anthropogenic items in their diet (Figure 9) in comparison to SDR cats (Table 2). Although the locations from which the SDR cats were caught are unknown, it is presumed majority were located within the peri-urban areas of the region. In comparison, DDS cats were in a remote rural area that was largely unmodified and otherwise natural (Allen et al. 2014). These results agree with previous literature that shows unowned cats consume less anthropogenic items than those inhabiting modified or semi-modified environments (Denny 2005; Crawford et al. 2020).

5.3 Limitations and recommendations

The sample size for SDR cats was considered adequate given previous studies had similar or smaller sample sizes (Jones 1977; Martin et al. 1996; Yip et al. 2014; Read et al. 2019). However, in the present study, 45% of cats were juveniles. In some cases, juveniles are still partially or fully dependent on their mothers for food (Devillard et al. 2003). Additionally, it was clear from the veterinary admission information that some juveniles sampled were from the same litter. As a result, these cats often had the same GIT contents and would likely have the same GIT contents as their mother. Hence, future studies should aim to include a larger sample of adult cats and obtain a clearer picture on the origin of and relationship between sampled cats.

The present study assessed a snapshot of each individual cat’s diet. Gastrointestinal transit time for cats is between 26.5 and 35.7 hours (Peachey et al. 2000). The results from the present study showed mammals were an important food item in unowned cat diet. If a cat had consumed prey prior to this time, it would not have been recorded for this study. It is therefore plausible to assume more cats had been consuming live prey than recorded.

This study did not assess prey abundance within the SDR. Prey abundance measures were also not available for the DDS dataset, although the cats were shot during a rat plague, and so would be expected to contain a high FOO of rats. Establishing prey abundance would assist researchers to determine if prey selectivity is occurring (Dickman & Newsome 2015). Additionally, prey abundance can inform managers on whether cat management needs to take place. Likewise, this study did not assess the age of the prey consumed by unowned cats in the SDR or at DDS. Determining whether adult or juvenile prey are being consumed can also inform on whether cats in these areas are displaying prey selectivity and can also help determine the impact of cats on the demography of prey populations. Therefore, future studies should aim to incorporate prey abundance and determine prey age.

Conducting BCS assessments on deceased cats is different to conducting them on live cats. The cats in this study were frozen for varying amounts of time. Upon defrosting, many cats lost fluid. It is possible this impacted the BC scoring of the cats in this study.

Locations of where cats were captured was not available for the SDR cats. Understanding where these cats are coming from and their movement ecology can inform managers on areas of concern in addition to determining likely diet. This is particularly important considering the amount of commercial cat food found in the diet of cats in this study. The amount of commercial cat food observed in the diet of unowned cats during this study indicates humans are having a direct impact on unowned cat survival. For council to decrease the unowned cat population, public education programs on how human activity impacts cats should be implemented. Educating the public on cat management can limit the resources of unowned cats, leading to their decline. Additionally, the removal of carrion along roadsides may reduce this food source at specific sites if this indeed is where the cats are consuming them.

Chapter 6: Conclusions

The purpose of this research was to determine whether diet has an influence on BC in unowned cats in two locations, SDR and DDS. Unowned cat diet was assessed using FOO, VOL%, and IRI. BC was assessed using various indices and these indices were compared to determine their validity in measuring BC. BCS is a frequently used BCI and was used as a baseline to assess other BCIs against. Regression analyses between morphometric measurements and weight and KFI were also used.

The results from the SDR dataset found the FOO of commercial cat food in unowned cat diet to be higher than expected. It was hypothesised that small mammals would make up the majority of SDR unowned cat diet. Contrary to this, commercial cat food and small mammals had similar FOO and VOL% in diet. Additionally, large mammals had a higher FOO in diet than expected.

It is likely factors such as deliberate feeding, irresponsible pet ownership, carrion availability, and unowned cat management practices have influenced the results of this study. Additionally, due to sample size, it is unclear if the diet listed is comprehensive.

Contrary to the hypothesis that KFI would correlate with other BCIs, it did not correlate with BCS, nor was there a strong relationship between KFI and body weight. KFI did however, correlate with some morphometric residuals.

The distinct differences between KFI and other BCS suggests one of these BCIs is inaccurate. Using frozen specimens to complete BCSs may have impacted the results in addition to the sample size being small. Likewise, the residual method used for KFI may not have been an appropriate transformed measurement.

Diet was not a strong predictor of BC in unowned cats for either the SDR or DDS datasets. The exact reason for this is unclear however, this thesis offers low sample size and food intake regulation as potential explanations. Future studies should aim to use a larger sample, particularly of adults, than that used in this study. To determine whether KFI is a valid method of measuring BC in cats, a more robust method including measuring the total body fat of the cat should be conducted to compare KFI results to.

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Appendices

Appendix A: Non-domestic cat assessment form used by SDRC

Non Domestic Cat Assessment

Biosecurity Risk Assessment

1. Check for a microchip
    - Yes – Surrender to AWL
    - No – Continue Assessment

2. Is the Cat desexed?
    - Yes – Surrender to AWL
    - No – Continue Assessment

3. Does the cat have a collar?
    - Yes – Complete assessment 4
    - No – Cat displays no signs of ownership. Complete assessment 5

4. Does the cat collar have ownership details?

- Yes – Surrender to AWL
- No - Undertake domestic trait test

5. Does the Authorised Officer have a reason to believe the cat is Domestic?
    - No – Cat is considered non-domestic and therefore must be considered a restricted invasive animal under the Biosecurity Act 2014
    - Yes – Surrender to AWL (Officer to provide details of Decision below)

Domestic Trait Test

Complete Assessments 6 & 7

6. Visual Assessment

Overall health – Body Condition Scoring (BCS) using the Royal Canin Body Condition Score for Cats & observing for signs of illness	
Assessment	Score
ZERO POINTS • Unhealthy – BCS ≤3 out of 9; dirty/unkempt coat; one or more signs of moderate to severe illness e.g. cat flu, visible diarrhoea, unhealed wounds	
ONE POINT • Reasonably Healthy – BCS 3-4 out of 9; dirty/unkempt coat; no obvious signs of illness	
TWO POINTS • Healthy – BCS ≥4 out of 9; healthy coat; no overt signs of illness	

7. Behavioural Assessment

Body Posture	
Assessment	Score
ZERO POINTS • Climbing the walls of the trap/cage **AND/OR** • Cat throws itself around **AND/OR** • Distressed **AND/OR** • Ears are flat back **AND/OR** • Pupils dilated **AND/OR** • Head/neck pulled tight **AND/OR** • Facial muscles tense **AND/OR** • Body is flattened **AND/OR** • Teeth bared **AND/OR** • May roll over and show claws **AND/OR** • Vocalizes loudly & aggressively **AND/OR**	
ONE POINT • Frightened or tense stance **AND/OR** • Twitching tail	
TWO POINTS • Soft and relaxed posture	

When the Cage is Approached	
Assessment	Score
ZERO POINTS • Vocalising loudly and aggressively **AND/OR** • Tries to escape trap/cage **AND/OR** • Cat pins itself against the wall of the trap **AND/OR** • Cat crouches body stiff **AND/OR** • Swipes out, lunges or charges person approaching cage **AND/OR**	
ONE POINT • Does not approach **AND/OR** • Avoids eye contact but does not move away	
TWO POINTS • Comes to the front/side of trap/cage **AND/OR** • Solicits attention by rubbing/chirping/etc	

Response to touch with an object (do not “poke” the cat) – attempt to “pet” in a gentle manner	
Assessment	Score
ZERO POINTS • Swipes at the object and hisses **AND/OR** • Throws itself around the trap/cage **AND/OR** • Climbing walls to get away from the object **AND/OR** • Loudly and aggressively vocalizes **AND/OR** • Lunges	
ONE POINT • Does not seemed to be bothered by the object	
TWO POINTS • Indifferent or attempts to interact with the object positively	
TOTAL SCORE FOR ASSESSMENTS 6 & 7	

8. Domestic Trait Test Results
    - Assessment Score of 5 or more – Surrender to AWL

- Assessment Score of 4 or Less – Cats that receive a score of 4 or less out of 9 have failed the domestic trait test and must be considered a Restricted Invasive Animal under the *Biosecurity Act 2014*.

Appendix B: Vertebrate species list for SDR

Table 14. List of vertebrate species present within the SDR as of 2013 (Department of Environment and Science 2013b).

Class	**Family**	**Scientific Name**	**Common Name**
mammals	Acrobatidae	*Acrobates pygmaeus*	feathertail glider
mammals	Bovidae	*Bos* sp.	cattle
mammals	Bovidae	*Bos taurus*	European cattle
mammals	Bovidae	*Capra hircus*	goat
mammals	Burramyidae	*Cercartetus nanus*	eastern pygmy-possum
mammals	Canidae	*Canis familiaris*	dog
mammals	Canidae	*Canis familiaris (dingo)*	dingo
mammals	Canidae	*Canis* sp.	
mammals	Canidae	*Vulpes vulpes*	red fox
mammals	Cervidae	*Cervus elaphus*	red deer
mammals	Cervidae	*Dama dama*	fallow deer
mammals	Dasyuridae	*Antechinus flavipes flavipes*	yellow-footed antechinus (south-east Queensland)
mammals	Dasyuridae	*Antechinus flavipes sensu lato*	yellow-footed antechinus
mammals	Dasyuridae	*Antechinus stuartii*	brown antechinus
mammals	Dasyuridae	*Antechinus subtropicus*	subtropical antechinus
mammals	Dasyuridae	*Dasyurus hallucatus*	northern quoll
mammals	Dasyuridae	*Dasyurus maculatus maculatus*	spotted-tailed quoll (southern subspecies)
mammals	Dasyuridae	*Dasyurus* sp.	
mammals	Dasyuridae	*Phascogale tapoatafa tapoatafa*	brush-tailed phascogale
mammals	Dasyuridae	*Planigale maculata*	common planigale
mammals	Dasyuridae	*Planigale tenuirostris*	narrow-nosed planigale

mammals	Dasyuridae	*Sminthopsis murina*	common dunnart
mammals	Dasyuridae	*Sminthopsis murina murina*	common dunnart (SE mainland)
mammals	Emballonuridae	*Saccolaimus flaviventris*	yellow-bellied sheathtail bat
mammals	Equidae	*Equus caballus*	horse
mammals	Felidae	*Felis catus*	cat
mammals	Leporidae	*Lepus europaeus*	European brown hare
mammals	Leporidae	*Oryctolagus cuniculus*	rabbit
mammals	Macropodidae	*Macropus giganteus*	eastern grey kangaroo
mammals	Macropodidae	*Macropus* sp.	
mammals	Macropodidae	*Notamacropus agilis*	agile wallaby
mammals	Macropodidae	*Notamacropus dorsalis*	black-striped wallaby
mammals	Macropodidae	*Notamacropus parryi*	whiptail wallaby
mammals	Macropodidae	*Notamacropus rufogriseus*	red-necked wallaby
mammals	Macropodidae	*Osphranter robustus*	common wallaroo
mammals	Macropodidae	*Petrogale penicillata*	brush-tailed rock-wallaby
mammals	Macropodidae	*Petrogale* sp.	
mammals	Macropodidae	*Thylogale* sp.	
mammals	Macropodidae	*Thylogale stigmatica*	red-legged pademelon
mammals	Macropodidae	*Thylogale thetis*	red-necked pademelon
mammals	Macropodidae	*Wallabia bicolor*	swamp wallaby
mammals	Miniopteridae	*Miniopterus australis*	little bent-wing bat
mammals	Miniopteridae	*Miniopterus schreibersii oceanensis*	eastern bent-wing bat
mammals	Miniopteridae	*Miniopterus* sp.	
mammals	Molossidae	*Mormopterus norfolkensis*	east coast freetail bat
mammals	Molossidae	*Mormopterus ridei*	eastern free-tailed bat
mammals	Molossidae	*Mormopterus* sp.	
mammals	Molossidae	*Tadarida australis*	white-striped freetail bat

mammals	Muridae	*Conilurus albipes*	white-footed rabbit-rat
mammals	Muridae	*Hydromys chrysogaster*	water rat
mammals	Muridae	*Melomys cervinipes*	fawn-footed melomys
mammals	Muridae	*Melomys* sp.	
mammals	Muridae	*Mus musculus*	house mouse
mammals	Muridae	*Pseudomys desertor*	desert mouse
mammals	Muridae	*Pseudomys gracilicaudatus*	eastern chestnut mouse
mammals	Muridae	*Pseudomys novaehollandiae*	New Holland mouse
mammals	Muridae	*Pseudomys oralis*	Hastings River mouse
mammals	Muridae	*Rattus fuscipes*	bush rat
mammals	Muridae	*Rattus lutreolus*	swamp rat
mammals	Muridae	*Rattus rattus*	black rat
mammals	Muridae	*Rattus* sp.	
mammals	Muridae	*Rattus tunneyi*	pale field-rat
mammals	Ornithorhynchidae	*Ornithorhynchus anatinus*	platypus
mammals	Peramelidae	*Isoodon macrourus*	northern brown bandicoot
mammals	Peramelidae	*Perameles nasuta*	long-nosed bandicoot
mammals	Petauridae	*Petaurus australis australis*	yellow-bellied glider (southern subspecies)
mammals	Petauridae	*Petaurus breviceps sensu lato*	sugar glider
mammals	Petauridae	*Petaurus norfolcensis*	squirrel glider
mammals	Petauridae	*Petaurus* sp.	
mammals	Phalangeridae	*Trichosurus caninus*	short-eared possum
mammals	Phalangeridae	*Trichosurus* sp.	
mammals	Phalangeridae	*Trichosurus vulpecula*	common brushtail possum
mammals	Phascolarctidae	*Phascolarctos cinereus*	koala
mammals	Potoroidae	*Aepyprymnus rufescens*	rufous bettong
mammals	Potoroidae	*Potorous tridactylus tridactylus*	long-nosed potoroo

mammals	Pseudocheiridae	*Petauroides volans*	greater glider
mammals	Pseudocheiridae	*Petauroides volans volans*	southern greater glider
mammals	Pseudocheiridae	*Pseudocheirus peregrinus*	common ringtail possum
mammals	Pteropodidae	*Pteropus alecto*	black flying-fox
mammals	Pteropodidae	*Pteropus poliocephalus*	grey-headed flying-fox
mammals	Pteropodidae	*Pteropus scapulatus*	little red flying-fox
mammals	Pteropodidae	*Pteropus* sp.	
mammals	Rhinolophidae	*Rhinolophus megaphyllus*	eastern horseshoe-bat
mammals	Suidae	*Sus scrofa*	pig
mammals	Tachyglossidae	*Tachyglossus aculeatus*	short-beaked echidna
mammals	Vespertilionidae	*Chalinolobus dwyeri*	large-eared pied bat
mammals	Vespertilionidae	*Chalinolobus gouldii*	Gould's wattled bat
mammals	Vespertilionidae	*Chalinolobus morio*	chocolate wattled bat
mammals	Vespertilionidae	*Chalinolobus nigrogriseus*	hoary wattled bat
mammals	Vespertilionidae	*Chalinolobus picatus*	little pied bat
mammals	Vespertilionidae	*Falsistrellus tasmaniensis*	eastern false pipistrelle
mammals	Vespertilionidae	*Kerivoula papuensis*	golden-tipped bat
mammals	Vespertilionidae	*Myotis macropus*	large-footed myotis
mammals	Vespertilionidae	*Nyctophilus bifax*	northern long-eared bat
mammals	Vespertilionidae	*Nyctophilus corbeni*	eastern long-eared bat
mammals	Vespertilionidae	*Nyctophilus geoffroyi*	lesser long-eared bat
mammals	Vespertilionidae	*Nyctophilus gouldi*	Gould's long-eared bat
mammals	Vespertilionidae	*Nyctophilus* sp.	
mammals	Vespertilionidae	*Scoteanax rueppellii*	greater broad-nosed bat
mammals	Vespertilionidae	*Scotorepens balstoni*	inland broad-nosed bat
mammals	Vespertilionidae	*Scotorepens greyii*	little broad-nosed bat
mammals	Vespertilionidae	*Scotorepens orion*	south-eastern broad-nosed bat

mammals	Vespertilionidae	*Scotorepens* sp.	
mammals	Vespertilionidae	*Scotorepens* sp. (Parnaby)	central-eastern broad-nosed bat
mammals	Vespertilionidae	*Vespadelus darlingtoni*	large forest bat
mammals	Vespertilionidae	*Vespadelus pumilus*	eastern forest bat
mammals	Vespertilionidae	*Vespadelus regulus*	southern forest bat
mammals	Vespertilionidae	*Vespadelus* sp.	
mammals	Vespertilionidae	*Vespadelus troughtoni*	eastern cave bat
mammals	Vespertilionidae	*Vespadelus vulturnus*	little forest bat
mammals	Vombatidae	*Lasiorhinus krefftii*	northern hairy-nosed wombat
mammals	Vombatidae	*Vombatus ursinus*	common wombat
birds	Acanthizidae	*Acanthiza apicalis*	inland thornbill
birds	Acanthizidae	*Acanthiza chrysorrhoa*	yellow-rumped thornbill
birds	Acanthizidae	*Acanthiza lineata*	striated thornbill
birds	Acanthizidae	*Acanthiza nana*	yellow thornbill
birds	Acanthizidae	*Acanthiza pusilla*	brown thornbill
birds	Acanthizidae	*Acanthiza reguloides*	buff-rumped thornbill
birds	Acanthizidae	*Acanthiza* sp.	
birds	Acanthizidae	*Acanthiza uropygialis*	chestnut-rumped thornbill
birds	Acanthizidae	*Aphelocephala leucopsis*	southern whiteface
birds	Acanthizidae	*Gerygone fusca*	western gerygone
birds	Acanthizidae	*Gerygone mouki*	brown gerygone
birds	Acanthizidae	*Gerygone olivacea*	white-throated gerygone
birds	Acanthizidae	*Hylacola pyrrhopygia*	chestnut-rumped heathwren
birds	Acanthizidae	*Pyrrholaemus sagittatus*	speckled warbler
birds	Acanthizidae	*Sericornis citreogularis*	yellow-throated scrubwren
birds	Acanthizidae	*Sericornis frontalis*	white-browed scrubwren
birds	Acanthizidae	*Sericornis magnirostra*	large-billed scrubwren

birds	Acanthizidae	*Smicrornis brevirostris*	weebill
birds	Accipitridae	*Accipiter cirrocephalus*	collared sparrowhawk
birds	Accipitridae	*Accipiter fasciatus*	brown goshawk
birds	Accipitridae	*Accipiter novaehollandiae*	grey goshawk
birds	Accipitridae	*Accipiter* sp.	
birds	Accipitridae	*Aquila audax*	wedge-tailed eagle
birds	Accipitridae	*Aviceda subcristata*	Pacific baza
birds	Accipitridae	*Circus approximans*	swamp harrier
birds	Accipitridae	*Circus assimilis*	spotted harrier
birds	Accipitridae	*Elanus axillaris*	black-shouldered kite
birds	Accipitridae	*Elanus scriptus*	letter-winged kite
birds	Accipitridae	*Erythrotriorchis radiatus*	red goshawk
birds	Accipitridae	*Haliaeetus leucogaster*	white-bellied sea-eagle
birds	Accipitridae	*Haliastur sphenurus*	whistling kite
birds	Accipitridae	*Hamirostra melanosternon*	black-breasted buzzard
birds	Accipitridae	*Hieraaetus morphnoides*	little eagle
birds	Accipitridae	*Lophoictinia isura*	square-tailed kite
birds	Accipitridae	*Milvus migrans*	black kite
birds	Accipitridae	*Pandion cristatus*	eastern osprey
birds	Acrocephalidae	*Acrocephalus australis*	Australian reed-warbler
birds	Aegothelidae	*Aegotheles cristatus*	Australian owlet-nightjar
birds	Alaudidae	*Mirafra javanica*	Horsfield's bushlark
birds	Alcedinidae	*Ceyx azureus*	azure kingfisher
birds	Anatidae	*Anas castanea*	chestnut teal
birds	Anatidae	*Anas gracilis*	grey teal
birds	Anatidae	*Anas platyrhynchos*	northern mallard
birds	Anatidae	*Anas superciliosa*	Pacific black duck

birds	Anatidae	*Aythya australis*	hardhead
birds	Anatidae	*Biziura lobata*	musk duck
birds	Anatidae	*Chenonetta jubata*	Australian wood duck
birds	Anatidae	*Cygnus atratus*	black swan
birds	Anatidae	*Dendrocygna arcuata*	wandering whistling-duck
birds	Anatidae	*Dendrocygna eytoni*	plumed whistling-duck
birds	Anatidae	*Malacorhynchus membranaceus*	pink-eared duck
birds	Anatidae	*Spatula rhynchotis*	Australasian shoveler
birds	Anatidae	*Stictonetta naevosa*	freckled duck
birds	Anatidae	*Tadorna tadornoides*	Australian shelduck
birds	Anhingidae	*Anhinga novaehollandiae*	Australasian darter
birds	Apodidae	*Apus pacificus*	fork-tailed swift
birds	Apodidae	*Hirundapus caudacutus*	white-throated needletail
birds	Ardeidae	*Ardea alba modesta*	eastern great egret
birds	Ardeidae	*Ardea intermedia*	intermediate egret
birds	Ardeidae	*Ardea pacifica*	white-necked heron
birds	Ardeidae	*Botaurus poiciloptilus*	Australasian bittern
birds	Ardeidae	*Bubulcus ibis*	cattle egret
birds	Ardeidae	*Egretta garzetta*	little egret
birds	Ardeidae	*Egretta novaehollandiae*	white-faced heron
birds	Ardeidae	*Ixobrychus dubius*	Australian little bittern
birds	Ardeidae	*Ixobrychus flavicollis*	black bittern
birds	Ardeidae	*Nycticorax caledonicus*	nankeen night-heron
birds	Artamidae	*Artamus cinereus*	black-faced woodswallow
birds	Artamidae	*Artamus cyanopterus*	dusky woodswallow
birds	Artamidae	*Artamus leucorynchus*	white-breasted woodswallow
birds	Artamidae	*Artamus minor*	little woodswallow

birds	Artamidae	*Artamus personatus*	masked woodswallow
birds	Artamidae	*Artamus* sp.	
birds	Artamidae	*Artamus superciliosus*	white-browed woodswallow
birds	Artamidae	*Cracticus nigrogularis*	pied butcherbird
birds	Artamidae	*Cracticus torquatus*	grey butcherbird
birds	Artamidae	*Gymnorhina tibicen*	Australian magpie
birds	Artamidae	*Strepera graculina*	pied currawong
birds	Artamidae	*Strepera graculina graculina*	pied currawong (eastern Australia)
birds	Atrichornithidae	*Atrichornis rufescens*	rufous scrub-bird
birds	Burhinidae	*Burhinus grallarius*	bush stone-curlew
birds	Cacatuidae	*Cacatua galerita*	sulphur-crested cockatoo
birds	Cacatuidae	*Cacatua sanguinea*	little corella
birds	Cacatuidae	*Cacatua* sp.	
birds	Cacatuidae	*Cacatua tenuirostris*	long-billed corella
birds	Cacatuidae	*Calyptorhynchus banksii*	red-tailed black-cockatoo
birds	Cacatuidae	*Calyptorhynchus funereus*	yellow-tailed black-cockatoo
birds	Cacatuidae	*Calyptorhynchus lathami*	glossy black-cockatoo
birds	Cacatuidae	*Calyptorhynchus lathami lathami*	glossy black-cockatoo (eastern)
birds	Cacatuidae	*Eolophus roseicapilla*	galah
birds	Cacatuidae	*Nymphicus hollandicus*	cockatiel
birds	Campephagidae	*Coracina lineata*	barred cuckoo-shrike
birds	Campephagidae	*Coracina maxima*	ground cuckoo-shrike
birds	Campephagidae	*Coracina novaehollandiae*	black-faced cuckoo-shrike
birds	Campephagidae	*Coracina papuensis*	white-bellied cuckoo-shrike
birds	Campephagidae	*Coracina tenuirostris*	cicadabird
birds	Campephagidae	*Lalage leucomela*	varied triller
birds	Campephagidae	*Lalage tricolor*	white-winged triller

birds	Casuariidae	*Dromaius novaehollandiae*	emu
birds	Charadriidae	*Charadrius ruficapillus*	red-capped plover
birds	Charadriidae	*Elseyornis melanops*	black-fronted dotterel
birds	Charadriidae	*Erythrogonys cinctus*	red-kneed dotterel
birds	Charadriidae	*Vanellus miles*	masked lapwing
birds	Charadriidae	*Vanellus miles novaehollandiae*	masked lapwing (southern subspecies)
birds	Charadriidae	*Vanellus tricolor*	banded lapwing
birds	Ciconiidae	*Ephippiorhynchus asiaticus*	black-necked stork
birds	Cisticolidae	*Cisticola exilis*	golden-headed cisticola
birds	Climacteridae	*Climacteris erythrops*	red-browed treecreeper
birds	Climacteridae	*Climacteris picumnus*	brown treecreeper
birds	Climacteridae	*Climacteris picumnus victoriae*	brown treecreeper (south-eastern)
birds	Climacteridae	*Cormobates leucophaea*	white-throated treecreeper
birds	Climacteridae	*Cormobates leucophaea metastasis*	white-throated treecreeper (southern)
birds	Columbidae	*Chalcophaps indica*	emerald dove
birds	Columbidae	*Columba leucomela*	white-headed pigeon
birds	Columbidae	*Columba livia*	rock dove
birds	Columbidae	*Geopelia cuneata*	diamond dove
birds	Columbidae	*Geopelia humeralis*	bar-shouldered dove
birds	Columbidae	*Geopelia striata*	peaceful dove
birds	Columbidae	*Geophaps scripta*	squatter pigeon
birds	Columbidae	*Geophaps scripta scripta*	squatter pigeon (southern subspecies)
birds	Columbidae	*Leucosarcia melanoleuca*	wonga pigeon
birds	Columbidae	*Lopholaimus antarcticus*	topknot pigeon
birds	Columbidae	*Macropygia amboinensis*	brown cuckoo-dove
birds	Columbidae	*Ocyphaps lophotes*	crested pigeon
birds	Columbidae	*Phaps chalcoptera*	common bronzewing

birds	Columbidae	*Ptilinopus magnificus*	wompoo fruit-dove
birds	Columbidae	*Ptilinopus regina*	rose-crowned fruit-dove
birds	Columbidae	*Ptilinopus superbus*	superb fruit-dove
birds	Columbidae	*Streptopelia chinensis*	spotted dove
birds	Coraciidae	*Eurystomus orientalis*	dollarbird
birds	Corcoracidae	*Corcorax melanorhamphos*	white-winged chough
birds	Corcoracidae	*Struthidea cinerea*	apostlebird
birds	Corvidae	*Corvus coronoides*	Australian raven
birds	Corvidae	*Corvus orru*	Torresian crow
birds	Corvidae	*Corvus* sp.	
birds	Cuculidae	*Cacomantis flabelliformis*	fan-tailed cuckoo
birds	Cuculidae	*Cacomantis pallidus*	pallid cuckoo
birds	Cuculidae	*Cacomantis variolosus*	brush cuckoo
birds	Cuculidae	*Centropus phasianinus*	pheasant coucal
birds	Cuculidae	*Chalcites basalis*	Horsfield's bronze-cuckoo
birds	Cuculidae	*Chalcites lucidus*	shining bronze-cuckoo
birds	Cuculidae	*Chalcites minutillus*	little bronze-cuckoo
birds	Cuculidae	*Chalcites minutillus barnardi*	Eastern little bronze-cuckoo
birds	Cuculidae	*Chalcites osculans*	black-eared cuckoo
birds	Cuculidae	*Chalcites* sp.	
birds	Cuculidae	*Cuculus optatus*	oriental cuckoo
birds	Cuculidae	*Eudynamys orientalis*	eastern koel
birds	Cuculidae	*Scythrops novaehollandiae*	channel-billed cuckoo
birds	Dasyornithidae	*Dasyornis brachypterus*	eastern bristlebird
birds	Dicruridae	*Dicrurus bracteatus*	spangled drongo
birds	Estrildidae	*Lonchura castaneothorax*	chestnut-breasted mannikin
birds	Estrildidae	*Neochmia modesta*	plum-headed finch

birds	Estrildidae	*Neochmia temporalis*	red-browed finch
birds	Estrildidae	*Poephila cincta cincta*	black-throated finch (white-rumped subspecies)
birds	Estrildidae	*Stagonopleura guttata*	diamond firetail
birds	Estrildidae	*Taeniopygia bichenovii*	double-barred finch
birds	Estrildidae	*Taeniopygia guttata*	zebra finch
birds	Eurostopodidae	*Eurostopodus argus*	spotted nightjar
birds	Eurostopodidae	*Eurostopodus mystacalis*	white-throated nightjar
birds	Falconidae	*Falco berigora*	brown falcon
birds	Falconidae	*Falco cenchroides*	nankeen kestrel
birds	Falconidae	*Falco hypoleucos*	grey falcon
birds	Falconidae	*Falco longipennis*	Australian hobby
birds	Falconidae	*Falco peregrinus*	peregrine falcon
birds	Falconidae	*Falco* sp.	
birds	Falconidae	*Falco subniger*	black falcon
birds	Fringillidae	*Carduelis carduelis*	European goldfinch
birds	Gruidae	*Antigone rubicunda*	brolga
birds	Halcyonidae	*Dacelo leachii*	blue-winged kookaburra
birds	Halcyonidae	*Dacelo novaeguineae*	laughing kookaburra
birds	Halcyonidae	*Todiramphus macleayii*	forest kingfisher
birds	Halcyonidae	*Todiramphus pyrrhopygius*	red-backed kingfisher
birds	Halcyonidae	*Todiramphus sanctus*	sacred kingfisher
birds	Hirundinidae	*Cheramoeca leucosterna*	white-backed swallow
birds	Hirundinidae	*Hirundo neoxena*	welcome swallow
birds	Hirundinidae	*Hirundo rustica*	barn swallow
birds	Hirundinidae	*Petrochelidon ariel*	fairy martin
birds	Hirundinidae	*Petrochelidon nigricans*	tree martin
birds	Jacanidae	*Irediparra gallinacea*	comb-crested jacana

birds	Laridae	*Chlidonias hybrida*	whiskered tern
birds	Laridae	*Chroicocephalus novaehollandiae*	silver gull
birds	Laridae	*Gelochelidon nilotica*	gull-billed tern
birds	Laridae	*Hydroprogne caspia*	Caspian tern
birds	Laridae	*Thalasseus bergii*	crested tern
birds	Maluridae	*Malurus cyaneus*	superb fairy-wren
birds	Maluridae	*Malurus lamberti*	variegated fairy-wren
birds	Maluridae	*Malurus leucopterus*	white-winged fairy-wren
birds	Maluridae	*Malurus melanocephalus*	red-backed fairy-wren
birds	Maluridae	*Malurus* sp.	
birds	Maluridae	*Malurus splendens*	splendid fairy-wren
birds	Maluridae	*Stipiturus malachurus*	southern emu-wren
birds	Megaluridae	*Cincloramphus cruralis*	brown songlark
birds	Megaluridae	*Cincloramphus mathewsi*	rufous songlark
birds	Megaluridae	*Megalurus gramineus*	little grassbird
birds	Megaluridae	*Megalurus timoriensis*	tawny grassbird
birds	Megapodiidae	*Alectura lathami*	Australian brush-turkey
birds	Meliphagidae	*Acanthagenys rufogularis*	spiny-cheeked honeyeater
birds	Meliphagidae	*Acanthorhynchus tenuirostris*	eastern spinebill
birds	Meliphagidae	*Anthochaera carunculata*	red wattlebird
birds	Meliphagidae	*Anthochaera chrysoptera*	little wattlebird
birds	Meliphagidae	*Anthochaera phrygia*	regent honeyeater
birds	Meliphagidae	*Caligavis chrysops*	yellow-faced honeyeater
birds	Meliphagidae	*Entomyzon cyanotis*	blue-faced honeyeater
birds	Meliphagidae	*Epthianura albifrons*	white-fronted chat
birds	Meliphagidae	*Epthianura tricolor*	crimson chat
birds	Meliphagidae	*Gavicalis virescens*	singing honeyeater

birds	Meliphagidae	*Grantiella picta*	painted honeyeater
birds	Meliphagidae	*Lichenostomus melanops*	yellow-tufted honeyeater
birds	Meliphagidae	*Lichmera indistincta*	brown honeyeater
birds	Meliphagidae	*Manorina flavigula*	yellow-throated miner
birds	Meliphagidae	*Manorina melanocephala*	noisy miner
birds	Meliphagidae	*Manorina melanophrys*	bell miner
birds	Meliphagidae	*Meliphaga lewinii*	Lewin's honeyeater
birds	Meliphagidae	*Melithreptus albogularis*	white-throated honeyeater
birds	Meliphagidae	*Melithreptus brevirostris*	brown-headed honeyeater
birds	Meliphagidae	*Melithreptus gularis*	black-chinned honeyeater
birds	Meliphagidae	*Melithreptus gularis gularis*	black-chinned honeyeater (eastern)
birds	Meliphagidae	*Melithreptus gularis laetior*	golden-backed honeyeater
birds	Meliphagidae	*Melithreptus lunatus*	white-naped honeyeater
birds	Meliphagidae	*Myzomela obscura*	dusky honeyeater
birds	Meliphagidae	*Myzomela sanguinolenta*	scarlet honeyeater
birds	Meliphagidae	*Nesoptilotis leucotis*	white-eared honeyeater
birds	Meliphagidae	*Philemon citreogularis*	little friarbird
birds	Meliphagidae	*Philemon corniculatus*	noisy friarbird
birds	Meliphagidae	*Phylidonyris niger*	white-cheeked honeyeater
birds	Meliphagidae	*Phylidonyris novaehollandiae*	New Holland honeyeater
birds	Meliphagidae	*Plectorhyncha lanceolata*	striped honeyeater
birds	Meliphagidae	*Ptilotula flavescens*	yellow-tinted honeyeater
birds	Meliphagidae	*Ptilotula fusca*	fuscous honeyeater
birds	Meliphagidae	*Ptilotula penicillata*	white-plumed honeyeater
birds	Meliphagidae	*Ptilotula plumula*	grey-fronted honeyeater
birds	Meliphagidae	*Purnella albifrons*	white-fronted honeyeater
birds	Meliphagidae	*Sugomel niger*	black honeyeater

birds	Menuridae	*Menura alberti*	Albert's lyrebird
birds	Menuridae	*Menura novaehollandiae*	superb lyrebird
birds	Meropidae	*Merops ornatus*	rainbow bee-eater
birds	Monarchidae	*Carterornis leucotis*	white-eared monarch
birds	Monarchidae	*Grallina cyanoleuca*	magpie-lark
birds	Monarchidae	*Monarcha melanopsis*	black-faced monarch
birds	Monarchidae	*Myiagra cyanoleuca*	satin flycatcher
birds	Monarchidae	*Myiagra inquieta*	restless flycatcher
birds	Monarchidae	*Myiagra rubecula*	leaden flycatcher
birds	Monarchidae	*Symposiachrus trivirgatus*	spectacled monarch
birds	Motacillidae	*Anthus novaeseelandiae*	Australasian pipit
birds	Nectariniidae	*Dicaeum hirundinaceum*	mistletoebird
birds	Neosittidae	*Daphoenositta chrysoptera*	varied sittella
birds	Oriolidae	*Oriolus sagittatus*	olive-backed oriole
birds	Oriolidae	*Sphecotheres vieilloti*	Australasian figbird
birds	Orthonychidae	*Orthonyx temminckii*	Australian logrunner
birds	Pachycephalidae	*Colluricincla harmonica*	grey shrike-thrush
birds	Pachycephalidae	*Colluricincla megarhyncha*	little shrike-thrush
birds	Pachycephalidae	*Falcunculus frontatus*	crested shrike-tit
birds	Pachycephalidae	*Oreoica gutturalis*	crested bellbird
birds	Pachycephalidae	*Pachycephala olivacea*	olive whistler
birds	Pachycephalidae	*Pachycephala pectoralis*	golden whistler
birds	Pachycephalidae	*Pachycephala pectoralis youngi*	golden whistler (south-eastern Australia)
birds	Pachycephalidae	*Pachycephala rufiventris*	rufous whistler
birds	Paradisaeidae	*Ptiloris paradiseus*	paradise riflebird
birds	Pardalotidae	*Pardalotus punctatus*	spotted pardalote
birds	Pardalotidae	*Pardalotus striatus*	striated pardalote

birds	Passeridae	*Passer domesticus*	house sparrow
birds	Pelecanidae	*Pelecanus conspicillatus*	Australian pelican
birds	Petroicidae	*Eopsaltria australis*	eastern yellow robin
birds	Petroicidae	*Melanodryas cucullata*	hooded robin
birds	Petroicidae	*Microeca fascinans*	jacky winter
birds	Petroicidae	*Petroica boodang*	scarlet robin
birds	Petroicidae	*Petroica goodenovii*	red-capped robin
birds	Petroicidae	*Petroica phoenicea*	flame robin
birds	Petroicidae	*Petroica rosea*	rose robin
birds	Petroicidae	*Tregellasia capito*	pale-yellow robin
birds	Phalacrocoracidae	*Microcarbo melanoleucos*	little pied cormorant
birds	Phalacrocoracidae	*Phalacrocorax carbo*	great cormorant
birds	Phalacrocoracidae	*Phalacrocorax sulcirostris*	little black cormorant
birds	Phalacrocoracidae	*Phalacrocorax varius*	pied cormorant
birds	Phasianidae	*Coturnix pectoralis*	stubble quail
birds	Phasianidae	*Coturnix ypsilophora*	brown quail
birds	Phasianidae	*Excalfactoria chinensis*	king quail
birds	Pittidae	*Pitta versicolor*	noisy pitta
birds	Podargidae	*Podargus strigoides*	tawny frogmouth
birds	Podicipedidae	*Podiceps cristatus*	great crested grebe
birds	Podicipedidae	*Poliocephalus poliocephalus*	hoary-headed grebe
birds	Podicipedidae	*Tachybaptus novaehollandiae*	Australasian grebe
birds	Pomatostomidae	*Pomatostomus superciliosus*	white-browed babbler
birds	Pomatostomidae	*Pomatostomus temporalis*	grey-crowned babbler
birds	Pomatostomidae	*Pomatostomus temporalis temporalis*	grey-crowned babbler (eastern)
birds	Procellariidae	*Pterodroma nigripennis*	black-winged petrel
birds	Psittacidae	*Alisterus scapularis*	Australian king-parrot

birds	Psittacidae	*Aprosmictus erythropterus*	red-winged parrot
birds	Psittacidae	*Barnardius zonarius*	Australian ringneck
birds	Psittacidae	*Cyclopsitta diophthalma coxeni*	Coxen's fig-parrot
birds	Psittacidae	*Glossopsitta concinna*	musk lorikeet
birds	Psittacidae	*Lathamus discolor*	swift parrot
birds	Psittacidae	*Melopsittacus undulatus*	budgerigar
birds	Psittacidae	*Neophema pulchella*	turquoise parrot
birds	Psittacidae	*Northiella haematogaster*	blue bonnet
birds	Psittacidae	*Parvipsitta porphyrocephala*	purple-crowned lorikeet
birds	Psittacidae	*Parvipsitta pusilla*	little lorikeet
birds	Psittacidae	*Platycercus adscitus*	pale-headed rosella
birds	Psittacidae	*Platycercus adscitus palliceps*	pale-headed rosella (southern form)
birds	Psittacidae	*Platycercus elegans*	crimson rosella
birds	Psittacidae	*Platycercus elegans elegans*	crimson rosella (nom. subsp.)
birds	Psittacidae	*Platycercus eximius*	eastern rosella
birds	Psittacidae	*Platycercus* sp.	
birds	Psittacidae	*Psephotus haematonotus*	red-rumped parrot
birds	Psittacidae	*Trichoglossus chlorolepidotus*	scaly-breasted lorikeet
birds	Psittacidae	*Trichoglossus haematodus moluccanus*	rainbow lorikeet
birds	Psophodidae	*Cinclosoma punctatum*	spotted quail-thrush
birds	Psophodidae	*Psophodes olivaceus*	eastern whipbird
birds	Ptilonorhynchidae	*Ailuroedus crassirostris*	green catbird
birds	Ptilonorhynchidae	*Ptilonorhynchus maculatus*	spotted bowerbird
birds	Ptilonorhynchidae	*Ptilonorhynchus violaceus*	satin bowerbird
birds	Ptilonorhynchidae	*Sericulus chrysocephalus*	regent bowerbird
birds	Rallidae	*Fulica atra*	Eurasian coot
birds	Rallidae	*Gallinula tenebrosa*	dusky moorhen

birds	Rallidae	*Gallirallus philippensis*	buff-banded rail
birds	Rallidae	*Lewinia pectoralis*	Lewin's rail
birds	Rallidae	*Porphyrio melanotus*	purple swamphen
birds	Rallidae	*Porzana fluminea*	Australian spotted crake
birds	Rallidae	*Porzana pusilla*	Baillon's crake
birds	Rallidae	*Tribonyx ventralis*	black-tailed native-hen
birds	Recurvirostridae	*Himantopus himantopus*	black-winged stilt
birds	Recurvirostridae	*Recurvirostra novaehollandiae*	red-necked avocet
birds	Rhipiduridae	*Rhipidura albiscapa*	grey fantail
birds	Rhipiduridae	*Rhipidura leucophrys*	willie wagtail
birds	Rhipiduridae	*Rhipidura rufifrons*	rufous fantail
birds	Rostratulidae	*Rostratula australis*	Australian painted snipe
birds	Scolopacidae	*Calidris acuminata*	sharp-tailed sandpiper
birds	Scolopacidae	*Calidris ferruginea*	curlew sandpiper
birds	Scolopacidae	*Calidris ruficollis*	red-necked stint
birds	Scolopacidae	*Gallinago hardwickii*	Latham's snipe
birds	Scolopacidae	*Gallinago* sp.	
birds	Scolopacidae	*Tringa nebularia*	common greenshank
birds	Scolopacidae	*Tringa stagnatilis*	marsh sandpiper
birds	Strigidae	*Ninox boobook*	southern boobook
birds	Strigidae	*Ninox connivens*	barking owl
birds	Strigidae	*Ninox strenua*	powerful owl
birds	Sturnidae	*Acridotheres tristis*	common myna
birds	Sturnidae	*Sturnus vulgaris*	common starling
birds	Threskiornithidae	*Platalea flavipes*	yellow-billed spoonbill
birds	Threskiornithidae	*Platalea regia*	royal spoonbill
birds	Threskiornithidae	*Plegadis falcinellus*	glossy ibis

birds	Threskiornithidae	*Threskiornis molucca*	Australian white ibis
birds	Threskiornithidae	*Threskiornis spinicollis*	straw-necked ibis
birds	Timaliidae	*Zosterops lateralis*	silvereye
birds	Timaliidae	*Zosterops lateralis cornwalli*	silvereye (eastern)
birds	Turdidae	*Turdus merula*	common blackbird
birds	Turdidae	*Zoothera heinei*	russet-tailed thrush
birds	Turdidae	*Zoothera lunulata*	Bassian thrush
birds	Turdidae	*Zoothera lunulata lunulata*	bassian thrush (south-east Queensland)
birds	Turdidae	*Zoothera* sp.	
birds	Turnicidae	*Turnix melanogaster*	black-breasted button-quail
birds	Turnicidae	*Turnix pyrrhothorax*	red-chested button-quail
birds	Turnicidae	*Turnix varius*	painted button-quail
birds	Turnicidae	*Turnix velox*	little button-quail
birds	Tytonidae	*Tyto delicatula*	eastern barn owl
birds	Tytonidae	*Tyto novaehollandiae*	masked owl
birds	Tytonidae	*Tyto novaehollandiae novaehollandiae*	masked owl (southern subspecies)
birds	Tytonidae	*Tyto tenebricosa tenebricosa*	sooty owl
reptiles	Agamidae	*Amphibolurus burnsi*	Burns's dragon
reptiles	Agamidae	*Amphibolurus muricatus*	jacky lizard
reptiles	Agamidae	*Amphibolurus* sp.	
reptiles	Agamidae	*Diporiphora australis*	tommy roundhead
reptiles	Agamidae	*Diporiphora nobbi*	nobbi
reptiles	Agamidae	*Intellagama lesueurii*	eastern water dragon
reptiles	Agamidae	*Lophognathus gilberti sensu lato*	Gilbert's dragon
reptiles	Agamidae	*Lophosaurus spinipes*	southern angle-headed dragon
reptiles	Agamidae	*Pogona barbata*	bearded dragon
reptiles	Boidae	*Antaresia maculosa*	spotted python

reptiles	Boidae	*Morelia spilota*	carpet python
reptiles	Carphodactylidae	*Saltuarius swaini*	southern leaf-tailed gecko
reptiles	Carphodactylidae	*Saltuarius wyberba*	granite leaf-tailed gecko
reptiles	Carphodactylidae	*Uvidicolus sphyrurus*	border thick-tailed gecko
reptiles	Chelidae	*Chelodina expansa*	broad-shelled river turtle
reptiles	Chelidae	*Chelodina longicollis*	eastern snake-necked turtle
reptiles	Chelidae	*Emydura macquarii macquarii*	Murray turtle
reptiles	Chelidae	*Emydura* sp.	
reptiles	Chelidae	*Wollumbinia belli*	Bell's turtle
reptiles	Chelidae	*Wollumbinia latisternum*	saw-shelled turtle
reptiles	Colubridae	*Boiga irregularis*	brown tree snake
reptiles	Colubridae	*Dendrelaphis punctulatus*	green tree snake
reptiles	Colubridae	*Tropidonophis mairii*	freshwater snake
reptiles	Diplodactylidae	*Amalosia jacovae*	clouded gecko
reptiles	Diplodactylidae	*Amalosia lesueurii*	Lesueur's velvet gecko
reptiles	Diplodactylidae	*Amalosia rhombifer*	zig-zag gecko
reptiles	Diplodactylidae	*Diplodactylus* sp.	
reptiles	Diplodactylidae	*Diplodactylus vittatus*	wood gecko
reptiles	Diplodactylidae	*Nebulifera robusta*	robust velvet gecko
reptiles	Diplodactylidae	*Oedura marmorata sensu lato*	marbled velvet gecko
reptiles	Diplodactylidae	*Oedura tryoni*	southern spotted velvet gecko
reptiles	Diplodactylidae	*Strophurus williamsi*	soft-spined gecko
reptiles	Elapidae	*Acanthophis antarcticus*	common death adder
reptiles	Elapidae	*Brachyurophis australis*	coral snake
reptiles	Elapidae	*Cacophis harriettae*	white-crowned snake
reptiles	Elapidae	*Cacophis krefftii*	dwarf crowned snake
reptiles	Elapidae	*Cacophis squamulosus*	golden crowned snake

reptiles	Elapidae	*Cryptophis nigrescens*	eastern small-eyed snake
reptiles	Elapidae	*Demansia psammophis*	yellow-faced whipsnake
reptiles	Elapidae	*Furina diadema*	red-naped snake
reptiles	Elapidae	*Hemiaspis signata*	black-bellied swamp snake
reptiles	Elapidae	*Hoplocephalus bitorquatus*	pale-headed snake
reptiles	Elapidae	*Hoplocephalus stephensii*	Stephens' banded snake
reptiles	Elapidae	*Notechis scutatus*	eastern tiger snake
reptiles	Elapidae	*Pseudechis guttatus*	spotted black snake
reptiles	Elapidae	*Pseudechis porphyriacus*	red-bellied black snake
reptiles	Elapidae	*Pseudonaja textilis*	eastern brown snake
reptiles	Elapidae	*Suta dwyeri*	Dwyer's snake
reptiles	Elapidae	*Suta suta*	myall snake
reptiles	Elapidae	*Tropidechis carinatus*	rough-scaled snake
reptiles	Elapidae	*Vermicella annulata*	bandy-bandy
reptiles	Gekkonidae	*Gehyra dubia*	dubious dtella
reptiles	Gekkonidae	*Heteronotia binoei*	Bynoe's gecko
reptiles	Pygopodidae	*Delma inornata*	patternless delma
reptiles	Pygopodidae	*Delma plebeia*	common delma
reptiles	Pygopodidae	*Delma* sp.	
reptiles	Pygopodidae	*Delma tincta*	excitable delma
reptiles	Pygopodidae	*Delma torquata*	collared delma
reptiles	Pygopodidae	*Lialis burtonis*	Burton's legless lizard
reptiles	Pygopodidae	*Pygopus lepidopodus*	common scaly-foot
reptiles	Scincidae	*Acritoscincus platynotum*	red-throated skink
reptiles	Scincidae	*Anomalopus leuckartii*	two-clawed worm-skink
reptiles	Scincidae	*Anomalopus verreauxii*	three-clawed worm-skink
reptiles	Scincidae	*Bellatorias frerei*	major skink

reptiles	Scincidae	*Bellatorias major*	land mullet
reptiles	Scincidae	*Calyptotis scutirostrum*	scute-snouted calyptotis
reptiles	Scincidae	*Carlia munda*	shaded-litter rainbow-skink
reptiles	Scincidae	*Carlia pectoralis*	open-litter rainbow skink
reptiles	Scincidae	*Carlia pectoralis sensu lato*	
reptiles	Scincidae	*Carlia* sp.	
reptiles	Scincidae	*Carlia tetradactyla*	southern rainbow-skink
reptiles	Scincidae	*Carlia vivax*	tussock rainbow-skink
reptiles	Scincidae	*Coeranoscincus reticulatus*	three-toed snake-tooth skink
reptiles	Scincidae	*Concinnia brachysoma*	northern bar-sided skink
reptiles	Scincidae	*Concinnia martini*	dark bar-sided skink
reptiles	Scincidae	*Concinnia tenuis*	bar-sided skink
reptiles	Scincidae	*Cryptoblepharus pulcher pulcher*	elegant snake-eyed skink
reptiles	Scincidae	*Cryptoblepharus virgatus sensu lato*	
reptiles	Scincidae	*Ctenotus eurydice*	brown-backed yellow-lined ctenotus
reptiles	Scincidae	*Ctenotus* sp.	
reptiles	Scincidae	*Ctenotus spaldingi*	straight-browed ctenotus
reptiles	Scincidae	*Ctenotus taeniolatus*	copper-tailed skink
reptiles	Scincidae	*Cyclodomorphus gerrardii*	pink-tongued lizard
reptiles	Scincidae	*Egernia cunninghami*	Cunningham's skink
reptiles	Scincidae	*Egernia mcpheei*	eastern crevice-skink
reptiles	Scincidae	*Egernia striolata*	tree skink
reptiles	Scincidae	*Eulamprus quoyii*	eastern water skink
reptiles	Scincidae	*Eulamprus* sp.	
reptiles	Scincidae	*Harrisoniascincus zia*	rainforest cool-skink
reptiles	Scincidae	*Karma murrayi*	Murray's skink
reptiles	Scincidae	*Lampropholis amicula*	friendly sunskink

reptiles	Scincidae	*Lampropholis delicata*	dark-flecked garden sunskink
reptiles	Scincidae	*Lampropholis guichenoti*	pale-flecked garden sunskink
reptiles	Scincidae	*Lampropholis* sp.	
reptiles	Scincidae	*Lerista fragilis*	eastern mulch slider
reptiles	Scincidae	*Lerista timida*	timid slider
reptiles	Scincidae	*Liopholis modesta*	eastern ranges rock-skink
reptiles	Scincidae	*Liopholis whitii*	White's skink
reptiles	Scincidae	*Lygisaurus foliorum*	tree-base litter-skink
reptiles	Scincidae	*Menetia greyii*	common dwarf skink
reptiles	Scincidae	*Morethia boulengeri*	south-eastern morethia skink
reptiles	Scincidae	*Morethia taeniopleura*	fire-tailed skink
reptiles	Scincidae	*Ophioscincus truncatus*	short-limbed snake-skink
reptiles	Scincidae	*Saiphos equalis*	three-toed skink
reptiles	Scincidae	*Saproscincus challengeri*	orange-tailed shadeskink
reptiles	Scincidae	*Saproscincus rosei*	Rose's shadeskink
reptiles	Scincidae	*Saproscincus* sp.	
reptiles	Scincidae	*Saproscincus spectabilis*	pale-lipped shadeskink
reptiles	Scincidae	*Tiliqua scincoides*	eastern blue-tongued lizard
reptiles	Typhlopidae	*Anilios ligatus*	robust blind snake
reptiles	Typhlopidae	*Anilios nigrescens*	blackish blind snake
reptiles	Typhlopidae	*Anilios proximus*	proximus blind snake
reptiles	Typhlopidae	*Anilios* sp.	
reptiles	Typhlopidae	*Anilios wiedii*	brown-snouted blind snake
reptiles	Varanidae	*Varanus gouldii*	sand monitor
reptiles	Varanidae	*Varanus panoptes*	yellow-spotted monitor
reptiles	Varanidae	*Varanus tristis*	black-tailed monitor
reptiles	Varanidae	*Varanus varius*	lace monitor

amphibians	Hylidae	*Cyclorana alboguttata*	greenstripe frog
amphibians	Hylidae	*Litoria caerulea*	common green treefrog
amphibians	Hylidae	*Litoria chloris*	orange eyed treefrog
amphibians	Hylidae	*Litoria dentata*	bleating treefrog
amphibians	Hylidae	*Litoria fallax*	eastern sedgefrog
amphibians	Hylidae	*Litoria gracilenta*	graceful treefrog
amphibians	Hylidae	*Litoria latopalmata*	broad palmed rocketfrog
amphibians	Hylidae	*Litoria nasuta*	striped rocketfrog
amphibians	Hylidae	*Litoria pearsoniana*	cascade treefrog
amphibians	Hylidae	*Litoria peronii*	emerald spotted treefrog
amphibians	Hylidae	*Litoria revelata*	whirring treefrog
amphibians	Hylidae	*Litoria rubella*	ruddy treefrog
amphibians	Hylidae	*Litoria* sp.	
amphibians	Hylidae	*Litoria subglandulosa*	New England treefrog
amphibians	Hylidae	*Litoria tyleri*	southern laughing treefrog
amphibians	Hylidae	*Litoria verreauxii*	whistling treefrog
amphibians	Hylidae	*Litoria wilcoxii*	eastern stony creek frog
amphibians	Limnodynastidae	*Adelotus brevis*	tusked frog
amphibians	Limnodynastidae	*Lechriodus fletcheri*	black soled frog
amphibians	Limnodynastidae	*Limnodynastes dumerilii*	grey bellied pobblebonk
amphibians	Limnodynastidae	*Limnodynastes fletcheri*	barking frog
amphibians	Limnodynastidae	*Limnodynastes peronii*	striped marshfrog
amphibians	Limnodynastidae	*Limnodynastes* sp.	
amphibians	Limnodynastidae	*Limnodynastes tasmaniensis*	spotted grassfrog
amphibians	Limnodynastidae	*Limnodynastes terraereginae*	scarlet sided pobblebonk
amphibians	Limnodynastidae	*Neobatrachus sudellae*	meeowing frog
amphibians	Limnodynastidae	*Philoria kundagungan*	red-and-yellow mountainfrog

amphibians	Limnodynastidae	*Platyplectrum ornatum*	ornate burrowing frog
amphibians	Myobatrachidae	*Crinia parinsignifera*	beeping froglet
amphibians	Myobatrachidae	*Crinia signifera*	clicking froglet
amphibians	Myobatrachidae	*Crinia* sp.	
amphibians	Myobatrachidae	*Mixophyes fasciolatus*	great barred frog
amphibians	Myobatrachidae	*Mixophyes fleayi*	Fleay's barred frog
amphibians	Myobatrachidae	*Mixophyes iteratus*	giant barred frog
amphibians	Myobatrachidae	*Mixophyes* sp.	
amphibians	Myobatrachidae	*Pseudophryne coriacea*	red backed broodfrog
amphibians	Myobatrachidae	*Pseudophryne major*	great brown broodfrog
amphibians	Myobatrachidae	*Pseudophryne* sp.	
amphibians	Myobatrachidae	*Uperoleia fusca*	dusky gungan
amphibians	Myobatrachidae	*Uperoleia laevigata*	eastern gungan
amphibians	Myobatrachidae	*Uperoleia rugosa*	chubby gungan

Appendix C: Diagram showing how cats were allocated BCSs.

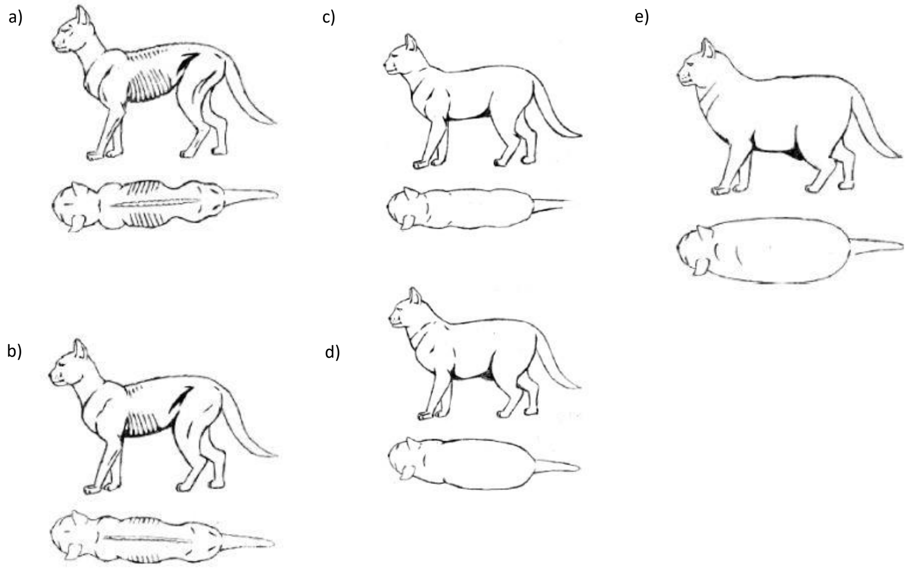

Figure 10. BCS 1: emaciated, ribs, spine and pelvis clearly visible and easily palpable. Abdomen is clearly concave (a). BCS 2: poor, ribs, spine and pelvis partially visible and palpable with thin layer of tissue. Abdomen is concave (b). BCS 3: ideal, ribs, spine and pelvis not visible but can be felt with layer of tissue covering them. Abdomen is slightly concave (c). BCS 4: overweight, ribs, spine and pelvis are not visible or palpable. Abdomen is slightly convex (d). BCS 5: obese, ribs, spine and pelvis are not visible or palpable, fat flap on ventral side present. Abdomen is clearly convex (e) (adapted from Animal Welfare Victoria 2021).

Appendix D: Vertebrate species list for DDS

Table 15. List of vertebrate species present at DDS as of 2013 (Department of Environment and Science 2013a).

Class	Family	Scientific Name	Common Name
mammals	Bovidae	*Bos taurus*	European cattle
mammals	Suidae	*Sus scrofa*	pig
mammals	Felidae	*Felis catus*	cat
mammals	Canidae	*Canis familiaris*	dog
mammals	Canidae	*Canis dingo*	dingo
mammals	Muridae	*Mus musculus*	house mouse
mammals	Muridae	*Rattus villosissimus*	long-haired rat
mammals	Thylacomyidae	*Macrotis lagotis*	greater bilby
mammals	Dasyuridae	*Antechinomys laniger*	kultarr
mammals	Dasyuridae	*Dasyuroides byrnei*	kowari
mammals	Dasyuridae	*Planigale tenuirostris*	narrow-nosed planigale
mammals	Dasyuridae	*Sminthopsis crassicaudata*	fat-tailed dunnart
mammals	Dasyuridae	*Sminthopsis macroura*	stripe-faced dunnart
birds	Motacillidae	*Anthus novaeseelandiae*	Australasian pipit
birds	Estrildidae	*Taeniopygia guttata*	zebra finch
birds	Nectariniidae	*Dicaeum hirundinaceum*	mistletoebird
birds	Hirundinidae	*Petrochelidon ariel*	fairy martin
birds	Hirundinidae	*Petrochelidon nigricans*	tree martin
birds	Megaluridae	*Cincloramphus cruralis*	brown songlark
birds	Megaluridae	*Cincloramphus mathewsi*	rufous songlark
birds	Megaluridae	*Megalurus gramineus*	little grassbird

birds	Megaluridae	*Megalurus timoriensis*	tawny grassbird
birds	Acrocephalidae	*Acrocephalus australis*	Australian reed-warbler
birds	Alaudidae	*Mirafra javanica*	Horsfield's bushlark
birds	Petroicidae	*Microeca fascinans*	jacky winter
birds	Petroicidae	*Petroica goodenovii*	red-capped robin
birds	Monarchidae	*Grallina cyanoleuca*	magpie-lark
birds	Monarchidae	*Myiagra inquieta*	restless flycatcher
birds	Corvidae	*Corvus bennetti*	little crow
birds	Corvidae	*Corvus coronoides*	Australian raven
birds	Corvidae	*Corvus orru*	Torresian crow
birds	Corvidae	*Corvus* sp.	
birds	Rhipiduridae	*Rhipidura albiscapa*	grey fantail
birds	Rhipiduridae	*Rhipidura leucophrys*	willie wagtail
birds	Artamidae	*Artamus cinereus*	black-faced woodswallow
birds	Artamidae	*Artamus leucorynchus*	white-breasted woodswallow
birds	Artamidae	*Artamus minor*	little woodswallow
birds	Artamidae	*Artamus personatus*	masked woodswallow
birds	Artamidae	*Artamus superciliosus*	white-browed woodswallow
birds	Artamidae	*Gymnorhina tibicen*	Australian magpie
birds	Pachycephalidae	*Pachycephala rufiventris*	rufous whistler
birds	Campephagidae	*Coracina novaehollandiae*	black-faced cuckoo-shrike
birds	Campephagidae	*Lalage tricolor*	white-winged triller
birds	Psophodidae	*Cinclosoma cinnamomeum*	cinnamon quail-thrush
birds	Meliphagidae	*Acanthagenys rufogularis*	spiny-cheeked honeyeater
birds	Meliphagidae	*Ashbyia lovensis*	gibberbird
birds	Meliphagidae	*Epthianura albifrons*	white-fronted chat
birds	Meliphagidae	*Epthianura aurifrons*	orange chat

birds	Meliphagidae	*Epthianura crocea*	yellow chat
birds	Meliphagidae	*Epthianura tricolor*	crimson chat
birds	Meliphagidae	*Gavicalis virescens*	singing honeyeater
birds	Meliphagidae	*Manorina flavigula*	yellow-throated miner
birds	Meliphagidae	*Ptilotula penicillata*	white-plumed honeyeater
birds	Pardalotidae	*Pardalotus rubricatus*	red-browed pardalote
birds	Acanthizidae	*Acanthiza apicalis*	inland thornbill
birds	Acanthizidae	*Acanthiza nana*	yellow thornbill
birds	Acanthizidae	*Acanthiza reguloides*	buff-rumped thornbill
birds	Maluridae	*Malurus lamberti sensu lato*	variegated fairy-wren
birds	Maluridae	*Malurus leucopterus*	white-winged fairy-wren
birds	Meropidae	*Merops ornatus*	rainbow bee-eater
birds	Halcyonidae	*Todiramphus pyrrhopygius*	red-backed kingfisher
birds	Tytonidae	*Tyto delicatula*	eastern barn owl
birds	Tytonidae	*Tyto longimembris*	eastern grass owl
birds	Strigidae	*Ninox boobook*	southern boobook
birds	Cuculidae	*Cacomantis pallidus*	pallid cuckoo
birds	Cuculidae	*Scythrops novaehollandiae*	channel-billed cuckoo
birds	Psittacidae	*Barnardius zonarius*	Australian ringneck
birds	Psittacidae	*Melopsittacus undulatus*	budgerigar
birds	Cacatuidae	*Cacatua sanguinea*	little corella
birds	Cacatuidae	*Calyptorhynchus banksii*	red-tailed black-cockatoo
birds	Cacatuidae	*Eolophus roseicapilla*	galah
birds	Cacatuidae	*Nymphicus hollandicus*	cockatiel
birds	Laridae	*Chlidonias hybrida*	whiskered tern
birds	Glareolidae	*Stiltia isabella*	Australian pratincole
birds	Turnicidae	*Turnix velox*	little button-quail

birds	Scolopacidae	*Calidris acuminata*	sharp-tailed sandpiper
birds	Rostratulidae	*Rostratula australis*	Australian painted snipe
birds	Pedionomidae	*Pedionomus torquatus*	plains-wanderer
birds	Charadriidae	*Charadrius australis*	inland dotterel
birds	Charadriidae	*Charadrius veredus*	oriental plover
birds	Charadriidae	*Elseyornis melanops*	black-fronted dotterel
birds	Charadriidae	*Erythrogonys cinctus*	red-kneed dotterel
birds	Charadriidae	*Vanellus miles novaehollandiae*	masked lapwing (southern subspecies)
birds	Charadriidae	*Vanellus tricolor*	banded lapwing
birds	Otididae	*Ardeotis australis*	Australian bustard
birds	Rallidae	*Gallinula tenebrosa*	dusky moorhen
birds	Rallidae	*Porphyrio melanotus*	purple swamphen
birds	Rallidae	*Porzana fluminea*	Australian spotted crake
birds	Rallidae	*Porzana tabuensis*	spotless crake
birds	Rallidae	*Tribonyx ventralis*	black-tailed native-hen
birds	Gruidae	*Antigone rubicunda*	brolga
birds	Falconidae	*Falco berigora*	brown falcon
birds	Falconidae	*Falco cenchroides*	nankeen kestrel
birds	Falconidae	*Falco hypoleucos*	grey falcon
birds	Falconidae	*Falco subniger*	black falcon
birds	Accipitridae	*Accipiter fasciatus*	brown goshawk
birds	Accipitridae	*Aquila audax*	wedge-tailed eagle
birds	Accipitridae	*Circus approximans*	swamp harrier
birds	Accipitridae	*Circus assimilis*	spotted harrier
birds	Accipitridae	*Elanus axillaris*	black-shouldered kite
birds	Accipitridae	*Elanus scriptus*	letter-winged kite
birds	Accipitridae	*Haliastur sphenurus*	whistling kite

birds	Accipitridae	*Milvus migrans*	black kite
birds	Threskiornithidae	*Platalea flavipes*	yellow-billed spoonbill
birds	Threskiornithidae	*Platalea regia*	royal spoonbill
birds	Threskiornithidae	*Threskiornis molucca*	Australian white ibis
birds	Ardeidae	*Ardea alba modesta*	eastern great egret
birds	Ardeidae	*Ardea pacifica*	white-necked heron
birds	Ardeidae	*Egretta novaehollandiae*	white-faced heron
birds	Ardeidae	*Nycticorax caledonicus*	nankeen night-heron
birds	Pelecanidae	*Pelecanus conspicillatus*	Australian pelican
birds	Phalacrocoracidae	*Microcarbo melanoleucos*	little pied cormorant
birds	Anhingidae	*Anhinga novaehollandiae*	Australasian darter
birds	Apodidae	*Apus pacificus*	fork-tailed swift
birds	Eurostopodidae	*Eurostopodus argus*	spotted nightjar
birds	Columbidae	*Geopelia cuneata*	diamond dove
birds	Columbidae	*Geopelia striata*	peaceful dove
birds	Columbidae	*Ocyphaps lophotes*	crested pigeon
birds	Columbidae	*Phaps chalcoptera*	common bronzewing
birds	Columbidae	*Phaps histrionica*	flock bronzewing
birds	Podicipedidae	*Poliocephalus poliocephalus*	hoary-headed grebe
birds	Podicipedidae	*Tachybaptus novaehollandiae*	Australasian grebe
birds	Anatidae	*Anas gracilis*	grey teal
birds	Anatidae	*Anas superciliosa*	Pacific black duck
birds	Anatidae	*Aythya australis*	hardhead
birds	Anatidae	*Chenonetta jubata*	Australian wood duck
birds	Anatidae	*Dendrocygna eytoni*	plumed whistling-duck
birds	Anatidae	*Malacorhynchus membranaceus*	pink-eared duck
birds	Phasianidae	*Coturnix pectoralis*	stubble quail

birds	Phasianidae	*Coturnix ypsilophora*	brown quail
birds	Casuariidae	*Dromaius novaehollandiae*	emu
reptiles	Elapidae	*Demansia rimicola*	soil-crack whipsnake
reptiles	Elapidae	*Oxyuranus microlepidotus*	western taipan
reptiles	Elapidae	*Suta suta*	myall snake
reptiles	Boidae	*Antaresia stimsoni*	Stimson's python
reptiles	Scincidae	*Austroablepharus kinghorni*	red-tailed soil-crevice skink
reptiles	Scincidae	*Ctenotus lateralis*	gravelly-soil ctenotus
reptiles	Scincidae	*Ctenotus* sp.	
reptiles	Scincidae	*Eremiascincus richardsonii*	broad-banded sand swimmer
reptiles	Scincidae	*Menetia greyii*	common dwarf skink
reptiles	Varanidae	*Varanus gouldii*	sand monitor
reptiles	Varanidae	*Varanus spenceri*	Spencer's monitor
reptiles	Agamidae	*Tympanocryptis lineata*	lined earless dragon
reptiles	Agamidae	*Tympanocryptis tetraporophora*	Eyrean earless dragon
reptiles	Diplodactylidae	*Diplodactylus* sp.	
reptiles	Gekkonidae	*Heteronotia binoei*	Bynoe's gecko
amphibians	Limnodynastidae	*Limnodynastes tasmaniensis*	spotted grassfrog

Appendix E: Results from Shapiro-Wilk normality tests

Table 16. Shapiro-Wilk normality test results for each group and measurement taken. P-value < 0.050 indicates data is not normally distributed.

Measure	Group	w	df	p-value
Weight	Adult male	0.978	19	0.923
	Adult female	0.947	19	0.345
	Juvenile male	0.893	14	0.089
	Juvenile female	0.922	16	0.178
Nose-tail	Adult male	0.917	18	0.113
	Adult female	0.908	18	0.081
	Juvenile male	0.939	14	0.409
	Juvenile female	0.923	16	0.187
Forearm	Adult male	0.905	18	0.069
	Adult female	0.633	18	<0.001
	Juvenile male	0.932	14	0.327
	Juvenile female	0.909	16	0.111
Femur	Adult male	0.897	18	0.051
	Adult female	0.975	18	0.883
	Juvenile male	0.916	14	0.194
	Juvenile female	0.879	16	0.038
Head width	Adult male	0.952	18	0.460
	Adult female	0.939	18	0.28
	Juvenile male	0.92	14	0.222
	Juvenile female	0.924	16	0.198
KFI	Adult male	0.925	18	0.157
	Adult female	0.893	18	0.044
	Juvenile male	0.713	14	<0.001
	Juvenile female	0.676	16	<0.001
BCS	Adult male	0.717	18	<0.001
	Adult female	0.849	18	0.008
	Juvenile male	0.75	14	0.001
	Juvenile female	0.759	16	<0.001

Appendix F: Necropsy data

Table 17. Data taken during necropsy of SDR unowned cats

Cat_ID	Weight(g)	Sex	Age	BCS	Neutered	Cat_type_colour	KFI	Femur_length (mm)	Nose_tail (cm)	Forearm(mm)	Head_width (mm)	Stomach full(y/n)	Teeth	Location	Injuries_SoF	Notes
1	3750	f	2	3	n	tortoiseshell	21.89	100	80	47.05	67.5	y	broken, plaque, missing incisors	STANTHORPE	red front paws	n/a
2	3100	f	2	1	n	black	5.62	110	82.5	112	68.63	y	n/a	STANTHORPE	n/a	just finished lactating, discoloured kidney
3	3100	f	2	3	n	tortoiseshell & white	16.56	100	80	104	69	y	broken upper right canine	STANTHORPE	n/a	lactating
4	4950	m	2	2	n	tabby	19.62	115	89	120	75	y	n/a	STANTHORPE	n/a	cat food
5	825	f	1	2	n	brown tabby	6.99	79	47.5	70	46	y	n/a	STANTHORPE	n/a	kitten
6	470	m	1	2	n	ginger	5.73	48	40	50	42	y	n/a	STANTHORPE	n/a	kitten
7	795	m	1	2	n	ginger & white	6.40	62	46.3	62	48	y	n/a	STANTHORPE	n/a	kitten
8	550	f	1	2	n	black tortoiseshell	2.80	54	42.5	54	43	y	n/a	STANTHORPE	n/a	kitten
9	1700	m	1	2	n	cinnamon	34.78	83	64	87	58	y	n/a	STANTHORPE	n/a	kitten

10	3290	m	2	2	n	black & white	18.29	101	75	114	66	y	no incisors	STANTHORPE	n/a	cat food
11	1200	f	1	2	n	tortoiseshell	5.38	80	59.5	85	54	y	n/a	STANTHORPE	n/a	kitten, cat food
12	2800	f	2	2	n	ginger/cinnamon	25.15	112	80.5	116	67	y	little wear	STANTHORPE	n/a	surgery, had litter previously, cat food
13	1300	f	1	2	n	black & white	8.92	65	49	69.13	47.54	y	n/a	STANTHORPE	n/a	n/a
14	3900	f	2	3	n	grey & white	91.82	104	75	102	66	y	n/a	STANTHORPE	n/a	pregnant, lots of fat
15	1600	f	1	3	n	grey tabby	24.24	75	52	78	52	y	n/a	STANTHORPE	n/a	n/a
16	1600	f	1	3	n	grey tabby	18.75	70	51	74	51	y	n/a	STANTHORPE	n/a	n/a
17	1750	m	1	3	n	grey tabby	28.27	75	55	82	53	y	n/a	STANTHORPE	n/a	n/a
18	5250	m	2	3	n	tabby	15.31	116	84	120	77	y	n/a	STANTHORPE	n/a	multiple rodents (mice)
19	900	f	1	2	n	black	20.22	42	36	46	42	y	n/a	STANTHORPE	n/a	milk
20	890	m	1	2	n	tabby	15.64	40	33.2	42.5	40	y	n/a	STANTHORPE	n/a	milk
21	800	f	1	2	n	black	-75.24	41	31	42.5	41	n	newborn no molars	STANTHORPE	n/a	no stomach contents, newborn
22	1800	m	1	3	n	tabby DLH	39.20	79	55	76	53.73	n	n/a	STANTHORPE	n/a	Roundworm, TAM SDRC ID: 08, No stomach contents
23	1200	m	1	3	n	black & white	23.75	59	44.5	63	47	y	n/a	STANTHORPE	n/a	TAM ID SDRC: 20

24	900	f	1	2	n	black	27.22	38	31.7	42	39	n	n/a	STANTHORPE	n/a	empty stomach
25	1100	m	1	2	n	black & white	42.65	68	53	71	52	y	n/a	STANTHORPE	n/a	rubber band piece in stomach
26	3700	f	3	2	n	brown tabby	45.27	101	82	115	71	y	no canines or incisors	STANTHORPE	n/a	hair loss, fleas, previous litter/s, reptiles in stomach contents
27	5500	m	2	3	n	grey tabby	23.17	114	88.5	124	83	y	n/a	STANTHORPE	n/a	fleas
28	5500	m	2	3	y	grey & white tabby	13.54	119	82	128	91	y	n/a	STANTHORPE	n/a	surgery
29	5900	m	2	3	n	black	23.10	122	91	126	95	y	n/a	STANTHORPE	scabs	scabs all over coat
30	850	m	1	2	n	cinnamon & white tabby	8.84	40	36	48	40	n	n/a	STANTHORPE	n/a	red liquid in stomach but empty otherwise
31	825	m	1	2	n	ginger & white	7.65	42	39	46	39	n	n/a	STANTHORPE	n/a	n/a
32	2000	f	1	3	n	tabby	18.80	83	64	88	55	n	n/a	WARWICK	n/a	grass only item in stomach, sub-adult
33	2050	f	1	3	n	tabby	33.54	81	66	86	56	n	n/a	WARWICK	n/a	sub-adult
34	2200	f	1	2	n	white & ginger	42.51	85	65	92	56	n	n/a	WARWICK	n/a	sub-adult
35	3500	f	2	3	n	tabby	11.86	95	73.5	114	63	n	n/a	STANTHORPE	n/a	pregnant, SDRC TAM ID: 29

36	4600	m	2	3	n	tabby	9.24	117	78	129	75	y	n/a	WARWICK	n/a	grass only item in stomach cat food
37	4000	m	2	3	n	tabby & white	32.91	118	87.5	124	72	y	broken left canine	WARWICK	n/a	grass, invertebrate, pregnant rodent in stomach
38	3800	m	2	3	n	black & white	13.31	118	65	116	74	n	rotted & missing teeth	WARWICK	half tail missing, vertebrae visible but healed	If using nose-tail measurement, keep in mind, tail half missing
39	4750	m	2	3	n	ginger & white	33.48	113	87	124	76	y	n/a	WARWICK	n/a	red stomach
40	4700	m	2	2	y	tabby & white	22.55	131	92	132	76	y	n/a	ALLORA	n/a	Fleas, ear tattoo and neutered-previously owned, rubber in stomach
41	4900	m	2	3	n	ginger/cinnamon	#VALUE!	112	86.5	119	71	y	n/a	STANTHORPE	n/a	lots of fat under skin, ripped both kidneys during dissection hence no data

42	1550	m	1	3	n	black	17.04	66	53	75	52	n	n/a	WARWICK	n/a	n/a
43	1800	f	1	3	n	black	25.05	72	59.5	80	54	y	n/a	WARWICK	n/a	n/a
44	1200	f	1	1	n	tabby	10.11	69	54.5	70	49	y	n/a	STANTHORPE	n/a	grass in stomach
45	1050	m	1	2	n	black	14.89	54	45	61	44	y	n/a	STANTHORPE	n/a	n/a
46	3750	m	2	3	n	tabby	19.68	103	84	117	65	n	n/a	STANTHORPE	n/a	Bandage on arm, lots of fat and muscle
47	850	m	1	1	n	tabby & white	-74.68	45	39	53	42	n	n/a	STANTHORPE	n/a	Liquid stomach contents only, no fat on kidney
48	3400	f	2	3	n	grey tabby	31.49	100	n/a	109	59	y	n/a	STANTHORPE	Half tail missing	No measure for n-t as tail partially missing, some hair in stomach contents, lactating
49	2520	f	2	2	n	tabby	37.54	104	68	106	57	n	n/a	STANTHORPE	n/a	Liquid stomach contents only
50	2450	f	2	2	n	brown & black	18.77	89	65	91	58	y	n/a	STANTHORPE	n/a	mince or cat food?

51	2500	f	2	3	n	brown tabby	33.77	94	66	101	61	n	n/a	WARWICK	n/a	External parasites
52	2500	f	2	1	n	black & white	8.81	107	76	111	64	y	rotted & missing teeth	STANTHORPE	n/a	coat covered in cobbles pegs, external parasites
53	2900	f	2	3	n	brown tabby	42.42	98	76	103	56	n	n/a	STANTHORPE	n/a	TAM ID SDRC: 28
54	3500	m	2	3	n	tabby & white	15.40	101	75	104	70	n	n/a	STANTHORPE	n/a	n/a
55	3700	f	2	2	n	tabby	19.86	110	83	116	66	y	n/a	STANTHORPE	n/a	Stomach huge, full of cat food?
56	4200	f	2	3	n	ginger tabby	29.71	113	82	106	63	y	n/a	STANTHORPE	n/a	Previous litter, fleas, cat food, stomach 140mm
57	4500	m	2	4	n	ginger tabby	38.42	113	74	110	64	n	n/a	STANTHORPE	n/a	Internal parasite
58	3600	f	2	2	y	tortoiseshell	46.71	102	85	110	68	y	n/a	WARWICK	Internal bleeding	Ear tattoo, internal bleeding, came from other Warwick vet, possibly stray hit by car?

59	6300	m	2	3	n	black & white	21.96	115	90	129	85	y	n/a	STANTHORPE	broken front claw	fleas
60	2700	f	2	3	n	grey tabby	20.92	95	69	106	57	n	bottom incisors missing, plaque	WARWICK	n/a	elongated nippled-previously pregnant
61	3100	f	2	3	n	white & brown tabby	42.03	98	80	105	64	y	n/a	WARWICK	n/a	almonds and small rope in stomach contents
62	900	m	1	2	n	black & white	22.31	44	35.5	47	43	n	n/a	STANTHORPE	n/a	n/a
63	900	f	1	3	n	black	25.87	45	35.5	47	42	n	n/a	STANTHORPE	n/a	n/a
64	950	f	1	2	n	black	24.26	45	35.5	52	48	n	n/a	STANTHORPE	n/a	n/a
65	5000	m	2	3	n	brown tabby	22.23	112	88	128	78	y	n/a	STANTHORPE	n/a	Vegetables and vegetation in stomach contents
66	2800	m	2	3	n	ginger	31.87	88	70	99	61	y	n/a	STANTHORPE	n/a	small testes, fleas, dirty, little in stomach
67	3300	f	2	4	n	ginger	70.00	105	77	102	63	n	n/a	STANTHORPE	n/a	lots of fat in abdomen
68	5200	m	2	4	n	black & white	47.48	115	75	125	76	y	n/a	STANTHORPE	n/a	cat food?

Appendix G: Food items consumed by SDR unowned cats

Table 18. List of food items categorised by food type consumed by SDR unowned cats.

Food type	Food item
Mammal	*Felis catus*
	Macropod sp.
	Macropus giganteus
	Notamacropus parryi
	Osphranter robustus
	Notamacropus rufogriseus
	Mus musculus
	Oryctolagus cuniculus
	Petaurus breviceps
	Rattus rattus
	Sus scrofa
	Trichosurus vulpecula
	Vulpes vulpes
	Wallabia bicolor
Bird	Unidentified bird species
Reptile	Dragon (possible *Pogona* sp.)
Invertebrate	Coleoptera
	Orthoptera
Vegetation	Unidentified vegetation species
Anthropogenic	Commercial cat food
	Food scraps
	Glass
	Human hair
	Nylon wadding
	Peanuts
	Plastic
	Rope
	Rubber band
	String

Appendix H: Regression plots

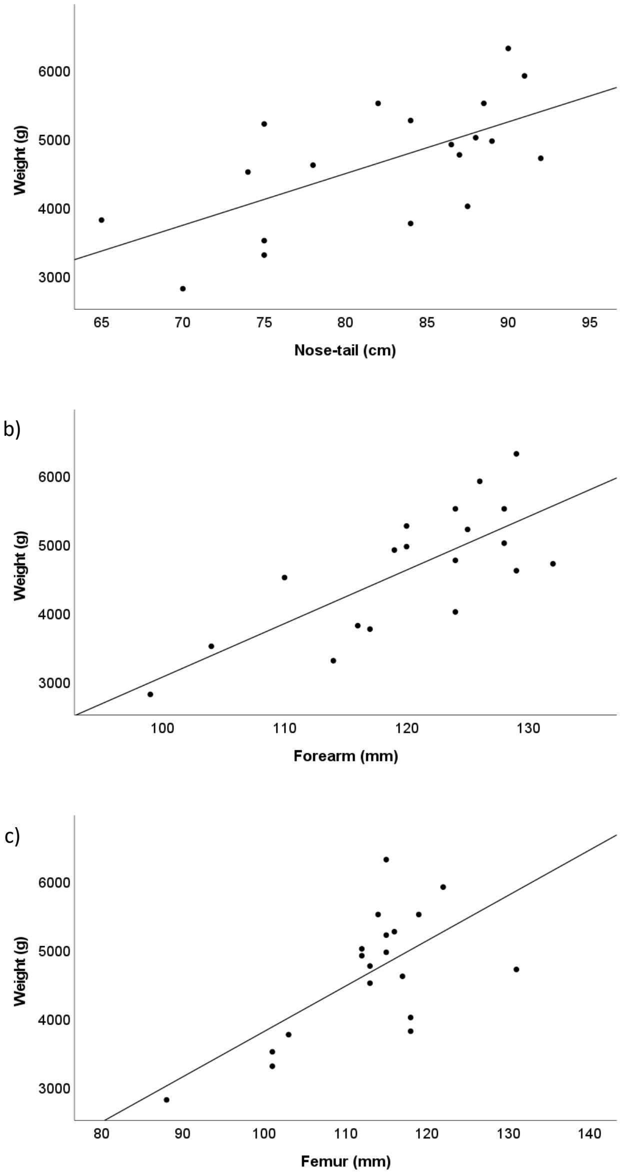

Figure 11. Relationship between weight and nose-tail length in adult male cats. Regression line is described as y = -1540 + 75.26x (a). Relationship between weight and forearm length in adult male cats. Regression line is described as y = -4730 + 77.85x (b). Relationship between weight and femur length in adult male cats. Regression line is described as y = -2120 + 60.09x (c).

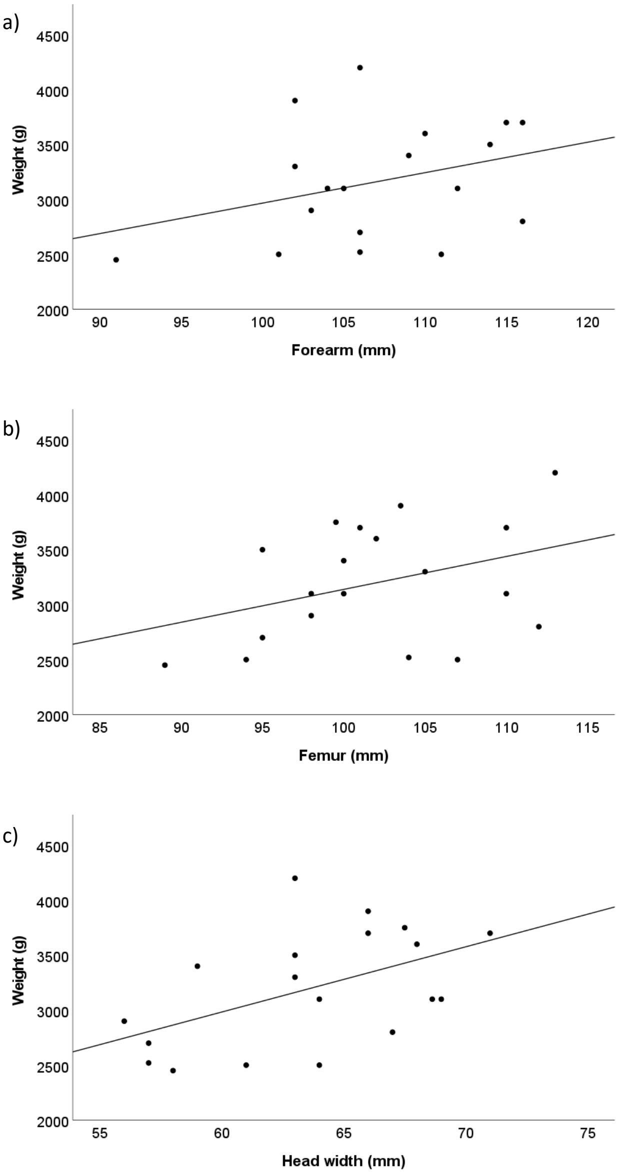

Figure 12. Relationship between weight and forearm length in adult female cats. Regression line is described as y = 193 + 27.74x (a). Relationship between weight and femur length in adult females. Regression line is described as y = 145 + 29.94x (b). Relationship between weight and head width in adult females. Regression line is described as y = -576 + 59.31x (c).

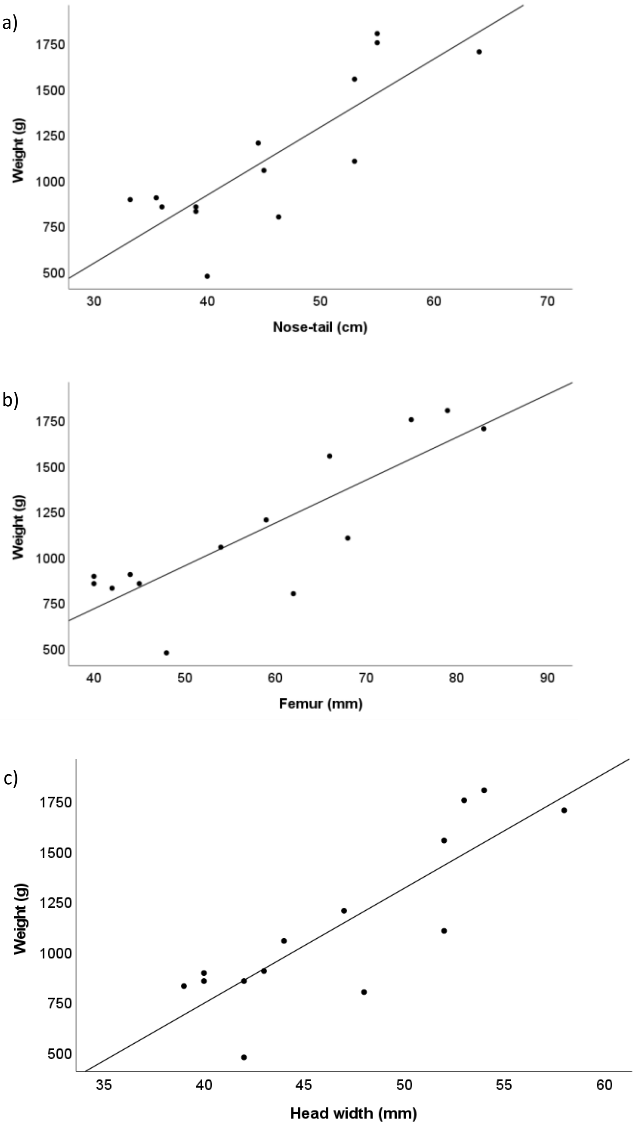

Figure 13. Relationship between weight and nose-tail length in juvenile male cats. Regression line is described as y = 578 + 37.3x (a). Relationship between weight and femur length in juvenile male cats. Regression line is described as y = -229 + 23.52x (b). Relationship between weight and head width in juvenile male cats. Regression line is described as y = -1550 + 57.24x (c).

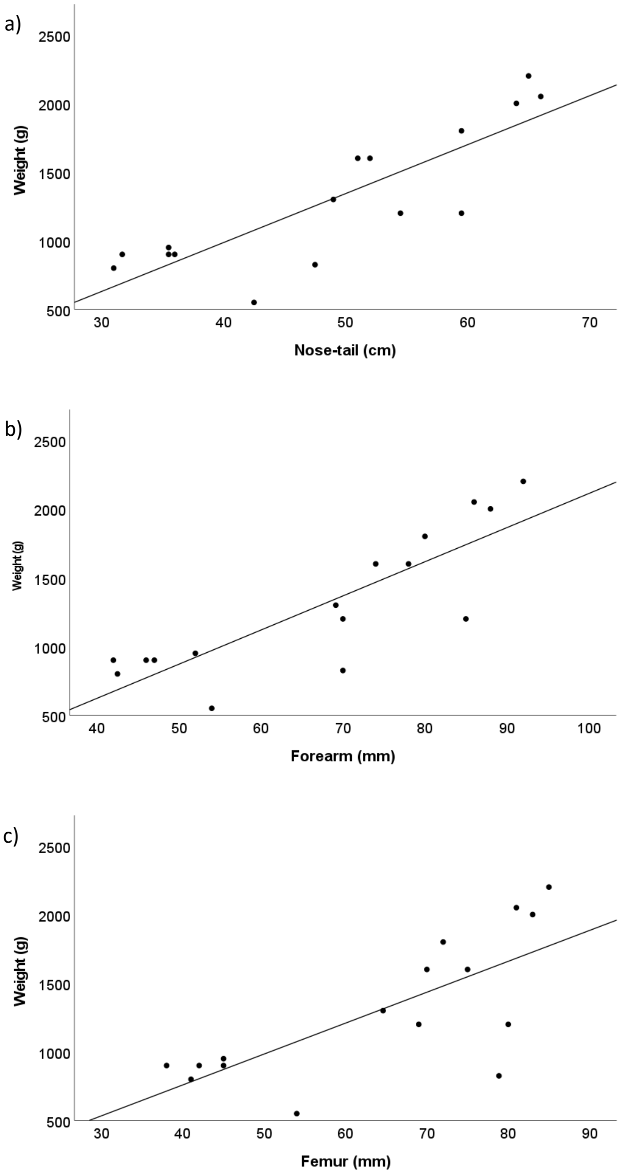

Figure 14. Relationship between weight and nose-tail length in juvenile female cats. Regression line is described as y = -439 + 35.64x (a). Relationship between weight and forearm length in juvenile female cats. Regression line is described as y = -370 + 24.82x (b). Relationship between weight and femur length in juvenile female cats. Regression line is described as y = -141 + 22.51x (c).