UNIVERSITY
of
OTAGO

SAPERE AUDE

Te Whare Wānanga o Otāgo

Does host diet govern the structure and diversity of tapeworm assemblages in sharks? Insights from the literature and a model shark species; *Cephaloscyllium isabellum*

Trent Rasmussen (ID: 6737930)

A thesis submitted for the degree of Masters of Science in Ecology
University of Otago, Dunedin, New Zealand

Supervisors: Haseeb Randhawa and Steve Wing

Abstract

Previous research has shed some light on what phylogenetic and ecological factors may be important determinants of tapeworm parasite diversity in elasmobranchs (sharks, skates and rays). However, several potentially key factors for tapeworm transmission, including the breadth and composition of host's diets, have been recognised as crucial gaps in our understanding.

The main objective of this research was to investigate the relative importance of sharks' diets for the structure and diversity of their tapeworm assemblages. First, the literature was searched for information on tapeworms and host features for a large subset of different shark species, and aspects of shark's diets (including their diet breadth, diet composition and trophic level) were assessed for their relative importance as predictors of tapeworm diversity. Second, literature records were used to conduct a comprehensive analysis of the relationship between shark's diet compositions and tapeworm compositions. Finally, the importance of host diet was examined as a potential encounter filter for restricting tapeworm diversity in a model shark species, Cephaloscyllium isabellum.

The results of this study revealed diet breadth to be a key predictor of tapeworm richness in sharks, indicating that sharks with broader diets generally harbour more tapeworm species. The composition of tapeworms infecting a shark species was found to be related to its diet composition, and moreover, certain tapeworm taxa were found to be useful indicators of the host species' ecology and evolutionary history. The research on C. isabellum here offered only limited insights into the potential importance of diet as an encounter filter for the shark, but provided some new important data on both the diet and parasites of this species.

Ultimately, the observational studies carried out within this research emphasise that aspects of sharks' diets can have important implications for their tapeworm parasite assemblages. Further exploration of these patterns with experimental research may be able to validate the influence of these patterns in nature.

Acknowledgements

I would firstly like to thank the University for selecting me as a candidate for an MSc Research Year Scholarship, which supported me throughout this project. To my supervisors Haseeb Randhawa and Steve Wing, I cannot thank you enough for providing me with your critical support and feedback. Overall, I could not have hoped for better mentors. Thanks to Haseeb for his constant guidance and advice all the way from the inception of ideas for this research to the analyses and final stages of writing. Thanks to Steve for his advice and ideas, especially on the scientific methods for data collection, and for his support in overseeing the logistics for conducting this research.

I would like to say a big thank you to the staff in the Marine Science and Botany departments for helping me to setup and carry out the practical parts of this research. Thanks to Doug Mackie, Daryl Coup and Sean Heseltine for their help in organising shark prey collections, and to Peter Batson and Adelle O'Neill for their assistance in collection trips and setup of my laboratory workspace. In addition, thanks to Gavin Heineman (Echo F/V) for his help in collections of sharks and some invertebrates. I am grateful for Vickey Tomlinson of the Botany department, for helping to provide me with the needed workspace and resources for some of the practical work, and for Michelle McKinlay (Botany) and Christoph Matthaei (Zoology) for their guidance on the logistics of completing and submitting this project.

Thank you to my colleagues and professors who got involved with this research throughout various stages. I would like to thank Keith Probert for his advice regarding some of the identifications of collected invertebrate species and my fellow students Thibaut Anglade, Steph Bennington and Zuri Burns who helped me with some of the practical work. I would also like to extend my thanks to Robert Poulin and various members of the Evolutionary and Ecological Parasitology Group for their valuable discussions and insights regarding some of the ideas and methods used in this project.

Finally, I would like to thank my partner Ashley, and my friends and family for being there for me during each stage of this project. My long and arduous journey towards completing this thesis was made possible thanks to their continued support and encouragement.

Table of Contents

List of figures

List of tables

Chapter 1: Introduction

1.1. General introduction

Parasites comprise an extremely diverse group of organisms and account for a large portion of our total global biodiversity (Dobson et al. 2008; Poulin & Morand 2014). Traditionally, parasites have been viewed as a massive burden to our environment. We have sought, and in many cases, we have succeeded in their eradication (see Bowman 2006). Nevertheless, many ecological studies have shown that parasites are a critical part of ecosystems, serving an important role in the regulation of food webs and host populations, and mediating energy flow through trophic levels (Lafferty et al. 2006; Lafferty et al. 2008; Amundsen et al. 2009). Recent notions in parasite ecology have also highlighted the potential consequences of parasite loss from our ecosystems, indicating that the influence of parasites on overall communities and ecosystems may often be underestimated (Holt 2010; Wood & Johnson 2015). With these points in mind, there is a clear need for further research to better describe the parasite diversity in our ecosystems, and likewise, to determine what factors govern parasite diversity.

Determining what host features influence parasite species richness has been the subject of a broad range of ecological studies, conducted in different ecosystems and animal groups. A recent meta-analysis looking at parasite richness across animal, plant and fungal hosts found that three features of hosts, namely body size, population density and geographical range, could be generally considered as universal predictors of parasite species richness (Kamiya et al. 2014). However, notwithstanding the general significance of these features, their relative influence on parasite diversity can vary considerably among different studies, and in many cases, other less prominent variables may be better predictors of parasite richness among host species e.g. temperature (Poulin & Rohde 1997), anthropogenic changes to land (Mitchell et al. 2010), diet breadth and vulnerability of hosts to predators (Locke et al. 2014). The significance, direction and strength of predictors may largely depend on what type of hosts and parasites are involved (Lindenfors et al. 2007; Poulin & Morand 2014). It is also worth noting that several host features with potentially large impacts on parasite diversity are seldom included in studies looking at determinants of parasite diversity. For instance, it is difficult to evaluate the predictive strength of host diet, home range size or metabolic rate on parasite richness, because they have rarely been taken in account in comparative studies (Kamiya et al. 2014). To gauge the generality of these factors, more emphasis must be put on their inclusion in research going forward.

Sharks are important apex predators in all of the world's oceans and are distributed across a broad range of depths, latitudes and habitats (Froese & Pauly 2017). Their exploitation over the past few decades has left many sharks vulnerable and endangered, producing unpredictable and ecosystem-wide consequences (Stevens et al. 2000; Ward & Myers 2005, Myers et al. 2007; Ferretti et al. 2010). Sharks present an important system for studying determinants of parasite diversity, due not only to their importance as apex predators and integral role in food webs, but also because their associated tapeworms are an extremely diverse and significant group of parasites (Caira & Healy 2004). These parasites may offer key insights from an evolutionary perspective given that elasmobranchs have been hosts of tapeworms for an estimated 270 million years, and they represent the earliest fossil record of tapeworm parasitism of vertebrates (Dentzien-Dias et al. 2013). Previous records show there are about one thousand tapeworm species so far described from elasmobranchs (Caira & Jensen 2014), with thousands more to be described (Randhawa & Poulin 2010). These parasites hold additional importance in marine ecosystems in that they are all trophically transmitted through food webs and affect an array of other marine species as larvae (Caira & Jensen 2017). Hundreds of intermediate hosts have already been described for elasmobranch tapeworms, including various species of teleost fishes, cephalopods, crustaceans and reptiles (Palm 2004; Caira & Jensen 2017).

Previous research has begun to shed some light on what phylogenetic and ecological factors may be important determinants of tapeworm infections in elasmobranchs (McVicar & Fletcher 1970; Randhawa et al. 2007; Randhawa & Burt 2008; Randhawa & Poulin 2010). A recent study found that host size, latitude and depth may each influence tapeworm diversity in elasmobranchs (Randhawa & Poulin 2010). However, the influence of these factors clearly depends on the type of elasmobranch host involved (shark or batoid), and after correcting for phylogenetic influences in sharks, only host size has been demonstrated to significantly impact tapeworm richness (Randhawa & Poulin 2010). Notably, there are still many factors that remain to be comparatively tested for their influence on tapeworm diversity. It has been pointed out that host diet breadth and composition especially warrant further investigation (Randhawa & Poulin 2010).

The aim of this thesis is to assess what factors govern the structure and diversity of tapeworm parasite assemblages in sharks, with particular reference to the importance of host diet. Two main approaches will be used to achieve this. The first will be to look at records from the literature across many shark species and their prey to determine whether aspects of host diet are strong predictors of tapeworm diversity and composition in comparison with other relevant host features. This approach will serve to tell us whether factors such as diet breadth, diet composition and trophic level appear to be generally important barriers for tapeworm establishment in sharks. The second approach will complement the first approach by more specifically analysing the importance of diet for parasite composition in a local shark species, the draughtsboard shark Cephaloscyllium isabellum. This species could be a good model for investigating what factors restrict parasite establishment in sharks because it is known to have a broad diet (Horn 2016), but has fewer tapeworm species than would be expected for a shark of its size (Randhawa 2014, unpublished data; see also Poulin et al. 2011b). Thus, it can be investigated whether its diet is a large factor restricting encounters with different tapeworm parasites, or alternatively, whether strong compatibility filters in the species prevent parasite associations. Notably, this investigation could also provide valuable biological information on the shark, which is currently scarce (Horn 2016).

1.2. Host specificity and barriers to parasite establishment

As described in the above introduction, testing what factors predict parasite richness among host species can help us to understand patterns of parasite biodiversity. For a complementary approach to understanding these patterns of diversity, we can look at a trait known as "host specificity" and analyse what factors act as barriers to infection for certain parasites. Host specificity is a fundamental property of parasites (Kosoy et al. 1997; Dyer et al. 2007; Poulin 2011). Although there are many definitions for this property (see Poulin & Mouillot 2005; Poulin et al. 2011a), it can generally be defined as the extent to which different host species are used by a parasite (Combes 2001). Host specificity exhibits great variation among different species and groups of parasites. At one end of the spectrum, we have parasites which infect only one species. Most monogenean ecto-parasites of fish, for example, are restricted to a single host species (Ŝimková et al. 2006). On the other end, some parasites infect a multitude of different hosts. For instance, the Asian tapeworm Bothriocephalus acheilognathi has been described from more than a hundred different fish species (Salgado Maldonado & Pineda-López 2003).

Understanding what ecological and evolutionary factors determine parasite-host specificity is a common goal for research in parasitology. On the theoretical side, knowing what factors prevent a parasite from expanding its host range is key to learning about the community structure of parasites and host-parasite co-evolution (Poulin et al. 2011a). On the practical side, knowing what ecological and evolutionary factors underpin host specificity may guide scientists towards better prediction and control of the transmission of infectious diseases (Poulin 1992; Taraschewski 2006; Lootvoet et al. 2013). Of course, the ability of a parasite to successfully spread and establish in a new area largely depends on what ecological or phylogenetic barriers may prevent the parasite from establishing and spreading to alternative hosts (Taraschewski 2006; Dunn 2009).

Euzet and Combes' filter paradigm offers a simple but effective illustration of what factors determine the host specificity of a parasite (Euzet & Combes 1980). The idea is that there is a two-step filter for parasite-host compatibility. The first step is an 'encounter filter', which excludes hosts from a parasite's potential host spectrum that don't come in physical contact with the parasite (illustrated by circles in Figure 1.1). A host may not be encountered by a parasite either because they live in different ecosystems, or alternatively, because host behaviours such as diet and niche segregation prevent contact with infective stages of the parasite (Combes 2001). The second step of the filter paradigm is a 'compatibility filter', which excludes any hosts from a parasite's potential host spectrum that are incompatible with the parasite (illustrated by triangular section in Figure 1.1). Hosts may be incompatible either because they do not provide adequate spatial or metabolic resources for the parasite, or because immunological or other defense mechanisms of the host kill the parasite (Combes 2001). Upon consideration of each of these filters, it is clear that the range of hosts actually used by a parasite represents only a small subset of their potential host species (small shaded triangular area in Figure 1.1).

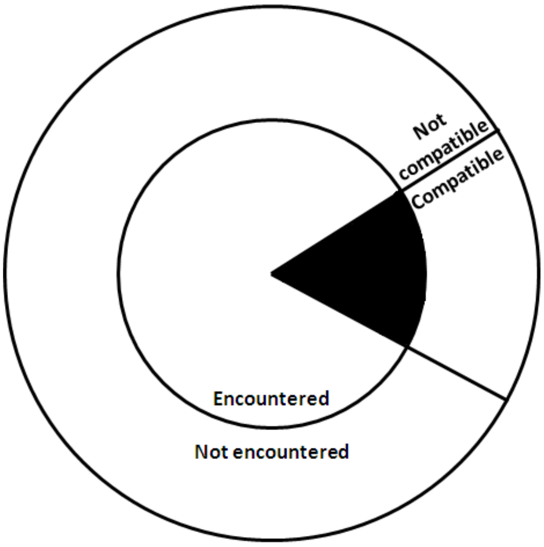

Figure 1.1: Diagram illustrating the filter paradigm of host-parasite specificity. (adapted from Combes 2001). A parasite's host spectrum (range of hosts it uses) represents a small subset of its potential host species (small shaded area) that are encountered (within the small inner circle) as well as compatible (within the triangular area).

The compatibility filter concept has been applied in research over the past few decades as a basic tool for understanding determinants of host specificity (Kuris et al. 2007; Randhawa & Burt 2008; Lagrue et al. 2011). A handful of studies have shown that the high specificity of tapeworms in elasmobranchs is, to some extent, the result of compatibility filters. For example, immune response in elasmobranchs has been demonstrated to cause mortality for certain tapeworm species (McVicar & Fletcher 1970; Randhawa & Burt 2008). Attachment site morphology in elasmobranch host mucosa may also determine whether some tapeworm species can live in the host (Williams 1960, 1966, 1968). Although it has been suggested that attachment site morphology is more of a determinant for attachment site specificity rather than for host specificity (Randhawa and Burt 2008). These studies clearly demonstrate that compatibility filters have an influence in restricting the host specificity of elasmobranch tapeworms. However, the role of these compatibility filters in comparison with encounter filters is unclear.

It is unknown whether encounter filters (e.g. diet) are as restrictive for tapeworm establishment in elasmobranchs as parts of the compatibility filters (e.g. immune response). This is an important gap in knowledge since some encounter filters, such as host substrate preference and diet, have been suggested as potentially important ecological determinants of host specificity (Randhawa et al. 2008). It is likely that both filters play key roles as barriers to parasite establishment, but by assessing the relative importance of each filter we may be able to better predict the consequences of ecological changes on the transmission of these parasites to other hosts in future.

1.3. The influence of host diet on parasite diversity

Helminth parasites with complex life cycles rely on trophic interactions for transmission to their final host (Simkova et al. 2001; Cirtwill et al. 2016).
Thus, the diversity of these parasites is unlikely to be randomly distributed in food webs, but is rather expected to be concentrated in parts of a food web that favour transmission (Locke et al. 2014). Following from this, it would make sense that hosts diets have a large influence on their acquisition of parasites, and consequently, the diversity of their parasite assemblages. Many previous studies support this notion, showing that variation in diet breadth and trophic positions among host species can explain a large amount of interspecific variation in the richness of their trophically-transmitted parasites (Klimpel et al. 2006; Chen et al., 2008; Poulin & Leung, 2011; Locke et al., 2014).

Diet breadth could be one of the most important factors in determining the diversity of fish parasites. In theory, a fish species that feeds on many different prey species should be exposed to a greater range of larval parasites compared to a host species with a restricted or specialised diet. Because of this, fish species with broad diets could accumulate a larger variety of trophically transmitted adult parasites (Kennedy et al. 1986; Lo et al. 1998; Locke et al. 2014). The trophic level of a host might be similarly important, since fish occupying higher trophic positions have access to more prey, and parasites tend to exploit host species that are highly connected (Chen et al. 2008). The diet composition of a host may also play a large role in its accumulation of different parasites. Trophically transmitted parasites typically infect a limited number of intermediate hosts, meaning that the variety of parasites in the final host may depend on whether these intermediate hosts are an important component of the definitive host's diet (Kennedy et al. 1986; Marques et al. 2011).

The influence of dietary factors on the diversity of tapeworms infecting sharks is currently unknown (Randhawa & Poulin 2010). There is good reason, however, to think that diet may be an important factor for tapeworms in elasmobranchs, since all tapeworm species are acquired by elasmobranchs via trophic transmission from intermediate or paratenic hosts (Willams 2002).

1.4. Life cycles of shark tapeworms

From what is currently known of tapeworms infecting sharks, the life cycles of these parasites are typically complex and involve several invertebrate and vertebrate intermediate hosts (Sakanari & Moser 1989; Palm 2004; Randhawa 2011; Caira & Jensen 2017). In general, these parasites are highly host specific as adults (i.e. are restricted to one or a few host species), but show considerably lower host specificity in intermediate hosts (Palm & Caira 2008; Jensen & Bullard 2010). There also appears to be considerable variation in life cycles among different tapeworm species, where some species use many more intermediate and paratenic hosts than others (Palm 2004).

To get an idea of a general shark tapeworm life cycle, we can look at one of the earliest described examples involving the trypanorhynch tapeworm Lacistorhynchus dollfusi infecting the leopard shark Triakis semifasciata. Adult tapeworms live in the spiral intestine of its definitive elasmobranch host (Figure 1.2a), and once mature, pass eggs out through the shark's faeces (Figure 1.2b-c). These eggs are then consumed by a first intermediate host, which is often a small crustacean such as a copepod or amphipod (Palm 2004) (Figure 1.2d). The first intermediate host is consumed by a second, larger intermediate host, such as a teleost fish, where the tapeworm develops from a procercoid to a plerocercoid larva (Figure 1.2e). The tapeworm then completes its life cycle once the shark host eats the plerocercoid-infected fish (Figure 1.2). Life cycles of tapeworms can be more complex than the one described above, and prey other than crustaceans and fish, including reptiles, mammals and cephalopods, have also been identified as intermediate hosts (Palm

2004; Randhawa 2011; Caira & Jensen 2017). Moreover, there are variations in life cycles where the second intermediate host is not a fish, and is instead another larger invertebrate like a shrimp or crab (Palm 2004).

An overwhelming majority of the life cycles of elasmobranch tapeworms are still unknown or poorly described (Caira & Reyda, 2005; Jensen & Bullard 2010). The main reason for this is the difficulty of identifying larval stages, since most tapeworms have larvae that don't look like the adult based on morphology (trypanorhynchs are an exception) (Jensen & Bullard 2010). This difficulty has sparked the need for alternative approaches to identify tapeworm larvae, including molecular tools (Poulin & Keeney 2008; Jensen & Bullard 2010; Randhawa 2011) and in-vitro growth (e.g. Presswell et al. 2012). These alternatives to morphological identification could be pivotal for researchers aiming to better understand the ecology and evolution of tapeworm life cycles in future (Palm & Caira 2008).

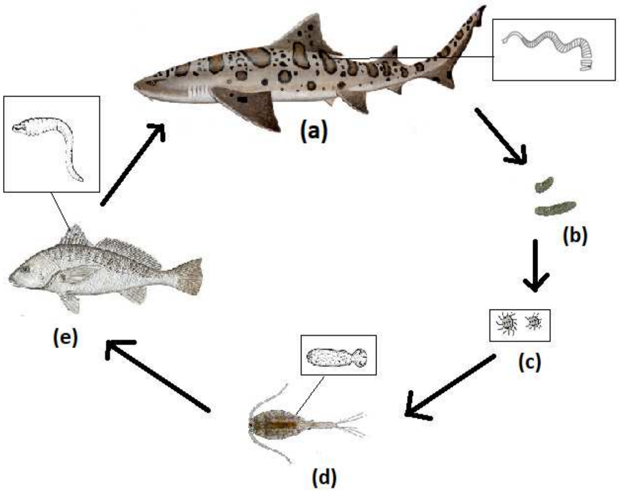

Figure 1.2: Diagram illustrating the life cycle of a shark trypanorhynch tapeworm: *Lacistorhynchus dollfusi* infecting the leopard shark *Triakis semifasciata* (Modified from Sakanari & Moser (1989)). (a) Adult tapeworms live in the spiral valve of the definitive shark host. (b) gravid proglottids pass out in shark faeces, releasing eggs. (c) Ciliated coracidia hatch from operculated eggs and are consumed by copepods (d), where they develop into procercoid larvae. Copepods are eaten by teleosts such as white croakers (e), and the procercoids develop into plerocerci inside blastocysts. When infected fish is consumed by the shark host, the tapeworm lifecycle is completed.

1.5. Biology of the model species: *Cephaloscyllium isabellum*

The draughtsboard shark Cephaloscyllium Isabellum (Bonnaterre, 1788) is a scyliorhinid catshark species, of the Order Carcharhiniformes, that is thought to be endemic to New Zealand (Cox & Francis 1997) (Figure 1.3). This species is sometimes referred to as the New Zealand carpet shark, but it is not a member of the carpet shark Order Orectolobiformes. To avoid confusion, the species will be hereafter referred to as the draughtsboard shark or C. isabellum.

Figure 1.3: Image of the New Zealand draughtsboard shark (*Cephaloscyllium isabellum).* Image credit: Chris 2012*,* http://www.surfcaster.co.nz/

Like other members of the Cephaloscyllium genus, C. isabellum is oviparous (Dulvy & Reynolds 1997), and is it thought to have year-round reproduction (Horn 2016). Individuals of this species generally range from 20cm in length as juveniles up to a metre in length as adults, with male sharks generally not growing as large as females (Cox & Francis 1997; Horn 2016). Draughtsboard sharks live around New Zealand coasts in shallow depths out to around 200 metres, but also occur (mostly as larger individuals) in deeper waters out to 500 metres (Francis et al. 2002; Horn 2016). They are known to be demersal hunter and scavenger, and feed on a variety of prey including fishes, crustaceans, molluscs and other invertebrates (Horn 2016). Cephaloscyllium species can detect weak bioelectric fields of prey (Tricas 1982), which likely assists them in their ambush predation and scavenging. Like other species of this genus, C. isabellum is thought to forage mainly at night, whilst taking refuge in caves or reefs during the day (Nelson & Johnson, 1970; Awruch et al. 2012; Horn 2016).

Several endoparasite species have been described in C. isabellum; two digenean trematodes (in the stomach), three nematodes (in the stomach and sometimes intestine), and one tapeworm species, Calyptrobothrium chalarosomum (in the spiral intestine) (Hewitt & Hine 1972; Hine et al. 2000). Cephaloscyllium isabellum has a notably low diversity of intestinal tapeworms (one) compared to other sharks, which on average, are infected by around 6 different tapeworm species (Randhawa and Poulin 2010). This shark species has also been identified as a cold spot in shark tapeworm diversity, which means that it has fewer tapeworm species than would be expected for a shark of its size (Poulin et al. 2011b). This is surprising given that the species demonstrates a broad diet (Horn 2016), and it could potentially encounter many tapeworm larvae in different prey. It is unknown, however, whether many different tapeworm larvae are present in its favoured prey. The above points indicate that C. isabellum should be a useful model for looking at determinants of tapeworm host specificity. Its diet may be a large factor restricting encounters with different tapeworm parasites, or alternatively, there are perhaps strong compatibility filters in the species preventing parasite establishment. This poses a key question: Which filter is more restrictive of parasite establishment in C. isabellum?

1.6. Study location

All samples for this research were obtained between the Otago Peninsula region of Dunedin, and Curio Bay in Southland, New Zealand (Figure 1.4). Draughtsboard sharks and their prey were sampled from between Curio Bay and Taiaroa Head, Dunedin. Lab work, including most measurements and dissections, was conducted at Portobello Marine Lab, which is in close proximity to the sampling sites (Figure 1.4). However, some practical work, primarily the identification of shark prey items, also took place in the University of Otago Botany Department.

Figure 1.4: Map of the Otago region displaying the sampling site locations; Curio Bay to Nugget Point to Taiaroa Head, and the sites for lab work; Portobello Marine Lab and the University of Otago Botany Department.

1.7. Aims, objectives and hypotheses

The overall aim of this research was to provide insights into what factors govern the composition and diversity of tapeworm parasites in sharks, with particular reference to the influence of host diet. This aim was explored on a large scale by using published literature to conduct comparative analyses across a broad range of different shark species. In addition, this aim was examined on a smaller scale, by testing links between the diet and tapeworms of a model shark species (C. isabellum).

The objective of the comparative analyses was to determine whether certain aspects of host diet (such as diet breadth, composition or trophic level) are important predictors of tapeworm diversity or composition, particularly in comparison with other host variables. Additionally, larval shark tapeworm records in the literature were investigated to ascertain whether intermediate hosts of shark tapeworms show strong links with the diet of their shark hosts. These objectives were carried out by compiling comprehensive datasets on diet and other features of shark species from the literature and testing the variables of interest for their predictive strength on measures of tapeworm diversity among host species (e.g. tapeworm richness).

The objective of research on the model species Cephaloscyllium isabellum was to find out what factors act to restrict parasite establishment in the species, something which may also be relevant for other sharks. Moreover, this part of the study attempted to elucidate parts of the life cycle of the shark's only known tapeworm (Calyptrobothrium chalarosomum). To complete these objectives, a number of individual sharks were examined for their diets and parasite fauna, and their known prey items were surveyed for parasite larvae to determine their potential as intermediate hosts. This was done to give some indication of how many larval parasites are likely to be encountered via their diet, and thus, whether there are mechanisms other than diet (e.g. host immune response) that heavily restrict parasite establishment.

1.8. Overview of thesis chapters

This thesis is presented in five chapters; a general introduction (this chapter), three chapters devoted to three individual studies, and a general discussion chapter.

Chapter two investigated aspects of host diet as determinants of tapeworm diversity in sharks. Data was obtained from the literature on the diet breadth, trophic level and diet composition of as many shark species as possible in order to test whether these variables are strong predictors of tapeworm diversity. The importance of these factors were also assessed relative to many other potentially significant host features (e.g. host size and phylogeny).

Chapter three examined the influence of diet on the composition of shark tapeworm assemblages. Similarly to chapter two, this involved compiling data from the literature on the diet composition of different sharks. However, this chapter analysed what factors may shape shark tapeworm compositions rather than analysing which factors are important for general tapeworm diversity. This chapter also examined larval shark tapeworm records in the literature to determine whether known intermediate hosts of shark tapeworms show a strong presence in the diets of their definitive shark hosts.

Chapter four focused on the model species C. isabellum, and primarily investigated whether this shark's diet is an important factor restricting its encounters with potential tapeworm parasites. This involved a look at both the parasites and diet of this shark species, and an examination of larval parasites in its known prey items, which could serve as potential intermediate hosts.

The final chapter summarised key findings from all of these thesis chapters, discussed their implications, and provided suggestions for further research.

Chapter 2: Host diet as a determinant of tapeworm diversity in sharks

2.1. Introduction

The diversity of parasite species in a host, like the diversity of free-living species in an ecosystem, is shaped by a range of different ecological and evolutionary features. Exploring how these features relate to parasite diversity is fundamental to our understanding of why particular host species evolve with richer or more diverse parasite faunas than others (Poulin 2004). Research over the past several decades has already identified several widely important host features that can influence the diversity of parasite assemblages, including body size, lifespan, population density, geographical range and diet (Morand et al. 2000; Vitone et al. 2004; Kamiya et al. 2014). A few of these features; body size, population density and geographical range, have even been recognised as ‘universal’ determinants of parasite species richness (Kamiya et al. 2014). Despite the general significance of these few host features, however, their relative importance is known to vary considerably among different host-parasite systems (e.g. Poulin et al. 2011). Depending on what hosts and parasites are involved, other less-generalised factors may also have a large influence on parasite diversity. For instance, in anthropoid primates, parasite species richness is influenced largely by social group size (Vitone et al. 2004). In addition, a number of host features, which could potentially have a significant impact on parasite diversity, are often left out of studies looking at determinants of parasite diversity. For example, host basal metabolic rate (BMR) is potentially a very important factor for parasite diversity. Higher BMRs are associated with higher rates of energy processing and resource availability, and consequently, animals with a high BMR may be able to support richer parasite assemblages (Brown et al. 2004). However, since BMR has been scarcely included as a factor in comparative analyses of parasite diversity, it is difficult to assess its predictive strength and relative importance compared to other factors (Kamiya et al. 2014). Likewise, host diet breadth may be an important predictor of parasite diversity because species with broad diets can encounter a greater range of parasite species from different prey (Locke et al. 2014). Yet, diet breadth has been rarely included in comparative analyses of parasite diversity, making it difficult to know how important diet breadth is for parasite diversity on a larger scale (Kamiya et al. 2014). In light of these issues, there remains a need for research to assess the relative importance of many different host features in various host-parasite systems to better understand emerging diseases and their transmission dynamics.

Sharks and their tapeworm species assemblages provide a useful system for looking at large-scale patterns of parasite diversity for a number of reasons. First, the tapeworm parasites of these elasmobranchs show substantial diversity and exhibit high host specificity. They are the most diverse group of parasites infecting elasmobranchs, with 977 different species and 201 genera known from nine established orders (Caira and Healy 2004; Caira and Jensen 2014). Based on the number of host species sampled for parasites to date, it is also estimated that an astounding 3600 tapeworm species in described elasmobranchs have yet to be recorded (Randhawa and Poulin 2010). Almost all tapeworm species exhibit a high degree of host specificity in sharks, with most being restricted to a single host species or a few closely related hosts (Caira and Jensen 2014). However, it is also worth noting that these tapeworms are generally less specific in intermediate hosts and can have consequences for thousands of marine species other than elasmobranchs, including a broad range of teleost fishes, molluscs, crustaceans, mammals, reptiles and other invertebrates (Palm and Caira 2008; Jensen 2009). Second, sharks are a group of animals that have features with broad and easily measurable variation across species. Shark species display a broad range of sizes, are found at nearly all marine habitats, depths and latitudes, and show marked variation in the breadth of their depth and latitudinal distributions (Froese and Pauly 2015). There is also considerable variation in diet and trophic level among different species (Cortés 1999). Third and finally, sharks are apex predators of marine ecosystems and they, along with their many parasites, exert considerable influences throughout marine food-webs. Thus, sharks and their parasites stand as an important model for understanding determinants of marine diversity.

From previous research looking at tapeworm diversity in sharks, several host features, including latitude, depth, and habitat, appear to have little influence on their parasite diversity (Randhawa and Poulin 2010). Host size has been identified as a significant predictor, but no other ecological or host biological features examined so far have demonstrated significant effects on shark tapeworm diversity (Randhawa and Poulin 2010). Additionally, it has been shown that shared host evolutionary history, i.e. shark phylogeny, outweighs ecological variables as a predictor of tapeworm diversity in sharks (Poulin et al. 2011). Nevertheless, there are a number of potentially important host features that have yet to be examined for their influence on tapeworm diversity. Host population density is a feature of likely importance, but data on population density is generally unavailable for different shark species, making it difficult to investigate its influence on parasite diversity. Of the features for which data are available, aspects of host diet are arguably the prominent features warranting investigation. Host diet hasn't previously been assessed as a determinant of shark tapeworm diversity, but it is likely to have a large impact considering that all tapeworm species in elasmobranchs are acquired via ingestion of infected prey (which are intermediate or paratenic hosts) (Williams et al. 1994). Essentially, the number of tapeworm species that infect a shark species should directly depend on what prey, and ultimately how many different prey, a shark consumes as part of its regular diet.

2.1.1. Objectives of chapter 2

The main aim of this chapter was to examine whether three major aspects of host diet; (1) breadth of diet, (2) trophic level, and (3) diet composition, influence the species diversity of tapeworm assemblages in sharks. Although all three of these factors convey information on the diet of sharks, each is distinctly different in what it tells us. Breadth of diet is a general measure of how many different prey are consumed by a shark species. It was hypothesised that shark species with broader diets (diets including more different prey taxa) would harbour a greater diversity of tapeworm species than those with restricted diets. This was based on the fact that they would encounter more tapeworm intermediate hosts through their diet, and thus, could come in contact with a greater diversity of parasites. Trophic level is a measure of a species' position in food webs, and indicates overall what type of ecological groups are most important in their diet (see Cortés 1999). It was hypothesised that shark species occupying higher trophic levels would harbour more diverse tapeworm assemblages than those occupying lower trophic levels, since they have access to more trophic links and additional intermediate hosts from higher trophic levels. Diet composition is a measure of what specific taxa are the most dominant in the diet of a host species. In contrast to trophic level, which reflects the position of a shark's prey in the food web, diet composition tells us which specific taxonomic groups of prey (e.g. teleosts, cephalopods, crustaceans) comprise most of the diet. I predicted that shark species feeding predominantly on teleost fishes would have more tapeworm species than other groups because most currently described intermediate hosts for marine tapeworms are teleost fishes (Palm 2004; Jensen 2009). Overall, it was predicted that each of these measures of host diet would have more significant, higher magnitude impacts on tapeworm diversity than host features examined in previous studies.

2.2. Methods

2.2.1. Tapeworm data collection

All tapeworm species diversity data used in this study were compiled by revising and updating a comprehensive elasmobranch tapeworm dataset made available by Randhawa and Poulin (2010). This original data set obtained tapeworm richness estimates for a large range of shark species by searching through Zoological Records on ISI Web of Knowledge and compiling available data from 1864 to 2008. To ensure that the data set would be accurate and up to date, the original data set was revised by changing tapeworm richness estimates to include all new shark-tapeworm records published in Zoological Records from 2008 to 2017. Notably, a number of shark species were included in the present data set for which tapeworm species records had become available since 2008. Using the same method as Randhawa and Poulin (2010), new tapeworm records for each host species were found by searching the shark taxa (Latin name plus all known synonyms) combined with keywords ‘‘Parasit* OR disease OR pathog*’’.

Since measures of parasite diversity are often greatly influenced by sampling/study effort (Walther et al. 1995), correcting for sampling effort can give a more accurate measure of diversity (Poulin 2004; Luque and Poulin 2007). An ideal measure of parasite diversity sampling effort would be the sum of host individuals examined for parasites, but unfortunately, these data are seldom available from studies describing parasite records. As such, sampling effort for parasite diversity of each host species was measured as the total number of references obtained by searching the host’s tapeworm records on ISI Web of Knowledge (using the search parameters defined above). Compared with other estimates of sampling effort, this measure has been previously demonstrated as a stronger correlate with tapeworm richness (Randhawa and Poulin 2010).

To complement tapeworm species richness as a measure of parasite diversity, the average taxonomic distinctness (TD) of tapeworm assemblages was also calculated for each shark species. This index is used to measure the average taxonomic distance between the parasite species of an assemblage, with greater TD values indicating greater average taxonomic difference between species in the assemblage (Luque et al. 2004). TD is a different measure of diversity to richness in that is thought to be more sensitive to host ecology (Luque et al. 2004, Luque and Poulin 2008). It is measured as the average number of steps up the taxonomic hierarchy (Phylum, Class, Order, Family, Genus, and Species) in order to reach a taxonomic level common to two species, and is calculated for all pairs of species in the assemblage being examined (Warwick and Clarke 2001). Variance in TD was computed to accompany the measure of average TD for each shark species. Variance in TD can be used to provide information on the taxonomic heterogeneity among host species, basically showing how even the distribution of taxa across the taxonomic tree is (Warwick and Clarke 2001). Tapeworm TD was calculated (with the associated variance) for each host species harbouring at least 3 tapeworm species, using the programme ‘Taxobiodiv 1.2’ (available at <www.otago.ac.nz/ zoology/downloads/poulin/ TaxoBiodiv1.2>).

2.2.2. Host features data collection

For all shark species with an available estimate of tapeworm richness, a number of important host features were recorded: (1) diet breadth, measured as the total number of prey families in a shark’s diet; (2) diet TD, measured as the average taxonomic distance between all prey families in a shark’s diet; (3) trophic level, measured as the number of energy-transfer steps to the shark’s food chain position; (4) diet composition, given as the taxonomic prey category composing most of the diet (of nine groups: teleost fishes, cephalopods, crustaceans, mammals, chondrichthyan fishes, reptiles, birds, other molluscs, other invertebrates); (5) habitat, given as which zone of the ocean a shark species is most associated with inhabiting (of seven categories: reef-associated, demersal, pelagic-oceanic, pelagic-neritic, benthopelagic, bathypelagic, bathydemersal); (6) host total length (cm); (7) latitudinal range, measured by the number of degrees of latitude spanning the shark’s geographic distribution; (8) depth range, measured as the distance in metres between shallowest and deepest points at which they occur; (9) depth mid-point, measured as the mid-point of the shark’s preferred depth distribution; and (10) phylogeny, represented by the Genus, Family and Order of the host. It is worth noting that many of these host characteristics (host length, latitudinal range, depth range, depth mid-point, and habitat) have been previously assessed as predictors of tapeworm diversity (Randhawa and Poulin 2010), and were also included in the present data set to determine their relative importance compared with the dietary features of primary interest here.

Data on most host features (phylogeny, trophic level, host length, latitudinal range, depth range, depth mid-point, and habitat) were obtained directly from recent species records listed on FishBase in March 2017 (Froese and Pauly 2017). In cases where data on one or more of these host features were not available for a species, the shark species was excluded from the data set. For all data on host diet, a comprehensive dataset was compiled from ISI Web of Knowledge. To do this, the taxa of each shark was searched (Latin name plus all known synonyms) combined with keywords ‘‘diet* OR feed* OR prey*’’ on ISI Web of Knowledge (all databases) and all available references from 1864 to 2017 were compiled (Searches were conducted April 2017). Every reference listed was searched for information on the diet of sharks, and out of 2,081 references listed across all species, 361 had relevant data on diet that could be included in this study (see references in Table A.1 in Appendix).

Unsurprisingly, the level of taxonomic definition for prey varied among studies; where some sharks had prey recorded mostly to Family or Order level, others had prey items known to the level of species. Considering this potential bias in records, it was decided that the best measure of diet breadth would be at the taxonomic level of Family. At this level there were records for most species that had tapeworm diversity estimates and there was also a large range of taxonomic groups (603 different families of prey) to give a good measure of variability in diet breadth among sharks. To ensure further accuracy in diet breadth comparisons, the final data set was also restricted to only include host species with at least one family of prey recorded (n = 91). Every family of prey identified for each shark species was recorded from each reference. Notably, some diet records were old, and taxonomic changes had been made to certain prey taxa since their publication. To address this, all prey taxa were checked in the World Register of Marine Species (WoRMS) database and updated to their current taxonomical nomenclature (WoRMS Editorial Board 2017).

Similarly to measures of parasite diversity, measures of diet breadth may be substantially affected by sampling effort (Randall and Myers 2001). Diet breadth is likely to increase as more individual hosts are examined for diet contents. Correcting for this influence of sampling effort should therefore provide a better measure of host diet (Cortés 1999). In contrast to studies describing parasites from sharks, the standard protocol for dietary studies is to include the number of hosts examined. Thus, it was possible to measure diet sampling effort as the sum of stomachs containing food (i.e. the number of non-empty stomachs) examined across all diet records for each shark species. For a few references, the number of non-empty stomachs examined was not provided, and therefore had to be excluded from further analyses. All the references were checked thoroughly in the methods and results sections to make sure that diet information was not duplicated among studies. In cases where studies gave duplicate data, the data was cited from the original reference only, and the number of hosts examined was only included for the original reference to avoid overestimation of sampling efforts.

Taxonomic distinctness (TD) of diet was calculated to complement number of prey families as a measure of diet breadth. This was measured as the average number of steps up the taxonomic hierarchy in order to reach a taxonomic level common to two prey families, and was calculated for all prey family pairs in a shark's prey assemblage. Similarly to the TD calculations for tapeworm assemblages, TD of prey family assemblages and associated variance were computed for each shark species with at least 3 families of prey in their diet, using 'Taxobiodiv 1.2'. For information on diet composition, the percentage composition of nine different prey groups was recorded from each reference (teleost fishes, cephalopods, crustaceans, mammals, chondrichthyan fishes, reptiles, birds, other molluscs, other invertebrates). The overall composition of these groups for each shark species was then calculated by taking the average compositions across all studies, weighted by the number of non-empty stomachs examined for diet. Similar to the approach of Cortés (1999), compound indices were used to estimate composition where available (e.g. the index of relative importance %IRI), and otherwise, single indices, such as percent frequency of occurrence (%O), percent number (%N) percent weight (%W), or percent volume (%V) were used individually. Where two of these single indices were available, an average was calculated (e.g. %O + %W/2). Plant materials, detritus and non-organic materials were not included in composition estimates as the present study was only interested in prey which are potential intermediate hosts for shark tapeworms.

2.2.3. Data analysis

All statistical tests were carried out in the R environment (R Development Core Team 2012). Prior to analysing relationships between measures of tapeworm diversity and recorded host features, regressions were run to determine the influence of sampling effort on measures of tapeworm diversity and diet breadth. As expected, for both tapeworm richness and diet breadth, the relationships between diversity and sampling effort were best characterized by positive curves where diversity increased with increasing sampling effort, slowing towards an asymptote at higher effort values. To determine the significance of the associations, simple quadratic regressions were run for each diversity measure on their associated measures of sampling effort. Host-parasite sampling effort was found to have a significant influence on tapeworm species richness ($r^2 = 0.539$, $p < 0.001$), but not on tapeworm TD ($r^2 = 0.007$, $p = 0.825$). To correct for this influence in further analyses, tapeworm richness was from here on measured by residuals for its quadratic regression on host-parasite sampling effort (a plot of this regression can be seen in Appendix Figure A.1.). Diet breadth was also influenced by sampling effort. The number of stomachs sampled for diet showed a strongly positive association with number of recorded prey families ($r^2 = 0.601$, $p < 0.001$), although not with prey family TD ($r^2 = 0.047$, $p = 0.272$). Thus, there was a need to correct diet breadth, but not diet TD, in further analyses. From this point forwards, diet breadth (prey family richness) was measured by residuals for its quadratic regression on diet sampling effort (see regression plot for diet breadth and associated study effort in Appendix Figure A.2.).

Linear mixed-effects models (LMMs) were used to analyse relationships between measures of tapeworm diversity and all recorded host features, each with identity link functions and Gaussian error distribution. The first main LMM looked at the influence of host features on tapeworm richness (response variable). This model included seven continuous (fixed) predictor variables (diet breadth, diet TD, trophic level, host length, depth mid-point, depth range, and latitudinal range) and three categorical (random) predictor variables (habitat type, dominant diet group, and host phylogeny [which was measured as host genus nested within host family, nested within host order]). The second main LMM looked at tapeworm TD as a response with the same predictors as the above model. To supplement the analysis of tapeworm TD, a model was also run where tapeworm TD and prey TD variables were replaced with associated variances in TD. This was done to gauge the taxonomic 'evenness' among host species. In addition to these LMMs, a "tips" analysis was performed, which involved re-running each model with the random effects removed. This analysis serves as an approach to examining differences to the importance of predictor variables when the phylogenetic relationships among hosts are not considered (See Figure 2.1 for species phylogeny).

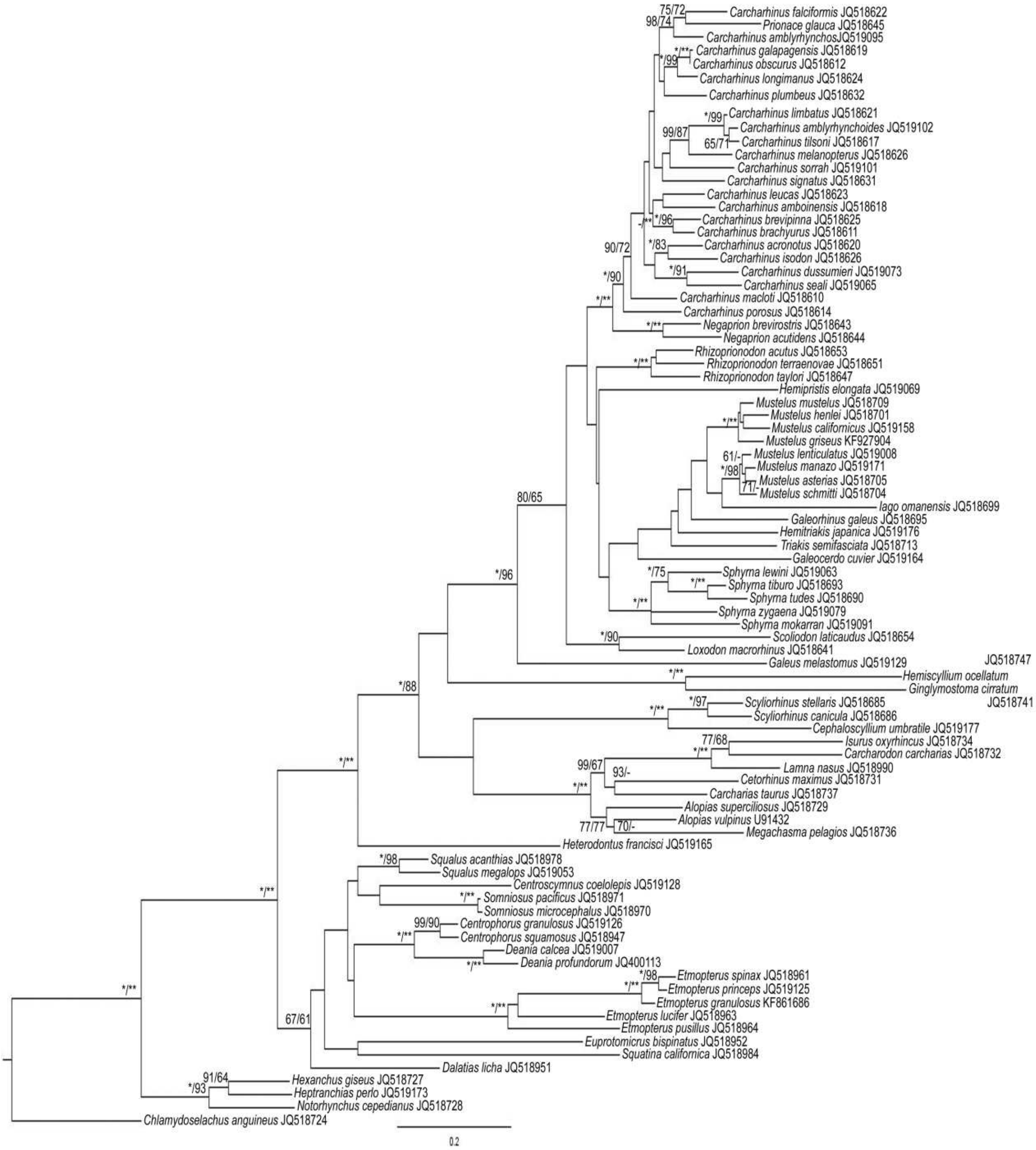

Figure 2.1: Phylogenetic tree displaying relationships among hosts included in this study (taken from Rasmussen and Randhawa 2017, submitted). The shark phylogeny was generated using Bayesian Inference from which contrasts were derived for phylogenetic independent contrast analyses (Rasmussen & Randhawa 2017, submitted). Numbers next to species names correspond to GenBank accession numbers for individual NADH2 sequences. Nodal support is expressed as posterior probabilities/bootstrap support; *, 100% posterior probability; **100% bootstrap support.

LMMs were each analysed in detail using the “MuMIn” package in R (Barton 2013). Corrected Akaike’s information criterion (AICc) and Akaike model-averaged weights (w+(i)) were calculated for all possible linear regression models (models with all possible combinations of the predictors), and used to determine the best models as well as the rank and relative importance of each individual predictor in each model. Model averaged parameter estimates and 95% confidence intervals were also calculated for each variable using methods summarised in Anderson (2008). To determine the interaction terms to be included in each model, a priori sets of second order interactions were selected for combinations of predictor variables that were thought to be relevant based on biological and ecological principles. For instance, there is a known association between host size and trophic level that should be accounted for (Poulin and Leung 2011). AICc values were compared between models including these sets and the models with only combinations of individual predictors to determine whether the inclusion of the interaction terms significantly improved models. All other potential interactions between predictors were assessed in the same way to make sure that no important interactions were missed. From these analyses it was decided that four interaction terms were to be included in the model predicting tapeworm richness (habitat & depth mid-point, diet breadth & diet TD, diet breadth & trophic level, trophic level & host size) and three were to be included in the model predicting tapeworm TD (diet breadth & diet TD, host size & trophic level, latitudinal range & depth range).

2.3. Results

Across the 91 different shark species analysed in the present data set, there was a total of 570 tapeworm-host associations. Based on the raw data, shark species harboured 6.26 tapeworm species on average (± 6.00 SD, range = 1 to 24) (Figure 2.2), and the average TD (taxonomic distinctness) and variance in TD of tapeworm assemblages was 3.39 (± 0.39 SD, range = 1.90 to 4.17) and 0.74 (± 0.51 SD, range = 0.00 to 2.53) hierarchical steps, respectively. Host species were commonly infected by a single tapeworm species and there was a positive skew in the number of tapeworm species infecting sharks (frequency of shark species decreased with increasing tapeworm richness) (Figure 2.2). In total, the host-parasite study effort across the 91 shark species included 2068 records, which equated to 22.73 mean records per host (± 35,67 SD).

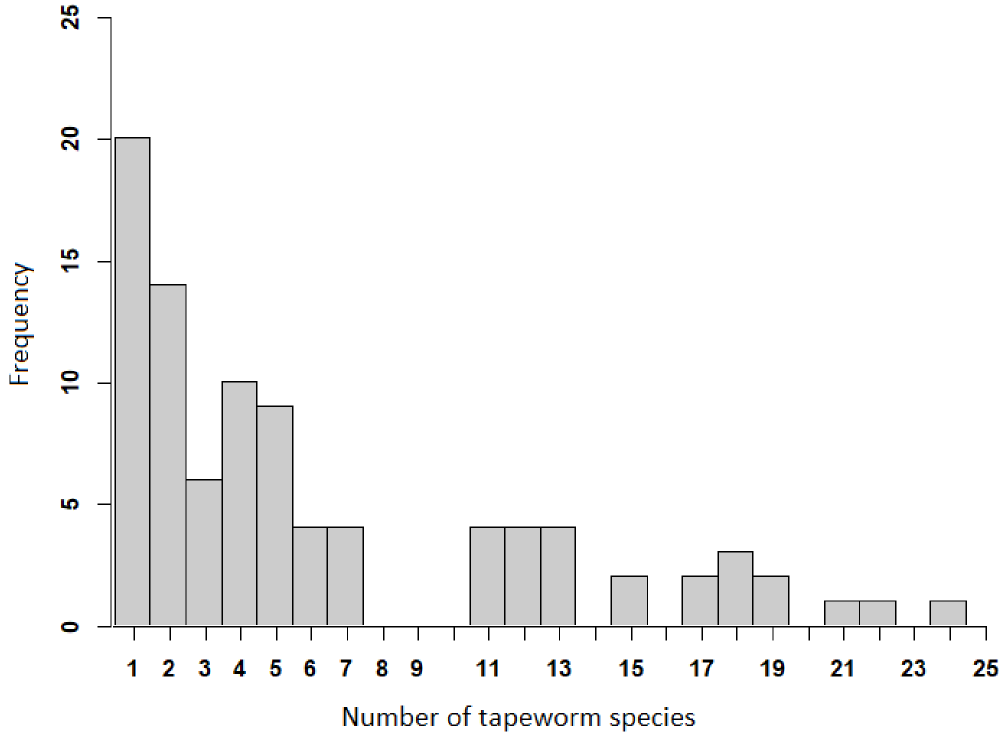

Figure 2.2. Frequency distribution of the number of shark species infected with different numbers of tapeworm species (n=91).

A total of 603 families of prey (within 163 orders; 39 classes; 16 phyla) were recorded in the data set. Shark species had records, on average, of 39.49 families of prey (± 34.16 SD, range = 1 to 145) (Figure 2.3), and the average TD and variance in TD of prey family assemblages was 2.81 (± 0.56 SD, range = 0 to 4.00) and 1.12 (± 0.42 SD, range = 0 to 2.05) hierarchical steps, respectively. As illustrated by Figure 2.3, diet breadth (prey family richness) was distributed with a positive skew across the shark species examined. In total, the number of stomachs examined across all shark species was more than 170,000, with 110,005 stomachs containing food. Notably, the number of stomachs with food examined was highly variable among species (mean = 1208.85 ± 4502.72 SD), and the spiny dogfish Squalus acanthias on its own had data from 40,698 stomachs containing food. The average trophic level of sharks in this dataset was 4.14 (± 0.34 SD, range = 3.20 to 4.94).

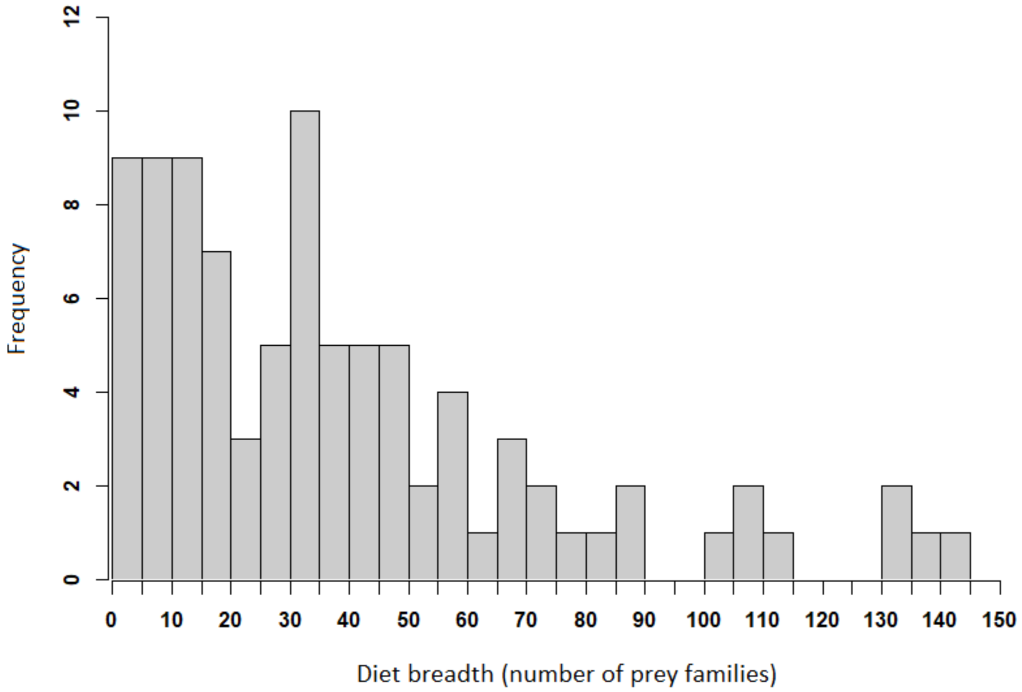

Figure 2.3. Frequency distribution of the number of shark species consuming various quantities of prey families (n=91).

2.3.1. Determinants of tapeworm richness

In the LMM analysis looking at tapeworm richness, it was found that random effects explained a total of 19.1% of observed variation in tapeworm richness, with the interaction of habitat and depth mid-point explaining 13.26%, host phylogeny explaining 5.84%, but habitat and diet composition each did not explain any variation (Table 2.1). The top AIC model explaining variation in shark tapeworm richness (AICc = 508.59) included a combination of only three fixed predictors, diet breadth, diet TD and trophic level. The top five best AICc models were very close (ΔAICc < 1), and included various combinations of diet breadth, diet TD, trophic level and the interaction effect of diet breadth and diet TD. The model-averaged Akaike weights analysis also showed that these diet related variables were the best predictors of tapeworm richness. The factor with the highest relative variable weight was diet breadth (w+(i) = 0.99), followed by diet TD (w+(i) = 0.71), trophic level (w+(i) = 0.59), and the interaction between diet breadth and diet TD (w+(i) = 0.37) (Table 2.1). It was interesting to note that although the association between diet breadth and tapeworm richness was positive (tapeworm increased with increasing diet breadth), the association between diet TD and tapeworm richness was negative (diet TD decreased with increasing tapeworm richness) (Table 2.1). Most other variables, including latitudinal range, depth mid-point, depth range and host size, were generally poor predictors of tapeworm richness in comparison (all w+(i) ≤ 0.01).

Table 2.1. Summary of host features as predictors for tapeworm species richness in sharks. The relative importance of predictors is compared by model-averaged weights (w+(i)s), ranks, parameter estimates, and 95% confidence intervals (CIs). 95% CIs in bold indicate statistical significance.

Random effects

Variable	Number of levels	Variance explained (%)
Host Phylogeny (Genus/Family/Order)	(n=40)	5.84%
Habitat	(n=7)	0.00%
Diet composition	(n=6)	0.00%
Habitat*Depth Mid-Point	(n=81)	13.26%

Fixed effects

variable	w+(i)	Rank	Parameter estimate	95% CI
Diet Breadth	0.99	1	0.288	(-0.332, 0.909)
Diet TD	0.71	2	-2.047	(-4.705, 0.611)
Trophic Level	0.59	3	0.806	(-1.758, 3.369)
Diet Breadth*Diet TD	0.37	4	-0.124	**(-0.213, -0.037)**
Diet Breadth*Trophic Level	0.14	5	-0.115	**(-0.211, -0.019)**
Latitudinal Range	0.01	6	0.005	(-0.020, 0.031)
Depth Range	<0.01	7	-0.001	**(-0.002, -0.000)**
Host Size	<0.01	8	-0.003	(-0.007, 0.002)
Depth Mid-Point	<0.01	9	-0.001	(-0.004, 0.001)
Host Size*Trophic Level	<0.01	10	0.000	(-0.000, 0.000)

Three predictors had significant effects on tapeworm richness, the interaction between diet breadth and diet TD, the interaction between diet breadth and trophic level, and depth range (their 95% confidence intervals were all bounded away from “0”) (Table 2.1). However, it should be noted that the effect size for depth range was small and it was only marginally significant (95% CI = (-0.002, -0.000), p = 0.044) (Table 2.1). The relationship between diet breadth and tapeworm richness was further analysed to determine the strength of the association. From a plotted linear regression (see Figure 2.4) it can be seen that there is a significant positive correlation between the variables with a moderate amount of variability surrounding the linear trend line ($r^2 = 0.220$, $p<0.001$). When the analysis was repeated with phylogeny and the other random effects removed, diet breadth, diet TD, the interaction between diet breadth and diet TD, and the interaction between diet breadth and trophic level all became highly significant predictors of tapeworm richness ($p <0.001$, Table 2.2). In contrast, depth range became non-significant (p = 0.276, Table 2.2).

Table 2.2. Summary of "tips" analysis assessing predictors of tapeworm richness (random effects of main models are excluded in this analysis). The relative importance of predictors is compared by model-averaged weights (w+(i)s), ranks, parameter estimates, and 95% confidence intervals (CIs). 95% CIs in bold indicate statistical significance.

Tapeworm Richness Fixed effects

variable	w+(i)	Rank	Parameter estimate	95% CI
Diet Breadth	1.00	1	0.926	**(0.279, 1.574)**
Diet TD	0.98	2	-3.046	**(-5.118, -1.115)**
Diet Breadth*Diet TD	0.94	3	-0.124	**(-0.219, -0.043)**
Trophic Level	0.89	4	0.283	(-2.074, 2.714)
Diet Breadth*Trophic Level	0.84	5	-0.109	**(-0.221, -0.039)**
Depth Range	0.71	6	-0.001	(-0.002, 0.000)
Depth Mid-Point	0.30	7	0.000	(-0.003, 0.004)
Host Size	0.29	8	0.000	(-0.017, 0.017)
Latitudinal Range	0.28	9	0.002	(-0.017, 0.034)
Host Size*Trophic Level	0.06	10	0.000	(-0.010, 0.008)

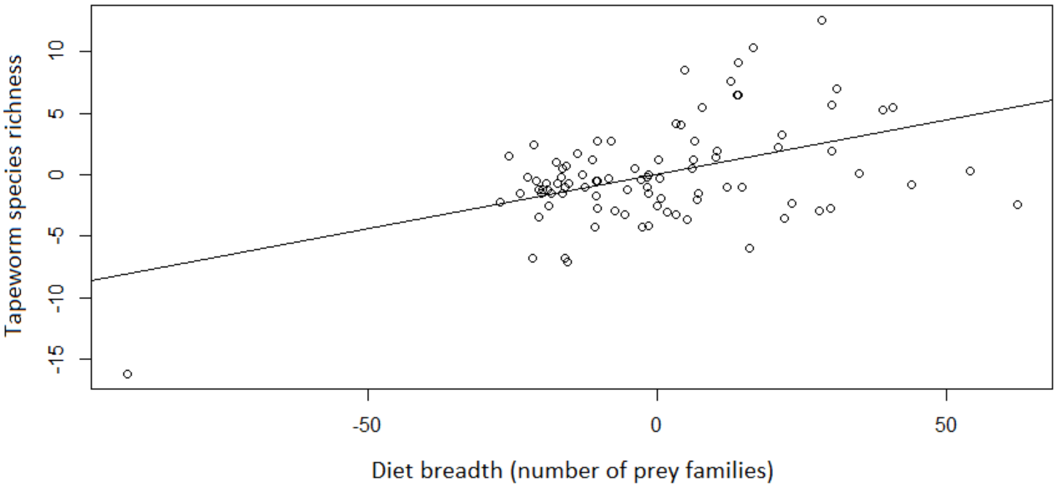

Figure 2.4. Scatter plot showing the association between tapeworm richness (measured as the residuals for the quadratic regression of number of tapeworm species on parasite sampling effort) and shark diet breadth (measured as the residuals for the quadratic regression of number of prey families on prey sampling effort). Solid line shows linear regression between the variables ($r^2 = 0.220$, $p<0.001$).

2.3.2. Determinants of tapeworm TD

In the LMM analysis looking at predictors of tapeworm taxonomic distinctness (TD), it was found that random effects overall accounted for 26.91% of the variation in tapeworm TD, and host phylogeny explained a much larger proportion of variation in tapeworm TD (20.15%) compared to tapeworm richness (Table 2.3). The top AIC model explaining variation in tapeworm TD (AICc = 71.75) was the null model (model including none of the fixed predictors in the data set). Notably, the null model was considerably better than all other AIC models (all others ΔAICc >2). Diet TD and trophic level were the best predictors of tapeworm TD included in the model, with the highest relative variable weights across all models (diet TD (w+(i) = 0.23) and trophic level (w+(i) = 0.10)). However, these model weights were still low, and ultimately, all variables included in the analysis were poor predictors of tapeworm TD. In addition, no variables demonstrated statistical significance for tapeworm TD (Table 2.3).

Table 2.3. Summary of various host features as predictors for tapeworm species taxonomic distinctness (TD) in sharks. Relative importance of these predictors is compared by model-averaged weights (w+(i)s), ranks, parameter estimates, and 95% confidence intervals (CIs). 95% CIs in bold indicate statistical significance.

Random effects

Variable	Number of levels	Variance explained (%)
Host Phylogeny (Genus/Family/Order)	(n=29)	20.15%
Habitat	(n=6)	2.25%
Diet composition	(n=5)	4.51%

Fixed effects

variable	w+(i)	Rank	Parameter estimate	95% CI
Diet TD	0.23	1	-0.210	(-0.447, 0.027)
Trophic Level	0.10	2	0.012	(-0.331, 0.356)
Diet Breadth	<0.01	3	0.001	(-0.004, 0.005)
Latitudinal Range	<0.01	4	-0.001	(-0.004, 0.002)
Host Size	<0.01	5	0.000	(-0.001, -0.001)
Depth Mid-Point	<0.01	6	0.000	(-0.000, 0.001)
Depth Range	<0.01	7	-0.000	(-0.000, 0.000)
Diet Breadth*Diet TD	0.00	8	0.000	(-0.000, 0.000)
Host Size*Trophic Level	0.00	8	0.000	(-0.000, 0.000)
Latitudinal range*Depth Range	0.00	8	0.000	(-0.000, 0.000)

When the analysis was repeated with phylogeny and the other random effects removed, Latitudinal range (w+(i) = 0.45) and depth range (w+(i) = 0.41) became the best predictors of tapeworm TD (Table 2.4). However, all variables remained poor predictors of tapeworm TD overall, and none of the host features included demonstrated significance in this analysis (Table 2.4).

Table 2.4. Summary of "tips" analysis assessing predictors of the average TD of shark tapeworm assemblages (random effects of main models are excluded in this analysis). The relative importance of predictors is compared by model-averaged weights (w+(i)s), ranks, parameter estimates, and 95% confidence intervals (CIs). 95% CIs in bold indicate statistical significance.

Tapeworm TD Fixed effects

variable	w+(i)	Rank	Parameter estimate	95% CI
Latitudinal Range	0.45	1	-0.003	(-0.009, 0.002)
Depth Range	0.41	2	-0.000	(-0.001, 0.000)
Depth Mid-Point	0.36	3	0.000	(-0.000, 0.001)
Trophic Level	0.33	4	-0.151	(-0.548, 0.246)
Diet TD	0.32	5	-0.097	(-0.355, 0.162)
Host Size	0.27	6	-0.000	(-0.003, 0.002)
Diet Breadth	0.26	7	-0.001	(-0.021, 0.018)
Latitudinal range*Depth Range	0.15	8	0.000	(-0.000, 0.000)
Host Size*Trophic Level	0.03	9	0.001	(-0.001, 0.002)
Diet Breadth*Diet TD	0.03	10	0.006	(-0.008, 0.020)

The model looking at determinants of variance in tapeworm TD was similar to the LMM for TD, with the null model being favoured. A large portion of variation (58.64%) in variance of TD was explained by host phylogeny (no other random effects explained variance), but all fixed variables were non-significant and poor predictors of variance in TD (Table 2.5). When the analysis was repeated with random effects removed, all factors remained poor predictors of variance in tapeworm TD (Table 2.6).

Table 2.5. Summary of host features as predictors for variance in the taxonomic distinctness (TD) of tapeworm assemblages in sharks. The relative importance of predictors is compared by model-averaged weights (w+(i)s), ranks, parameter estimates, and 95% confidence intervals (CIs). 95% CIs in bold indicate statistical significance.

Random effects

Variable	Number of levels	Variance explained (%)
Host Phylogeny (Genus/Family/Order)	(n=29)	58.64%
Habitat	(n=6)	0.00%
Diet composition	(n=5)	0.00%

Fixed effects

variable	w+(i)	Rank	Parameter estimate	95% CI
Trophic Level	0.22	1	-0.270	(-0.661, 0.121)
Variance in Diet TD	0.10	2	0.046	(-0.292, 0.385)
Diet TD	0.09	3	-0.061	(-0.365, 0.243)
Latitudinal Range	<0.01	4	-0.003	(-0.008, 0.002)
Diet Breadth	<0.01	5	0.002	(-0.004, 0.008)
Host Size	<0.01	6	-0.000	(-0.001, 0.000)
Depth Mid-Point	<0.01	7	-0.000	(-0.001, 0.000)
Depth Range	<0.01	8	-0.000	(-0.000, 0.000)
Diet Breadth*Diet TD	0.00	9	0.000	(-0.000, 0.000)
Host Size*Trophic Level	0.00	9	0.000	(-0.000, 0.000)
Latitudinal range*Depth Range	0.00	9	0.000	(-0.000, 0.000)

Table 2.6. Summary of "tips" analysis assessing predictors of variance in TD of shark tapeworm assemblages (random effects of main models are excluded in this analysis). The relative importance of predictors is compared by model-averaged weights (w+(i)s), ranks, parameter estimates, and 95% confidence intervals (CIs). 95% CIs in bold indicate statistical significance.

Variance in TD Fixed effects				
variable	w+(i)	Rank	Parameter estimate	95% CI
Depth Range	0.39	1	0.000	(-0.000, 0.001)
Trophic Level	0.33	2	-0.172	(-0.618, 0.274)
Latitudinal Range	0.27	3	-0.000	(-0.006, 0.006)
Diet Breadth	0.27	4	0.004	(-0.022, 0.031)
Diet TD	0.26	5	0.029	(-0.312, 0.371)
Depth Mid-Point	0.25	6	0.000	(-0.001, 0.001)
Host Size	0.25	7	0.000	(-0.002, 0.002)
Variance in Diet TD	0.24	8	-0.066	(-0.312, 0.371)
Latitudinal range*Depth Range	0.04	9	0.000	(-0.000, 0.000)
Diet Breadth*Diet TD	0.03	10	-0.010	(-0.028, 0.009)
Host Size*Trophic Level	0.02	11	-0.000	(-0.002, 0.001)

2.4. Discussion

The main objective of this study was to determine what host features influence the diversity of tapeworm assemblages in sharks, with focus on the influence of certain aspects of host diet, such as diet breadth, composition, and trophic level. In accordance with this, it was found that the breadth of a shark's diet, measured by its diversity of prey families, was a better predictor of tapeworm richness than any other host feature examined to date (Table 2.1). This outcome was robust with both parasite richness and diet breadth corrected to prevent confounding by their associated sampling efforts. Moreover, diet breadth showed a highly significant positive association with tapeworm richness after adjusting data to account for phylogenetic relationships between shark species (Table 2.2). Thus, the findings here support the hypothesis that shark species with broader diets encounter and subsequently acquire more tapeworm species than those with restricted diets**.** So far only a few empirical studies in other host-parasite systems have shown diet breadth to be important for the diversity of trophically-transmitted parasites (e.g. Chen et al. 2008; Locke et al. 2014). However, it is also worth noting that other studies looking at determinants of parasite diversity have rarely included diet breadth, and in fact, too few comparative studies have involved diet for meta-analysis to sufficiently assess its strength as a predictor of parasite richness (Kamiya et al. 2014). In such cases, there is an eminent need for more research into the diet of hosts. Where diet records are available, on the other hand, it is suggested that future studies involving trophically-transmitted parasites should consider finding ways to analyse diet breadth as a potential predictor of parasite richness among host species.

Using the average taxonomic distinctness (TD) of species assemblages to complement simpler measures of species diversity (i.e. species richness) has been a common practice in ecological studies for the past couple of decades (Von Euler & Svensson 2001; Heino et al. 2005; Winter et al. 2013). But despite its potential application as a measure for diversity in species diets, to my knowledge TD has only been implemented as a measure of diet breadth in one recent diet study involving the diet of turtles (Stringell et al. 2016). In the present study it was found that diet TD (the taxonomic distinctness among prey families in the diet) was the second most important predictor of tapeworm species richness in sharks following diet breadth (prey family richness). Interestingly, diet TD displayed a negative estimate in the LMM of tapeworm richness, entailing a net decrease in tapeworm richness with increasing diet TD. In addition, there was a significant negative interaction between diet breadth and diet TD. These results are somewhat peculiar given that prior to the analyses I predicted that tapeworm richness would increase with both the general diet breadth and diet TD of sharks. One possible reason for this discrepancy could be that most of these shark tapeworms have a high host specificity (Palm & Caira 2008), and from an evolutionary perspective, these tapeworms are likely to exploit a narrower (more closely related) range of intermediate hosts than other more generalist parasites. Thus, if transmission of tapeworms is generally limited to more closely related intermediate hosts (low diet TD), perhaps parasite speciation has also been favoured more in closely related hosts, leading to infection with multiple congeners in these hosts (i.e. high tapeworm richness within low diet TD). This could mean that certain taxonomic groups are generally devoid of tapeworm larval stages and may inflate prey TD to be higher in certain sharks that eat them, despite not being relevant for tapeworm transmission. Nevertheless, without knowing the relative importance of these taxonomic groups as intermediate hosts for tapeworms, reasons for the discrepancy found between diet breadth and diet TD here can only be speculated. Importantly, for many elasmobranch tapeworm species, intermediate hosts are completely unknown (Palm 2004; Jensen & Bullard 2010; Caira & Jensen 2014), and further research on the life cycles of these tapeworms could be key towards better understanding the relative importance of these diet aspects for tapeworm richness in sharks.

Large-scale food web analyses have previously highlighted trophic level as an important aspect of host diet that can drive patterns of parasite richness (Lafferty et al. 2006; Chen et al. 2008). Trophic level was found to be the third best predictor of shark tapeworm richness in this study (Table 2.1), but unlike measures of diet breadth, trophic level did not have a significant effect when data were adjusted to account for phylogenetic relationships between hosts (Table 2.2). It is worth noting, however, that trophic level and diet breadth had a significant interaction in the model, indicating that although these variables measure different diet aspects (trophic level reflects the position of a shark's prey in the food web, where general diet breadth does not), they are related on some level, and may both gauge how broad a shark's diet is. When considering this, one could posit that tapeworm richness in sharks is determined more by the breadth of different prey in a host species' diet than by the position of these prey in food webs. A study of other marine fishes has also observed trophic level to have less impact on parasite richness compared with breadth of diet (Locke et al. 2014). Even so, there is a question of why trophic level has shown to be a key driver of parasite richness in network studies, while appearing to be of less importance here. Locke et al. (2014) have offered a few plausible explanations: (1) trophic level is less relevant in the context of a fish community because it varies much less than in larger networks of species, and (2) links found between parasite richness and trophic level in other studies may reflect their association with diet breadth, meaning that diet breadth may actually be the underlying predictor of significance for parasite richness. It is also worth considering that trophic level may not appear as important in vertebrates where species at higher trophic levels have their parasite faunas restricted by more complex and effective immune responses (Benesh et al. 2014). There may be some truth to each of these explanations. In any case, further research will be necessary to fully understand the relative influences of diet breadth and trophic level on parasite richness, and considering the results here, it is recommended that such studies look simultaneously at both factors as predictors of parasite richness (and account for their interaction).

All variables other than host diet breadth, trophic level, and diet TD were comparatively poor predictors of tapeworm species richness in this study. However, it is worth pointing out that when the effect of phylogeny was removed from the analyses, many factors became more important (Table 2.2). The depth range of a shark species was a poor predictor of tapeworm richness (despite being marginally significant), but became considerably more important when data were adjusted to remove host phylogeny. Little is known about how depth factors regulate parasite diversity in the marine environment, but it has been suggested that depth-driven temperature gradients could influence tapeworm richness in elasmobranchs (Randhawa & Poulin 2010). Prior to this study, I also thought that differences in depth might reflect differences in habitat or diet among hosts. Yet, the results here showed conversely that although there was an interaction between habitat and the mid-point of shark depth ranges, habitat itself did not explain any variation in tapeworm richness. Likewise, diet composition, which was represented by the shark's preferred taxonomic group of prey, did not account for any variation in tapeworm richness (Table 2.1). As such, despite measures of diet breadth demonstrating large influences on tapeworm species richness in sharks, the importance of the sharks' preferred prey and habitats appeared to be negligible (Table 2.1).

Prior to this study, it was predicted that shark species feeding predominantly on teleost fishes would have more tapeworm species than sharks feeding mainly on other prey groups because most currently described intermediate hosts for tapeworms are teleost fishes (Palm 2004; Jensen 2009). With the present results running counter to this proposal, there is a question of why the dominant prey group of sharks does not appear to be a major determinant of tapeworm richness. One thing worth noting is that crustaceans and cephalopods (the two most commonly dominant prey groups of sharks other than teleosts) have been described as intermediate hosts for many tapeworms, but are generally less studied for parasites than teleost fishes, meaning that they likely harbour larval stages of substantially more species of tapeworms than have been currently described (Jensen 2009). Another possibility is that the lack of importance for diet composition reflects the limitations of simplifying composition into such large taxonomic groups. Perhaps more specific diet composition involving comparisons at the species or genus level could more accurately reflect differences in tapeworm richness. Analysing diet composition at this level would prove extremely challenging, however, since a large portion of studies on shark diets do not provide composition for individual species, genera, families, or even orders, and instead only estimate the full composition of major taxonomic groups. The importance of habitat for tapeworm richness may similarly be limited by the simplification of shark's ecologies. Although habitat categories such as “Demersal” and “Bentho-pelagic” summarise the areas where these sharks are distributed, there can be substantial differences in the foraging behaviours, dietary preferences and distributions of sharks living in these same habitats.

The lack of importance of host size for parasite richness in this study was surprising given that host size is a key predictor of parasite richness for a vast range of taxa (Kamiya et al. 2014). Larger host species generally provide more space and a greater diversity of niches for parasites to exploit (Kuris et al. 1980; Poulin & Morand 2004). Interestingly, however, Randhawa and Poulin (2010) have found host size to be a non-significant predictor of tapeworm richness in sharks, but found that it became a highly significant predictor when using Phylogenetic Independent Contrasts analyses to control confounding of host's phylogenetic relationships (Randhawa & Poulin 2010). Here a different method was used to examine the importance of variables without the influence of phylogeny (a “tips” analysis to remove phylogeny from the LMM), but host size was still non-significant for tapeworm richness (Table 2.2). This inconsistency highlights the difference between these methods to adjust for phylogenetic relationships. Phylogeny in the analysis here was measured by host genus nested within family, nested within order, and this assumes that species in these groups are equally related. On the other hand, the PIC method uses a phylogenetic tree which accounts for the genetic differences within each of these taxonomic groups (see Figure 2.1). Based on this, perhaps one could say that host size is probably a key predictor of tapeworm richness after accounting for phylogeny, because the

Phylogenetic Independent Contrasts result corrects for the effects of phylogeny using more precise data. However, it must be also be noted that Phylogenetic independent contrasts analyses are more sensitive to phylogeny due to the assumptions of accurate branch lengths and correct topology (Felsenstein 1985, Ackerly 2000), so any taxon bias or phylogenetic uncertainty, e.g. polytomies, might lead to questionable results that should be interpreted with caution (Ackerly & Reich 1999). In any case, increased sampling of shark diets and parasite assemblages across all shark orders should provide further insight into the importance of phylogeny and host size for tapeworm richness.

Like host size, geographical range is generally known to be a key predictor of parasite richness for a broad range of taxa (Kamiya et al. 2014), but latitudinal range was found to be of little importance for tapeworm richness in sharks here. It was thought that shark species spanning greater ranges of latitude would likely encounter more prey taxa, and consequently encounter a greater diversity of parasite species than sharks with covering limited ranges in latitude. Yet, given that diet breadth, but not latitudinal range, had a significant influence on tapeworm richness, perhaps latitudinal range is not a good predictor since latitude indicates only one dimension of a shark's distribution, whereas the diversity of prey taxa encountered by a shark would also vary with their longitudinal and depth distributions, in addition to their temporal patterns in foraging. Some other measures may be much better proxies for total geographical range of shark species (e.g. total area distribution). However, information on these other aspects of geographical range are unknown for many sharks, and would need to become available for further analysis.

The LMM looking at predictors of the average taxonomic distinctness (TD) of tapeworm assemblages found that no factors included in this study were good predictors of tapeworm TD (Table 2.3). This was somewhat surprising given that a few of the same variables (especially diet breadth) were considerably more important predictors of tapeworm species richness (Table 2.1), but as previous research has illustrated, parasite richness and average TD of parasite assemblages are sensitive to different host features (Luque et al. 2004; Heino et al. 2005; Luque & Poulin 2008; Randhawa & Poulin 2010). It is possible that diet breadth has much less influence on tapeworm TD than tapeworm richness because the distinctness of tapeworm assemblages is driven more by other factors that were not accounted for in this study. For instance, tapeworm TD may be driven more by the compatibility of tapeworms with shark hosts. From an evolutionary perspective, hosts are more likely to be compatible with closely related tapeworm species, and as such, a shark could have a broad diet and encounter many tapeworm species, but only tapeworms within a limited range of taxa may actually be able to exploit the shark i.e. they could have species rich parasite assemblages with overall low TD consisting of many congener species. Notably, immune response has already been demonstrated to prevent infection by certain cestode species in elasmobranchs (McVicar & Fletcher 1970; Randhawa & Burt 2008). However, the importance of host compatibility in determining the taxonomic diversity of tapeworms in sharks warrants further investigation. In line with the results for average tapeworm TD, there were no good predictors observed for variance in tapeworm TD. This indicates that the taxonomic evenness of tapeworm assemblages in sharks is unlikely to be influenced by the factors examined. However, the present dataset including variance in tapeworm TD was considerably more restricted (n= 57 species) and this variable may be worth revisiting in future when more shark tapeworm records are described.

2.4.1. Conclusions

A total of 91 different shark species were examined in this study, which is less than a fifth of all described shark species known to date (Randhawa et al. 2015). However, a more complete analysis of diet's influence on tapeworm richness would require host diet and tapeworm records to become available for many more shark species which are currently data deficient. The present study is the first to examine the influence of host diet and trophic level on parasite diversity in elasmobranchs, and to my knowledge, is the most comprehensive analysis of parasite diversity in sharks thus far. This gives credence to the key finding that the diet of a shark species, and particularly the breadth of its diet, has important consequences for the diversity of its trophically transmitted parasites. The intricacies of this link between host diet breadth and tapeworm diversity in sharks warrant deeper exploration. Despite having records of prey for the shark species examined here, the parasites of these prey items are generally unknown, and it remains uncertain whether these prey are actually intermediate hosts contributing to shark tapeworm richness. Indeed, several studies have pointed out that there are major gaps in our knowledge of the life cycles of elasmobranch tapeworms (Palm 2004; Jensen & Bullard 2010; Randhawa & Brickle 2011; Caira & Jensen 2014). These life cycles will need to be elucidated for a more in-depth understanding of diet breadth's influence on tapeworm diversity.

Chapter 3: Linking the diet composition of sharks to their tapeworm compositions and use of prey as intermediate hosts

3.1. Introduction

In the past few decades parasites have gained increased appreciation as a vital component of food webs (Marcogliese and Cone 1997; Lafferty et al. 2008; Dunne et al. 2013). The collective biomass of parasites distributed throughout a food web has been shown to exceed that of top predators in certain ecosystems (Kuris et al. 2008) and contribute substantially to energy flow through species networks (Johnson et al. 2010; Goedknegt et al. 2012; Thieltges et al. 2013). Moreover, parasites can largely influence the structure of free-living communities and impact the strength of trophic links among species (Marcogliese 2003; Thompson et al. 2005; Lefèvre et al. 2009; Poulin 2010). For instance, larval trematode parasites have been shown to induce behavioral changes in their intermediate hosts that increase their susceptibility to bird definitive hosts, consequently strengthening the link between these different animals (Lafferty & Morris 1996). This is one of many examples of this type of influence (Aeby 1991; Thomas and Poulin 1998; Thomas et al. 2010; Bakker et al. 2017). On the flip side of these interactions, the structure of food webs may shape the ecology of these parasites, and their ability to strengthen trophic links may have ultimately arisen from selection pressures on parasites to reach their definitive hosts (Lafferty 1999; Lefèvre et al. 2009). Food web structure especially may have implications for the ecology of parasites with highly complex life cycles that depend on several different interactions among free-living species to reach their definitive hosts.

Certain prey in food webs provide better routes than others for trophically-transmitted parasites to the definitive hosts (Thompson et al. 2013). For trophically-transmitted parasites, infecting intermediate hosts that are closely linked with their desired definitive hosts should increase the probability of completing their life cycle. Therefore, we might expect larval stages of trophically-transmitted parasites to occur more frequently in intermediate hosts that are abundant, high in biomass, or constitute a large proportion of their definitive host’s diet (Cirtwill et al. 2017). However, parasites also face phylogenetic constraints that limit which prey are suitable intermediate hosts (Euzet and Combes 1980; Combes 2001). Acanthocephalan parasites, for instance, are restricted to using arthropod intermediate hosts to reach their vertebrate definitive hosts (Near 2002). In some cases, host evolutionary history may be even more important in determining the range of intermediate hosts used by parasites than the strength of their trophic links to the definitive host (Cirtwill et al. 2017). Nevertheless, the importance of ecology and host evolutionary history for intermediate host-use may vary considerably among different parasites and hosts, and is yet to be investigated in many ecosystems.

Tapeworms infecting elasmobranchs (sharks, skates and rays) represent an incredibly diverse and speciose group of marine parasites (Caira and Healy 2004; Caira and Jensen 2014). These parasites all have complex life-cycles and use a range of different invertebrate and vertebrate prey as intermediate (or paratenic/non-obligate) hosts, including, but not limited to, a variety of teleost and elasmobranch fishes, crustaceans, and molluscs (Palm 2004; Caira and Jensen 2017). In general, these tapeworms are highly host specific as adults, living in the spiral intestines of their definitive elasmobranch hosts, but show much lower specificity as larval stages in their respective intermediate hosts (Palm & Caira 2008; Jensen & Bullard 2010). Several cosmopolitan elasmobranch tapeworm species with lower host specificity may be exceptions to this, but these species also show considerably lower specificity in intermediate hosts (Palm & Caira 2008). Tentacularia coryphaenae, for instance, has been described as adults from more than 10 different elasmobranch species. Their larval stages have been described from more than 80 different intermediate host species in more than 40 different taxonomic families (Palm & Caira 2008). From what is currently known of elasmobranch tapeworm life cycles, different taxonomic groups of the parasites may use distinctly different types of intermediate hosts (Palm 2004; Palm & Caira 2008). For example, certain trypanorhynch tapeworms in the families Eutetrarhynchidae and Aporhynchidae are known to use almost exclusively crustaceans (e.g. crabs and shrimps) as their second intermediate hosts, whereas trypanorhynch tapeworms of the family Lacistorhynchidae seem to use generally teleost fishes as their second intermediate hosts (Palm 2004). These patterns have been established from limited information on certain species, however, and until further research is done to elucidate the life cycles of more tapeworms, it cannot be said whether these patterns of intermediate host use are consistent within taxonomic groups (Palm 2004). Notably, the life cycles of different elasmobranch tapeworms have been poorly described, and in many cases their intermediate hosts are completely unknown (Caira & Reyda 2005; Jensen & Bullard 2010).

Ongoing research in parasitology is continuing to reveal new records for elasmobranch tapeworms in both intermediate and definitive hosts (Caira and Jensen 2017). However, there remain significant gaps in knowledge regarding what factors underpin these patterns of host use. Questions remain as to which described intermediate hosts may represent “dead ends” for the parasites (ecological sinks), and which hosts are likely to transmit parasites to suitable paratenic or definitive hosts (ecological links) (Jensen and Bullard 2010). Likewise, it is unknown whether intermediate host use by these parasites is strongly linked to the diets of their definitive elasmobranch hosts, or alternatively, whether the links between intermediate and definitive hosts are dynamically weak as has been observed in other food webs (Cirtwill et al. 2017). Palm et al. (2017) have provided some recent insights, showing that the depth, diet, and habitat of sharks are major factors influencing the composition of their trypanorhynch tapeworm assemblages. Yet, the influence of diet has only been examined at a very coarse level (between vertebrate and invertebrate feeding sharks) (Palm et al. 2017), and many questions remain about how more specific taxonomic groups of prey in shark diets might influence their tapeworm compositions. Answering these questions may be critical towards better understanding and predicting the life cycles of these important marine parasites.

3.1.1. Objectives of chapter 3

In chapter two of this thesis, it was found that the dominant prey group in shark diets (e.g. crustaceans, cephalopods or teleosts) had little bearing on the overall diversity of their tapeworm assemblages. Diet composition may be substantially more important for the composition of their respective tapeworm assemblage, however, since as noted above, tapeworms in different taxonomic groups may use different types of intermediate hosts (i.e. some tapeworms may only use crustaceans as second intermediate hosts whereas others may use cephalopods or teleosts). The objective of this chapter was to investigate association between the diet and tapeworm composition of sharks. The first part of this chapter involved the use of ordination methods to map known tapeworm and prey taxa across different shark species. These methods served to analyse whether similarities in diet composition among sharks are correlated with their similarities in tapeworm composition, and more specifically, whether tapeworm assemblages show distinct differences among different feeding groups of sharks (e.g. teleost-feeding and cephalopod-feeding sharks). The second part of this chapter looked more specifically at whether intermediate host use by tapeworm species is associated with their contribution to shark diets i.e. whether families of prey that harbour many different tapeworms are also important contributors to the diets of sharks that the tapeworms infect as adults. This entailed searching the literature for tapeworm records in families of intermediate hosts that could be compared with shark diet records for the same animal families. Only trypanorhynch tapeworms (Order Trypanorhyncha) were examined for their larval records in this part because unlike other tapeworms, they have consistent morphology between larval stages and adults, and thus, have been more consistently identified from intermediate hosts (Palm 2004; Jensen & Bullard 2010). Intermediate host records for other tapeworms are rare due to their reliance on molecular tools for identification, making it difficult to be explore their life cycle patterns based on current records (Jensen & Bullard 2010). In any case, patterns of intermediate host use by trypanorhynchs especially warrant investigation because the Order Trypanorhyncha is a basal tapeworm group of elasmobranchs and is the most speciose tapeworm Order in elasmobranchs known to date (Mariaux & Olson 2001; Hoberg & Klassen 2002; Caira & Jensen 2017; Palm et al. 2017).

3.2. Methods

3.2.1. Data collection of shark diets and tapeworms

All data on tapeworms infecting sharks were compiled from a revised dataset of tapeworms in elasmobranchs that is publicly available from Randhawa and Poulin (2010). The final dataset included tapeworm species records for more than a hundred different shark species listed in Zoological Records on ISI Web of Knowledge from 1864 to 2017 (see methods in 2.2.1 for more detail). Searches for tapeworm records were conducted for each shark species on ISI Web of Knowledge by searching its Latin name and all known synonyms combined with keywords ‘‘Parasit* OR disease OR pathog*’’. All data on prey families of sharks were compiled in a similar fashion; for each shark species which had tapeworm records, prey records were obtained from all published records listed across all databases on ISI Web of Knowledge from 1864 to 2017. Diet records were obtained for individual shark species by searching its Latin name and all known synonyms combined with keywords ‘‘diet* OR feed* OR prey*’’. The final dataset included all known prey families for each of 91 different shark species (for a list of prey families for each of the sharks see Table A.1 in Appendix). Given that many diet studies were published decades ago and taxonomic changes had been made to certain prey taxa since their publication, all prey taxa recorded were checked in the World Register of Marine Species (WoRMS) database and updated to reflect their currently accepted taxonomical nomenclature (WoRMS Editorial Board 2017). For information on the dominant prey groups of each shark species, the percentage composition of nine different prey groups were recorded from each diet reference (teleost fishes, cephalopods, crustaceans, mammals, chondrichthyan fishes, reptiles, birds, other molluscs, other invertebrates). The overall composition of these groups for each shark species was calculated by taking the average compositions across all studies, weighted by the number of non-empty stomachs examined for diet (See 2.2.2 for details on how composition was quantified). Overall diet composition was also recorded for the five families of prey most dominant in each shark species’ diet (this was relevant for later analyses linking intermediate hosts of trypanorhynch tapeworms to shark prey; see 3.2.4.).

In preparation for ordination analyses, tapeworm and diet data were formatted into presence/absence matrices in Microsoft Excel. For tapeworm data, a binary matrix was constructed giving the presence/absence of tapeworm families for each shark species (shark species x tapeworm families), where presence of a tapeworm family was indicated by “1” and absence of a tapeworm family was indicated by “0”. Tapeworm composition was analysed at the family level since tapeworms are highly host specific in elasmobranchs and, consequently, there is generally low overlap of tapeworms among different shark species (Caira and Jensen 2008). There is considerably more overlap of tapeworm families among sharks, allowing more meaningful comparisons in composition among hosts. Presence/absence of tapeworm families was used to represent composition rather than the species richness per family because the main interest of the present study was to explore the influence of diet on tapeworm composition in sharks, rather than tapeworm diversity which was already explored in detail within chapter two of this thesis. Only shark species with at least two available adult tapeworm records were included in the final matrix (akin to the methodology of Palm et al. 2017). This restriction was made to ensure more reliable comparisons in the ordination since species with fewer than two tapeworm records could have just been poorly studied for parasites. Based on this minimum criterion, the analysis of tapeworm composition included a total of 272 host/tapeworm family records, involving 22 different tapeworm families recorded across 61 different shark species.

For diet data, a binary matrix was constructed giving the presence/absence of prey families for each shark (shark species x prey families), where presence of a prey family was indicated by “1” and absence of a prey family was indicated by “0”. Consistent with the tapeworm data, only shark species with at least two prey families described were included in the final matrix. This restriction was made because shark species with fewer than two prey family records may have been poorly studied for diet, and thus, removing them ensured more reliable comparisons in the ordination. Based on this minimum criterion, the analysis of diet composition included a total of 2,793 shark/prey family records, involving 398 different families of prey recorded across 61 different shark species (importantly, these 61 shark species were the same species analysed for tapeworm composition).

3.2.2. Analysis of diet vs. tapeworm composition

All statistical tests were carried out in the R environment (R Development Core Team 2012). Prey and tapeworm family matrices were uploaded into R and the package “betapart” (Baselga & Orme 2012) was used to transform each presence-absence matrix into a distance matrix of pair-wise dissimilarities among shark species. The Jaccard similarity index was used to generate dissimilarities. This index was selected for its simplicity and widespread application to presence/absence data, and for this study, it specifies the number of prey (or tapeworm) families shared by two shark species divided by the total number of prey (or tapeworm) families found across both shark species. Similarity measures are usually not independent of richness gradients among sites (in the case of this research, similarity would not be independent of gradients in prey and tapeworm family richness among shark species), meaning that differences in composition among sites can be obscured by differences in richness among sites (Baselga et al. 2007; Carvalho et al. 2012). One method that has been proposed to resolve this issue is to quantify dissimilarities by two different components; nestedness and turnover (Baselga 2010). The nestedness component of dissimilarity accounts for differences in composition that are due to species loss between sites (in this case, prey/tapeworm family loss between shark species), whereas the turnover component, which is independent of potential differences in richness between sites, reflects replacement of species between sites (in this case, the replacement of prey/tapeworm families by other families between sharks) (Koleff et al. 2003; Baselga 2010; Baeten et al. 2012). For this study, both prey and tapeworm distance matrices were partitioned into nestedness and turnover components of dissimilarity using the "betapart" package (Baselga & Orme 2012), and since the main interest was in quantifying differences in tapeworm composition among sharks (not differences in richness), further analyses of prey and tapeworm composition were conducted specifically on turnover dissimilarities.

The "metaMDS" function of the "vegan" package in R (Oksanen 2013) was used to generate two-dimensional nMDS ordinations plotting dissimilarities in tapeworm and prey composition among shark species. Both prey and tapeworm nMDS ordinations found global solutions within 20 runs. Initial ordinations displayed the species names for each shark, but they were subsequently re-plotted with two factors (dominant diet group and shark order) superimposed on the plot to show composition differences among sharks with different dominant prey groups and different orders (each shark species was coded with a symbol to represent its associated group). To test for significant differences in composition among sharks with different dominant prey groups and sharks with different orders, the "adonis" function was used to run non-parametric (permutational) MANOVAs for both factors (999 permutations). Pairwise dissimilarities among the groups were calculated by re-running the function on each individual pair of groups (e.g. for the difference among two shark orders, the data was adjusted to include only sharks from those two orders). In instances where significant differences were seen between groups, SIMPER analyses (Clarke 1993) were performed to explore which prey families, or which tapeworm families, contributed most to compositional differences. In these analyses prey and tapeworm families were ordered by increasing contribution (%) to the total dissimilarity observed between groups. To determine overall correlation between the tapeworm and prey dissimilarities of sharks, the Mantel statistic was used (function "mantel" in the "vegan" package).

3.2.3. Data collection on intermediate host families

A list of trypanorhynch tapeworm species was compiled for the 91 shark species with recorded diet information (only tapeworms known to infect spiral valves of these sharks as adults). Intermediate host records were obtained for each species from Palm (2004), which provides a comprehensive list of intermediate hosts described for trypanorhynchs up until 2004. Additional records published after 2004 (between 2004 and 2017) were compiled from all databases of ISI Web of Knowledge. References were found on Web of Knowledge by searching the taxa of each tapeworm (Latin name plus all known synonyms) combined with keywords “larva* OR plerocerc* OR merocerc* OR procerc* OR cysticerc* OR "intermediate host"” (searches were all conducted in September 2017). Overall, 186 different animal families were recorded as intermediate hosts across 74 of the 139 trypanorhynch species infecting the 91 included shark species (see hosts and associated references in Appendix Table A.2).

The occurrence of animal families as intermediate hosts to trypanorhynch tapeworms is likely to be substantially influenced by each family’s sampling effort for parasites. Considering this, a measure of sampling effort was obtained for each animal family from ISI Web of Knowledge by searching each family name and all its described genera (all currently accepted genera described on WoRMS) combined with keywords ‘‘Parasit* OR disease OR pathog*’’. Several different genera were synonymous with taxa from unrelated families. In light of this potential error in searches, the list of taxa returned for each search was checked to make sure all included species and genera were part of the family in question. Where taxa were not part of the family, the search was refined to exclude them and their associated references. Families of terrestrial mammals recorded from shark diets were not included due to their potential inflation of sampling effort, and their unlikelihood to serve as intermediate hosts to shark tapeworms.

3.2.4. Analysis of intermediate host importance

All statistical tests were conducted in the R environment. To investigate the relationship between intermediate host use by trypanorhynch tapeworms and importance to shark’s diets, a linear mixed model (LMM) was run with trypanorhynch richness of animal families as a response. To correct for the influence of sampling effort, trypanorhynch richness was measured by the residuals for its quadratic regression on sampling effort (a plot of this regression can be seen in Appendix Figure A.3). Prey phylogeny was included as a random effect in the model (measured as prey order, nested within prey class, nested within prey phylum). Two fixed predictors were included; (1) the overall occurrence of the animal family as prey across the 91 shark's diets, and (2) the occurrence of the animal family as important prey across the 91 shark's diets (important prey families were defined as those within a shark's top five prey families by overall composition). The interaction between these fixed effects was included in the model since common prey families are also likely to be important in shark's diets.

To determine whether sharks feeding on trypanorhynch-rich prey had more trypanorhynch tapeworms as adults, the average trypanorhynch richness across a shark's top five prey families by composition (adjusted for sampling effort as stated above) was plotted against its richness of adult trypanorhynchs (measured as residuals for the quadratic regression of richness on sampling effort (regression plot available in Appendix, Figure A.4). Significance was determined by a linear regression between these variables.

3.3. Results

3.3.1. The influence of sharks' diets on their tapeworm compositions

The 61 different shark species included in the analyses comparing diet and tapeworm compositions (Figures 3.1 & 3.2) showed considerable bias towards shark species with certain diets. Sharks with diets dominated by teleost fishes were by far the most frequent in the data (n=38), followed by sharks with diets dominated by crustaceans (n=14), and sharks with diets dominated by cephalopods (n=5). Sharks with diets dominated by other groups were poorly represented in comparison (chondrichthyan fishes (n=2) and other invertebrates (n=2)). The species were also biased towards certain taxonomic orders. Notably, more than two thirds of the sharks included were of the Order Carcharhiniformes (n=41). Lamniformes (n=7) and Squaliformes (n=6) were reasonably well represented, but all other orders (Hexanchiformes, Heterodontiformes, Orectolobiformes and Squatiniformes) were poorly represented, each with three or fewer species included.

There was a significant difference in prey family compositions among sharks with different dominant prey groups (ADONIS; R^2=0.188, p<0.001) (Figure 3.1B). Pairwise comparisons indicated that there was a notably large difference in prey family composition between crustacean-feeding and teleost-feeding sharks (ADONIS; R^2=0.124, p<0.001), and between crustacean-feeding and cephalopod-feeding sharks (ADONIS; R^2=0.191, p=0.002). However, prey composition was not significantly different between teleost-feeding and cephalopod-feeding sharks (ADONIS; R^2=0.035, p=0.161) (Figure 3.1B), indicating substantial overlap in the families of prey consumed between sharks feeding primarily on teleosts and sharks feeding primarily on cephalopods.

Prey family composition showed a significant difference among sharks' taxonomic orders (ADONIS; R^2=0.179, p<0.001) (Figure 3.1C). Pairwise comparisons revealed significant differences in prey family composition between squaliform and lamniform sharks (ADONIS; R^2=0.141, p=0.032), between carcharhiniform and squaliform sharks (ADONIS; R^2=0.082, p<0.001), and between carcharhiniform and lamniform sharks (ADONIS; R^2=0.044, p=0.021), (comparisons for other orders were not included due to their small sample sizes). SIMPER analysis determined that the five prey families contributing most to dissimilarity between lamniform and squaliform sharks were teleost fish families Macrouridae (cumsum=0.015, p<0.001), Myctophidae (cumsum=0.027, p=0.017), and Callionymidae (cumsum=0.061, p=0.005), and the cephalopod families Histioteuthidae (cumsum=0.050, p=0.031) and Sepiolidae (cumsum=0.039, p=0.003). All five of these families were significantly more common in the diets of squaliform sharks (in 100%, 100%, 66.7%, 100% and 66.7%, respectively) than in the diets of lamniform sharks (in 14.3%, 28.6%, 0.0%, 42.9% and 0.0%, respectively). The five prey families contributing most to dissimilarity between carcharhiniform and squaliform sharks were the teleost fish families Macrouridae (cumsum=0.014, p<0.001), Myctophidae (cumsum=0.038, p=0.002), and Gadidae (cumsum=0.057, p=0.008), and the cephalopod families Histioteuthidae (cumsum=0.026, p=0.002) and Sepiolidae (cumsum=0.048, p=0.003). All five of these families were significantly more common in the diets of squaliform sharks (in 100%, 100%, 66.7%, 100% and 66.7%, respectively) than in the diets of carcharhiniform sharks (in 12.2%, 22.0%, 19.5%, 42.9% and 19.5%, respectively).

The five families contributing most to dissimilarity between carcharhiniform and lamniform sharks were the teleost fish families Merlucciidae (cumsum=0.011, p<0.001), Sebastidae (cumsum=0.022, p<0.001), Sciaenidae (cumsum=0.041, p=0.084) and Paralepididae (cumsum=0.050, p=0.004), and the crustacean family Penaeidae (cumsum=0.032, p=0.196). The teleost families (Merlucciidae, Sebastidae, Sciaenidae and Paralepididae) were more common in the diets of lamniform sharks (in 85.7%, 71.4%, 71.4% and 57.1%, respectively) than in the diets of carcharhiniform sharks (in 26.8%, 7.3%, 51.2% and 2.4%, respectively). Conversely, Penaeidae was present more in the diets of carcharhiniform sharks (68.3%) than in lamniform sharks (42.9%).

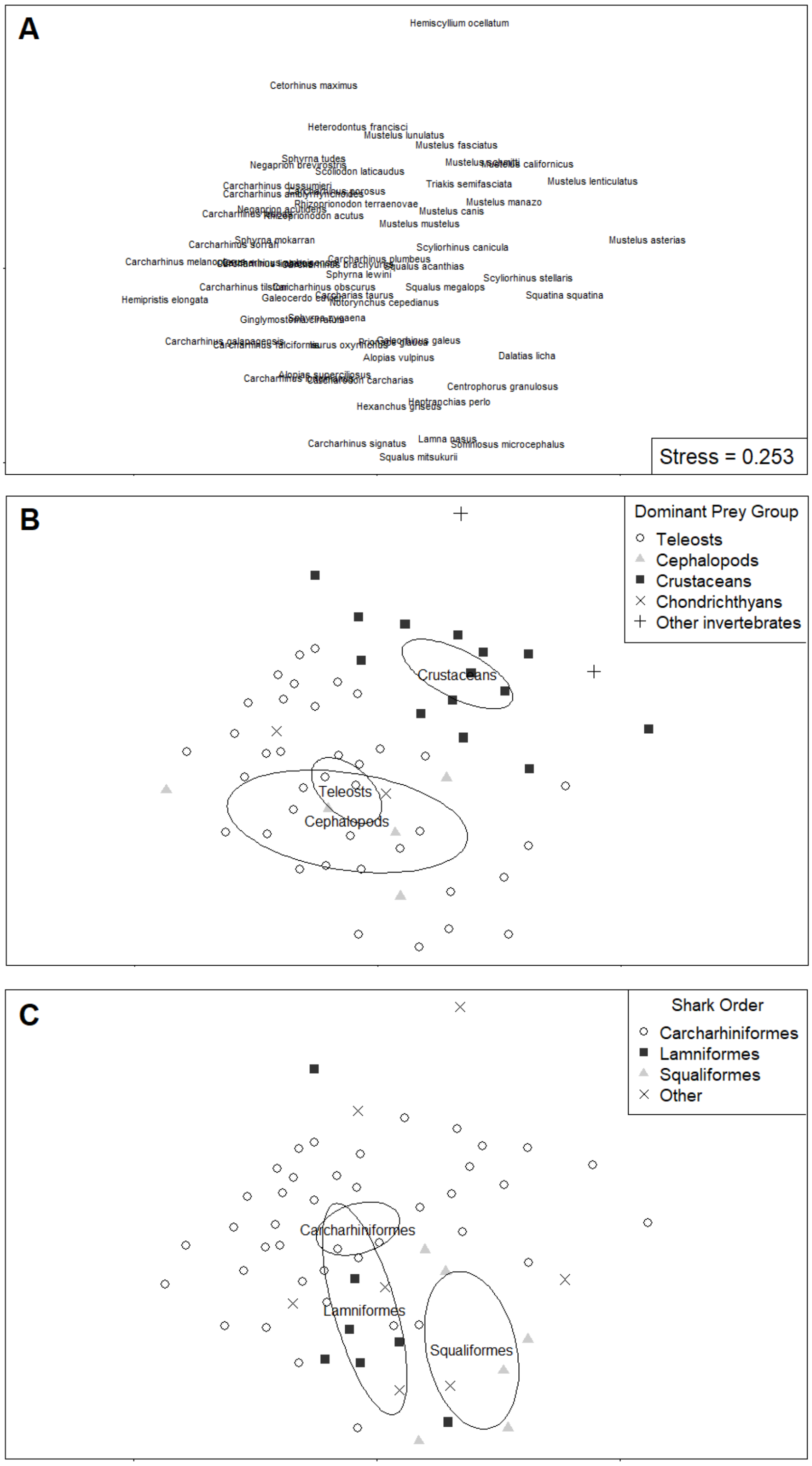

Figure 3.1. Non-metric multidimensional scaling (nMDS) plots displaying the similarity in diet (prey family) composition among shark species (n=61), grouped by shark species (A), shark diet composition (B), and shark Order (C). Distances are based on Jaccard similarities of spatial turnover among shark species. In B and C, 95% CI ellipses are displayed to compare groups.

The tapeworm family assemblages of sharks were substantially different among dominant prey groups (ADONIS; R^2=0.167, p<0.001) (Figure 3.2B). However, pairwise comparisons revealed that there was only a significant difference in tapeworm composition between crustacean-feeding and teleost-feeding sharks (ADONIS; R^2=0.430, p<0.001), and not between crustacean-feeding and cephalopod-feeding sharks (ADONIS; R^2=0.099, p=0.162), or teleost-feeding and cephalopod-feeding sharks (ADONIS; R^2=0.022, p=0.487). These differences can be clearly seen in the associated nMDS plot (Figure 3.2B), which shows relatively distinct groupings for teleost-feeding and crustacean-feeding sharks, with cephalopod-feeding sharks in-between and overlapping both groups. Notably, the two chondrichthyan-feeding sharks overlapped with teleost-feeding sharks in their tapeworm composition, whereas the two sharks feeding on other invertebrates were closest to crustacean-feeding sharks (Figure 3.2B).

SIMPER analysis revealed that the five tapeworm families contributing most to dissimilarity between teleost-feeding and crustacean-feeding sharks were the diphyllidean family Echinobothriidae (cumsum=0.116, p<0.001), the trypanorhynch families Lacistorhynchidae (cumsum=0.212, p=0.245) and Tentaculariidae (cumsum=0.300, p=0.728), the phyllobothriidean family Phyllobothriidae (cumsum=0.388, p=0.682), and the onchoproteocephalidean family Onchobothriidae (cumsum=0.473, p=0.771). Echinobothriidae was particularly common in the tapeworm assemblages of crustacean-feeding sharks (harboured by 71.4% of crustacean-feeding sharks) but completely absent from assemblages of teleost feeding sharks. Tentaculariidae and Phyllobothriidae were also more common in crustacean-feeding sharks (harboured by 71.4% and 85.7% of crustacean-feeding sharks, respectively) than in teleost-feeding sharks (harboured by 57.9% and 55.2%, respectively). On the other hand, Lacistorhynchidae and Onchobothriidae were more common in teleost-feeding sharks (harboured by 71.1% and 44.7%, respectively) than in crustacean-feeding sharks (each harboured by 42.9% of crustacean-feeding sharks).

Tapeworm family composition showed a significant difference among shark orders (ADONIS; R^2=0.319, p<0.001) (Figure 3.2C). Pairwise comparisons demonstrated that tapeworm compositions were significantly different between carcharhiniform and lamniform sharks (ADONIS; R^2=0.145, p<0.001), and between carcharhiniform and squaliform sharks (ADONIS; R^2=0.122, p<0.001), but not between lamniform and squaliform sharks (ADONIS; R^2=0.132, p=0.203). SIMPER analysis revealed that the five tapeworm families contributing most to dissimilarity between carcharhiniform and lamniform sharks were all trypanorhynch families; Sphyriocephalidae (cumsum=0.110, p<0.001), Tentaculariidae (cumsum=0.200, p=0.331), Lacistorhynchidae (cumsum=0.289, p=0.388), Otobothriidae (cumsum=0.374, p=0.260) and Gymnorhynchidae (cumsum=0.452, p<0.001). Sphyriocephalidae and Gymnorhynchidae were significantly more common in lamniform sharks (71.4% and 57.1%, respectively) compared with carcharhiniform sharks (each harboured by only 2.4% of carcharhiniform sharks). Conversely, Tentaculariidae, Lacistorhynchidae and Otobothriidae were all more common in the assemblages of carcharhiniform sharks (73.2%, 65.9% and 51.2%, respectively) than in the assemblages of lamniform sharks (42.9%, 42.9% and 14.3%, respectively). SIMPER analysis determined that the five tapeworm families contributing most to tapeworm dissimilarity between carcharhiniform and squaliform sharks were the trypanorhynch families Tentaculariidae (cumsum=0.116, p=0.002), Lacistorhynchidae (cumsum=0.216, p=0.023), Sphyriocephalidae (cumsum=0.313, p=0.004) and Otobothriidae (cumsum=0.405, p=0.045), and the phyllobothriidean family, Phyllobothriidae (cumsum=0.492, p=0.216). Tentaculariidae, Lacistorhynchidae and Otobothriidae were all more common in carcharhiniform sharks (73.2%, 65.9%, and 51.2%, respectively) than in squaliform sharks (16.7%, 33.3%, and 0.0%, respectively). In contrast, Sphyriocephalidae and Phyllobothriidae were more common in squaliform sharks (50.0% and 66.7%, respectively) than in carcharhiniform sharks (2.4% and 63.4%, respectively).

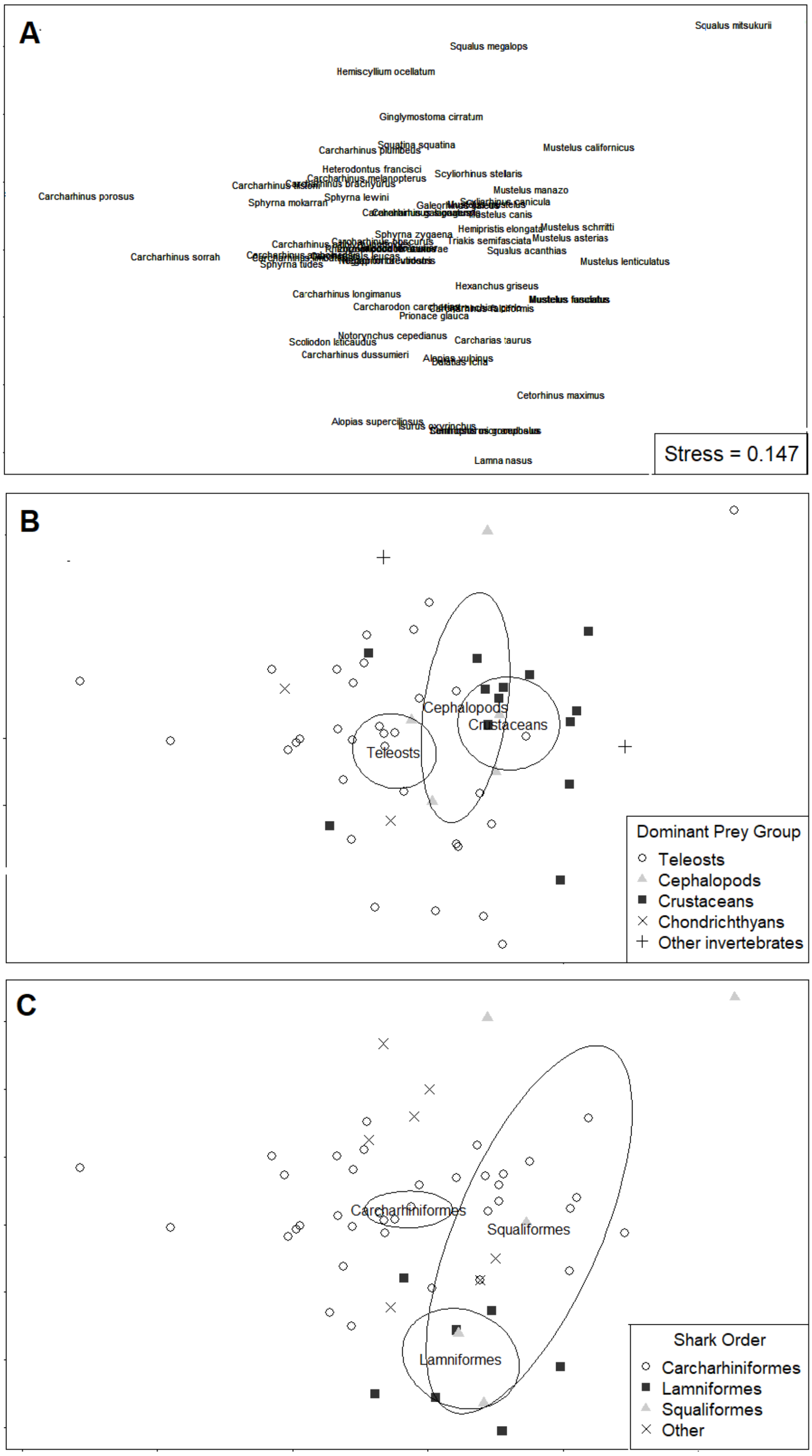

Figure 3.2. Non-metric multidimensional scaling (nMDS) plots displaying the similarity in tapeworm family composition among shark species (n=61), grouped by shark species (A), shark diet composition (B), and shark order (C). Distances are based on Jaccard similarities of spatial turnover among shark species. In B and C, 95% CI ellipses are displayed to compare groups.

Overall, dissimilarity in tapeworm family composition was significantly positively correlated with dissimilarity in prey family composition (Mantel R^2=0.118, p<0.001, permutations=999). In other words, tapeworm family assemblages were more similar in composition between sharks with similar diets (Figure 3.3).

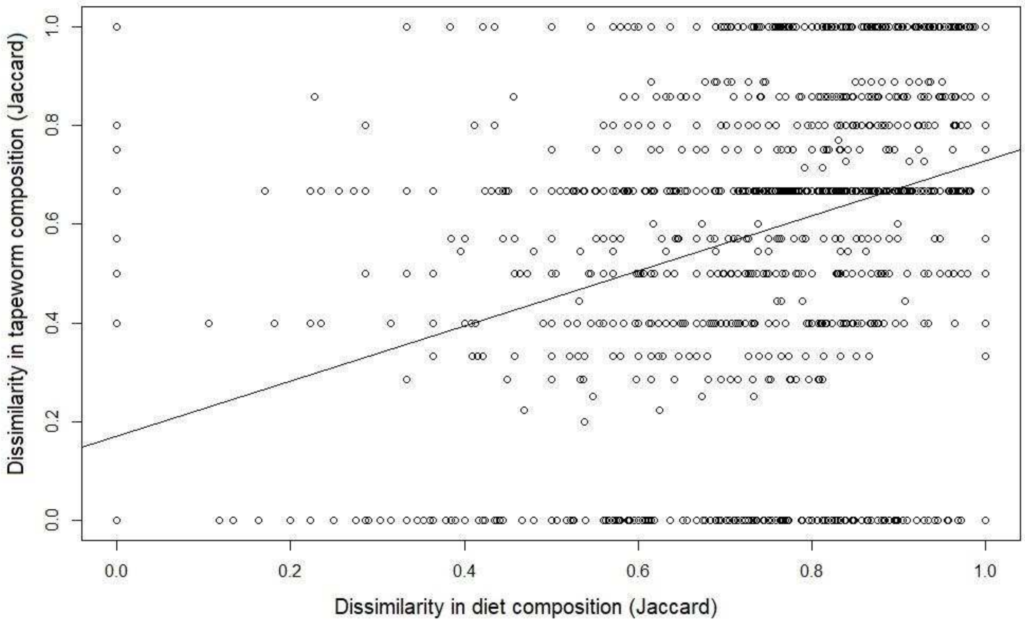

Figure 3.3. Scatter plot showing the relationship between similarity in diet composition (prey family assemblages) and similarity in tapeworm composition (tapeworm family assemblages) among sets of shark species (n=61 sharks, 3721 comparisons). Dissimilarities were derived using the Jaccard index and specifically reflect spatial turnover. Solid line shows linear regression between the two variables.

3.3.2. Linking trypanorhynch intermediate hosts with shark diet composition

Data on diet and tapeworm composition was obtained for a total of 91 different shark species and across these shark species there were 139 different trypanorhynch tapeworm species described as adults from spiral valves. Seventy-four (53.2%) of these trypanorhynch species had one or more families of marine animals described as their intermediate hosts in the literature. Overall, these trypanorhynch larvae were recorded across 186 different families of marine animals (Appendix Table A.2). Most of these intermediate host families were teleost fishes (n=134), and the remainder were comprised of families of chondrichthyan fishes (n=23), crustaceans (n=16), cephalopods (n=7), other molluscs (n=3) and reptiles (n=3). After cross referencing trypanorhynch intermediate host families with families recorded from the diets of the 91 shark host species analysed, a sum of 613 animal families could be compared for their use as intermediate hosts of shark trypanorhynchs and their use as prey by sharks.

The number of trypanorhynch species described to infect prey families as larval stages ranged from 0 in most families, up to more than 20 species in some families of teleost fishes. Once this number was corrected by each family's associated sampling effort (using residuals for the quadratic regression of trypanorhynch species richness on sampling effort), it was revealed that the families with the most trypanorhynch species larvae described per sampling effort were teleost fishes of the Order Perciformes (Figure 3.4A). In fact, of the 20 families with the highest species richness of shark trypanorhynch larvae (per sampling effort), 13 families were perciform fishes, five were families of teleost fishes from other orders, and only two families were of other taxonomic groups (elasmobranchs and cephalopods, respectively) (Figure 3.4A). There was considerable overlap between the most trypanorhynch-rich intermediate hosts and the most common prey of sharks (Figure 3.4B). Seven of the 20 families most rich in shark trypanorhynch tapeworm larvae (Carangidae, Sciaenidae, Scombridae, Trichiuridae, Serranidae, Merlucciidae, and Ommastrephidae) were also observed among the twenty most common prey families recorded from the diets of sharks (Figures 3.4A & 3.4B). Similarly, there was considerable overlap between the most trypanorhynch-rich families and the families most commonly comprising important shark prey (important prey families were defined as one of the five prey families with overall highest composition in a shark's diet) (Figures 3.4A & 3.4C). In contrast with the 20 families most rich in shark trypanorhynch tapeworm larvae, there was more variation in taxonomy among the 20 families most common as shark prey and important shark prey, with several families of cephalopods and crustaceans being much more common as shark prey than as intermediate hosts to their trypanorhynch tapeworm larvae (Figure 3.4).

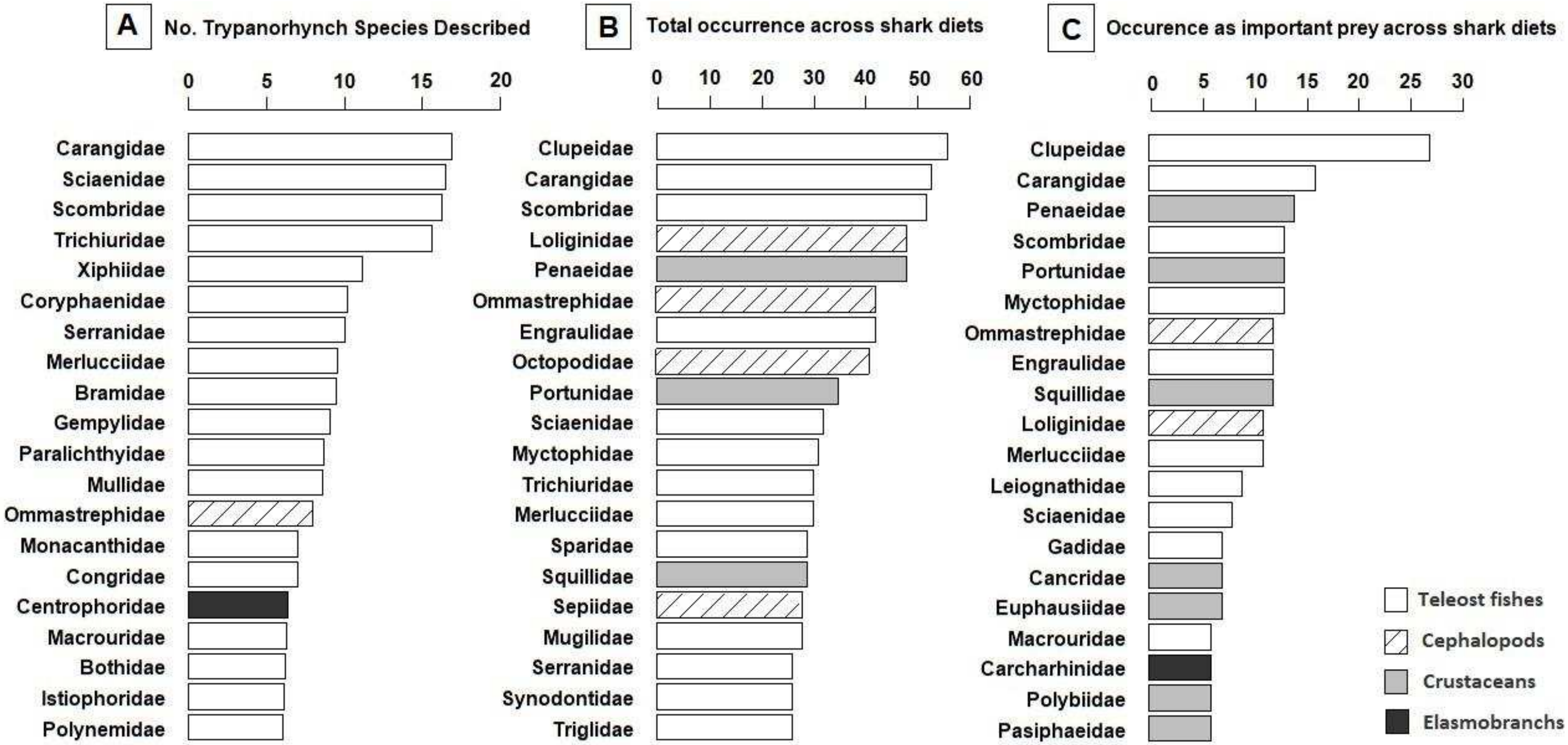

Figure 3.4: Frequency distributions showing the twenty animal families with (A) the most described larval trypanorhynch tapeworm species known to infect 91 different shark species as adults (adjusted to show highest numbers of trypanorhynch tapeworm species described per sampling effort), (B) the highest total occurrence as prey across the same 91 shark species, and (C) the highest occurrence as important prey across the same 91 shark species, defined by their frequency as one of the top five prey families by overall composition in shark's diets. Legend (lower right) gives the higher taxonomic groupings of prey families.

For the LMM looking at factors influencing richness of shark trypanorhynch tapeworm larvae, the full model (AICc = 2600.139) was considerably better than the null model (AICc = 2817.743) and the model including the random effect only (AICc = 2740.764). The evolutionary history of intermediate host families (measured as prey order nested within prey class nested within prey phylum) was found to explain a considerable amount of the variance in richness (11.39%). Of the fixed effects, occurrence as shark prey had a significant positive influence on trypanorhynch richness (t_{448} = 8.315, P<0.001), but occurrence as important shark prey, and the interaction between the fixed effects, were both statistically non-significant (Table 3.1).

Table 3.1: Summary of Linear mixed model (LMM) showing the effects of variables on the number of shark Trypanorynch tapeworm species (corrected for sampling effort) known to infect various families of marine animals as larval stages. Frequency of shark prey is the overall number of occurrences of an animal family across the diets of 91 different shark species, and frequency as important shark prey represents the number of occurrences in the top five prey families of sharks by composition. The model estimates, t-values, degrees of freedom (df) and p-values are shown for each variable.

Random effect

Variable	Number of levels	Variance explained (%)
Prey Phylogeny (Order/Class/Phylum)	(n=162)	11.39%

Fixed effects

Variable	**Estimate**	**t-value**	**df**	**p-value**
Occurrence as shark prey	0.138	8.315	448	<0.001
Occurrence as important shark prey	0.010	0.099	448	0.921
Fixed effects interaction	0.000	0.086	448	0.932

It was found that the average trypanorhynch tapeworm richness across a shark’s most important prey (top 5 prey families by composition) was positively associated with the overall richness of trypanorhynch tapeworms known to infect the shark (t_{69}=2.592, P=0.012) (Figure 3.5). In other words, sharks infected by high numbers of trypanorhynch species as adults appeared to feed often on families of prey harbouring more trypanorhynch species as larval stages. However, it must be noted that this association was relatively weak overall (R^2=0.089), and there was considerable variation among sharks (Figure 3.5).

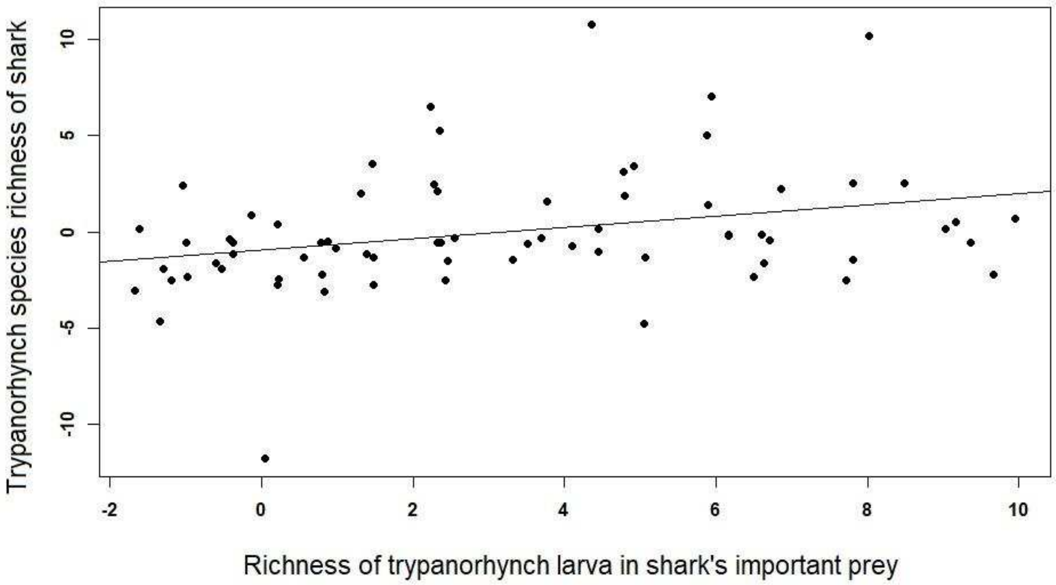

Figure 3.5. Scatter plot showing the relationship between number of trypanorhynch tapeworm species infecting shark species and number of trypanorhynch tapeworm species infecting their important prey families as larval stages (important prey families are defined as families that are in the top five families by overall composition in the shark's diet). Both measures of trypanorhynch tapeworm richness represent the residuals for their quadratic regressions on sampling effort.

3.4. Discussion

The results of this study reveal some important links between the diet of sharks and their tapeworm assemblages. First, tapeworm families were found to have substantially more overlap between shark species with similar diets. This pattern was demonstrated by a significantly positive correlation between shark similarities in tapeworm composition and similarities in diet composition (Figure 3.3). Additionally, nMDS analyses revealed distinct differences in shark species' tapeworm compositions depending on what taxonomic group of prey was dominant in their diets (Figure 3.2B). Taken together, these findings support the notion that tapeworms may be useful indicators of the feeding ecology of their definitive shark hosts, and vice versa (Palm et al. 2017). However, these findings must also be viewed in light of host evolutionary histories. Tapeworm compositions showed significant differences among sharks of different taxonomic orders (Figure 3.2C), and shark diets were also largely dependent on their evolutionary history (Figure 3.1C). These relationships make it difficult to decipher the relative impacts of host ecology and evolutionary history on the structure of shark tapeworm assemblages. Ultimately, tapeworm assemblages may be similar between related shark species partly due to their shared evolutionary history with tapeworms, and partly due to the fact that they feed on the same type of intermediate hosts. Nevertheless, host ecology is evidently more important in some cases. In the present study, for example, there was substantial variation in tapeworm families among different lamniform shark species, and some lamniform sharks were shown to have tapeworm compositions closer to those of some squaliform shark species with similar feeding ecologies than those of other lamniform sharks. Likewise, other research has found that trypanorhynch tapeworms show substantial overlap among ecologically similar shark species even when they are phylogenetically-distant hosts (Palm et al. 2017). These instances likely reflect the occurrence of host switches between shark species with different evolutionary histories (Palm et al. 2017). Phylogenetic research of trypanorhynch tapeworms suggests that host switches have occurred for tapeworms even between exceptionally different elasmobranchs e.g. host switches between sharks and rays (Palm et al. 2009).

3.4.1. The influence of sharks' diets on their tapeworm compositions

Tapeworm families were observed to be remarkably different between crustacean-feeding and teleost-feeding sharks in this study (Figure 3.2B). This pattern is likely the result of life-cycle differences among tapeworm families, since some tapeworm taxa appear to exclusively use crustaceans as second intermediate hosts, whereas others may only use teleosts as second intermediate hosts (see Palm 2004). For illustration, the trypanorhynch family Lacistorhynchidae was observed to be much more common among teleost-feeding shark species than crustacean-feeding species, which is consistent with previous findings that tapeworms of this family generally use teleosts as their second intermediate hosts (Palm 2004; Jakob & Palm 2006; Jensen

2009; see also Appendix Table A.2). In contrast, diphyllidean tapeworms of the family Echinobothriidae were observed to be very common in crustacean-feeding shark species, but completely absent from teleost-feeding sharks. A majority of the echinobothriid tapeworms infecting sharks in this study were of the genus Coronocestus, for which intermediate hosts are completely unknown. In other genera of echinobothriid tapeworms, crustaceans and molluscs have been described as second intermediate hosts (Vivares 1971; Cake 1976; Tyler 2006), and based on findings here, it is likely that Coronocestus species also use these invertebrates as second intermediate hosts. Phyllobothriid tapeworms were also more common in crustacean-feeding sharks than teleost-feeding sharks. However, larval stages of phyllobothriid tapeworms have been recorded from a range of different marine fauna including crustaceans (Vivares 1971), mammals (Aznar et al. 2007), teleost fishes, and molluscs (cephalopods and bivalves) (Jensen 2009; Jensen & Bullard 2010; Randhawa & Brickle 2011). Notably, these records from different fauna involve different phyllobothriid genera, indicating that different taxa within this family could have very different life cycles. Certain phyllobothriid genera may typically use crabs as second intermediate hosts (Vivares 1971), whereas others infecting primarily lamniform sharks (e.g. Clistobothrium) use squid as second intermediate hosts (Randhawa & Brickle 2011). However, further research is needed to elucidate their poorly studied life cycles.

The families Tentaculariidae and Onchobothriidae showed minor differences in their presence between teleost-feeding and crustacean-feeding sharks (tentaculariids were present more in crustacean-feeders, whereas onchobothriids were slightly more common in teleost-feeders), and overall both families were relatively common amongst sharks of both feeding types. This is perhaps unsurprising for tentaculariid tapeworms, which generally use teleosts as second intermediate hosts, but show cosmopolitan distributions across a range of other marine animals including cephalopods, other molluscs, chondrichthyans and reptiles (Palm 2004; Palm & Caira 2008; Jensen 2009; see also Appendix Table A.2). Some genera within Tentaculariidae are probably more host specific than others, but most members of this family have low host specificity, and some species (e.g. Tentacularia sp.) appear to have flexible life cycles with many potential paratenic hosts (Palm 2004; Palm & Caira 2008). The typically low host specificity of these species perhaps explains why they are common across shark species with very different feeding habits. Interestingly, onchobothriid tapeworms are characterised by a high degree of host specificity (Caira & Jensen 2001; Caira et al. 2001), but are also common across sharks with different feeding habits. As described above for Phyllobothriidae, it is likely that different genera within this family have contrasting life-cycles. For example, the genus Acanthobothrium is found in crustacean-feeding carcharhiniform sharks (Mustelus and Scyliorhinus spp.), and some species in this genus have been found to commonly use benthic molluscs (gastropods and bivalves) as intermediate hosts (although it must be noted that there is a paucity of information on the life cycles of most Acanthobothrium species) (Cake 1976; Holland & Wilson 2009; Jensen 2009). Sharks with diets dominated by crustaceans are generally benthic foragers that often eat families of bivalves and gastropods (see Mustelus and Scyliorhinus species in Appendix Table A.1 for examples), and thus, crustacean-feeding sharks are likely to also frequently encounter tapeworm larval stages from these mollusc intermediate hosts. On the other hand, the onchobothriid genus Phoreiobothrium is known from teleost-feeding carcharhiniform sharks (e.g. Carcharhinus and Sphyrna spp.) and has been recorded to generally utilise teleost second intermediate hosts (Jensen & Bullard 2010). Based on these different associations, the feeding ecology of shark hosts could provide useful insights for onchobothriid genera with completely unknown life cycles. For instance, intermediate hosts have not been described for tapeworms of the genus Platybothrium (Healy 2003), but given that tapeworms of this genus are common in teleost-feeding sharks (e.g. Carcharhinus spp. and Sphyrna spp.) and are frequently found in co-infections with species of Phoreiobothrium (Caira et al. 2017), they most likely use teleosts as intermediate hosts.

The nMDS analyses revealed that tapeworm families of cephalopod-feeding sharks were not distinctly different from those of either teleost-feeding or crustacean-feeding sharks (Figure 3.2B). This pattern may be in part due to the low sample size here for sharks feeding primarily on cephalopods (n = 5). However, there was also strong overlap in the prey families of cephalopod-feeding and teleost-feeding sharks (Figure 3.1B), indicating that sharks with these different dominant prey groups have very similar diets overall. This pattern was also anecdotally supported by the diet composition data obtained for this study, which showed that the diet of sharks feeding mainly on cephalopods typically had high overall compositions of teleost fishes (15-30%). Pelagic cephalopods are commonly intermediate hosts to trypanorhynch tapeworms that also use teleost second intermediate hosts (Palm 2004; Appendix Table A.2), and many studies suggest that cephalopods are frequent paratenic hosts for these trypanorhynch tapeworms (Stunkard 1977; Brickle et al. 2001; Shukhgalter & Nigmatullin 2001; Palm et al. 2017). Hence, tapeworm assemblages may not be distinctly different between cephalopod-feeding and teleost-feeding sharks since cephalopods are often used as second intermediate hosts (facultative or obligate) and paratenic hosts in the same life-cycles as teleost fishes. Notably, this would not explain why no significant difference was observed in tapeworm compositions between cephalopod-feeding and crustacean-feeding sharks. The diet composition data showed that cephalopod-feeding sharks had variable, but generally lower diet compositions of crustaceans (0-15%), compared to teleosts (15-30%). Based on this, there is less support for cephalopods and crustaceans being intermediate or paratenic hosts in the same life cycles. However, given the current paucity of information on marine tapeworm life cycles, further investigation is needed.

Tapeworm families showed strong distinctions among the different shark Orders, with significant differences observed between carcharhiniform and lamniform sharks, as well as between carcharhiniform and squaliform sharks. The analyses revealed that carcharhiniform sharks were characterised by the trypanorhynch tapeworm families Tentaculariidae, Lacistorhynchidae and Otobothriidae. These findings mirrored those of Palm et al. (2017), who found that genera within these and other trypanorhynch families could be useful indicators of the ecology of carcharhiniform shark hosts. For example, the genera Tentacularia and Heteronybelinia (Tentaculariidae), Otobothrium (Otobothriidae), Callitetrarhynchus, Floriceps and Dasyrhynchus (Lacistorhynchidae) are all typical of pelagic teleost-feeding carcharhiniform sharks, whereas the genera Dollfusiella and Trigonolobium (Eutetrarhynchidae), Lacistorhynchus and Diesingium (Lacistorhynchidae) are all typical of benthic invertebrate-feeding carcharhiniform sharks (Palm et al. 2017). Although these disparities are also underpinned by further phylogenetic differences within the Order e.g. sharks of the Family Carcharhinidae are typically teleost-feeding sharks, whereas sharks of the Family Triakidae are typically invertebrate-feeding sharks (Palm et al. 2017), these tapeworm genera could clearly be very useful for predicting the general feeding habits of a host species. To illustrate this, there is a paucity of information on the feeding habits of the triakid shark Mustelus mento (Fishbase 2017), but this species is known to harbour tapeworms of the trypanorhynch genera Lacistorhynchus (Gibson et al. 2005) and Dollfusiella (Alves et al. 2017), which both suggest that this shark has a diet dominated by benthic invertebrates. In a similar vein, the feeding habits of a shark host could be used for making inferences about their tapeworm assemblages. For example, to my knowledge the carcharhinid shark Rhizoprionodon longurio has not yet been studied for tapeworm parasites, but it is known to feed mainly on pelagic teleost fishes (Márquez-Farías et al. 2005). From this we could infer that the tapeworm assemblage of this shark is likely to include tapeworms of the aforementioned genera common to teleost-feeding carcharhiniform sharks.

Both squaliform and lamniform sharks were characterised by the trypanorhynch tapeworm family Sphyriocephalidae (which was rare in carcharhiniform sharks). The common occurrence of this family across both lamniform and squaliform sharks may at least partially explain why tapeworm compositions were overall not significantly different among sharks of these orders. Tapeworms of this family are known to use teleost fishes, elasmobranchs, and cephalopods as intermediate or paratenic hosts (Jensen & Bullard 2010; Dallarés et al. 2017; Appendix Table A.2). Interestingly, sphyriocephalid tapeworms have also been described from many of the same intermediate host species as tapeworms characteristic of carcharhiniform sharks (species of tentaculariids, lacistorhynchids and otobothriids) (see Appendix Table A.2). Considering this overlap in intermediate hosts, one might ask why sphyriocephalid tapeworms are not common in carcharhiniform shark species too. One possibility is that these tapeworms might be incompatible with carcharhiniform sharks due to differences in immune response (McVicar & Fletcher 1970; Randhawa & Burt 2008) or other filters that have yet to be examined in this host-parasite system. However, this can only be speculated without further investigation. Importantly, the patterns of host use established here are based on small subsets of shark species from all orders other than Carcharhiniformes, and may become much clearer with larger sample sizes. This will require increased sampling of sharks’ diets and parasite assemblages across different orders in future.

3.4.2. Linking trypanorhynch intermediate hosts with shark diet composition

One of the primary aims of this study was to investigate links between intermediate host use by trypanorhynch tapeworms and the diets of their definitive shark hosts. The present study demonstrated considerable overlap between the most trypanorhynch-rich families of intermediate hosts and the most common prey families of sharks (Figure 3.4). Moreover, the occurrence of animal families as shark prey was overall statistically significant and positively correlated with the number of trypanorhynch species known to infect them (adjusted by the family's sampling effort for parasites) (Table 3.1). Based on these findings, trypanorhynch tapeworms of sharks appear to use intermediate hosts which have a high likelihood of being consumed by shark definitive hosts. From an evolutionary standpoint, this could suggest that these tapeworms increase the probability of completing their life cycles by infecting intermediate hosts which have strong trophic links to suitable definitive hosts (Cirtwill et al. 2017). However, this is not to say that trypanorhynch tapeworms specifically target prey with strong trophic links to their definitive shark hosts. Notably, many of the families of marine animals that are important hosts of trypanorhynch larvae are also some of the most common, speciose, and widespread animals across marine ecosystems (e.g. Scombridae and Sciaenidae (Nelson et al. 2016)), and consequently, are likely to be frequently encountered by sharks. Thus, it is possible that the links observed between intermediate host use in trypanorhynch tapeworms and shark's diets are a product of the wide distribution and abundance of these prey families (see Cirtwill et al. 2017).

The evolutionary history of an animal has an important influence on its use by trypanorhynch tapeworm species (Table 3.1). In line with previous records, families of teleost fishes were by far the most common intermediate hosts for shark trypanorhynch tapeworms (Palm 2004; Jensen & Bullard 2010), and made up more than two thirds of all described intermediate host families in the present study. Fishes of the Order Perciformes were particularly important intermediate hosts (Figure 3.4a, which as noted above, may partly be a consequence of the size, abundance and wide distributions of members within the Order. Families of perciform fishes could also be more suitable intermediate hosts for phylogenetic reasons e.g. they may provide more adequate resources for larval development. Although most trypanorhynch tapeworms exhibit low host specificity as larval stages, some species (e.g. Gymnorhynchus gigas) appear to have higher host specificity as larvae, perhaps due to such phylogenetic constraints (Palm & Caira 2008). Nevertheless, trypanorhynch tapeworm patterns of host use in teleosts are almost certainly more influenced by the ecology, rather than the phylogeny, of their intermediate hosts (Jakob and Palm 2006). Based on the comparative findings here it is difficult to establish the extent to which phylogenetic constraints might influence patterns of host use, and further insights would require experimental infection studies which could test compatibility of larval tapeworms among different potential intermediate hosts. To my knowledge, no such studies have been conducted to date.

The average richness of trypanorhynch tapeworms (larval stages) across a shark's preferred families of prey was found to be positively associated with the richness of adult trypanorhynch tapeworm species known to infect the shark (Figure 3.5). However, it must also be cautioned that the overall association between these factors was relatively weak ($r^2 = 0.089$), and there was a large amount of variation among different species. Many factors might account for this variation, including all the factors analysed within chapter two of this thesis (e.g. the phylogeny, size, trophic level, and overall diet breadth of the shark), as well as factors which have not yet been examined, such as host-parasite compatibility. It is also possible that some of this variability is the result of limitations in the diet records obtained for this study. The overall diet compositions of shark species here reflect the sum of their feeding habits across all examined individuals for a species, regardless of their sex, size and locality. Unfortunately, many further diet studies of shark species would be required to assess the influence of these factors because a substantial amount of currently published records have not provided data on shark's feeding habits for each of these sub-populations. Furthermore, it is very uncommon for studies of elasmobranch stomach contents to include information on the intestinal parasites of individuals (see the following for some notable exceptions; Klimpel et al. 2003; Gracan et al. 2014; Isbert et al. 2015).

3.4.3. Conclusions

The present study revealed several important links between the diets of shark species and their tapeworm parasite assemblages. Firstly, there was substantial overlap found between the tapeworm families infecting a shark species and the prey families comprising their diet. Second, it was established that certain tapeworm lineages may serve as useful indicators of the feeding habits and evolutionary history of shark host species. Finally, it was observed that trypanorhynch tapeworm species that mature in sharks often use intermediate hosts which are commonly preyed upon by shark hosts. As a whole these findings demonstrate that both evolutionary history and ecology have important influences in shaping the tapeworm parasite assemblages of shark species. Importantly, the patterns of host use established here are conspicuously biased towards shark species of the Order Carcharhiniformes. Sharks of this order are generally the most well-studied and are arguably some of the most important, abundant and widespread species inhabiting our oceans (Compagno 2001). However, to further our understanding of the evolution of parasitic life cycles in sharks, it is recommended that sampling of diets and parasite assemblages prioritise shark species across more different Orders in future.

Chapter 4: Analysing host diet and parasites in a model species; the draughtsboard shark *Cephaloscyllium isabellum*

4.1. Introduction

Parasite species demonstrate substantial heterogeneity in their distributions among host species, and consequently, certain hosts harbour disproportionately more parasite species than others (Poulin 2011). This heterogeneity in parasite diversity among host species is underpinned by a number of ecological and phylogenetic factors, which act to restrict the range of hosts that can be used by parasite species i.e. their host specificity (Combes 2001; Poulin 2011; Poulin et al. 2011a). In 1980, Euzet and Combes proposed that these determinants of host use can be thought of as two types of "filters"; namely encounter filters, which are factors that prevent physical contact between hosts and parasites, and compatibility filters, which are factors that prevent parasites from forming an association with hosts, even if they do come in contact (Euzet & Combes 1980; Combes 2001). Testing the relative strength of encounter and compatibility filters has since become a common feature of studies aiming to understand patterns of host use by different parasites (Kuris et al. 2007; Lagrue et al. 2011; Medeiros et al. 2013). By assessing the relative importance of these filters, researchers may gain key insights towards understanding, predicting and controlling outbreaks of infectious diseases in the future (Poulin 1992; Taraschewski 2006; Lootvoet et al. 2013).

A common method used to test the relative importance of compatibility and encounter filters is to control for the encounter filter via experimental infection of a host with a parasite it does not naturally encounter (Komar et al. 2003; Perlman & Jaenike 2003; Kuris et al. 2007; Dick et al. 2009; Van Oosten et al. 2016). A number of studies employing this method have established the importance of encounter filters, showing that many parasites are compatible with hosts that they don't normally come in contact with (Komar et al. 2003; Perlman & Jaenike 2003; Dick et al. 2009). However, this is not to say compatibility filters are less important in preventing parasite infections. Studies have also demonstrated substantial variation in the compatibility of parasites among different host species, with compatibility suggested to depend heavily on the taxonomic distance between hosts (Komar et al. 2003; Perlman & Jaenike 2003; Medeiros et al. 2013). Unfortunately, assessing the importance of encounter and compatibility filters relative to each other has proven more challenging, largely due to the difficulty of quantifying host-parasite encounters

(Combes 2001). Although the natural distribution of parasites in different hosts gives some clue as to how many parasites are encountered by a host, the full extent of encounters cannot be estimated without knowing how many parasites are encountered that fail to infect the host (Kuris et al. 2007). Researchers must address this issue and look for ways to reliably measure host-parasite encounters in order to gain a better understanding of the relative roles of encounter and compatibility filters in different ecosystems.

Tapeworms (cestodes) are arguably the most important group of parasites infecting elasmobranchs. These parasites are known to have typically high host specificity as adults (are limited to one or a few host species) and showcase an incredible diversity of species across various elasmobranch hosts (Caira & Healy 2004; Caira & Jensen 2010). Several studies have demonstrated that the high specificity of tapeworms in elasmobranchs is at least partially the consequence of compatibility filters. For example, immune response in elasmobranchs has been shown to reject certain tapeworm species (McVicar & Fletcher 1970; Randhawa & Burt 2008). On the other hand, the importance of encounter filters for tapeworm infection is generally unknown, and it is possible that some encounter filters, such as host diet, are more restrictive for tapeworm establishment than compatibility filters (Randhawa et al. 2008). These tapeworms all have complex life-cycles, involving at a minimum three different host species, and are trophically-transmitted to elasmobranchs from their intermediate or paratenic host prey (Willams 2002), and thus, it would not be surprising for their diet to play a large role in restricting parasite encounters.

The draughtsboard shark Cephaloscyllium Isabellum (Bonnaterre, 1788) is a scyliorhinid catshark species, of the Order Carcharhiniformes, endemic to New Zealand (Cox & Francis 1997). This species is abundant around New Zealand’s continental shelf, and acts as a demersal hunter and scavenger, foraging down to depths of 500 m (Francis et al. 2002; Horn 2016). Their diet is dominated by benthic invertebrates, but overall consists of a broad range of prey species, including but not limited to, fishes, crustaceans, molluscs, tunicates, echinoderms and priapulid worms (Horn 2016). Despite their broad diet, however, only one tapeworm species, Calyptrobothrium chalarosomum, has been described from C. isabellum (Hewitt and Hine 1972; Hine et al. 2000). This is interesting given that most other studied sharks harbour several different tapeworm species on average (Randhawa and Poulin 2010). Moreover, C. isabellum has been identified as a cold spot in shark tapeworm diversity, entailing that it is infected by fewer tapeworm species than expected for a shark of its size (~1m in length) (Randhawa 2014, unpublished data; see Poulin et al. 2011b for details on hot and cold spots in parasite diversity). In essence, this makes C. isabellum a good model for studying the importance of encounter and compatibility filters for elasmobranch tapeworms. On one hand, the shark species could be a coldspot for tapeworm diversity because it does not encounter other tapeworm species through its diet, and conversely, different tapeworm species may be encountered through its diet, but their infection of the shark could be inhibited by strong compatibility filters such as host immune response. The life cycle of the shark's known tapeworm C. chalarosomum is completely unknown, and the intermediate hosts used by this tapeworm could consist of any number of the many different species included in the shark's diet. It is also unknown whether any of its prey species harbour the larval stages of other tapeworm species, although given the breadth of its diet, there is a high possibility that other tapeworms are also encountered by the shark.

4.1.1. Objectives of chapter 4

The primary objective of this chapter was to provide some insight into the importance of encounter and compatibility filters for the tapeworm infection of the draughtsboard shark C. isabellum. This objective was addressed through a series of steps. The first step was to analyse the diet and parasites of C. isabellum individuals to get an idea of what prey the shark encounters, and thus determine which prey species might serve as intermediate hosts for tapeworms. Previous research has shown that the intermediate hosts of trophically transmitted parasites are not always important prey of their final host (Cirtwill et al. 2017). However, important shark prey were targeted for parasite examination in this study to give some indication of how many parasites C. isabellum might regularly encounter via its diet. The second step was to examine samples of the sharks favoured prey, to determine whether they host any larval tapeworms. This included looking for larval stages of the shark's only known tapeworm species, C. chalarosomum. The third and final step relied on whether any tapeworm larvae other than C. chalarosomum were recovered from the prey of C. isabellum. If other tapeworm larvae were found in prey, they were tested for their compatibility with the shark host via exposure to the host’s blood serum (following procedures described in Randhawa and Burt 2008).

4.2. Methods

4.2.1. Surveying parasites of *C. isabellum*

A total of 24 adult draughtsboard sharks were examined for this study. These sharks were trawled from approximately 80 metres deep by local fishermen off the coast of Otago, New Zealand, on two separate trips. Thirteen of the sharks were caught on the 25th of April 2015, between Curio Bay (46°66S, 169°10E), and Dunedin (45°87S, 170°54E). The other 11 sharks were caught on the 26th of June 2015, a few kilometres north of Nugget Point (46°44S, 169°82E). Individuals were collected from the fishermen immediately following each trip and transported to the laboratory to be necropsied without delay.

Prior to dissection, each individual shark was measured for total length (in cm) and sex was determined based on the presence of distinct claspers in males (Horn 2016). Following these measurements, the individuals were flipped upside down and an incision was made along the ventral body surface to allow inspection of the body and organs for any parasites. The entire spiral valve was removed, carefully cut open, and placed inverted into a 1L container filled with saline solution and 1 tsp of sodium bicarbonate (baking soda). This container was sealed and shaken vigorously for a few minutes to separate tapeworms from mucous, then left to sit for at least 2 hours. Any tapeworms not detached from the gut mucosa were extracted from the tissue using forceps. All recovered parasites were then placed in formalin to be later counted under a dissection microscope.

4.2.2. Diet analysis of *C. isabellum*

Once measured and surveyed for parasites, the stomach of each shark was removed and opened via an incision along the dorsal surface. Stomach contents were placed into a fine sieve (500µm mesh size), rinsed with tap water, and transferred into a container with 70% ethanol for later measurement and identification. Prey were identified under dissection microscope to the lowest taxonomic level possible. A range of available guides and taxonomic keys from literature were used to aid in identifications for crustaceans (Schembri & McLay 1983; Naylor et al. 2005; Ahyong 2012; Wilkens & Ahyong 2015), molluscs (Powell 1979) and teleosts (Lourie et al. 2004; Roberts et al. 2014). Prey items were counted and weighed (wet weight, to nearest .01 g) for subsequent quantitative analyses.

The diet composition of different shark prey groups was quantified by percentage number (%N), percentage mass (%M), and frequency of occurrence (%O). These indices were also used to calculate the index of relative importance [IRI = (%N + %M) × %O] (Cortés 1997). % IRI was calculated for the contribution of overall groups (teleosts, molluscs, crustaceans, unidentified organic matter and plants) as well as separately for individual prey within each group.

4.2.3. Surveying known prey items for parasites

Based on the quantitative diet analyses here, combined with findings of Horn's (2016) diet analysis on a larger subset of C. isabellum individuals, it was decided that parasite surveys of prey should focus on crab species, and especially, hermit crabs (Paguridae), which make up a considerable amount of the diet of C. isabellum. Most of the prey collected for this study were obtained on the 14th of March 2016 by the vessel "RV Polaris II" operating Northeast of Taiaroa Head, off Otago's Shelf (45°48S, 170°55E). More than a hundred individual hermit crabs, as well as small numbers ($n<25$) of several other crab species and ascidians, were caught as by-catch in beam trawls at 92 metres depth. A small sample ($n=10$) of long-legged masking crabs (Leptomithrax longipes) was obtained separately from local fishermen, who found the crabs in a trawl with draughtsboard sharks. This trawl was conducted on the 7th of December 2015 offshore Otago peninsula at 50 m depth (45°87S, 170°75E).

All invertebrates were kept alive on capture and transported to an aerated holding tank with seawater in Portobello Marine Lab, where they were surveyed for macroparasites within two weeks of capture. It was important they were kept alive so that any parasites were still alive upon inspection. Similar with the stomach contents of C. isabellum, invertebrates were identified to the lowest taxonomic level possible using guides and taxonomic keys from literature (Schembri & McLay 1983; Naylor et al.

2005; Ahyong 2012; Wilkens & Ahyong 2015). Tunicates were measured by length and dissected via longitudinal incision to reveal internal organs. Crabs were euthanised by piercing the cephalic ganglion with a knife, measured (carapace length) and determined for sex where possible. Following measurements, crabs were placed in a petri dish with seawater and the carapace was removed to expose organs for dissection. The full body was checked for macroparasites under a light dissection microscope and any recovered worms were placed in 5ml Eppendorf tubes containing 70% ethanol.

4.2.4. Host compatibility tests

Due to the complete absence of any tapeworm larval stages recovered from prey items, serological tests to analyse host compatibility (exposure of parasites to the host's blood serum) could not be carried out in this study.

4.3. Results

Of the 24 sharks caught in this study, there were 14 males and 10 females. The mean length of individuals was 72.40cm (± 6.25cm SD, range = 53.6-82.1cm). There was no significant difference in size between males (72.05 ± 3.81cm SD) and females (72.88 ± 8.85cm SD) (t^{22} = 0.31, p = 0.76), although size was notably more variable among female sharks.

4.3.1. Parasites of *C. isabellum*

All 24 draughtsboard sharks were infected by the tapeworm Calyptrobothrium chalarosomum in their spiral valves (100% prevalence), but no other tapeworm species were recovered from the sharks' intestines. In total, 1,609 adult C. chalarosomum were recovered. The mean abundance of tapeworms per shark was 67.04 (± 49.90 SD) and intensity ranged from 16 to 241 tapeworms per host. Sharks caught between Curio Bay and Dunedin in April generally had a higher intensity of infection (mean = 82.46 ± 60.17 SD) compared with sharks caught North of Nugget Point in June (mean = 48.82 ± 26.59 SD) (Figure 4.1). However, there was considerable variation among individuals from each location and the difference was statistically non-significant between the two samples (t^{22} = 1.71, p=0.10). Male sharks had generally higher intensity of infection (mean = 82.79 ± 58.15 SD) than females (mean = 45.00 ± 23.53 SD) (Figure 4.1), but there was a similarly high variation within each sex and the overall difference was marginally non-significant (t^{22} = 1.94, p=0.06). There was also no significant association between infection intensity and host length (Figure 4.2) (R^2 = 0.0003, p=0.94).

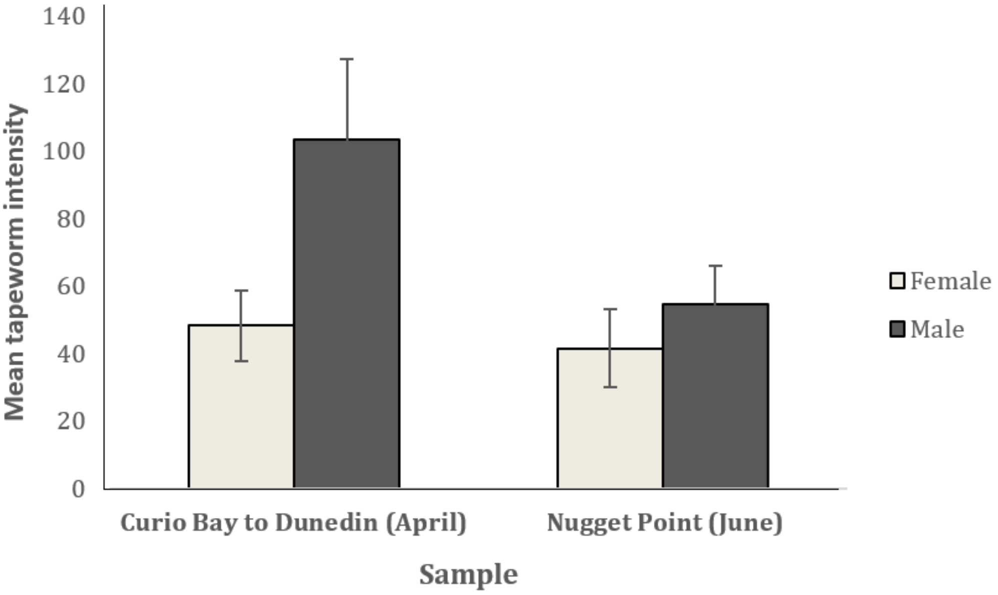

Figure 4.1. Mean infection intensity of the tapeworm *Calyptrobothrium chalarosomum* and the sex and sample of their draughtsboard shark hosts (*Cephaloscyllium isabellum*). Sample sizes for the Curio Bay to Dunedin sharks were (n=5) for females and (n=8) for males. Sample Sizes for the Nugget Point sharks were (n=5) for females and (n=6) for males. Error bars show ± 1 Standard Error.

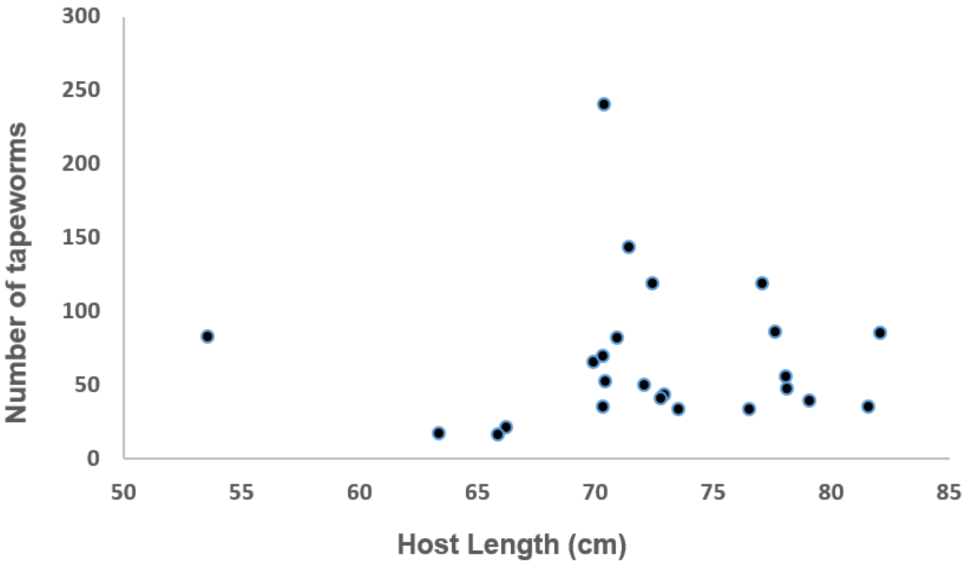

Figure 4.2. Relationship between the infection intensity of the tapeworm *Calyptrobothrium chalarosomum* and the total length (cm) of their draughtsboard shark hosts (*Cephaloscyllium isabellum*). There was no significant relationship between the variables (R^2 = 0.0003, p=0.94).

Although no parasite other than C. chalarosomum was recovered from the spiral valve of C. isabellum, there were a few parasites recovered from other parts of the shark. Five individuals were each infected by a single digenean trematode in the stomach (Prevalence = 20.83%). This trematode species was not identified, but could possibly be Otodistomum veliporum, which has been previously described from the stomach of C. isabellum (Hewitt & Hine 1972). Two female sharks (both from North of Nugget Point) also had a few (3 and 4) larval Anisakis sp. nematodes embedded in their body wall. The only other parasite recovered from C. isabellum was a singular immature (plerocercoid) tapeworm, identified as Hepatoxylon trichiuri (Palm 2004), which was found in the body cavity of a male shark caught North of Nugget Point.

4.3.2. The diet of *C. isabellum*

All 24 sharks sampled in this study had stomachs containing food. Stomach contents included remains from a variety of different taxa, including teleost fishes, bivalves, cephalopods, crabs and shrimps (Table 4.1). Overall, teleosts, molluscs and crustaceans were all similarly important prey groups in diet of C. isabellum, with their %IRI's being 29.69%, 38.39%, and 29.73%, respectively (Table 4.1). Crustaceans were the most important prey group by frequency of occurrence (%O) with 16 of the

24 sharks sampled (66.67%) containing some crustacean remains in their stomachs. Teleosts were most important in terms of overall mass (%M) (49.68% of total prey mass), and molluscs contributed the most to the diet by percentage in number (%N) (Table 4.1).

Table 4.1. Diet composition of 24 draughtsboard sharks (*Cephaloscyllium isabellum*) caught off the Coast of Otago, New Zealand. Composition is expressed as percentage in number (%N), mass (%M), percentage occurrence (%O), and by index of relative importance (%IRI).

Prey group	**Species**	**%N**	**%M**	**%O**	**%IRI**
Teleosts		**15.15**	**49.68**	**41.67**	**29.69**
Sea horse (Syngnathidae)	*Hippocampus abdominalis*	1.52	0.03	4.17	0.12
Unidentified teleost remains		13.64	49.37	37.5	42.29
Molluscs		**42.42**	**41.39**	**41.67**	**38.39**
Bivalve siphons (Hiatellidae)	*Panopea zelandica*	31.82	40.64	29.17	37.83
Unidentified Cephalopods		4.55	0.25	12.5	1.07
Unidentified Mollusc remains		6.06	0.5	12.5	1.47
Crustaceans		**33.33**	**7.24**	**66.67**	**29.73**
Policeman crabs (Goneplacidae)	*Neommatocarcinus huttoni*	4.55	2.00	8.33	0.98
Hairy red swimming crab (Ovalipidae)	*Nectocarcinus antarcticus*	3.03	1.70	8.33	0.71
Hermit crabs (Paguridae)	*Pagurus sp.*	6.06	0.51	16.67	1.96
Unidentified crabs		13.64	1.84	33.33	9.24
Banded mantis shrimp (Lysiosquillidae)		1.52	1.15	4.17	0.20
Unidentified shrimp		1.52	0.01	4.17	0.11
Other crustacea remains		3.03	0.03	8.33	0.46
Unidentified organic matter		**7.58**	**1.68**	**20.83**	**2.12***
Plant material		**1.52**	**0.00**	**4.17**	**0.07***

Total number of prey items	**66**
Total mass of prey	**494.76g**
Number of stomachs sampled (none empty)	**24**

*Calculated as overall group IRIs, not compared against individual components

Teleosts and crustaceans found in stomach contents included a range of different species, although most were unidentifiable parts, or their digestion was too far advanced for specific identification. In contrast, molluscs were represented almost entirely by a single bivalve species, Panopea zelandica (Table 4.1). Only siphons of this bivalve were found in stomachs, except for one individual which had its shell attached. Crabs were the most important group of crustaceans within the diet of C. isabellum, and of the identified crabs, hermit crabs (Pagurus sp.) made up the most %O (Table 4.1). The only teleost species identified from stomach contents was the seahorse Hippocampus abdominalis, but it was of low overall importance to the diet with only one specimen observed (Table 4.1).

4.3.3. Survey of prey items

Five different crab species and one ascidian species were trawled off the coast of Otago at depths where C. isabellum is common (50-100m) (Table 4.2). Most of the crabs sampled (n=128) were hermit crabs (Pagurus sp.), which as described above, make up a large component of the diet of C. isabellum. The other crab species and ascidian that were sampled in smaller numbers (n=5-25) have not been explicitly identified from the shark's stomach contents. However, given that other ascidians and a large amount of unidentified crabs are important parts of its diet, C. isabellum is very likely to encounter and eat these species too.

There was a general absence of parasites in these invertebrates, and overall, no tapeworm larval stages were recovered from any of the sampled crabs or ascidians. All species were found to have some individuals with very small nematodes (≤ 1mm in size), and some individual hermit crabs and ascidians also harboured small mites. The prevalence and abundance of these small nematodes was generally low for each crab species (Prevalence mostly below 20%), except for camouflage crabs (prevalence = 76.92%) (Table 4.2). A few mites were recovered from one orange ascidian and two individual hermit crabs, and their total prevalence and abundance also was very low (Table 4.2). Only one other symbiont was recovered from the invertebrates examined; a nematode found only in orange ascidians. This unidentified nematode species was much larger than the other nematode observed from crabs and ascidians (it reached a few millimetres in length). Four specimens of this nematode were recovered from each of four individual ascidians (26.67% prevalence).

Table 4.2. Prevalence and abundance of symbionts recovered from crabs and ascidians caught off the Coast of Otago, New Zealand. All prey species were caught via beam trawl from the Otago Shelf, ~NE off Taiaroa Head, at 92 m depth, except for long-legged masking crabs (*Leptomithrax longipes*), which were caught in a trawl with sharks offshore Otago peninsula, at 50 m depth. Mean host size given represents the carapace length of crabs, and the longest width dimension of ascidians. Numbers in parentheses indicate associated standard deviations.

Prey (Species)	**Sample size** (n)	**Mean host size** (mm)	**Nematodes (≤1mm)**		**Mite symbionts**	
			Prevalence	**Abundance**	**Prevalence**	**Abundance**
Orange Ascidian *(Cnemidocarpa bicornuta)*	15	51.67 (16.01)	40.00%	0.87 (1.30)	6.67%	0.07 (0.26)
Hermit crabs *(Pagurus spinulimanus)*	128	11.02 (3.58)	3.91%	0.06 (0.39)	1.56%	0.03 (0.28)
Triangle crabs **(***Eurynolambrus australis*)	25	27.40 (9.45)	8.00%	0.28 (1.21)	-	-
Long-legged masking crabs *(Leptomithrax longipes)*	11	67.45 (17.49)	9.09%	0.18 (0.60)	-	-
Long-handed masking crabs *(Leptomithrax longimanus)*	5	23.60 (7.64)	20.00%	0.60 (1.34)	-	-
Camouflage crabs *(Notomithrax sp.)*	13	36.38 (8.90)	76.92%	3.00 (3.36)	-	-

4.4. Discussion

The main objective of this study was to provide some insight into the importance of encounter and compatibility filters for tapeworm infection in the draughtsboard shark Cephaloscyllium isabellum. Regarding this aim, the findings of this study offer a small glimpse into the importance of diet as an encounter filter, showing that some of the most important prey items of draughtsboard sharks (hermit crabs Pagurus spinulimanus) are unlikely to be intermediate hosts to tapeworms. It is possible that despite their broad diet, draughtsboard sharks have a low diversity of tapeworms because the prey species they exploit generally don't harbour many tapeworm larvae. However, this can only be speculated based on the data analysed in this study. As demonstrated here and in other research, this shark consumes a variety of teleost, mollusc and other invertebrate prey (Horn 2016), none of which have been surveyed for parasites. Consequently, a far more comprehensive survey of prey would be needed to fully quantify the shark's tapeworm encounters, and accordingly, to determine the full importance of diet as an encounter filter for tapeworms. As highlighted by previous research on parasite encounter and compatibility filters, quantifying all parasite encounters is logistically challenging and requires a considerable amount of time and resources (Combes 2001; Kuris et al. 2007). Nevertheless, the data here provide a useful starting point for such research.

Prior to this research, C. isabellum was considered a cold spot in shark tapeworm diversity because it has fewer described tapeworm species than expected for a shark of its size (Randhawa 2014, unpublished data). The results here are consistent with this notion, given that all 24 of the sharks' spiral valves examined were exclusively infected by their only known tapeworm, Calyptrobothrium chalarosomum. It is still possible that the parasite assemblages of C. isabellum vary on a greater spatial scale. These sharks are known to occur across the entire coastal shelf around the North, South and Stewart Islands of New Zealand (Horn 2016), but so far records of their tapeworms have only been obtained from sharks examined off the east coast in Otago (present study), Oamaru, and from sharks inhabiting waters at the bottom of the North Island (Alexander 1963). To fully confirm the shark's low diversity of tapeworm species, individuals should be examined across their entire range, including waters off the West Coast, upper North Island, and offshore islands.

There was considerable variation in the intensity of Calyptrobothrium chalarosomum infection among the sharks inspected in this study, but the size, sex and sample of individuals did not significantly affect intensity. The intensity of tapeworm infections has been shown to generally increase with host size in fishes (see Poulin 2000). However, studies of elasmobranchs have shown mixed results (Cislo & Caira 1993; Sanmartin et al. 2000; Friggens & Brown 2005), and consistent with findings herein, some demonstrate that host length has little impact on tapeworm intensity of infection (Cislo & Caira 1993; Randhawa and Poulin 2009). It has been noted that the relationship may be in part masked by differences in tapeworm size, since there is a trade-off between the size and number of worms in the limited space and resources of a host, i.e. "crowding effect" (Read 1951; Roberts 2000; Randhawa and Poulin 2009). Regarding the effect of host sex on tapeworm intensity, it was interesting that although the difference was marginally non-significant here, male draughtsboard sharks generally harboured more tapeworms than females. Differences in infection intensity between sexes may reflect physiological or behavioural differences between male and female hosts (Zuk 1990; Poulin 1996). It is possible for example, that male and female draughtsboard sharks have different dietary preferences, whereby males are exposed to more tapeworm larva in their prey. Diet has not been analysed per sex in C. isabellum, however (Horn 2016), and in any case, high variation in tapeworm intensity was also observed within each sex and sample of sharks. Thus, larger sample sizes are needed to determine whether the differences seen here are consistent on a larger scale.

The overall diet composition of C. isabellum showed substantial overlap with previous records, reinforcing the notion that crustaceans, teleosts and molluscs are all important prey groups for this shark (Graham 1956; Horn 2016). Much like Horn's (2016) New Zealand-wide diet analysis, the present study showed that crabs dominate the diet in terms of occurrence (%O), and several crab species identified here had already been described as prey of C. isabellum. On the other hand, it was found here that the siphons of geoduck clams (Panopea zelandica) constituted a large part of the diet for individuals caught off Otago; this is the first record of these molluscs as prey. Given that there was a general absence of the clam's shells and other parts in stomach contents, it is suggested that the sharks likely bite off P. zelandica siphons while foraging, rather than eating them whole. Horn (2016) noted that similarly with findings here, despite feeding on many hermit crabs and gastropods, there are rarely shells or shell fragments in C. isabellum stomach contents. Like other scyliorhinid sharks, draughtsboard sharks may actively use their snouts to flip over protected invertebrates and eat only the exposed prey (Brightwell 1953; Horn 2016). This would allow them to avoid hard shells and materials that are difficult to digest and have little to no nutritional value. Remarkably, many other bivalve species have been recorded as intermediate hosts of elasmobranch tapeworms, but the infection of tapeworm larvae is generally restricted to the gonads, digestive gland and digestive tract of these bivalve hosts (Lauckner 1983; Hine & Thorne 2000; Vázquez & Cremonte 2017). This indicates that feeding on these siphons is unlikely to yield in tapeworm transmission for C. isabellum.

The complete absence of tapeworm larval stages that was observed in hermit crabs Pagurus spinulimanus suggests that they are unlikely intermediate hosts of elasmobranch tapeworms off the coast of Otago. By having a diet that consists largely of these hermit crabs, C. isabellum may inadvertently avoid potential encounters with tapeworms. However, this is not to say that all hermit crabs in their diet are scarce sources of tapeworms, since C. isabellum probably eat many other species which have not been surveyed for parasites. Overseas, several hermit crab species have been confirmed to host procercoid and plerocercoid larval stages of tapeworms that mature in sharks (Abbott 1987; Cherry et al. 1991; Smolowitz et al. 1993; McDermott et al. 2010). The most studied tapeworm species in hermit crabs, Calliobothrium verticillatum, has showed more than 95% prevalence in Pagurus sp. from the Woods Hole region, Massachusetts (Cherry et al. 1991). Interestingly, these tapeworms have also been described from brachyuran crabs (Fyler 2007), highlighting the potential for both hermit crabs and brachyuran crabs to be intermediate hosts of the same tapeworm larva. In the several small samples of brachyuran crabs examined for parasites in this study, no tapeworm larval stages were found. Nevertheless, the crab species examined from this area of Otago may harbour tapeworm larvae at a low prevalence, and hence, considerably larger sample sizes of these species must be surveyed to determine whether they host tapeworm larval stages. It is also worth noting that individuals in other areas may be more heavily infected, given that the prevalence and abundance of marine parasites often varies substantially across spatial and temporal scales (Smith 2001; Latham & Poulin 2003; Byers et al. 2008). Accordingly, further research on the parasites of these crabs would likely benefit from surveying individuals across other areas and during different seasons.

Despite not recovering any larval tapeworms from the ascidians, hermit crabs and brachyuran crabs examined, it is worth noting that all species were found to be infected with small nematodes, and some individual hermit crabs and ascidians also harboured small mites. Hermit crabs and other decapod crustaceans are known to sometimes harbour juvenile nematodes that mature in marine fishes (Poinar and Kuris 1975; Moravec et al. 2003; McDermott et al. 2010). Conversely, the nematodes of crabs examined herein were not encysted, and are probably not parasitic. However, further insights into the nature of this relationship will require additional studies and molecular tools for specific identification of the nematodes. Based on mites known from other hermit crabs, the mites found on P. spinulimanus in this study are also unlikely to be parasitic, and are probably symbiotic or commensal associates (McDermott 2010). Though it has been pointed out that the nature of relationships between mites and hermit crabs are generally unknown and warrant further investigation (O'Connor 1982, McDermott 2010).

4.4.1. Conclusions

The limited application of this study toward determining the importance of encounter filters for C. isabellum emphasises that analysing encounter filters for parasite infection is a very challenging task. Difficulties in estimating parasite encounters have been acknowledged by many researchers seeking to understand the importance of encounter and compatibility filters in hosts (Combes 2001; Kuris et al. 2007; Lagrue et al. 2011; Medeiros et al. 2013). In some host-parasite systems it may be possible to find good proxies for parasite encounter rates (Medeiros et al. 2013), but this is not possible for trophically transmitted parasites that use many different intermediate hosts to encounter their final host. In tapeworms of elasmobranchs, for example, the importance of encounter filters is hard to estimate without conducting comprehensive surveys of parasites from all of the elasmobranch's prey. Surveys can focus on prey species that are known to be most important in the final host's diet, since these species are the most encountered, and therefore, the quantity of larval parasites they harbour should be good indicators of how many parasites are typically encountered by the host. However, the most important prey in the diet are not necessarily the most important intermediate hosts (Cirtwill et al. 2017), and as illustrated by the findings here, there may be important prey which don't harbour larval stages of parasites known to infect the final host as adults. Ultimately, where there is sufficient time and resources available for researchers to quantify a host's encounters with trophically-transmitted parasites, it would be ideal to survey as many of their common prey for parasites as possible. However, it is suggested that smaller scale studies can still prove to be a valuable tool for analysing parasite encounters. Despite not finding any links in the life-cycles of local elasmobranch tapeworms, the results of this study provide new ecological and parasitological data for draughtsboard sharks and some of their selected prey, and offer some important insights that may aide further research towards better understanding the influence of encounter and compatibility filters of tapeworms in the model species C. isabellum and other South Island (NZ) elasmobranchs.

Chapter 5: Discussion

5.1. Summary of main findings and their implications

The overall objective of this thesis was to assess what factors influence the structure and diversity of tapeworm parasite assemblages in sharks, with focus on the importance of host diet. Previous research has already provided insights on several important determinants for tapeworm infections in elasmobranchs. These include a combination of phylogenetic factors, such as the interface between tapeworm attachment structures and host gut morphology (Williams 1960; 1966; 1968; Randhawa & Burt 2008), immune response (McVicar & Fletcher 1970; Randhawa & Burt 2008) and ecological factors, such as host size, depth, and latitude (Randhawa & Poulin 2010). However, a number of potentially key determinants, including the breadth and composition of host diet, have been recognised as crucial gaps in knowledge (Randhawa & Poulin 2010). The present research aimed to investigate these gaps on a broad scale by assessing the relative importance of host diet features (diet breadth, diet composition and trophic level) as predictors of tapeworm diversity across a large subset of different shark species (Chapter 2). Furthermore, literature records were used to conduct a comprehensive analysis of the relationship between shark's diet compositions and tapeworm compositions (Chapter 3). And finally, these gaps in knowledge were investigated on a smaller scale by analysing the importance of host diet for tapeworm encounter in a local shark species, Cephaloscyllium isabellum (Chapter 4). This shark species was selected as a model given its relatively broad diet and restricted tapeworm diversity.

The present research revealed that the diet breadth of a shark species, measured as its number of known prey families, is a better predictor of tapeworm species richness than other host features examined to date (Chapter 2). Thus, it is suggested that shark species with broad diets generally encounter and become infected with more tapeworm species than those with restricted or specialised diets. This has important implications not only for tapeworm diversity in sharks, but also for the diversity of trophically transmitted parasites in general. This study is the first to examine the influence of host diet on tapeworm diversity in elasmobranchs, and to my knowledge, is the most comprehensive analysis of tapeworm diversity in sharks to date. Few empirical studies in other host-parasite systems have demonstrated diet breadth to be important for the diversity of trophically-transmitted parasites (e.g. Chen et al. 2008;

Locke et al. 2014). Moreover, other comparative studies looking at determinants of parasite diversity have rarely analysed diet breadth as a factor (Kamiya et al. 2014). Thus, the findings herein emphasise that diet breadth could be very important for parasite diversity in other systems too, and should be more seriously considered in comparative studies among other commonly tested predictors of parasite diversity (e.g. host size, geographical range and population density).

Although the findings here establish diet breadth to be a key determinant of tapeworm richness in sharks, it was also made evident that it cannot be considered as a universal determinant of tapeworm diversity in all shark species. There are some shark species which are clear exceptions to the rule, demonstrated in chapter four with the model species C. isabellum. C. isabellum was found to have a relatively broad diet consisting of a variety of teleost fishes, crustaceans and other benthic invertebrates. Yet, it was shown to have a limited tapeworm fauna, with all individuals examined being exclusively infected by one species; Calyptrobothrium chalarosomum. This finding ultimately reinforces the notion that C. isabellum is a cold spot in shark tapeworm diversity, meaning that it harbours fewer tapeworm species than is expected for a shark of its size (Randhawa 2014, unpublished data; see Poulin et al. 2011b). Prior to this research, it was thought that C. isabellum could be a cold spot in tapeworm diversity due to compatibility filters (e.g. immune response) that eliminate potential tapeworm infections. However, given that no larval tapeworms were recovered in this research, testing for such compatibility filters was beyond the scope of the present study and still requires further investigation. The lack of tapeworm larval stages observed in some of the local prey species of this shark (hermit crabs and various brachyuran crabs) could also indicate that C. isabellum is a cold spot in tapeworm diversity because its preferred prey taxa are not intermediate hosts for tapeworms that mature in elasmobranchs. Without further surveys of its other prey, this can only be speculated. However, these results highlight the fact that certain prey taxa in a shark’s diet are likely to be considerably more relevant for tapeworm encounters than others.

The potential for highly variable distributions of tapeworm larval stages among prey is perhaps best illustrated in chapter three, where it was found that certain families of perciform fishes are substantially more important intermediate hosts of trypanorhynch-species larvae than other prey families. This notion has also been supported by extensive surveys of some fish families which have revealed particular taxa to be devoid of any tapeworm larvae (Beveridge et al. 2014). In chapter two, a negative relationship was found between the tapeworm richness of sharks and the average TD (taxonomic distinctness) of prey in their diets, which could also reflect this minor importance of certain prey as intermediate hosts. It was reasoned that sharks eating very unique and taxonomically different prey might often consume taxa that are irrelevant for tapeworm transmission, therefore resulting in species-poor tapeworm assemblages. Whether or not this is the case, this result clearly demonstrates that general diet breadth (richness of prey families) and diet TD, despite both being relative measures of diversity, reflect very different properties. Although the average TD of species assemblages is often included in ecological studies to complement simpler measures of diversity (i.e. species richness) (Von Euler & Svensson 2001; Heino et al. 2005; Winter et al. 2013), to my knowledge TD has only been implemented to measure aspects of diet in one other study comparing the diets of turtles (Stringell et al. 2016). The different patterns observed in this research for diet breadth and diet TD indicate that diet TD can provide unique and interesting insights into the feeding ecology of animals. As such, I encourage future comparative diet studies to consider investigating diet TD in conjunction with other aspects of diet.

It was clear from the present research that sharks' diets not only have important implications for the diversity of their tapeworms, but also have large consequences for the structure and composition of their tapeworm assemblages (Chapter 3). A significant positive correlation was found between similarities in shark diet composition and similarities in tapeworm composition, meaning that shark species are more likely to share tapeworm taxa if they have similar diets. This relationship supports the idea that tapeworms can be useful indicators of the feeding ecology of their definitive shark hosts (Palm et al. 2017). Further support for this point can be seen in comparisons of tapeworm families among sharks with different dominant prey groups. Tapeworm families were observed to be remarkably different between crustacean-feeding and teleost-feeding shark species, reflecting underlying differences in the life cycles of their tapeworms. Chapter three used several examples to illustrate that the tapeworm families common in crustacean-feeding sharks generally use crustaceans as second intermediate hosts, whereas tapeworm families of teleost-feeding sharks generally use teleosts or cephalopods as second intermediate hosts (see Palm 2004). These associations have important implications for ecologists who want to know the general feeding habits of poorly studied shark species. Notably, certain tapeworm taxa may be used to make predictions about a host's diet. For example, if a host is known to harbour echinobothriid tapeworm species, we could say that its diet is likely dominated by crustaceans and other benthic invertebrates, which are known to be their second intermediate hosts (Vivares 1971; Cake 1976; Tyler 2006) (see 3.4.1 for more examples). In some cases, these predictions may also work in reverse to inform ecologists of the likely intermediate hosts of certain tapeworms. Such information is extremely valuable given the current paucity of information on the life cycles of marine tapeworms (Caira & Reyda 2005; Jensen & Bullard 2010; Caira & Jensen 2017). Research on tapeworm life cycles has also been declining over the last few decades and is failing to keep up with new species descriptions, further highlighting the importance of this research (Blasco-Costa & Poulin 2017).

Tapeworm compositions were demonstrated to significantly differ between sharks of different taxonomic Orders, with carcharhiniform sharks being characterised by different tapeworm families than both lamniform and squaliform sharks (Chapter 3). This highlights the fact that tapeworm assemblages are fundamentally shaped both by host ecology and host evolutionary history, and emphasises the need to consider both aspects when looking at parasites as indicators of host ecology. Although previous research has given some insight into the importance of host diet and evolutionary history for the structure of trypanorhynch tapeworm assemblages in sharks (see Palm et al. 2017), the present research is the first to use ecological and phylogenetic data of host species to make inferences about the potential life cycles of tapeworms in other taxonomic Orders. Furthermore, this research offers new insights on more specific aspects of host diets, which have only been examined prior at a very basic level (between vertebrate and invertebrate feeding sharks) (Palm et al. 2017). For instance, the tapeworms of cephalopod-feeding sharks were found to overlap considerably with both teleost-feeding and crustacean-feeding sharks, indicating that perhaps cephalopods are commonly intermediate or paratenic hosts in the same life cycles of teleosts and crustaceans. Overall, these insights provide a considerable step towards a better understanding of the complex relationships that exist between the ecology and evolution of elasmobranch hosts and the composition of their tapeworm parasite assemblages.

A more comprehensive survey of the prey of C. isabellum is still clearly needed to determine the relative importance of its diet as an encounter filter for tapeworm parasite diversity. However, although the prey survey conducted here offered very limited insights regarding the life-cycles of local elasmobranch tapeworms, important new data were provided on both the parasites and diet of C. isabellum. The geoduck clam Panopea zelandica was revealed as a new diet record for C. isabellum, and was found to be very important prey for individuals inhabiting the Otago region. Panopea zelandica has been recognised to lack current ecological information, but has recently gained increased attention as a potential target for commercial exploitation (Gribben & Heasman 2015). Likewise, C. isabellum has been identified as one of the shark species most at risk to commercial fishery impacts in New Zealand, but is also acknowledged that significant knowledge gaps remain regarding its basic biology and ecology (Ford et al. 2015; Horn 2016). Thus, the diet information herein may be of use for fisheries involving these species. On top of this, these findings provide a small step for further research towards better understanding the influence of encounter and compatibility filters of tapeworms in C. isabellum and other South Island (NZ) elasmobranchs.

5.2. Further research

The current research established some strong ecological patterns that demonstrate host diet as an important factor governing tapeworm assemblages in sharks. These patterns were established by large-scale comparative analyses across different species and by some limited observations in the model shark species C. isabellum. Nevertheless, there is still great room for future comparative studies to extend on this work and provide a better understanding of the intricacies within these patterns. Furthermore, there is a need for the patterns established here to be backed up with experimental evidence. Notably, questions remain as to whether encounter filters or compatibility filters are more important barriers to tapeworm infections in C. isabellum and other elasmobranchs. These questions cannot be fully answered by observational studies alone, and will require explorations of host-parasite compatibility in future experimental studies.

This research identified several key knowledge gaps in the literature that, if addressed, may considerably improve our understanding of the relationship between the diet of shark hosts and the diversity and structure of their parasite assemblages. First and foremost, available data on hosts' diets and parasites are conspicuously biased towards shark species of the Order Carcharhiniformes. It could be argued that this is the result of carcharhiniform species being some of the most important, abundant and widespread sharks inhabiting our oceans (Compagno 2001). However, to further our knowledge of the diversity and evolution of parasite lineages in sharks, there is a need to prioritise the sampling of diets and parasites across shark species of other taxonomic Orders. The diets of some lesser-studied shark species could be poorly known for conservational or ethical reasons (since most diet sampling is lethal). However, several studies have demonstrated the efficacy of non-lethal methods of diet sampling e.g. gastric lavage (Foster 1977; Barnett et al. 2010; Hammerschlag & Sulikowski 2011). These methods warrant consideration in further research aiming to elucidate the diets of important shark species, especially those that are endangered or identified as conservation risks.

Another key knowledge gap that may be of interest to future research is how intraspecific variation in the diets of elasmobranchs affects their tapeworm assemblages. In the present study, tapeworms and prey were viewed for each shark species as the sum of all records known for the species. However, diets may vary substantially with the size or age of individuals (e.g. Lowe et al. 1996; Ebert 2002), between different sexes (e.g. Klimley 1987; Borrell 2011), between seasons (e.g. Cortes et al. 1996), and between different geographical areas (e.g. Simpfendorfer et al. 2001; Bethea et al. 2007). The present research indicates that disparities in diet among these different sub-populations could have large consequences for their parasite assemblages. Though diet studies often look at differences in sharks' stomach contents among these different sub-populations, few diet studies have looked at the intestinal parasites of individuals in conjunction with their diets (for notable exceptions see Klimpel et al. 2003; Gracan et al. 2014; Isbert et al. 2015). I recommend that future studies examining elasmobranch stomach contents should also examine the spiral valves of individuals for any potential parasites. Such studies will provide valuable insights into the effects of intra-specific diet variation on the diversity and composition of elasmobranch tapeworm assemblages. Furthermore, even where researchers are specifically interested in revealing species diets (i.e. have no interest in parasitological data), as demonstrated within this research, tapeworms can provide additional information on species' feeding habits. In some cases, tapeworm assemblages could be even more informative given that stomach contents provide only a limited snapshot of a host's last meal, whereas tapeworms have the potential to reflect their longer-term feeding habits. The inclusion of parasite data in future diet studies will require raising the awareness of marine ecologists to these potential insights, and ultimately, better collaborations between marine ecologists and marine parasitologists (Poulin et al. 2016).

Future studies integrating diet and parasite data could perhaps also benefit from looking at how diet variation among closely related elasmobranchs affects their respective tapeworm assemblages. In chapter three of this thesis it was shown that deciphering the relative influence of feeding ecology and host evolutionary history on shark tapeworm assemblages can be very challenging. However, it would be possible to explore the relative influence of host ecology and evolutionary history in some species by looking at how tapeworm compositions vary among closely related sharks with different feeding habits. For example, in hammerhead sharks (Sphryna sp.), there are some clear differences in feeding habits between species. Sphyrna lewini, S. mokkaran and S. zygaena all feed predominantly on fishes and cephalopods (Stevens & Lyle 1989; Smale 1991), whereas S. tiburo is a durophagous predator that feeds mainly on crustaceans (Cortés et al. 1996; Bethea et al. 2007). The dataset herein indicates that the three teleost-feeding hammerhead species each have relatively high tapeworm species richness, but S. tiburo appears to have low tapeworm richness (although it requires further study for parasites). A possible reason for this discrepancy could be that the ancestors of S. tiburo were mainly teleost-feeders, but S. tiburo has gone down an evolutionary path where it switched to feeding on crustaceans (see Lim et al. 2010). Considering this, the evolution of S. tiburo may have broken life cycles of ancestral tapeworms involving teleost fishes, resulting in a lower diversity of tapeworms for the species compared with other Sphyrna species.

These points can only be speculated without further research looking more specifically at the variations in diet and parasites among these species.

The present research showed a novel approach towards understanding encounter and compatibility filters for parasites in C. isabellum, by analysing host diet to determine parasite encounters. However, there is still a long way to go before the relative importance of these filters can be established. More comprehensive parasite surveys of the shark's prey will be needed to determine the relative influence of encounter filters. This will demand a considerable amount of time and resources (Combes 2001; Kuris et al. 2007). Nevertheless, no previous studies have quantified parasite encounters for elasmobranchs in this way, and the findings of such research could significantly advance our understanding of parasite filters in elasmobranchs. In addition, where larval parasites are recovered from prey in these surveys, this would open opportunities for experimental studies to test the strength of compatibility filters in elasmobranch hosts. For instance, larval tapeworms from known prey could be exposed to the host's blood sera and observed for mortality to determine whether immune response restricts potential tapeworm establishment i.e. whether immune response is a strong compatibility filter for the species (McVicar & Fletcher 1970; Randhawa & Burt 2008). Other compatibility filters could also be tested by setting up experiments where sharks are held in vivo and given anthelmintic drugs to remove their tapeworms. Subsequently, prey infected by larval tapeworms (that are not naturally found in the host) could be fed to individual hosts and assessed for whether they can successfully establish in the shark's digestive system as mature adult worms. These tests could potentially be done in species other than C. isabellum. However, the findings of this thesis reinforce that C. isabellum would be an ideal model for these studies given its relatively broad diet and low tapeworm diversity.

5.3. Conclusions

The observational studies carried out within this project emphasise that various aspects of sharks' diets can have important implications for their tapeworm parasite assemblages. It was demonstrated that the diversity of tapeworms infecting a shark species largely depends on the breadth of its diet. Furthermore, it was shown that certain prey taxa are considerably more important as intermediate hosts for tapeworms than others, and ultimately, even sharks with broad diets may have a limited diversity of tapeworm species depending on their dietary preferences. Finally, it was found that certain tapeworm taxa in sharks are useful indicators of their ecology and evolutionary history, and may be used to make predictions about the host’s feeding habits. Together these findings demonstrate some strong ecological patterns that establish host diet as an important determinant of tapeworm composition and diversity in sharks. However, there is a need for further exploration of these patterns with experimental research to validate the influence of these patterns in nature.

References

Abbott, D. P., & Hilgard, G. H. (1987). Observing marine invertebrates: drawings from the laboratory. Stanford University Press.

Ackerly, D. D. (2000). Taxon sampling, correlated evolution, and independent contrasts. Evolution, 54, 1480-1492.

Ackerly, D. D. & Reich, P. B. (1999). Convergence and correlations among leaf size and function in seed plants: a comparative test using independent contrasts. American Journal of Botany, 86, 1272-1281.

Aeby, G. S. (1991). Behavioral and ecological relationships of a parasite and its hosts within a coral reef system. Pacific Science, 45(3), 263-269.

Alexander, C. G. (1963). Tetraphyllidean and diphyllidean cestodes' of New Zealand selachians. In Transactions of the Royal Society of New Zealand, 3 (12), 117-142.

Alves, P. V., de Chambrier, A., Scholz, T., & Luque, J. L. (2017). Annotated checklist of fish cestodes from South America. ZooKeys, (650), 1.

Ahyong, S. T. (2012). The marine fauna of New Zealand: mantis shrimps (Crustacea: Stomatopoda). National Institute of Water and Atmospheric Research Biodiversity Memoir, 125.

Amundsen, P. A., Lafferty, K. D., Knudsen, R., Primicerio, R., Klemetsen, A., & Kuris, A. M. (2009). Food web topology and parasites in the pelagic zone of a subarctic lake. Journal of Animal Ecology, 78(3), 563-572.

Anderson, D. R. (2008). Model based inference in the life sciences: a primer on evidence. Springer Science & Business Media, Berlin.

Awruch, C. A., Frusher, S. D., Stevens, J. D., & Barnett, A. (2012). Movement patterns of the draughtboard shark Cephaloscyllium laticeps (Scyliorhinidae) determined by passive tracking and conventional tagging. Journal of fish biology, 80(5), 1417-1435.

Aznar, F. J., Agustí, C., Littlewood, D. T. J., Raga, J. A., & Olson, P. D. (2007). Insight into the role of cetaceans in the life cycle of the tetraphyllideans (Platyhelminthes: Cestoda). International Journal for Parasitology, 37(2), 243-255.

Baeten, L., Vangansbeke, P., Hermy, M., Peterken, G., Vanhuyse, K., & Verheyen, K. (2012). Distinguishing between turnover and nestedness in the quantification of biotic homogenization. Biodiversity and Conservation, 21(6), 1399-1409.

Bakker, T., Frommen, J. G., & Thünken, T. (2017). Adaptive parasitic manipulation as exemplified by acanthocephalans. Ethology, 123(11), 779-784.

Barnett, A., Redd, K. S., Frusher, S. D., Stevens, J. D., & Semmens, J. M. (2010). Non-lethal method to obtain stomach samples from a large marine predator and the use of DNA analysis to improve dietary information. Journal of experimental marine biology and ecology, 393(1), 188-192.

Barton, K. (2013). MuMIn: Multi-model inference. R package version 1.9.5. – < cran.rproject.org/web/packages/MuMIn >.

Baselga, A. (2010). Partitioning the turnover and nestedness components of beta diversity. Global Ecology and Biogeography, 19(1), 134-143.

Baselga, A., Jiménez-Valverde, A., & Niccolini, G. (2007). A multiple-site similarity measure independent of richness. Biology Letters, 3(6), 642-645.

Baselga, A., & Orme, C. D. L. (2012). betapart: an R package for the study of beta diversity. Methods in Ecology and Evolution, 3(5), 808-812.

Benesh, D. P., Chubb, J. C., & Parker, G. A. (2014). The trophic vacuum and the evolution of complex life cycles in trophically transmitted helminths. Proceedings of the Royal Society of London B: Biological Sciences, 281(1793), 20141462.

Bethea, D. M., Hale, L., Carlson, J. K., Cortés, E., Manire, C. A., & Gelsleichter, J. (2007). Geographic and ontogenetic variation in the diet and daily ration of the bonnethead shark, Sphyrna tiburo, from the eastern Gulf of Mexico. Marine Biology, 152(5), 1009-1020.

Beveridge, I., Bray, R. A., Cribb, T. H., & Justine, J. L. (2014). Diversity of trypanorhynch metacestodes in teleost fishes from coral reefs off eastern Australia and New Caledonia. Parasite, 21, 60.

Blasco-Costa, I., & Poulin, R. (2017). Parasite life-cycle studies: a plea to resurrect an old parasitological tradition. Journal of Helminthology, 91, 647–656.

Borrell, A., Aguilar, A., Gazo, M., Kumarran, R. P., & Cardona, L. (2011). Stable isotope profiles in whale shark (Rhincodon typus) suggest segregation and dissimilarities in the diet depending on sex and size. Environmental Biology of Fishes, 92(4), 559-567.

Bowman, D. D. (2006). Successful and currently ongoing parasite eradication programs. Veterinary parasitology, 139(4), 293-307.

Brickle, P., Olson, P. D., Littlewood, D. T. J., Bishop, A., & Arkhipkin, A. I. (2001). Parasites of Loligo gahi from waters off the Falkland Islands, with a phylogenetically based identification of their cestode larvae. Canadian Journal of Zoology, 79(12), 2289-2296.

Brightwell, L. E. (1953). Further notes on the hermit crab Eupagurus bernhardus and associated animals. Journal of Zoology, 123(1), 61-64.

Brown, J. H., Gillooly, J. F., Allen, A. P., Savage, V. M., & West, G. B. (2004). Toward a metabolic theory of ecology. Ecology, 85(7), 1771-1789.

Byers, J. E., Blakeslee, A. M., Linder, E., Cooper, A. B., & Maguire, T. J. (2008). Controls of spatial variation in the prevalence of trematode parasites infecting a marine snail. Ecology, 89(2), 439-451.

Caira, J. N. & Healy, C. J. (2004). Elasmobranchs as hosts of metazoan parasites. Biology of sharks and their relatives, (eds Carrier, J. C., Musick, J. A., & Heithaus, M. R.), pp. 523-551. CRC Press, Florida.

Caira, J. N., & Jensen, K. (2001). An investigation of the co-evolutionary relationships between onchobothriid tapeworms and their elasmobranch hosts. International Journal for Parasitology, 31(9), 960-975.

Caira, J. N., & Jensen, K. (2014). A digest of elasmobranch tapeworms. The Journal of parasitology, 100(4), 373-391.

Caira, J. N., & Jensen, K. (2017). Planetary Biodiversity Inventory (2008–2017): Tapeworms from Vertebrate Bowels of the Earth. University of Kansas, Natural History Museum Special Publication, 25, KS, USA, pp. 279-304.

Caira, J. N., Jensen, K., & Healy, C. J. (2001). Interrelationships among tetraphyllidean and lecanicephalidean cestodes. Interrelationships of the Platyhelminthes, 1, 135-158.

Caira, J. N., Jensen, K., & Ivanov, V. A. (2017). Onchoproteocephalidea II Caira, Jensen, Waeschenbach, Olson & Littlewood, 2014. In: Planetary Biodiversity Inventory (2008-2017): Tapeworms from Vertebrate Bowels of the Earth. J. N. Caira & K. Jensen (eds). University of Kansas, Natural History Museum Special Publication, 25, KS, USA, pp. 279-304.

Caira, J. N., & Reyda, F. B. (2005). Eucestoda (true tapeworms). In: Marine parasitology, K. Rohde (ed.). CSIRO Publishing, Collingwood, Australia, 92-

104.

Cake, E. W. (1976). A key to larval cestodes of shallow-water, benthic mollusks of the northern Gulf of Mexico. Proceedings of the Helminthological Society of Washington, 43, 160-171.

Carvalho, J. C., Cardoso, P., & Gomes, P. (2012). Determining the relative roles of species replacement and species richness differences in generating beta-diversity patterns. Global Ecology and Biogeography, 21(7), 760-771.

Chen, H. W., Liu, W. C., Davis, A. J., Jordán, F., Hwang, M. J., & Shao, K. T. (2008). Network position of hosts in food webs and their parasite diversity. Oikos, 117(12), 1847-1855.

Cherry, B., Neese, A. S., Bullis, R. A., & Schad, G. A. (1991). Investigations into the life cycle of Calliobothrium verticillatum, a tapeworm of Mustelus canis. The Biological Bulletin, 181(2), 358-358.

Cirtwill, A. R., Stouffer, D. B., Poulin, R., & Lagrue, C. (2016). Are parasite richness and abundance linked to prey species richness and individual feeding preferences in fish hosts?. Parasitology, 143(1), 75-86.

Cirtwill, A. R., Lagrue, C., Poulin, R., & Stouffer, D. B. (2017). Host taxonomy constrains the properties of trophic transmission routes for parasites in lake food webs. Ecology, 98(9), 2401–2412.

Cislo, P. R., & Caira, J. N. (1993). The parasite assemblage in the spiral intestine of the shark Mustelus canis. The Journal of parasitology, 886-899.

Clarke, K. R. (1993). Non‐parametric multivariate analyses of changes in community structure. Austral Ecology, 18(1), 117-143.

Combes, C. (2001). The ecology and evolution of intimate interactions. University of Chicago Press.

Compagno, L. J. (2001). Sharks of the world: an annotated and illustrated catalogue of shark species known to date (Vol. 1). Food & Agriculture Org.

Cortés, E. (1997). A critical review of methods of studying fish feeding based on analysis of stomach contents: application to elasmobranch fishes. Canadian Journal of Fisheries and Aquatic Sciences, 54(3), 726-738.

Cortés, E. (1999). Standardized diet compositions and trophic levels of sharks. ICES Journal of marine science, 56(5), 707-717.

Cortés, E., Manire, C. A., & Hueter, R. E. (1996). Diet, feeding habits, and diel feeding chronology of the bonnethead shark, Sphyrna tiburo, in southwest Florida. Bulletin of Marine Science, 58(2), 353-367.

Cox, G. J., & Francis, M. (1997). Sharks and rays of New Zealand. Canterbury University Press.

Dallarés, S., Carrassón, M., & Schaeffner, B. C. (2017). Revision of the family Sphyriocephalidae Pintner, 1913 (Cestoda: Trypanorhyncha), with the description of Heterosphyriocephalus encarnae n. sp. and redescriptions of two species of Sphyriocephalus. Parasitology international, 66(1), 843-862.

Dentzien-Dias, P. C., Poinar Jr, G., de Figueiredo, A. E. Q., Pacheco, A. C. L., Horn, B. L., & Schultz, C. L. (2013). Tapeworm eggs in a 270 million-year-old shark coprolite. PLoS One, 8(1), e55007.

Dick, C. W., Esbérard, C. E. L., Graciolli, G., Bergallo, H. G., & Gettinger, D. (2009). Assessing host specificity of obligate ectoparasites in the absence of dispersal barriers. Parasitology research, 105(5), 1345.

Dobson, A., Lafferty, K. D., Kuris, A. M., Hechinger, R. F., & Jetz, W. (2008). Homage to Linnaeus: How many parasites? How many hosts? Proceedings of the National Academy of Sciences, 105 (1), 11482-11489.

Dulvy, N. K., & Reynolds, J. D. (1997). Evolutionary transitions among egg-laying, live–bearing and maternal inputs in sharks and rays. Proceedings of the Royal Society of London B: Biological Sciences, 264(1386), 1309-1315.

Dunn, A. M. (2009). Parasites and biological invasions. Advances in parasitology, 68, 161-184.

Dunne, J. A., Lafferty, K. D., Dobson, A. P., Hechinger, R. F., Kuris, A. M., Martinez, N. D., ... & Stouffer, D. B. (2013). Parasites affect food web structure primarily through increased diversity and complexity. PLoS biology, 11(6), e1001579.

Dyer, L. A., Singer, M. S., Lill, J. T., Stireman, J. O., Gentry, G. L., Marquis, R. J., and Coley, P. D. (2007). Host specificity of Lepidoptera in tropical and temperate forests. Nature, 448(7154), 696-699.

Ebert, D. A. (2002). Ontogenetic changes in the diet of the sevengill shark (Notorynchus cepedianus). Marine and Freshwater Research, 53(2), 517-523.

Euzet, L., and Combes, C. (1980). Les proble`mes de l'espe`ce chez les animaux parasites. Bulletin de la Société zoologique de France, 40, 239–285.

Felsenstein, J. (1985). Phylogenies and the comparative method. The American Naturalist, 125, 1-15.

Ferretti, F., Worm, B., Britten, G. L., Heithaus, M. R., & Lotze, H. K. (2010). Patterns and ecosystem consequences of shark declines in the ocean. Ecology letters, 13(8), 1055-1071.

Foster, J. R. (1977). Pulsed gastric lavage: an efficient method of removing the stomach contents of live fish. The Progressive Fish-Culturist, 39(4), 166-169.

Francis, M. P., Hurst, R. J., McArdle, B. H., Bagley, N. W., & Anderson, O. F. (2002). New Zealand demersal fish assemblages. Environmental Biology of Fishes, 65(2), 215-234.

Friggens, M. M., & Brown, J. H. (2005). Niche partitioning in the cestode communities of two elasmobranchs. Oikos, 108(1), 76-84.

Froese, R. and D. Pauly. Editors. (2017). FishBase. World Wide Web electronic publication. www.fishbase.org, version (06/2017).

Fyler, C. A. (2007). Comparison of microthrix ultrastructure and morphology on the plerocercoid and adult scolex of Calliobothrium cf. verticillatum (Tetraphyllidea: Onchobothriidae). Journal of Parasitology, 93(1), 4-11.

Gibson, D. I., Bray, R. A., & Harris, E. A. (Compilers) (2005). Host-Parasite Database of the Natural History Museum, London. http://www.nhm.ac.uk. [Accessed December 2017].

Goedknegt, A., Welsh, J., & Thieltges, D.W. (2012). Parasites as prey. In: eLS. John Wiley & Sons Ltd., Chichester.

Gracan, R., Mladineo, I., & Lazar, B. (2014). Insight into the diet composition and gastrointestinal parasite community of the common smooth-hound, Mustelus mustelus (Carcharhiniformes: Triakidae), in the northern Adriatic Sea. Natura Croatica, 23(1), 35.

Graham, D. H. (1956). A treasury of New Zealand fishes. AH & AW Reed.

Gribben, P. E., & Heasman, K. G. (2015). Developing fisheries and aquaculture industries for Panopea zelandica in New Zealand. Journal of Shellfish Research, 34(1), 5-10.

Hammerschlag, N., & Sulikowski, J. (2011). Killing for conservation: the need for alternatives to lethal sampling of apex predatory sharks. Endangered Species Research, 14(2), 135-140.

Healy, C. J. (2003). A revision of Platybothrium Linton, 1890 (Tetraphyllidea:

Onchobothriidae), with a phylogenetic analysis and comments on host-parasite associations. Systematic Parasitology, 56(2), 85-139.

Heino, J., Soininen, J., Lappalainen, J., & Virtanen, R. (2005). The relationship between species richness and taxonomic distinctness in freshwater organisms. Limnology and Oceanography, 50(3), 978-986.

Hewitt, G. C., & Hine, P. M. (1972). Checklist of parasites of New Zealand fishes and of their hosts. New Zealand journal of marine and freshwater research, 6(1-2), 69-114.

Hine, P. M., Jones, J. B., & Diggles, B. K. (2000). A checklist of the parasites of New Zealand fishes, including previously unpublished records. National Institute of Water and Atmospheric Research. 95pp.

Hine, P. M., & Thorne, T. (2000). A survey of some parasites and diseases of several species of bivalve mollusc in northern Western Australia. Diseases of aquatic organisms, 40(1), 67-78.

Hoberg, E. P., & Klassen, G. J. (2002). Revealing the faunal tapestry: co-evolution and historical biogeography of hosts and parasites in marine systems. Parasitology, 124(7), 3-22.

Holland, N. D., & Wilson, N. G. (2009). Molecular identification of larvae of a tetraphyllidean tapeworm (Platyhelminthes: Eucestoda) in a razor clam as an alternative intermediate host in the life cycle of Acanthobothrium brevissime. Journal of Parasitology, 95(5), 1215-1217.

Holt, R. D. (2010). A World free of parasites and vectors: Would it be heaven, or would it be hell? Israel Journal of Ecology & Evolution, 56, 239-250.

Horn, P. L. (2016). Biology of the New Zealand carpet shark Cephaloscyllium isabellum. Journal of Ichthyology, 56(3), 336-347.

Isbert, W., Rodríguez-Cabello, C., Frutos, I., Preciado, I., Montero, F. E., & Pérez, A. (2015). Metazoan parasite communities and diet of the velvet belly lantern shark Etmopterus spinax (Squaliformes: Etmopteridae): a comparison of two deep-sea ecosystems. Journal of fish biology, 86(2), 687-706.

Jakob, E., & Palm, H. W. (2006). Parasites of commercially important fish species from the southern Java coast, Indonesia, including the distribution pattern of trypanorhynch cestodes. Verhandlungen der Gesellschaft fr Ichthyologie, 5, 165-191.

Jensen, K. (2009). Cestoda (Platyhelminthes) of the Gulf of Mexico. Gulf of Mexico. Origin, Waters, and Biota. (eds Felder, D. L., & Camp, D. K). Pp. 487-522. Texas A & M University Press: Texas.

Jensen, K., & Bullard, S. A. (2010). Characterization of a diversity of tetraphyllidean and rhinebothriidean cestode larval types, with comments on host associations and life-cycles. International Journal for Parasitology, 40(8), 889-910.

Johnson, P. T., Dobson, A., Lafferty, K. D., Marcogliese, D. J., Memmott, J., Orlofske, S. A., & Thieltges, D. W. (2010). When parasites become prey: ecological and epidemiological significance of eating parasites. Trends in ecology & evolution, 25(6), 362-371.

Kamiya, T., O'dwyer, K., Nakagawa, S., & Poulin, R. (2014). What determines species richness of parasitic organisms? A meta-analysis across animal, plant and fungal hosts. Biological Reviews, 89(1), 123-134.

Kennedy, C. R., Bush, A. O., & Aho, J. M. (1986). Patterns in helminth communities: why are birds and fish different?. Parasitology, 93(1), 205-215.

Klimley, A. P. (1987). The determinants of sexual segregation in the scalloped hammerhead shark, Sphyrna lewini. Environmental Biology of Fishes, 18(1), 27-40.

Klimpel, S., Palm, H. W., & Seehagen, A. (2003). Metazoan parasites and food composition of juvenile Etmopterus spinax (L., 1758)(Dalatiidae, Squaliformes) from the Norwegian Deep. Parasitology Research, 89(4), 245-251.

Klimpel, S., Rückert, S., Piatkowski, U., Palm, H. W., & Hanel, R. (2006). Diet and metazoan parasites of silver scabbard fish Lepidopus caudatus from the Great Meteor Seamount (North Atlantic). Marine Ecology Progress Series, 315, 249-257.

Koleff, P., Gaston, K. J., & Lennon, J. J. (2003). Measuring beta diversity for presence–absence data. Journal of Animal Ecology, 72(3), 367-382.

Komar, N., Langevin, S., Hinten, S., Nemeth, N., Edwards, E., Hettler, D., ... & Bunning, M. (2003). Experimental infection of North American birds with the New York 1999 strain of West Nile virus. Emerging infectious diseases, 9(3), 311.

Kosoy, M. Y., Regnery, R. L., Tzianabos, T., Marston, E. L., Jones, D. C., Green, D. and Childs, J. E. (1997). Distribution, diversity, and host specificity of Bartonella in rodents from the southeastern United States. The American journal of tropical medicine and hygiene, 57(5), 578-588.

Kuris, A. M., Blaustein, A. R., & Alio, J. J. (1980). Hosts as islands. The American Naturalist, 116(4), 570-586.

Kuris, A. M., Goddard, J. H., Torchin, M. E., Murphy, N., Gurney, R., and Lafferty, K. D. (2007). An experimental evaluation of host specificity: the role of encounter and compatibility filters for a rhizocephalan parasite of crabs. International journal for parasitology, 37(5), 539-545.

Kuris, A. M., Hechinger, R. F., Shaw, J. C., Whitney, K. L., Aguirre-Macedo, L., Boch, C. A., ... & Lorda, J. (2008). Ecosystem energetic implications of parasite and free-living biomass in three estuaries. Nature, 454(7203), 515-518.

Lafferty, K. D. (1999). The evolution of trophic transmission. Parasitology Today, 15(3), 111-115.

Lafferty, K. D., & Morris, A. K. (1996). Altered behavior of parasitized killifish increases susceptibility to predation by bird final hosts. Ecology, 77(5), 1390-1397.

Lafferty, K. D., Dobson, A. P., & Kuris, A. M. (2006). Parasites dominate food web links. Proceedings of the National Academy of Sciences, 103(30), 11211-11216.

Lafferty, K.D., Allesina, S., Arim, M., Briggs, C.J., De Leo, G., Dobson, A.P., Dunne, J.A., Johnson, P.T., Kuris, A.M., Marcogliese, D.J. and Martinez, N.D. (2008). Parasites in food webs: the ultimate missing links. Ecology letters, 11(6), 533-546.

Lagrue, C., Kelly, D. W., Hicks, A., & Poulin, R. (2011). Factors influencing infection patterns of trophically transmitted parasites among a fish community: host diet, host parasite compatibility or both? Journal of fish biology, 79(2), 466-485.

Latham, A. D. M., & Poulin, R. (2003). Spatiotemporal heterogeneity in recruitment of larval parasites to shore crab intermediate hosts: the influence of shorebird definitive hosts. Canadian Journal of Zoology, 81(7), 1282-1291.

Lauckner, G. (1983). Chapter 13. Diseases of Mollusca: Bivalvia. In: O. Kinne (Ed.), Vol. II. Diseases of Marine Animals. pp. 477-879, Biologische Anstalt Helgoland, Hamburg.

Lefèvre, T., Lebarbenchon, C., Gauthier-Clerc, M., Missé, D., Poulin, R., & Thomas, F. (2009). The ecological significance of manipulative parasites. Trends in Ecology & Evolution, 24(1), 41-48.

Lim, D. D., Motta, P., Mara, K., & Martin, A. P. (2010). Phylogeny of hammerhead sharks (Family Sphyrnidae) inferred from mitochondrial and nuclear genes. Molecular Phylogenetics and Evolution, 55(2), 572-579.

Lindenfors, P., Nunn, C. L., Jones, K. E., Cunningham, A. A., Sechrest, W., & Gittleman, J. L. (2007). Parasite species richness in carnivores: effects of host body mass, latitude, geographical range and population density. Global Ecology and Biogeography, 16(4), 496-509.

Lo, C. M., Morand, S., & Galzin, R. (1998). Parasite diversity host age and size relationship in three coral-reef fishes from French Polynesia. International Journal for Parasitology, 28(11), 1695-1708.

Locke, S. A., Marcogliese, D. J., & Valtonen, E. T. (2014). Vulnerability and diet breadth predict larval and adult parasite diversity in fish of the Bothnian Bay. Oecologia, 174(1), 253-262.

Lootvoet, A., Blanchet, S., Gevrey, M., Buisson, L., Tudesque, L., and Loot, G. (2013). Patterns and processes of alternative host use in a generalist parasite: insights from a natural host–parasite interaction. Functional Ecology, 27(6), 1403-1414.

Lourie, S. A., Foster, S. J., Cooper, E. W., & Vincent, A. C. (2004). A guide to the identification of seahorses. Project Seahorse and TRAFFIC North America, 114.

Lowe, C. G., Wetherbee, B. M., Crow, G. L., & Tester, A. L. (1996). Ontogenetic dietary shifts and feeding behavior of the tiger shark, Galeocerdo cuvier, in Hawaiian waters. Environmental Biology of Fishes, 47(2), 203-211.

Luque, J. L., Mouillot, D., & Poulin, R. (2004). Parasite biodiversity and its determinants in coastal marine teleost fishes of Brazil. Parasitology, 128(6), 671-682.

Luque, J. L., & Poulin, R. (2007). Metazoan parasite species richness in Neotropical fishes: hotspots and the geography of biodiversity. Parasitology, 134(6), 865-878.

Luque, J. L., & Poulin, R. (2008). Linking ecology with parasite diversity in Neotropical fishes. Journal of Fish Biology, 72(1), 189-204.

Marcogliese, D. J. (2003). Food webs and biodiversity: are parasites the missing link. Journal of Parasitology, 89(6), 106-113.

Marcogliese, D. J., & Cone, D. K. (1997). Food webs: a plea for parasites. Trends in Ecology & Evolution, 12(8), 320-325.

Mariaux, J., & Olson, P. D. (2001). Cestode systematics in the molecular era. Interrelationships of the Platyhelminthes, 127-134.

Marques, J. F., Santos, M. J., Teixeira, C. M., Batista, M. I., & Cabral, H. N. (2011). Host-parasite relationships in flatfish (Pleuronectiformes) - the relative importance of host biology, ecology and phylogeny. Parasitology, 138(1), 107-121.

Márquez-Farías, J. F., Corro-Espinosa, D., & Castillo-Géniz, J. L. (2005). Observations on the biology of the Pacific sharpnose shark (Rhizoprionodon longurio, Jordan and Gilbert 1882), captured in southern Sinaloa, Mexico.

Journal of Northwest Atlantic Fishery Science, 55(1), 107-114.

McDermott, J. J., Williams, J. D., & Boyko, C. B. (2010). The unwanted guests of hermits: a global review of the diversity and natural history of hermit crab parasites. Journal of Experimental Marine Biology and Ecology, 394(1), 2-44.

McVicar, A. H., & Fletcher, T. C. (1970). Serum factors in Raja radiata toxic to Acanthobothrium quadripartitum (Cesoda: Tetraphyllidea), a parasite specific to R. naevus. Parasitology, 61(1), 55-63.

Medeiros, M. C., Hamer, G. L., & Ricklefs, R. E. (2013). Host compatibility rather than vector–host-encounter rate determines the host range of avian Plasmodium parasites. Proceedings of the Royal Society B: Biological Sciences, 280(1760), 1-8.

Mitchell, C. E., Blumenthal, D., Jarošík, V., Puckett, E. E., & Pyšek, P. (2010). Controls on pathogen species richness in plants' introduced and native ranges: roles of residence time, range size and host traits. Ecology letters, 13(12), 1525-1535.

Morand, S., Cribb, T. H., Kulbicki, M., Rigby, M. C., Chauvet, C., Dufour, V., ... & Pichelin, S. (2000). Endoparasite species richness of New Caledonian butterfly fishes: host density and diet matter. Parasitology, 121(1), 65-73

Moravec, F., Fredensborg, B. L., Latham, A. D. M., & Poulin, R. (2003). Larval spirurida (Nematoda) from the crab Macrophthalmus hirtipes in New Zealand. Folia Parasitologica, 50(2), 109-114.

Myers, R. A., Baum, J. K., Shepherd, T. D., Powers, S. P., & Peterson, C. H. (2007). Cascading effects of the loss of apex predatory sharks from a coastal ocean. Science, 315(5820), 1846-1850.

Naylor, J., Webber, W. & Booth, J. (2005). A guide to common offshore crabs in New Zealand waters. New Zealand Aquatic Environment and Biodiversity Report No. 2. 47 p.

Near, T. J. (2002). Acanthocephalan phylogeny and the evolution of parasitism. Integrative and Comparative Biology, 42(3), 668-677.

Nelson, J. S., Grande, T. C., & Wilson, M. V. (2016). Fishes of the World. John Wiley & Sons.

Nelson, D. R., & Johnson, R. H. (1970). Diel activity rhythms in the nocturnal, bottom-dwelling sharks, Heterodontus francisci and Cephaloscyllium ventriosum. Copeia, 732-739.

O'Connor, B. M. (1982). Evolutionary ecology of astigmatid mites. Annual review of entomology, 27(1), 385-409.

Oksanen, J., Blanchet, F. G., Kindt, R., Legendre, P., Minchin, P. R., O'hara, R. B., ... & Oksanen, M. J. (2013). *Package 'vegan'*. Community ecology package, version, 2(9).

Palm, H. (2004). The Trypanorhyncha Diesing, 1863. PKSPL -IPB Press, Bogor, 710 pp.

Palm, H. W., & Caira, J. N. (2008). Host specificity of adult versus larval cestodes of the elasmobranch tapeworm order Trypanorhyncha. International Journal for Parasitology, 38(3), 381-388.

Palm, H. W., Waeschenbach, A., Olson, P. D., & Littlewood, D. T. J. (2009). Molecular phylogeny and evolution of the Trypanorhyncha (Platyhelminthes: Cestoda). Molecular Phylogenetics and Evolution, 52(2), 351-367.

Palm, H. W., Yulianto, I., & Piatkowski, U. (2017). Trypanorhynch Assemblages Indicate Ecological and Phylogenetical Attributes of Their Elasmobranch Final Hosts. Fishes, 2(2), 8.

Perlman, S. J., & Jaenike, J. (2003). Infection success in novel hosts: an experimental and phylogenetic study of Drosophila-parasitic nematodes. Evolution, 57(3), 544-557.

Poinar, G. O., & Kuris, A. M. (1975). Juvenile Ascarophis (Spirurida: Nematoda) parasitizing intertidal decapod Crustacea in California: with notes on prevalence and effects on host growth and survival. Journal of Invertebrate Pathology, 26(3), 375-382.

Posada, D. (2008). jModelTest: phylogenetic model averaging. Molecular biology and evolution, 25(7), 1253-1256.

Poulin, R. (1992). Determinants of host-specificity in parasites of freshwater fishes. International journal for parasitology, 22(6), 753-758.

Poulin, R. (1996). Sexual inequalities in helminth infections: a cost of being a male? The American Naturalist, 147(2), 287-295.

Poulin, R. (2000). Variation in the intraspecific relationship between fish length and intensity of parasitic infection: biological and statistical causes. Journal of Fish Biology, 56(1), 123-137.

Poulin, R. (2004). Macroecological patterns of species richness in parasite assemblages. Basic and Applied Ecology, 5(5), 423-434.

Poulin, R. (2010). Network analysis shining light on parasite ecology and diversity. Trends in parasitology, 26(10), 492-498.

Poulin, R. (2011). Evolutionary ecology of parasites. Princeton university press.

Poulin, R., Blasco-Costa, I., & Randhawa, H. S. (2016). Integrating parasitology and marine ecology: Seven challenges towards greater synergy. Journal of Sea Research, 113, 3-10.

Poulin, R., Krasnov, B. R., & Mouillot, D. (2011a). Host specificity in phylogenetic and geographic space. Trends in parasitology, 27(8), 355-361.

Poulin, R., Guilhaumon, F., Randhawa, H. S., Luque, J. L., and Mouillot, D. (2011b). Identifying hotspots of parasite diversity from species–area relationships: host phylogeny versus host ecology. Oikos, 120(5), 740-747.

Poulin, R., & Keeney, D. B. (2008). Host specificity under molecular and experimental scrutiny. Trends in parasitology, 24(1), 24-28.

Poulin, R., & Leung, T. L. F. (2011). Body size, trophic level, and the use of fish as transmission routes by parasites. Oecologia, 166(3), 731-738.

Poulin, R., & Morand, S. (2014). Parasite biodiversity. Smithsonian Institution.

Poulin, R., & Mouillot, D. (2005). Combining phylogenetic and ecological information into a new index of host specificity. Journal of Parasitology, 91(3), 511-514.

Poulin, R., & Rohde, K. (1997). Comparing the richness of metazoan ectoparasite communities of marine fishes: controlling for host phylogeny. Oecologia, 110(2), 278-283.

Powell, A. W. B. (1979). New Zealand Mollusca: marine, land, and freshwater shells. Collins.

Presswell, B., Poulin, R., & Randhawa, H. S. (2012). First report of a gryporhynchid tapeworm (Cestoda: Cyclophyllidea) from New Zealand and from an eleotrid fish, described from metacestodes and in vitro-grown worms. Journal of helminthology, 86(4), 453-464.

R Development Core Team (2012). R: A language and environment for statistical computing. Version 2.15.0. – <www.r-project.org>.

Randall, P. J., & Myers, A. A. (2001). Effects of resource matrix, gut region analysed and sample size on diet statistics in co-existing species of flatfish. Journal of the Marine Biological Association of the United Kingdom, 81(6), 1041-1048.

Randhawa, H. S. (2011). Insights using a molecular approach into the life cycle of a tapeworm infecting great white sharks. The Journal of parasitology, 97(2), 275-280.

Randhawa, H. S., & Brickle, P. (2011). Larval parasite gene sequence data reveal cryptic trophic links in life cycles of porbeagle shark tapeworms. Marine Ecology Progress Series, 431, 215-222.

Randhawa, H. S., & Burt, M. D. (2008). Determinants of host specificity and comments on attachment site specificity of tetraphyllidean cestodes infecting rajid skates from the northwest Atlantic. Journal of Parasitology, 94(2), 436-461.

Randhawa, H. S., & Poulin, R. (2009). Determinants and consequences of interspecific body size variation in tetraphyllidean tapeworms. Oecologia, 161(4), 759-769.

Randhawa, H. S., & Poulin, R. (2010). Determinants of tapeworm species richness in elasmobranch fishes: untangling environmental and phylogenetic influences. Ecography, 33(5), 866-877.

Randhawa, H. S., Poulin, R., & Krkošek, M. (2015). Increasing rate of species discovery in sharks coincides with sharp population declines: implications for biodiversity. Ecography, 38(1), 96-107.

Randhawa, H. S., Saunders, G. W., & Burt, M. D. B. (2007). Establishment of the onset of host specificity in four phyllobothriid tapeworm species (Cestoda: Tetraphyllidea) using a molecular approach. Parasitology, 134(9), 1291-1300.

Randhawa, H. S., Saunders, G. W., Scott, M. E., & Burt, M. D. B. (2008). Redescription of Pseudanthobothrium hanseni Baer, 1956 and description of P. purtoni n. sp.(Cestoda: Tetraphyllidea) from different pairs of rajid skate hosts, with comments on the host-specificity of the genus in the northwest Atlantic. Systematic parasitology, 70(1), 41-60.

Rasmussen, T. K., & Randhawa H. S. (2017). Host diet influences parasite diversity: A case study looking at tapeworm diversity among sharks. (research submitted to Oecologia for publication).

Read, C. P. (1951). The" crowding effect" in tapeworm infections. The Journal of Parasitology, 37(2), 174-178.

Roberts, L. S. (2000). The crowding effect revisited. Journal of Parasitology, 86(2), 209-211.

Roberts, C. D., Stewart, A. S., & Struthers, C. D. (2014). Checklist of the fishes of New Zealand. Museum of New Zealand Te Papa Tongarewa. [Accessed online 8th August 2015 at: http://www.tepapa.govt.nz/SiteCollection Documents/Research/Checklist-of-the-Fishes-of-New-Zealand.pdf].

Sakanari, J. A., & Moser, M. (1989). Complete life cycle of the elasmobranch cestode, Lacistorhynchus dollfusi Beveridge and Sakanari, 1987 (Trypanorhyncha). The Journal of parasitology, 806-808.

Salgado-Maldonado, G., and Pineda-López, R. F. (2003). The Asian fish tapeworm Bothriocephalus acheilognathi: a potential threat to native freshwater fish species in Mexico. Biological Invasions, 5(3), 261-268.

Sanmartın, M. L., Alvarez, M. F., Peris, D., Iglesias, R., & Leiro, J. (2000). Parasite community study of the undulate ray Raja undulata in the Ría of Muros (Galicia, northwest Spain). Aquaculture, 184(3), 189-201.

Schembri, P. J., & McLay, C. L. (1983). An annotated key to the hermit crabs (Crustacea: Decapoda: Anomura) of the Otago region (southeastern New Zealand). New Zealand journal of marine and freshwater research, 17(1), 27-35.

Shukhgalter, O. A., & Nigmatullin, C. M. (2001). Parasitic helminths of jumbo squid Dosidicus gigas (Cephalopoda: Ommastrephidae) in open waters of the central east Pacific. Fisheries Research, 54(1), 95-110.

Ŝimková, A., Morand, S., Matejusová, I., Jurajda, P. V., & Gelnar, M. (2001). Local and regional influences on patterns of parasite species richness of central European fishes. Biodiversity & Conservation, 10(4), 511-525.

Ŝimková, A., Verneau, O., Gelnar, M., and Morand, S. (2006). Specificity and specialization of congeneric monogeneans parasitizing cyprinid fish. Evolution, 60(5), 1023-1037.

Simpfendorfer, C. A., Goodreid, A. B., & McAuley, R. B. (2001). Size, sex and geographic variation in the diet of the tiger shark, Galeocerdo cuvier, from Western Australian waters. Environmental Biology of Fishes, 61(1), 37-46.

Smale, M. J. (1991). Occurrence and feeding of three shark species, Carcharhinus brachyurus, C. obscurus and Sphyrna zygaena, on the Eastern Cape coast of South Africa. South African Journal of Marine Science, 11(1), 31-42.

Smith, N. F. (2001). Spatial heterogeneity in recruitment of larval trematodes to snail intermediate hosts. Oecologia, 127(1).

Smolowitz, R. M., Bullis, R. A., Abt, D. A., & Leibovitz, L. (1993). Pathologic observations on the infection of Pagurus spp. by plerocercoids of Calliobothrium verticillatum (Rudolphi, 1819; Van Benden, 1850). Journal of Invertebrate Pathology, 62(2), 185-190.

Stevens, J. D., Bonfil, R., Dulvy, N. K., & Walker, P. A. (2000). The effects of fishing on sharks, rays, and chimaeras (chondrichthyans), and the implications for marine ecosystems. ICES Journal of Marine Science, 57(3), 476-494.

Stevens, J. D., & Lyle, J. M. (1989). Biology of three hammerhead sharks (Eusphyra blochii, Sphyrna mokarran and S. lewini) from northern Australia. Marine and Freshwater Research, 40(2), 129-146.

Stringell, T. B., Clerveaux, W. V., Godley, B. J., Kent, F. E., Lewis, E. D., Marsh, J. E., ... & Broderick, A. C. (2016). Taxonomic distinctness in the diet of two sympatric marine turtle species. Marine Ecology, 37(5), 1036-1049.

Stunkard, H. W. (1977). Studies on Tetraphyllidean and Tetrarhynchidean metacestodes from squids taken on the New England coast. The Biological Bulletin, 153(2), 387-412.

Taraschewski, H. (2006). Hosts and parasites as aliens. Journal of Helminthology, 80(02), 99-128.

Thieltges, D. W., Amundsen, P. A., Hechinger, R. F., Johnson, P. T., Lafferty, K. D., Mouritsen, K. N., ... & Poulin, R. (2013). Parasites as prey in aquatic food webs: implications for predator infection and parasite transmission. Oikos, 122(10), 1473-1482.

Thomas, F., & Poulin, R. (1998). Manipulation of a mollusc by a trophically transmitted parasite: convergent evolution or phylogenetic inheritance?. Parasitology, 116(5), 431-436.

Thomas, F., Poulin, R., & Brodeur, J. (2010). Host manipulation by parasites: a multidimensional phenomenon. Oikos, 119(8), 1217-1223.

Thompson, R. M., Mouritsen, K. N., & Poulin, R. (2005). Importance of parasites and their life cycle characteristics in determining the structure of a large marine food web. Journal of Animal Ecology, 74(1), 77-85.

Thompson, R. M., Poulin, R., Mouritsen, K. N., & Thieltges, D. W. (2013). Resource tracking in marine parasites: going with the flow?. Oikos, 122(8), 1187-1194.

Tricas, T. C. (1982). Bioelectric-mediated predation by swell sharks, Cephaloscyllium ventriosum. Copeia, 1982(4), 948-952.

Tyler II G.A. (2006). Tapeworms of Elasmobranchs (Part II). A monograph on the Diphyllidea (Platyhelminthes, Cestoda). Bulletin of the University of Nebraska State Museum, 20, 142 pp.

Van Oosten, A. R., Heylen, D. J., Elst, J., Philtjens, S., & Matthysen, E. (2016). An experimental test to compare potential and realised specificity in ticks with different ecologies. Evolutionary ecology, 30(3), 487-501.

Vázquez, N., & Cremonte, F. (2017). Review of Parasites and Pathologies of the Main Bivalve Species of Commercial Interest of Argentina and Uruguay, Southwestern Atlantic Coast. Arch Parasitol, 1(112), 2.

Vitone, N. D., Altizer, S., & Nunn, C. L. (2004). Body size, diet and sociality influence the species richness of parasitic worms in anthropoid primates. Evolutionary Ecology Research, 6(2), 183-199.

Vivares, C. (1971). Study of the parasites of brachyuran decapod crustaceans: Nemertea and cestode larvae. Annales de Parasitologie Humaine et Comparee, 46(1), 1-9.

Von Euler, F., & Svensson, S. (2001). Taxonomic distinctness and species richness as measures of functional structure in bird assemblages. Oecologia, 129(2), 304-311.

Walther, B. A., Cotgreave, P., Price, R. D., Gregory, R. D., & Clayton, D. H. (1995). Sampling effort and parasite species richness. Parasitology Today, 11(8), 306-310.

Ward, P., & Myers, R. A. (2005). Shifts in open-ocean fish communities coinciding with the commencement of commercial fishing. Ecology, 86(4), 835-847.

Warwick, R. M., & Clarke, K. R. (2001). Practical measures of marine biodiversity based on relatedness of species. Oceanography and Marine Biology, 39, 207-231.

Wilga, C. D., & Motta, P. J. (2000). Durophagy in sharks: feeding mechanics of the hammerhead Sphyrna tiburo. Journal of Experimental Biology, 203(18), 2781-2796.

Wilkens, S. & Ahyong, S. (2015). Coastal crabs: a guide to the crabs of New Zealand. National Institute of Water and Atmospheric Research, 1. [Accessed online 16th August 2015 from: https://www.niwa.co.nz/coasts-and-oceans/marine-identification-guides-and-fact-sheets].

Williams, H. (1960). The Intestine in Members of the Genus Raja and Host-specificity in the Tetraphyllidea. Nature, 188: 514–516.

Williams, H. (1966). The ecology, functional morphology and taxonomy of Echeneibothrium Beneden, 1849 (Cestoda: Tetraphyllidea), a revision of the genus and comments on Discobothrium Beneden, 1870, Pseudanthobothrium Baer, 1956, and Phormobothrium Alexander, 1963. Parasitology, 56(02), 227-286.

Williams, H. (1968). The taxonomy, ecology and host-specificity of some Phyllobothriidae (Cestoda: Tetraphyllidea), a critical revision of Phyllobothrium Beneden, 1849 and comments on some allied genera.

Philosophical Transactions of the Royal Society of London. Series B, Biological Sciences, 231-307.

Williams, H. H., Jones, A., & Crompton, D. W. T. (1994). Parasitic worms of fish. Taylor & Francis, London.

Winter, M., Devictor, V., & Schweiger, O. (2013). Phylogenetic diversity and nature conservation: where are we?. Trends in Ecology & Evolution, 28(4), 199-204.

Wood, C. L., & Johnson, P. T. (2015). A world without parasites: exploring the hidden ecology of infection. Frontiers in Ecology and the Environment, 13(8), 425-434.

WoRMS Editorial Board 2017. World Register of Marine Species. <www.marinespecies.org>.

Zuk, M. (1990). Reproductive strategies and disease susceptibility: an evolutionary viewpoint. Parasitology today, 6(7), 231-233.

Appendix

.

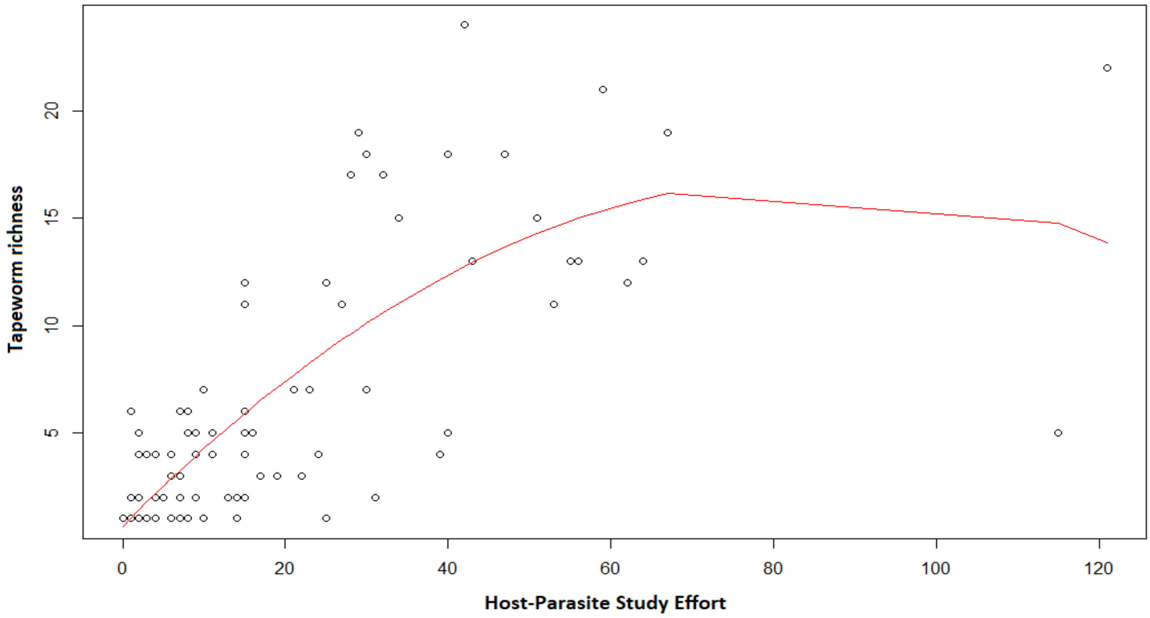

Figure A.1. Scatter plot showing the relationship between tapeworm richness and host-parasite study effort of shark species (n = 91). Tapeworm richness represents the sum of all tapeworm species known to infect the shark species (as adult stages) and host-parasite study effort is the total number of references obtained by searching parasite records for the host species on ISI Web of Knowledge. The solid line shows the fit of a quadratic regression of tapeworm richness on study effort.

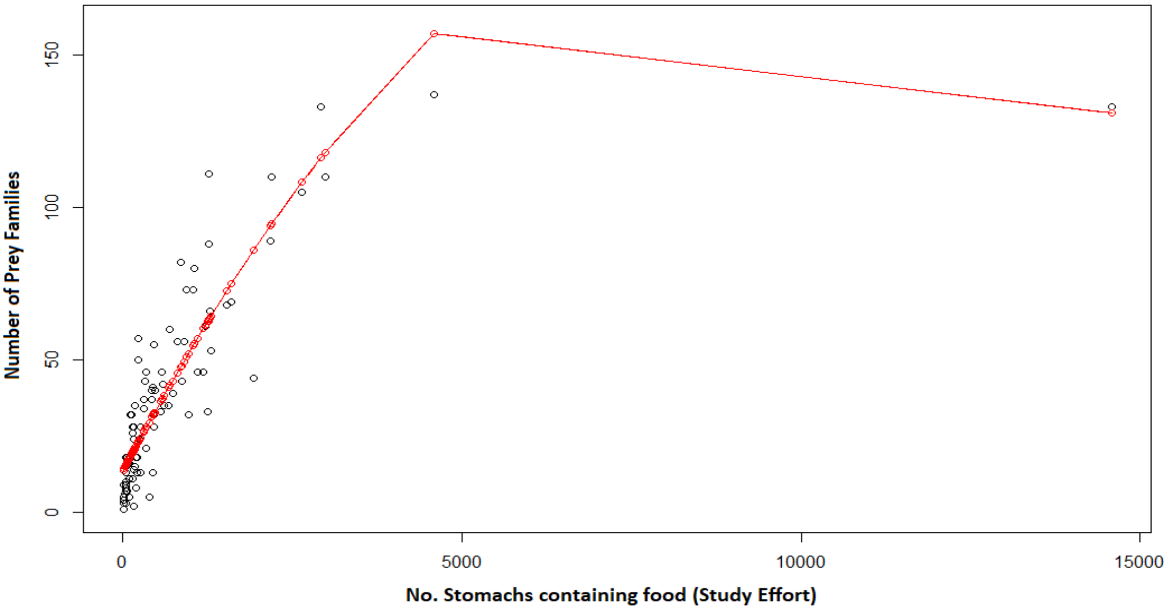

Figure A.2. Scatter plot showing the relationship between number of prey families (diet breadth) and the number of stomachs examined containing food (study effort for diet) for 91 shark species. Number of prey families is how many different taxonomic families were recorded across all diet references available for a species on ISI Web of Knowledge and number of stomachs containing food represents the sum of how many stomachs were examined for diet contents across these references. The solid line shows the fit of a quadratic regression of number of prey families on study effort.

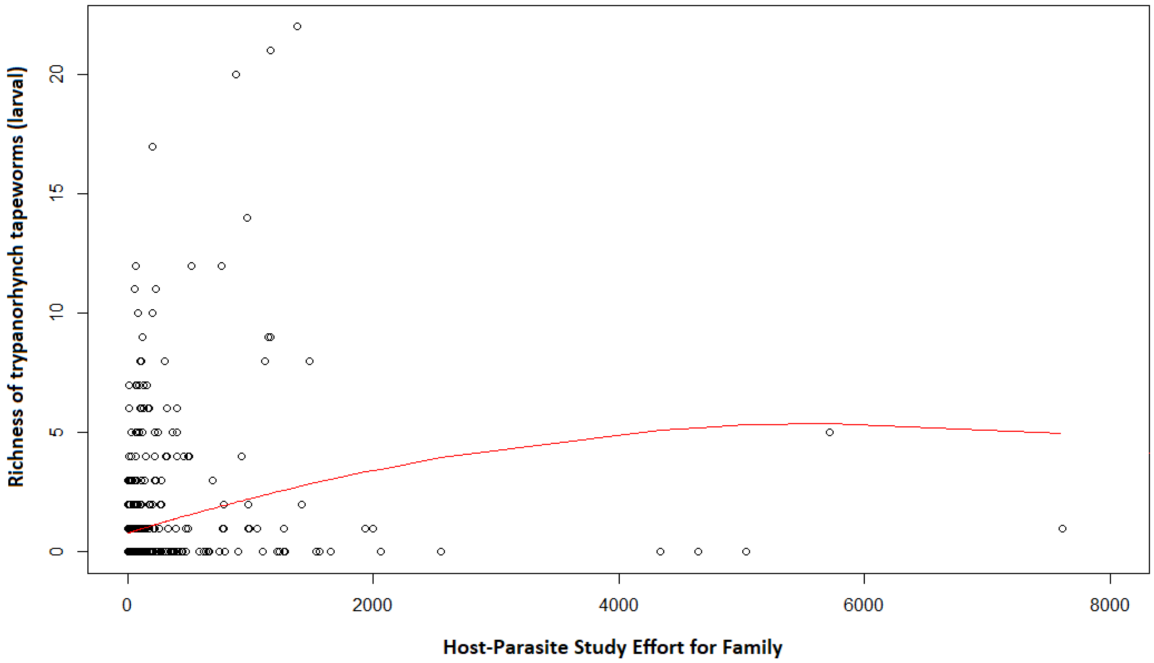

Figure A.3. Scatter plot showing the relationship between richness of trypanorhynch tapeworms (larval stages) known to infect an animal family and the study effort of the family for parasites (n = 626). Study effort represents the total number of references obtained by searching parasite records for the animal family on ISI Web of Knowledge. The solid line shows the fit of a quadratic regression of trypanorhynch tapeworm richness on study effort.

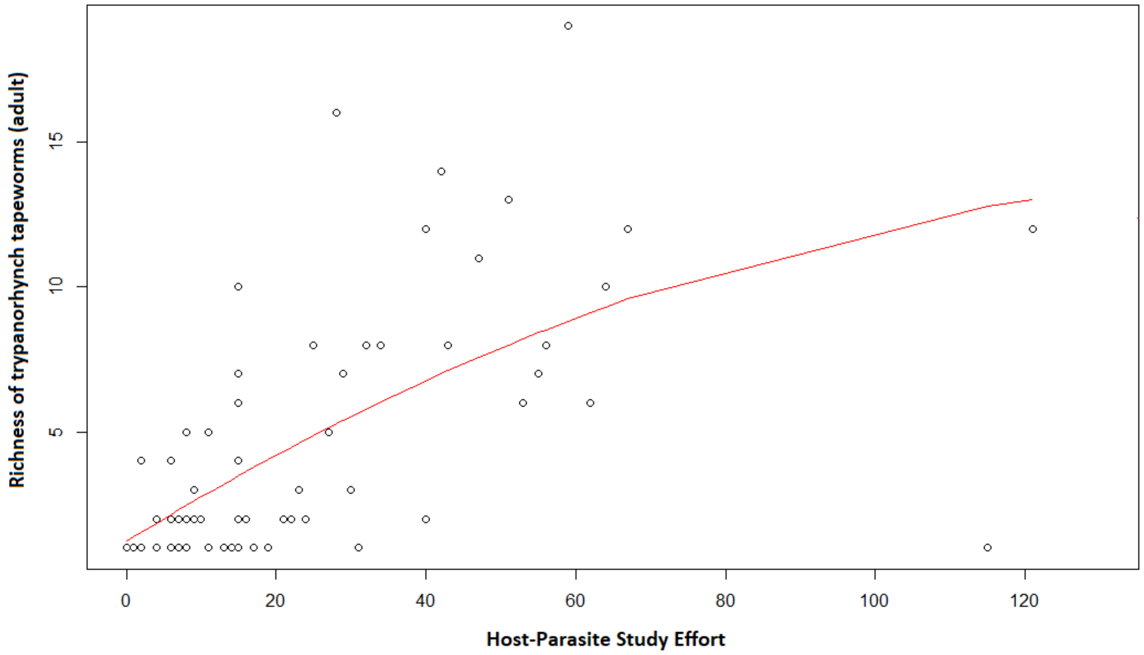

Figure A.4. Scatter plot showing the relationship between richness of trypanorhynch tapeworms (adult stages) known to infect a shark species and the study effort of the shark species for parasites (n = 71). host-parasite study effort is the total number of references obtained by searching parasite records for the shark species on ISI Web of Knowledge. The solid line shows the fit of a quadratic regression of trypanorhynch tapeworm richness on study effort.

Table A.1. Table displaying tapeworm species richness and diet breadth (prey family richness) for 91 different shark species. For each shark host species, families of prey are listed with their respective references indicated in brackets following each name. This dataset on shark families was obtained from ISI Web of Knowledge. Taxa of each shark here (Latin name plus all known synonyms) combined with keywords ‘‘diet* OR feed* OR prey*’’ were searched on ISI Web of Knowledge (all databases) and all available references from 1864 to 2017 were perused for records on shark diets (Searches conducted April 2017). The total number of references listed, references with diet information, and sum of stomachs examined containing food are given for each shark species. The total sum of stomachs examined (including empty stomachs) is included in parentheses and indicates minimum estimates.

Shark Species	**Total No. Prey Families**	**List of prey families in diet of shark species** [references]	**Total No. Refs**	**Refs with diet info**	**Stomachs examined with food** (total Inc. empty)
Alopias superciliosus	42	Alepisauridae [8], Ancistrocheiridae [1-2,6], Belonidae [1-2,5-6], Carangidae [4], Clupeidae [1-2,4-6,8], Coryphaenidae [1-2,5-6], Cranchiidae [4], Echeneidae [1-2,5], Engraulidae [4], Enoploteuthidae [1-2,6], Exocoetidae [1,5-6], Fistulariidae [1-2,5-6], Gonatidae [4], Hemiramphidae [5], Histioteuthidae [1-2,4-6], Istiophoridae [8], Loliginidae [2,4-5], Lutjanidae [1,6], Mastigoteuthidae [1-2,5-6], Merlucciidae [1-2,4-6], Munididae [4], Myctophidae [1-2,5], Octopoteuthidae [1,4,6], Ommastrephidae [1-2,4-7], Ophichthidae [1-2,6], Ophidiidae [5], Paralepididae [4], Penaeidae [5], Pholidoteuthidae [2], Sciaenidae [1-2,5-6], Scomberesocidae [4], Scombridae [1-2,4-6,8], Scorpaenidae [1,6], Sebastidae [4], Serranidae [1,6], Solenoceridae [6], Steniteuthidae [1], Synodontidae [1], Tetraodontidae [1-2,5-6], Thysanoteuthidae [2], Trachipteridae [4,6], Trichiuridae [3],	26	8	448 (523)
Alopias vulpinus	21	Arripidae [9], Atherinidae [10,12], Berycidae [9], Carangidae [9-10,12-13], Clupeidae [9-12], Engraulidae [9-12], Gadidae [13], Gonatidae [12], Loliginidae [10-12], Luvaridae [12], Merlucciidae [10-12], Munididae [12], Ommastrephidae [10-11], Paralepididae [10], Paralichthyidae [10,12], Sciaenidae [12], Scomberesocidae [10], Scombridae [11-13], Sebastidae [10-12], Sphyraenidae	31	5	349 (506)

		[12], Stromateidae [12]			
Carcharhinus acronotus	5	Loliginidae [14], Muraenidae [14], Octopodidae [14], Pomacentridae [14], Serranidae [14]	12	1	19 (86)
Carcharhinus amblyrhynchoides	14	Ariidae [17], Carangidae [17], Clupeidae [17], Hemiramphidae [36], Labridae [17], Leiognathidae [17,36], Ophichthidae [17], Penaeidae [17,36], Platycephalidae [17], Plotosidae [17], Scombridae [17], Sillaginidae [17], Squillidae [17], Terapontidae [17]	2	2	163 (241)
Carcharhinus amblyrhynchos	24	Acanthuridae [16], Apogonidae [37], Ariidae [38,285], Carangidae [38,285], Chaetodontidae [16], Clupeidae [38], Elapidae [37], Gerreidae [36], Hemiramphidae [38], Holocentridae [16], Leiognathidae [36], Monacanthidae [16], Mugilidae [38], Mullidae [36], Muraenidae [16,37-38], Octopodidae [37], Palinuridae [15], Penaeidae [36,285], Pomacentridae [16], Scaridae [16], Scombridae [17], Scorpaenidae [16], Sepiidae [37], Zanclidae [16]	25	9	173 (400)
Carcharhinus amboinensis	50	Acanthuridae [19], Ariidae [17-19], Carangidae [18-19], Carcharhinidae [18-20], Chanidae [18], Chirocentridae [19], Clupeidae [17], Cynoglossidae [18], Dasyatidae [19], Delphinidae [18], Diodontidae [18], Elapidae [19-20], Engraulidae [20], Gobiidae [19], Gymnuridae [18], Haemulidae [18], Istiophoridae [17], Labridae [19], Leiognathidae [18-19], Loliginidae [17,19], Megalopidae [19], Mugilidae [18], Myliobatidae [18], Octopodidae [18], Ommastrephidae [19], Oplegnathidae [18], Penaeidae[19], Platycephalidae [18], Pomacanthidae [19], Pomatomidae [18], Polynemidae [17,19-20], Portunidae [17], Psettodidae [19], Rhinobatidae [18], Sciaenidae [18-19], Scombridae [17-19], Scyliorhinidae [18], Sepiidae [18-19], Sepiolidae [19], Serranidae [18], Sparidae [18-19], Sphyrnidae [18], Squatinidae [18], Squillidae [19], Synodontidae [18], Terapontidae [19], Tetraodontidae [19], Triacanthidae [20], Triacanthodidae [20], Trichiuridae [18]	9	4	227 (338)
Carcharhinus brachyurus	56	Alcyoniidae [22], Apogonidae [23], Arhynchobatidae [22], Arripidae [9], Atherinidae [22-23], Batrachoididae [22], Callianassidae [23], Callorhinchidae [22-23], Carangidae [9,21,23], Cheilodactylidae [23], Clupeidae [9,21-23],	12	4	895 (2264)

		Congridae [22], Delphinidae [21], Dussumieriidae [21,23], Engraulidae [9,22-23], Gadidae [9], Gempylidae [9], Haemulidae [21,23], Hemiramphidae [9], Labridae [9], Loliginidae [9,21,23,39], Merlucciidae [23], Monacanthidae [9], Mugilidae [9,21,23], Mullidae [21], Myliobatidae [21-23], Nassariidae [22], Octopodidae [23], Odacidae [9], Ommastrephidae [9,23], Ophidiidae [9,23], Ovalipidae [9], Penaeidae [9,23], Pentanchidae [21], Percophidae [22], Pinguipedidae [22], Platycephalidae [9], Plotosidae [9], Pomatomidae [21], Rajidae [9,22], Rhinobatidae [21,23], Sciaenidae [9,22-23], Scombridae [9,21,23], Scorpaenidae [9], Sepiidae [9,21,23], Serranidae [22-23], Sillaginidae [9], Sparidae [9,21-23], Sphyraenidae [9], Squalidae [21,23], Squatinidae [21-22], Stromateidae [22], Terapontidae [9], Triakidae [22], Trichiuridae [21], Urolophidae [9]			
Carcharhinus brevipinna	33	Ancistrocheiridae [26], Ariidae [25-26], Carangidae [17,24,26], Carcharhinidae [26], Clupeidae [17,24-25], Engraulidae [24-27], Exocoetidae [24], Gerreidae [17,26], Haemulidae [24,26], Leiognathidae [17,26], Loliginidae [26], Monacanthidae [17], Monodactylidae [26], Mugilidae [24,26], Mullidae [17,26], Muraenidae [24], Nemipteridae [17], Octopodidae [24,26], Paralichthyidae [24], Pomacanthidae [26], Pomatomidae [26], Rhinobatidae [26], Sciaenidae [25-26], Scombridae [17,24,26-27], Sepiidae [26], Soleidae [26], Sparidae [26], Sphyraenidae [24,26], Sphyrnidae [26], Syngnathidae [25], Synodontidae [24,26], Trichiuridae [26], Triglidae [26]	27	6	559 (2038)
Carcharhinus dussumieri	40	Anguillidae [17], Apogonidae [17], Balistidae [17], Bothidae [17], Bregmacerotidae [17], Callianassidae [17], Callionymidae [17], Carangidae [38], Centriscidae [17], Clupeidae [17], Congridae [17], Cynoglossidae [17], Engraulidae [17,38,40], Fistulariidae [17], Gerreidae [36], Gobiidae [17,38], Hemiramphidae [36], Hypoptychidae [17], Labridae [17,38], Leiognathidae [17,36,38,40], Loliginidae [17], Monacanthidae [17,36], Mugilidae [17,36], Mullidae [17], Muraenesocidae [17], Muraenidae [17], Myctophidae [17], Nemipteridae [17], Ogcocephalidae [17], Penaeidae [17,36, 40], Platycephalidae [17], Portunidae [17], Priacanthidae [17], Raninidae [17], Scombridae [17],	4	4	470 (695)

		Scorpaenidae [17], Squillidae [17], Synodontidae [17], Tetraodontidae [17], Uranoscopidae [17]			
Carcharhinus falciformis	44	Alloposidae [29], Amphitretidae [29], Ancistrocheiridae [2,28-30], Argonautidae [2,17,28-30], Balistidae [2,17,28,30-31], Carangidae [2,28-30], Clupeidae [30], Coryphaenidae [2,28-30], Diodontidae [29], Echeneidae [29], Enoploteuthidae [28], Exocoetidae [29-31], Galatheidae [29], Gempylidae [28], Gobiidae [31], Gonatidae [2,30], Haemulidae [30], Hemiramphidae [29], Histioteuthidae [2,30], Istiophoridae [29], Kyphosidae [29], Labridae [2,30], Mastigoteuthidae [29], Molidae [29], Monacanthidae [17,28], Mugilidae [41], Munididae [29-30], Myctophidae [28-29], Nomeidae [28-29], Octopodidae [28], Octopoteuthidae [29], Ommastrephidae [2,28-31], Onychoteuthidae [2,28-29], Ostraciidae [29], Penaeidae [2], Polynemidae [31], Portunidae [17,28-29,31], Priacanthidae [2,28,30], Scombridae [2,27-31], Sepiidae [17], Sphyraenidae [28,30], Tetraodontidae [29-30], Tremoctopodidae [29], Vitreledonellidae [29]	31	7	864 (1447)
Carcharhinus galapagensis	16	Acanthuridae [32], Balistidae [32], Belonidae [32], Carangidae [32], Clupeidae [32], Diodontidae [32], Holocentridae [32], Lethrinidae [32], Monacanthidae [32], Muraenidae [32], Pomacentridae [32], Priacanthidae [32], Scaridae [32], Scombridae [32], Serranidae [32], Synodontidae [32]	10	3	96 (178)
Carcharhinus isodon	11	Carangidae [25,33], Carcharhinidae [34], Clupeidae [25,33-35], Elopidae [25], Engraulidae [25], Penaeidae [34-35], Portunidae [33], Sciaenidae [25,33-34], Scombridae [33-34], Sparidae [25], Syngnathidae [25]	10	4	142 (293)
Carcharhinus leucas	73	Achiridae [46], Ambassidae [19], Anguillidae [19], Ariidae [19,42,45-47], Batrachoididae [46], Bradypodidae [44], Carangidae [19,42,45-46], Carcharhinidae [19,42,45-46], Centropomidae [19,44], Characidae [44], Cheilodactylidae [42], Cheloniidae [42,44,47], Cichlidae [42,44], Clariidae [42], Clupeidae [42-43,46-47], Coryphaenidae [42], Crocodylidae [47], Dasyatidae [19,42,45-46], Dermochelyidae [42], Dinopercidae [42], Drepaneidae [42], Elapidae [43], Eleotridae [44], Elopidae [46], Engraulidae [42], Ephippidae [42], Gecarcinidae [44], Gerreidae [45], Gobiidae [19,45], Haemulidae [42,44-45],	88	7	1034 (1967)

		Hemiramphidae [47], Hominidae [42], Labridae [42], Lamnidae [42], Latidae [47], Leiognathidae [19], Lepisosteidae [44], Lutjanidae [44], Megalopidae [44], Monodactylidae [42], Mugilidae [42,44,46-47], Muraenidae [42], Myliobatidae [42,45], Octopodidae [42], Odontaspididae [42], Ophichthidae [46], Osteoglossidae [19,47], Ovalipidae [42], Palaemonidae [19,47], Penaeidae [46], Platycephalidae [42], Plotosidae [42], Polynemidae [47], Pomatomidae [42], Portunidae [45-46], Pristidae [44,47], Rhinobatidae [42], Sciaenidae [42,45-47], Scombridae [19,42,45], Scyliorhinidae [42], Sepiidae [42], Serranidae [42], Sesarmidae [42], Sparidae [42,46], Sphyrnidae [42,45], Squalidae [42], Squatinidae [42], Stegostomatidae [42], Suidae [47], Synbranchidae [19], Synodontidae [42,46-47], Triakidae [42], Tyrannidae [44]			
Carcharhinus limbatus	72	Acanthuridae [54], Achiridae [33], Albulidae [54,56], Ariidae [25,38,48,51,55], Atherinopsidae [53], Balistidae [48,50,56], Batrachoididae [55], Belonidae [48,54], Berycidae [48], Blenniidae [48], Bothidae [33,53], Carangidae [25,48,50,53-54], Carcharhinidae [48,55], Chaetodontidae [54,56], Cichlidae [48], Clupeidae [25,33,35,38,40,48,50-51,53-56], Cynoglossidae [38,48], Dasyatidae [33], Delphinidae [48], Dinopercidae [48], Echeneidae [48], Elopidae [25,48], Engraulidae [25,48,50,53], Ephippidae [51], Gerreidae [54,56], Gymnuridae [48], Haemulidae [48,50,54,56], Hemiramphidae [50], Holocentridae [56], Kyphosidae [48], Labridae [48], Leiognathidae [40,48], Lethrinidae [38], Loliginidae [25,48,50,53], Lutjanidae [25,48,54,56-57], Megalopidae [51], Monodactylidae [48], Mugilidae [48], Mullidae [56], Muraenidae [49], Myliobatidae [50,55], Octopodidae [48], Ophichthidae [33], Oplegnathidae [48], Ostraciidae [48], Palinuridae [48], Paralichthyidae [50-51,55], Penaeidae [25,33,50-53], Plotosidae [48], Pomacanthidae [48], Pomacentridae [48,54], Pomatomidae [48], Rajidae [50], Rhinobatidae [48], Scaridae [54,56], Sciaenidae [25,33,38,48,50-51,53,55], Scombridae [27,48,55-57], Scyliorhinidae [48], Sepiidae [48], Serranidae [48], Sillaginidae [38], Sparidae [25,48,54,56-57], Sphyraenidae [49], Sphyrnidae [48,55], Squillidae [52], Stromateidae [33,53], Syngnathidae [25], Synodontidae	64	16	1624 (3056)

		[25,40,48], Terapontidae [40], Tetraodontidae [48], Triacanthidae [38], Trichiuridae [33,48,50,53]			
Carcharhinus longimanus	9	Alepisauridae [58], Berycidae [27], Bramidae [58], Carangidae [27], Clupeidae [58], Coryphaenidae [59], Pomatomidae [27], Scombridae [58], Sparidae [27]	20	3	44 (89)
Carcharhinus macloti	11	Carangidae [17], Chirocentridae [17], Clupeidae [17], Dasyatidae [17], Engraulidae [17], Hoplichthyidae [17], Leiognathidae [17], Nemipteridae [17], Platycephalidae [17], Scombridae [17], Synodontidae [17]	2	1	91 (216)
Carcharhinus melanopterus	37	Acanthuridae [60], Aplysinidae [60], Apogonidae [63], Acrochordidae [63], Atherinidae [37], Balistidae [60], Carangidae [36], Chanidae [63], Elapidae [37,63], Ephippidae [63], Fistulariidae [37], Gerreidae [36], Haemulidae [36], Haliotidae [37], Labridae [37,60,63], Leiognathidae [36], Lethrinidae [60], Loliginidae [37], Lutjanidae [37,63], Monacanthidae [63], Mullidae [36,60], Muraenidae [37], Muridae [61], Octopodidae [37], Platycephalidae [63], Pomacanthidae [37], Portunidae [60], Scaridae [37,60], Sparidae [63], Sternidae [37], Sulidae [61], Synodontidae [37],	45	6	133 (231)
Carcharhinus obscurus	105	Acanthuridae [67,319], Achiridae [66], Albulidae [65], Anguillidae [65,319], Apogonidae [65], Ariidae [23,64], Atherinidae [66], Aulopidae [65], Balistidae [64,66], Belonidae [319], Cancridae [66], Carangidae [9,50,64-65,67], Carcharhinidae [27,50,64-65,67], Cheilodactylidae [64-65], Cheloniidae [50], Chirocentridae [64], Chlorophthalmidae [64], Cichlidae [64], Clupeidae [9,23,64-65,67], Congridae [50,319], Cynoglossidae [64-65], Dactylopteridae [319], Dasyatidae [23,64-66], Delphinidae [64], Elopidae [64,67], Engraulidae [23,50,64,66-67], Enoploteuthidae [319], Ephippidae [50,319], Exocoetidae [64], Fistulariidae [68], Gempylidae [9,64], Gerreidae [64-65], Gobiidae [23,67], Gymnuridae [64], Haemulidae [23,64,67], Hemiramphidae [65], Heterodontidae [9,65], Istiophoridae [319], Kyphosidae [64-65], Labridae [65], Lamnidae [319], Leiognathidae [64], Lethrinidae [67], Limulidae [66], Loliginidae [23,50,64,66-67], Lophiidae [66], Lutjanidae [50], Lycoteuthidae [319], Macrouridae [64], Matutidae [67], Monacanthidae [65], Mugilidae [23,64-65,67], Mullidae [50,64-	41	10	2466 (6467)

		65,67], Muraenidae [64], Myctophidae [319], Myliobatidae [64-65], Nassariidae [66-67], Neosebastidae [65], Octopodidae [9,23,64,67,319], Octopoteuthidae [319], Odacidae [9], Odontaspididae [64], Ommastrephidae [9,64], Ophidiidae [9], Oplegnathidae [64], Ostraciidae [64,68], Ovalipidae [9,66], Palinuridae [23,64,67], Paralichthyidae [50,65-66], Penaeidae [9,67], Pentacerotidae [65], Pentanchidae [64], Peristediidae [319], Platycephalidae [65,319], Pleuronectidae [319], Plotosidae [64-65], Pomacentridae [65,319], Pomatomidae [64,66-68], Priacanthidae [319], Pristigasteridae [64], Rajidae [50,64,66], Rhinobatidae [23,64-65], Scaridae [65], Sciaenidae [23,50,64,66-67], Scombridae [9,23,64-65,67], Scyliorhinidae [319], Sepiidae [9,23,64,67], Serranidae [23,64-65], Sillaginidae [9], Sparidae [9,23,64,67], Sphyraenidae [9,64,67], Sphyrnidae [64-65], Spirulidae [67], Squalidae [319], Squatinidae [64], Stromateidae [66], Syngnathidae [65,319], Synodontidae [50,64,67], Terapontidae [319], Triakidae [64-65,67], Trichiuridae [23,50,67,319], Triglidae [50,66-67,319], Uranoscopidae [9,65-66], Urolophidae [65], Zeidae [23]			
Carcharhinus plumbeus	110	Acanthuridae [70], Achiridae [69], Acropomatidae [73], Ammodytidae [17,69], Anguillidae [69,74-75], Apogonidae [73], Atherinopsidae [74], Aulostomidae [70], Balistidae [17,70,72], Belonidae [70,72], Bothidae [17,70-71], Callianassidae [69], Callionymidae [70], Cancridae [69,71,75], Caproidae [17], Carangidae [17,69-73], Carcharhinidae [69], Carcinidae [69], Centrolophidae [73], Chaetodontidae [70], Champsodontidae [17], Chauliodontidae [71], Clupeidae [69,71-72,74-75,81], Congridae [17,69-71,73], Cottidae [71], Crangonidae [69], Cynoglossidae [69], Dasyatidae [69,71], Diodontidae [70], Engraulidae [69,73-74,81], Ephippidae [69], Epialtidae [69], Exocoetidae [70], Fistulariidae [69-70], Fundulidae [69,74-75], Gadidae [71], Gonatidae [71], Holocentridae [70], Labridae [17,70-71], Leiognathidae [17], Lethrinidae [17], Leucosiidae [69], Limulidae [69], Littorinidae [69], Loliginidae [17,69,72,74,81], Lophiidae [69,71], Lutjanidae [70], Lysmatidae [69], Mactridae [69], Majidae [69], Merlucciidae [71], Monacanthidae [70], Monocentridae [17], Moronidae [69], Mugilidae [69,72-	87	10	2022 (2757)

		73,81], Mullidae [17,70,72], Muraenidae [70], Myliobatidae [69], Mytilidae [69], Nassariidae [69], Naticidae [69], Nemipteridae [17], Nuculidae [69], Octopodidae [72,76], Ommastrephidae [69,71,73], Ophidiidae [69,71], Ophichthidae [71], Ophiuridae [70], Ostraciidae [70], Ovalipidae [69,74-75], Paguridae [69], Paralichthyidae [69,74], Penaeidae [17,69,72], Pharidae [69], Phocoenidae [70], Phycidae [69,81], Pinguipedidae [17], Pleuronectidae [69,71], Pomacanthidae [17], Pomatomidae [69,71,74-75], Portunidae [69,74-75], Priacanthidae [17,70], Rachycentridae [69], Rajidae [69,71-72], Scaridae [70,76], Sciaenidae [69,74-75,81], Scombridae [17,70-72], Scophthalmidae [69], Scorpaenidae [70], Scutellidae [71], Scyliorhinidae [17], Sepiidae [72], Serranidae [69], Soleidae [72], Sparidae [69,72,81], Sphyraenidae [70], Squalidae [71], Squatinidae [81], Squillidae [69,72,74-75], Stromateidae [71,73,81], Syngnathidae [69-70,74], Synodontidae [17,70-71], Tetraodontidae [17,69-70,75], Triakidae [69,71,74,81], Trichiuridae [17,73], Triglidae [69-71,81], Upogebiidae [69], Uranoscopidae [69], Zanclidae [70], Zoarcidae [71]			
Carcharhinus porosus	15	Achiridae [77], Ariidae [77], Carcharhinidae [77], Clupeidae [77], Dasyatidae [77], Engraulidae [77], Ephippidae [77], Loliginidae [77], Mugilidae [77], Penaeidae [77], Polynemidae [77], Portunidae [77], Sciaenidae [77], Stromateidae [77], Trichiuridae [77]	4	1	171 (684)
Carcharhinus sealei	6	Carangidae [78], Clupeidae [78], Hemiramphidae [78], Labridae [78], Lutjanidae [78], Nemipteridae [78]	1	1	30 (108)
Carcharhinus signatus	13	Acanthuridae [79], Bramidae [79], Chiroteuthidae [79], Cranchiidae [79], Histioteuthidae [79], Howellidae [79], Myctophidae [79], Octopodidae [79], Octopoteuthidae [79], Ommastrephidae [79], Scombridae [79], Serranidae [79], Xiphiidae [79]	6	1	215 (415)
Carcharhinus sorrah	39	Ammodytidae [80], Apogonidae [20], Ariidae [80], Balistidae [80], Bothidae [20,80], Carangidae [20,36,80], Centriscidae [80], Chirocentridae [80], Clupeidae [20,80], Congridae [80], Dactylopteridae [80], Diodontidae [80], Exocoetidae [80], Fistulariidae [80], Gerreidae [20,80], Haemulidae [20,36], Leiognathidae	6	5	731 (1388)

		[20,36,40,80], Lethrinidae [80], Loliginidae [20], Menidae [20,80], Monacanthidae [36,80], Mullidae [20,36,80], Nemipteridae [80], Ostraciodontidae [80], Penaeidae [20,36,38,40], Polynemidae [20], Portunidae [80], Priacanthidae [80], Scaridae [80], Sciaenidae [20], Scombridae [80], Sillaginidae [20], Soleidae [20], Spirulidae [20], Synodontidae [40,80], Terapontidae [20], Tetraodontidae [20,80], Trichiuridae [80], Triglidae [80]			
Carcharhinus tilstoni	46	Anguillidae [80], Ariidae [20,38,80], Balistidae [80], Bothidae [80], Carangidae [20,80], Carcharhinidae [80], Clupeidae [20,38,40,80], Coryphaenidae [80], Cynoglossidae [38], Dactylopteridae [80], Diodontidae [80], Elapidae [80], Engraulidae [20], Exocoetidae [80], Gerreidae [20,36], Haemulidae [36,80], Hemigaleidae [80], Hemiramphidae [36,80], Leiognathidae [20,36,40,80], Lethrinidae [38,80], Loliginidae [20,80], Lutjanidae [80], Monacanthidae [80], Mullidae [20,36,80], Muraenesocidae [80], Myctophidae [80], Nemipteridae [80], Paralepididae [80], Penaeidae [20,36], Platycephalidae [80], Polynemidae [20], Priacanthidae [80], Psettodidae [80], Scaridae [80], Sciaenidae [36,38,80], Scombridae [20,40,80], Scorpaenidae [80], Sillaginidae [38], Sphyraenidae [80], Synodontidae [20,80], Terapontidae [40], Tetraodontidae [80], Triacanthidae [38,80], Trichiuridae [80], Triglidae [80], Uranoscopidae [80]	4	5	1192 (2402)
Carcharias taurus	56	Achiridae [66], Aphroditidae [83], Arhynchobatidae [83], Ariidae [82], Atherinidae [83], Batrachoididae [81,83], Bothidae [82], Carangidae [81-83], Carcharhinidae [66,81,82], Cheilodactylidae [82-83], Clupeidae [66,81-83], Congridae [83], Ctenodiscidae [83], Cynoglossidae [82], Dasyatidae [82], Dussumieriidae [82], Echeneidae [82], Gonorynchidae [82], Haemulidae [82], Hexanchidae [83], Labridae [66], Loliginidae [39,66,81-82], Lophiidae [66], Merlucciidae [82], Mugilidae [82], Myliobatidae [66,82-83], Nassariidae [83], Octopodidae [82], Ophidiidae [66,82], Paguridae [66,83], Paralichthyidae [66,83], Pentanchidae [82], Percophidae [81,83], Phycidae [83], Pinguipedidae [83], Platyxanthidae [83], Pomatomidae [66,82-83], Rajidae [66,82-83], Rhinobatidae [82], Sciaenidae [66,81-83], Scombridae [66,81-82], Scophthalmidae [66],	46	5	801 (1000)

		Scyliorhinidae [82], Sepiidae [82], Serranidae [83], Soleidae [82], Sparidae [66,81-82], Squalidae [82], Squatinidae [66,83], Stromateidae [66,81,83], Synodontidae [66], Torpedinidae [82], Triakidae [66,81-83], Trichiuridae [81], Triglidae [66,82-83], Uranoscopidae [83]			
Carcharodon carcharias	43	Acipenseridae [94], Alopiidae [91], Ariidae [84], Asteriidae [85], Bursidae [94], Cancridae [85,88,94], Carcharhinidae [84], Cetorhinidae [89,94], Chiroteuthidae [91], Clupeidae [84-85,89,94], Cottidae [89,94], Dasyatidae [84,94], Delphinidae [84,90], Engraulidae [94], Haemulidae [91], Hexagrammidae [89,94], Loliginidae [91], Merlucciidae [85,88], Moronidae [94], Myliobatidae [84,89,94,96], Odontaspididae [84], Otariidae [84,87,94-95], Phocidae [88,92-94,97], Phocoenidae [86], Phycidae [85], Pleuronectidae [85,92], Pomatomidae [85], Rajidae [85], Rhincodontidae [91], Rhinobatidae [84], Salmonidae [88,92,94], Sciaenidae [84,89,94,96], Scombridae [84,96,98], Sebastidae [88,94], Sepiidae [84], Serranidae [91], Sparidae [84], Sphyraenidae [84], Sphyrnidae [84,91], Squalidae [84,89,94], Stromateidae [85], Triakidae [85,89,94,96], Triglidae [85]	266	17	329 (512)
Centrophorus granulosus	28	Apogonidae [100], Argentinidae [99], Brachioteuthidae [100], Calappidae [99], Callionymidae [99], Clupeidae [100], Cranchiidae [100], Etmopteridae [99], Gadidae [99], Geryonidae [99], Histioteuthidae [100], Lepidoteuthidae [100], Macrouridae [99], Myctophidae [99], Nephropidae [99], Octopoteuthidae [100], Ommastrephidae [99], Pandalidae [99], Pentanchidae [99], Phosichthyidae [99], Phycidae [99], Polybiidae [99], Rajidae [99], Scombridae [100], Sepiidae [99], Sepiolidae [99], Soleidae [100], Trachichthyidae [99]	11	3	153 (194)
Centrophorus squamosus	16	Alepocephalidae [102], Argentinidae [104], Bramidae [101], Carangidae [101,103], Chimaeridae [102], Congridae [101], Diretmidae [103], Gadidae [102], Macrouridae [102-104], Merlucciidae [101,103], Moridae [104], Onychoteuthidae [103], Oreosomatidae [101], Penaeidae [103], Sebastidae [101,104], Squalidae [101]	12	4	85 (466)
Centroscymnus coelolepis	18	Acanthephyridae [105], Alepocephalidae [102], Bramidae [103], Epigonidae [103], Gadidae [102], Histioteuthidae [105], Ipnopidae [105], Lotidae [104],	14	4	222 (379)

		Lycoteuthidae [103], Macrouridae [102,104], Merlucciidae [103], Moridae [104], Myctophidae [104], Octopoteuthidae [103], Ommastrephidae [105], Pleuronectidae [104] Sepiolidae [105], Stomiidae [105]			
Cephaloscyllium isabellum	28	Buccinidae [106], Cancridae [106], Carangidae [106], Centriscidae [106], Engraulidae [106], Goneplacidae [106], Macrouridae [106], Monacanthidae [106], Moridae [106], Munididae [106], Octopodidae [106], Ommastrephidae [106], Ophidiidae [106], Ostreidae [106], Ovalipidae [106], Palinuridae [106], Percophidae [106], Pinguipedidae [106], Pleuronectidae [106], Pseudarchasteridae [106], Pyuridae [106], Rajidae [106], Scyllaridae [106], Squalidae [106], Squillidae [106], Syngnathidae [106], Triakidae [106], Urechidae [106]	1	1	261 (278)
Cetorhinus maximus	3	Penaeidae [107], Sergestidae [108], Temoridae [109]	87	3	3 (6)
Chlamydoselachus anguineus	7	Chiroteuthidae [110], Gonatidae [110], Histioteuthidae [110], Mastigoteuthidae [110], Ommastrephidae [110], Onychoteuthidae [110], Rostellariidae [110]	6	1	37 (139)
Dalatias licha	26	Alpheidae [111], Aristeidae [111], Axiidae [111], Carapidae [113], Centrophoridae [101], Chlorophthalmidae [112], Epigonidae [111], Etmopteridae [111,113], Gadidae [111], Histioteuthidae [111], Loliginidae [112,115], Macrouridae [111], Merlucciidae [101,112,114], Moridae [111,113], Myctophidae [111-112], Nephropidae [113], Paralepididae [111], Pasiphaeidae [111,113], Penaeidae [112], Pentanchidae [111-113], Phycidae [111], Pyrosomatidae [113], Sepiolidae [111-112,114], Sergestidae [111], Stomiidae [111], Trichiuridae [115]	16	8	151 (210)
Deania calcea	32	Acanthephyridae [116], Alepocephalidae [117], Aphroditidae [117], Carangidae [116,118], Clupeidae [120], Cranchiidae [116], Cyttidae [116], Gadidae [102,117], Gempylidae [120], Gonostomatidae [117], Histioteuthidae [117], Lotidae [102], Macrouridae [116], Merlucciidae [116], Myctophidae [102-103,116-120], Notosudidae [116], Ommastrephidae [116-117,119], Onychoteuthidae [116],	11	7	455 (814)

		Oplophoridae [116], Pandalidae [120], Paralepididae [103], Pasiphaeidae [116-117], Penaeidae [103], Phosichthyidae [116], Phycidae [102], Scombridae [102,117], Sebastidae [102], Sergestidae [102,116-117], Sternoptychidae [116], Stomiidae [103,117], Trachichthyidae [116], Zeidae [116]			
Deania profundorum	3	Enoploteuthidae [103], Myctophidae [103], Ommastrephidae [103]	1	1	43 (78)
Etmopterus granulosus	7	Batoteuthidae [121], Brachioteuthidae [121], Histioteuthidae [103,121], Mastigoteuthidae [121], Myctophidae [103,121], Paralepididae [103], Penaeidae [103]	1	2	67 (191)
Etmopterus lucifer	5	Enoploteuthidae [122], Euphausiidae [122-123], Histioteuthidae [122], Lophogastridae [123], Myctophidae [122]	8	2	385 (681)
Etmopterus princeps	7	Acanthephyridae [124], Bathylagidae [102], Gadidae [124], Myctophidae [124], Notosudidae [124], Ommastrephidae [124], Paralepididae [124]	5	2	55 (98)
Etmopterus pusillus	13	Enoploteuthidae [125], Gadidae [125], Gonostomatidae [125], Histioteuthidae [125], Merlucciidae [103], Myctophidae [103,125], Ommastrephidae [125], Onychoteuthidae [125], Pandalidae [125], Pasiphaeidae [125], Polybiidae [125], Sepiolidae [125], Trichiuridae [125]	1	2	448 (605)
Etmopterus spinax	35	Alepocephalidae [127], Alpheidae [133], Aristeidae [126,133], Callionymidae [126], Centrolophidae [127], Crangonidae [126,131,133], Enoploteuthidae [133], Etmopteridae [126], Euphausiidae [102,117,127-133], Gadidae [102,117,126,131], Gnathophausiidae [117,127], Gobiidae [129], Goneplacidae [126], Histioteuthidae [117,129-130,133], Munididae [127], Myctophidae [102,126-127,129-130,133], Mysidae [102], Oplophoridae [117], Pandalidae [126,128,130-131], Paralepididae [130], Pasiphaeidae [127-131,133], Pentanchidae [126], Phosichthyidae [126], Phycidae [102,126-127], Processidae [127], Scombridae [128], Sepiidae [126], Sepiolidae [126,128-130,133], Sergestidae [102,129-130,133], Solenoceridae [127], Sternoptychidae [102,128,131-132], Stomiidae [130,133], Synaphobranchidae [102],	42	12	533 (866)

		Trachichthyidae [126], Trichiuridae [130]			
Euprotomicrus bispinatus	5	Bramidae [136], Myctophidae [136], Oncaeidae [136], Phosichthyidae [136], Sternoptychidae [136]	3	1	12 (12)
Galeocerdo cuvier	111	Acanthuridae [138], Ancistrocheiridae [332], Anguillidae [17,148], Argonautidae [332], Ariidae [76], Aulostomidae [138], Balistidae [17,50,76,138,140], Batrachoididae [137,143], Belonidae [138,143], Bovidae [138,141,147,153-155], Busyconidae [150], Canidae [138], Carangidae [27,50,137-138,147,156], Carcharhinidae [50,62,137,154], Chelonibiidae [17], Cheloniidae [137-138,149,153-154], Chirocentridae [156,332], Cirolanidae [139], Clupeidae [27,62,140,154,156], Columbidae [150,152], Congridae [138], Coryphaenidae [138], Cranchiidae [332], Cuculidae [150], Cycloteuthidae [332], Dasyatidae [50,76,145,154], Delphinidae [17,27,137], Diodontidae [27,50,76,137-138,143,145,147], Diomedeidae [17], Dugongidae [143,145,148], Elapidae [17,143,145,148,151], Elopidae [144], Engraulidae [156], Enoploteuthidae [332], Ephippidae [50,140], Equidae [138,153], Fasciolariidae [150], Felidae [138], Fistulariidae [138,147], Gerreidae [50,140], Gorgoniidae [137], Haemulidae [140], Hemiramphidae [143,145], Herpestidae [138], Histioteuthidae [332], Hominidae [138,146], Istiophoridae [17,138], Joubiniteuthidae [332], Labridae [138], Lamnidae [141], Laridae [137], Limulidae [137], Loliginidae [50,140,332], Luidiidae [137], Lutjanidae [145], Meropidae [141], Molidae [27], Monacanthidae [138,140], Muridae [138], Mugilidae [50,137], Mullidae [138], Muraenidae [138], Myliobatidae [50,145,154], Naticidae [137], Octopodidae [137,332], Octopoteuthidae [332], Ommastrephidae [332], Onychoteuthidae [332], Ostraciidae [17,138], Ovalipidae [137], Palinuridae [138,145], Paralichthyidae [137], Parulidae [150], Pelecanidae [149], Phalacrocoracidae [137], Phocidae [157], Phocoenidae [154], Pholidoteuthidae [332], Platycephalidae [27], Pleuronectidae [138], Pomacanthidae [17], Pomacentridae [138], Pontoporiidae [144], Portunidae [17,27,50,137,149], Pristidae [62], Procellariidae [27], Rachycentridae [137], Rajidae [137], Rallidae [142],	208	30	1359 (1944)

		Raninidae [147], Rhinobatidae [17,145], Sciaenidae [27,50,137,140,144,156], Scaridae [138], Scombridae [17,27,137-138,154], Scyllaridae [17,138,147], Sepiidae [332], Serranidae [137], Sparidae [50,137], Sphyraenidae [138], Sphyrnidae [50,154], Squillidae [17,137], Suidae [147], Sulidae [140], Stromateidae [137,144,156], Syngnathidae [145], Tachyglossidae [17], Tetraodontidae [17,27,60,138,140,145], Trichiuridae [156], Triglidae [50], Turdidae [150], Volutidae [145]			
Galeorhinus galeus	66	Agonidae [163], Argentinidae [101], Arhynchobatidae [158], Atherinidae [158], Batrachoididae [158-159], Belonidae [159], Bramidae [101], Callionymidae [163], Callorhinchidae [158], Caproidae [161], Carangidae [81,101,158,161,164], Centriscidae [161], Chimaeridae [159], Clinidae [159], Clupeidae [81,158-160,162-163], Congridae [158], Cottidae [159,162], Cynoglossidae [158], Eledonidae [163], Embiotocidae [159], Engraulidae [81,158-160,162], Exocoetidae [159], Gadidae [163], Gempylidae [160,164], Gonatidae [162], Kyphosidae [159], Loliginidae [158,163,165], Macrouridae [101,161], Merlucciidae [81,101,160,162], Moridae [101], Mugilidae [158], Mullidae [158], Myliobatidae [158], Nephropidae [101], Nototheniidae [159], Octopodidae [158,161], Ommastrephidae [101,158,160,165], Onychoteuthidae [101], Ophidiidae [158], Palinuridae [164], Pandalidae [101], Paralichthyidae [158,160], Percophidae [81,158], Phycidae [161], Pinguipedidae [158], Pleuronectidae [162-163], Pomacentridae [159], Pomatomidae [81,158], Porpitidae [162], Rajidae [158], Salmonidae [159], Salpidae [101], Sciaenidae [81,158-159], Scombridae [159,161,163], Sebastidae [101], Sepiolidae [163], Serranidae [158], Sparidae [159,161], Sphyraenidae [159], Squatinidae [81], Sternoptychidae [161], Stromateidae [158], Synodontidae [161], Triakidae [81,158], Trichiuridae [81,161], Triglidae [81,163]	34	12	1280 (1976)
Galeus melastomus	110	Acanthephyridae [105,117,166], Alepocephalidae [117,167], Alpheidae [105,129-130,167], Aphroditidae [117], Argentinidae [169], Argonautidae [166,171], Aristeidae [130,166], Astrorhizidae [169], Axiidae [105,129-130,169],	49	14	2983 (3431)

Bathypolypodidae [105,117], Benthesicymidae [129-130,166], Bothidae [166], Brachioteuthidae [130,166,171], Bythitidae [166], Carangidae [117,167], Chiroteuthidae [105,166], Chlorophthalmidae [166], Chtenopterygidae [166], Cirolanidae [105,117], Cliidae [166], Cranchiidae [105], Crangonidae [117,129,169,359], Cymbuliidae [117], Cynoglossidae [166], Diphyidae [105], Eledonidae [172], Enoploteuthidae [130,166,171], Epimeriidae [117], Eucopiidae [105], Euphausiidae [105,117,129-130,169], Eusiridae [169], Gadidae [117,130,166-167,169,359], Gammaridae [169], Geryonidae [105,117,130], Gnathophausiidae [117], Gobiidae [166,169], Goneplacidae [167], Gonostomatidae [130], Hauerinidae [105], Histioteuthidae [105,129,166,170-171], Hyperiidae [105,117], Ipnopidae [166], Leuconidae [105], Loliginidae [166], Lophogastridae [129-130,169], Lotidae [105,166,169], Lysianassidae [169], Macrouridae [105,117,129,166,359], Merlucciidae [166-167], Moridae [105,117,359], Munididae [102,117,166-167,169], Myctophidae [102,105,117,129-130,166-167,170], Mysidae [105,169], Nassariidae [166], Nebaliidae [169], Nemichthyidae [129], Octopodidae [166,172], Octopoteuthidae [166], Ommastrephidae [105,117,129-130,166,168], Onuphidae [169], Onychoteuthidae [129-130,166], Opisthoteuthidae [172], Oplophoridae [129,166], Paguridae [105,117,129-130,167], Pandalidae [129,166,169,359], Paralepididae [105,166], Parapaguridae [117], Parasquillidae [166], Pardaliscidae [105], Pasiphaeidae [105,117,129-130,166-167,170,359], Penaeidae [130], Phasianidae [105], Phosichthyidae [166], Phronimidae [105,129], Phrosinidae [105], Phycidae [117], Platyscelidae [105], Pleuronectidae [129], Polybiidae [117,130,167], Polychelidae [117,129], Poromyidae [166], Processidae [105,129-130,166-167], Pyrosomatidae [129,166], Pyroteuthidae [166,359], Rajidae [130], Rhabdamminidae [105,169], Rissoidae [105], Saccamminidae [169], Salpidae [105,117,129], Scinidae [105], Scomberesocidae [167], Scombridae [167], Scorpaenidae [166], Scyllaridae [166], Sebastidae [166], Sepiidae [172], Sepiolidae [105,129-130,166,168-169,171-172,359], Sergestidae

		[105,117,129-130,166-167,170], Serpulidae [105], Sigalionidae [169], Solenoceridae [167,359], Sternoptychidae [117,129,167,169], Stomiidae [105,129-130,166], Synaphobranchidae [117], Terebratulidae [105], Trachichthyidae [166], Tubulariidae [166], Uristidae [117,169], Uvigerinidae [105], Xanthidae [105]			
Ginglymostoma cirratum	18	Batrachoididae [173], Belonidae [173], Carangidae [173], Epialtidae [173], Haemulidae [173], Labridae [173], Loliginidae [173], Lutjanidae [173], Majidae [173], Octopodidae [173], Ophichthidae [173], Palinuridae [173], Rachycentridae [173], Rajidae [173], Scaridae [173], Scombridae [173], Sparidae [173], Syngnathidae [173]	80	1	41 (91)
Hemipristis elongata	5	Congridae [17], Dasyatidae [17], Diodontidae [17], Lutjanidae [17], Muraenidae [17]	2	2	86 (114)
Hemiscyllium ocellatum	10	Alpheidae [174], Amphinomidae [174], Callionymidae [174], Gammaridae [174], Gonodactylidae [174], Lysiosquillidae [174], Ocypodidae [174], Portunidae [174], Terebellidae [174], Xanthidae [174]	15	1	51 (53)
Hemitriakis japanica	13	Alpheidae [175], Bothidae [175], Callianassidae [175], Engraulidae [175], Gobiidae [175], Hippolytidae [175], Mysidae [175], Ogyrididae [175], Palaemonidae [175], Pasiphaeidae [175], Penaeidae [175], Sciaenidae [175], Upogebiidae [175]	3	2	51 (57)
Heptranchias perlo	18	Acropomatidae [176], Centrolophidae [176], Enoploteuthidae [176], Gempylidae [176], Macrouridae [176], Merlucciidae [176], Myctophidae [176], Narcinidae [176], Octopodidae [176], Ommastrephidae [176-177], Ophidiidae [176], Oplophoridae [176], Paraulopidae [176], Scombridae [177], Serranidae [176], Solenoceridae [176], Trichiuridae [176], Triglidae [176]	9	2	89 (117)
Heterodontus francisci	8	Aegidae [178-179], Calyptraeidae [178-179], Fissurellidae [178], Octopodidae [178-179], Penaeidae [178-179], Portunidae [178-179], Sipunculidae [178-179], Syngnathidae [178-179]	30	2	193 (219)
Hexanchus griseus	28	Callorhinchidae [180], Carangidae [181], Chimaeridae [184], Clupeidae [180-181], Delphinidae [180], Dussumieriidae [180], Echinorhinidae [184], Engraulidae	33	8	90 (162)

		[180,182], Gempylidae [180], Inachoididae [183], Loliginidae [180-181], Lophiidae [183], Macrouridae [180,183], Merlucciidae [180-182,184], Myctophidae [180], Myxinidae [184], Ommastrephidae [180], Otariidae [180], Petromyzontidae [184], Pomatomidae [182], Phycidae [183], Rajidae [180], Scombridae [180-182], Scyliorhinidae [180], Sebastidae [180], Squalidae [180-184], Triakidae [180-181], Xiphiidae [181]			
Iago omanensis	12	Cranchiidae [185], Eledonidae [185], Enoploteuthidae [185], Histioteuthidae [185], Loliginidae [185], Majidae [185], Myctophidae [185-186], Ommastrephidae [185], Portunidae [185], Sepiolidae [185], Solenoceridae [186], Squillidae [185]	11	2	256 (279)
Isurus oxyrinchus	89	Alepisauridae [190,193,195,197,202], Ammodytidae [194], Amphitretidae [10], Ancistrocheiridae [1,201], Anguillidae [202], Argentinidae [73], Argonautidae [10], Atelecyclidae [197], Axiidae [197], Balistidae [190,197], Belonidae [1,191,197,201], Berycidae [27], Bramidae [10,190,192-193,195,199], Carangidae [1,9-10,27,73,191,193,195,197,199-201], Carcharhinidae [10,191,194,201-202], Centrolophidae [27], Cheloniidae [190], Clupeidae [10,27,191,194,198,202], Coryphaenidae [1], Dalatiidae [195], Dasyatidae [191], Delphinidae [9-10,189,193,196], Diodontidae [27,195], Diomedeidae [193], Engraulidae [10,73], Enoploteuthidae [10], Ephippidae [191], Gadidae [202], Gempylidae [3,9,27,191,195,199], Gnathophausiidae [192], Gonatidae [10,202], Haemulidae [1,191,201], Histioteuthidae [1,10,165,193,197,199,201-202], Istiophoridae [191,195,201], Kyphosidae [10], Lamnidae [191,193], Loliginidae [10,191,194,197,201-202], Lycoteuthidae [165,199,201], Macrouridae [10], Malacanthidae [194], Mastigoteuthidae [1], Merlucciidae [191,194,202], Monacanthidae [1,9], Mugilidae [10,27,73], Myliobatidae [1,191,201-202], Nomeidae [1,193,195], Octopoteuthidae [1,10,201], Odontaspididae [191], Ommastrephidae [1,9-10,73,165,192-194,197,199,201-203], Onychoteuthidae [1,10,202], Ophidiidae [10,191], Opisthoteuthidae [1], Oplegnathidae [191], Ostraciidae [201], Otariidae [188], Paralepididae [10], Paralichthyidae [10],	106	23	2177 (3776)

		Penaeidae [27], Phycidae [197], Pomatomidae [194,202], Priacanthidae [1], Pseudocarchariidae [193], Rajidae [197,202], Regalecidae [1,27], Sciaenidae [191,201], Scomberesocidae [10,190,197-198,202], Scombridae [1,3,9-10,27,73,190-195,197,199-202], Scorpaenidae [202], Scyliorhinidae [191,201], Sebastidae [10,202], Sepiidae [187,201], Serranidae [194], Solenoceridae [193-194], Sparidae [27,191,194,197,202], Sphyrnidae [191,201], Spirulidae [197], Squalidae [191,194,201], Squillidae [197], Stromateidae [202], Syngnathidae [202], Tetragonuridae [195], Tetraodontidae [1,27], Thysanoteuthidae [1], Trachipteridae [199], Trichiuridae [3,27,73,199], Triglidae [194,202], Xiphiidae [193-195,197,202], Zeidae [191], Zoarcidae [194,202]			
Lamna nasus	44	Alepisauridae [204], Ammodytidae [204], Anarhichadidae [204], Anguillidae [204], Anotopteridae [204], Argentinidae [209], Belonidae [209], Brachioteuthidae [121], Bramidae [200,206], Carangidae [200], Centrolophidae [200], Channichthyidae [121], Chiroteuthidae [121], Clupeidae [204,208-209,211], Congiopodidae [121], Cottidae [204], Cranchiidae [121], Cyclopteridae [204], Eledonidae [208], Euphausiidae [207], Gadidae [204,209], Gempylidae [121], Gonatidae [121], Hemitripteridae [204], Histioteuthidae [121], Loliginidae [211], Mastigoteuthidae [121], Merlucciidae [204,206], Myctophidae [121,204], Nemichthyidae [204], Neoteuthidae [121], Ommastrephidae [121,200,204], Onychoteuthidae [121,205,210], Oregoniidae [204], Paralepididae [200], Petromyzontidae [204], Phosichthyidae [206], Pleuronectidae [209], Scomberesocidae [204], Scombridae [204,208,211], Sebastidae [204,211], Squalidae [204], Stomiidae [121], Trachipteridae [206]	30	10	1928 (4891)
Loxodon macrorhinus	46	Alpheidae [17], Ammodytidae [17], Apogonidae [17], Bothidae [17], Bregmacerotidae [17], Caesionidae [17], Calappidae [212], Callianassidae [17], Callionymidae [17,212], Carangidae [17], Champsodontidae [17], Congridae [17], Creediidae [212], Engraulidae [213], Euphausiidae [17], Gobiidae [17], Labridae [17,212], Leiognathidae [17], Loliginidae [212-213], Mugilidae [212], Mullidae [17], Mussidae [213], Nannosquillidae [212], Ocypodidae [213], Ophichthidae	4	3	345 (449)

		[212], Palinuridae [213], Pegasidae [17,212], Penaeidae [17,212-213], Phascolosomatidae [213], Platycephalidae [17], Portunidae [212-213], Scaridae [17], Scombridae [212], Scorpaenidae [17], Sepiidae [213], Sepiolidae [17,212], Siganidae [212], Sillaginidae [212], Sparidae [212], Squillidae [17,212], Syngnathidae [17], Synodontidae [212], Tetraodontidae [17,212], Triglidae [17,212], Upogebiidae [17], Uranoscopidae [17]			
Megachasma pelagios	1	Euphausiidae [214-216]	19	3	3(3)
Mustelus asterias	15	Agonidae [163], Atelecyclidae [163], Axiidae [163], Cancridae [163], Corystidae [163], Donacidae [163], Galatheidae [207], Majidae [163], Oregoniidae [163], Paguridae [163], Pandalidae [163], Pilumnidae [163], Polybiidae [163,207], Upogebiidae [163], Xanthidae [163]	8	2	48 (49)
Mustelus californicus	8	Batrachoididae [217], Blepharipodidae [217], Callianassidae [217], Cancridae [217], Clupeidae [217], Crangonidae [217], Urechidae [217], Varunidae [217]	5	1	49 (52)
Mustelus canis	55	Achiridae [66], Ammodytidae [66,218], Anguillidae [218], Ariidae [222], Atherinopsidae [224], Busyconidae [66], Calappidae [223], Cancridae [66,218-220,224], Carcinidae [224], Clupeidae [218], Congridae [223], Crangonidae [66,220,224], Diodontidae [223], Diogenidae [223], Engraulidae [220,222], Epialtidae [66,219-220,224], Fundulidae [224], Gadidae [66], Gammaridae [220], Geryonidae [223], Glyceridae [220], Gonodactylidae [223], Limulidae [66,219], Loliginidae [66,165,218,220,223], Majidae [218,223], Merlucciidae [218], Munididae [223], Mysidae [220], Mytilidae [219-220], Naticidae [218,220], Nephropidae [218-219], Octopodidae [223], Ommastrephidae [218], Ophidiidae [66], Ovalipidae [66,219,224], Paguridae [66,218-220,224], Palaemonidae [224], Panopeidae [218-219,224], Paralichthyidae [66,218,220], Parasquillidae [223], Penaeidae [220], Pharidae [220,224], Portunidae [66,223-224], Priacanthidae [223], Rajidae [218], Sciaenidae [220], Sepiolidae [165], Solenidae [66], Sparidae [66,218], Squalidae [218], Squillidae [66,219-220,222-223], Stromateidae [218], Triglidae [218], Upogebiidae [219-220,224], Xanthidae [223]	45	8	453 (514)

Mustelus fasciatus	9	Aethridae [225], Batrachoididae [225], Diogenidae [225], Epialtidae [225], Leucosiidae [225], Olividae [225], Paralichthyidae [225], Penaeidae [225], Portunidae [225]	1	1	14 (17)
Mustelus griseus	35	Alpheidae [175], Bothidae [175], Callianassidae [175], Callionymidae [175], Cancridae [175], Carangidae [175], Crangonidae [175], Diogenidae [175], Dorippidae [175], Epialtidae [175], Euryplacidae [175], Galatheidae [175], Galenidae [175], Hexapodidae [175], Hippolytidae [175], Holognathidae [175], Leucosiidae [175], Lysmatidae [175], Menippidae [175], Mysidae [175], Ogyrididae [175], Ommastrephidae [175], Palaemonidae [175], Parthenopidae [175], Pasiphaeidae [175], Penaeidae [175], Pinnotheridae [175], Polybiidae [175], Portunidae [175], Sepiidae [175], Sepiolidae [175], Squillidae [175], Upogebiidae [175], Urechidae [175], Varunidae [175]	3	1	181 (187)
Mustelus henlei	42	Blepharipodidae [217], Bothidae [217,227], Calappidae [226-227], Callianassidae [229-230], Cancridae [217,229-230], Chasmocarcinidae [228], Cottidae [229], Crangonidae [217,229-230], Diogenidae [226], Dromiidae [228], Embiotocidae [230], Engraulidae [217,229-230], Epialtidae [229], Ethusidae [227], Eurysquillidae [226], Galatheidae [226-227], Gobiidae [217,230], Grapsidae [230], Loliginidae [217,228], Lophiidae [227], Molgulidae [230], Munididae [226,228], Nereididae [230], Octopodidae [226], Ophidiidae [227], Paguridae [228], Pandalidae [227], Paralichthyidae [226-227,230], Parasquillidae [227], Penaeidae [227-228], Pinnotheridae [229], Portunidae [226-228], Scombridae [226], Scorpaenidae [226], Serranidae [226-227], Sicyoniidae [228], Solenoceridae [226-227], Squillidae [226-228], Syngnathidae [229], Upogebiidae [229-230], Uranoscopidae [226], Varunidae [217,229-230]	13	6	585 (768)
Mustelus lenticulatus	37	Aphroditidae [232], Arenicolidae [232], Axiidae [232], Cancridae [232], Crangonidae [232], Diogenidae [232], Eunicidae [232], Glyceridae [232], Goneplacidae [232], Goniadidae [232], Hiatellidae [232], Hymenosomatidae [232], Laomediidae [232], Macrophthalmidae [232], Majidae [232], Maldanidae [232], Mesodesmatidae [232], Olividae [232], Ommastrephidae [232], Ovalipidae	2	1	428 (428)

		[232], Paguridae [232], Palaemonidae [232], Palinuridae [232], Pinnotheridae [232], Priapulidae [232], Sabellariidae [232], Sigalionidae [232], Squillidae [232], Struthiolariidae [232], Terebellidae [232], Tetrasquillidae [232], Trochidae [232], Turritellidae [232], Upogebiidae [232], Urechidae [232], Varunidae [232], Volutidae [232]			
Mustelus lunulatus	37	Aethridae [233-234], Albulidae [233], Axiidae [228], Batrachoididae [235], Blepharipodidae [235], Buccinidae [234], Calappidae [228,235], Cancridae [233], Carangidae [233], Chasmocarcinidae [228], Congridae [235], Dromiidae [228], Dussumieriidae [233], Gobiidae [228], Hemiramphidae [233], Hemisquillidae [233], Hippidae [234], Kyphosidae [233], Leucosiidae [228], Loliginidae [228], Munididae [228,233], Octopodidae [233], Paguridae [228], Palaemonidae [234], Parthenopidae [228], Penaeidae [228,234], Portunidae [228,234], Pseudorhombilidae [228], Pseudosquillidae [234], Scombridae [233], Scorpaenidae [235] Sicyoniidae [228,233], Squillidae [228,234], Stromateidae [233], Synodontidae [233], Terebridae [234], Xanthidae [228]	7	4	309 (498)
Mustelus manazo	68	Aegidae [236], Alpheidae [175,236-237], Ammodytidae [175,236], Axiidae [236], Blepharipodidae [237], Calappidae [175,236], Callianassidae [175], Cancridae [73,175,236-237], Carangidae [236-237], Carditidae [236], Cheiragonidae [236], Cirolanidae [175], Clupeidae [236-237], Cottidae [236], Crangonidae [175,236-237], Diogenidae [175,236-237], Dorippidae [175,236-237], Engraulidae [73,236], Epialtidae [175], Euphausiidae [236], Euryplacidae [175,236], Galatheidae [175,236], Galenidae [175], Gobiidae [236], Goneplacidae [73,175,236], Hexapodidae [175], Hippolytidae [175], Holognathidae [175], Inachidae [236], Leucosiidae [175,236-237], Lysmatidae [175], Majidae [236], Menippidae [175], Monacanthidae [237], Moridae [237], Munididae [73], Mysidae [175], Nephropidae [236], Ogyrididae [175], Ommastrephidae [73], Ophiolepididae [236], Oregoniidae [236], Ovalipidae [237], Paguridae [236-237], Palaemonidae [175], Pandalidae [236], Parthenopidae [175,236], Pasiphaeidae [175], Penaeidae [175,236-237], Pinnotheridae [236], Polybiidae [73,175,236],	12	4	1539 (1602)

		Porcellanidae [175], Portunidae [175,236-237], Raninidae [236-237], Scomberesocidae [73,236], Scombridae [237], Scyllaridae [175], Semelidae [236], Sicyoniidae [237], Solenoceridae [73], Squillidae [175,236-237], Synodontidae [236], Trichiuridae [73], Trichopeltariidae [236], Upogebiidae [175,236-237], Urechidae [175], Varunidae [175], Xanthidae [175,236-237]			
Mustelus mustelus	88	Alpheidae [238-239], Aphroditidae [242], Atelecyclidae [238,242], Belonidae [239], Bothidae [238], Calappidae [238-240], Callianassidae [240], Carangidae [239-240,242], Carcinidae [241], Centracanthidae [239-240,242], Cerithiidae [238], Cheilodactylidae [240], Clupeidae [238-240,242], Congridae [238-239], Congiopodidae [240], Corystidae [238], Crangonidae [238-239,242], Dairoididae [240], Dentaliidae [238], Diodontidae [240], Diogenidae [238], Donacidae [238], Dorippidae [238-239], Dromiidae [239], Dussumieriidae [240], Eledonidae [239], Engraulidae [238,240-242], Enteroctopodidae [240], Eriphiidae [239], Ethusidae [238-239], Euryplacidae [239], Gadidae [241-242], Geryonidae [239], Gobiidae [239], Goneplacidae [238-240,242], Haemulidae [240], Inachidae [238], Labridae [239], Leucosiidae [238,240], Loliginidae [168,238-240,242], Lophogastridae [238], Mactridae [238], Majidae [238-239], Merlucciidae [240,242], Mugilidae [238-240], Mullidae [239], Munididae [242], Mytilidae [240], Nannosquillidae [238], Nephropidae [242], Nuculidae [238], Octopodidae [239-240], Ommastrephidae [168,242], Ophichthidae [238], Ophiotrichidae [240], Ovalipidae [240], Paguridae [238-239], Palaemonidae [239], Palinuridae [240], Parasquillidae [238], Parthenopidae [238-239], Pectinidae [238], Penaeidae [238-240], Pilumnidae [241], Plagusiidae [240], Polybiidae [238-239,241-242], Pomatomidae [240], Portunidae [238-241], Processidae [238], Sciaenidae [240], Scombridae [238-239], Scorpaenidae [239], Scyllaridae [238,240], Sebastidae [240], Sepiidae [238-241], Sepiolidae [242], Serranidae [239], Sicyoniidae [238-239], Soleidae [239], Sparidae [238-240,242], Squillidae [238-242], Syngnathidae [239-240], Tellinidae [238], Thiidae [240], Turritellidae [238], Upogebiidae [238-241], Varunidae [238,240], Xanthidae [239,242]	22	8	1271 (1444)

Mustelus schmitti	80	Alpheidae [247], Ampeliscidae [243], Ampharetidae [243], Atherinidae [247], Balanidae [243], Batrachoididae [243], Belliidae [243,245], Blepharipodidae [243,246], Branchiostomatidae [243], Calappidae [243], Capitellidae [243], Caprellidae [245], Carangidae [243], Cirolanidae [243,245], Clupeidae [247], Columbellidae [243], Cynoglossidae [243], Diogenidae [243,245], Echiuridae [243], Engraulidae [243,246], Epialtidae [243,245], Eunicidae [243], Flabelligeridae [243], Gammaridae [243,245], Glyceridae [243-244,246], Goniadidae [243], Idoteidae [246], Inachoididae [243], Lithodidae [243], Loliginidae [243-245,247], Lumbrineridae [243], Mactridae [243], Majidae [243,247], Maldanidae [243], Marginellidae [243], Merlucciidae [243], Munididae [243], Muricidae [244], Mytilidae [243], Myxinidae [243], Nassariidae [243], Naticidae [243], Nephtyidae [243], Nereididae [243], Nototheniidae [246], Octopodidae [243,245-246], Ommastrephidae [243], Onuphidae [243,246], Opheliidae [243], Ophidiidae [243], Paguridae [243-244], Panopeidae [243], Paralichthyidae [243], Pectinariidae [243], Penaeidae [243,245-246], Phyllodocidae [243], Pilumnidae [243], Pilumnoididae [243,245], Pinnotheridae [243], Platyxanthidae [243,245], Polybiidae [243], Porcellanidae [243,246], Portunidae [243], Sabellidae [243], Sergestidae [243,245], Serolidae [243,245,247], Serranidae [243], Sesarmidae [243], Sipunculidae [243,245], Solenidae [243,245], Solenoceridae [243], Spionidae [243], Squillidae [243], Tetrasquillidae [243,245-246], Travisiidae [243], Trichopeltariidae [243,247], Varunidae [245-247], Veneridae [243,245], Volutidae [243], Zoarcidae [247]	12	5	1056 (1126)
Negaprion acutidens	18	Ariidae [38], Atherinidae [248], Balistidae [60], Belonidae [38,248], Carangidae [38], Clupeidae [38,248], Drepaneidae [38], Gobiidae [38], Labridae [248], Mugilidae [38], Penaeidae [38], Portunidae [248], Rhinobatidae [248], Scaridae [60], Scombridae [38], Sillaginidae [248], Sparidae [248], Syngnathidae [248]	22	4	68 (98)
Negaprion brevirostris	46	Albulidae [249], Alpheidae [249,253], Arenicolidae [249], Ariidae [250], Atherinidae [249-250], Atherinopsidae [251], Balistidae [249], Batrachoididae [249-251], Belonidae [249-250], Blenniidae [250], Bothidae [249], Carangidae	164	5	576 (855)

		[249-250,253], Centropomidae [250], Clupeidae [253], Cyprinodontidae [249-251], Dasyatidae [249], Elopidae [250], Engraulidae [251], Ephippidae [250], Fundulidae [253], Gerreidae [249-251,253], Gobiidae [249-250], Gonodactylidae [249], Haemulidae [249-251], Hemiramphidae [249-250], Labridae [249], Loliginidae [249], Lutjanidae [249-250], Monacanthidae [250], Mugilidae [250,252], Octopodidae [250], Ocypodidae [249], Ophichthidae [249-250,253], Ostraciidae [250], Palinuridae [249], Penaeidae [249-251], Pomacentridae [249], Portunidae [249-250,253], Pseudosquillidae [249], Scaridae [249-250], Soleidae [249-250], Sparidae [249-251], Sphyraenidae [249-250], Synodontidae [249], Tetraodontidae [250], Xanthidae [249]			
Notorynchus cepedianus	82	Acipenseridae [259], Aegidae [176], Anguillidae [254], Aplodactylidae [176], Arhynchobatidae [256], Ariidae [258], Arripidae [176,254], Atherinidae [255-257,259], Balaenopteridae [254], Batrachoididae [81], Callorhinchidae [176,254,257-258], Cancridae [259], Carangidae [176,254,256-258], Carcharhinidae [258], Centriscidae [176], Centrolophidae [176,254,257], Cheilodactylidae [176], Clupeidae [81,254,258], Congridae [254,258], Cottidae [259], Dasyatidae [254,258], Delphinidae [254,258], Embiotocidae [259], Emmelichthyidae [254], Engraulidae [254], Enteroctopodidae [259], Gempylidae [176,254], Geotriidae [254], Gobiidae [176], Haemulidae [258], Hexanchidae [176,254,258-259], Labridae [254], Latridae [254], Loliginidae [81,176,258], Macropodidae [254], Merlucciidae [257-258], Moridae [254], Mugilidae [254,258], Myliobatidae [176,254,256,258-259], Mytilidae [258], Myxinidae [257-258], Narcinidae [254], Narkidae [258], Nassariidae [256,258], Octopodidae [258], Odacidae [254], Ommastrephidae [176,254,257], Ophidiidae [257-258], Otariidae [176,255-258], Paguridae [256], Palinuridae [176], Paralichthyidae [257], Parascylliidae [176], Pentacerotidae [176], Percophidae [81], Petromyzontidae [259], Phocidae [257,259], Pinguipedidae [257], Plagusiidae [258], Platycephalidae [254], Polyprionidae [257], Pontoporiidae [256], Pristiophoridae [176,254], Rajidae [176,254,256-259], Rhinobatidae [258],	38	8	855 (1371)

		Salmonidae [254,259], Sciaenidae [81,256,258], Scombridae [254,257-258], Scyliorhinidae [176,254,258], Sebastidae [259], Serranidae [176,254,257], Sertulariidae [256], Sillaginidae [176], Sparidae [176,258], Squalidae [176,254,257-259], Squatinidae [81,176,256], Stromateidae [257], Torpedinidae [258], Triakidae [81,176,254,256-259], Trichiuridae [258], Triglidae [176,254], Urolophidae [176,254]			
Prionace glauca	133	Alepisauridae [58,192,200,261,264,268,274], Alloposidae [2,10,165,199,260-262,266,268,271-272], Amphitretidae [2,10,260-262,268,272], Ancistrocheiridae [1-2,165,199,260,262,266,268,272], Anoplopomatidae [275], Anotopteridae [280], Architeuthidae [165,199,260-261], Argonautidae [1-2,10,165,192,199,260,262,270,272,275], Ariommatidae [261], Balistidae [265], Bathylagidae [260], Batrachoididae [260,275], Belonidae [260,278], Brachioteuthidae [271], Brachyscelidae [192], Bramidae [192,199,261,264,268,274], Buccinidae [278], Cancridae [260], Caproidae [272], Carangidae [1-2,10,27,192,199,200,260-262,264,277-278], Carcharhinidae [262,264], Centriscidae [272], Chiroteuthidae [165,199,260-261,265-266,268,272,277], Chtenopterygidae [272], Cionidae [278], Cinclidae [262], Cirolanidae [199], Cirroteuthidae [264,268], Clupeidae [10,27,58,162,192,260,269,271,275,278], Congridae [199], Coryphaenidae [1], Cottidae [260], Cranchiidae [2,260-262,266,268,272,277,279], Cyclopteridae [269,276], Cycloteuthidae [268,272], Dasyatidae [261], Delphinidae [192,260,264], Dermochelyidae [264], Diodontidae [27,192,199,261,265], Diretmidae [272], Echeneidae [2,58,262], Embiotocidae [281], Engraulidae [1,162,260,262,267,270,275,277,281], Enoploteuthidae [10,260-261,268], Euphausiidae [10,266,275], Exocoetidae [268,277], Gadidae [269,271,278], Galatheidae [278], Gempylidae [192,199,261,264,274], Gnathophausiidae [260,274], Gonatidae [1-2,10,260-262,266,268,271-272,274-275,279], Halosauridae [271], Hexagrammidae [260], Histioteuthidae [1-2,10,165,192,199,260-262,266,268,271-273,275,277,279], Hyperiidae [266,271],	167	34	2921 (4259)

		Idoteidae [260], Istiophoridae [261], Joubiniteuthidae [272], Lamnidae [261,264], Lampridae [10,261], Loliginidae [10,260,275,277], Lophiidae [269], Lophogastridae [262], Luvaridae [10], Lycoteuthidae [165,199], Lysianassidae [277], Macrouridae [272], Mastigoteuthidae [1-2,260,272,277], Merlucciidae [1-2,10,162,260,262,269,275,277], Mimidae [263], Molidae [10,260], Monacanthidae [199,261,265], Moronidae [278], Mugilidae [10,260,278], Munididae [2,10,260,262], Muraenidae [272], Myctophidae [58,260-261,268,272,274-275,280], Neoteuthidae [260], Nomeidae [260,264], Octopodidae [192,260-261,275,277], Octopoteuthidae [1,10,165,199,260-261,268,271-272,275,279], Ocythoidae [200,260-261,266,272,274], Ommastrephidae [1-2,10 ,165,192,199,260-262,264-265,267,271,274-275,277,279], Onuphidae [278], Onychoteuthidae [1-2,10,162,165,199,260-262,266,268,272,274-275,277], Ophidiidae [1,275], Opisthoteuthidae [1,10], Ostraciidae [58], Otariidae [265], Pandalidae [275,277], Paralichthyidae [162,275], Pentacerotidae [274], Petromyzontidae [275], Pholidoteuthidae [1-2,261-262], Platyscelidae [199,261], Pleuronectidae [162,269,271,275,278], Polybiidae [278], Pomacentridae [277], Procellariidae [261,271], Psammobiidae [278], Rajidae [269], Regalecidae [27], Renillidae [277], Salmonidae [268-269], Salpidae [261], Sciaenidae [275,281], Scomberesocidae [10,260,269,271,280], Scombridae [1-2,10,27,192,200,260-262,264,268-269,271,276,278], Sebastidae [10,260,269,275,277], Sepiidae [278-279], Sepiolidae [279], Solenoceridae [264], Sphyraenidae [272], Squalidae [10,269,275,277], Squillidae [2,262], Sternidae [10], Stauroteuthidae [268], Sternoptychidae [260,268,274], Stomiidae [260], Sulidae [58], Syngnathidae [271,275-277], Tetragonuridae [192], Tetraodontidae [1,264], Thysanoteuthidae [261-262,277], Trachichthyidae [192], Trachipteridae [2,261-262], Tremoctopodidae [165,199,261], Triakidae [10,270], Trichiuridae [192,199,265,271-272], Triglidae [278], Vampyroteuthidae [1-2,10,260-262,264,266,268,272,275,277], Veneridae [278], Xiphiidae [261,264]			
Rhizoprionodon	61	Alpheidae [38], Apogonidae [38], Ariidae [17], Aristeidae [282], Atherinidae	20	8	1223 (4656)

acutus		[38,248], Batrachoididae [17], Bothidae [20], Bregmacerotidae [17], Callionymidae [20], Carangidae [17,38,213,282], Centropomidae [248], Clupeidae [17,20,38,40,213,248,282], Congridae [17], Cynoglossidae [282], Dactylopteridae [17], Draconettidae [20], Elapidae [20], Elopidae [282], Engraulidae [20,38,40,213,282], Gerreidae [36,38,213,282], Gobiidae [38], Haemulidae [20,36,282], Hemiramphidae [36,38], Labridae [248,282], Leiognathidae [17,20,36,38,40], Lethrinidae [213], Loliginidae [17,20], Lutjanidae [213], Monacanthidae [17,36], Monocentridae [17], Moronidae [282], Mugilidae [20,36,213,282], Mullidae [17,20,36,282], Muraenesocidae [282], Muraenidae [17,282], Nassariidae [282], Nemipteridae [17], Octopodidae [282], Ommastrephidae [282], Penaeidae [17,20,36,38,40,248], Pinguipedidae [17], Platycephalidae [17,38], Polybiidae [282], Polynemidae [282], Pomatomidae [282], Portunidae [248,282], Pristigasteridae [17], Ranellidae [282], Sciaenidae [38,282], Scombridae [17,20], Scorpaenidae [17], Sepiidae [213,282], Sillaginidae [17,38,248], Soleidae [20,282], Sparidae [282], Sphyraenidae [282], Strombidae [282], Syngnathidae [17], Synodontidae [17,20], Terapontidae [38,248], Tetraodontidae [17]			
Rhizoprionodon terraenovae	53	Aethridae [51,283], Alpheidae [284], Ariidae [25,33,51,284], Balistidae [283], Bothidae [33,283], Calappidae [24], Cancridae [66], Carangidae [24-25,33,51,284], Cerithiidae [25], Cheloniidae [283], Clupeidae [25,33,51,66,283-284], Congridae [283], Cynoglossidae [25,66,284], Dasyatidae [33,284], Elopidae [24,33], Engraulidae [24-25,33,51,66,283-284], Epialtidae [66], Gerreidae [284], Gobiidae [51], Gonodactylidae [24], Haemulidae [24,66,283-284], Hippidae [284], Loliginidae [24-25,33,66,284], Lutjanidae [25,51], Lysiosquillidae [283], Mugilidae [51], Muraenidae [24,283], Nassariidae [66], Naticidae [284], Octopodidae [24], Ophichthidae [25,33,51,284], Ophidiidae [66], Ovalipidae [66], Paguridae [66,283], Paralichthyidae [24,51,66,284], Penaeidae [25,33,51,66,283-284], Portunidae [25,33,51,284], Priacanthidae [66], Rajidae [283], Sciaenidae [25,33,51,66,283-284], Scombridae [24,33,51,283], Scophthalmidae [66],	35	8	1306 (2287)

		Serranidae [51,66], Sicyoniidae [283], Sparidae [51,66,284], Sphyraenidae [283], Squillidae [25,33,51,283], Syngnathidae [33,284], Synodontidae [51,283-284], Terebellidae [284], Triacanthodidae [283], Trichiuridae [33,51], Triglidae [33,66]			
Rhizoprionodon taylori	24	Apogonidae [286], Chirocentridae [286], Clupeidae [17,20,286], Cynoglossidae [286], Engraulidae [20,285-286], Haemulidae [286], Hemiramphidae [286], Leiognathidae [17,20,286], Leptobramidae [286], Loliginidae [286], Monacanthidae [17], Mullidae [20,286], Penaeidae [17,286], Plotosidae [286], Polynemidae [20,286], Portunidae [286], Pristigasteridae [17], Scombridae [17,20], Sillaginidae [286], Syngnathidae [286], Synodontidae [286], Terapontidae [286], Triacanthidae [286], Trichiuridae [286]	8	4	238 (536)
Scoliodon laticaudus	32	Ambassidae [287], Bregmacerotidae [287], Carangidae [287], Clupeidae [287-288,290-291], Cynoglossidae [288], Cypridinidae [290], Engraulidae [287-288,290-291], Eucalanidae [290], Glyceridae [290], Gobiidae [287-288], Hyperiidae [290], Leiognathidae [287], Loliginidae [287-288], Mugilidae [287,290], Muraenesocidae [290], Nereididae [290], Ophichthidae [290], Palaemonidae [290], Pasiphaeidae [290], Penaeidae [287-288,290-291], Sciaenidae [287-288,290-291], Scyllaridae [291], Sepiidae [288], Sergestidae [287-288], Solenoceridae [288,290], Sphyraenidae [290], Squillidae [288,290-291], Synodontidae [288,290], Temoridae [290], Trachipteridae [291], Trichiuridae [287-288,290], Triglidae [290]	10	4	964 (1867)
Scyliorhinus canicula	135	Aegidae [300], Alepocephalidae [167], Alpheidae [129,167,294-297,301], Agonidae [163,292], Ammodytidae [163,292], Ampeliscidae [294,298], Aphroditidae [129,163,292,298,301], Arenicolidae [163,302], Argentinidae [129,163,300], Atelecyclidae [129,163,292,294], Axiidae [163], Belonidae [296], Bonelliidae [301], Bothidae [295,301], Branchiostomatidae [292], Buccinidae [163,292,298], Callianassidae [163], Callionymidae [129,163,292,294,296,298], Cancridae [163], Caproidae [129,297], Carangidae [129,163,167,295-297 ,301], Cardiidae [292], Centracanthidae [129], Centriscidae [301], Cepolidae [129,295-297], Cerithiidae [302], Chaetopteridae [300], Chlorophthalmidae [300],	164	14	14579 (16876)

Cirolanidae [294,300], Clupeidae [163,292,294,300-301], Corystidae [163,302], Cottidae [292], Crangonidae [129,163,292,298], Cucumariidae [294,298], Cynoglossidae [129], Diogenidae [129,294,301], Echinasteridae [292], Eledonidae [163], Engraulidae [297,300-301], Eriphiidae [301], Eunicidae [292], Euphausiidae [129,294-296], Euphrosinidae [300], Gadidae [129,163,167,292,294-297,300,302], Galatheidae [129,163,292,295,300-302], Gammaridae [292], Glyceridae [292 ,301], Glycymerididae [292], Gobiesocidae [292], Gobiidae [129,163,292,295,300], Golfingiidae [292], Goneplacidae [129,163,167,294.297,300], Gonostomatidae [300], Haliotidae [294], Inachidae [163,301], Laomediidae [163,294], Leucosiidae [129,292,294], Liparidae [163], Loliginidae [163,294,300], Lophogastridae [129,294-296], Lumbrineridae [292], Macrouridae [300], Mactridae [292], Majidae [163], Merlucciidae [167,295,297,300-302], Mullidae [301], Munididae [129,163,167,295-297], Myctophidae [167,300], Myidae [163,292], Mysidae [295], Mytilidae [163], Naticidae [300], Nephropidae [163,295,301-302], Nephtyidae [292,298], Nereidae [163], Nereididae [292,301-302], Octopodidae [294,297,301], Oenonidae [292], Ommastrephidae [168,295,300-301], Opheliidae [292], Ophichthidae [129,301], Ophidiidae [163], Ophiuridae [292], Onuphidae [294], Oregoniidae [163,292,298], Paguridae [129,163,167,292,294,296-298,301-302], Palaemonidae [302], Palinuridae [129], Pandalidae [129,163,292,296-298,300], Parechinidae [301], Parthenopidae [129], Pasiphaeidae [129,163,167,294,297], Pectinidae [292], Penaeidae [300-301], Peristediidae [300], Pharidae [163,292], Pholidae [163,292], Phronimidae [129], Phyllophoridae [292,302], Pinnotheridae [292], Pleuronectidae [129,163,292,294], Polybiidae [129,163,167,292,294-298,300-302], Polynoidae [292], Porcellanidae [292], Portunidae [129,294,300], Processidae [129,167,295,297], Rajidae [294], Sabellidae [302], Salpidae [129], Scomberesocidae [167], Scombridae [163,167,294], Scophthalmidae [129], Scyliorhinidae [292,297], Scyllaridae [129,301], Sepiidae [168,294-295,300-301], Sepiolidae [129,163,292,295,300-301], Sergestidae [167], Sipunculidae [129,293-

		294,301], Soleidae [163,301], Solenoceridae [129,167,294-295,297], Sparidae [129,301], Squillidae [129,163,295], Sternaspidae [294-295], Sternoptychidae [167,295], Stichaeidae [163,292], Stomiidae [129], Synaphobranchidae [296], Syngnathidae [129,294], Thiidae [294], Thoridae [292], Trachinidae [163], Triglidae [163,301-302], Upogebiidae [163,292,294-295,298,301-302], Xanthidae [129,163,292]			
Scyliorhinus stellaris	32	Ammodytidae [303], Aphroditidae [303], Atelecyclidae [303], Buccinidae [163,303], Callionymidae [303], Cancridae [163], Carangidae [303], Clupeidae [303], Crangonidae [303], Eledonidae [163], Gadidae [163,303], Galatheidae [163], Loliginidae [163], Majidae [163], Merlucciidae [163,303], Myidae [163], Nephropidae [163], Nephtyidae [303], Oregoniidae [163], Paguridae [163,303], Palaemonidae [303], Pandalidae [303], Parechinidae [303], Pleuronectidae [163], Polybiidae [163,303], Rajidae [163], Scombridae [163,303], Sepiolidae [163], Soleidae [303], Squillidae [163], Triglidae [163], Upogebiidae [163]	13	2	112 (126)
Somniosus microcephalus	57	Acanthephyridae [304], Agonidae [304], Anarhichadidae [304-307,309], Architeuthidae [121], Argentinidae [309], Arhynchobatidae [121], Balaenopteridae [305], Bathylagidae [304], Bathypolypodidae [304], Brachioteuthidae [121], Buccinidae [308], Chimaeridae [309], Clupeidae [309], Cottidae [304,307-308], Cranchiidae [121], Cyclopteridae [304,307-309], Cycloteuthidae [121], Etmopteridae [309], Gadidae [304-305,307,309], Gonatidae [121,305,307], Gorgonocephalidae [305,307], Histioteuthidae [121], Liparidae [304], Lithodidae [307], Lotidae [304,309], Lysianassidae [306], Macrouridae [304,307,310], Mastigoteuthidae [121], Monodontidae [306], Myctophidae [309], Myxinidae [307], Neoteuthidae [121], Nototheniidae [121], Octopodidae [304], Octopoteuthidae [121], Ommastrephidae [121], Onychoteuthidae [121], Ophiactidae [305], Ophiuridae [306], Opisthoteuthidae [304], Oplophoridae [304], Oregoniidae [305,307], Otariidae [121], Pandalidae [304], Phocidae [304-305,307-308,310-312], Pleuronectidae [304-305,307-309,311], Psychrolutidae [304], Rajidae [304-305,307-309], Salmonidae	41	10	230 (256)

		[310,312], Sebastidae [304-305,307,309], Solasteridae [310], Somniosidae [305-306,310], Stichaeidae [304], Strongylocentrotidae [305,308], Uristidae [306], Ursidae [307,309], Zoarcidae [304,307-308,310]			
Somniosus pacificus	28	Anoplopomatidae [314], Cirolanidae [304], Clupeidae [316], Cranchiidae [316], Crangonidae [315], Delphinidae [317], Enteroctopodidae [304,314-315], Gadidae [304,313-316], Gonatidae [304,316], Liparidae [313,316], Macrouridae [304,313,316], Myctophidae [316], Nototheniidae [317], Octopodidae [318], Ommastrephidae [304], Onychoteuthidae [316], Oregoniidae [318], Paguridae [315], Phocidae [314,318], Pleuronectidae [314-316], Psychrolutidae [316], Ranellidae [315,318], Salmonidae [313-316], Sebastidae [314-315], Spongiidae [316], Squalidae [314], Uristidae [316], Zoarcidae [316]	28	9	461 (545)
Sphyrna lewini	137	Acanthuridae [2,320-321,329-330], Achiridae [325,328], Alloposidae [79,165], Alpheidae [50,323,329-330], Amphitretidae [79], Ancistrocheiridae [2,321,326,332], Anguillidae [319], Apogonidae [329-331], Argonautidae [321,324,326,328], Ariidae [320,323], Atherinidae [329-330], Aulostomidae [49], Balistidae [50,319,321,324,328,331], Batrachoididae [2,321], Belonidae [320-321,331], Bothidae [320,323-324,328,330-331], Brachioteuthidae [332], Callionymidae [2,331], Carangidae [2,24,79,320-326,328,331], Carcharhinidae [320], Centropomidae [328], Chaetodontidae [330], Champsodontidae [331], Chanidae [330], Cheilodactylidae [320], Chirocentridae [319], Chiroteuthidae [79,332], Chlorophthalmidae [319], Cichlidae [319], Clupeidae [2,320-324,326,328-329,331], Congridae [320,330-331], Coryphaenidae [2,321,326,328], Cranchiidae [79,332], Cynoglossidae [320,325,328], Dactylopteridae [320,331], Dasyatidae [320], Delphinidae [319], Diodontidae [331], Dussumieriidae [2,320], Echeneidae [321], Elopidae [319], Engraulidae [40,50,320,323-326,328-330], Enoploteuthidae [2,320-321,324,328,330], Ephippidae [319], Exocoetidae [2,320-321,323,326,331], Gempylidae [2,79,319], Gerreidae [319,322-323,328], Gobiidae [329-330], Gonatidae [2,321,326], Gymnuridae [320], Haemulidae [24,50,320,323], Hemiramphidae	106	28	4157 (5320)

[2,321,326,328,330], Histioteuthidae [2,79,235,326,330,332], Holocentridae [328], Istiophoridae [319], Kyphosidae [320], Labridae [321,324,328,330], Lamnidae [319], Leiognathidae [40,320,331], Lethrinidae [331], Loliginidae [2,50,320-321,323-328,332], Lutjanidae [2,79,325,331], Lycoteuthidae [165,320,332], Lysiosquillidae [329], Malacanthidae [328], Mastigoteuthidae [2,321,326], Menidae [331], Merlucciidae [2,320,326], Monacanthidae [79,331], Mugilidae [24,320,322,324-326,328-329], Mullidae [235,320,328,330-331], Munididae [2,321,324,328], Muraenesocidae [27,320], Muraenidae [2,24,79,319,321,324,326,328], Muricidae [325], Myctophidae [319,326], Myliobatidae [319], Nemipteridae [331], Octopodidae [2,79,235,320,325-326,332], Octopoteuthidae [2,79,320,326,332], Odontaspididae [319], Ommastrephidae [2,79,165,319,321,326,328,330,332], Onychoteuthidae [326], Ophichthidae [2,324,326,328], Ophidiidae [328], Opisthoteuthidae [332], Oplegnathidae [320], Ostraciidae [320,331], Palaemonidae [323,329], Pandalidae [2], Paralichthyidae [24,50,321-324,326,328], Penaeidae [2,24,40,50,321-326,328,331], Pentanchidae [320], Peristediidae [320], Pholidoteuthidae [2,326], Pinguipedidae [320], Platycephalidae [320,331], Pleuronectidae [320,328], Plotosidae [320], Polynemidae [331], Pomacentridae [320,330], Pomatomidae [320], Portunidae [50,323,328-329], Priacanthidae [2,320,331], Processidae [325,328], Pseudocarchariidae [320], Rajidae [50,320], Rhinobatidae [320], Scaridae [79,329-331], Sciaenidae [2,50,320,322-326,328], Scombridae [2,79,235,320-324,326,328,331], Scorpaenidae [2], Scyliorhinidae [320], Sepiidae [320,331-332], Sepiolidae [165], Serranidae [2,24,320-321,326,328], Sicyoniidae [323-325,328], Sillaginidae [331], Solenoceridae [2,50,325], Sparidae [50,320,323], Sphyraenidae [79,320,328], Sphyrnidae [319,330], Squalidae [27,320-321,323,331], Squillidae [235,324-325,327-329,331], Squatinidae [320], Stromateidae [324,328], Syngnathidae [320], Synodontidae [2,50,320-321,323-324,326,328-331], Terapontidae [320], Tetraodontidae [331], Thysanoteuthidae [2,325-326], Triacanthidae [331], Trichiuridae [50,320,331], Triglidae [24,320],

		Vampyroteuthidae [235], Xanthidae [2], Yoldiidae [323]			
Sphyrna mokarran	40	Ancistrocheiridae [332], Ariidae [333], Balistidae [331], Carcharhinidae [333], Cynoglossidae [331], Dasyatidae [333], Diodontidae [331], Echeneidae [333], Gymnuridae [333], Haemulidae [331,333], Istiophoridae [331], Labridae [331], Latidae [331], Loliginidae [332], Lutjanidae [331], Myliobatidae [333], Nemipteridae [331], Octopodidae [332], Octopoteuthidae [332], Ommastrephidae [332], Oplegnathidae [333], Ostraciidae [331], Palinuridae [331,333], Penaeidae [331], Pentanchidae [333], Platycephalidae [333], Portunidae [331], Psettodidae [331], Rajidae [333], Rhinobatidae [333], Sciaenidae [331,333], Scombridae [331], Scyliorhinidae [333], Scyllaridae [331], Sepiidae [332-333], Sphyraenidae [331], Squillidae [331], Tetraodontidae [331], Trichiuridae [331], Triglidae [333]	27	3	423 (493)
Sphyrna tiburo	33	Aethridae [334], Batrachoididae [334], Bothidae [334], Calappidae [334], Callianassidae [334], Cancridae [334], Clupeidae [51], Congridae [337], Elopidae [334], Engraulidae [337], Epialtidae [334,336], Leucosiidae [334,336], Limulidae [336], Loliginidae [51,334,336], Menippidae [336], Mithracidae [334], Mugilidae [51], Octopodidae [334], Onuphidae [334], Ophichthidae [334,336], Ophiolepididae [51], Ovalipidae [334], Paguridae [334,336], Palinuridae [334,337], Pandalidae [334], Panopeidae [334], Penaeidae [51,334-337], Portunidae [51,334-337], Sciaenidae [51,334,337], Sicyoniidae [334], Squillidae [51,334,336], Syngnathidae [334], Xanthidae [51,334]	65	5	1262 (1317)
Sphyrna tudes	2	Ariidae [338], Penaeidae [338]	8	1	155 (155)
Sphyrna zygaena	73	Achiridae [340], Amathinidae [340], Amphitretidae [2,340], Anisakidae [341], Ancistrocheiridae [2,332,340], Argonautidae [2,23,332], Ariidae [23], Arripidae [9], Atherinopsidae [50], Balistidae [2,339], Belonidae [9,27,235], Berycidae [9,27], Carangidae [2,23,27,49-50,340-341], Chaetodontidae [23], Chiroteuthidae [23,339], Clupeidae [2,23,27,50,340-341], Congiopodidae [23], Congridae [23], Coryphaenidae [2,340], Cranchiidae [235,332], Cynoglossidae [23], Diodontidae [339], Dussumieriidae [23], Engraulidae [9,23,340], Enoploteuthidae [2,332],	45	13	943 (1463)

		Exocoetidae [2,340], Fistulariidae [2], Gempylidae [2,9], Gerreidae [2], Gonatidae [2,340], Haemulidae [340-341], Hemiramphidae [2,9,50,340-341], Histioteuthidae [2,235,332,340], Loliginidae [2,9,23,39,50,332,340-341], Lycoteuthidae [23,332], Macrouridae [23], Mastigoteuthidae [2,340], Merlucciidae [2,23], Monacanthidae [9,27,339], Mugilidae [2,23,27,50], Myctophidae [340], Myliobatidae [235], Naticidae [340], Nuculanidae [340], Octopodidae [332], Octopoteuthidae [2,332,340], Ocythoidae [23], Ommastrephidae [2,9,23,50,332,339-340], Onychoteuthidae [2,235,332,340], Ophichthidae [2,340], Ophidiidae [2,23,340], Ovalipidae [9], Paralichthyidae [2,340], Pempheridae [9], Penaeidae [9], Pholidoteuthidae [2], Phosichthyidae [2], Platycephalidae [27], Portunidae [341], Sciaenidae [2,23,50,340-341], Scomberesocidae [23,27], Scombridae [2,23,27,50,58,340], Sepiidae [9,23,332], Serranidae [2], Sparidae [9,23,50], Sphyraenidae [27], Squalidae [23], Syngnathidae [9], Synodontidae [2], Terapontidae [9], Thysanoteuthidae [2,340], Trichiuridae [23,50,341], Triglidae [23,27,340]			
Squalus acanthias	145	Actiniidae [342], Alpheidae [343], Ammodytidae [163,218,221,345,348,352,357-358], Ampeliscidae [346], Anguillidae [218,346], Anoplopomatidae [357], Aphroditidae [351,354], Argentinidae [116], Atherinidae [346], Batrachoididae [342,351,357], Belonidae [163], Beroidae [353,358], Bolinopsidae [353,357], Bothidae [343], Buccinidae [358], Callianassidae [163], Callionymidae [103,163,352,358], Cancridae [162-163,218,221,348,355,358], Carangidae [116,163,342-343,349,358], Centracanthidae [343], Centriscidae [116], Centrolophidae [342], Cepolidae [343], Cheilodactylidae [354], Chimaeridae [116,357], Cirolanidae [342], Clupeidae [103,162-163,218,221,342-349,352-353,355,357-358], Congridae [342-343], Cottidae [345,348], Cranchiidae [116], Crangonidae [116,163,349], Cryptacanthodidae [221], Cyllopodidae [116], Cymothoidae [346], Cynoglossidae [344-345], Echiuridae [342], Dussumieriidae [103], Eledonidae [163,343,352], Embiotocidae [357], Emmelichthyidae [116], Engraulidae [103,162-163,342-344,346,348-349,351,354], Enteroctopodidae	334	23	40827 (72204)

[351,353], Epialtidae [342,354], Eunicidae [116], Euphausiidae [116,162,221,342,345,347-348,350,354-355,357], Gadidae [163,221,342-343,347,349,352,357-358], Gammaridae [221,342], Gempylidae [346], Glyceridae [342,346], Gobiidae [343,345-346,349], Gonatidae [353], Goneplacidae [116], Hexagrammidae [162,347], Histioteuthidae [348], Hoplichthyidae [116], Hyperiidae [116,162,342,351,353-355], Latridae [346], Limacinidae [162], Lithodidae [342], Litocheiridae [346], Loliginidae [163,218,221,342-343,351,353-354,357], Lophiidae [221], Lotidae [349], Macrouridae [116], Majidae [116,342], Merlucciidae [103,116,162,218,221,342-343,348,351-354,356-358], Metridiidae [358], Moridae [116,342,355], Moronidae [344], Mullidae [343,349], Munididae [116,342,351,353-355], Muricidae [349], Myctophidae [103,116,342,354-355], Myxinidae [342,354], Nassariidae [346], Naticidae [345], Nephropidae [116,163], Nereididae [358], Nototheniidae [342,351,353-354], Octopodidae [342-343,346,352,354-355,357], Ommastrephidae [103,116,218,221,342-343,346,351,353-355], Onuphidae [116,343], Onychoteuthidae [116], Ophidiidae [116,342,345,351], Oplophoridae [116], Oreosomatidae [116], Osmeridae [348,357], Ovalipidae [355], Paguridae [163,354,358], Palaemonidae [343], Palinuridae [116], Pandalidae [116,162-163,357], Paralichthyidae [162,218,221,344-345,351], Pasiphaeidae [163], Penaeidae [103,346,354], Percophidae [342,355], Petromyzontidae [348], Phycidae [221,345], Phyllodocidae [346], Pleurobrachiidae [351,355,357-358], Pleuronectidae [162-163,221,352,354], Polybiidae [163], Polymixiidae [345], Polyprionidae [342], Porpitidae [162], Portunidae [345], Rajidae [342], Salmonidae [347], Salpidae [353-355], Sciaenidae [342,344], Scomberesocidae [103,352], Scombridae [163,221,342,352,358], Scophthalmidae [221], Scorpaenidae [116,343,347,349,357], Sebastidae [116,221,342], Sepiadariidae [355], Sepiidae [343], Sepiolidae [116,163,345,351,353-354], Sergestidae [342], Serolidae [342,351,354], Serranidae [116,342], Sillaginidae [346], Sipunculidae [342], Soleidae [343], Solenoceridae [342,351,354], Sparidae [218,342-343],

		Squalidae [103,342,346,349,355,357], Squillidae [103,346], Sternoptychidae [103,116], Stichaeidae [163], Stromateidae [218,342,351,353], Syngnathidae [345,349], Terebellidae [346], Tetrarogidae [346], Tetrasquillidae [355], Thalasseleotrididae [355], Trichiuridae [103], Trichopeltariidae [116,353-354], Triglidae [218,342-343,345,351,358], Upogebiidae [163,349], Uranoscopidae [349], Urechidae [355], Varunidae [346], Zeniontidae [342], Zoarcidae [221,351,357]			
Squalus blainville	46	Alpheidae [294,360], Aphroditidae [360], Argentinidae [359-360], Astropectinidae [360], Atelecyclidae [360], Blenniidae [360], Bothidae [360], Callionymidae [360], Calliostomatidae [294], Carangidae [360], Centracanthidae [360], Cepolidae [360-361], Cirolanidae [294], Citharidae [360], Clupeidae [360-361], Congridae [360], Crangonidae [360], Echinasteridae [360], Eledonidae [360], Engraulidae [360-361], Euphrosinidae [361], Gadidae [359-360], Gobiidae [360-361], Goneplacidae [360-361], Haliotidae [294], Inachidae [360], Leucosiidae [360], Loliginidae [294,360], Merlucciidae [360], Mullidae [360-361], Nereididae [360], Octopodidae [294,360], Paguridae [294], Palaemonidae [294], Penaeidae [360-361], Polybiidae [294,360-361], Portunidae [294], Scyliorhinidae [361], Sepiidae [294,360-361], Sepiolidae [360], Serranidae [360], Sicyoniidae [360], Sipunculidae [294,360], Soleidae [294,360], Sparidae [360], Squillidae [360]	11	4	1103 (1471)
Squalus megalops	60	Acropomatidae [299], Alpheidae [299], Aphroditidae [299], Apogonidae [299], Argentinidae [73,299], Callionymidae [103], Carangidae [299], Centriscidae [299], Cirolanidae [299], Clupeidae [299], Congridae [299], Crangonidae [73], Cyttidae [299], Diogenidae [299], Dussumieriidae [103], Engraulidae [73,103], Eunicidae [299], Fasciolariidae [299], Gempylidae [299], Gerreidae [299], Histioteuthidae [299], Leucosiidae [299], Loliginidae [73], Lumbrineridae [299], Macrouridae [299], Merlucciidae [299], Myctophidae [73,103,299], Nannosquillidae [299], Narcinidae [299], Nereididae [299], Octopodidae [73,103,299], Ommastrephidae [73,299], Ophichthidae [299], Otariidae [299], Paguridae [299], Palaemonidae	9	3	694 (1076)

		[299], Palinuridae [299], Paraulopidae [299], Penaeidae [103,299], Philinidae [299], Pilumnidae [299], Platycephalidae [299], Portunidae [299], Rajidae [299], Scomberesocidae [73], Scombridae [73,299], Scorpaenidae [299], Sebastidae [299], Serrivomeridae [299], Sillaginidae [299], Sipunculidae [299], Solenoceridae [299], Squalidae [299], Squillidae [103,299], Triakidae [299], Trichiuridae [73,103], Triglidae [299], Turbinidae [299], Urolophidae [299], Volutidae [299]			
Squalus mitsukurii	34	Benthesicymidae [289], Berycidae [289], Bothidae [289], Callanthiidae [289], Calappidae [103], Callionymidae [103], Clupeidae [103], Congridae [103,289], Echeneidae [289], Emmelichthyidae [103,289], Enoploteuthidae [289], Epigonidae [289], Euphausiidae [289], Gnathophausiidae [289], Histioteuthidae [289], Macrouridae [103], Merlucciidae [103], Monacanthidae [289], Myctophidae [103,289], Ommastrephidae [103,289], Oplophoridae [289], Paralepididae [289], Parapaguridae [103], Phronimidae [289], Pyrosomatidae [289], Salpidae [289], Scomberesocidae [103], Sebastidae [103], Sepiolidae [289], Sergestidae [289], Sternoptychidae [289], Stomiidae [289], Trichiuridae [103], Zeidae [103]	10	2	312 (564)
Squatina californica	18	Batrachoididae [231], Carangidae [231], Clupeidae [231], Dussumieriidae [231], Enoploteuthidae [231], Holocentridae [231], Labridae [231], Mastigoteuthidae [231], Mugilidae [231], Muraenidae [231], Ophidiidae [231], Pomacentridae [231], Scombridae [231], Serranidae [231], Sicyoniidae [231], Stromateidae [231], Synodontidae [231], Triglidae [231]	9	1	190 (414)
Squatina squatina	4	Majidae [163], Pleuronectidae [163], Polybiidae [163], Triglidae [163]	7	1	18 (19)
Triakis semifasciata	35	Atherinidae [62,230], Batrachoididae [62,230], Blepharipodidae [62], Bothidae [62], Callianassidae [62,230], Cancridae [62,134-135,230], Clupeidae [62,230], Cottidae [62,135], Crangonidae [62,230], Cynoglossidae [62], Embiotocidae [62,135,230], Engraulidae [62,230], Gobiidae [62,230], Grapsidae [62,134], Hippidae [62], Loliginidae [135], Mactridae [135], Myliobatidae [135,230], Nereididae [230], Octopodidae [62,230], Ophidiidae [135], Paralichthyidae [135,230], Petromyzontidae [135], Pinnotheridae [135], Rajidae [135],	49	4	676 (827)

Rhinobatidae [62], Sciaenidae [62], Scorpaenidae [62], Solenidae [135], Squalidae [135], Syngnathidae [135], Triakidae [230], Upogebiidae [62,135,230], Urechidae [62,134-135,230], Varunidae [62,134]

References:

1. Rosas-Luis, R., Loor-Andrade, P., Carrera-Fernández, M., Pincay-Espinoza, J. E., Vinces-Ortega, C., & Chompoy-Salazar, L. (2016). Cephalopod species in the diet of large pelagic fish (sharks and billfishes) in Ecuadorian waters. Fisheries Research, 173, 159-168.
2. Galván-Magaña, F., Polo-Silva, C., Hernández-Aguilar, S. B., Sandoval-Londoño, A., Ochoa-Díaz, M. R., Aguilar-Castro, N., ... & Abitia-Cárdenas, L. A. (2013). Shark predation on cephalopods in the Mexican and Ecuadorian Pacific Ocean. Deep Sea Research Part II: Topical Studies in Oceanography, 95, 52-62.
3. Gorni, G. R., Goitein, R., & Amorim, A. F. D. (2013). Description of diet of pelagic fish in the southwestern Atlantic, Brazil. Biota Neotropica, 13(1), 61-69.
4. Preti, A., Kohin, S., Dewar, H., & Ramon, D. (2008). Feeding habits of the bigeye thresher shark (Alopias superciliosus) sampled from the California-based drift gillnet fishery. CalCOFI Rep, 49, 202-211.
5. Polo-Silva, C., Rendón, L., & Galván-Magaña, F. (2009). Descripción de la dieta de los tiburones zorro (Alopias pelagicus) y (Alopias superciliosus) durante la época lluviosa en aguas ecuatorianas. Pan-American Journal of Aquatic Sciences, 4(4), 556-571.
6. Polo-Silva, C., Baigorri-Santacruz, A., Galvan-Magana, F., Grijalba-Bendeck, M., & Sanjuan-Munoz, A. (2007). Food habits of thresher shark Alopias superciliosus (Lowe, 1839) in Ecuadorian Pacific. Revista de Biologia Marina y Oceanografia, 42(1), 59.
7. Gruber, S. H., & Compagno, L. (1981). Taxonomic status and biology of the bigeye thresher Alopias superciliosus (Mediterranean Sea, New Zealand. Fishery Bulletin, 79(4), 617-640.
8. Stillwell, C. E., & Casey, J. G. (1976). Observations on bigeye thresher shark, Alopias superciliosus, in Western North-Atlantic. Fishery Bulletin, 74(1), 221-225.
9. Rogers, P. J., Huveneers, C., Page, B., Hamer, D. J., Goldsworthy, S. D., Mitchell, J. G., & Seuront, L. (2012). A quantitative comparison of the diets of sympatric pelagic sharks in gulf and shelf ecosystems off southern Australia. ICES Journal of Marine Science: Journal du Conseil, 69(8), 1382-1393.
10. Preti, A., Soykan, C. U., Dewar, H., Wells, R. D., Spear, N., & Kohin, S. (2012). Comparative feeding ecology of shortfin mako, blue and thresher sharks in the California Current. Environmental Biology of Fishes, 95(1), 127-146.
11. Preti, A., Smith, S. E., & Ramon, D. A. (2004). Diet differences in the thresher shark (Alopias vulpinus) during transition from a warm-water regime to a cool-water regime off California-Oregon, 1998-2000. California Cooperative Oceanic Fisheries Investigations Report, 45, 118.
12. Preti, A., Smith, S. E., & Ramon, D. A. (2001). Feeding habits of the common thresher shark (Alopias vulpinus) sampled from the California-based drift gill net fishery, 1998-1999. California Cooperative Oceanic Fisheries Investigations Report, 145-152.
13. Pascoe, P. L. (1986). Fish otoliths from the stomach of a thresher shark, Alopias vulpinus. Journal of the Marine Biological Association of the United Kingdom, 66(2), 315-317.
14. Fischer, A. F., Hazin, F. H. V., Carvalho, F., Viana, D. L., Rêgo, M. G., & Wor, C. (2009). Biological aspects of sharks caught off the Coast of Pernambuco, Northeast Brazil. Brazilian Journal of Biology, 69(4), 1173-1181.
15. Papastamatiou, Y. P., Wetherbee, B. M., Lowe, C. G., & Crow, G. L. (2006). Distribution and diet of four species of carcharhinid shark in the Hawaiian Islands: evidence for resource partitioning and competitive exclusion. Marine Ecology Progress Series, 320, 239-251.

16. Wetherbee, B. M., Crow, G. L., & Lowe, C. G. (1997). Distribution, reproduction and diet of the gray reef shark Carcharhinus amblyrhynchos in Hawaii. Marine Ecology Progress Series, 151, 181-189.
17. Stevens, J. D., & McLoughlin, K. J. (1991). Distribution, size and sex composition, reproductive biology and diet of sharks from northern Australia. Marine and Freshwater Research, 42(2), 151-199.
18. Cliff, G., & Dudley, S. F. J. (1991a). Sharks caught in the protective gill nets off Natal, South Africa. 5. The Java shark Carcharhinus amboinensis (Müller & Henle). South African Journal of Marine Science, 11(1), 443-453.
19. Tillett, B. J., Meekan, M. G., & Field, I. C. (2014). Dietary overlap and partitioning among three sympatric carcharhinid sharks. Endangered Species Research, 25(3), 283-293.
20. Kinney, M. J., Hussey, N. E., Fisk, A. T., Tobin, A. J., & Simpfendorfer, C. A. (2011). Communal or competitive? Stable isotope analysis provides evidence of resource partitioning within a communal shark nursery. Marine Ecology Progress Series, 439, 263-276.
21. Cliff, G., & Dudley, S. F. J. (1992). Sharks caught in the protective gill nets off Natal, South Africa. 6. The copper shark Carcharhinus brachyurus (Günther). South African Journal of Marine Science, 12(1), 663-674.
22. Lucifora, L. O., García, V. B., Menni, R. C., Escalante, A. H., & Hozbor, N. M. (2009a). Effects of body size, age and maturity stage on diet in a large shark: ecological and applied implications. Ecological Research, 24(1), 109-118.
23. Smale, M. J. (1991). Occurrence and feeding of three shark species, Carcharhinus brachyurus, C. obscurus and Sphyrna zygaena, on the Eastern Cape coast of South Africa. South African Journal of Marine Science, 11(1), 31-42.
24. Avendaño-Alvarez, José Otilio, Horacio Pérez-España, David Salas-Monreal, and Emiliano García-Rodríguez. Captures and Diet of Three Sharks Species in the Veracruz Reef System. Growth, 23, 25.
25. Bethea, D. M., Buckel, J. A., & Carlson, J. K. (2004). Foraging ecology of the early life stages of four sympatric shark species. Marine Ecology Progress Series, 268, 245-264.
26. Allen, B. R., & Cliff, G. (2000). Sharks caught in the protective gill nets off Kwazulu-Natal, South Africa. 9. The spinner shark Carcharhinus brevipinna (Müller and Henle). South African Journal of Marine Science, 22(1), 199-215.
27. Stevens, J. D. (1984a). Biological observations on sharks caught by sport fisherman of New South Wales. Marine and Freshwater Research, 35(5), 573-590.
28. Varghese, S. P., Gulati, D. K., Unnikrishnan, N., & Ayoob, A. E. (2015). Biological aspects of silky shark Carcharhinus falciformis in the eastern Arabian Sea. Journal of the Marine Biological Association of the United Kingdom, 96(7), 1437-1447.
29. Duffy, L. M., Olson, R. J., Lennert-Cody, C. E., Galván-Magaña, F., Bocanegra-Castillo, N., & Kuhnert, P. M. (2015). Foraging ecology of silky sharks, Carcharhinus falciformis, captured by the tuna purse-seine fishery in the eastern Pacific Ocean. Marine Biology, 162(3), 571-593.
30. Cabrera-Chávez-Costa, A. A., Galván-Magaña, F., & Escobar-Sánchez, O. (2010). Food habits of the silky shark Carcharhinus falciformis (Müller & Henle, 1839) off the western coast of Baja California Sur, Mexico. Journal of Applied Ichthyology, 26(4), 499-503.
31. Nancy-Elizabeth, R. P., Cerdenares-Ladron de Guevara, G., Liney Lopez-Herrera, D., & Altamirano-Ramirez, I. R. (2016). Trophic interactions between five pelagic fish species cohabiting in the coast of Oaxaca, Mexico. Hidrobiologica, 26(1), 77-85.

32. Wetherbee, B. M., Crow, G. L., & Lowe, C. G. (1996). Biology of the Galapagos shark, Carcharhinus galapagensis, in Hawai'i. Environmental biology of fishes, 45(3), 299-310.
33. Hoffmayer, E. R., & Parsons, G. R. (2003). Food habits of three shark species from the Mississippi Sound in the northern Gulf of Mexico. Southeastern Naturalist, 2(2), 271-280.
34. Castro, J. I. (1993). The biology of the finetooth shark, Carcharhinus isodon. Environmental Biology of Fishes, 36(3), 219-232.
35. Gurshin, C. W. (2007). Shark nursery grounds in Sapelo Island National Estuarine Research Reserve, Georgia. In American Fisheries Society Symposium, 50, 141.
36. Salini, J. P., Blaber, S. J. M., & Brewer, D. T. (1992). Diets of sharks from estuaries and adjacent waters of the north-eastern Gulf of Carpentaria, Australia. Marine and Freshwater Research, 43(1), 87-96.
37. Frisch, A. J., Ireland, M., Rizzari, J. R., Lönnstedt, O. M., Magnenat, K. A., Mirbach, C. E., & Hobbs, J. P. A. (2016). Reassessing the trophic role of reef sharks as apex predators on coral reefs. Coral Reefs, 35(2), 459-472.
38. Salini, J. P., Blaber, S. J. M., & Brewer, D. T. (1990). Diets of piscivorous fishes in a tropical Australian estuary, with special reference to predation on penaeid prawns. Marine Biology, 105(3), 363-374.
39. Lipinski, M. R., & Soule, M. (2007). Disintegration of a large concentration of loliginid squid as a response to predation. Reviews in Fish Biology and Fisheries, 17(2-3), 477-485.
40. Simpfendorfer, C. A., & Milward, N. E. (1993). Utilisation of a tropical bay as a nursery area by sharks of the families Carcharhinidae and Sphyrnidae. Environmental Biology of Fishes, 37(4), 337-345.
41. Garibaldi, F. & Relini, L. (2012). Record of Carcharhinus falciformis (Bibron in Müller & Henle, 1839), in Italian waters (Ligurian Sea, Northwestern Mediterranean). Cybium, 36(2), 399-400.
42. Cliff, G., & Dudley, S. F. J. (1991b). Sharks caught in the protective gill nets off Natal, South Africa. 4. The bull shark Carcharhinus leucas Valenciennes. South African Journal of Marine Science, 10(1), 253-270.
43. Werry, J. M., Lee, S. Y., Otway, N. M., Hu, Y., & Sumpton, W. (2011). A multi-faceted approach for quantifying the estuarine–nearshore transition in the life cycle of the bull shark, Carcharhinus leucas. Marine and Freshwater Research, 62(12), 1421-1431.
44. Tuma, R. E. (1976). An investigation of the feeding habits of the bull shark, Carcharhinus leucas, in the Lake Nicaragua-Rio San Juan system. Investigations of the Ichthyofauna of Nicaraguan Lakes, 39.
45. Sadowsky, V. (1971). Notes on the bull shark Carcharhinus leucas in the lagoon region of Cananéia, Brazil. Boletim do Instituto Oceanográfico, 20(2), 71-78.
46. Snelson, F. F., Mulligan, T. J., & Williams, S. E. (1984). Food habits, occurrence, and population structure of the bull shark, Carcharhinus leucas, in Florida coastal lagoons. Bulletin of Marine Science, 34(1), 71-80.
47. Thorburn, D. C., & Rowland, A. J. (2008). Juvenile bull sharks' Carcharhinus leucas' (Valenciennes, 1839) in northern Australian rivers. Beagle: Records of the Museums and Art Galleries of the Northern Territory, 24, 79.
48. Dudley, S. F. J., & Cliff, G. (1993). Sharks caught in the protective gill nets off Natal, South Africa. 7. The blacktip shark Carcharhinus limbatus

(Valenciennes). South African Journal of Marine Science, 13(1), 237-254.

49. Crow, G. L., Lowe, C. G., & Wetherbee, B. M. (1996). Shark records from longline fishing programs in Hawai'i with comments on Pacific Ocean distributions. Pacific Science, 50(4), 382-392.
50. Bornatowski, H., Braga, R. R., Abilhoa, V., & Corrêa, M. F. M. (2014). Feeding ecology and trophic comparisons of six shark species in a coastal ecosystem off southern Brazil. Journal of Fish Biology, 85(2), 246-263.
51. Plumlee, J. D., & Wells, R. D. (2016). Feeding ecology of three coastal shark species in the northwest Gulf of Mexico. Marine Ecology Progress Series, 550, 163-174.
52. Abdurahiman, K. P., Nayak, T. H., Zacharia, P. U., & Mohamed, K. S. (2010). Trophic organisation and predator–prey interactions among commercially exploited demersal finfishes in the coastal waters of the southeastern Arabian Sea. Estuarine, Coastal and Shelf Science, 87(4), 601-610.
53. Barry, K. P., Condrey, R. E., Driggers, W. B., & Jones, C. M. (2008). Feeding ecology and growth of neonate and juvenile blacktip sharks Carcharhinus limbatus in the Timbalier–Terrebone Bay complex, LA, USA. Journal of Fish Biology, 73(3), 650-662.
54. Tavares, R. (2008). Occurrence, diet and growth of juvenile blacktip sharks, Carcharhinus limbatus, from Los Roques Archipelago National Park, Venezuela. Caribbean Journal of Science, 44(3), 291-302.
55. Castro, J. I. (1996). Biology of the blacktip shark, Carcharhinus limbatus, off the southeastern United States. Bulletin of marine science, 59(3), 508-522.
56. Tavares, R., & Provenzano, F. (2000). Alimentación de los juveniles del tiburón macuira, Carcharhinus limbatus (Valenciennes, 1839), en el Parque Nacional Archipiélago de Los Roques, Venezuela. Acta Biologica Venezuelica, 20(1), 59-67.
57. Zhang, Y., Peimao, C., & Xiaoyun, L. (2006). Relationship between food and food habits for main fish species in the transitional waters around coral reefs in the South China Sea. Periodical of Ocean University of China, 36(4), 635-638.
58. Strasburg, D. W. (1958). Distribution, abundance, and habits of pelagic sharks in the central Pacific Ocean. Fisheries, 1, 2S.
59. Schuck, H. A., & Clark, J. R. (1951). Record of a white-tipped shark, Carcharhinus longimanus, from the northwestern Atlantic. Copeia, 1951(2), 172-172.
60. Stevens, J. D. (1984b). Life-history and ecology of sharks at Aldabra Atoll, Indian Ocean. Proceedings of the Royal Society of London B: Biological Sciences, 222(1226), 79-106.
61. Papastamatiou, Y. P., Caselle, J. E., Friedlander, A. M., & Lowe, C. G. (2009). Distribution, size frequency, and sex ratios of blacktip reef sharks Carcharhinus melanopterus at Palmyra Atoll: a predator-dominated ecosystem. Journal of Fish Biology, 75(3), 647-654.
62. Talent, L. G. (1976). Food habits of the leopard shark, Triakis semifasciata, in Elkhorn Slough, Monterey Bay, California. California Fish and Game, 62(4), 286-298.
63. Lyle, J. M. (1987). Observations on the biology of Carcharhinus cautus (Whitley), C. melanopterus (Quoy & Gaimard) and C. fitzroyensis (Whitley) from northern Australia. Marine and Freshwater Research, 38(6), 701-710.
64. Dudley, S. F. J., Cliff, G., Zungu, M. P., & Smale, M. J. (2005). Sharks caught in the protective gill nets off KwaZulu-Natal, South Africa. 10. The dusky shark Carcharhinus obscurus (Lesueur 1818). African Journal of Marine Science, 27(1), 107-127.
65. Simpfendorfer, C. A., Goodreid, A., & McAuley, R. B. (2001a). Diet of three commercially important shark species from Western Australian waters.

Marine and freshwater research, 52(7), 975-985.

66. Gelsleichter, J., Musick, J. A., & Nichols, S. (1999). Food habits of the smooth dogfish, Mustelus canis, dusky shark, Carcharhinus obscurus, Atlantic sharpnose shark, Rhizoprionodon terraenovae, and the sand tiger, Carcharias taurus, from the northwest Atlantic Ocean. Environmental Biology of Fishes, 54(2), 205-217.
67. Van der Elst, R. P. (1979). A proliferation of small sharks in the shore-based Natal sport fishery. Environmental Biology of Fishes, 4(4), 349-362.
68. Schwartz, F. J., & Jensen, C. (1996). Coronetfishes (Fistularia, Fistulariidae) as Food of the Dusky Shark, Carcharhinus obscurus (Carcharhinidae), from North Carolina, and Problems Identifying Fistularids from only Heads. Journal of the Elisha Mitchell Scientific Society, 112, 40-44.
69. Ellis, J. K., & Musick, J. A. (2007). Ontogenetic changes in the diet of the sandbar shark, Carcharhinus plumbeus, in lower Chesapeake Bay and Virginia (USA) coastal waters. Environmental biology of fishes, 80(1), 51-67.
70. McElroy, W. D., Wetherbee, B. M., Mostello, C. S., Lowe, C. G., Crow, G. L., & Wass, R. C. (2006). Food habits and ontogenetic changes in the diet of the sandbar shark, Carcharhinus plumbeus, in Hawaii. Environmental Biology of Fishes, 76(1), 81-92.
71. Stillwell, C. E., & Kohler, N. E. (1993). Food habits of the sandbar shark Carcharhinus plumbeus off the U. S. northeast coast, with estimates of daily ration. Fishery Bulletin, 91(1), 138-150.
72. Saidi, B., Bradai, M. N., Bouain, A., & Capape, C. (2007). Feeding habits of the sandbar shark Carcharhinus plumbeus (Chondrichthyes: Carcharhinidae) from the Gulf of Gabes, Tunisia. Cahiers de biologie marine, 48(2), 139-144.
73. Huh, S. H., Park, J. M., Park, S. C., Kim, J. H., & Baeck, G. W. (2010). Feeding Habits of 6 Shark Species in the Southern Sea of Korea. Korean Journal of Fisheries and Aquatic Sciences, 43(3), 254-261.
74. Medved, R. J., Stillwell, C. E., & Casey, J. J. (1985). Stomach contents of young sandbar sharks, Carcharhinus plumbeus, in Chincoteague Bay, Virginia. Fishery Bulletin, 83(3), 395-402.
75. Medved, R. J., & Marshall, J. A. (1981). Feeding behavior and biology of young sandbar sharks, Carcharhinus plumbeus (Pisces, Carcharhinidae), in Chincoteague Bay, Virginia. Fishery bulletin United States, National Marine Fisheries Service, 79, 441-447.
76. Shibuya, A., Rosa, R. S., & Gadig, O. B. F. (2005). Stomach contents of Galeocerdo cuvier and Carcharhinus plumbeus (Elasmobranchii: Carcharhinidae) caught of Paraíba State, Brazil. Arquivos de Ciências do Mar, 38, 105-107.
77. Lessa, R., & Almeida, Z. (1997). Analysis of stomach contents of the smalltail shark Carcharhinus porosus from northern Brazil. Cybium, 21(2), 123-133.
78. Darracott, A. (1977). Availability, morphometrics, feeding and breeding activity in a multi-species, demersal fish stock of the Western Indian Ocean. Journal of Fish Biology, 10(1), 1-16.
79. Vaske Júnior, T., Vooren, C. M., & Lessa, R. P. (2009a). Feeding strategy of the night shark (Carcharhinus signatus) and scalloped hammerhead shark (Sphyrna lewini) near seamounts off northeastern Brazil. Brazilian Journal of oceanography, 57(2), 97-104.
80. Stevens, J. D., & Wiley, P. D. (1986). Biology of two commercially important carcharhinid sharks from northern Australia. Marine and Freshwater Research, 37(6), 671-688.
81. Praderi, R. (1985). Relaciones entre Pontoporia blainvillei (Mammalia: Cetacea) y tiburones (Selachii) de aguas Uruguayas. Comunicaciones zool. del

Museo de historia natural de Montevideo, 11(151).
82. Smale, M. J. (2005). The diet of the ragged-tooth shark Carcharias taurus Rafinesque 1810 in the Eastern Cape, South Africa. African Journal of Marine Science, 27(1), 331-335.
83. Lucifora, L. O., García, V. B., & Escalante, A. H. (2009b). How can the feeding habits of the sand tiger shark influence the success of conservation programs?. Animal Conservation, 12(4), 291-301.
84. Cliff, G., Dudley, S. F. J., & Davis, B. (1989). Sharks caught in the protective gill nets off Natal, South Africa. 2. The great white shark Carcharodon carcharias (Linnaeus). South African Journal of Marine Science, 8(1), 131-144.
85. Casey, J. G., & Pratt Jr, H. L. (1985). Distribution of the white shark, Carcharodon carcharias, in the western North Atlantic. Memoirs of the Southern California Academy of Sciences, 9, 2-14.
86. Arnold, P. W. (1972). Predation on harbour porpoise, Phocoena phocoena, by a white shark, Carcharodon carcharias. Journal of the Fisheries Board of Canada, 29(8), 1213-1214.
87. Shaughnessy, P. D., Dennis, T. E., & Berris, M. (2007). Predation on Australian sea lions Neophoca cinerea by white sharks Carcharodon carcharias in South Australia. Australian Mammalogy, 29, 69-75.
88. LeMier, E. H. (1951). Recent records of the great white shark, Carcharodon carcharias, on the Washington Coast. Copeia, 1951(3), 249-249.
89. Tricas, T. C., & McCosker, J. E. (1984). Predatory behavior of the white shark (Carcharodon carcharias), with notes on its biology. Proceedings of the California Academy of Sciences, 43(14), 221-238.
90. Bruce, B. D. (1992). Preliminary observations on the biology of the white shark, Carcharodon carcharias, in south Australian waters. Marine and Freshwater Research, 43(1), 1-11.
91. Hussey, N. E., McCann, H. M., Cliff, G., Dudley, S. F., Wintner, S. P., & Fisk, A. T. (2012). Size-based analysis of diet and trophic position of the white shark (Carcharodon carcharias) in South African waters. In: Global Perspectives on the Biology and Life History of the White Shark, pp. 27-49. CRC Press.
92. Martin, R. A. (2004). Northerly distribution of white sharks, Carcharodon carcharias, in the eastern Pacific and relation to ENSO events. Marine Fisheries Review, 66(1), 16-26.
93. Stewart, B. S., & Yochem, P. K. (1985). Radio-tagged harbor seal, Phoca vitulina richardsi, eaten by white shark, Carcharodon carcharias in the Southern California Bight. California Fish and Game, 71(2), 113-115.
94. Klimley, A. P. (1985). The areal distribution and autoecology of the white shark, Carcharodon carcharias, off the west coast of North America. Memoirs of the Southern California Academy of Sciences, 9, 15-40.
95. Scholl, J. P. (1983). Skull fragments of the California sea lion (Zalophus californianus) in stomach of a white shark (Carcharodon carcharias). Journal of Mammalogy, 64(2), 332-332.
96. Santana-Morales, O., Sosa-Nishizaki, O., Escobedo-Olvera, M. A., Oñate-González, E. C., O’Sullivan, J. B., & Cartamil, D. (2012). Incidental catch and ecological observations of juvenile white sharks, Carcharodon carcharias, in western Baja California, Mexico. In Global Perspectives on the Biology and Life History of the White Shark, pp. 187-198. CRC Press.

97. LeBoeuf, B. J., Riedman, M., & Keyes, R. S. (1982). White shark predation on pinnipeds in California coastal waters. Fishery Bulletin, 80(4), 891-895.
98. Kabasakal, H. (2008). Two recent records of the great white sharks, Carcharodon carcharias(Linnaeus, 1758)(Chondrichthyes: Lamnidae), caught in turkish waters. In Annales. Anali za istrske in mediteranske studije. Series historia naturalis, 18, 11-16.
99. Capapé, C. (1985). Nouvelle description de Centrophorus granulosus (Schneider, 1801) (Pisces, Squalidae). Données sur la biologie de la reproduction et le régime alimentaire des spécimens des côtes tunisiennes. Bulletin de l'Institut national scientifique et technique d'océanographie et de pêche, 12, 97-141
100. Megalofonou, P., & Chatzispyrou, A. (2006). Sexual maturity and feeding of the gulper shark, Centrophorus granulosus, from the eastern Mediterranean Sea. Cybium, 30(4), 67-74.
101. Dunn, M. R., Szabo, A., McVeagh, M. S., & Smith, P. J. (2010). The diet of deepwater sharks and the benefits of using DNA identification of prey. Deep Sea Research Part I: Oceanographic Research Papers, 57(7), 923-930.
102. Mauchline, J., & Gordon, J. D. M. (1983). Diets of the sharks and chimaeroids of the Rockall Trough, northeastern Atlantic Ocean. Marine Biology, 75(2-3), 269-278.
103. Ebert, D. A., Compagno, L. J. V., & Cowley, P. D. (1992). A preliminary investigation of the feeding ecology of squaloid sharks off the west coast of southern Africa. South African Journal of Marine Science, 12(1), 601-609.
104. Hareide, N. R., Garnes, G., Langedal, G., & Clarke, M. (2006). Aspects of the biology of two deepwater sharks Centrophorus squamosus and Centroscymnus coelolepis from hatton bank and the mid-Atlantic Ridge. In FAO Fisheries Proceedings (FAO). FAO.
105. Carrassón, M., Stefanescu, C., & Cartes, J. E. (1992). Diets and bathymetric distributions of two bathyal sharks of the Catalan deep sea (western Mediterranean). Marine ecology progress series. Oldendorf, 82(1), 21-30.
106. Horn, P. L. (2016). Biology of the New Zealand carpet shark Cephaloscyllium isabellum. Journal of Ichthyology, 56(3), 336-347.
107. Soto, J. M. R. (2000). Sobre a presença do tubarão-peregrino, Cetorhinus maximus (Gunnerus, 1765)(Lamniformes, Cetorhinidae), em águas brasileiras. Biotemas, 13(1), 73-88.
108. Mutoh, M., & Omori, M. (1978). Two records of patchy occurrences of the oceanic shrimp Sergestes similis Hansen off the east coast of Honshu, Japan. Journal of Oceanography, 34(1), 36-38.
109. Loas, V. (1958). Plancton et squales. Revue des Travaux de l'Institut des Pêches Maritimes, 22(3), 333-335.
110. Kubota, T., Shiobara, Y. and Kubodera, T. (1991). Food habits of the frilled shark Chlamydoselachus anguineus collected from Suruga Bay, central Japan. *日本水産学会誌*, 57(1), 15-20.
111. Matallanas, J. (1982). Feeding habits of Scymnorhinus licha in Catalan waters. Journal of Fish Biology, 20(2), 155-163.
112. Kabasakal, H., & Kabasakal, E. (2002). Morphometrics of young kitefin sharks, Dalatias licha (Bonnaterre, 1788), from Northeastern Aegean Sea, with notes on its biology. In Annales (Annals for istran and mediterranean studies), Series historia naturalis, 12(2), 161-166.
113. Navarro, J., López, L., Coll, M., Barría, C., & Sáez-Liante, R. (2014). Short-and long-term importance of small sharks in the diet of the rare deep-sea shark Dalatias licha. Marine biology, 161(7), 1697-1707.
114. Capapé, C., Hemida, F., Quignard, J., Ben Amor, M., & Reynaud, C. (2008). Biological observations on a rare deep-sea shark, Dalatias licha

(Chondrichthyes: Dalatiidae), off the Maghreb coast (south-western Mediterranean). Pan-American Journal of Aquatic Sciences, 3(3), 355-360.

115. Silva, H. M. (1983). Preliminary studies of the exploited stock of kitefin shark Scymnorhinus licha (Bonnaterre, 1788) in the Azores. International Council for the Exploration of the Sea, 1983/G:18, 13
116. Dunn, M. R., Stevens, D. W., Forman, J. S., & Connell, A. (2013). Trophic interactions and distribution of some squaliforme sharks, including new diet descriptions for Deania calcea and Squalus acanthias. PloS one, 8(3), e59938.
117. Preciado, I., Cartes, J. E., Serrano, A., Velasco, F., Olaso, I., Sánchez, F., & Frutos, I. (2009). Resource utilization by deep-sea sharks at the Le Danois Bank, Cantabrian Sea, north-east Atlantic Ocean. Journal of fish biology, 75(6), 1331-1355.
118. Blaber, S. J. M., & Bulman, C. M. (1987). Diets of fishes of the upper continental slope of eastern Tasmania: content, calorific values, dietary overlap and trophic relationships. Marine Biology, 95(3), 345-356.
119. Kazunari, Y. (1991). Catch distribution, stomach contents and size at maturity of two squaloid sharks, Deania calceus and D. crepidalbus, from the southeast Atlantic off Namibia. Bulletin of the Japanese Society of Fisheries Oceanography, 55(3), 189-196.
120. Kobayashi, H. (1986). Studies on deep-sea sharks in Kumano-nada region. Bulletin of the Faculty of Fisheries, Mie University, 13, 25-133.
121. Cherel, Y., & Duhamel, G. (2004). Antarctic jaws: cephalopod prey of sharks in Kerguelen waters. Deep Sea Research Part I: Oceanographic Research Papers, 51(1), 17-31.
122. Baba, O., Taniuchi, T., & Nose, Y. (1987). Depth distribution and food habits of three species of small squaloid sharks [Etmopterus lucifer, E. unicolor and Centroscyllium ritteri] off Choshi [Japan]. Bulletin of the Japanese Society of Scientific Fisheries (Japan), 53(3), 417-424.
123. Ozawa, T., & Zinno, H. (1990). Studies on the bottom fishes of continental slope off Makurazaki, southern Japan, 2: Stomach content analysis. Bulletin of the Japanese Society of Fisheries Oceanography (Japan), 54(3), 255-270.
124. Jakobsdóttir, K. B. (2001). Biological aspects of two deep-water squalid sharks: Centroscyllium fabricii (Reinhardt, 1825) and Etmopterus princeps (Collett, 1904) in Icelandic waters. Fisheries Research, 51(2), 247-265.
125. Xavier, J. C., Vieira, C., Assis, C., Cherel, Y., Hill, S., Costa, E., ... & Coelho, R. (2012). Feeding ecology of the deep-sea lanternshark Etmopterus pusillus (Elasmobranchii: Etmopteridae) in the northeast Atlantic. Scientia Marina, 76(2), 301-310.
126. Capapé, C., Guélorget, O., Reynaud, C., Marquès, A., Bouchereau, J. L., & Zaouali, J. (2003). Effects of reproductive factors on interrelationships among three deep water sharks from northern Tunisia (Central Mediterranean). In Annales, series Historia Naturalis, 13(2), 109-120.
127. Isbert, W., Rodríguez-Cabello, C., Frutos, I., Preciado, I., Montero, F. E., & Pérez-del-Olmo, A. (2015). Metazoan parasite communities and diet of the velvet belly lantern shark Etmopterus spinax (Squaliformes: Etmopteridae): a comparison of two deep-sea ecosystems. Journal of fish biology, 86(2), 687-706.
128. Renwart, M., & Mallefet, J. (2013). First study of the chemistry of the luminous system in a deep-sea shark, Etmopterus spinax Linnaeus, 1758 (Chondrichthyes: Etmopteridae). Journal of Experimental Marine Biology and Ecology, 448, 214-219.
129. Valls, M., Quetglas, A., Ordines, F., & Moranta, J. (2011). Feeding ecology of demersal elasmobranchs from the shelf and slope off the Balearic Sea (western Mediterranean). Scientia Marina, 75(4), 633-639.
130. Fanelli, E., Rey, J., Torres, P., & Gil de Sola, L. (2009). Feeding habits of blackmouth catshark Galeus melastomus Rafinesque, 1810 and velvet belly

lantern shark Etmopterus spinax (Linnaeus, 1758) in the western Mediterranean. Journal of Applied Ichthyology, 25(s1), 83-93.

131.Bergstad, O. A., Wik, A. D., & Hildre, O. (2003). Predator-prey relationships and food sources of the Skagerrak deep-water fish assemblage. Journal of Northwest Atlantic Fishery Science, 31, 165.

132. Klimpel, S., Palm, H. W., & Seehagen, A. (2003). Metazoan parasites and food composition of juvenile Etmopterus spinax (L., 1758)(Dalatiidae, Squaliformes) from the Norwegian Deep. Parasitology Research, 89(4), 245-251.

133. Bello, G. (1998). The feeding ecology of the velvet belly, Etmopterus spinax (Chondrichthyes: Squalidae), of the Adriatic Sea on the basis of its stomach contents. Atti della Società italiana di scienze naturali e del museo civico di storia naturale di Milano, 139, 187-193.

134. Barry, J. P., Yoklavich, M. M., Cailliet, G. M., Ambrose, D. A., & Antrim, B. S. (1996). Trophic ecology of the dominant fishes in Elkhorn Slough, California, 1974–1980. Estuaries and Coasts, 19(1), 115-138.

135. Ebert, D. A., & Ebert, T. B. (2005). Reproduction, diet and habitat use of leopard sharks, Triakis semifasciata (Girard), in Humboldt Bay, California, USA. Marine and freshwater research, 56(8), 1089-1098

136. Hubbs, C. L., Iwai, T., & Matsubara, K. (1967). External and internal characters, horizontal and vertical distributions, luminescence, and food of the dwarf pelagic shark, Euprotomicrus bispinatus. Scripps Institution of Oceanography.

137. Schwartz, F. J. (2000). Food of tiger sharks, Galeocerdo cuvier (Carcharhinidae) from the northwest Atlantic Ocean, off North Carolina. Journal of the Elisha Mitchell Scientific Society. 116 (4) 351-355.

138. Lowe, C. G., Wetherbee, B. M., Crow, G. L., & Tester, A. L. (1996). Ontogenetic dietary shifts and feeding behavior of the tiger shark, Galeocerdo cuvier, in Hawaiian waters. Environmental Biology of Fishes, 47(2), 203-211.

139. Briones-Fourzan, P., & Lozano-Alvarez, E. (1991). Aspects of the biology of the giant isopod Bathynomus giganteus A. Milne Edwards, 1879 (Flabellifera: Cirolanidae), off the Yucatan Peninsula. Journal of Crustacean Biology, 11(3), 375-385.

140. Bornatowski, H., Robert, M. C., & Costa, L. (2007a). Dados sobre a alimentação de jovens de tubarão-tigre, Galeocerdo cuvier (Péron & Lesueur) (Elasmobranchii, Carcharhinidae), do sul do Brasil. Pan-American Journal of Aquatic Sciences, 2(3), 10-13.

141. Yosef, R., Zakai, D., Rydberg-Hedaen, M., & Nikolajsen, R. (2002). An unusual record of a European Bee-eater Merops apiaster from Eilat-inside a Tiger Shark Galeocerdo cuvier. Sandgrouse, 24(2), 141-141

142. Carlson, J. K., Grace, M. A., & Lago, P. K. (2002). An observation of juvenile tiger sharks feeding on clapper rails off the southeastern coast of the United States. Southeastern Naturalist, 1(3), 307-310.

143. Heithaus, M. R. (2001). The biology of tiger sharks, Galeocerdo cuvier, in Shark Bay, Western Australia: sex ratio, size distribution, diet, and seasonal changes in catch rates. Environmental Biology of Fishes, 61(1), 25-36.

144. Di Beneditto, A. P. M. (2004). Presence of franciscana dolphin (Pontoporia blainvillei) remains in the stomach of a tiger shark (Galeocerdo cuvieri) captured in Southeastern Brazil. Aquatic Mammals, 30(2), 311-314.

145. Simpfendorfer, C. A., Goodreid, A. B., & McAuley, R. B. (2001b). Size, sex and geographic variation in the diet of the tiger shark, Galeocerdo cuvier, from Western Australian waters. Environmental Biology of Fishes, 61(1), 37-46.

146. Rathbun, T. A., & Rathbun, B. C. (1984). Human remains recovered from a shark's stomach in South Carolina. Journal of Forensic Science, 29(1), 269-

276.
147. Randall, J. E. (1992). Review of the biology of the tiger shark (Galeocerdo cuvier). Marine and Freshwater Research, 43(1), 21-31.
148. Simpfendorfer, C. (1992). Biology of tiger sharks (Galeocerdo cuvier) caught by the Queensland shark meshing program off Townsville, Australia. Marine and Freshwater Research, 43(1), 33-43.
149. Small, P. 1991. Caretta-caretta loggerhead predation. Herpetological review, 22(3), 97.
150. Dodrill, J. W., & Gilmore, R. G. (1978). Land birds in the stomachs of tiger sharks Galeocerdo cuvieri (Peron and Lesueur). The Auk, 1978, 585-586
151. Heatwole, H., Heatwole, E., & Johnson, C. R. (1974). Shark predation on sea snakes. Copeia, 1974(3), 780-781.
152. Brooke, R.K. (1976). Shark predation on seabirds in Natal waters. Ostrich, 47(2-3), 126.
153. Gudger, E. W. (1949). Natural history notes on tiger sharks, Galeocerdo tigrinus, caught at Key West, Florida, with emphasis on food and feeding habits. Copeia, 1949(1), 39-47.
154. Gudger, E. W. (1948). The tiger shark, Galeocerdo tigrinus, on the North Carolina coast and its food and feeding habits there. Journal of the Elisha Mitchell Scientific Society, 64(2), 221-233.
155. Chidambaram, K. (1946). Note on the food of tiger sharks (Galeocerdo spp.) of the Madras coasts. Journal of the Bombay Natural History Society, 45(1), 247.
156. Sarangdhar, P. N. (1943). Tiger shark-Galeocerdo tigrinus Muller and Henle. Feeding and breeding habits. Journal of the Bombay Natural History Society, 44(1), 102-110.
157. Rosas, F. C. W., Capistrano, L. C., Di Beneditto, A. P., & Ramos, R. (1992). Hydrurga leptonyx recovered from the stomach of a tiger shark captured off the Rio de Janeiro coast, Brazil. Mammalia, 56(1), 153-155.
158. Lucifora, L. O., García, V. B., Menni, R. C., & Escalante, A. H. (2006). Food habits, selectivity, and foraging modes of the school shark Galeorhinus galeus. Marine Ecology Progress Series, 315, 259-270.
159. Ripley, W. E. (1946). The soupfin shark and the fishery. Fishery Bulletin, 64, 7-37.
160. Reyes, P. R. (2005). Preliminaries antecedents on the feeding of the tope shark Galeorhinus galeus (Linnaeus, 1758) (Carcharhiniformes: Triakidae) in Central Southern Chile. Revista de Biologia Marina y Oceanografia, 40(1), 83.
161. Morato, T., Sola, E., Gros, M. P., & Menezes, G. (2003). Diets of thornback ray (Raja clavata) and tope shark (Galeorhinus galeus) in the bottom longline fishery of the Azores, northeastern Atlantic. Fishery Bulletin, 101(3), 590-602.
162. Brodeur, R. D., Lorz, H. V., & Pearcy, W. G. (1987). Food habits and dietary variability of pelagic nekton off Oregon and Washington, 1979-1984. NOAA Technical Report NMFS, 57, 1-32.
163. Ellis, J. R., Pawson, M. G., & Shackley, S. E. (1996). The comparative feeding ecology of six species of shark and four species of ray (Elasmobranchii) in the north-east Atlantic. Journal of the Marine Biological Association of the United Kingdom, 76(1), 89-106.
164. Olsen, A. M. (1984). Synopsis of biological data on the school shark, Galeorhinus australis (Macleay 1881). FAO Fisheries Synopsis 139. FAO Fisheries, Rome
165. dos Santos, R. A., & Haimovici, M. (2002). Cephalopods in the trophic relations off southern Brazil. Bulletin of Marine Science, 71(2), 753-770.

166. Anastasopoulou, A., Mytilineou, C., Lefkaditou, E., Dokos, J., Smith, C. J., Siapatis, A. & Papadopoulou, K. N. (2013). Diet and feeding strategy of blackmouth catshark Galeus melastomus. Journal of fish biology, 83(6), 1637-1655.
167. Olaso, I., Velasco, F., Sánchez, F., Serrano, A., Rodríguez-Cabello, C., & Cendrero, O. (2005). Trophic relations of lesser-spotted catshark (Scyliorhinus canicula) and blackmouth catshark (Galeus melastomus) in the Cantabrian Sea. Journal of Northwest Atlantic Fishery Science, 35, 481-494.
168. Kabasakal, H. (2002a). Cephalopods in the stomach contents of four Elasmobranch species from the northern Aegean Sea. Acta Adriatica, 43(1), 17-24.
169. Mattson, S. (1981). The food of Galeus melastomus, Gadiculus argenteus thori, Trisopterus esmarkii, Rhinonemus cimbrius, and Glyptocephalus cynoglossus (Pisces) caught during the day with shrimp trawl in a West-Norwegian fjord. Sarsia, 66(2), 109-127.
170. Neves, A., Figueiredo, I., Moura, T., Assis, C., & Gordo, L. S. (2007). Diet and feeding strategy of Galeus melastomus in the continental slope off southern Portugal. Vie et milieu, 57(3), 165-169.
171. Bello, G. (1995). Cephalopods in the stomach contents of Galeus melastomus (Selachii, Scyliorhinidae) from the Adriatic Sea. Atti della Società italiana di scienze naturali e del museo civico di storia naturale di Milano, 134(1), 33-40.
172. Velasco, F., Olaso, I., & Sánchez, F. (2001). The role of cephalopods as forage for the demersal fish community in the southern Bay of Biscay. Fisheries Research, 52(1), 65-77.
173. Castro, J. I. (2000). The biology of the nurse shark, Ginglymostoma cirratum, off the Florida east coast and the Bahama Islands. Environmental Biology of Fishes, 58(1), 1-22.
174. Heupel, M. R., & Bennett, M. B. (1998). Observations on the diet and feeding habits of the epaulette shark, Hemiscyllium ocellatum (Bonnaterre), on Heron Island Reef, Great Barrier Reef, Australia. Marine and Freshwater Research, 49(7), 753-756.
175. Kamura, S., & Hashimoto, H. (2004). The food habits of four species of triakid sharks, Triakis scyllium, Hemitriakis japanica, Mustelus griseus and Mustelus manazo, in the central Seto Inland Sea, Japan. Fisheries science, 70(6), 1019-1035.
176. Braccini, J. M. (2008). Feeding ecology of two high-order predators from south-eastern Australia: the coastal broadnose and the deepwater sharpnose sevengill sharks. Marine Ecology Progress Series, 371, 273-284.
177. Henderson, A. C., & Williams, R. S. (2001). A new record of the sharpnose seven-gill shark Heptranchias perlo, from the north-east Atlantic. Marine Biological Association of the United Kingdom. Journal of the Marine Biological Association of the United Kingdom, 81(4), 707.
178. Segura Zarzosa, J. C., Galván Magaña, F., & Abitia Cárdenas, L. A. (1997). Observaciones sobre la alimentación del tiburón Heterodontus francisci Girard 1854 (Chondrichthyes: Heterodontidae), en Laguna de San Ignacio, Baja California Sur, México. Ciencias Marinas, 23(1).
179. Cruz-Escalona, V. H., Campos-Dávila, L., & Zetina-Rejón, M. J. (2010). Repartición de recursos alimentarios entre la ictiofauna dominante de una laguna templada de Baja California Sur, México. CICIMAR Oceánides, 25(1), 1-15.
180. Ebert, D. A. (1994). Diet of the sixgill shark Hexanchus griseus off southern Africa. South African Journal of Marine Science, 14(1), 213-218.
181. Kabasakal, H. (2006). Distribution and biology of the bluntnose sixgill shark, Hexanchus griseus (Bonnaterre, 1788)(Chondrichthyes: Hexanchidae), from Turkish waters. In Annales Series Historia Naturalis, 16(1), 29.
182. Kabasakal, H. (2004). Preliminary observations on the reproductive biology and diet of the Bluntnose sixgill shark, Hexanchus griseus (Bonnaterre, 1788)(Chondrichthyes: Hexanchidae), in Turkish Seas. Acta adriatica, 45(2), 187-196.

183. Soto, J. (2001a). Deep-sea fishes in the stomach contents of the bluntnose sixgill shark Hexanchus griseus (Bonnaterre, 1788)(Chondrichthyes, Hexanchidae), caught off southern Brazilian coast. Mare Magnum, 1(2), 171.
184. Ebert, D. A. (1986a). Biological aspects of the sixgill shark, Hexanchus griseus. Copeia, 131-135.
185. Waller, G. N. H., & Baranes, A. (1994). Food of Iago omanensis, a deep water shark from the northern Red Sea. Journal of fish biology, 45(1), 37-45.
186. Nair, R. V., & Appukkuttan, K. K. (1973). observation on the food of deep sea sharks halaelvrus hispidus (alcock), eridacnis radcliffei smith and Iago omanensis compagno and springer. Indian Journal of Fisheries, 20(2), 575-583.
187. Tunçer, S., & Kabasakal, H. (2016). Capture of a juvenile shortfin mako shark, isurus oxyrinchus rafinesque, 1810 (chondrichthyes: lamnidae) in the bay of Edremit, Northern Aegean Sea (Turkey). Annales Series Historia Naturalis, 26, 1.
188. Lyons, K., Preti, A., Madigan, D. J., Wells, R. J. D., Blasius, M. E., Snodgrass, O. E., ... & MacKenzie, K. (2015). Insights into the life history and ecology of a large shortfin mako shark Isurus oxyrinchus captured in southern California. Journal of fish biology, 87(1), 200-211.
189. Porsmoguer, S. B., Banaru, D., Boudouresque, C. F., Dekeyser, I., Viricel, A., & Merchán, M. (2015). DNA evidence of the consumption of short-beaked common dolphin Delphinus delphis by the shortfin mako shark Isurus oxyrinchus. Marine Ecology Progress Series, 532, 177-183.
190. Porsmoguer, S. B., Bănaru, D., Bearez, P., Dekeyser, I., Fornelino, M. M., & Boudouresque, C. F. (2014). Unexpected headless and tailless fish in the stomach content of shortfin mako Isurus oxyrinchus. PloS one, 9(2), e88488.
191. Groeneveld, J. C., Cliff, G., Dudley, S. F. J., Foulis, A. J., Santos, J., & Wintner, S. P. (2014). Population structure and biology of shortfin mako, Isurus oxyrinchus, in the south-west Indian Ocean. Marine and Freshwater Research, 65(12), 1045-1058.
192. Young, J. W., Lansdell, M. J., Campbell, R. A., Cooper, S. P., Juanes, F., & Guest, M. A. (2010). Feeding ecology and niche segregation in oceanic top predators off eastern Australia. Marine Biology, 157(11), 2347-2368.
193. Lopez, S., Meléndez, R., & Barría, P. (2009). Feeding of the shortfin mako shark Isurus oxyrinchus Rafinesque, 1810 (Lamniformes: Lamnidae) in the Southeastern Pacific. Revista de biología marina y oceanografía, 44(2), 439-451.
194. Wood, A. D., Wetherbee, B. M., Juanes, F., Kohler, N. E., & Wilga, C. (2009). Recalculated diet and daily ration of the shortfin mako (Isurus oxyrinchus), with a focus on quantifying predation on bluefish (Pomatomus saltatrix) in the northwest Atlantic Ocean. Fishery Bulletin, 107(1), 76-88.
195. Mucientes-Sandoval, G. R., & Saborido-Rey, F. (2008). Acercamiento a la composición de la dieta de Isurus oxyrinchus Rafinesque, 1810 (Elasmobranchii: Lamnidae) en aguas internacionales del Pacífico sur central. Rev. Invest. Mar, 29(2), 145-150.
196. Monteiro, M. S., Júnior, T. V., Barbosa, T. M., Danise, M., & Alves, O. (2006). Predation by a shortfin mako shark, Isurus oxyrinchus, Rafinesque, 1810, on a pantropical spotted dolphin, Stenella attenuata, calf in central Atlantic waters. LAJAM 5(2): 141-144.
197. Maia, A., Queiroz, N., Correia, J. P., & Cabral, H. (2006). Food habits of the shortfin mako, Isurus oxyrinchus, off the southwest coast of Portugal. Environmental Biology of Fishes, 77(2), 157-167.
198. Sepulveda, C. A., Kohin, S., Chan, C., Vetter, R., & Graham, J. B. (2004). Movement patterns, depth preferences, and stomach temperatures of free-swimming juvenile mako sharks, Isurus oxyrinchus, in the Southern California Bight. Marine Biology, 145(1), 191-199.
199. Vaske-Júnior, T. & Rincón-Filho, G (1998). Stomach content of blue sharks (Prionace glauca) and anequim (Isurus oxyrinchus) from oceanic waters of

southern Brazil. Revista Brasileira de Biologia, 58(3), 445-452.

200. Yatsu, A. (1995). The role of slender tuna, Allothunnus fallai, in the pelagic ecosystems of the South Pacific Ocean. Japanese Journal of Ichthyology, 41(4), 367-377.
201. Cliff, G., Dudley, S. F. J., & Davis, B. (1990). Sharks caught in the protective gill nets off Natal, South Africa. 3. The shortfin mako shark Isurus oxyrinchus (Rafinesque). South African Journal of Marine Science, 9(1), 115-126.
202. Stillwell, C. E., & Kohler, N. E. (1982). Food, feeding habits, and estimates of daily ration of the shortfin mako (Isurus oxyrinchus) in the northwest Atlantic. Canadian Journal of Fisheries and Aquatic Sciences, 39(3), 407-414.
203. Logan, J. M., Toppin, R., Smith, S., Galuardi, B., Porter, J., & Lutcavage, M. (2013). Contribution of cephalopod prey to the diet of large pelagic fish predators in the central North Atlantic Ocean. Deep Sea Research Part II: Topical Studies in Oceanography, 95, 74-82.
204. Joyce, W. N., Campana, S. E., Natanson, L. J., Kohler, N. E., Pratt, H. L., & Jensen, C. F. (2002). Analysis of stomach contents of the porbeagle shark (Lamna nasus Bonnaterre) in the northwest Atlantic. ICES Journal of Marine Science: Journal du Conseil, 59(6), 1263-1269.
205. van der Vyver, J. S. F., De Bruyn, P. J. N., Lipinski, M. R., & Leslie, R. W. (2015). First record of a porbeagle shark Lamna nasus (Bonnaterre, 1788) stranding at sub-Antarctic Marion Island. Marine Biodiversity Records, 8, e67.
206. Francis, M. P., Campana, S. E., & Jones, C. M. (2007). Age under-estimation in New Zealand porbeagle sharks (Lamna nasus): is there an upper limit to ages that can be determined from shark vertebrae?. Marine and Freshwater Research, 58(1), 10-23.
207. Henderson, A. C., Flannery, K., & Dunne, J. (2003). Biological observations on shark species taken in commercial fisheries to the west of Ireland. In Biology and Environment: Proceedings of the Royal Irish Academy (pp. 1-7). Royal Irish Academy.
208. Ellis, J. R., & Shackley, S. E. (1995). Notes on porbeagle sharks, Lamna nasus, from the Bristol Channel. Journal of Fish Biology, 46(2), 368-370.
209. Gauld, J. A. (1989). Records of porbeagles landed in Scotland with observations on the biology, distribution and exploitation of the species. In Scottish Fisheries Research Report, 45, 1-15.
210. Stevens, J. D., Dunning, M. C., & Machida, S. (1983). Occurrence of the porbeagle shark, Lamna nasus, in the Tasman Sea. Japanese Journal of Ichthyology, 30(3), 301-307.
211. Scattergood, L. W. (1949). Notes on the Maine Shark fishery. Copeia, 1949(1), 69-71.
212. Gutteridge, A. N., Bennett, M. B., Huveneers, C., & Tibbetts, I. R. (2011). Assessing the overlap between the diet of a coastal shark and the surrounding prey communities in a sub-tropical embayment. Journal of Fish Biology, 78(5), 1405-1422.
213. Jabado, R. W., Al Ghais, S. M., Hamza, W., Henderson, A. C., & Al Mesafri, A. A. (2015). Diet of two commercially important shark species in the United Arab Emirates: milk shark, Rhizoprionodon acutus (Rüppell, 1837), and slit-eye shark, Loxodon macrorhinus (Müller & Henle, 1839). Journal of Applied Ichthyology, 31(5), 870-875.
214. Sawamoto, S., & Matsumoto, R. (2012). Stomach contents of a megamouth shark Megachasma pelagios from the Kuroshio Extension: evidence for feeding on a euphausiid swarm. Plankton and Benthos Research, 7(4), 203-206.
215. Yano, K., Yabumoto, Y., Tanaka, S., Tsukada, O., & Furuta, M. (1997). Capture of a mature female megamouth shark, Megachasma pelagios, from Mie, Japan. In Proceedings of the 5th Indo-Pacific Conference, Nouméa (pp. 335-349).

216. Taylor, L. R., Compagno, L. J., & Struhsaker, P. J. (1983). Megamouth--a new species, genus, and family of lamnoid shark (Megachasma pelagios, family Megachasmidae) from the Hawaiian Islands. Proceedings of the California Academy of Sciences, 43(8), 87-110.
217. Talent, L. G. (1982). Food habits of the gray smoothhound, Mustelus californicus, the brown smoothhound, Mustelus henlei, the shovelnose guitarfish, Rhinobatos productus, and the bat ray, Myliobatis californica, in Elkhorn Slough, California. California fish and game, 68(4), 224-234.
218. Taylor, D. L., Kutil, N. J., Malek, A. J., & Collie, J. S. (2014). Mercury bioaccumulation in cartilaginous fishes from Southern New England coastal waters: contamination from a trophic ecology and human health perspective. Marine environmental research, 99, 20-33.
219. Montemarano, J. J., Havelin, J., & Draud, M. (2016). Diet composition of the smooth dogfish (Mustelus canis) in the waters of Long Island, New York, USA. Marine Biology Research, 12(4), 435-442.
220. Woodland, R. J., Secor, D. H., & Wedge, M. E. (2011). Trophic resource overlap between small elasmobranchs and sympatric teleosts in Mid-Atlantic Bight nearshore habitats. Estuaries and Coasts, 34(2), 391-404.
221. Link, J. S., Garrison, L. P., & Almeida, F. P. (2002). Ecological interactions between elasmobranchs and groundfish species on the northeastern US continental shelf. I. Evaluating predation. North American Journal of Fisheries Management, 22(2), 550-562.
222. Gaitan-Espitia, J. D. (2008). The diet of neonates of the smooth dogfish (Mustelus canis) during its in the breeding area, ecorregion Palomino, Colombian Caribbean. Dieta de neonatos de tiburon toyo (Mustelus canis) durante su permanencia en area de crianza, ecorregion Palomino, Caribe colombiano. III National Symposium on Sharks and Rays. 25-29 August 2008, Faculty of Science, UNAM, Ciudad Unversitaria, Mexico D.F. , p.220-223.
223. Vianna, M., Arfelli, C. A., & Amorim, A. F. (2000). Feeding of Mustelus canis (Elasmobranchii, Triakidae) caught off south-southeast coast of Brazil. Boletim do Instituto de Pesca, 26(1), 79-84.
224. Rountree, R. A., & Able, K. W. (1996). Seasonal abundance, growth, and foraging habits of juvenile smooth dogfish, Mustelus canis, in a New Jersey estuary. Fishery Bulletin, 94(3), 522-534.
225. Soto, J. M. (2001b). Distribution and reproductive biology of the striped smooth-hound Mustelus fasciatus (Garman, 1913) (Carcharhiniformes, Triakidae). Mare Magnum, 1(2), 129-34.
226. Rodríguez-Romero, J., Álvarez-Bauman, E., Ochoa-Díaz, M. R., López-Martínez, J., & Maldonado-García, M. (2013). Feeding habits of Mustelus henlei on the western coast of Baja California Sur, México. Rev. biol. mar. oceanogr, 48(2), 261-271.
227. Espinoza, M., Clarke, T. M., Villalobos-Rojas, F., & Wehrtmann, I. S. (2012). Ontogenetic dietary shifts and feeding ecology of the rasptail skate Raja velezi and the brown smoothhound shark Mustelus henlei along the Pacific coast of Costa Rica, Central America. Journal of fish biology, 81(5), 1578-1595.
228. Gómez, G. O., Zapata, L. A., Franke, A., & Ramos, G. E. (2003). Food habits of Mustelus Lunulatus and M. henlei (Pisces: Triakidae) colected in Parque Nacional Natural Gorgona, Colombian Pacific. Boletín de Investigaciones Marinas y Costeras-INVEMAR, 32(1), 219-229.
229. Haeseker, S. L., & Cech Jr, J. J. (1993). Food habits of the brown smoothhound shark (Mustelus henlei) from two sites in Tomales Bay. California Fish and Game, 79(3), 89-95.

230. Russo, R. A. (1975). observations on food-habits of leopard sharks (Triakis semifasciata) and brown smoothhounds (Mustelus henlei). california fish and game, 61(2).
231. Escobar-Sánchez, O., Abitia-Cardenas, L. A., & Galvan-Magaña, F. (2006). Food habits of the Pacific angel shark Squatina californica in the southern Gulf of California, Mexico. Cybium, 30(4 SUPPL.), 91-97.
232. King, K. J., & Clark, M. R. (1984). The food of rig (Mustelus lenticulatus) and the relationship of feeding to reproduction and condition in Golden Bay. New Zealand journal of marine and freshwater research, 18(1), 29-42.
233. Moreno-Sanchez, X. G., Escobar-Sánchez, O., Abitia-Cardenas, L. A., & Cruz-Escalona, V. H. (2012). Diet composition of the sicklefin smooth-hound shark Mustelus lunulatus caught off El Pardito Island, Baja California Sur, Mexico. Marine Biodiversity Records, 5, e67.
234. Navia, A. F., Giraldo, A., & Mejía-Falla, P. A. (2006). Notas sobre la biología y dieta del toyo vieja (Mustelus lunulatus) en la zona central de pesca del Pacífico colombiano. Investigaciones marinas, 34(2), 217-222.
235. Galván-Magaña, F., Nienhuis, H. J., & Klimley, A. P. (1989). Seasonal abundance and feeding habits of sharks of the lower Gulf of California, Mexico. California Fish and Game, 75(2), 74-84.
236. Yamaguchi, A., & Taniuchi, T. (2000). Food variations and ontogenetic dietary shift of the starspotted-dogfish Mustelus manazo at five locations in Japan and Taiwan. Fisheries science, 66(6), 1039-1048.
237. Taniuchi, T., Kuroda, N., & Nose, Y. (1983). Age, growth, reproduction, and food-habits of the star-spotted dogfish mustelus-manazo collected from choshi. Bulletin of the Japanese Society of Scientific Fisheries, 49(9), 1325-1334.
238. Morte, S., Redon, M. J., & Sanz-Brau, A. (1997). Feeding habits of juvenile Mustelus mustelus (Carcharhiniformes, Triakidae) in the western Mediterranean. Cahiers de biologie marine, 38(2), 103-107.
239. Saidi, B., Enajjar, S., Bradai, M. N., & Bouain, A. (2009). Diet composition of smooth-hound shark, Mustelus mustelus (Linnaeus, 1758), in the Gulf of Gabès, southern Tunisia. Journal of Applied Ichthyology, 25(s1), 113-118.
240. Smale, M. J., & Compagno, L. J. V. (1997). Life history and diet of two southern African smoothhound sharks, Mustelus mustelus (Linnaeus, 1758) and Mustelus palumbes Smith, 1957 (Pisces: Triakidae). South African Journal of Marine Science, 18(1), 229-248.
241. Gracan, R., Mladineo, I., & Lazar, B. (2014). Insight into the diet composition and gastrointestinal parasite community of the common smooth-hound, Mustelus mustelus (Carcharhiniformes: Triakidae), in the northern Adriatic Sea. Natura Croatica, 23(1), 35.
242. Jardas, I., Šantić, M., Nerlović, V., & Pallaoro, A. (2007). Diet of the smooth-hound Mustelus mustelus (Chondrichthyes: Triakidae) in the eastern Adriatic Sea. Cybium, 31(4), 459-464.
243. Belleggia, M., Figueroa, D. E., Sánchez, F., & Bremec, C. (2012a). The feeding ecology of Mustelus schmitti in the southwestern Atlantic: geographic variations and dietary shifts. Environmental Biology of Fishes, 95(1), 99-114.
244. Bonelli, A. G., Giachetti, C. B., Jaureguizar, A. J., & Milessi, A. C. (2016). First report of predation by a small shark on the invasive rapa whelk Rapana venosa (Valenciennes, 1846) in Argentinean waters. BioInvasions Records, 5(3), 169–172.
245. Molina, J. M., & Cazorla, A. L. (2011). Trophic ecology of Mustelus schmitti (Springer, 1939) in a nursery area of northern Patagonia. Journal of sea research, 65(4), 381-389.

246. Van der Molen, S., & Caille, G. (2001). Bahía Engaño: a north Patagonian nursery area for the smoothhound shark Mustelus schmitti (Carcharhiniformes: Triakidae). Journal of the Marine Biological Association of the UK, 81(05), 851-855.
247. Chiaramonte, G. E., & Pettovello, A. D. (2000). The biology of Mustelus schmitti in southern Patagonia, Argentina. Journal of Fish Biology, 57(4), 930-942.
248. White, W. T., Platell, M. E., & Potter, I. C. (2004). Comparisons between the diets of four abundant species of elasmobranchs in a subtropical embayment: implications for resource partitioning. Marine Biology, 144(3), 439-448.
249. Newman, S. P., Handy, R. D., & Gruber, S. H. (2010). Diet and prey preference of juvenile lemon sharks Negaprion brevirostris. Marine Ecology Progress Series, 398, 221-234.
250. Cortés, E., & Gruber, S. H. (1990). Diet, feeding habits and estimates of daily ration of young lemon sharks, Negaprion brevirostris (Poey). Copeia, 204-218.
251. Schmidt, T. W. (1986). Food of young juvenile lemon sharks, Negaprion brevirostris (Poey), near Sandy Key, western Florida Bay. Florida Scientist, 49(1), 7-10.
252. Banner, A. (1968). Attraction of young lemon sharks, Negaprion brevirostris, by sound. Copeia, 1968(4), 871-872.
253. Banner, A. (1972). Use of sound in predation by young lemon sharks, Negaprion brevirostris (Poey). Bulletin of Marine Science, 22(2), 251-283.
254. Barnett, A., Abrantes, K., Stevens, J. D., Yick, J. L., Frusher, S. D., & Semmens, J. M. (2010a). Predator–prey relationships and foraging ecology of a marine apex predator with a wide temperate distribution. Marine Ecology Progress Series, 416, 189-200.
255. Barnett, A., Redd, K. S., Frusher, S. D., Stevens, J. D., & Semmens, J. M. (2010b). Non-lethal method to obtain stomach samples from a large marine predator and the use of DNA analysis to improve dietary information. Journal of experimental marine biology and ecology, 393(1), 188-192.
256. Lucifora, L. O., Menni, R. C., & Escalante, A. H. (2005). Reproduction, abundance and feeding habits of the broadnose sevengill shark Notorynchus cepedianus in north Patagonia, Argentina. Marine Ecology Progress Series, 289, 237-244.
257. Crespi-Abril, A. C., García, N. A., Crespo, E. A., & Coscarella, M. A. (2003). Consumption of marine mammals by broadnose sevengill shark Notorynchus cepedianus in the northern and central Patagonian shelf. Latin American Journal of Aquatic Mammals, 2(2), 101-107.
258. Ebert, D. A. (1991). Diet of the seven gill shark Notorynchus cepedianus in the temperate coastal waters of southern Africa. South African Journal of Marine Science, 11(1), 565-572.
259. Ebert, D. A. (1986b). Aspects on the biology of hexanchid sharks along the California coast. In Indo-Pacific Fish Biology: Proceedings of the Second International Conference on Indo-*Pacific Fishes’.(Eds T. Uye*no, R. Arai, T. Taniuchi and K. Matsuura.). pp. 437-449.
260. Markaida, U., & Sosa-Nishizaki, O. (2010). Food and feeding habits of the blue shark Prionace glauca caught off Ensenada, Baja California, Mexico, with a review on its feeding. Journal of the marine biological association of the United Kingdom, 90(05), 977-994.
261. Vaske Júnior, T., Lessa, R. P., & Gadig, O. B. F. (2009b). Feeding habits of the blue shark (Prionace glauca) off the coast of Brazil. Biota Neotropica, 9(3), 55-60
262. Hernández-Aguilar, S. B., Escobar-Sánchez, O., Galván-Magaña, F., & Abitia-Cárdenas, L. A. (2016). Trophic ecology of the blue shark (Prionace glauca) based on stable isotopes (δ 13 C and δ 15 N) and stomach content. Journal of the Marine Biological Association of the United Kingdom,

96(7), 1403-1410.

263. Jaime-Rivera, M., Hernández-Vázquez, S., Galván-Magaña, F., & Pérez-Lezama, E. L. (2015). A Mockingbird Mimus polyglottos (Linnaeus, 1758) found in a stomach of a blue shark Prionace glauca (Linnaeus, 1758). CICIMAR Oceánides, 30(2), 53-54.

264. Lopez, S., Meléndez, R., & Barría, P. (2010). Preliminary diet analysis of the blue shark Prionace glauca in the eastern South Pacific. Revista de Biología Marina y Oceanografía, 45(S1), 745-749.

265. Bornatowski, H., & Schwingel, P. R. (2008). Alimentação e reprodução do tubarão-azul, Prionace glauca (Linnaeus, 1758), capturado na costa Sudeste e Sul do Brasil. Arquivos de Ciências do Mar, 41(1), 98-103

266. Pusineri, C., Chancollon, O., Ringelstein, J., & Ridoux, V. (2008). Feeding niche segregation among the Northeast Atlantic community of oceanic top predators. Marine Ecology Progress Series, 361, 21-34.

267. Pardo-Gandarillas, M. C., Duarte, F., Chong, J., & Ibáñez, C. M. (2007). Dieta de tiburones juveniles Prionace glauca (Linnaeus, 1758)(Carcharhiniformes: Carcharhinidae) en la zona litoral centro-sur de Chile. Revista de biología marina y oceanografía, 42(3), 365-369.

268. Kubodera, T., Watanabe, H., & Ichii, T. (2007). Feeding habits of the blue shark, Prionace glauca, and salmon shark, Lamna ditropis, in the transition region of the Western North Pacific. Reviews in Fish Biology and Fisheries, 17(2-3), 111-124.

269. McCord, M. E., & Campana, S. E. (2003). A quantitative assessment of the diet of the blue shark (Prionace glauca) off Nova Scotia, Canada. Journal of Northwest Atlantic Fishery Science, 32, 57.

270. Pastorino, G., & Tamini, L. (2002). Argonauta nodosa Solander, 1786 (Cephalopoda: Argonautidae) in Argentine waters. Journal of Conchology, 37(5), 477-482

271. Henderson, A. C., Flannery, K., & Dunne, J. (2001). Observations on the biology and ecology of the blue shark in the North-east Atlantic. Journal of Fish Biology, 58(5), 1347-1358.

272. Clarke, M. R., Clarke, D. C., Martins, H. R., & Silva, H. M. (1996). The diet of the blue shark (Prionace glauca L.) in Azorean waters. ARQUIPÉLAGO. Ciências Biológicas e Marinhas. Life and Marine Sciences, 14, 41-56.

273. Bello, G. (1994). Histioteuthis bonnellii (Cephalopoda: Histioteuthidae) in the Adriatic Sea: Evidence from predator stomach contents. Bollettino Malacologico, 30, 43-48.

274. Seki, M. P. (1993). The role of neon flying squid, Ommastrephes bartrami, in the North Pacific pelagic food web. International North Pacific Fisheries Commission Bulletin, 53.

275. Harvey, J. T. (1989). Food habits, seasonal abundance, size, and sex of the blue shark, Prionace glauca, in Monterey bay, California. California Fish and Game, 75(1), 33-44.

276. Dorman, J. (1987). Notes on the stomach contents of some blue sharks (Prionace glauca, L.), caught in Courtmacsherry Bay, County Cork. Irish naturalists' journal, 22(5), 207.

277. Tricas, T. C. (1979). Relationships of the blue shark, Prionace glauca, and its prey species near Santa Catalina Island, California. Fishery Bulletin, 77(1), 175-182.

278. Stevens, J. D. (1973). Stomach contents of the blue shark (Prionace glauca L.) off south-west England. Journal of the marine biological association of

the United Kingdom, 53(02), 357-361.

279. Clarke, M. R., & Stevens, J. D. (1974). Cephalopods, blue sharks and migration. Journal of the marine biological association of the United Kingdom, 54(04), 949-957.
280. LeBrasseur, R. J. (1964). Stomach contents of blue shark (Prionace glauca L.) taken in the Gulf of Alaska. Journal of the Fisheries Board of Canada, 21(4), 861-862.
281. Kanakoff, G. (1940). Stomach contents of the great blue shark. Southern California Academy of Sciences, 38(3), 205.
282. Ba, B. A., Diop, M. S., Diatta, Y., Justine, D., & Ba, C. T. (2013). Diet of the milk shark, Rhizoprionodon acutus (Rüppel, 1837)(Chondrichthyes: Carcharhinidae), from the Senegalese coast. Journal of Applied Ichthyology, 29(4), 789-795.
283. Delorenzo, D. M., Bethea, D. M., & Carlson, J. K. (2015). An assessment of the diet and trophic level of Atlantic Sharpnose Shark Rhizoprionodon terraenovae. Journal of fish biology, 86(1), 385-391.
284. Bethea, D. M., Carlson, J. K., Buckel, J. A., & Satterwhite, M. (2006). Ontogenetic and site-related trends in the diet of the Atlantic sharpnose shark Rhizoprionodon terraenovae from the northeast Gulf of Mexico. Bulletin of Marine Science, 78(2), 287-307.
285. Salini, J. P., Brewer, D. T., & Blaber, S. J. M. (1998). Dietary studies on the predatory fishes of the Norman River Estuary, with particular reference to penaeid prawns. Estuarine, Coastal and Shelf Science, 46(6), 837-847.
286. Simpfendorfer, C. A. (1999). Diet of the Australian sharpnose shark, Rhizoprionodon taylori, from northern Queensland. Marine and Freshwater Research, 49(7), 757-761.
287. Wai, T. C., Yeung, J. W., Lam, V. Y., Leung, K. M., Dudgeon, D., & Williams, G. A. (2012). Monsoons and habitat influence trophic pathways and the importance of terrestrial-marine linkages for estuary sharks. Ecosphere, 3(1), 1-31.
288. Fofandi, M., Zala, M. S., & Koya, M. (2013). Observations on selected biological aspects of the spadenose shark (Scoliodon laticaudus Müller & Henle, 1838), landed along Saurashtra coast. Indian Journal of Fisheries, 60(1), 51-54.
289. Wilson, C. D., & Seki, M. P. (1994). Biology and population characteristics of Squalus mitsukurii from a seamount in the central North Pacific Ocean. Fishery Bulletin, 92(4), 851-864.
290. Jun, W., Shuyuan, Q., Yuling, H., Shengyun, Y., Xiaochun, L., & Mingru, C. (1996). Feeding Habits of spadenose shark, Scoliodon laticaudus from southern coast of Fujian. Journal of Oceanography in Taiwan Strait, 15(4), 400-406.
291. Devadoss, P. (1989). Observations on the length-weight relationship and food and feeding habits of spade nose shark, Scoliodon laticaudus Muller and Henle. Indian Journal of Fisheries, 36(2), 169-174.
292. Lyle, J. M. (1983). Food and feeding habits of the lesser spotted dogfish, Scyliorhinus canicula (L.), in Isle of Man waters. Journal of fish biology, 23(6), 725-737.
293. Özcan, E. İ., & Başusta, N. (2015). Kuzeydoğu Akdeniz Bölgesi'nde yaşayan Benekli Kedibalığı,(Scyliorhinus canicula (Linnaeus, 1758))'nın mide içeriği. Ege Journal of Fisheries and Aquatic Sciences, 32(4), 193-195.
294. Martinho, F., Sá, C., Falcão, J., Cabral, H. N., & Pardal, M. Â. (2012). Comparative feeding ecology of two elasmobranch species, Squalus blainville and Scyliorhinus canicula, off the coast of Portugal. Fishery Bulletin, 110(1), 71-84.

295. Šantić, M., Rađa, B., & Pallaoro, A. (2012). Feeding habits of small-spotted catshark (Scyliorhinus canicula Linnaeus, 1758) from the eastern central Adriatic Sea. Marine Biology Research, 8(10), 1003-1011.
296. Rodríguez-Cabello, C., Sánchez, F., & Olaso, I. (2007). Distribution patterns and sexual segregations of Scyliorhinus canicula (L.) in the Cantabrian Sea. Journal of Fish Biology, 70(5), 1568-1586.
297. Olaso, I., Velasco, F., & Pérez, N. (1998). Importance of discarded blue whiting (Micromesistius poutassou) in the diet of lesser spotted dogfish (Scyliorhinus canicula) in the Cantabrian Sea. ICES Journal of Marine Science: Journal du Conseil, 55(3), 331-341.
298. Kaiser, M. J., & Spencer, B. E. (1994). Fish scavenging behaviour in recently trawled areas. Marine ecology progress series. Oldendorf, 112(1), 41-49.
299. Braccini, J. M., Gillanders, B. M., & Walker, T. I. (2005). Sources of variation in the feeding ecology of the piked spurdog (Squalus megalops): implications for inferring predator–prey interactions from overall dietary composition. ICES Journal of Marine Science: Journal du Conseil, 62(6), 1076-1094.
300. Kabasakal, H. (2001). Preliminary data on the feeding ecology of some selachians from the north-eastern Aegean Sea. Acta Adriatica, 42(2), 15-24.
301. Mnasri, N., El Kamel, O., Boumaiza, M., Reynaud, C., & Capape, C. (2012). Food and feeding habits of the small-spotted catshark, Scyliorhinus canicula (Chondrichthyes: Scyliorhinidae) from the northern coast of Tunisia (central Mediterraneean). Cahiers de biologie marine, 53(1), 139-150.
302. Henderson, A. C., & Dunne, J. J. (1999). Food of the lesser-spotted dogfish Scyliorhinus canicula (L.), in Galway Bay. The Irish Naturalists' Journal, 191-194.
303. Caut, S., Jowers, M. J., Michel, L., Lepoint, G., & Fisk, A. (2013). Diet-and tissue-specific incorporation of isotopes in the shark Scyliorhinus stellaris, a North Sea mesopredator. Marine Ecology. Progress Series, 492, 185-198.
304. Yano, K., Stevens, J. D., & Compagno, L. J. V. (2007). Distribution, reproduction and feeding of the Greenland shark Somniosus (Somniosus) microcephalus, with notes on two other sleeper sharks, Somniosus (Somniosus) pacificus and Somniosus (Somniosus) antarcticus. Journal of fish biology, 70(2), 374-390.
305. Leclerc, L. M. E., Lydersen, C., Haug, T., Bachmann, L., Fisk, A. T., & Kovacs, K. M. (2012). A missing piece in the Arctic food web puzzle? Stomach contents of Greenland sharks sampled in Svalbard, Norway. Polar biology, 35(8), 1197-1208.
306. McMeans, B. C., Arts, M. T., & Fisk, A. T. (2015). Impacts of food web structure and feeding behavior on mercury exposure in Greenland Sharks (Somniosus microcephalus). Science of the Total Environment, 509, 216-225.
307. Nielsen, J., Hedeholm, R. B., Simon, M., & Steffensen, J. F. (2014). Distribution and feeding ecology of the Greenland shark (Somniosus microcephalus) in Greenland waters. Polar biology, 37(1), 37-46.
308. McMeans, B. C., Arts, M. T., & Fisk, A. T. (2012). Similarity between predator and prey fatty acid profiles is tissue dependent in Greenland sharks (Somniosus microcephalus): Implications for diet reconstruction. Journal of Experimental Marine Biology and Ecology, 429, 55-63.
309. McMeans, B. C., Svavarsson, J., Dennard, S., & Fisk, A. T. (2010). Diet and resource use among Greenland sharks (Somniosus microcephalus) and teleosts sampled in Icelandic waters, using δ13C, δ15N, and mercury. Canadian Journal of fisheries and aquatic sciences, 67(9), 1428-1438.
310. Fisk, A. T., Tittlemier, S. A., Pranschke, J. L., & Norstrom, R. J. (2002). Using anthropogenic contaminants and stable isotopes to assess the feeding ecology of Greenland sharks. Ecology, 83(8), 2162-2172.

311. Ridoux, V., Hall, A. J., Steingrimsson, G., & Olafsson, G. (1998). An inadvertent homing experiment with a young ringed seal, Phoca hispida. Marine Mammal Science, 14(4), 883-888.

312. Beck, B., & Mansfield, A. W. (1969). Observations on the Greenland shark, Somniosus microcephalus, in northern Baffin Island. Journal of the Fisheries Board of Canada, 26(1), 143-145.

313. Orlov, A. M., & Baitalyuk, A. A. (2014). Spatial distribution and features of biology of Pacific sleeper shark Somniosus pacificus in the North Pacific. Journal of ichthyology, 54(8), 526-546.

314. Sigler, M. F., Hulbert, L. B., Lunsford, C. R., Thompson, N. H., Burek, K., O'Corry-Crowe, G., & Hirons, A. C. (2006). Diet of Pacific sleeper shark, a potential Steller sea lion predator, in the north-east Pacific Ocean. Journal of Fish Biology, 69(2), 392-405.

315. Yang, M. S., & Page, B. N. (1999). Diet of Pacific sleeper shark, Somniosus pacificus, in the Gulf of Alaska. Fishery bulletin-national oceanic and atmospheric administration, 97, 406-409.

316. Orlov, A. M., & Moiseev, S. I. (1999). Some biological features of Pacific sleeper shark, Somniosus pacificus (Bigelow et Schroeder 1944)(Squalidae) in the Northwestern Pacific Ocean. Oceanological Studies, 28(1-2), 3-16.

317. Crovetto, A., Lamilla, J., & Pequeño, G. (1992). Lissodelphis peronii, Lacépède 1804 (Delphinidae, Cetacea) within the stomach contents of a sleeping shark, Somniosus cf. pacificus, Bigelow and Schroeder 1944, in Chilean waters. Marine mammal science, 8(3), 312-314.

318. Bright, D. B. (1959). The occurrence and food of the sleeper shark, Somniosus pacificus, in a central Alaska bay. Copeia, 1959(1), 76-77.

319. Hussey, N. E., Dudley, S. F., McCarthy, I. D., Cliff, G., & Fisk, A. T. (2011). Stable isotope profiles of large marine predators: viable indicators of trophic position, diet, and movement in sharks?. Canadian Journal of Fisheries and Aquatic Sciences, 68(12), 2029-2045.

320. De Bruyn, P., Dudley, S. F. J., Cliff, G., & Smale, M. J. (2005). Sharks caught in the protective gill nets off KwaZulu-Natal, South Africa. 11. The scalloped hammerhead shark Sphyrna lewini (Griffith and Smith). African Journal of Marine Science, 27(3), 517-528.

321. Torres-Rojas, Y. E., Páez Osuna, F., Camalich, J., & Galván Magaña, F. (2015). Diet and trophic level of scalloped hammerhead shark (Sphyrna lewini) from the Gulf of California and Gulf of Tehuantepec, Mexico. Iranian Journal of Fisheries Sciences, 14(3), 767-785.

322. Bergés-Tiznado, M. E., Márquez-Farías, F., Lara-Mendoza, R. E., Torres-Rojas, Y. E., Galván-Magaña, F., Bojórquez-Leyva, H., & Páez-Osuna, F. (2015). Mercury and selenium in muscle and target organs of Scalloped Hammerhead Sharks Sphyrna lewini of the SE Gulf of California: dietary intake, molar ratios, loads, and human health risks. Archives of environmental contamination and toxicology, 69(4), 440-452.

323. Bethea, D. M., Carlson, J. K., Hollensead, L. D., Papastamatiou, Y. P., & Graham, B. S. (2011). A comparison of the foraging ecology and bioenergetics of the early life-stages of two sympatric hammerhead sharks. Bulletin of Marine Science, 87(4), 873-889.

324. Torres-Rojas, Y. E., Hernández-Herrera, A., Galván-Magaña, F., & Alatorre-Ramírez, V. G. (2010). Stomach content analysis of juvenile, scalloped hammerhead shark Sphyrna lewini captured off the coast of Mazatlán, Mexico. Aquatic Ecology, 44(1), 301-308.

325. Zanella Cesarotto, I., López Garro, A., & Arauz Vargas, R. (2010). The feeding habits of juvenile scalloped hammerhead sharks (Sphyrna lewini) caught in the gulf of nicoya, costa rica. Boletín de Investigaciones Marinas y Costeras-INVEMAR, 39(2), 447-453.

326. Estupiñán-Montaño, C., Cedeño-Figueroa, L. G., & Galván-Magaña, F. (2009). Hábitos alimentarios del tiburón martillo Sphyrna lewini (Griffith & Smith, 1834)(Chondrichthyes) en el Pacífico ecuatoriano. Revista de biología marina y oceanografía, 44(2), 379-386.

327. Pena-Vargas V. (2008). Feeding behaviour of juveniles of the bronze hammerhead shark (Sphyrna lewini Griffith y Smith, 1834) on the coast of Chiapas, Mexico. [III National Symposium on Sharks and Rays. 25-29 August 2008, Faculty of Science, UNAM, Ciudad Unversitaria, Mexico D.F., 109-111.
328. Torres-Rojas, Y., Hernandez-Herrera, A., & Galvan-Magaña, F. (2006). Feeding habits of the scalloped hammerhead shark, Sphyrna lewini, in Mazatlán waters, southern Gulf of California, Mexico. Cybium, 30(4), 85-90.
329. Bush, A. (2003). Diet and diel feeding periodicity of juvenile scalloped hammerhead sharks, Sphyrna lewini, in Kāne'ohe Bay, Ō'ahu, Hawai'i. Environmental biology of fishes, 67(1), 1-11.
330. Clarke, T. A. (1971). The Ecology of the Scalloped Hammerhead Shark, Sphyrna lewini, in Hawaii. Pacific Science, 25, 133-144.
331. Stevens, J. D., & Lyle, J. M. (1989). Biology of three hammerhead sharks (Eusphyra blochii, Sphyrna mokarran and S. lewini) from northern Australia. Marine and Freshwater Research, 40(2), 129-146.
332. Smale, M. J., & Cliff, G. (1998). Cephalopods in the diets of four shark species (Galeocerdo cuvier, Sphyrna lewini, S. zygaena and S. mokarran) from KwaZulu-Natal, South Africa. South African Journal of Marine Science, 20(1), 241-253.
333. Cliff, G. (1995). Sharks caught in the protective gill nets off KwaZulu-Natal, South Africa. 8. The great hammerhead shark Sphyrna mokarran (Rüppell). South African Journal of Marine Science, 15(1), 105-114.
334. Bethea, D. M., Hale, L., Carlson, J. K., Cortés, E., Manire, C. A., & Gelsleichter, J. (2007). Geographic and ontogenetic variation in the diet and daily ration of the bonnethead shark, Sphyrna tiburo, from the eastern Gulf of Mexico. Marine Biology, 152(5), 1009-1020.
335. Reis-Filho, J. A., Sampaio, C. L., Leite, L., Oliveira, G. S., Loiola, M., & José de Anchieta, C. C. (2014). Rediscovery of bonnethead shark Sphyrna tiburo after more than two decades of non-record on central coast of Brazil. Marine Biodiversity Records, 7.
336. Cortes, E., Manire, C. A., & Hueter, R. E. (1996). Diet, feeding habits, and diel feeding chronology of the bonnethead shark, Sphyrna tiburo, in southwest Florida. Bulletin of Marine Science, 58(2), 353-367.
337. Lessa, R. P. (1998). feeding habits of the bonnethead shark, Sphyrna tiburo, from northern brazil. Cybium, 22(4), 383-394.
338. Castro, J. I. (1989). The biology of the golden hammerhead, Sphyrna tudes, off Trinidad. Environmental Biology of Fishes, 24(1), 3-11.
339. Bornatowski, H., & Schwingel, P. R. (2009). Sobre a alimentação do tubarão-martelo, Sphyrna zygaena (Linnaeus, 1758), capturado por espinhel pelágico no sudeste e sul do brasil. Arquivos de Ciências do Mar, 42(2), 89-92.
340. Bolaño Martínez, N. (2006). Hábitos alimenticios en juveniles de Sphyrna Zygaena Linnaeus 1758 (Chondrichthyes: Sphyrnidae) en el puerto pesquero de Santa Rosa de Salinas, Guayas durante mayo–diciembre 2004 (Bachelor's thesis).
341. Bornatowski, H., Costa, L., Robert, M. C., & Pina, J. V. (2007). Hábitos alimentares de tubarões-martelo jovens, Sphyrna zygaena (Carcharhiniformes: Sphyrnidae), no litoral sul do Brasil. Biota Neotropica, 7(1), 213-6.
342. Belleggia, M., Figueroa, D. E., Sánchez, F., & Bremec, C. (2012b). Long-term changes in the spiny dogfish (Squalus acanthias) trophic role in the southwestern Atlantic. Hydrobiologia, 684(1), 57-67.
343. Gračan, R., Zavodnik, D., Krstinić, P., Dragičević, B., & Lazar, B. (2017). Feeding ecology and trophic segregation of two sympatric mesopredatory sharks in the heavily exploited coastal ecosystem of the Adriatic Sea. Journal of Fish Biology, 90(1), 167-184.

344. Bangley, C. W., & Rulifson, R. A. (2014). Feeding habits, daily ration, and potential predatory impact of mature female spiny dogfish in North Carolina coastal waters. North American Journal of Fisheries Management, 34(3), 668-677.

345. Bangley, C. W., Rulifson, R. A., & Overton, A. S. (2013). Evaluating the efficiency of flushed stomach-tube lavage for collecting stomach contents from dogfish sharks. Southeastern Naturalist, 12(3), 523-533.

346. Yick, J. L., Barnett, A., & Tracey, S. R. (2012). The trophic ecology of two abundant mesopredators in south-east coastal waters of Tasmania, Australia. Marine biology, 159(6), 1183-1196.

347. Sturdevant, M. V., Orsi, J. A., & Fergusson, E. A. (2012). Diets and trophic linkages of epipelagic fish predators in coastal Southeast Alaska during a period of warm and cold climate years, 1997–2011. Marine and Coastal Fisheries, 4(1), 526-545.

348. Brodeur, R. D., Fleming, I. A., Bennett, J. M., & Campbell, M. A. (2009). Summer distribution and feeding of spiny dogfish off the Washington and Oregon coasts. Biology and management of dogfish sharks. American Fisheries Society, Bethesda, 39-51.

349. Demirhan, S. A., & Seyhan, K. (2007). Life history of spiny dogfish, Squalus acanthias (L. 1758), in the southern Black Sea. Fisheries Research, 85(1), 210-216.

350. Yang, M. S., Dodd, K., Hibpshman, R., & Whitehouse, A. (2006). Food habits of groundfishes in the Gulf of Alaska in 1999 and 2001. US Department of Commerce, National Oceanic and Atmospheric Administration, National Marine Fisheries Service, Alaska Fisheries Science Center, Pp. 199.

351. Koen Alonso, M., Alberto Crespo, E., Aníbal García, N., Noemí Pedraza, S., Ariel Mariotti, P., & Judith Mora, N. (2002). Fishery and ontogenetic driven changes in the diet of the spiny dogfish, Squalus acanthias, in Patagonian waters, Argentina. Environmental Biology of Fishes, 63(2), 193-202.

352. Henderson, A. C., Dunne, J., & Flannery, K. (2002). Stomach contents of spiny dogfish Squalus acanthias L. off the west coast of Ireland. The Irish Naturalists' Journal, 101-105.

353. Laptikhovsky, V. V., Arkhipkin, A. I., & Henderson, A. C. (2001). Feeding habits and dietary overlap in spiny dogfish Squalus acanthias (Squalidae) and narrowmouth catshark Schroederichthys bivius (Scyliorhinidae). Journal of the Marine Biological Association of the United Kingdom, 81(6), 1015.

354. García de La Rosa, S. B., & Sánchez, M. F. (1997). Alimentación de Squalus acanthias y predación sobre Merluccius hubbsi en el Mar Argentino entre 34° 47'-47° S. Rev. Invest. Des. Pesq. 11: 119-133.

355. Hanchet, S. (1991). Diet of spiny dogfish, Squalus acanthias Linnaeus, on the east coast, South Island, New Zealand. Journal of fish biology, 39(3), 313-323.

356. Livingston, M. E. (1990). Spawning hoki (Macruronus novaezelandiae Hector) concentrations in Cook Strait and off the east coast of the South Island, New Zealand, August-September 1987. New Zealand journal of marine and freshwater research, 24(4), 503-517.

357. Jones, B. C., & Geen, G. H. (1977). Food and feeding of spiny dogfish (Squalus acanthias) in British Columbia waters. Journal of the Fisheries Board of Canada, 34(11), 2056-2066.

358. Holden M. J. (1966). The food of the spurdog, Squalus acanthias (L.). Journal du Conseil Permanent International pour l'Exploration de la Mer, 30,

255-266.

359. Özütemiz, Ş., Kaya, M., & Özaydın, O. (2009). Growth and Feeding Characteristics of Two Sharks Species [Galeus melastomus Rafinesque, 1810 and Squalus blainvillei (Risso, 1826)] from Sığacık Bay (Aegean Sea). Ege Journal of Fisheries and Aquatic Sciences, 26(3), 211-217.

360. Capapé, C. (1975). Etude du régime alimentaire de Squalus blainvillei (Risso, 1826) des côtes tunisiennes. Bulletin de l'Institut national scientifique et technique d'océanographie et de pêche, 4, 61-73.

361. Kabasakal, H. (2002b). Stomach contents of the longnose spurdog Squalus blainvillei (Risso, 1826) from the North-Eastern Aegean Sea. In Annales (Annals for Istran and Mediterranean Studies), Series historia naturalis, 12(2), 161-166.

Table A.2. Table showing intermediate hosts of trypanorhynch tapeworms, which infect various shark species with known diet information. For each tapeworm species, the larval stage(s) are given alongside the known definitive hosts and intermediate hosts obtained via the extensive records in Palm (2004) and from ISI Web of Knowledge references 2004-present.

Trypanorhynch tapeworm species	**Tapeworm Family**	**Definitive shark hosts with diet records**	**Intermediate hosts (family** -species) listed with references Key to larval stage: pro = procercoid, pls = plerocercus, pld = plerocercoid, mer = merocercoid
Aporhynchus norvegicus	Aporhynchidae	*Etmopterus spinax*	Crustaceans: **Euphausiidae** -*Meganyctiphanes norvegica* (pls) [1]; **Calanidae** -Calanus finmarchicus (pro) [1]
Dollfusiella lineata	Eutetrarhynchidae	*Ginglymostoma cirratum*	Teleosts: **Sciaenidae** -*Sciaenops ocellatus* (pls) [1]
Dollfusiella martini	Eutetrarhynchidae	*Carcharhinus brachyurus*	Crustaceans: **Carcinidae** -*Carcinus maenas (pld) [2-3]*
Eutetrarhynchus ruficollis	Eutetrarhynchidae	*Mustelus canis, Mustelus mustelus, Squalus acanthias*	Crustaceans: **Cancridae** -*Cancer pagurus* (pls) [1]; **Carcinidae** -*Carcinus maenas* (pls) [1]; **Oregoniidae** -*Hyas araneus* (pls) [1]; **Inachidae** -*Inachus dorsettensis* (pls) [1], *Macropodia longirostris* (pls) [1], *Macropodia rostrata* (pls) [1] **Polybiidae** - *Liocarcinus marmoreus* (pls) [1], *Liocarcinus depurator* (pls) [1]; **Paguridae** -*Pagurus bernhardus* (pls) [1]; **Penaeidae** -*Penaeus kerathurus* (pls) [1]; **Pilumnidae** -*Pilumnus hirtellus* (pls) [1]; Other mollusks: **Ostreidae** - *Ostrea edulis* (pls) [1];

Parachristianella dimegacantha	Eutetrarhynchidae	*Sphyrna zygaena*	Crustaceans: **Penaeidae** -*Penaeus aztecus* (pls) [1], *Penaeus duorarum* (pls) [1]; Other mollusks: **Veneridae** -*Chione cancellate* (pls) [1]; **Mactridae** -*Spisula solidissima* (pls) [1]
Prochristianella tumidula	Eutetrarhynchidae	*Carcharhinus obscurus, Mustelus canis, Mustelus mustelus*	Teleosts: **Batrachoididae** -*Opsanus tau* (pls) [1];
Gilquinia squali	Gilquiniidae	*Etmopterus granulosus, Mustelus mustelus, Scyliorhinus stellaris, Squalus acanthias, Squalus blainville*	Teleosts: **Gadidae** -*Merlangius merlangus* (pls) [1]; **Salmonidae** -*Oncorhynchus tshawytscha* (pls) [1];
Gymnorhynchus gigas	Gymnorhynchidae	*Carcharodon carcharias, Isurus oxyrinchus, Lamna nasus*	Teleosts: **Bramidae** -*Brama brama* (mer) [1]; **Xiphiidae** -Xiphias gladius (pld) [4]; **Trichiuridae** -*Lepidopus caudatus* (pld) [5]
Molicola horridus	Gymnorhynchidae	*Carcharodon carcharias, Isurus oxyrinchus, Prionace glauca*	Teleosts: **Diodontidae** - *Cyclichthys orbicularis* (mer) [1], *Diodon holocanthus* (mer) [1], *Diodon hystrix* (mer) [1,11], *Diodon liturosus* (mer) [11]; **Molidae** -*Masturus lanceolatus* (mer) [1], *Mola mola* (mer) [1,7]; **Xiphiidae** -*Xiphias gladius* (pld) [4,6];
Molicola uncinatus	Gymnorhynchidae	*Alopias vulpinus*	Teleosts: **Scombridae** -*Allothunnus fallai* (mer) [1]; **Molidae** -*Mola mola* (mer) [1]; **Bramidae** -*Taractes rubescens* (mer) [1], *Taractichthys steindachneri* (mer) [1]; **Gempylidae** -*Thyrsites atun* (mer) [1,8]; **Xiphiidae** -*Xiphias gladius* (mer) [1]

Callitetrarhynchus gracilis	Lacistorhynchidae	*Carcharhinus amblyrhynchoides, Carcharhinus amboinensis, Carcharhinus leucas, Carcharhinus limbatus, Carcharhinus melanopterus, Carcharhinus obscurus, Mustelus canis, Negaprion brevirostris, Prionace glauca, Rhizoprionodon acutus, Rhizoprionodon terraenovae, Sphyrna lewini, Sphyrna zygaena*	Teleosts: **Belonidae** -*Ablennes hians* (pls) [1], *Tylosurus crocodilus crocodilus* (pls) [1]; **Carangidae** -*Alepes djedaba* (pls) [1], *Alectis alexandrina* (pls) [1], *Carangoides otrynter* (pls) [1], *Carangoides bajad* (pls) [1], *Carangoides fulvoguttatus* [11], *Caranx crysos* (pls) [1], *Caranx hippos* (pls) [1], *Caranx ignobilis* (pls) [1], *Caranx rhonchus* (pls) [1], *Caranx ruber* (pls) [1], *Caranx senegallus* (pls) [1], *Caranx sexfasciatus* (pls) [1*], Caranx papuensis [11], Caranx sp*. (pls) [1] Caranx latus (pls) [19], *Chloroscombrus chrysurus* (pls) [1], Lichia amia (pls) [1], *Megalaspis cordyla* (pls) [1,11], *Oligoplites palometa* (pls) [1], *Oligoplites saurus* (pls) [1], *Scomberoides commersonnianus* (pls) [1], *Scomberoides lysan* (pls) [1], *Scomberoides tala* (pls) [1], *Selene vomer* (pls) [1], Seriola quinqueradiata (pls) [1], Seriola sp. (pls) [1], *Trachinotus goodei* (pls) [1], *Trachinotus ovatus* (pls) [1], *Trachurus capensis* (pls) [1], *Trachurus trachurus* (pls) [1], *Atule mate* [11]; **Serranidae** -*Alphestes afer* (pls) [1], *Cephalopholis fulva* (pls) [1], *Cephalopholis taeniops* (pls) [1], *Cephalopholis miniata* (pls) [11,17], *Cephalopholis boenak* [11], *Cephalopholis cyanostigma* [11], *Cephalopholis spiloparaea* [11], *Hyporthodus niveatus* (pls) [1], *Epinephelus adscensionis* (pls) [1], *Epinephelus aeneus* (pls) [1], *Epinephelus akaara* (pls) [1], *Epinephelus maculatus* (pls) [1], *Epinephelus striatus* (pls) [1], *Epinephelus chlorostigma* [11], *Epinephelus fasciatus* [11], *Epinephelus retouti* [11], *Epinephelus rivulatus* [11], *Epinephelus polyphekadion* (pls) *[17] Epinephelus summana* (pls) [17]*, Epinephelus sp.* (pls) [1], *Hemilutjanus macrophthalmos* (pls) [1], *Mycteroperca bonaci* (pls) [1], *Mycteroperca interstitialis* (pls) [1], *Mycteroperca tigris* (pls) [1], *Mycteroperca venenosa* (pls) [1], *Paralabrax humeralis* (pls) [1], *Paranthias furcifer* (pls) [1], *Cromileptes altivelis* [11], *Plectropomus maculatus* [11], *Variola louti* [11]; **Sciaenidae** -*Argyrosomus regius* (pls) [1], *Cynoscion guatucupa* (pls) [1,20], *Cynoscion jamaicensis* (pls) [1], *Larimus breviceps* (pls) [1] *Macrodon ancylodon* (pls) [1,20], *Micropogonias furnieri* (pls) [1,20], *Otolithes ruber* (pls) [1,13,15], *Paralonchurus peruanus* (pls) [1], *Pennahia anea* (pls) [1], *Pennahia argentata* (pls)

[1], *Paralonchurus peruanus* (pls) [1], *Sciaena deliciosa* (pls) [1], *Johnius borneensis* [11];

Ariidae -*Ariopsis felis* (pls) [1], *Netuma thalassina* (pls) [1], *Bagre marinus* (pls) [1], *Genidens barbus* (pls) [1];

Arripidae -*Arripis truttacea* (pls) [1];

Scombridae -*Auxis rochei rochei* (pls) [1], *Euthynnus affinis* (pls) [1], *Euthynnus alletteratus* (pls) [1,9], *Euthynnus sp.* (pls) [1], *Scomber colias* (pls) [1], *Scomberomorus cavalla* (pls) [1], *Scomberomorus commerson* (pls) [1,11,21], *Scomberomorus guttatus* (pls) [1], *Scomberomorus maculatus* (pls) [1], *Scomberomorus munroi* (pls) [1], *Scomberomorus niphonius* (pls) [1], *Scomberomorus queenslandicus* [11], *Thunnus albacares* (pls) [1], *Thunnus thynnus* (pls) [1], *Thunnus sp.* (pls) [1];

Balistidae -*Balistes sp.* (pls) [1];

Bramidae -*Brama brama* (pls) [1];

Centropomidae -*Centropomus undecimalis* (pls) [1];

Bothidae -*Chascanopsetta lugubris* (pls) [1];

Psettodidae - *Psettodes erume*i (pls) [10,15],

Chirocentridae -*Chirocentrus dorab* (pls) [1,11], *Chirocentrus nudus* (pld) [15];

Chloropthalmidae -*Chlorophthalmus agassizi* (pls) [1];

Sparidae -*Chrysoblephus puniceus* (pls) [1], *Chrysophrys auratus* (pls) [1], *Pagrus pagrus* (pls) [1], *Porcostoma dentata* (pls) [1];

Labridae -*Bodianus axillaris* (pls) [1], *Choerodon cyanodus* [11];

Exocoetidae -*Cypselurus poecilopterus* (pls) [1];

Moronidae -*Dicentrarchus labrax* (pls) [1];

Dinopercidae -*Dinoperca petersi* (pls) [1];

Elopidae -*Elops saurus* (pls) [1];

Engraulidae -*Engraulis japonicus* (pls) [1];

Clupeidae -*Ethmalosa fimbriate* (pls) [1], *Harengula clupeola* (pls) [1], *Opisthonema oglinum* (pls) [1], *Sardina pilchardus* (pls) [1], *Sardinella maderensis* (pls) [1];

Ammodytidae - *Gymnammodytes cicerelus* (pls) [1];

Haemulidae -*Haemulon aurolineatum* (pls) [1];

Istiophoridae - *Istiophorus platypterus* (pls) [1], *Istiompax indica* (pls) [1], *Makaira mazara* (pls) [1];

Latidae - *Lates calcarifer* (pls) [1];

Leiognathidae - *Leiognathus equulus* (pls) [1], *Secutor ruconius* (pls) [1];

Trichiuridae -*Lepturacanthus savala* (pls) [1], *Trichiurus lepturus* (pls, pld) [1,14,56];

Macrouridae -*Malacocephalus laevis* (pls) [1];

Lethrinidae -*Lethrinus erythracanthus* (pls) [1], *Lethrinus miniatus* [11], *Lethrinus nebulosus* [15], *Lethrinus mahsena* (pls) [17], *Lethrinus variegatus* (pls) [17];

Lutjanidae -*Lutjanus analis* (pls) [1], *Lutjanus campechanus* (pls) [1], *Lutjanus fulgens* (pls) [1], *Lutjanus goreensis* (pls) [1], *Lutjanus griseus* (pls) [1], *Lutjanus vitta* (pls) [1,11], *Lutjanus carponotatus* [11], *Lutjanus johnii* (pls) [15], *Lutjanus sp.* (pls) [1], *Ocyurus chrysurus* (pls) [1], *Pristipomoides multidens* (pls) [1];

Merlucciidae -*Merluccius gayi peruanus* (pls) [1], *Merluccius gayi gayi* (pls) [1];

Mullidae -*Mullus barbatus* (pls) [1];

Muraenesocidae -*Muraenesox cinereus* (pls) [1];

Nemipteridae - *Nemipterus japonicus* (pls) [1], *Nemipterus furcosus* [11];

Paralichthyidae -*Paralichthys dentatus* (pls) [1], *Paralichthys olivaceus* (pls) [1], *Paralichthys isosceles* (pls) [18];

Platycephalidae -*Platycephalus fuscus* (pls) [1];

Pomatomidae -*Pomatomus saltatrix* (pls) [1,11,19];

Priacanthidae -*Priacanthus hamrur* (pls) [1];

Rachycentridae -*Rachycentron canadum* (pls) [1];

			Synodontidae -*Saurida tumbil* (pls) [1,15], *Saurida undosquamis* (pls) [1], *Synodus lucioceps* (pls) [1]; **Sphyraenidae -** *Sphyraena acutipinnis* (pls) [1], *Sphyraena barracuda* (pls) [1], *Sphyraena guachancho* (pls) [1], *Sphyraena novaehollandiae* (pls) [1], *Sphyraena pinguis* (pls) [1], *Sphyraena obtusata* [11]; **Monacanthidae** -(unidentified) (pls) [1]; **Pomacentridae** -*Abudefduf whitleyi* [11]; **Apogonidae** -*Apogon poecilopterus* [11], *Ostorhinchus fasciatus* [11]; **Caesionidae** -*Caesio cuning* [11]; **Moridae** -*Lotella rhacina* [11]; **Acanthuridae** -*Naso vlamingii* [11]; **Polynemidae** -*Filimanus heptadactyla* [11], *Eleutheronema tetradactylum* [16]; **Triodontidae** -*Triodon macropterus* [11]; **Phycidae** -*Urophycis brasiliensis* [12]; **Ophidiidae** -*Genypterus brasiliensis* (pls) [22]; **Coryphaenidae -***Coryphaena hippurus* [28], *Coryphaena equiselis* [28]; Chondrichthyan fishes **Ginglymostomatidae** -*Nebrius ferrugineus* (pls) [1]; Reptiles: **Colubridae** -*Cerberus rynchops* (pls) [1];
Callitetrarhynchus speciosus	Lacistorhynchidae	*Carcharhinus obscurus, Negaprion brevirostris*	Teleosts: **Ariidae -***Netuma thalassina* (pls) [1], *Genidens barbus* (pls) [1]; **Platycephalidae** -*Cociella punctata* (pls) [1], *Cymbacephalus beauforti* [11], *Platycephalus indicus* (pls) [1]; **Congridae** -*Conger cinereusi* (pls) [17]; **Pomatomidae** –*Pomatomus saltatrix* (pls) [1,19]; **Sciaenidae** -*Cynoscion guatucupa* (pls) [1,20], *Cynoscion regalis* (pls) [1], *Micropogonias furnieri* (pls) [1,20], *Nibea albiflora* (pls) [1]; **Balistidae** -*Canthidermis maculata* (pls) [1];

			Serranidae -*Cephalopholis hemistiktos* (pls) [1], *Epinephelus adscensionis* (pls) [1], *Epinephelus areolatus* (pls) [1], *Epinephelus maculatus* (pls) [1], *Epinephelus morio* (pls) [1], *Epinephelus multinotatus* (pls) [1], *Epinephelus striatus* (pls) [1], *Epinephelus tukula* (pls) [1], *Mycteroperca interstitialis* (pls) [1], *Mycteroperca phenax* (pls) [1], *Mycteroperca venenosa* (pls) [1]; **Sparidae** -*Chrysoblephus anglicus* (pls) [1]; **Coryphaenidae** -*Coryphaena hippurus* (pls) [1]; **Echeneidae** -*Echeneis naucrates* (pls) [1], **Haemulidae** -*Haemulon album* (pls) [1]; **Priacanthidae** -*Heteropriacanthus cruentatus* (pls) [1], *Priacanthus arenatus* (pls) [23]; **Scombridae** -*Katsuwonis pelamis* (pls) [1], *Scomber japonicus* (pls) [1], *Scomberomorus commerson* (pls) [1], *Scomberomorus guttatus* (pls) [1], *Thunnus thynnus* (pls) [1]; **Labridae** -*Lachnolaimus maximus* (pls) [1]; **Lethrinidae** -*Lethrinus nebulosus* (pls) [1]; **Lutjanidae** -*Lutjanus analis* (pls) [1], *Lutjanus campechanus* (pls) [1], *Lutjanus griseus* (pls) [1], *Lutjanus synagris* (pls) [1], *Lutjanus argentimaculatus* [25], *Ocyurus chrysurus* (pls) [1]; **Muraenesocidae** -*Muraenesox cinereus* (pls) [1]; **Triglidae** -*Prionotus carolinus* (pls) [1]; **Carangidae** -*Selene vomer* (pls) [1], *Seriola dumerili* (pls) [1], *Trachinotus goodei* (pls) [1]; **Monacanthidae** -*Stephanolepis hispidus* (pls) [1], *Aluterus monoceros* (pls) [24]; **Trichiuridae** -*Trichiurus lepturus* (pls) [1]; **Xiphiidae** -*Xiphias gladius* (pls) [1];
Dasyrhynchus giganteus	Lacistorhynchidae	*Carcharhinus leucas, Carcharhinus melanopterus,*	Teleosts: **Carangidae** -*Caranx hippos* (pls) [1], *Oligoplites saliens* (pls) [1], *Seriola dumerili* (pls) [1];

		Carcharhinus plumbeus, Negaprion brevirostris, Rhizoprionodon terraenovae	**Polynemidae** -*Polydactylus quadrifilis* (pls) [1]; **Xiphiidae** -*Xiphias gladius* (pls) [1];
Dasyrhynchus pacificus	Lacistorhynchidae	*Carcharhinus brachyurus, Carcharhinus limbatus, Carcharhinus obscurus, Carcharhinus plumbeus, Sphyrna lewini*	Teleosts: **Sciaenidae** -*Cynoscion guatucupa* (pls) [1,20], *Cynoscion jamaicensis* (pls) [1], *Macrodon ancylodon* (pls) [1,20], *Micropogonias furnieri* [20], *Menticirrhus americanus* (pls) [1,20], *Argyrosomus japonicus* (pls) [1], *Argyrosomus hololepidotus* (pls) [1], *Sciaena deliciosa* (pls) [1]; **Monacanthidae** -*Acanthaluteres brownie* (pls) [1]; **Lutjanidae** -*Aprion virescens* (pls) [1]; **Lethrinidae** -*Lethrinus mahsena* (pls) [1]; **Mugilidae** -*Mugil curema* (pls) [1], *Mugil cephalus* (pls) [1];
Dasyrhynchus talismani	Lacistorhynchidae	*Carcharhinus brachyurus, Carcharhinus leucas, Carcharhinus longimanus, Prionace glauca*	Teleosts: **Scombridae** -*Thunnus albacares* (pls) [1], *Thunnus obesus* (pls) [1];
Dasyrhynchus varioucinatus	Lacistorhynchidae	*Carcharhinus amblyrhynchoides, Carcharhinus amblyrhynchos, Carcharhinus falciformis, Carcharhinus leucas, Carcharhinus limbatus, Carcharhinus plumbeus, Carcharodon carcharias, Negaprion brevirostris*	Teleosts: **Scombridae** -*Euthynnus affinis* (pls) [1]; **Carangidae** -*Carangoides ciliarius* (pls) [1], *Caranx sexfasciatus* (pls) [1], *Caranx sp.* (pls) [1];

Diesingium lomentaceum	Lacistorhynchidae	*Galeorhinus galeus, Mustelus canis, Mustelus mustelus*	Teleosts: **Carangidae** -*Carangoides fulvoguttatus* [11]; **Serranidae** -*Epinephelus chlorostigma* [11]
Floriceps minacanthus	Lacistorhynchidae	*Carcharhinus amblyrhynchos, Carcharhinus amboinensis, Carcharhinus brachyurus, Carcharhinus melanopterus*	Teleosts: **Serranidae** -*Cephalopholis hemistiktos* (pls) [1], *Cephalopholis urodeta [11], Cephalopholis miniate* (pld) *[11,26-27], Cephalopholis boenak [11], Cephalopholis cyanostigma [11], Cephalopholis sonnerati [11], Epinephelus quoyanus [11], Epinephelus coioides [11], Epinephelus cyanopodus [11], Epinephelus maculatus [11], Plectropomus areolatus* [11], *Plectropomus leopardus* (pls) [1,11], Plectropomus laevis [11], *Variola louti* [11]*;* **Scombridae** -*Euthynnus affinis* (pls) [1,11], *Euthynnus alletteratus* (pls) [1,11], *Grammatorcynus bicarinatus* [11], *Gymnosarda unicolor* (pls) [1]; **Lethrinidae** -*Lethrinus miniatus* (pls) [1,11], *Lethrinus mahsena* (pls,pld) [1,26]; **Nemipteridae** -*Nemipterus furcosus* (pls) [1,11]; **Sphyraenidae** -*Sphyraena flavicauda* [11], *Sphyraena putnamae* [11], *Sphyraena jello* [11], *Sphyraena novaehollandiae* (pls) [1]; **Belonidae** -*Tylosurus crocodilus crocodilus* [11]; **Carangidae -***Carangoides bajad* (pls,pld) [1,26]; **Platycephalidae** -*Platycephalus bassensis* (pls) [1], *Platycephalus laevigatus* (pls) [1], *Platycephalus sp.* (pls) [1]
Floriceps saccatus	Lacistorhynchidae	*Carcharhinus limbatus, Carcharhinus obscurus, Carcharhinus plumbeus, Carcharhinus signatus, Negaprion brevirostris, Notorynchus cepedianus, Prionace glauca*	Teleosts: **Carangidae** -*Caranx papuensis* [11], *Caranx hippos* (pls) [1], *Seriola lalandi* (pls) [1], *Trachinotus ovatus* (pls) [1]; **Diodontidae** -*Diodon holocanthus* (pls) [1], *Diodon liturosus* [11], *Diodon hystrix* (pls) [1,11]; **Monacanthidae** -*Aluterus monoceros* (pls) [24], *Aluterus sp.* (pls) [1]; **Coryphaenidae** -*Coryphaena hippurus* (pls) [1,28], *Coryphaena equiselis* (pls) [1]; **Sciaenidae** -*Argyrosomus regius* (pls) [1]; **Centropomidae** -Centropomus nigrescens (pls) [1];

			Gempylidae -*Gempylus serpens* (pls) [1,56]; **Pleuronectidae** -*Glyptocephalus stelleri* (pls) [1]; **Scombridae** -*Katsuwonis pelamis* (pls) [1]; **Molidae** -*Mola mola* (pls) [1]; **Tetraodontidae** -*Takifugu porphyreus* (pls) [1]; **Trichiuridae** -*Trichiurus lepturus* (pls) [1];
Grillotia acanthoscolex	Lacistorhynchidae	*Hexanchus griseus*	Teleosts: **Scorpaenidae** -*Scorpaena scrofa* (pls) [1]; **Lophiidae** -*Lophius piscatorius* (pls) [29]; Chondrichthyan fishes: **Torpedinidae** -*Tetronarce nobiliana* (pls) [1]; **Centrophoridae** -*Deania hystricosa* (pls) [1], *Deania profundorum* (pls) [1]; **Hexanchidae** -*Heptranchias perlo* (pls) [1];
Grillotia amblyrhynchos	Lacistorhynchidae	*Carcharhinus amblyrhynchos*	Chondrichthyan fishes: **Etmopteridae** -*Etmopterus sp*. (pls) [1];
Grillotia dolichocephala	Lacistorhynchidae	*Centrophorus squamosus*	Chondrichthyan fishes: **Somniosidae** -*Centroscymnus coelolepis* (pls) [29]; **Centrophoridae** -*Centrophorus squamosus* (pls) [29], *Deania profundorum* (pls) [29];
Grillotia dollfusi	Lacistorhynchidae	*Carcharhinus signatus*, *Heptranchias perlo*	Teleosts: **Macrouridae** -*Nezumia aequalis* (pls) [1]; **Merlucciidae** -*Merluccius gayi gayi* (pls) [1];
Grillotia erinaceus	Lacistorhynchidae	*Squalus acanthias*	Teleosts: **Agonidae** -*Agonus cataphractus* (pls) [1]; **Anarhichadidae** -*Anarhichas lupus* (pls) [1]; **Bothidae** -*Arnoglossus laterna* (pls) [1]; **Lotidae** -*Brosme brosme* (pls) [1], *Enchelyopus cimbrius* (pls) [1], *Lota lota* (pls) [1], *Molva molva* (pls) [1]; **Macrouridae** -*Coelorinchus fasciatus* (pls) [1], *Malacocephalus laevis* (pls) [1];

Callionymidae -*Callionymus lyra* (pls) [1];

Caproidae -*Capros aper* (pls) [1];

Cepolidae -*Cepola macrophthalma* (pls) [1];

Triglidae -*Chelidonichthys cuculus* (pls) [1], *Chelidonichthys lucerna* (pls) [1], *Eutrigla gurnardus* (pls) [1], *Trigloporus lastoviza* (pls) [1], *Prionotus carolinus* (pls) [1];

Clupeidae -*Clupea harengus* (pls) [1];

Congridae -*Conger conger* (pls) [1];

Serranidae -*Epinephelus marginatus* (pls) [1];

Gadidae -*Gadus morhua* (pls) [1], *Melanogrammus aeglefinus* (pls) [1], *Merlangius merlangus* (pls,pld) [1,30-31], *Pollachius pollachius* (pls) [1], *Pollachius virens* (pls) [1], *Trisopterus esmarkii* (pls) [1], *Trisopterus minutus* (pls) [1];

Pleuronectidae -*Glyptocephalus cynoglossus* (pls) [1], *Hippoglossoides platessoides* (pls) [1], *Hippoglossus hippoglossus* (pls) [1], *Limanda aspera* (pls) [1], *Limanda ferruginea* (pls) [1], *Limanda limanda* (pls) [1] *Microstomus kitt* (pls) [1], *Platichthys flesus* (pls) [1], *Pleuronectes platessa* (pls) [1], *Reinhardtius hippoglossoides* (pls) [1];

Ateleopodidae -*Guentherus altivela* (pls) [1];

Scophthalmidae -*Lepidorhombus Whiffiagonis* (pls) [1], *Scophthalmus aquosus* (pls) [1], *Scophthalmus maximus* (pls) [1], *Scophthalmus rhombus* (pls) [1];

Lophiidae -*Lophius piscatorius* (pls) [1];

Merlucciidae -*Merluccius bilinearis* (pls) [1], *Merluccius merluccius* (pls) [1];

Cottidae -*Myoxocephalus scorpius* (pls) [1], *Taurulus bubalis* (pls) [1];

Pholidae -*Pholis gunnellus* (pls) [1];

Batrachoididae -*Porichthys porosissimus* (pls) [1];

Salmonidae -*Salmo salar* (pls) [1];

Scombridae -*Scomber scombrus* (pls) [1];

Scorpaenidae -*Scorpaena plumieri* (pls) [1];

			Sebastidae -*Sebastes norvegicus* (pls) [1], *Sebastes mentella* (pls) [1], *Sebastes viviparous* (pls) [1]; **Soleidae** -*Solea solea* (pls) [1]; **Carangidae** -*Trachurus capensis* (pls) [1], *Trachurus trachurus* (pls) [1]; **Phycidae** -*Urophycis tenuis* (pls) [1]; **Xiphiidae** -*Xiphias gladius* (pls) [1]; **Nototheniidae** -*Dissostichus eleginoides* (pld) [32,34]; **Eleginopsidae** -*Eleginops maclovinus* (pld) [33]; <u>Crustaceans</u> **Acartidae** -Acartia longiremis (pro) [1]; **Paracalanidae** -*Paracalanus parvus* (pro) [1]; **Clausiocalanidae** -*Pseudocalanus elongatus* (pro) [1]; **Temoridae** -*Temora longicornis* (pro) [1]; <u>Chondrichthyan fishes:</u> **Odontaspididae** -*Carcharias Taurus* (pls) [1]; **Centrophoridae** -*Centrophorus squamosus* (pls) [1]; **Rajidae** -*Dipturus batis* (pls) [1]; **Arhynchobatidae** -*Sympterygia bonapartii* (pls) [1];
Grillotia heptanchi	Lacistorhynchidae	*Dalatias licha,* *Heptranchias perlo,* *Hexanchus griseus*	<u>Teleosts:</u> **Scophthalmidae** -*Lepidorhombus Whiffiagonis* (pls) [29]; **Lotidae** -*Molva dypterygia* (pls) [1,29]; **Ophidiidae** -*Genypterus chilensis* (pls) [1]; **Trachichthyidae** -*Hoplostethus atlanticus (pls) [1];* **Sciaenidae** -*Johnius coitori* (pls) [1]; **Trichiuridae** -*Lepidopus caudatus* (pls) [1]; **Lophiidae** -*Lophius piscatorius* (pls) [1];

			Merlucciidae -*Macruronus magellanicus* (pls) [1], *Merluccius australis* (pls) [1,47], *Merluccius capensis* (pls) [1], *Merluccius gayi gayi* (pls) [1], *Merluccius merluccius* (pls) [1], *Merluccius paradoxus* (pls) [1]; **Gadidae** -*Merlangius merlangus* (pls) [1], *Gadus chalcogrammus* (pls) [1]; **Centrarchidae** -*Micropterus salmoidesi* (pls) [1]; **Hexagrammidae** -*Ophiodon elongatus* (pls) [1]; Chondrichthyan fishes: **Hexanchidae** -*Hexanchus griseus* (pls) [1];
Grillotia smaris-gora	Lacistorhynchidae	*Squatina californica, Squatina squatina*	Teleosts: **Gadidae** -*Microgadus tomcod* (pls) [1]; **Paralichthyidae** -*Paralichthys dentatus* (pls) [1]; **Centracanthidae** -*Spicara maena* (pls) [1], *Spicara smaris* (pls) [1]; **Sparidae** -*Stenotomus chrysops* (pls) [1]; **Carangidae** -*Trachurus sp.* (pls) [1];
Grilotiella exile	Lacistorhynchidae	*Galeocerdo cuvier*	Teleosts: **Scombridae** -*Scomberomorus commerson* [11];
Lacistorhynchus dollfusi	Lacistorhynchidae	*Galeorhinus galeus, Mustelus californicus, Mustelus henlei, Mustelus lunulatus, Triakis semifasciata*	Teleosts: **Sciaenidae** -*Atractoscion nobilis* (pls) [1], *Genyonemus lineatus* (pls) [1], *Cheilotrema fasciatum* (pls) [1]; **Labridae** -*Choerodon cyanodus* (pls) [1]; **Clupeidae** -*Clupea pallasii* (pls) [1]; **Embiotocidae** -*Cymatogaster aggregata* (pls) [1], *Cymatogaster sp.* (pls) [1], *Embiotoca jacksoni* (pls) [1], *Rhacochilus vacca* (pls) [1]; **Poeciliidae** -*Gambusia affinisi* (pls) [1]; **Atherinidae** -*Leuresthes tenuis* (pls) [1]; **Moronidae** -*Morone saxatilis* (pls) [1]; **Hexagrammidae** -*Ophiodon elongatus* (pls) [1]; **Paralichthyidae** -*Paralichthys adspersus* (pls) [1], *Paralichthys californicus* (pls) [1], *Citharichthys sordidus* (pld) [35-36];

			Pleuronectidae -*Platichthys stellatus* (pls) [1]; **Sebastidae** -*Sebastes atrovirens* (pls) [1], *Sebastes flavidus* (pls) [1], *Sebastes goodei* (pls) [1], *Sebastes paucispinis* (pls) [1]; **Synodontidae** -*Synodus lucioceps* (pls) [1]; Chondrichthyan fishes **Triakidae** -*Triakis semifasciata* (pls) [1]; Crustaceans: **Harpacticidae** -*Tigriopus californicus* (pro) [1];
Lacistorhynchus tenuis	Lacistorhynchidae	*Alopias vulpinus, Galeorhinus galeus, Mustelus canis, Mustelus mustelus, Squalus acanthias*	Teleosts: **Mugilidae** -*Aldrichetta forsteri* (pls) [1]; **Monacanthidae** -*Aluterus schoepfii* (pls) [1]; **Anguillidae** -*Anguilla rostrata* (pls) [1]; **Belonidae** -*Belone belonei* (pls) [1]; **Triglidae** -*Chelidonichthys cuculus* (pls) [1], *Eutrigla gurnardus* (pls) [1], *Chelidonichthys lucerna* (pls) [1], *Trigloporus lastoviza* (pls) [1], *Trigla lyra* (pls) [1], *Trigla sp.* (pls) [1]; **Clupeidae** -*Clupea harengus* (pls) [1,37]; **Congridae** -*Conger conger* (pls) [1]; **Moronidae** -*Dicentrarchus labrax* (pls) [1]; **Trachinidae** -*Echiichthys vipera* (pls) [1], *Trachinus draco* (pls) [1]; **Lotidae** -*Enchelyopus cimbrius* (pls) [1], *Molva macrophthalma* (pls) [1]; **Gadidae** -*Gadus morhua* (pls) [1], *Melanogrammus aeglefinus* (pls) [1], *Merlangius merlangus* (pls) [1], *Pollachius pollachius* (pls) [1], *Trisopterus luscus* (pls) [1]; **Poeciliidae** -*Gambusia affinis* (pls) [1]; **Gasterosteidae** -*Gasterosteus aculeatus aculeatus* (pls) [1]; **Gobiidae** -*Gobius sp.* (pls) [1]; **Labrisomidae** -*Labrisomus philippii* (pls) [1,38]; **Labridae** -*Labrus bergylta* (pls) [1], *Labrus merula* (pls) [1], *Symphodus tinca* (pls) [1], *Tautoga onitis* (pls) [1];

			Pleuronectidae -*Limanda limanda* (pls) [1]; **Lophiidae** -*Lophius piscatorius* (pls) [1]; **Scophthalmidae** - *Scophthalmus aquosus* (pls) [1]; **Merlucciidae** -*Merluccius gayi gayi* (pls) [1], *Merluccius merluccius* (pls) [1], *Merluccius gayi peruanus* (pls) [1]; **Mullidae** -*Mullus barbatus* (pls) [1]; **Cottidae** -*Myoxocephalus octodecemspinosus* (pls) [1], *Myoxocephalus Scorpius* (pls) [1]; **Atherinidae** -*Odontesthes regia* (pls) [1]; **Osmeridae** -*Osmerus eperlanus* (pls) [1]; **Scombridae** -*Scomber scombrus* (pls) [1], *Scomberomorus maculatus* (pls) [1]; **Sparidae** -*Sparus aurata* (pls) [1]; **Gempylidae** -*Thyrsites atun* (pls) [1]; **Carangidae** -*Trachurus trachurus* (pls) [1]; **Zeidae** -*Zenopsis nebulosa* (pls) [1], *Zeus faber* (pls) [1]; Chondrichthyan fishes: **Etmopteridae** -*Etmopterus spinax* (pls) [1]; **Triakidae** -*Mustelus canis* (pls) [1]; Cephalopods: **Loliginidae** -*Doryteuthis (Amerigo) pealeii* (pls) [1]; **Ommastrephidae** -*Illex illecebrosus* (pls) [1]; Crustaceans: **Acartiidae** -*Acartia (Acanthacartia) tonsa* (pro) [1]; **Harpacticidae** -Tigriopus fulvus (pro) [1];
Pseudogrillotia basipunctata	Lacistorhynchidae	*Carcharhinus amblyrhynchos*	Teleosts: **Diodontidae** -*Diodon hystrix* (pls) [1];
Pseudogrillotia epinepheli	Lacistorhynchidae	*Carcharhinus leucas*	**Serranidae** -*Epinephelus flavocaeruleus* (pls) [1], *Epinephelus marginatus* (pls) [1], *Serranus atricauda* (pls) [39]; **Muraenidae** -*Muraena Helena* (pls) [1];

Pseudogrillotia perelica	Lacistorhynchidae	*Carcharhinus leucas, Carcharhinus limbatus, Negaprion brevirostris*	Teleosts: **Carangidae** -*Caranx senegallus* (pls) [1]; **Mugilidae** -*Liza dumerili* (pls) [1], *Liza macrolepis* (pls) [1], *Liza richardsonii* (pls) [1], *Mugil cephalus* (pls) [1], *Myxus capensis* (pls) [1], *Valamugil buchanani* (pls) [1], *Valamugil cunnesius* (pls) [1], *Valamugil robustus* (pls) [1];
Pseudolacistorhynchus noodti	Lacistorhynchidae	*Ginglymostoma cirratum*	Teleosts: **Monacanthidae** -*Aluterus schoepfii* (pls) [1]; **Muraenidae** -*Gymnothorax funebris* (pls) [1]; **Mullidae** -*Pseudupeneus maculatus* (pls) [1]; **Scombridae** -*Scomberomorus maculatus* (pls) [1];
Otobothrium alexanderi	Otobothriidae	*Carcharhinus melanopterus*	Teleosts: **Belonidae** -*Tylosurus crocodilus crocodilus* (pls) [1,11,40];
Otobothrium carcharidis	Otobothriidae	*Carcharhinus amblyrhynchoides, Carcharhinus brachyurus, Carcharhinus dussumieri, Carcharhinus limbatus, Carcharhinus macloti, Carcharhinus melanopterus, Carcharhinus sealei, Carcharhinus sorrah, Rhizoprionodon acutus, Rhizoprionodon taylori, Scoliodon laticaudus, Sphyrna lewini*	Teleosts: **Balistidae** -*Abalistes stellatus* (pls) [1]; **Lethrinidae** -*Lethrinus ornatus* (pls) [1];

Otobothrium cysticum	Otobothriidae	*Carcharhinus amboinensis, Carcharhinus leucas, Carcharhinus longimanus, Carcharhinus melanopterus, Carcharhinus obscurus, Galeocerdo cuvier, Rhizoprionodon terraenovae, Sphyrna zygaena*	Teleosts: **Ophidiidae** -*Genypterus brasiliensis* (pls) [22]; **Scombridae** -*Scomberomorus commerson* (pls) [1,21], *Euthynnus alletteratus* (pls) [1,40], *Sarda sarda* (pls) [1,40], *Scomberomorus cavalla* (pls) [1,40], *Scomberomorus regalis* [40], *Scomberomorus maculatus* (pls) [1]; **Coryphaenidae** -*Coryphaena hippurus (pls)* [1,28,40]; **Acanthuridae** -*Acanthurus coeruleus (pls)* [1,40]; **Monacanthidae** -*Aluterus schoepfii (pls)* [1,40]; **Ariidae** -*Ariopsis felis* [40], *Bagre marinus* (pls) [1,40], *Neoarius graeffei* (pls) [1], *Arius latiscutatus* (pls) [1]; **Sciaenidae** -*Bairdiella chrysoura* (pls) [1,40], *Cynoscion nebulosus* (pls) [1,40], *Cynoscion regalis* (pls) [1,40], *Cynoscion arenarius* (pls) [1], *Leiostomus xanthurus* (pls) [1,40], *Micropogonias undulatus* (pls) [1,40], *Pseudotolithus elongatus* (pls) [1], *Pseudotolithus senegallus* (pls) [1], *Pseudotolithus typus* (pls) [1]; **Balistidae** -*Balistes capriscus* (pls) [1,40]; **Carangidae** -*Caranx crysos* (pls) [1,40], *Caranx senegallus* (pls) [1], *Trachurus trecae* (pls) [1]; **Sparidae** -*Diplodus sargus sargus* (pls) [1,40], *Lagodon rhomboides* (pls) [1,40], *Dentex macrophthalmus* (pls) [1], *Pagrus pagrus* (pls) [1]; **Fundulidae** -*Fundulus heteroclitus heteroclitus (pls)* [1,40]; **Triglidae** -*Lepidotrigla faurei* (pls) [1,40]; **Lobotidae** -*Lobotes surinamensis* (pls) [1,40]; **Acropomatidae** -*Neoscombrops cynodont* [40]; **Lutjanidae** -*Ocyurus chrysurus* (pls) [1,40], *Lutjanus campechanus* (pls) [1]; **Batrachoididae** -*Opsanus tau* (pls) [1,40]; **Haemulidae** -*Orthopristis chrysoptera* (pls) [1,40], *Haemulon parra* (pls) [1]; **Paralichthyidae** -*Paralichthys albigutta* (pls) [1,40], *Paralichthys dentatus* (pls) [1,40], *Paralichthys lethostigma* (pls) [1];

Stromateidae -*Peprilus paru* (pls) [1,40], Peprilus burti (pls) [1], *Peprilus triacanthus (pls) [1,40]*;
Pomatomidae -*Pomatomus saltatrix (pls)* [1,40];
Trichiuridae -*Trichiurus lepturus* (pls) [1,40];
Siluridae -*Wallago attu* (pls) [1,40];
Xiphiidae -*Xiphias gladius* (pls) [1,40];
Cepolidae -*Cepola macrophthalma* (pls) [1];
Muraenesocidae -*Cynoponticus ferox* (pls) [1];
Serranidae -*Epinephelus striatus* (pls) [1], *Mycteroperca bonaci* (pls) [1], *Sacura boulengeri* (pls) [1];
Lophiidae -*Lophius piscatorius* (pls) [1];
Merlucciidae -Merluccius capensis (pls) [1], Merluccius merluccius (pls) [1];
Monodactylidae -*Monodactylus sebae* (pls) [1];
Sphyraenidae -*Sphyraena guachancho* (pls) [1];
Uranoscopidae -*Uranoscopus scaber* (pls) [1], *Uranoscopus sp*. (pls) [1];
<u>Chondrichthyans:</u>
Carcharhinidae -*Carcharhinus melanopterus* (pls) [1,40], *Carcharhinus limbatus* (pls) [1,40], *Carcharhinus obscurus* (pls) [1,40], *Carcharhinus plumbeus* (pls) [1], *Rhizoprionodon terraenovae* (pls) [1,40];
Dasyatidae -*Dasyatis margarita* (pls) [1];
Triakidae -*Mustelus canis (pls)* [1,40], *Mustelus mustelus* (pls) [1,40];
Squalidae -*Squalus acanthias* (pls) [1,40];
Lamnidae -*Carcharodon carcharias* (pls) [1];
<u>Cephalopods:</u>
Ommastrephidae -*Illex illecebrosus* (pls) [1];
Loliginidae -*Doryteuthis pealeii* (pls) [1,40];
<u>Reptiles:</u>
Crocodylldae -*Osteolaemus tetraspis* (pls) [1];
Cheloniidae -*Eretmochelys imbricata* (pls) [1], *Chelonia mydas* (pls) [1];

Otobothrium insigne	Otobothriidae	*Carcharhinus leucas, Carcharhinus limbatus, Carcharhinus obscurus, Rhizoprionodon terraenovae, Sphyrna tudes*	<u>Teleosts:</u> **Ariidae** -*Ariopsis felis* (pls) [1,40], *Arius sp.* (pls) [1]; **Balistidae** -*Balistes polylepis* [40]
Otobothrium minutum	Otobothriidae	*Carcharhinus limbatus, Carcharhinus macloti, Rhizoprionodon acutus*	<u>Teleosts:</u> **Carangidae** -*Parastromateus niger* (pls) [1];
Otobothrium mugilis	Otobothriidae	*Carcharhinus limbatus, Sphyrna mokarran*	<u>Teleosts:</u> **Mugilidae** -*Mugil cephalus* (pls) [1,40]; **Ariidae** -*Neoarius graeffei* (pls) [1,40], *Netuma thalassina* (pls) [1], *Arius sp.* (pls) [1]; **Sciaenidae** -*Otolithes ruber* (pls) [1,40]; **Clupeidae** -*Tenualosa ilisha* (pls) [1];
Otobothrium penetrans	Otobothriidae	*Carcharhinus leucas, Carcharhinus limbatus, Carcharhinus longimanus, Carcharhinus melanopterus, Carcharodon carcharias, Negaprion brevirostris, Rhizoprionodon terraenovae, Sphyrna lewini, Sphyrna zygaena*	<u>Teleosts:</u> **Belonidae** -*Tylosurus crocodilus crocodilus* (pls) [1,11,40,56], *Tylosurus acus acus* (pls) [1,40], *Platybelone sp.* (pls) [1,40]; **Hemiramphidae** -*Hyporhamphus dussumieri* (pls) [1,40];
Poecilancistrium caryophyllum	Otobothriidae	*Carcharhinus brachyurus, Carcharhinus leucas,*	<u>Teleosts:</u>

		Carcharhinus limbatus, Negaprion brevirostris, Rhizoprionodon acutus, Rhizoprionodon terraenovae	**Sciaenidae** -*Micropogonias furnieri* [20], Micropogonias altipinnis (pls) [1], *Micropogonias undulatus* (pls) [1]; *Macrodon ancylodon* (pld) [41] *Argyrosomus hololepidotus* (pls) [1], *Argyrosomus japonicus* (pls) [1], *Bairdiella chrysoura* (pls) [1], *Cilus gilberti* (pls) [1], *Cynoscion arenarius* (pls) [1], *Cynoscion nebulosus* (pls) [1], *Cynoscion nothus* (pls) [1], *Cynoscion regalis* (pls) [1], *Leiostomus xanthurus* (pls) [1], *Menticirrhus americanus* (pls) [1], *Nibea maculata* (pls) [1], *Pennahia anea* (pls) [1], *Pennahia argentata* (pls) [1], *Pogonias cromis* (pls) [1], *Protonibea diacanthus* (pls) [1], *Pseudotolithus senegalensis* (pls) [1], *Sciaenops ocellatus* (pls) [1], *Umbrina coroides* (pls) [1]; **Polynemidae** -*Eleutheronema tetradactylum* (pls) [1], *Polydactylus opercularis* (pls) [1]; **Serranidae** -*Epinephelus coioides* (pls) [1]; **Latidae** -*Lates calcarifer* (pls) [1]; **Pomatomidae** -*Pomatomus saltatrix* (pls) [1]; **Sillaginidae** -*Sillago robusta* (pls) [1]; **Clupeidae** -*Tenualosa ilisha* (pls) [1]; <u>Crustaceans:</u> **Acartiidae** -*Acartia (Acanthacartia) tonsa* (pro) [1]; **Pseudodiaptomidae** -*Pseudodiaptomus sp.* (pro) [1]; **Harpacticidae** -*Tigriopus californicus* (pro) [1];
Proemotobothrium southwelli	Otobothriidae	*Carcharhinus limbatus*	<u>Teleosts:</u> **Sciaenidae** -*Johnius borneensis* (pls) [1,11]; **Istiophoridae** -*Istiophorus platypterus* (pls) [1];
Pterobothrium pearsoni	Pterobothriidae	*Mustelus manazo*	<u>Teleosts:</u> **Sphyraenidae** -*Sphyraena jello* [11], ***Polynemidae*** -*Eleutheronema tetradactylum* [16]; **Sciaenidae** -*Cynoscion virescens* (pls) [1], *Otolithes ruber* (pls) [1], *Protonibea diacanthus* (pls) [1]; **Leiognathidae** -*Gazza minuta* (pls) [1];

			Carangidae -*Pseudocaranx dentex* (pls) [1]; **Scombridae** -*Scomberomorus guttatus* (pls) [1]; **Clupeidae** -*Tenualosa ilisha* (pls) [1];
Pintneriella musculicola	Rhopalothylacidae	*Carcharias taurus*	Teleosts: **Sparidae** -*Chrysophrys auratus* (pls) [1]; **Serranidae** -*Epinephelus akaara* (pls) [1], *Epinephelus chlorostigma* (pls) [1], *Epinephelus tauvina* (pls) [1]; **Lethrinidae** -*Lethrinus nebulosus* (pls) [1];
Hepatoxylon megacephalum	Sphyriocephalidae	*Carcharodon carcharias,* *Notorynchus cepedianus,* *Prionace glauca*	Teleosts: **Gadidae** -*Pollachius virens* (pld) [1]; **Scorpaenidae** -*Scorpaena porcus* (pld) [1]; **Trichomycteridae** -*Trichomycterus punctulatus* (pld) [1]; **Xiphiidae** -*Xiphias gladius* (pld) [1]; Chondrichthyan fishes: **Squalidae** -*Squalus acanthias* (pld) [1], *Squalus megalops* (pld) [1]; **Squatinidae** -*Squatina australis* (pld) [1]; **Torpedinidae** -*Torpedo marmorata* (pld) [1]; **Scyliorhinidae** -*Scyliorhinus canicula* (pld) [1], *Scyliorhinus stellaris* (pld) [1]; **Sphyrnidae** -*Sphyrna zygaena* (pld) [1]; **Carcharhinidae** -*Carcharhinus obscurus* (pld) [1], *Prionace glauca* (pld) [1]; **Dalatiidae** -*Dalatias licha* (pld) [1]; **Centrophoridae** -*Deania calcea* (pld) [1]; **Rajidae** -*Dipturus oxyrinchus* (pld) [1], *Raja clavata* (pld) [1], *Raja sp.* (pld) [1]; **Etmopteridae** -*Etmopterus spinax* (pld) [1]; **Triakidae** -*Galeorhinus galeus* (pld) [1], *Mustelus mustelus* (pld) [1]; **Pentanchidae** -*Galeus melastomus* (pld) [1]; **Hexanchidae** -*Heptranchias perlo* (pld) [1], *Hexanchus griseus* (pld) [1], *Notorynchus cepedianus* (pld) [1]; **Lamnidae** -*Isurus oxyrinchus* (pld) [1];

Hepatoxylon trichiuri	Sphyriocephalidae	*Alopias vulpinus, Carcharodon carcharias, Isurus oxyrinchus, Lamna nasus, Prionace glauca, Somniosus microcephalus*	Teleosts: **Diodontidae** -*Diodon hystrix* [11]; **Istiophoridae** -*Tetrapturus angustirostris* [11]; **Scombridae** -*Thunnus obesus* [11], *Thunnus alalunga* (pld) [1], *Thunnus albacares* (pld) [1], *Thunnus thynnus* (pld) [52], *Katsuwonus pelamis* (pld) [1], *Scomber japonicus* (pld) [1]; **Ophidiidae** -*Genypterus brasiliensis* (pld) [1,22], *Genypterus blacodes* (pld) [1], *Genypterus chilensis* (pld) [1]; **Coryphaenidae** -Coryphaena hippurus (pld) [1,28]; **Nototheniidae** -Dissostichus eleginoides (pld) [34,48]; **Alepisauridae** -*Alepisaurus ferox* (pld) [1]; **Argentinidae** -*Argentina elongata* (pld) [1]; **Berycidae** -*Beryx splendens* (pld) [1]; **Bramidae** -*Brama brama* (pld) [1], *Taractes rubescens* (pld) [1], *Taractichthys steindachneri* (pld) [1], *Brama australis* [1], *Unidentified bramid* (pld) [1]; **Macrouridae** -*Coelorinchus australis* (pld) [1], *Coelorinchus chilensis* (pld) [51], *Lepidorhynchus denticulatus* (pld) [1]; **Serranidae** -*Caesioperca lepidoptera* (pld) [1], *Lepidoperca pulchella* (pld) [1]; **Cyttidae** -*Cyttus novaezealandiae* (pld) [1], *Cyttus traversi* (pld) [1]; **Gadidae** -*Gadus morhua* (pld) [1], *Melanogrammus aeglefinus* (pld) [1], *Micromesistius australis* (pld) [1,47], *Pollachius virens* (pld) [1]; **Gempylidae** -*Gempylus serpens* (pld) [1,56], *Rexea solandri* (pld) [1], *Thyrsites atun* (pld) [1]; **Geotriidae** -*Geotria australis* (pld) [1]; **Pleuronectidae** -*Hippoglossus hippoglossus* (pld) [1]; **Hoplichthyidae** -*Hoplichthys haswelli* (pld) [1]; **Trachichthyidae** -*Hoplostethus atlanticus* (pld) [1]; **Centrolophiidae** -*Hyperoglyphe antarctica* (pld) [1]; **Haemulidae** -*Isacia conceptionis* (pld) [1];

Trichiuridae -*Lepidopus caudatus* (pld) [1];

Merlucciidae -*Macruronus novaezelandiae* (pld) [1], *Macruronus magellanicus* (pld) [46-47], *Merluccius australis* (pld) [1,45,47], *Merluccius capensis* (pld) [1], *Merluccius gayi gayi* (pld) [1,47], *Merluccius hubbsi* (pld) [1], *Merluccius merluccius* (pld) [1], *Merluccius paradoxus* (pld) [1], *Merluccius polli* (pld) [1];

Lotidae -*Molva sp.* (pld) [1];

Moridae -*Mora moro* (pld) [1], *Pseudophycis bachus* (pld) [1];

Cheilodactylidae -*Nemadactylus macropterus* (pld) [1];

Salmonidae -*Oncorhynchus keta* (pld) [1], *Oncorhynchus tshawytscha* (pld) [1], *Salmo salar* (pld) [1], *Salmo carpio* (pld) [1], *Salmo trutta trutta* (pld) [1];

Oplegnathidae -*Oplegnathus conwayi* (pld) [1];

Polyprionidae -*Polyprion oxyge*neios (pld) [1];

Scophthalmidae -*Scophthalmus maximus* (pld) [1];

Sebastidae -*Sebastes norvegicus* (pld) [1], *Sebastes mentella* (pld) [1];

Carangidae -*Seriola lalandi* (pld) [1], *Trachurus murphyi* (pld) [1];

Trachipteridae -*Trachipterus arcticus* (pld) [1];

Xiphiidae -*Xiphias gladius* (pld) [1,43];

Notacanthidae -*Notacanthus sexspinis* [51];

Chondrichthyan fishes:

Alopiidae -*Alopias vulpinus* (pld) [1], *Alopias superciliosus* (pld) [1];

Carcharhinidae -*Carcharhinus plumbeus* (pld) [1], *Prionace glauca* (pld) [1,42];

Lamnidae -*Carcharodon carcharias* (pld) [1], *Isurus oxyrinchus* (pld) [1], *Lamna nasus* (pld) [1];

Somniosidae -*Centroscymnus coelolepis* (pld) [1], *Centroscymnus owstonii* (pld) [1], *Somniosus microcephalus* (pld) [1]; *Somniosus pacificus* (pld) [1];

Chlamydosechalidae -*Chlamydoselachus anguineus* (pld) [1];

Dalatiidae -*Dalatias licha* (pld) [1];

Centrophoridae -*Deania calcea* (pld) [1];

Triakidae -*Galeorhinus galeus* (pld) [1], *Mustelus mustelus* (pld) [1];

			Pentanchidae -*Galeus melastomus* (pld) [1]; **Hexanchidae** -*Hexanchus griseus* (pld) [1]; **Squalidae** -*Squalus acanthias* (pld) [1], *unidentified squalid* (pld) [1]; **Torpedinidae** -*Tetronarce fairchildi* (pld) [1]; **Scyliorhinidae** -*Scyliorhinus canicula* (pld) [1]; Cephalopods: **Architeuthidae** -*Architeuthis dux* (pld) [1]; **Ommastrephidae** -*Illex argentinus* (pld) [1], *Sthenoteuthis pteropus* (pld) [1], *Todarodes angolensis* (pld) [1], *Dosidicus gigas* (pld) [49-50]; Mammals: **Hominidae** -*Homo sapiens* (pld) [1];
Heterosphyriocephalus tergestinus	Sphyriocephalidae	*Alopias vulpinus,* *Euprotomicrus bispinatus,* *Isurus oxyrinchus*	Teleosts: **Trichiuridae** -*Aphanopus carbo* (pld) [1], *Lepidopus caudatus* (pld) [1], *Aphanopus carbo* [54]; **Bramidae** -*Brama brama* (pld) [1], *Brama dussumieri* (pld) [1,56], *Taractichthys steindachneri* (pld) [1]; **Merlucciidae** -*Macruronus novaezelandiae* (pld) [1]; **Carangidae** -*Trachurus picturatus* (pld) [1]; **Congridae** -*Conger conger* [53]; **Scombridae** -*Sarda chiliensis* (pld) [55];
Sphyriocephalus dollfusi	Sphyriocephalidae	*Alopias superciliosus*	Teleosts: **Alepisauridae** -*Alepisaurus ferox* (pld) [1,56]; **Bramidae** -*Taractichthys steindachneri* (pld) [1]; **Scombridae** -*Thunnus obesus* (pld) [1];
Sphyriocephalus viridis	Sphyriocephalidae	*Alopias superciliosus,* *Alopias vulpinus,* *Centrophorus granulosus,*	Teleosts: **Xiphiidae** -*Xiphias gladius* [43]; **Alepocephalidae** -*Alepocephalus rostratus* (pld) [1]; **Synaphobranchidae** -*Synaphobranchus brevidorsalis* (pld) [1]; **Macrouridae** -*Trachyrincus scabrus* (pld) [1];

		Dalatias licha, Isurus oxyrinchus	<u>Chondrichthyan fishes:</u> **Centrophoridae** -*Centrophorus granulosus* (pld) [1]; **Somniosidae** -*Centroscymnus coelolepis* (pld) [1]; **Dalatiidae** -*Dalatias licha* (pld) [1]; **Pentanchidae** -*Galeus melastomus* (pld) [1]; **Pseudotriakidae** -*Pseudotriakis microdon* (pld) [1]; **Squalidae** -*Squalus acanthias* (pld) [1];
Heteronybelinia estigmena	Tentaculariidae	*Carcharhinus amblyrhynchoides, Carcharhinus leucas, Carcharhinus limbatus, Carcharhinus obscurus, Prionace glauca*	<u>Teleosts:</u> **Scombridae** -*Sarda australis* (pld) [1,11], *Scomberomorus maculatus* (pld) [1], *Thunnus albacares* (pld) [1]; **Carangidae** -*Atule mate* [11], *Selar crumenophthalmus* (pld) [1,11], *Alectis alexandrina* (pld) [1], *Caranx rhonchus* (pld) [1], *Selene setapinnis* (pld) [1], *Seriola dumerili* (pld) [1], *Trachurus murphyi* (pld) [1]; **Sciaenidae** *Cynoscion jamaicensis* (pld) [1,20], *Genyonemus lineatus* (pld) [1], *Otolithes ruber* (pld) [1]; **Clupeidae** -*Herklotsichthys quadrimaculatus* [11]; **Sphyraenidae** - *Sphyraena putnamae* [11], *Sphyraena guachancho* (pld) [1]; **Trichiuridae** -*Trichiurus lepturus* [11]; **Coryphaenidae** -*Coryphaena hippurus* (pld) [1,28], *Coryphaena equiselis* (pld) [1,28]; **Monacanthidae** -*Aluterus monoceros* (pld) [1]; **Sparidae** -*Boops boops* (pld) [1]; **Bramidae** -*Brama dussumieri* (pld) [1,56], *Taractichthys steindachneri* (pld) [1], *Unidentified Bramid* (pld) [1]; **Echeneidae** -*Echeneis naucrates* (pld) [1], *Remora sp*. (pld) [1]; **Serranidae** -*Epinephelus fasciatus* (pld) [1]; **Fistulariidae** -*Fistularia tabacaria* (pld) [1]; **Haemulidae** -*Haemulon plumierii* (pld) [1], *Pomadasys incisus* (pld) [1]; **Pomatomidae** -*Pomatomus saltatrix* (pld) [1];

			Xiphiidae -*Xiphias gladius* (pld) [1];
Heteronybelinia heteromorphi	Tentaculariidae	*Sphyrna mokarran*	Teleosts: **Bothidae** -*Bothus podas* (pld) [1]; **Nemipteridae** -*Nemipterus furcosus* (pld) [1];
Heteronybelinia nipponica	Tentaculariidae	*Carcharhinus signatus,* *Sphyrna lewini*	Teleosts: **Paralichthyidae** -*Paralichthys isosceles* (pls) [18], *Pseudorhombus pentophthalmus* (pld) [1]; **Sciaenidae** -*Menticirrhus americanus* (pld) [1,20], *Umbrina canosai* (pld) [1,20]; **Ophidiidae** -*Genypterus brasiliensis* (pls) [22], *Neobythites macrops* (pld) [1]; **Argentinidae** -*Argentina kagoshimae* (pld) [1]; **Macrouridae** -*Coelorinchus caelorhincus* (pld) [1]; **Pleuronectidae** -*Eopsetta grigorjewi* (pld) [1]; **Sebastidae** -*Helicolenus dactylopterus* (pld) [1]; **Trachichthyidae** -*Hoplostethus mediterraneus mediterraneus* (pld) [1]; **Tetraodontidae** -*Sphoeroides pachygaster* (pld) [1];
Heteronybelinia overstreeti	Tentaculariidae	*Carcharhinus limbatus*	Teleosts: **Mullidae** -*Pseudupeneus maculatus* (pld) [1];
Heteronybelinia palliata	Tentaculariidae	*Notorynchus cepedianus,* *Sphyrna zygaena*	Teleosts: **Alepisauridae** -*Alepisaurus ferox* (pld) [1]; **Sciaenidae** -*Cynoscion regalise* (pld) [1]; **Paralichthyidae** -*Paralichthys dentatus* (pld) [1]; Chondrichthyan fishes: **Triakidae** -*Mustelus canis* (pld) [1];
Heteronybelinia perideraeus	Tentaculariidae	*Notorynchus cepedianus*	Teleosts: **Leiognathidae** -*Secutor ruconius* (pld) [1];
Heteronybelinia robusta	Tentaculariidae	*Carcharhinus limbatus,* *Mustelus asterias*	Teleosts: **Carangidae** -*Caranx rhonchus* (pld) [1]; **Bothidae** -*Chascanopsetta lugubris* (pld) [1];

			Lophiidae -*Lophiodes mutilus* (pld) [1], *Lophius piscatorius* (pld) [1]; **Merlucciidae** -*Merluccius capensis* (pld) [1]; **Polymixiidae** -*Polymixia nobilis* (pld) [1]; **Congridae** -*Bassanago albescens* (pld) [1]; **Peristediidae** -*Satyrichthys adeni* (pld) [1]; **Synodontidae** -*Saurida undosquamis* (pld) [1];
Heteronybelinia yamagutii	Tentaculariidae	*Carcharhinus signatus,* *Sphyrna lewini*	Teleosts: **Trichiuridae** -*Aphanopus carbo* [54], *Benthodesmus elongatus* (pld) [1]; **Berycidae** -*Beryx splendens* (pld) [1]; **Macrouridae** -*Coelorinchus flabellispinnis* (pld) [1]; **Chaunacidae** -*Chaunax pictus* (pld) [1]; **Coryphaenidae** -*Coryphaena hippurus* (pld) [1]; **Derichthyidae** -*Derichthys serpentinus* (pld) [1]; **Gempylidae** -*Gempylus serpens* (pld) [1,56], *Thyrsitoides marleyi* (pld) [1,56]; **Gonostomatidae** -*Gonostoma elongatum* (pld) [1]; **Lycoteuthidae** -*Lycoteuthis springeri* (pld) [1]; **Myctophidae** -*Metelectrona ventralis* (pld) [1]; **Nemichthyidae** -*Nemichthys scolopaceus* (pld) [1]; **Derichthyidae** -*Nessorhamphus ingolfianus* (pld) [1]; **Sternoptychidae** -*Polyipnus polli* (pld) [1]; **Polymixiidae** -*Polymixia nobilis* (pld) [1]; **Synodontidae** -*Saurida undosquamis* (pld) [1]; **Tetraodontidae** -*Sphoeroides pachygaster* (pld) [1]; Cephalopods: **Ommastrephidae** -*Sthenoteuthis oualaniensis* (pld) [1], *Sthenoteuthis pteropus* (pld) [1], *Todarodes angolensis* (pld) [1]; **Loliginidae** -*Doryteuthis (Amerigo) pealeii* (pld) [1];
Mixonybelinia californica	Tentaculariidae	*Isurus oxyrinchus*	Teleosts: **Pleuronectidae** -*Eopsetta jordani* (pld) [1];

			Sciaenidae -*Genyonemus lineatus* (pld) [1];
Mixonybelinia edwinlintoni	Tentaculariidae	*Sphyrna lewini, Sphyrna tudes*	Teleosts: **Bothidae** -*Arnoglossus sp.* (pld) [1]; **Mullidae** -*Pseudupeneus maculatus* (pld) [1]; Chondrichthyan fishes **Carcharhinidae** -*Carcharhinus melanopterus* (pld) [1], *Carcharhinus sorrah* (pld) [1]; **Rhinobatidae** -*Rhynchobatus djiddensis* (pld) [1];
Mixonybelinia lepturi	Tentaculariidae	*Alopias superciliosus, Sphyrna lewini*	Teleosts: **Alepisauridae** -*Alepisaurus ferox* (pld) [1,56]; **Coryphaenidae** -*Coryphaena hippurus* (pld) [1]; **Gempylidae** -*Gempylus serpens* (pld) [1,56], *Thyrsitoides marleyi* (pld) [1,56]; **Trichiuridae** -*Trichiurus lepturus* (pld) [1,56]; **Bramidae** -*Brama dussumieri* (pld) [56]; Cephalopods: **Ommastrephidae** -*Sthenoteuthis oualaniensis* (pld) [1];
Mixonybelinia southwelli	Tentaculariidae	*Galeocerdo cuvier*	Teleosts: **Labridae** -*Choerodon venustus* (pld) [1,11]; **Serranidae** -*Epinephelus longispinis* (pld) [1]; **Istiophoridae** -*Istiompax indica* (pld) [1]; **Trichiuridae** -*Trichiurus lepturus* (pld) [1]; **Mullidae** -*Upeneus sulphureus* (pld) [1], *Upeneus vittatus* (pld) [1];
Nybelinia africana	Tentaculariidae	*Alopias superciliosus, Carcharhinus leucas, Carcharhinus melanopterus, Carcharhinus obscurus, Mustelus canis*	Teleosts: **Gempylidae** -*Gempylus serpens* (pld) [1,56], *Thyrsitoides marleyi* (pld) [1]; **Trichiuridae** -*Trichiurus lepturus* (pld) [1,56], *Benthodesmus elongatus* (pld) [1]; **Bramidae** -*Brama dussumieri* (pld) [1,56]; **Alepisauridae** -*Alepisaurus ferox* (pld) [1,56]; **Paralepididae** -*Arctozenus risso* (pld) [1], *Lestrolepis intermedia* (pld) [1]; **Congridae** -*Conger cinereus* (pld) [1];

			Coryphaenidae -*Coryphaena hippurus* (pld) [1]; **Polynemidae** -*Galeoides decadactylus* (pld) [1]; **Mullidae** -*Mullus barbatus* (pld) [1], *Pseudupeneus maculatus* (pld) [1]; **Sparidae** -*Pagellus sp.* (pld) [1]; **Sternoptychidae** -*Polyipnus polli* (pld) [1]; **Polymixiidae** -*Polymixia nobilis* (pld) [1]; **Serranidae** -*Serranus cabrilla* (pld) [1]; **Triglidae** -*Trigla sp.* (pld) [1]; Cephalopods: **Ommastrephidae** -*Sthenoteuthis oualaniensis* (pld) [1], *Todarodes angolensis* (pld) [1];
Nybelinia anthicosum	Tentaculariidae	*Heterodontus francisci, Prionace glauca, Triakis semifasciata*	Teleosts: **Embiotocidae** -*Amphistichus rhodoterus* (pld) [1], *Cymatogaster aggregata* (pld) [1], *Hyperprosopon argenteum* (pld) [1], *Rhacochilus vacca* (pld) [1]; **Sciaenidae** -*Genyonemus lineatus* (pld) [1]; **Pleuronectidae** -*Glyptocephalus zachirus* (pld) [1], *Lyopsetta exilis* (pld) [1]; **Paralichthyidae** -*Paralichthys californicus* (pld) [1]; **Cottidae** -*Scorpaenichthys marmoratus* (pld) [1]; Chondrichthyan fishes: **Squalidae** -*Squalus acanthias* (pld) [1];
Nybelinia gopalai	Tentaculariidae	*Sphyrna lewini, Sphyrna zygaena*	Teleosts: **Macrouridae** -*Coelorinchus flabellispinnis* (pld) [1], *Ventrifossa nasuta* [1]; **Hoplichthyidae** -*Hoplichthys acanthopleurus* (pld) [1]; **Peristediidae** -*Satyrichthys adeni* (pld) [1], *Satyrichthys welchi* (pld) [1]; **Synodontidae** -*Saurida undosquamis* (pld) [1];
Nybelinia goreensis	Tentaculariidae	*Sphyrna lewini*	Teleosts: **Lethrinidae** -*Lethrinus genivittatus* [11], *Lethrinus rubrioperculatus* [11]; **Nemipteridae** -*Nemipterus furcosus* (pld) [1,11];

			Mullidae - *Parupeneus barberinus* [11], *Parupeneus multifasciatus* [11], *Upeneus vittatus* (pld) [1]; **Paralichthyidae** -*Pseudorhombus arsius* (pld) [1], *Pseudorhombus dupliciocellatus* (pld) [1];
Nybelinia indica	Tentaculariidae	*Alopias superciliosus,* *Carcharhinus leucas,* *Carcharhinus limbatus,* *Rhizoprionodon acutus*	Teleosts: **Carangidae** -*Caranx sexfasciatus* [11], *Alepes djedaba* (pld) [1], *Selar crumenophthalmus* (pld) [1]; **Diodontidae** -*Diodon hystrix* (pld) [1,11], *Diodon liturosus* (pld) [1]; **Tetraodontidae** -*Lagocephalus sceleratus* [11]; **Leiognathidae** -*Leiognathus fasciatus* [11]; **Nemipteridae** - *Nemipterus furcosus* [11], *Nemipterus japonicus* (pld) [1]; **Polynemidae** -*Eleutheronema tetradactylum* [16]; **Gempylidae** -*Gempylus serpens* (pld) [1,56]; **Balistidae** -*Balistes capriscus* (pld) [1]; **Congridae** -*Conger cinereus* (pld) [1]; **Coryphaenidae** -*Coryphaena hippurus* (pld) [1]; **Serranidae** - *Epinephelus coioides* (pld) [1], *Epinephelus tauvina* (pld) [1]; **Istiophoridae** -*Istiophorus platypterus* (pld) [1], *Istiompax indica* (pld) [1]; **Latidae** -*Lates calcarifer* (pld) [1]; **Sciaenidae** -*Pennahia anea* (pld) [1]; **Platycephalidae**- *Platycephalus indicus* (pld) [1]; **Sternoptychidae** -*Polyipnus polli* (pld) [1]; **Priacanthidae** -*Priacanthus hamrur* (pld) [1]; **Paralichthyidae** -*Pseudorhombus dupliciocellatus* (pld) [1]; **Mullidae** -*Pseudupeneus maculatus* (pld) [1], *Upeneus japonicus* (pld) [1], *Upeneus sulphureus* (pld) [1], *Upeneus tragula* (pld) [1], *Upeneus vittatus* (pld) [1]; **Synodontidae** -*Saurida undosquamis* (pld) [1], *Trachinocephalus myops* (pld) [1]; **Scombridae** -*Scomberomorus commerson* (pld) [1]; **Trichiuridae** -*Trichiurus lepturus* (pld) [1];

			Cephalopods: **Ommastrephidae** -*Sthenoteuthis pteropus* (pld) [1];
Nybelinia jayapaulazariahi	Tentaculariidae	*Rhizoprionodon terraenovae*	Teleosts: **Cynoglossidae** -*Cynoglossus sp.* (pld) [1]; **Synodontidae** -*Harpadon nehereus* (pld) [1]; **Soleidae** -*Synclidopus macleayanus* (pld) [1], *Brachirus niger* (pld) [1];
Nybelinia lingualis	Tentaculariidae	*Carcharhinus leucas, Carcharhinus limbatus, Carcharhinus melanopterus, Carcharhinus obscurus, Carcharhinus plumbeus, Carcharodon carcharias, Hexanchus griseus, Isurus oxyrinchus, Mustelus canis, Mustelus schmitti, Notorynchus cepedianus, Scyliorhinus canicula*	Teleosts: **Paralichthyidae** -*Paralichthys isosceles* (pld) [18], *Paralichthys californicus* (pld) [1]; **Trichiuridae** -*Aphanopus carbo* (pld) [1,54], *Lepidopus caudatus* (pld) [1]; **Bothidae** -*Arnoglossus imperialis* (pld) [1]; **Berycidae** -*Beryx splendens* (pld) [1]; **Bramidae** -*Brama japonica* (pld) [1], *Unidentified Bramid* (pld) [1]; **Triglidae** -*Eutrigla gurnardus* (pld) [1], *Chelidonichthys lucerna* (pld) [1], *Trigla lyra* (pld) [1]; **Congridae** -*Conger conger* (pld) [1]; **Embiotocidae** -*Cymatogaster aggregata* (pld) [1]; **Sciaenidae** -*Cynoscion leiarchus* (pld) [1], *Genyonemus lineatus* (pld) [1]; **Ammodytidae** -*Hyperoplus lanceolatus* (pld) [1]; **Merlucciidae** -*Merluccius bilinearis* (pld) [1]; **Soleidae** -*Microchirus variegatus* (pld) [1], *Pegusa lascaris* (pld) [1]; **Mullidae** -*Mullus barbatus* (pld) [1], *Mullus surmuletus* (pld) [1], *Pseudupeneus maculatus* (pld) [1]; **Salmonidae** -*Oncorhynchus gorbuscha* (pld) [1], *Oncorhynchus keta* (pld) [1]; **Phycidae** -*Phycis blennoides* (pld) [1]; **Batrachoididae** -Porichthys porosissimus (pld) [1]; **Scombridae** -*Sarda sarda* (pld) [1], *Scomber scombrus* (pld) [1], *Thunnus thynnus* (pld) [1]; **Cottidae** -*Scorpaenichthys marmoratus* (pld) [1];

			Centracanthidae -*Spicara smaris* (pld) [1]; **Tetraodontidae** -*Torquigener pleurogramma* (pld) [1]; **Carangidae** -*Trachurus capensis* (pld) [1], *Trachurus murphyi* (pld) [1], *Trachurus picturatus* (pld) [1,57], *Trachurus trachurus* (pld) [1]; Chondrichthyan fishes: **Rajidae** -*Zearaja nasuta* (pld) [1], *Raja sp*. (pld) [1]; **Arhynchobatidae** -*Sympterygia bonapartii* (pld) [1]; **Scyliorhinidae** -*Scyliorhinus canicula* (pld) [1]; **Carcharhinidae** -*Prionace glauca* (pld) [1]; Cephalopods: **Eledonidae** -*Eledone cirrhosa* (pld) [1], *Eledone moschata* (pld) [1]; **Ommastrephidae** -*Eucleoteuthis luminosa* (pld) [1], *Ommastrephes bartramii* (pld) [1], *Sthenoteuthis oualaniensis* (pld) [1], *Sthenoteuthis pteropus* (pld) [1], *Todarodes angolensis* (pld) [1], *Todaropsis eblanae* (pld) [1]; **Loliginidae** -*Loligo vulgaris* (pld) [1]; **Octopodidae** -*Octopus vulgaris* (pld) [1]; **Sepiidae** -*Sepia elegans* (pld) [1,58], *Sepia officinalis* (pld) [1];
Nybelinia pinteri	Tentaculariidae	*Prionace glauca*	Teleosts: **Paralichthyidae** -*Paralichthys olivaceus* (pld) [1];
Nybelinia queenslandensis	Tentaculariidae	*Carcharhinus melanopterus*	Teleosts: **Nemipteridae** -*Nemipterus furcosus* [11]; **Apogonidae** - *Ostorhinchus cookie* [11], *Ostorhinchus properuptus* [11];
Nybelinia strongyla	Tentaculariidae	*Sphyrna tudes*	Teleosts: **Sciaenidae** -*Johnius borneensis* (pld) [1,11], *Argyrosomus hololepidotus* (pld) [1]; **Tetraodontidae** -*Sphoeroides pachygaster* (pld) [1];
Nybelinia syngenes	Tentaculariidae	*Sphyrna zygaena*	Teleosts: **Scorpaenidae** -*Dendrochirus zebra* (pld) [1]; Chondrichthyan fishes: **Sphyrnidae** -*Sphyrna zygaena* (pld) [1];

Nybelinia thyrsites	Tentaculariidae	*Carcharhinus brachyurus, Carcharhinus longimanus, Galeorhinus galeus*	Teleosts: **Trichiuridae** -*Aphanopus carbo* [54], *Lepidopus caudatus* (pld) [1]; **Arripidae** -*Arripis truttacea* (pld) [1]; **Gempylidae** -*Thyrsites atun* (pld) [1]; **Carangidae** -*Trachurus declivis* (pld) [1], *Trachurus novaezelandiae* (pld) [1]; **Zeidae** -*Zeus faber* (pld) [1]; Chondrichthyan fishes: **Squalidae** -*Squalus sp.* (pld) [1];
Tentacularia coryphaenae	Tentaculariidae	*Carcharhinus galapagensis, Carcharhinus limbatus, Carcharhinus melanopterus, Carcharhinus obscurus, Carcharhinus plumbeus, Carcharodon carcharias, Galeocerdo cuvier, Prionace glauca, Rhizoprionodon acutus, Sphyrna zygaena*	Teleosts: **Ophidiidae** -*Genypterus brasiliensis* (pls) [22]; **Coryphaenidae** -*Coryphaena hippurus* (pld) [1,28,61], *Coryphaena equiselis* (pld) [1,28]; **Trichiuridae** -*Aphanopus carbo* (pld) [1,54], *Trichiurus lepturus* (pld) [1,56]; **Scombridae** -*Sarda chiliensis* (pld) [1,55], *Sarda sarda* (pld) [1], *Acanthocybium solandri* (pld) [1], *Euthynnus affinis* (pld) [1], *Euthynnus alletteratus* (pld) [1], *Katsuwonis pelamis* (pld) [1], *Orcynopsis unicolor* (pld) [1], *Scomber japonicus* (pld) [1], *Scomber scombrus* (pld) [1], *Scomberomorus cavalla* (pld) [1], *Scomberomorus commerson* (pld) [1], *Scomberomorus guttatus* (pld) [1], *Thunnus alalunga* (pld) [1], *Thunnus albacares* (pld) [1], *Unidentified bonito* (pld) [1]; **Gempylidae** -*Gempylus serpens* (pld) [1,56], *Thyrsitoides marleyi* (pld) [1,56], *Lepidocybium flavobrunneum* (pld) [1], *Ruvettus pretiosus* (pld) [1], *Thyrsites atun* (pld) [1,62]; **Bramidae** -*Brama dussumieri* (pld) [1,56], *Brama brama* (pld) [1], *Taractichthys steindachneri* (pld) [1]; **Alepisauridae** -*Alepisaurus ferox* (pld) [1,56]; **Sciaenidae** -*Atractoscion aequidens* (pld) [1]; **Centropomidae** -*Centropomus nigrescens* (pld) [1]; **Stomiidae** -*Chauliodus sloani* (pld) [1], *Stomias boa* (pld) [1]; **Chaunacidae** -*Chaunax pictus* (pld) [1];

Carangidae -*Decapterus sp*. (pld) [1], *Scomberoides commersonnianus* (pld) [1], *Seriola dumerili* (pld) [1], *Trachurus capensis* (pld) [1], *Trachurus murphyi* (pld) [1], *Trachurus picturatus* (pld) [1], *Trachurus declivis* (pld) [1], *Unidentified Carangid* (pld) [1];
Echeneidae -*Echeneis naucrates* (pld) [1], *Remora remora* (pld) [1];
Gadidae -*Gadus morhua* (pld) [1];
Pleuronectidae -*Hippoglossus hippoglossus* (pld) [1];
Trachichthyidae -*Hoplostethus atlanticus* (pld) [1];
Lampridae -*Lampris guttatus* (pld) [1];
Lobotidae -*Lobotes surinamensis* (pld) [1];
Lophiidae -*Lophius piscatorius* (pld) [1];
Merlucciidae -*Macruronus novaezelandiae* (pld) [1], *Merluccius capensis* (pld) [1], *Merluccius gayi gayi* (pld) [1], *Merluccius gayi peruanus* (pld) [1];
Salmonidae -*Oncorhynchus tshawytscha* (pld) [1], *Salmo salar* (pld) [1];
Paralichthyidae -*Paralichthys dentatus* (pld) [1];
Nomeidae -*Psenes cyanophrys* (pld) [1];
Polynemidae -*Polydactylus opercularis* (pld) [1];
Polyprionidae -*Polyprion oxygeneios* (pld) [1];
Rachycentridae -*Rachycentron canadum* (pld) [1];
Peristediidae -*Satyrichthys adeni* (pld) [1];
Sternoptychidae -*Sternoptyx diaphana* (pld) [1];
Istiophoridae -*Kajikia albida* (pld) [1];
Cyprinidae -*Tinca tinca* (pld) [1];
Xiphiidae -*Xiphias gladius* (pld) [1,60];
Clupeidae -*Sardinops sagax* (pld) [59];
Chondrichthyan fishes:
Carcharhinidae -*Carcharhinus obscurus* (pld) [1], *Galeocerdo cuvier* (pld) [1];
Centrophoridae -*Centrophorus moluccensis* (pld) [1], *Deania calcea* (pld) [1], *Deania profundorum* (pld) [1];

Somniosidae -*Centroscymnus coelolepis* (pld) [1];
Rajidae -*Raja sp.* (pld) [1], *Rajella caudaspinosa* (pld) [1];
Rhinobatidae -*Rhynchobatus sp.* (pld) [1];
Hexanchidae -*Heptranchias perlo* (pld) [1];
Cephalopods:
Ommastrephidae -*Dosidicus gigas* (pld) [1,49-50,63], *Illex illecebrosus* (pld) [1], *Ommastrephes bartramii* (pld) [1,64], *Sthenoteuthis pteropus* (pld) [1], *Sthenoteuthis oualaniensis* (pld) [1], *Todarodes angolensis* (pld) [1];
Sepiidae -*Sepia unguiculata* (pld) [1];
Reptiles:
Cheloniidae -*Chelonia mydas* (pld) [1];

References:

1. Palm, H. W. (2004). The trypanorhyncha diesing, 1863. PKSPL-IPB.
2. Gurney, R. H., Nowak, B. F., Dykova, I., & Kuris, A. M. (2004). Histopathological effects of trypanorhynch metacestodes in the digestive gland of a novel host, Carcinus maenas (Decapoda). Diseases of aquatic organisms, 58(1), 63-69.
3. Gurney, R. H., Johnston, D. J., & Nowak, B. F. (2006). The effect of parasitism by trypanorhynch plerocercoids (Cestoda, Trypanorhyncha) on the digestive enzyme activity of Carcinus maenas (Linnaeus, 1758)(Decapoda, Portunidae). Crustaceana, 79(6), 663-675.
4. Muscolino, D., Giarratana, F., Giuffrida, A., & Panebianco, A. (2012). Inspective Investigation on Swordfish (Xiphias gladius) frozen Slices of Commerce: Anatomical-Histopatological Findings. Czech Journal of Food Science, 30(3).
5. Giarratana, F., Muscolino, D., Beninati, C., Ziino, G., Giuffrida, A., Trapani, M., & Panebianco, A. (2014). Gymnorhynchus gigas in Lepidopus caudatus (Actinopterygii: Perciformes: Trichiuridae): Prevalence and Related Effects on Fish Quality. Czech Journal of Food Science, 32(4).
6. Gòmez-Morales, M. A., Ludovisi, A., Giuffra, E., Manfredi, M. T., Piccolo, G., & Pozio, E. (2008). Allergenic activity of Molicola horridus (Cestoda, Trypanorhyncha), a cosmopolitan fish parasite, in a mouse model. Veterinary parasitology, 157(3), 314-320.
7. Fernández, I., Oyarzún, C., Valenzuela, A., Burgos, C., Guaquín, V., & Campos, V. (2016). Parásitos del pez luna Mola mola (Pisces: Molidae). Primer registro en aguas de la costa centro sur de Chile. Gayana (Concepción), 80(2), 192-197.
8. Nunkoo, M. A. I., Reed, C. C., & Kerwath, S. E. (2016). Community ecology of the metazoan parasites of snoek Thyrsites atun (Euphrasen, 1791)(Perciformes: Gempylidae) off South Africa. African Journal of Marine Science, 38(3), 363-371.
9. Abdelsalam, M., Abdel-Gaber, R., Mahmoud, M. A., Mahdy, O. A., Khafaga, N. I., & Warda, M. (2016). Morphological, molecular and pathological appraisal of Callitetrarhynchus gracilis plerocerci (Lacistorhynchidae) infecting Atlantic little tunny (Euthynnus alletteratus) in Southeastern Mediterranean. Journal of advanced research, 7(2), 317-326.
10. Haseli, M., Azimi, S., & Valinasab, T. (2016). Microthrix pattern of Pseudogilquinia thomasi (Palm, 2000) (Cestoda: Trypanorhyncha) and a review of surface ultrastructure within the family Lacistorhynchidae Guiart, 1927. Journal of morphology, 277(3), 394-404.
11. Beveridge, I., Bray, R. A., Cribb, T. H., & Justine, J. L. (2014). Diversity of trypanorhynch metacestodes in teleost fishes from coral reefs off eastern Australia and New Caledonia. Parasite, 21, 60.
12. Pereira, A. N., Pantoja, C., Luque, J. L., & Timi, J. T. (2014). Parasites of Urophycis brasiliensis (Gadiformes: Phycidae) as indicators of marine ecoregions in coastal areas of the South American Atlantic. Parasitology research, 113(11), 4281-4292.
13. Shohreh, P., Mousavi, H. E., Soltani, M., Mobedi, I., Ghadam, M., & Mood, S. M. (2014). Trypanorhych cestodes of the tiger tooth croaker (Otolithes ruber) in the Persian Gulf. Bulletin of the European Association of Fish Pathologists, 34(4), 109.

14. Carvalho, A. R., & Luque, J. L. (2011). Seasonal variation in metazoan parasites of Trichiurus lepturus (Perciformes: Trichiuridae) of Rio de Janeiro, Brazil. Brazilian Journal of Biology, 71(3), 771-782.
15. Haseli, M., Malek, M., Valinasab, T., & Palm, H. W. (2011). Trypanorhynch cestodes of teleost fish from the Persian Gulf, Iran. Journal of Helminthology, 85(2), 215-224.
16. Zischke, M. T., Cribb, T. H., Welch, D., Sawynok, W., & Lester, R. J. G. (2009). Stock structure of blue threadfin Eleutheronema tetradactylum on the Queensland east coast, as determined by parasites and conventional tagging. Journal of Fish Biology, 75(1), 156-171.
17. Abdou, N. S., & Palm, H. W. (2008). New record of two genera of Trypanorhynch cestodes infecting Red Sea fishes in Egypt. Journal of the Egyptian Society of Parasitology, 38(1), 281-292.
18. Felizardo, N. N., Torres, E. J. L., Fonseca, M. C. G., Pinto, R. M., Gomes, D. C., & Knoff, M. (2010). Cestodes of the flounder Paralichthys isosceles Jordan, 1890 (Osteichthyes-Paralichthyidae) from the state of Rio de Janeiro, Brazil. Neotropical Helminthology, 4(2), 113-125.
19. de Freitas Ferreira, M., de São Clemente, S. C., Tortelly, R., de Lima, F. C., do Nascimento, E. R., de Oliveira, G. A., & de Resende Lima, A. (2006). Parasitas da ordem Trypanorhyncha: sua importância na inspeção sanitária do pescado. Revista Brasileira de Ciência Veterinária, 13(3), 190-193.
20. Pereira Junior, J., & Boeger, W. A. P. (2005). Larval tapeworms (Platyhelminthes, Cestoda) from sciaenid fishes of the southern coast of Brazil. Zoosystema, 27(1), 5-25.
21. Williams, R. E., & Lester, R. J. G. (2006). Stock structure of Spanish mackerel Scomberomorus commerson along the Australian east coast deduced from parasite data. Journal of Fish Biology, 68(6), 1707-1712.
22. São Clemente, S. C., Knoff, M., Padovani, R. E., Lima, F. C., & Gomes, D. C. (2004). Cestóides Trypanorhyncha parasitos de Congro-rosa, Genypterus brasiliensis Regan, 1903 comercializados nos municípios de Niterói e Rio de Janeiro, Brasil. Revista Brasileira de Parasitologia Veterinária, 13(3), 97-102.
23. Knoff, M. F., Nilza N. G., Delir C. C., Sergio C. (2016). Callitetrarhynchus speciosus (linton, 1897) carvajal & rego, 1985 trypanorhyncha (cestoda) parasitizing priacanthus arenatus (cuvier, 1829) (osteichthyes, priacanthidae) from Rio de Janeiro coast, Brazil. Neotropical Helminthology, 10(1), 33-40.
24. Dias, F. D. J. E., São Clemente, S. C. D., & Knoff, M. (2010). Larvae of Anisakidae nematodes and Trypanorhyncha cestodes of public health importance in Aluterus monoceros (Linnaeus, 1758) in Rio de Janeiro State, Brazil. Revista Brasileira de Parasitologia Veterinária, 19(2), 94-97.
25. Rizwana, A. G., Khatoon, N., & Bilqees, F. M. (2009). Studies on the infestation in relation to sex of the host fish Lutjanus argentimaculatus (Forsk., 1775). International Journal of Biology and Biotechnology, 6(1-2), 71-74.
26. Abdou, E. N. (2005). Scanning electron microscopy of the plerocercoids of Floriceps minacanthus (Cestoda: Trypanorhyncha) parasitize the fish Cephalopholis micri, a new host record in the Red Sea–Egypt. Journal of the Egyptian German Society of Zoology, 47, 133-145.

27. Abdou, N. (2005). Transmission electron microscope study of plerocercoids of two Floriceps spp. (Cestoda: Trypanorhyncha) infecting certain fishes in the Red Sea. Journal of Union of Arab Biologists, 24, 83-97.
28. Williams Jr, E. H., & Bunkley-Williams, L. (2009). Checklists of the parasites of dolphin, Coryphaena hippurus, and pompano dolphin, C. equiselis with new records, corrections, and comments on the literature. Reviews in Fisheries Science, 18(1), 73-93.
29. Beveridge, I., & Campbell, R. A. (2013). A new species of Grillotia Guiart, 1927 (Cestoda: Trypanorhyncha) with redescriptions of congeners and new synonyms. Systematic parasitology, 85(2), 99-116.
30. Özer, A., Kornyychuk, Y. M., Öztürk, T., & Yurakhno, V. (2015). Comparative Study on Parasite Fauna of the Whiting Merlangius merlangus in the Northern and Southern Zones of the Black Sea. Turkish Journal of Fisheries and Aquatic Sciences, 15(2), 283-291.
31. Özer, A., Öztürk, T., Kornyushin, V. V., Kornyychuk, Y., & Yurakhno, V. (2014). Grillotia erinaceus (van Beneden, 1858)(Cestoda: Trypanorhyncha) from whiting in the Black Sea, with observations on seasonality and host-parasite interrelationship. Acta Parasitologica, 59(3), 420-425.
32. Brown, J., Brickle, P., & Scott, B. E. (2013). The parasite fauna of the Patagonian toothfish Dissostichus eleginoides off the Falkland Islands. Journal of helminthology, 87(4), 501-509.
33. Brickle, P., & MacKenzie, K. (2007). Parasites as biological tags for Eleginops maclovinus (Teleostei: Eleginopidae) around the Falkland Islands. Journal of Helminthology, 81(2), 147-153.
34. Brickle, P., MacKenzie, K., & Pike, A. (2006). Variations in the parasite fauna of the patagonian toothfish (Dissostichus eleginoides smitt, 1898), with length, season, and depth of habitat around the falkland islands. Journal of Parasitology, 92(2), 282-291.
35. Hogue, C., & Swig, B. (2007). Habitat quality and endoparasitism in the Pacific sanddab Citharichthys sordidus from Santa Monica Bay, southern California. Journal of Fish Biology, 70(1), 231-242.
36. Courtney-Hogue, C. (2016). Heavy metal accumulation in Lacistorhynchus dollfusi (Trypanorhyncha: Lacistorhynchidae) infecting Citharichthys sordidus (Pleuronectiformes: Bothidae) from Santa Monica Bay, southern California. Parasitology, 143(6), 794-799.
37. Campbell, N., Cross, M. A., Chubb, J. C., Cunningham, C. O., Hatfield, E. M., & MacKenzie, K. (2007). Spatial and temporal variations in parasite prevalence and infracommunity structure in herring (Clupea harengus L.) caught to the west of the British Isles and in the North and Baltic Seas: implications for fisheries science. Journal of helminthology, 81(2), 137-146.
38. Cruces, C., Chero, J., Iannacone, J., Sáez, G., & Alvariño, L. (2017). Community of endohelminth parasites of yellowmouth blenny labrisomus philippii (steindachner, 1866) (perciformes: labrisomidae) from the central coast of peru. The Biologist (Lima), 13(1), 91-109.
39. Costa, G., Khadem, M., Silva, S., Moreira, E. M., & Amélio, S. D. (2013). Endohelminth parasites of the blacktail comber Serranus atricauda (Pisces: Serranidae), from Madeira Archipelago (Atlantic Ocean). Diseases of aquatic organisms, 103(1), 55-64.

40. Schaeffner, B. C., & Beveridge, I. (2013). Redescriptions and new records of species of Otobothrium Linton, 1890 (Cestoda: Trypanorhyncha). Systematic parasitology, 84(1), 17-55.
41. Oliveira, S., São Clemente, S., Benigno, R., & Knoff, M. (2009). Poecilancistrium caryophyllum (Diesing, 1850)(Cestoda, Trypanorhyncha), parasite of Macrodon ancylodon (Bloch & Schneider, 1801) from the Northern littoral of Brazil. Revista Brasileira de Parasitologia Veterinária, 18(4), 71-73.
42. Borucinska, J. D., & Bogicevic, T. (2004). Gastric polyp in a wild-caught blue shark. Journal of Aquatic Animal Health, 16(1), 39-44.
43. Mattiucci, S., Garcia, A., Cipriani, P., Santos, M. N., Nascetti, G., & Cimmaruta, R. (2014). Metazoan parasite infection in the swordfish, Xiphias gladius, from the Mediterranean Sea and comparison with Atlantic populations: implications for its stock characterization. Parasite, 21, 35.
44. Oliva, M. E., Espinola, J. F., & Ñacari, L. A. (2016). Metazoan parasites of Brama australis from southern Chile: a tool for stock discrimination?. Journal of fish biology, 88(3), 1143-1148.
45. Torres, P., Puga, S., Castillo, L., Lamilla, J., & Miranda, J. C. (2014). Helmintos, myxozoos y microsporidios en músculos de peces comercializados frescos y su importancia como riesgo potencial para la salud humana en la ciudad de Valdivia, Chile. Archivos de medicina veterinaria, 46(1), 83-92.
46. MacKenzie, K., Brickle, P., Hemmingsen, W., & George-Nascimento, M. (2013). Parasites of hoki, Macruronus magellanicus, in the Southwest Atlantic and Southeast Pacific Oceans, with an assessment of their potential value as biological tags. Fisheries research, 145, 1-5.
47. Chávez, R. A., González, M. T., Oliva, M. E., & Valdivia, I. M. (2012). Endoparasite fauna of five Gadiformes fish species from the coast of Chile: host ecology versus phylogeny. Journal of helminthology, 86(1), 10-15.
48. Oliva, M. E., Fernández, I., Oyarzún, C., & Murillo, C. (2008). Metazoan parasites of the stomach of Dissostichus eleginoides Smitt 1898 (Pisces: Notothenidae) from southern Chile: A tool for stock discrimination?. Fisheries Research, 91(2), 119-122.
49. Pardo-Gandarillas, M. C., Lohrmann, K. B., Valdivia, A. L., & Ibáñez, C. M. (2009). First record of parasites of Dosidicus gigas (d'Orbigny, 1835)(Cephalopoda: Ommastrephidae) from the Humboldt Current system off Chile. Revista de Biología Marina y Oceanografía, 44(2), 397-408
50. Céspedes, R. E., Iannacone, J., & Salas, A. (2011). Helmintos parásitos de Dosidicus gigas" Pota" eviscerada en Arequipa, Perú. Ecología Aplicada, 10(1), 1-11.
51. Pardo-Gandarillas, M. C., González, K., Ibáñez, C. M., & George-Nascimento, M. (2008). Parasites of two deep-sea fish Coelorynchus chilensis (Pisces: Macrouridae) and Notacanthus sexspinis (Pisces: Notacanthidae) from Juan Fernández Archipelago, Chile. Marine Biodiversity Records, 1, 1-5.
52. Mladineo, I. (2006). Hepatoxylon trichiuri (Cestoda: Trypanorhyncha) plerocercoids in cage-reared northern bluefin tuna, Thunnus thynnus (Osteichthyes: Scombridae). ACTA adriatica, 47(1), 79-83.
53. Costa, G., Santos, M. J., Costa, L., Biscoito, M., de Carvalho, M. A. P., & Melo-Moreira, E. (2009). Helminth parasites from the stomach of conger eel, Conger conger, from Madeira Island, Atlantic Ocean. Journal of Parasitology, 95(4), 1013-1015.

54. Santos, M. J., Saraiva, A., Cruz, C., Eiras, J. C., Hermida, M., Ventura, C., & Soares, J. P. (2009). Use of parasites as biological tags in stock identification of the black scabbardfish, Aphanopus carbo Lowe, 1839 (Osteichthyes: Trichiuridae) from Portuguese waters. Scientia Marina, 73(S2), 55-62.
55. Chero, J., Sáez, G., Iannacone, J., Cruces, C., Alvariño, L., & Luque, J. (2016). Ecología Comunitaria de Metazoos Parásitos del Bonito Sarda chiliensis Cuvier, 1832 (Perciformes: Scombridae) de la Costa Peruana. Revista de Investigaciones Veterinarias del Perú, 27(3), 539-555.
56. Jakob, E., & Palm, H. W. (2006). Parasites of commercially important fish species from the southern Java coast, Indonesia, including the distribution pattern of trypanorhynch cestodes. Verhandlungen der Gesellschaft fr Ichthyologie, 5, 165-191.
57. Costa, G., Melo-Moreira, E., & de Carvalho, M. P. (2012). Helminth parasites of the oceanic horse mackerel Trachurus picturatus Bowdich 1825 (Pisces: Carangidae) from Madeira Island, Atlantic Ocean, Portugal. Journal of helminthology, 86(3), 368-372.
58. Bello, G. (2004). Sepia elegans (Cephalopoda: Sepiidae): a new host record for the parasite Nybelinia lingualis (Cestoda: Tentaculariidae). Atti della Società italiana di scienze naturali e del museo civico di storia naturale di Milano, 145(1), 225-228.
59. Reed, C., MacKenzie, K., & Van der Lingen, C. D. (2012). Parasites of South African sardines, Sardinops sagax, and an assessment of their potential as biological tags. Bulletin of the European Association of Fish Pathologists, 32(2), 41-48.
60. Muñoz G., García N. and Valdebenito V. (2012). Gastric helminths in the swordfish Xiphias gladius collected off the coast of central-south Chile. XI European Multicolloquium of Parasitology, 11, 55-58.
61. da Silva, A. M., Clemente, S. D. S., da Fonseca, M. C. G., Gomes, D. C., Justo, M. C. N., & Knoff, M. (2017). Morphological characters and hygienic-sanitary significance of Tentacularia coryphaenae in Coryphaena hippurus from Brazil. Boletim do Instituto de Pesca, 43(2), 266-273.
62. Nunkoo, M. A. I., Reed, C. C., & Kerwath, S. E. (2016). Community ecology of the metazoan parasites of snoek Thyrsites atun (Euphrasen, 1791)(Perciformes: Gempylidae) off South Africa. African Journal of Marine Science, 38(3), 363-371.
63. Iannacone, J. & Alvariño, L. (2009). Catastre of endoparasite fauna of jumbo flying squid Dosidicus gigas (Cephalopoda) in the north of Peru.|Catastro de la fauna endoparasitaria de la pota Dosidicus gigas (Cephalopoda) en el norte del Peru. Neotropical Helminthology, 2009, 3(2), 89-100.
64. Nigmatullin, C. M., Shchetinnikov, A. S., & Shukhgalter, O. A. (2009). On feeding and helminth fauna of neon flying squid Ommastrephes bartramii (Lesueur, 1821)(Cephalopoda: Ommastrephidae) in the southeastern Pacific. Revista de Biología Marina y Oceanografía, 44(1).