DIVERSITY AND ABUNDANCE OF SELECTED BITING FLIES IN A PERIDOMESTIC AND NATURAL HABITATS, AND THEIR POTENTIAL AS A PROTEIN SOURCE FOR FEED IN KIBWEZI WEST SUBCOUNTY, MAKUENI COUNTY, KENYA

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DECLARATION

Student

I, Fridah Karegi Njeru, hereby declare that, this thesis is my original work and has not been submitted elsewhere for examination, award of a degree or publication. Where other people's work has been used, this has been properly acknowledged and referenced in accordance with the requirements of the University of Nairobi.

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DEDICATION

To my late dad, family and friends

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ABBREVIATIONS AND ACRONYMS

AOAC -Association of Analytical Chemists

BT—Blue Tongue

CP—Crude Protein

DDT – Dichlorodiphenyltrichloroethane

ECI- Efficiency of Conversion of Ingested food

FAO – Food and Agriculture Organization

MUFA- Monounsaturated Fatty Acid

PUFA – Polyunsaturated Fatty Acids

SFA- Saturated Fatty Acid

SIT – Sterile Insect Technique

Abstract

Biting flies are a diverse assemblage of blood-sucking flies that are important vectors of human, livestock and wildlife diseases. They may be found in domestic or peridomestic habitats, where they are associated with humans and livestock, as well as in natural habitats away from human habitation. A study was carried out to determine the diversity and abundance of biting flies in a peridomestic and a natural habitat in Kiboko, Makueni County, Kenya. Nutritional profiles of these flies were also assessed to determine their potential as sources of protein. Wild biting flies were caught from a natural habitat (Kibwezi forest) and a peridomestic habitat (Woni farm) during a dry season and a wet season using four commonly used traps (Nzi, Ngu, Pyramidal and Biconical). Performance of the traps was assessed in replicated 4x4 Latin Square experiments. Proximate analysis was done on fly species caught in the highest numbers (*Glossina pallidipes)*, to determine its potential as cheap source of proteins for feed. Trap catches varied significantly among the traps with the highest catch recorded in Nzi trap (3,235) and the lowest in Pyramidal trap (959). However, Pyramidal trap recorded the least diverse catch with 14 taxa and a Shannon diversity index of H′= 0.66. Among the insect species collected, Nzi trap recorded the highest number of *G. pallidipes* (26,552 individuals). Habitat type significantly affected insect community ($F_{1,\ 3}$=15.2786; p=0.005). There was no evidence of effect of Season ($F_{1,\ 3}$=01.66; p=0.225) and trap type ($F_{1,\ 3}$=00.496; p=00.75) on insect community. Diversity and abundance of insect species collected varied among seasons both in peridomestic and natural habitat. Nutrient contents (protein, fat, moisture, fibre) varied among the groups of insects tested (biting flies and moths/ butterflies). Protein content of biting flies was 63.02% higher that of Omena (*Rastrineobola argentea)* which varies between 19.1 and 21.7% while moths/butterflies had 63.29%. The high protein content among other nutrients observed in biting flies and lepidopterans during this study suggests that they can be used as an alternative protein source in fish and chicken feed.

CHAPTER ONE

1.0 INTRODUCTION

1.1 Background of study

Biting flies loosely refers to a diverse assemblage of dipterans that feed on blood of vertebrates. Some of them are important vectors of human and livestock diseases all over the world (Rodhain, 2015), which they transmit mechanically and/or biologically through their bites. Some of the important biting flies from a medical or veterinary point of view include mosquitoes (Culicidae) known to transmit malaria, among other diseases, tsetse flies (Glossinidae) transmitting Human African Trypanosomiasis (sleeping sickness) and animal trypanosomiasis (Nagana), Phlebotomine (Sandflies), the vector of Leishmaniasis disease to humans and black flies (Simuliidae), the vectors of Onchocerciasis (filariasis) or river blindness (Birley,1993). Other important biting flies include deer flies, horse flies and yellow flies (Tabanidae), stable flies (Muscidae), snipe flies (Rhagionidae) and biting midges (Ceratopogonidae). Many biting flies have a cosmopolitan distribution with their respective numbers reportedly varying among different habitats depending on ecological conditions (Walsh, *et al*., 1993).

Knowledge of vector composition (diversity), abundance and distribution in areas where vector-borne diseases are endemic is an important aspect in the epidemiology of these diseases. This biological information is critical for successful management of the vectors and the diseases they transmit as it helps to improve the efficacy of the existing management techniques.

One of the most important livestock disease in the Kiboko area of Makueni County is trypanosomiasis, which is caused by a Kinetoplastida protozoan parasite of the genus *Trypanosoma* and transmitted cyclically by tsetse (*Glossina*) and mechanically by other hematophagous biting flies such as *Tabanus*, *Haematopota*, *Stomoxys*, and *Chrysops* (Koné *et al*., 2011; Cecchi *et al*., 2015). A prevalence of trypanosomiasis in livestock of 3.9% has was reported by Mochabo *et al*., (2010) in Makueni county, with more recent studies (Nthiwa *et al.*, 2015) reporting a trypanosome infection rate of 11.5% in *Glossina* spp. Current vector management strategies include reduction of biting flies densities using traps. It has been demonstrated in the past that some traps are capable of catching biting flies in very large numbers. Such catches may be better utilized to feed poultry directly or processed to supplement protein in feeds. However, to achieve the aforementioned, it important to profile nutritional composition of insects. It is noteworthy that current sources of proteins in feeds, which include soya beans and fish or fish products, are expensive. This raises production cost of feeds making them unaffordable to many farmers.

Human consumption of insects (entomophagy) has been practiced in many tropical countries in Africa (Walkley, 1952). Close to 2,000 edible insect species have been reported to be consumed worldwide (Ramos-Elorduy and Paoletti, 2005). These insect species belong to more than 14 described orders. They are consumed at various developmental stages; egg, larvae, pupae and adult. Some insects are consumed directly at more than one stage. For example, *Apis mellifera* is consumed directly at its egg, larvae and pupae stages with high levels of protein content being recorded in all the three stages. Under indirect entomophagy, target insects may be fed directly to animals such as poultry or fish, or processed as a feed component.

During processing, insects are dried, crushed as parts or whole or protein component extracted and incorporated into the feed as a supplement. It is on this background that this study was conceptualized and undertaken to determine diversity, abundance and potential of biting flies as alternative or supplemental source of protein in feed.

1.2 Problem statement and justification of study

Biting flies are important vectors of human and animal diseases in tropical Africa. Due to their economic importance, several management interventions have been initiated with limited success. Their persistence in different habitats is attributed to adaptation to colonize different ecological conditions. In Makueni, the selected study area, there is limited information of the diversity and abundance of biting flies in the both peridomestic and natural habitats. Lack of information on the diversity and abundance of biting flies in Makueni coupled with desire to develop sustainable biting fly management strategy informed the choice of this study.

High cost of poultry feeds has awakened interest in the search for alternative or supplementary sources of protein other than the currently utilized sources which are expensive. Biting flies, if caught in large enough numbers, can easily be used as a protein source for feeds manufacture to replace or supplement the currently used expensive protein sources from fish or soybeans. This research study therefore, investigated the diversity, abundance and nutritional profiles of biting flies caught in peridomestic and natural habitats in Kibwezi west sub county, Makueni County. The study also sought to compare the performance of the trapping devices commonly used in the control of these flies.

1.3 Research question

1. Does diversity and abundance of biting flies vary between peridomestic and natural habitats?
2. Is the crude protein content of biting flies high enough to warrant interest in them as a source of protein?

1.4 Objectives

1.4.1 General objective

The overall objective of this study was to determine the diversity and abundance of biting flies in peridomestic and natural habitats, and profile the nutrient composition of the dominant species with a view to assessing their suitability as protein source for use in feeds.

1.4.2 Specific objectives

i). To assess the performance of commonly used biting flies traps and diversity of their catch.

ii). To determine the diversity of biting flies in peridomestic and natural habitats.

iii). To profile nutrient composition of the dominant biting fly species in the peridomestic and natural habitats.

1.5 Research hypothesis

i. There is a difference in mean catches of commonly used biting fly traps.

ii. There is a difference in the diversity of biting flies and other insects in peridomestic and natural habitat

iii. There is a difference in nutrient composition of the dominant biting fly species in the peridomestic and natural habitats.

CHAPTER TWO

2.0 LITERATURE REVIEW

2.1 Common biting flies

Biting flies, as the name suggests, are haematophagous diptera that bite humans and other animals to suck blood which they feed on. There are many species of true flies that are collectively referred to as biting flies. They have a cosmopolitan distribution inhabiting both natural, peridomestic and domestic habitats. Some of the biting flies are capable of transmitting disease-causing pathogens through their bites as they feed. Transmission may be biological, where the pathogens undergo a cycle of development in these flies and/or mechanical, where the pathogens are just transferred mechanically to the next host without undergoing development in the flies. Commonly encountered biting flies include tsetse flies (Glossinidae), horse flies, deer flies (Tabanidae), stable flies (*Stomoxys*), black flies (Simuliidae), sand flies (Psychodidae), biting midges (Ceratopogonidae) and mosquitoes (Culicidae), among others.

2.1.1 Tabanids

Tabanids are livestock vectors belonging to the family Tabanidae with more than 4,500 species in three subfamilies: Chrysopsinae, Pangoniinae, Tabaninae (Daniels, 1989). Tabanids are large robust biting flies whose females need blood to support egg development while males feed on nectar (Figure 1). Tabanids undergo complete metamorphosis with the cycle taking 1-2 years (Foil and Hogsette, 1994). Mating process occurs in swarms and their eggs are eventually laid on stones and water or on vegetation near water bodies. Adult tabanids inflict painful bites to human and livestock creating feeding and oviposition sites for myiasis causing flies subsequently causing annoyance and anemia.

Tabanids mechanically transmit several disease pathogens to livestock and humans (Krinsky, 1976).

Figure2.1: *Tabanus* spp – a large Tabanid commonly known as a horse fly. (Source: naturespot.org.uk)

2.1.2 Tsetse flies

Tsetse flies (or simply tsetse) belong to the family Glossinidae which is divided into three species groups; Morsitans group (savanna species), Palpalis group (riverine species), Fusca group (forest species), with each group exhibiting distinct distribution patterns (Radostitis *et al*., 2007). These three groups are divided basing on climatic conditions, vegetation and ecological zones (Wint and Rogers, 2000). The Morsitans group (e.g. *Glossina morsitans* and *Glossina pallidipes*) inhabit wooden savanna save for the dry season which they are found near the water bodies. They are distributed in Sudan, East, West and Central Africa and Mozambique.

The Palpalis group (*G. palpalis*), are distributed in dense vegetation near the edge of rivers. They are distributed in the forest areas of West and Central Africa, the riverine forest penetrating into the savanna regions.

The Fusca group (e.g. *Glossina longipennis*) is mainly found where vegetation is dark shaded and in riverine thickets. *G. longipennis* is limited to Kenya, Ethiopia, South-Eastern Sudan, Southern Somali, North- Western Uganda and Northern Tanzania (Aksoy *et al*., 2003). Distribution range of these tsetse fly species extend to domestic or peridomestic habitats.

Both sexes of tsetse flies feed on blood, and are thus biological vectors of trypanosome parasites which cause animal trypanosomiasis (Nagana) and human trypanosomiasis (sleeping sickness) (Wall and Shearer, 1997). Disease causing blood parasites are transmitted to both animals and human through trypanosome infected tsetse bites (Lambrecht, 1968). Tsetse flies can be distinguished from other biting flies due to resting with their wings held over their back resembling a scissors and presence of hatchet shaped cell positioned at the Centre of the wing (Itard, 1989) (Figure 2.2). The fly undergoes a complete metamorphosis with its lifecycle taking about 50 days (Leak, 1999). Tsetse flies breeds all year round with highest fly numbers being recorded immediately after rainy season. Tsetse flies thrive well in temperatures ranging between 20 to 25 degrees Celsius (Radostitis *et al* 2007).

Figure 2.2: A resting tsetse fly, *Glossina* (Source: naturespot.org.uk)

2.1.3 Stable flies

Stable flies belong to genus *Stomoxys* in the family Muscidae. Found in all habitats, the genus *Stomoxys* has more than 18 species with *Stomoxys calcitrans* being the most dominant species (Zumpt, 1973). Both sexes of stable flies are not only blood feeders but also mechanical vectors of pathogens. In livestock, they cause disruption when feeding and resting, skin and tissue injury, reduction of food intake, anxiety, anemia, and a global immunosuppressive (Campbell *et al*., 2001). In addition to blood sucking, they mechanically transmit disease causing pathogens such as West Nile virus, Rift valley fever virus, blue tongue virus, Q fever, anthrax, animal trypanosomes, protozoa- *Leshmania tropica*, helminthes -*Habronima microstoma* (Baldacchino *et al*., 2013). A stable fly resembles housefly (*Musca domestica*) save for its proboscis which has a forward projecting piercing and sucking proboscis (Soulsby, 1982). Adult flies measures 8mm in length with short abdomen consisting of three dark sports on the second and the third segment (Taylor *et al*., 2016). The female lays eggs in decaying vegetation (Foil and Hogsette, 1994). Stable flies undergo a complete metamorphosis with the cycle taking between 56 to 77 days (Yeruham and Braverman, 1994).

Figure 2.3: Stable fly, *Stomoxys calcitrans* (source: naturespot.org.uk)

2.1.4 Black flies

Black flies (order Diptera) belong to the family Simuliidae which has more than 1700 species worldwide (Adler and McCreadie, 2019).They undergo a complete metamorphosis with female black flies requiring a blood meal to lay eggs thus. They suck blood from birds and humans and animals depending on the species preference. Black fly life cycle takes five to six months depending on water temperature and food supply. They are vectors of *Onchocerca volvulus*, human filarial *Mansonella ozzardi*, bird protozoa, Leucocytozoon in turkey, geese and ducks, and viruses responsible for vesicular stomatitis.

2.1.5 Sand flies

Sand flies belong to the family Psychodidae, sub-family Phlebotominae. This subfamily has more than 700 described species. but only two genera are of medical and veterinary importance.(*Phlebotomus* and *Lutzomyia).* They are small nocturnal insects measuring 2-3 mm with a characteristic of folding their hairy wings in V shape when at rest. Both adult males and females feed on nectar, fruits and plant juices to obtain carbohydrate nutrition (Hashiguchi and Gomez 1991). The females are haematophagus feeding on human, domestic and wild animals (Harwood and James 1979). Sandflies are nocturnal and inhabit animal burrows, caves, rocks and human habitats (Feliciangeli, 2004, Claborn *et al*., 2008). Sand flies are vectors of both zoonotic and anthropogenic diseseses caused by protozoa (Leishmaniasis (Kala- azar), bacteria (*Bartonella bacilliformis*, yellow fever, ephemeral fever, Oroya fever) and virus (Phlebovirus) (Alten *et al*., 2015). Sand flies also transmit several saliva borne viruses: Phlebovirus, Vesiculovirus and Orbivirus grouped into the families Bunyaviridae, Rhabdoviridae and Reoviridae respectively (Mihok and Carlson, 2007.)

Sandflies undergo complete metamorphosis ranging between 25 to 52 days depending on the species. Immature stages of sandflies require warm and moist environment. Female lays eggs in a mass of organic content for instance animal excreta. Eggs take an average of 10 days to hatch into larva. The larvae are caterpillar-shaped with distinctive caudal setae. Sandflies have four larva instars which then molts into pupa which has a cast-off exoskeleton for attaching the pupa to the substrate holding it upright.

2.1.6 Biting midges and mosquitoes

Biting midges are the most abundant hematophagous insects with a cosmopolitan distribution inhabiting aquatic, semiaquatic and terrestrial habitats. They belong to Ceratopogonidae, a family of tiny flies (1-3mm long), with a total of 1,358 described species (Borkent 1997). Females are obligate blood feeders getting their blood from a wide range of reptiles, mammals, birds, man, and even blood-engorged mosquitoes. The males feed on plant juice (Meiswinkel *et al*., 2004). Culicoides spp. are nuisance to humans and inflict allergic dermatitis in horses which subsequently results in scratching and hair loss on the affected area. They are biological vectors of viruses, protozoa and filarial nematodes affecting birds, humans, and other animals (Purse *et al*., 2015). They have been associated with transmission of bluetongue virus (BT), African horse sickness virus, haemorrhagic disease virus, equine encephalitis virus, Akabane virus, bovine ephemeral fever virus and Schmallenberg virus (Mellor *et al*., 2000, Elbers *et al*., 2013).

There are about 3,300 species of mosquitoes in the family Culicidae, belonging to 41 genera. Mosquitoes inhabit fresh water habitats and exhibit holometabolous development cycle. The larvae and pupa require an environment with standing water or slow flowing water for proper development. Some of the common mosquito habitats are along flowingrivers, ponded streams, lake edges, wetland and marshes, shallow ponds, surface pools, sporadic temporaly puddles and containers. Females lay eggs singly or in clusters. Male mosquitoes feed exclusively on nectar while the females feed on blood. Mosquitoes are known to transmit a number of diseases to human and livestock; filariasis, viral encephalitis, Malaria, dengue, Rift valley fever virus (Beaty and Marquardt 1996; Eldridge and Edman 2000).

2.2 Management of biting flies.

Biting flies can be managed through an integrated approach. Over time various ecological, chemical and biological or genetic methods have been developed for controlling these flies.

Ecological control of biting flies involves total clearance of vegetation or partial vegetation clearing which is used against tsetse flies: riverine species, elimination of wild animals like buffaloes or an integrated approach of all the above. Due to environmental concerns, the above activities are partially used. Human activities like smoking and use of continuous wooden or wall netting to protect livestock from the biting flies has reduced the number of tsetse flies significantly (Torr, 2011).

Biological control of biting flies involves use of flies reared in the laboratory for autocidal control or for population monitoring, management of natural enemies (parasites and predators) and microbial agent, and use of chemosterilants, insect growth regulators, and juvenile hormone analogues for non-radiation based auto related odour-baited targets other than the conventional biological methods discussed above, there is the sterile insect technique (SIT) used in the management of vectors of medical and veterinary importance. Control by sterile insect techniques (SIT). Laboratory reared male tsetse flies are sterilized through irradiation with Gamma rays (FAO 1992).

The efficiency of this technique is supported by the fact that tsetse fly mate once during their life time, and the female stores the sperm in the spermathecae. Sterilization of the males targets decrease in population and eventual eradication of tsetse flies. Tsetse flies are laboratory reared in large numbers then the pupae obtained is sterilized through radioactivity. Radiation induces sterility, but treated male flies can still fly and mate with the females. The sterile flies are dispersed into the field where both sterile and wild males compete for fertilization of females (Hawse, 2005). For effectiveness of this technique, the number of sterile males produced should be greater than the natural population. Although the technique is expensive, it is a safe and environmentally sound method. This method can be combined with targets and chemical control during the initial stages to minimize the frequency of releasing the male flies (Leak 1998).

Chemical control of biting flies comprises use of residual and persistent insecticides for spraying on the bushes, indoor residual spraying or spraying the domestic animals. Spraying can be aerial or ground spraying. Aerial application of insecticides to manage biting flies involves spraying areas where they inhabit using suitable insecticide which kills both adults and immatures stages once they emerge (Bourn and Scott, 1978). Selection of insecticides used is based on its efficiency and residual effect. Aerial spraying is the most ideal method where a large area has to be covered. For instance, species of *G. morsitans, Glossina sywnertoni* and *G. pallidipes* cover a wide area in terms of distribution, and thus, the use of aeroplane for aerial application of insecticide is the most efficient method (Afewerk, 1998). However, this method cannot be used to control biting flies living in large tall mature trees with closed canopy like *G. palpalis* (Rogers *et al*., 1994).

During such instances, ground spraying of biting flies is involved insecticides are applied on fly resting-places, on the soil to kill ovipositing females and in open paths in the forest. This method has been used effectively to manage the riverine species, like *G. palpalis* and *G. fuscipes*.

Initially, Dichlorodiphenyltrichloroethane (DDT) suspensions and emulsions were used for ground spraying; they have been replaced by 4% dieldrin. Use of residual insecticides has been successful on restricted biting fly habitats (Terblanche *et al*., 2008).

Insecticide impregnated Targets are pieces of insecticide treated blue and black cloth deployed in tsetse infested habitats (Hao *et al*., 2001). Thin steel bars are used to support the targets. Although in absence of steel bars wooden poles are used as a substitute (Welburn and Maudlin, 1999). Then hanged on tree branches, fixed to supporting poles or fixed plant stem. Targets are treated with 0.4% of deltamethrin solution sufficient to knock down and kill biting flies and non-biting flies that come into contact with it.

On the other hand, traps have been used for management of biting flies. Each of the biting fly traps was developed for trapping specific species of biting flies. Traps are devices composed of blue and black fabrics with or without white netting used for tsetse control. They are usually impregnated with insecticides, to increase their efficiency of killing flies. For instance, the flies which do not manage to be trapped but lands outside the trap. They are used for both control and monitoring of biting flies (Vale *et al*., 1999). Traps are designed to mimic host animals, thus attracting the tsetse and eventually capturing them. Tsetse flies are attracted to blue colour, thus all tsetse fly traps are blue. The black colour enables the fly to settle. Since insects have a tendency of flying up, tsetse flies subsequently move upwards following the light direction (Vanden-Bosche *et al*., 2001).

Attractants were used to lure flies into the trap includes cow urine, acetone octenol and phenols (Robertson, 1991). Over time trap designs have undergone modifications to suit their efficiency under prevailing local conditions.

various traps with unique descriptions have been used for catching biting flies: Malaise trap, Sticky trap, Sticky back screen, Harris trap, Monoconical trap, Vavoua trap, Monoscreen trap, H trap, S trap, Pyramidal and Bipyramidal trap, Biconical trap, Ngu trap, Nzi trap, and traps among others. The performance of these traps in the capture of biting flies vary greatly as most of these trapping devices have been developed for specific groups of flies, mainly tsetse flies. Trapping technology increasing becoming a very significant component of integrated biting flies management due to its environment friendly nature and modest cost as well as its effectiveness in reducing fly densities. Biconical, Ngu, Nzi, and Pyramidal traps are the commonly used traps for biting flies.

2.2.1 Biconical trap

The Biconical trap (Figure 1.4) was developed in West Africa for riverine species of tsetse such as *Glossina palpalis* (Brightwell *et al*.,1987) and *Glossina tachniodes*. The trap is recommended for both control and surveys of tsetse and other biting flies. The trap is made up of two cones, an upper netting cone, and a lower blue cloth cone which allows the fly to enter the trap, black cruciform and a central pole that supports the trap.

Figure 2.4: A Biconical trap

2.2.2 Ngu trap

Ngu trap (Figure 2.5) was developed in Kenya specifically for trapping *Glossina pallidipes* (a Morsitans group tsetse). The trap is made of two rear sides that are blue in colour. The black shelf - that slants into the trap from the top. The black target base is attached halfway along the base of its two sides and its top is fixed to the upper rear corner. Pyramidal shaped net cone with 12mm hole is fixed to its apex that allows flies into the polythene cage. A polythene cage in which is a modified tetrahedron is used to collect the trapped flies.

The trap cage and the cone are suspended by metallic or wooden poles. (Brightwell *et al.*, 1991). The trap is baited using cow urine, acetone or phenol.

Figure 2.5: Ngu trap

2.2.3 Nzi trap

Nzi trap (Figure 2.6) was developed in Kenya (Mihok *et al*., 1995) for *G. pallidipes, Stomoxys* and Tabanids, although it proved to be a more universal that catches biting flies in large numbers. It is triangular in cross section with two equal wings of blue cloth. Nzi is a simple, flexible cloth trap with combination of blue and black colors and white netting for attraction and contrast.

The trap is known to capture more biting fly species than any other trap. Octenol or fermented cow urine are used as an attractant (Mihok, 2002).

Figure 2.6 : Nzi Trap

2.2.4 Pyramidal trap.

Pyramidal trap (Figure 2.7) was developed for trapping *G. palpalis, G. fuscipes quanzensis* (riverine group). This trap type is simple to construct and easy to transport to the field giving it advantage over other traps types. The upper net cone is pyramidal with blue and black material reaching only halfway. The upper part of the trap are netting while the lower part has two horizontal wooden rods running diagonally across the base to prevent the trap from collapsing from wind.

The trap can be suspended freely on a tree using a string (Gouteux and Lancien, 1986).

Figure 2.7: Pyramidal trap

2.2.5 Live bait techniques

This technique involves application of insecticides to livestock to control the biting flies. Insecticides are constituted as per the manufactures instructions then applied to livestock through dipping, spraying or as pour-on, along the back (Leak *et al*., 1995). These insecticide treated animals are as the mobile targets and are more efficient than the targets and traps. This technique is used against tsetse flies, other biting flies, ticks and other nuisance flies (Aksoy *et al*., 2001).

2.3 Insectivory

Insectivory is the consumption of insects and arachnids (spider and scorpions) exclusively (obligate insectivores) or as a portion in diets (facultative insectivores). Insectivore is a carnivorous plant or animal that consumes insects (Gullan and Cranston, 2005). The term can also be used to refer to consumption of wide variety of crustaceans, annelids and arachnids. Some primates prey directly onto insects while others feed on insects indirectly in fruiting bodies and vegetative material. Insects consumed belong to wide range of orders including the solitary and the social living insects. Insects preyed on by primates belong to insect Orders Coleoptera (beetles), Hymenoptera (bees and ants), Isoptera (termites), Lepidoptera (butterflies and moths), and Orthoptera (grasshoppers and crickets) (McGrew, 2014). Primates eat insects for nutritional supplementation mostly protein, vitamins, and minerals. Some of the challenges facing insectivory are poor distribution of the edible insect species and seasonal availability of insects. Primates employ various insects capturing techniques. Some primates eats or lick insects from leaves, others forage actively in substrate, and others are destructive foragers with elongated fingers to locate insects in tree hollows for instance aye-aye (*Daubentonia madagascariensis*). Obligatory insectivorous primates use both audio and visual sensory techniques to detect and capture insect prey.

Insectivorous animals are important as biocontrol agent, these includes moles, shrews, bats and hedgehogs (Jaglan and Rolania, 2018). Various organisms feed on insects as primary diets while others feed on insects for protein supplements when they are breeding (Raubenheimer and Rothman, 2013). For instance, spiders, frogs, lizards, chameleons, swallows, anteaters, sloth bear bats dragonflies, hornets, and praying mantis. Some insects prey on other insects e.g. lady birds and praying mantis feed on crop pests. Land dwelling birds like purple martins, warblers and barn swallows are exclusive insectivores (Srivastava *et al.,* 2009). Savanna chimpanzees obtain nutrients for their growth from consuming termites which are rich in manganese, iron, zinc and lipids (Deblauwe and Janssens, 2008). Various insectivores have modifications on their mouthparts adapted for the food they take Mammal insectivores have a characteristic sharp pointed nose, tiny ears and small eyes. Mollossidae families (bats) have jaws modified for crushing hard- shelled insect prey. Beetle eaters have heavy jaws, well developed cranial crests, larger teeth. Whereas moth-eaters have thin jaws, little crest build-up, and more but smaller teeth (Freeman, 1979). Ants feed on tsetse pupae while wasps and spiders and robber flies feed on adult tsetse flies. Pata monkeys (*Erythrocebus patas*) are insectivorous feeding primarily on gum and small arthropods (Nash, 1986). Native birds controlled insects' pests on farms in North Central Florida (Jones *et al.,* 2005). Indian runner ducks are functional insectivores in that they are used to control mosquito pupae. Entomophagous nematodes and fungi obtain their nutrition from insects thus have been used as biological pest control agents. Controlling insects through use of insects as food has been practiced in Thailand during locust plagues (Yhoung-Aree *et al*., 1997, Van Huis, 2003). Therefore, integrating pest or vector control by indirect entomophagy may not only reduce disease threat and loss in productivity but also promote health by providing protein-rich food. Previous studies on dietary ecology shows that majority of primates consume insects in the orders Coleoptera, Hymenoptera, Isoptera, Lepidoptera and Orthopera (McGrew, 2014).

2.4 Entomophagy

Entomophagy is defined as consumption of insects by humans. Insects are the most abundant and diverse living organisms on earth with the number of their species exceeding 10 million (Chapman 2006). Their abundance and diversity has been attributed by short life spans, ability to colonize new niches, feeding on a wide variety of plants and animal species and powerful means of fighting infections e.g phagocytosis and lysis (Kelemu *et al*., 2015). Close to 2,000 edible insect species, spread over 14 orders (Table 2.1), are commonly consumed worldwide by over two billion people (Ramos-Elorduy and Paoletti, 2005). Out of these, about 400 are consumed in Africa (Allotey and Mpichane, 2003). Consumption of insects is dependent on taste, nutritional value, culture (values, customs ethnic values and prohibitions) and religion (Van, 2003). For example, the Western countries regard insects as pests and eating them is regarded disgusting (FAO, 2013).

Some of the reasons that make entomophagy popular are the fact that most of these edible insects are readily available in forested and aquatic habitats and can be collected in large numbers within a short time (Young-Aree *et al*., 1997). Edible insects require small spaces for rearing and multiplication and have a short life cycle and high intrinsic growth rate (Gahukar, 2016). They are more environmental friendly in terms of the amount of feed they consume and emission of greenhouse gases (Oonincx *et al*., 2010). For instance, the female house cricket *Acheta domesticus* lays more than 1300 eggs in 3–4 weeks and its water and feed requirements are relatively low compared to beef animals (Clifford, 1977). The efficiency of conversion of ingested food (ECI) of house cricket is double that of pigs and broiler chicken, four times that of sheep, and six times higher than that of steers (Capinera, 2004).

Table 2.1: Number of edible insect species reported worldwide (source: Ramos-Elorduy and Paoletti, 2005)

Order	Common name	Number of species
Zyngetoma	Silverfish	1
Anoplura	Lice	3
Ephemeroptera	Mayflies	19
Odonata	Dragonflies	29
Orthoptera	Grasshoppers, cockroaches, Crickets	267
Isoptera	Termites	61
Hemiptera	True bugs	102
Homoptera	Cicadas, Leafhoppers, Mealy bugs	78
Neuroptera	Dobson flies	5
Lepidoptera	Butterflies, moths (silkworms)	253
Trichoptera	Caddis flies	10
Diptera	Flies, Mosquitoes	34
Coleoptera	Beetles	468
Hymenoptera	Ants, bees, Wasps	351
Total		1,681

Previous studies have shown that availability of edible insects is seasonal; mostly coinciding with onset of rainy season when there is shortage of food. Entomophagy can be direct or indirect. In direct entomophagy, Insects are consumed at various developmental stages; egg, larvae, pupae and adult. Some insects are consumed directly at more than one developmental stage. For example, *Apis mellifera* is consumed directly at its egg, larvae and pupae stages with high levels of protein content being recorded in all the three stages. Moreover, insects can be consumed raw/processed or used as an ingredient or supplement in modern recipes like sauces and condiments to spice or enhance flavor to some foods E.g. ants incorporated in lollipop sweets. Insects can be consumed in flours, in form of granules and pastes. Edible insects can be deep fried (Odonata), roasted (Orthoptera) or used to prepare stews (Hymenoptera) (FAO, 2013).

Consuming insects as condiments and insects' preparations like baking using insect flour, making protein bars, crackers, muffins and meat loaves using cricket powder, and mixing termite powders with food ingredients, are some of the interventions used to address the disgust or "yuck" factor associated with use of insects as food in western countries. Silkworm (*Bombyx mori*) pupae have been used for food and medicine in majority of Asian countries for treatment of neurological disorders, stress and Alzheimer disease (Wattanathon *et al.*, 2012). Attention is shifting from direct consumption of insects to indirect entomophagy whereby insects are used as fish and poultry feed to supplement fishmeal and soymeal consequently improving nutritional composition of traditional food sources (Ayieko *et al*., 2012). Insects are dried, crushed as parts or whole or protein component extracted and incorporated in the feeds as supplement. Black soldier fly (*Hermetia illucens*), house fly maggot (*Musca domestica*), mealworm pupae (*Tenebrio molitor*), Mormon crickets (*Anabrus simplex*) have been used as feed for poultry, pig and fish. This has been necessitated by their being palatable and high protein content thus can replace soymeal and fish meal in feeds (Makkar *et al.*, 2014).

Entomophagy is a potential source of protein for human and livestock as insects are highly nutritious with high protein, fat and mineral content and therefore can be a solution to malnutrition in developing countries and also can improve health in western societies.

2.5 Nutritional Composition of Insects

Insects are a highly nutritive natural source of food (Ramos-Elorduy, 1997). They contain carbohydrates, vitamins, minerals, proteins (amino acids- methionine, cysteine, lysine and threonine), fibre, dietary energy and fats in varying percentages (Gahukar, 2011). Nutritional value of insects is dependent on several factors. The insect species, its developmental stage, habitat and diet coupled with the method of preparation and processing used by the consumer (Rumpold and Schlüter, 2013). Limited information is available on nutritional composition of edible insects in spite of several global investigations. This has impacted negatively on consumer confidence and thus the incorporation of these edible insects in food and feed.

Edible insects contain huge quantities of polyunsaturated fat for provision of the much needed energy for sustaining life. Previous studies have shown that some edible insects have higher fat content than livestock and vegetables (Krause and Mahan, 2003) making them a good option for substitution with plant and animal fat as a source of energy required by both animals and humans (Ramos-Elorduy *et al*., 1997). Polyunsaturated fatty acid minimizes sugar tolerance, consequently minimizing the risk of diabetes and hypertension (Sirtori and Galli, 2002). For example, the edible grasshopper (*Ruspolia differens*) has a fat content of 67% whereas that of the African palm weevil (*Rhynchophorus phoenicis*) is 54% (Womeni *et al*., 2009) (Table 2.2).

Elsewhere, studies conducted on determination of fat content and fatty acids present in numerous insect species eaten in Cameroon indicated that the oil content in edible insects ranged between 7-48% (Womeni *et al* ., 2009).

Table 2.2: Fatty acid profiles of some edible insects (source: *Womeni et al*., 2009).

Edible insect	Fat content (% of dry matter)	Composition of main fatty acids (% oil content)		Types of fatty acid
African palm weevil (*Rhynchophorus phoenicis*)	54	Palmitoleic acid	38%	MUFA
		Linoleic acid	45%	PUFA
Edible grasshopper (*Ruspolia differens*)	67	Palmitoleic acid	28%	MUFA
		Linoleic acid	46%	PUFA
		a-Linoleic acid		PUFA
Termites (*Macrotermes* sp.)	49	Palmitic acid		SFA
		Oleic acid		MUFA
		Stearic acid	9%	SFA
Saturniid caterpillar (*Imbrasia* sp.)	24	Palmitic acid	8%	SFA
		Oleic acid	9%	MUFA
		Linoleic acid	7%	PUFA
		a-Linoleic acid	38%	PUFA
Variegates grasshopper (*Zonocerus variegates*)	9	Palmitoleic acid	24%	MUFA
		Oleic acid	11%	MUFA
		Linoleic acid	21%	PUFA
		a-Linoleic acid	15%	PUFA
		g-Linoleic acid	23%	PUFA

MUFA, monounsaturated fatty acids; PUFA, polyunsaturated fatty acids; SFA, saturated fatty acids.

Edible insects energy content varies from one species to another with 89 Kcal/100 g fresh weight being recorded in raw grasshopper (*Cyrtacantharis tatarica*) to highs of 1272 Kcal/100 g fresh weight in raw green weaver ant (FAO, 2012). Table 2.3some of the edible insects sampled from various locations and their energy content Kcal/100g fresh weight.

Table 2.3: Energy content (Kcal/100g fresh weight) of some common edible insects (source FAO 2009)

Location	Common name	Scientific name	Energy content Kcal/100g fresh weight
Australia	Australia plague locust, raw	*Chortoicetes terminifera*	499
Australia	Green (weaver) ant, raw	*Oecophylla smaragdina*	1272
Canada, Quebec	Red- legged grasshopper, whole, raw	*Melanoplus femurrubrum*	160
The USA , Illinois	Yellow mealworm , larva, raw,	*Tenebrio molitor*	206
The USA , Illinois	Yellow mealworm , larva, adult,	*Tenebrio molitor*	138
Ivory coast	Termite, adult, de-winged, dried, flour,	*Macrotermes subhylinus*	535
Mexico Veracruz state	Leaf cutter ant, adult, raw.	*Atta Mexicana*	404
Mexico Hidalgo state	Honey ant , adult raw	*Myrmecocystus melliger*	116
Thailand	Field cricket, raw	*Gryllus bimaculatus*	120
Thailand	Giant water bug, raw	*Lethocerus indicus*	165
Thailand	Rice grasshopper, raw	*Oxa japonica*	149
Thailand	Grasshopper raw	*Cyrtacanthacris tatarica*	89
Thailand	Domestic silkworm pupa , raw	*Bombyx mori*	94

Insects are potentially an important source of protein for food and feed through direct consumption or feed supplements. Proteins consist of amino acids which are building blocks required for the biosynthesis of proteins through human metabolism consequently leading to proper growth and development. The nutritive value of protein depends on protein content of insect species, protein quality (which depends on the kind of amino acids present) and protein digestibility (Yi., et al 2013). In addition to amino acids, proteins contain enzymes, hormones and hemoglobin.

Moreover, proteins are important in biological function and are important in final products of the information pathways. They also play a role in expression of genetic information. Therefore, ensuring a stable source of protein is vital for providing energy for both humans and animals.

Previous studies shows that edible insects have higher protein content than plant and animal protein (Van Huis, 2013) and insect based protein having higher digestibility compared to animal protein (Finke, 2004). In other studies by Finke (2007), insects in the order Lepidoptera recorded highest amount of crude protein followed by Coleoptera, while the order Hymenoptera had the least. These results agree with other studies conducted by Teffo *et al*., (2007), which revealed that, edible insects have higher protein content, than beef, chicken, fish, soybeans, and maize. FAO (2004) did comparison studies of protein content of insects, reptiles, cattle, and fish which revealed that protein content in edible insects was higher than in fish and mammals. Table 2.4 gives a clear indication that edible insects are a potential source of protein and more research on their use as an alternative source of protein is needed.

Table 2.4: Range of Crude protein content of major edible insect orders (Source: Xiaoming *et al*., 2010)

Insect order	Stage	Range (% protein)
Coleoptera	Adults and larvae	23-66
Lepidoptera	Pupae and larvae	14-68
Hemiptera	Adults and larvae	42-74
Homoptera	Adult, larvae and eggs	45-57
Hymenoptera	Adult , pupae, larvae and eggs	13-77
Odonata	Adult and naiad	46-65

Carbohydrate content in edible insects is formed by chitin which is a macromolecular compound. Chitin is responsible for reduction of serum cholesterol (Burton and Zaccone P. 2007) and improvement of body's immune system (Sun *et al*., 2007).

Carbohydrate content ranges from lows of ranges from 6.71% in stink bug (*Tessaratoma papillosa*) to highs of 15.98% in cicada (*Meimuna opalifera*) (Raksakantong *et al*., 2010).

Analysis of mineral components in edible insects portrays several minerals which plays various metabolic and physiologic roles in human body (Baek *et al*., 2017). This study agrees with a similar one done by Banjo et al 2009 (Table 2.5). For instance sodium, calcium, copper, iron zinc, manganese and phosphorus as displayed in cricket nymphs, cricket adult, mealworm adult, giant mealworm (not arranged in order) being some examples of minerals found in edible insects (Oliveira *et* al.,1976, Van Huis, 2003, Sun, 2008). Minerals inhibit oxidation and cofactors of enzyme (Talwar *et al.,* 1989). Moreover, minerals responsible for preventing heart muscle disease, muscle atrophy retarded growth, impaired spermatogenesis, immunological abnormality, and inability to form blood clot (Chaturvedi *et al.,* 2004). Minerals present in edible insects are dependent on the developmental stage of the insect. For instance, cricket nymphs are rich in sodium whereas adults are a rich source of calcium, manganese, zinc and phosphorous (Van Huis *et al.,* 2013). Table 2.5 displays that edible insect species are rich in minerals with

Argiope trifasciata recording high levels of Calcium (61.28 mg/100g) and phosphorus. *Megachile nigeriensis* has 60.96 mg/100g levels of magnesium and 7.6 mg/100g ash content. Apis mellifera has iron content of 25.2 mg/100g.

Table 2.5: Mineral and ash (mg/100 g) contents in some edible insect species. (Source Banjo *et al.*, 2009).

Insect species	Ca	P	Fe	Mg	Ash
Megachile nigeriensis	1.00	14.90	9.56	60.96	7.60
Macrotermes bellicosus	21	136	27	0.15	2.90
Macrotermes natalensis	18	114	29	0.26	1.90
Brachytrupes Spp.	9.21	126.6	0.68	0.13	1.82
Circus aeruginosus	4.40	100.2	0.35	0.09	2.10
Zonocerus variegatus	42.40	131.2	1.96	8.21	1.20
Argiope trifasciata	61.28	136.4	18.2	6.14	4.21
Anaphe infracta	8.56	111.3	1.78	1.01	1.60
Annona reticulate	10.52	102.4	2.24	2.56	2.50
Lepidoptera litoralia	12.00	9.00	19.50	0.50	4.30
Anaphe venata	8.75	100.5	2.01	1.56	3.20
Cirina forda	8.24	111.0	1.79	1.87	1.50
Apis mellifera	15.4	125.5	25.2	5.23	2.20

Edible insects contain vitamin A, B2, C all which play a vital role in proper vision, normal blood clotting and as antioxidants respectively. Previous studies on Insect order hymenoptera contain vitamin A (3.24mg/100g content) and vitamin C (10.25 mg/100g content). While Orders Isoptera, Coleoptera, Orthoptera, Lepidoptera have recorded presence of vitamin A, B, C, between the ranges 0.03 to 8.06 mg/100g content (Igwe *et al*., 2011, Ifie and Emeruwa, 2011).

2.6 Insects as an alternative source of protein for feed

Global feed manufacturing is estimated to over one billion tonnes (International Feed Industry Federation). FAO approximates feed production to increase by over 70% to accommodate feeding the increasing population in 2050. Despite this challenge, little has been said about the prospect of insects as feed sources. Ingredients for animal and fish feed include fishmeal, fish oil, soybeans and a number of other grains. Currently Cost of feed is very high, this has been necessitated by high price of meat meal, fishmeal and soybean meal, which represent between 60–70% of production costs (Hardy, 2006). However, a major drawback to further expansion of meat and fish manufacturing is acreage for soybean cultivation is decreasing globally, while on the other hand, overexploitation of marine resources has continued to reduce the abundance of small pelagic forage fish which are major components of fish meal and fish oil (Tacon and Metian, 2013).

The increasing shortage of resources for production of these increasingly needed ingredients has doubled their prices in the last five years, while the feed cost is already exorbitant, signifying 60-70% of production costs (REF). It will therefore not be a justifiable decision to continue to bank on on fishmeal and soybean as protein source in feed production (Van Huis *et al*., 2013). In many sub-Saharan Africa (SSA) countries, poultry and fish industries are among the fastest developing agro-businesses with women accounting for over 60% of the producers (Ondwasy *et* *al.*, 2006). However, feed unavailability and poor quality have been regarded as the most significant limitations for growth of fish farming in several countries including Kenya and Uganda (Fiaboe and Nakimbugwe, 2017).

The use of costly inputs like soybeans and cereals as feed ingredients is threatening the existence of the feed industry and there is need to find practical and sustainable substitutes. The use of insect protein as a replacement for expensive protein sources from fish or plants is one possible solution. Insects are natural feed sources for fish and poultry. In Asia and the Pacific, grasshoppers, crickets, cockroaches, termites, lice, stink bugs, cicadas, aphids, scale insects, psyllids, beetles, caterpillars, flies, fleas, bees, wasps and ants have all been used as complementary feed sources for poultry (Ravindran and Blair, 1993).

The amino acids derived from most insects' protein are superior to those from plant supplements in poultry feed formulations(Ravindran and Blair, 1993; Bukkens and Paoletti, 2005). In addition various insect species have a higher percentage of protein content compared to conventional fish and soybean meals (Anand *et al*., 2008). Since protein is considered as the most expensive component in the diets of poultry, feeding them with insects appears to be an economically practical option. Moreover, insects can be used to transform different types of organic waste materials into animal biomass rich in proteins which can be used later in animal nutrition additionally providing environmentally friendly organic waste reprocessing opportunities (Ramos-Elorduy, 2005). Limited research has been conducted in East Africa to exploit insects as an alternative protein source in feeds. Reviews by FAO recognized the role of insect as food and feed(Van Huis *et al*., 2013). Additionally, chitin, a polysaccharide present in the exoskeleton of insects is reported to have an enhancing result on the operations of the immune systems of different organisms.

Therefore, by feeding insects to chicken, the routine use of antibiotics in the poultry production (which can speed up drug-resistance development in human-pathogenic bacterial strains) may be reduced (Leverstein-van Hall *et al*., 2011).

Previous research has shown that insects contain a high nutritive value in terms of proteins, fats, minerals and vitamins. For example, (Ramos-Elorduy *et al*., 1997) demonstrated that the several insect species energy content ranged between 293 and 762 kilocalories per 100 g of dry matter. Essential amino acids derived from insect protein supplements were also found to be superior to those obtained from plant protein supplements in poultry feed formulations(Ravindran and Blair, 1993; Bukkens and Paoletti, 2005). In Latin and North America as well as Asia, the carcass quality, growth rate and palatability of broilers and fish fed on insect-based feeds obtained from crickets, house flies, grasshoppers, locust, mealworm, and silkworms were higher than those fed on conventional feed (Teotia and Miller, 1973).

2.7 Fish and poultry feeds – the future

There is need to increase protein sources to feed the ever increasing world population. With over 2,000 insect species being consumed worldwide (Jongema, 2012). Direct or indirect entomophagy is increasingly being accepted as a viable option with great potential for food security. Many of these insects are collected from the wild (Boulidam, 2010).

Fish is a major source of food for humans (Ayoola, 2010a).The amount of wild fish catches cannot take care of the current market demands for fish, and thus aquaculture is currently being used as a substitute for the same.

Fishmeal prices are becoming unaffordable due to increased prices of feed ingredients, such as fish meal and soya and increased demand (Olsen *et al*., 2012) approximately 68% fishmeal and 89% fish oil are used annually for the aqua feeds (Tacon and Metian, 2009). Consequently small scale farmers are unable to afford these feeds. Increase in world soya prices especially in China has been registered due to high demand caused by growing world population and low production of soya (Lock *et al*., 2009). Reports have indicated that marine resources are being over utilize (Van Huis, 2013). Increasing the use of insect meal in salmon feed could reduce demand for fish meal and add value to low grade biodegradable waste streams. There is need to source for new protein sources which have comparable contents to fish meal, inexpensive, readily accessible, that promote optimum development, growth, and reproduction (Ayoola, 2010b).Soya bean meal in which is also a component of fish meals methionine, cysteine and contains some antinutrient substances such as trypsin inhibitor, haemaglutinin, and antivitamins (Tacon, 1995). More so the size of arable land for cultivating soya bean is diminishing (Han *et al*., 1999).

There is much concern in potential replacements for these expensive components (Van Huis, 2013). A number of insects have been used as replacement for meal and fish oil in animal diets. Various insect species have been used as food; house fly, lesser mealworm, yellow meal worm, silkworm and grasshoppers(Anand *et al*., 2008). Black soldier fly larvae are used to compost and sanitize waste into feed. Pupae and pre pupae of black solder fly are eaten by poultry, pig and fish and contain high levels protein amino acids, arginine, lysine and methionine (El Boushy, 1991). Housefly maggots feed to chicken improves the carcass quality and growth performance (Hwangbo *et al*., 2009). Grasshoppers are food for birds, lizards, snakes, amphibians and fish. (Anand *et al*., 2008). Yellow mealworm is feed to African cat fish (Ng *et al*., 2001) and chicken reared for meat (Ramos-Elorduy *et al*., 2002). Pests of maize, beans, and alfalfa have been harvested for feed in Mexico, Caribbean and Guatemala Island. The grasshoppers are harvested in the morning and sold to Mesoamericans for food. This practice has resulted in drastic decrease in egg densities of pests and reduction in the amount of chemical contamination on agricultural food (Cerritos and Cano-Santana, 2008).

Adapting insects as feed for fish and chicken by the consumers is likely to be embraced with a lot of positivity since insects belong to fish and poultry diets in their natural habitats. Moreover, insects as feed will not encounter animal welfare concerns and they are affordable to local farmers.

Insects for poultry and fish feeds could contribute to food security and provide solution to meat crisis through increased production of fish and poultry meat consequently controlling the disease vectors.

CHAPTER THREE

3.0 MATERIALS AND METHODS

3.1 Description of the study area

This study was carried out in Kibwezi forest and Woni farm both in Makueni County, located approximately 194 km from Nairobi (Figure 3.1). The climate is semiarid. The area experiences two modes of rainfall pattern with the short rains between October and December and long Rains occurring between March and May. The rainfall and the temperatures in the area are affected by the altitude and the season. The cold season occurs between July and August while the hottest months are September, January and February throughout the county. The area receives a mean annual rainfall of about 600mm. The frequency of the rainfall is unevenly distributed throughout the year. The average temperature is 23°C. During the hot season the temperature shoots to 32° C. The area experiences high temperatures during the day and low temperatures during the night.

The study area is covered exclusively by tertiary volcanic materials overlying folded Precambrian basement system rocks of the Mozambique Belt. The geology of the area is composed of tertiary sediments overlaying weathered biotite gneisses underlain by highly weathered and fractured gneisses overlaying the less weathered feldspathic gneisses. Weathered gneisses tending massive subsequently underlay the latter in that stratigraphic order (Saggerson, 1967). The soils vary from red to brown sandy clay soils. They are usually compact with massive structure that has a strong surface sealing. The soils are of volcanic origin which ranges between shallow to rocky. The rocks are highly permeable (Michieka and Van der Pouw 1977).

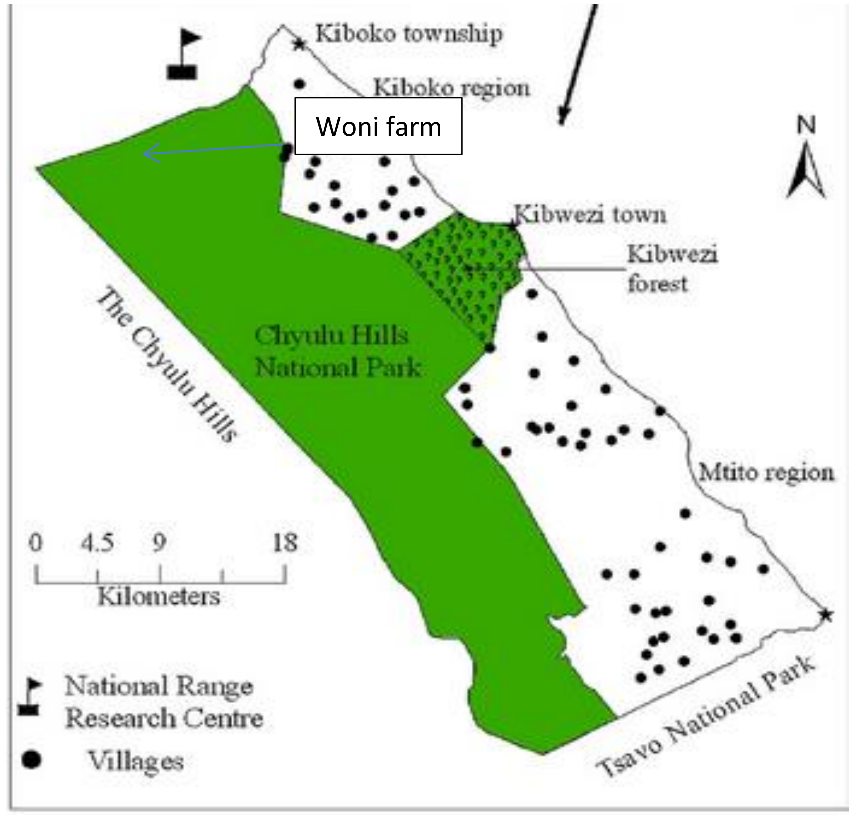

Figure 3.1: Map of the study area (source: www. Researchgate.net)

Kibwezi forest (S 02° 27.846', E 037° 56.203') is a protected area which was classified as a forest reserve in 1936 and covers 60 km^2. The vegetation of the area is controlled by climate, geological formation, soil type and presence or absence of ground water (Gachimbi 1990). The forest is described by *Acacia commiphora* woodland of varying densities, baobab trees (*Adansonia digitata*), perennial grasses like *Cenchrus ciliaris, Enteropogon macrostachyus, and Chloris roxburghiana.* Kibwezi forest is sustained by Umani springs which depend on rainfall from the Northern part of Chyulu hills. Kibwezi forest is a natural, undisturbed and is endowed with a rich biodiversity. It is home to baboons (*Papio Anubis*), leopards (*Panthera pardus*) Sykes (*Cercopithecus albogularis*) and velvet monkeys (*Chlorocebus pygerythrus*), crowned hornbills (*Tockus alboterminatus*), eagles (*Aquila rapax),* guinea fowls (*Numida meleagris*), elephants

(*Loxodanta Africana*) and buffaloes (*Syncerus caffer*). The forest is managed jointly by Kenya Forest Service (KFS) and David Sheldrick Wildlife Trust.

Woni farm (S 02° 13. 035ˋ, E37° 42. 729ˊ) measures about 160 acres. It borders KWS Kiboko sanctuary and KALRO -Kiboko. It lies in ecological zone V, with domestic animals (goats, sheep, cattle, donkeys and camels). It represented a peridomestic set up. The vegetation of this area has been degraded by human activities like illegal cutting down of trees for charcoal burning, overgrazing by domestic and wild animals (elephants, monkeys, baboons, giraffes). It's characterized by *Acacia commiphora*, baobab trees (*Adansonia digitata*), short thorny thickets and shrubs and perennial grasses. The community being pastoralists, the animals are grazed in the farm by herders. Occasionally, during the dry season, wild animals pass through the farm to graze and to take water. It is served by a permanent river; Kiboko. The community around Woni farm practice horticulture farming where French beans and passion fruits are grown for export through irrigation and also bee-keeping whereby they use a combination of modern langstroth hives and traditional log hives.

3.2 Comparison of performance of commonly used biting flies traps

Four commonly used biting flies traps (Biconical, Pyramidal, Nzi and Ngu) were used in a 4x4 Latin square experiment replicated three times in natural habitat (Kibwezi forest) and in a peridomestic set up (Woni farm). All the traps used for the experiment were standard in that same fabric and colour were used. The traps were made of phthalogen blue and black cotton and white polyester mosquito netting (Mihok, 2006). Metallic poles measuring 1.7m long with a metal spike was used for supporting the traps. Trap cages for the specific traps were of similar in size and quality. The traps were baited with an odor repertoire consisting of acetone with release rate of 500mg/h and octanol dispensed in sachets made of polyethylene with release rate of 0.03mg/h placed 20 cm from the ground level.

Each replicate consisted of four sites where the four traps were set up and rotated every day with respect to site such that each trap-type appeared in each one of the four sites only once in the four day testing period. Traps were set up in the morning (starting at 8.00 a.m.) and emptied at the same time after 24 hours. The experiment was repeated twice over a period of 40 days. Collected insects were transported to the Kiboko Zoological Laboratory and Technology Trial Centre in cool boxes where they were put in a freezer (-5°C) for 30 minutes to immobilize the flies before sorting and identifying them morphologically using standard identification keys (Zumpt, 1973). Catches from each trap type, site, day, habitat and season were carefully sorted out into species, sexed and recorded. Specimens that could not be identified to species level were categorized using their common names.

3.3 Nutrient composition analysis

All insects collected in the biting fly traps (section 3.2 above) were stored in a freezer at -4°C for use in nutrient profiling. However, only *G. pallidipes* and lepidopterans were caught in large enough numbers for nutritional profiling. *G. pallidipes* and lepidopterans (moths and butterflies) collected from the two habitats were defrosted at room temperature for proximate analysis. The weight of the insects was recorded before drying them for 24 hours in an oven at a temperature of 70°C. Dry weight was recorded before analysis. Proximate composition of dry flies was determined using standard methods. Samples were analyzed for moisture content, ash, fibre, fat and crude protein (C.P) according to the methods of the AOAC (1995).

Total moisture content was determined by oven drying method described by Memmert USA. Then cooled to room temperature and weighed on a weighing balance (Metler, Switzerland) to ensure accuracy (AOAC 2000). The formula below was used,

$$Moisture\ \% = \frac{W1 - W2}{W1} \times 100$$

W_1 = Weight (gm) of sample before drying
W_2 = Weight (gm) of sample after drying

Crude protein content was determined by the Biuret and micro Kjeldahl methods according to AOAC method 928.08 (AOAC, 1995). Factor 6.25 was used to convert percentage nitrogen to percentage protein. Protein content was determined as follows:

Total protein = % Nitrogen × 6.25

Total fat (lipid) content was determined by solvent extraction method after Soxhlet extraction according to AOAC method 991.38 (AOAC, 1995) and modified method of lipid extraction by Dyer and Bligh (1959). Lipid content was obtained as follows:

Lipid /100g sample = lipid tube× (volume chloroform in total/amount of sample weighed in (g) *(ml chloroform evaporated)

Total ash content was determined from pre-dried biting fly samples according to AOAC method 920.153(AOAC, 1995). The formula below was used:

$$Ash\ (\%) = \frac{Weight\ of\ ash}{Weight\ of\ sample} \times 100$$

Fibre content was determined by association of official analytical chemists' methods, using the following formula.

$$Crude\ fiber\ \% = \frac{\text{loss in weight on ignition} - \text{loss in weight of ceramic fiber blank}}{\text{weight of sample}} \times 100$$

Fatty acids were determined by gas chromatography

Total lipid content was determined by extraction in diethyl ether for a period of 40 minutes at a temperature of 90 °C. Later sample was put in an oven at a temperature of 70 °C for 30 minutes. Then cooled at room temperature in a desiccator and weighed. The percentage crude fat was then calculated as below:

$$\%\ crude\ fat = \frac{\text{original weight of the sample} - \text{weight of the sample after extraction}}{\text{original weight of the sample}} \times 100$$

3.4 Data analysis

The data on trap catches was tested for normality and subsequently normalized by arcsine transformation before analysis. Analysis of Variance was used to compare mean catches among traps and the Tukey test used to separate the means.

Collected insects were categorized and respective diversity indices computed (Alpha, Shannon, taxon (S)). Diversity indices were compared for each trap and habitat using t-test LSD.

Comparison of Shannon species diversity indices (H′) and abundance among the habitats and trap types was done using t-test

$$s^2 = \left[\sum ni \log ni - \left(\sum ni \log ni\right)^2 / N\right] / N^2$$

$$s_d = \sqrt{s_1^2 + s_2^2}$$

t is given by $t = \dfrac{H_1 - H_2}{s_d}$

The degrees of freedom (Df) in the t-test are calculated using the following formula

$$Df = \frac{\left(s_1^2 + s_2^2\right)^2}{\dfrac{\left(s_1^2\right)}{N_1} + \dfrac{\left(s_2^2\right)}{N_2}}$$

CHAPTER FOUR

4.0 RESULTS

4.1 Performance of commonly used traps in catching biting flies

Biting flies catches were very low in the peridomestic habitat with total mean catches ranging from 0.8 flies/trap/day in the Pyramidal trap to 2.2 flies/trap/day in the Ngu trap which caught significantly higher numbers than the other three traps during the dry season (Table 4.1). Catches rose to 4.4 flies/trap/day for both Nzi and Ngu traps, which were the best performing traps during the wet season. Catches of *Glossina* were very low (0.5 – 2.5 flies/trap/day) in both the dry and wet season and catches varied significantly across the trap types.

Table 4.1: Detransformed mean catches of biting flies (flies/trap/day) ±SE in commonly used traps in Woni farm (peridomestic) habitat, in Makueni County, Kenya, during dry and wet season

		Trap types			
Season	Biting flies	Nzi	Ngu	Biconical	Pyramidal
Dry	*Glossina*	0.5^{a}	0.5^{a}	0.5^{a}	0.5^{a}
	Stomoxys	2.5^{a}	6.5^{b}	2.5^{a}	1.5^{a}
	Tabanus	1.5^{a}	1.5^{a}	0.5^{a}	0.5^{a}
	Total	1.4 ± 0.25^{a}	2.2 ± 0.55^{b}	1.0 ± 0.29^{a}	0.77 ± 0.17^{a}
		(F= 0.594	$d.f$=3	P=0.636)	
Wet	*Glossina*	2.5^{a}	0.5^{b}	0.5^{b}	0.5^{b}
	Stomoxys	2.5^{a}	2.5^{a}	0.5^{b}	0.5^{b}
	Tabanus	9.5^{a}	9.5^{a}	0.5^{b}	0.5^{b}
	Total	4.4 ± 0.5^{a}	3.2 ± 0.69^{a}	0.71^{b}	0.71^{b}
		(F=2.821	$d.f$ =3	P=1.07)	

*Excludes six *Hematopota*, four of which were caught by Nzi and two by Ngu traps, as they were too few to be analyzed statistically. Means in the same row followed by a similar superscripted letter are not significantly different at 5% level.

Table 4.2: Detransformed mean catches of biting flies (flies/trap/day)±SE in commonly used traps in Kibwezi forest (natural) habitat, in Makueni County, Kenya, during dry and wet season.

		Trap types			
Season	Biting flies	Nzi	Ngu	Biconical	Pyramidal
Dry	*Glossina*	491.1^{a}	625.0^{b}	784.0^{c}	207.9^{d}
	Stomoxys	3.5^{a}	0.5^{b}	0.5^{b}	0.5^{b}
	Tabanus	0.5^{a}	0.5^{a}	0.5^{a}	0.5^{a}
	Total	67.90±6.97^{a}	77.65±8.10^{b}	96.24±9.10^{c}	27.88±4.57^{d}
		(*F*=0.070	*d.f*=3	*P*=0.975)	
Wet	*Glossina*	2,531.10^{a}	1,282.36^{b}	365.19^{c}	921.73^{d}
	Stomoxys	6.50^{a}	0.50^{b}	0.50^{b}	2.50c
	Tabanus	0.50^{a}	1.49^{b}	0.50^{a}	0.50^{a}
	Total	318.98±16.24^{a}	158.26±11.62^{b}	46.24±6.13^{c}	118.37±9.74^{d}
		(*F*= 0.157	*d.f*=3	*P*=0.922)	

In the natural habitat (Kibwezi forest) the total mean catch did not vary significantly across the traps (F=0.157, $d.f$=3, P=0.922) and ranged from 27 flies/trap/day in the Pyramidal trap to 96 flies/trap/day in the Biconical trap during the dry season. A similar pattern was observed during the wet season albeit with higher fly catches (Table 4.2), where the mean total catch varied from 118 flies/trap/day in the worst performing trap (Pyramidal) to 319 flies/trap/day in the best performing trap (Nzi). Catches of *Glossina* were high (208 – 784 flies/trap/day) during the dry season and even higher (365-2,531 flies/trap/day) during the wet season and varied significantly across the trap types, with Biconical trap performing the best during the dry season whereas the Nzi trap was the best performing trap during the wet season performing three time better than the Pyramidal trap.

4.2 Diversity and abundance of biting flies and other arthropods in peridomestic and natural habitats.

Insect species caught in different traps were compared using Shannon diversity *t*-test. Results showed that diversity and the number of insects caught varied significantly among the traps (Table 4.3). With a total catch of 3,235 the Nzi trap had the highest number of insects (including arachnids), distributed among 21 taxa with a Shannon index of H'=0.79. This was followed by Ngu trap with 2,477 individuals in 21 taxa too but a slightly lower Shannon index of H'=0.77. Biconical trap caught a total of 1,724 individuals belonging to 15 taxa (H'=0.47) whereas the Pyramidal trap caught the lowest number (959) with the lowest number of taxa (14) but with a Shannon index of H'=0.66.

Comparison of species diversity using Shannon diversity *t*-test between natural and peridomestic habitats revealed significant difference in diversity ($t_{918.81}$=18.182; p=2.4536E-63). Higher diversity was recorded in peridomestic habitat (H' =1.39) where on 21 taxa were identified from 797 individuals, compared to natural habitat (H' =0.39) in which 23 taxa were identified from 7,298 individuals collected (Table 4.5).

Table 4.3: Diversity and abundance of different groups of insects caught in different traps in Kibwezi forest and Woni farm in Makueni County.

Taxon	Pyramidal	Ngu	Nzi	Biconical
Glossina pallidipes	535	2,041	2,652	1,574
Glossina brevipalpis	0	9	25	15
Stomoxys calcitrans	0	6	16	2
Stomoxys inornatus	1	3	4	0
Stomoxys niger	0	0	1	0
Tabanus coniformis	0	1	0	0
Tabanus leucostomus	0	0	1	0
Tabanus taeniola	0	8	16	0
Tabanus gratus	0	2	3	0
Haematopota pluvialis	0	2	4	2
House fly	99	239	321	38
Ants	4	27	13	0
Beetle	1	4	3	3
Blow fly	1	0	3	1
Butterfly	1	13	35	3
Cockroach	0	0	0	1
Cricket	1	1	0	0
Dragon fly	2	0	3	1
Grasshopper	2	21	14	11
Honey bee	0	4	9	8
Moths	6	61	73	13
Praying mantis	0	1	0	0
Wasps	1	24	30	45
Scorpion	0	1	0	0
Spider	1	1	2	0
Tick	4	8	7	7
Taxa (S)	14	21	21	15
Individuals (n)	659	2,477	3,235	1,724
Shannon (H')	0.66	0.77	0.79	0.47
Fisher (α)	2.51	3.15	3.01	2.26
Berger-Parker (d)	0.82	0.82	0.82	0.91

Table 4.4: Effect of three environmental parameters on insect community composition based on Akaike Information Criteria (AIC).

Sources of variation	Df	AIC	*F*	*P*-value
<none>		206.13		
Season	1	206.33	01.6636	0.225
Habitat	1	196.32	15.2786	0.005**
Trap	3	210.26	0.496	0.75

Diversity and abundance of insect species collected varied among seasons both in peridomestic and natural habitat. The highest number of catches was recorded in the natural habitat. A total of 5,349 individuals and 1,949 individuals were collected in the wet and dry seasons, respectively. Low catches were recorded in the peridomestic habitat with 51 and 746 individuals were recorded in the dry and wet season, respectively. However, in both seasons the catches in the peridomestic habitat were diverse with 12 and 19 taxa recorded in the dry and wet season, respectively (Table 4.5).

Table 4.5: Diversity and abundance of different groups of insects caught in different habitats during dry and wet seasons in Kibwezi forest and Woni farm in Makueni County.

	Natural		Peridomestic	
Taxon	Dry season	Wet season	Dry season	Wet season
G. brevipalpis	12	35	0	2
G. pallidipes	1900	4902	0	0
H. pluviaris	2	1	5	0
M. domestica	14	163	21	499
S. calcitrans	4	5	7	8
S. inornatus	0	1	6	1
S. niger	0	1	0	0
T. coniformis	0	0	0	1
T. leucostomus	0	0	0	1
T. taeniola	0	4	1	19
T.gratus	0	1	0	4
Ants	0	37	0	7
Beetles	0	3	1	7
Bow fly	1	1	0	3
Butterfly	1	50	0	1
Cockroaches	0	1	0	0
Crickets	1	0	1	0
Dragon fly	1	0	4	1
Grasshopper	0	13	1	34
Honey bee	8	6	1	6
Moths	3	82	2	66
Praying mantis	0	0	0	1
Scorpion	0	1	0	0
Spiders	0	1	0	3
Ticks	0	26	0	0
Wasps	2	15	1	82
Taxa (S)	12	21	12	19
Individuals (n)	1949	5349	51	746
Shannon (H')	0.17	0.46	1.91	1.28
Fisher (α)	1.70	2.78	4.95	3.55
Berger-Parker (d)	0.97	0.92	0.41	0.67

Among the insect species collected, *G. pallidipes* was the most abundant species with a total of 6,802 flies caught by all four traps, with the catch varying significantly among the four traps (Pyramidal 659, Ngu 2,477, Nzi 3,235, Biconical 1,724 (Table 4.3). Of these, the Nzi trap caught the highest number of *G. pallidipes* (2,652) followed by Ngu trap (2,041), Biconical trap (1,574) whereas the Pyramidal trap caught the lowest number (535). The second most abundant species was *Musca domestica* (house fly) with a total of 470 flies, of which 239 were caught by the Ngu trap and 231 by the Nzi trap. Nzi trap had the most diverse species (H' =0.79) followed by Ngu (H'=0.77). Of the three environmental parameters (season, habitat, trap type) that impacted the composition of catches, only habitat has a significant effect (Table 4.4).

4.3 Nutrient composition analysis

Although there was no significant difference in crude protein content between biting flies (63.02%) and lepidopterans (63.29%), the content in both was significantly higher than in the silver cyprinid (20.8%) (Table 4.6 and Figure 4.1). However, Lepidopterans had a significantly higher fat content (21.84%) than the biting flies (17.9%) both of which were significantly higher than in the silver cyprinid (7.33%) (Table 4.6 and Figure 4.2).

Table 4.6: Proximate composition (%) of biting flies and Lepidopterans trapped in Kibwezi forest and Woni farm, Makueni County.

	Nutrient composition (%/100g dry weight)†				
Insect category	Protein	Fat	Ash	Moisture	Fibre
Biting flies (*Glossina* spp.)	63.02[a]	17.90[a]	3.57[a]	4.30[a]	15.80[b]
Lepidopterans (Moths & Butterflies)	63.29[a]	21.84[b]	4.16[b]	6.49[b]	13.55[c]
Silver cyprinid(Omena) (*Rastrineobola argentea*)	20.80[b]	7.33[c]	4.62[b]	69.5[c]	13.50[c]

† Means of percentage concentration of different nutrients based on 10 laboratory determinations were transformed [ln (x)] before statistical analysis. Tests of significance were performed on transformed means before converting back to the original scale. Means in a column followed by the same superscript alphabetical letter do not differ at $P < 0.05$ (Student's *t* test LSD). The silver cyprinid (Omena) was used as a standard for nutrient comparison.

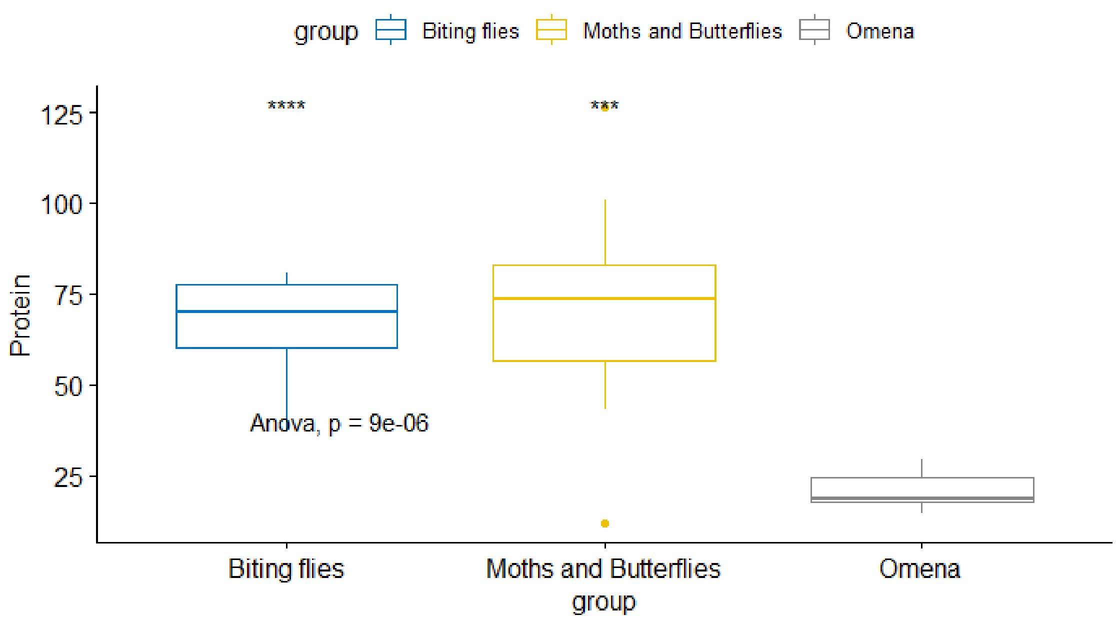

Figure 4.1: Comparative levels of crude protein content in biting flies, lepidopterans caught in Kiboko area of Makueni County.

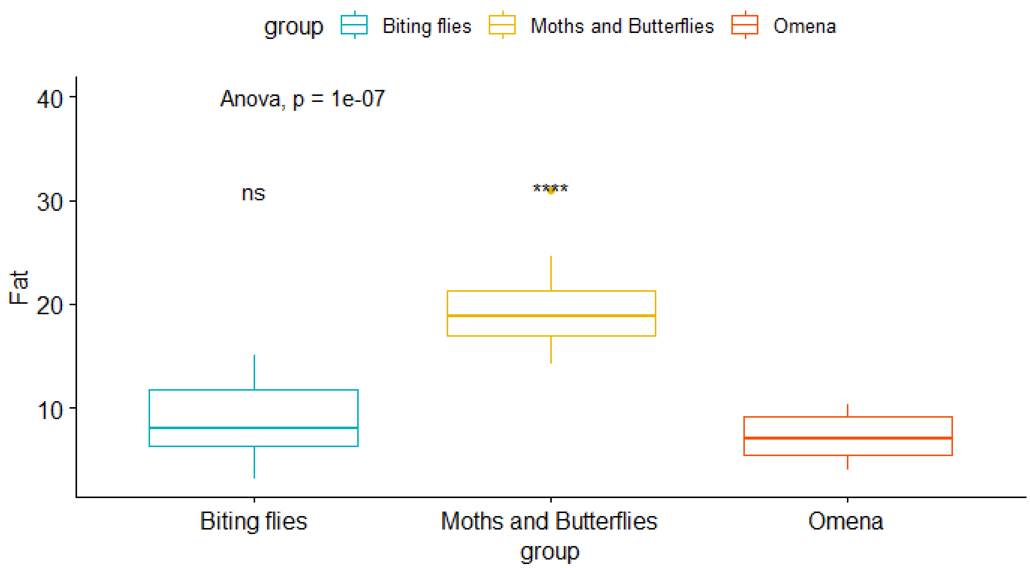

Figure 4.2: Comparative levels of fat content in biting flies, lepidopterans caught in Kiboko area of Makueni County.

There was a significant difference in the ash content within the three groups. Biting flies had a relatively lower (3.57%), though significant, ash content compared to lepidopterans (4.16%). The latter was however not significantly different from the 4.62% recorded in the silver cyprinid (Table 4.6 and Figure 4.3).

Percentage moisture content varied significantly among the three categories, with the biting flies having the lowest levels (4.30%), followed by lepidopterans at 6.49% whereas the silver cyprinid had the highest at 69.5% (Table 4.6 and Figure 4.4). At 15.80%, fibre content was significantly higher in biting flies than in either lepidopterans (13.55%) or the silver cyprinid (13.50%), both of which were significantly lower (Table 4.6 and Figure 4.5).

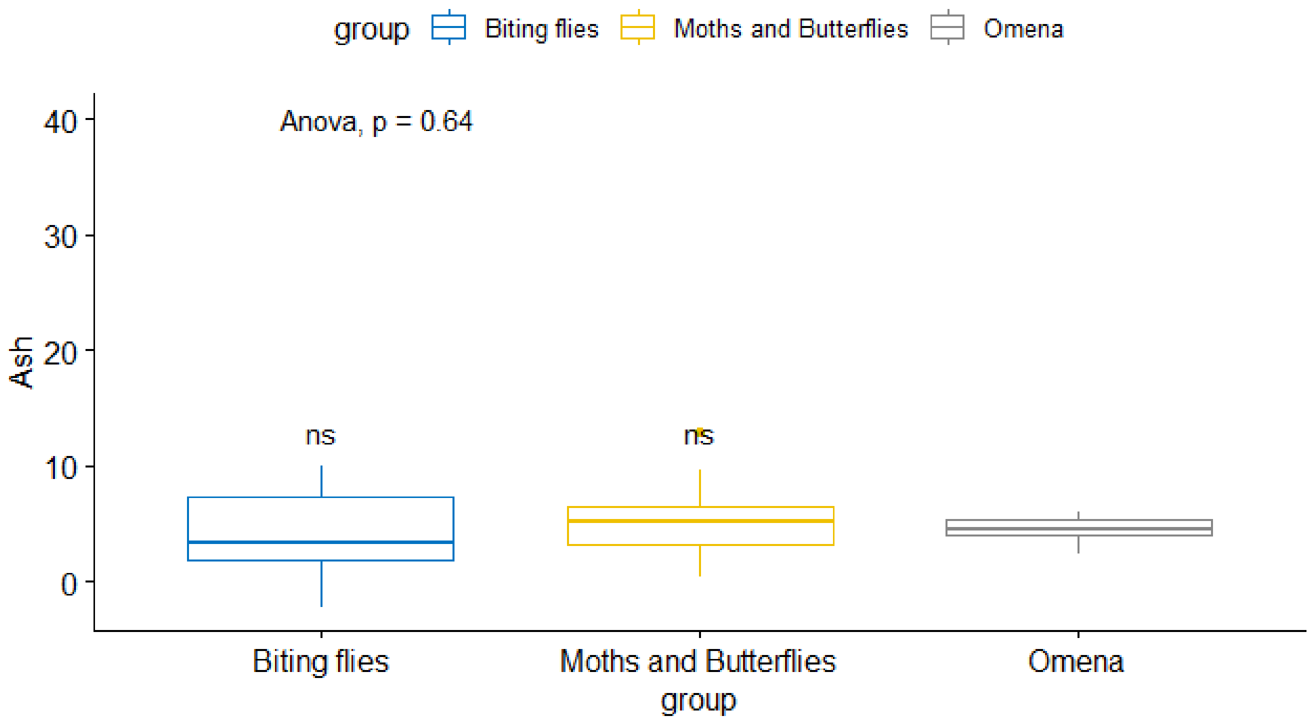

Figure 4.3: Comparative levels of ash content in biting flies, lepidopterans caught in Kiboko area of Makueni County.

There was a significant difference in the fibre content within the insect groups. The highest percentage being recorded in moths and butterflies at 18.33% and the least percentage being recorded in biting flies at 12.8%.

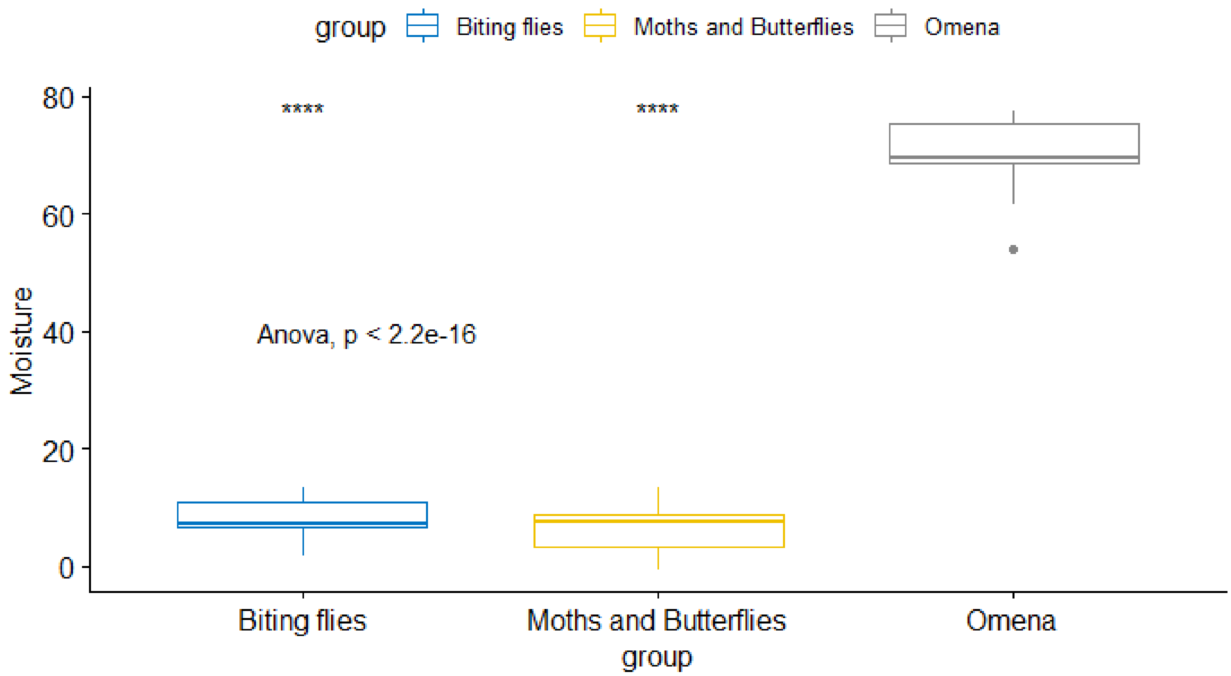

Figure 4.4: Comparative levels of moisture content in biting flies, lepidopterans caught in Kiboko area of Makueni County. The silver cyprinid (Omena) was used as a standard for comparison purpose.

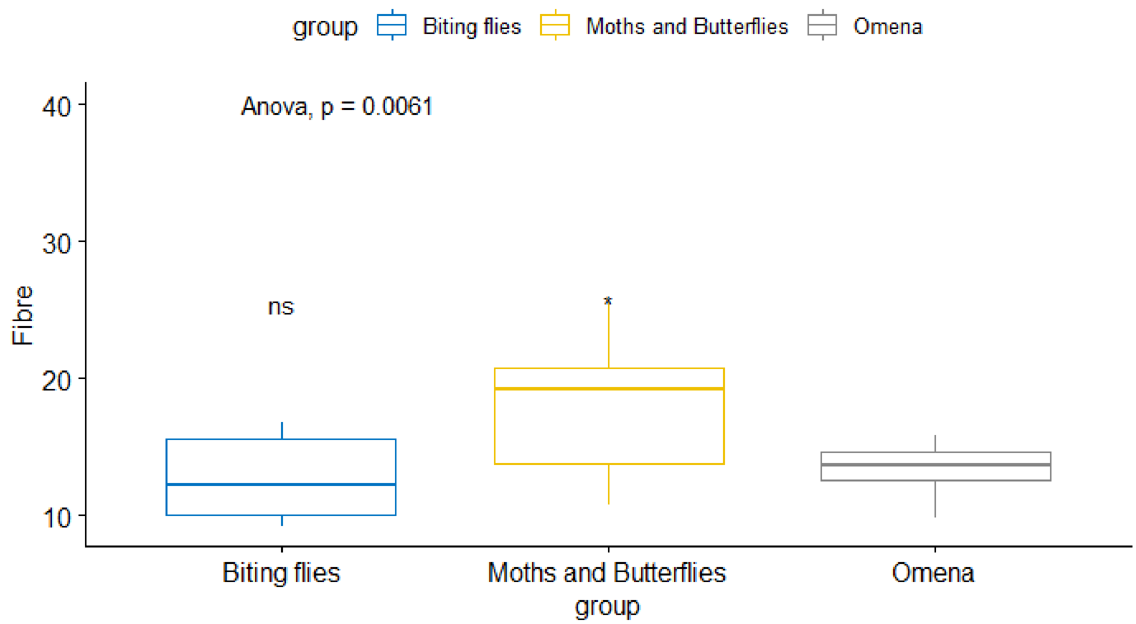

Figure 4.1: Comparative levels of fibre content in biting flies, lepidopterans caught in Kiboko area of Makueni County.

CHAPTER FIVE

5.0 DISCUSSION, CONCLUSIONS AND RECOMMENDATIONS

5.1 Discussion

This study determined the diversity and abundance of biting flies in both peridomestic and natural habitats. Higher diversity was recorded in natural habitat as compared to peridomestic habitat. Majority of the biting fly species were haematophagous and they included *G. brevipalpis, G. pallidipes, H. pluviaris, S. calcitrans, S. inornatus, S. niger, T. coniformis, T. leucostomus*, *T. taeniola* and *T.gratus*. The presence of high numbers of haematophagous flies in natural habitat could be attributed to abundance of hosts (e.g. buffalos and baboons) in the forest, which readily provided blood meal. On the other hand, the low catches of biting flies in peridomestic habitat could be attributed to localized grazing patterns and use of insecticides. Due to localized grazing pattern coupled with disturbance of vegetation cover through charcoal burning, a few or no hosts are available. Results of similar studies undertaken in Tanzania were consistent with findings of this study (Mihok 2007). Unlike this study, Mihok (2007) limited the study to natural habitat, an approach that limits comparison of diversity to natural habitats only.

Traps were used in this study to assess the diversity of biting flies, an approach common in many other similar studies. Unlike the other studies, a total of four different traps (Ngu, Nzi, Biconical and pyramidal) were used in a 4 x 4 Latin Square Design. Among these traps, Nzi and Ngu proved to be the most suitable in both habitats. Nzi trap can be used as a universal trap as it caught the most diverse number of biting fly species. This observation agrees with a study conducted in North America on performance of Nzi trap during which a high diversity biting fly species was recorded (Mihok, 2006). In addition to biting flies, other important insects collected in the aforementioned traps included house flies, ants, beetles, blow flies, butterflies, cockroaches, crickets, dragon flies, grasshoppers, honey bees, moths, praying mantis, scorpions, spiders, ticks, and wasps. Among the biting flies collected, *G. pallidipes* was the most abundant species in the natural habitat during this study. This concurs with studies done by Hyseni *et al*., (2012) who found that *G. pallidipes* is widely distributed in East Africa although later, Ngonyoka *et al*., (2017) showed that most *G. pallidipes* is mainly limited in the riverine woodland with the savanna. In view of these contrasting observations, there is need for further studies to assess and confirm the distribution range of *G. pallidipes* and whether it expands or shrinks with seasons.

Diversity and abundance of important biting flies varied significantly in both domestic and peridomestic habitats during the wet and the dry season across the four traps used. This study concides with similar one conducted in Maasai Steppe in Tanzania which showed that the abundance of tsetse and other biting flies was strongly affected by habitat type and different species were more common in some habitats than others (Ngonyoka *et al*., 2017). Findings showed that *S. calcitrans* existed in both habitats. Most of *G. pallidipes* were collected in the natural habitat (forest). This contrasts a similar observation in Nguruman valley (Brightwell *et al*., 1992).

Insects are the most diverse species in nature with numbers varying among species. Studies done previously indicate that the number of species and their respective abundance vary among habitats and seasons (Finke, 2004), results that are consistent with observations made during this study. The most abundant insects collected during this study were biting flies (*Glossina, Tabanus and Stomoxys*), houseflies and lepidopterans (moths and butterflies). Though abundant, these insects are not among insects considered as potential of source of protein in feed (Van Huis, 2013). These insects are fairly new in term s of consideration as source of protein in and/or feed.

Classical examples of insects so far considered as potential protein sources in food or feed include grasshoppers, locusts, termites and crickets among others with convincing results. These tests show that insects constitute important source of protein sometimes more than *R. argentea* (Comby, 1990). The observed high nutrient content of selected tested insect species is consistent with results in this study. Protein content for the biting flies was 63.2% compared to *R. argentea* which varies between 19.1 and 21.7% (Ogonda *et al*., 2014). These findings agree with results of Ramos-Elorduy 1987 who showed that protein content of edible insects' ranges between 44 and 70%. High protein content of termites, ants, caterpillars has necessitated their use as source of protein in chick feeds while Mormon crickets have been used to replace soybean meal as a major source of protein in chick feed (Finkel *et al.,* 1985). Other than protein, insects are important sources of carbohydrate fat, vitamins and mineral (Ueckert *et al.,* 1972). The observed high levels of ash, fat, moisture and fibre among insects is consistent with results of this study.

Previous studies done on nutritional potential of insects have established a basis of using insects as an alternative source of protein in animal nutrition. Selected insects can either be considered as edible and consumed directly or consumed indirectly as feed. In Botswana, ants and hawk moth larvae are often eaten by chicken i.e. indirect consumption (De Foliart, 1989). The high protein content among other nutrients observed in biting fly and moths during this study suggests that they can be used as an alternative protein source in fish and chicken diets.

5.2 Conclusions

1. This study attests that biting flies are most abundant in the natural habitat than in peridomestic habitats.
2. Nzi trap can be used to harvest biting flies in the wild as it catches a higher diversity of biting flies and other insects
3. The high crude protein content of biting flies makes them good candidates for alternative or supplemental sources of protein in food and feed,

5.3 Recommendations

1. Studies on laboratory rearing of several biting fly species should be done to ascertain the most appropriate one for food/feed.
2. Amino acid and fatty acid composition of biting flies should be investigated to determine their nutritional profile in detail.
3. Non-insecticide contaminated biting fly catches from traps in vector control programmes may be utilized as feed because of their high protein content.

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