SURVEY FOR HEMATOPHAGOUS FLIES AND *TRYPANOSOMA* SPECIES IN IKARA AND KUBAU LOCAL GOVERNMENT AREAS, KADUNA STATE, NIGERIA

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DEPARTMENT OF VETERINARY PARASITOLOGY AND ENTOMOLOGY
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SEPTEMBER, 2016

DECLARATION

I, hereby declare that the work in this Dissertation entitled "**Survey for Hematophagous Flies and *Trypanosoma* Species in Ikara and Kubau Local Government Areas, Kaduna State, Nigeria"** has been carried out by me in the Department of Veterinary Parasitology and Entomology, Ahmadu Bello University, Zaria. The information derived from the literature has been duly acknowledged in the text and a list of references provided. No part of this dissertation was previously presented for another degree or diploma at this or any institution.

Usunobun Collins INEGBENOSUN Date

CERTIFICATION

This dissertation titled **"SURVEY FOR HEMATOPHAGOUS FLIES AND *TRYPANOSOMA* SPECIES IN IKARA AND KUBAU LOCAL GOVERNMENT AREAS, KADUNA STATE, NIGERIA"** by Collins Usunobun INEGBENOSUN, Meets the regulations governing the award of the degree of Master of Science of Ahmadu Bello University, and is approved for its contribution to knowledge and literary presentation.

Prof. A.J. Natala Chairman, Supervisory Committee	_____ Signature	_____ Date
Dr. I.D.Jatau Member, Supervisory Committee	_____ Signature	_____ Date
Dr. O.O Okubanjo Head, Department of Veterinary Parasitology and Entomology	_____ Signature	_____ Date
Prof. K. Bala Dean, School of Postgraduate Studies Ahmadu Bello University, Zaria	_____ Signature	_____ Date

DEDICATION

This Dissertation is dedicated to God Almighty and my loving son Nathan Ose Inegbenosun, wife, Mrs. Constance E Inegbenosun for her understanding, encouragement and support throughout the period of the study.

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ABSTRACT

A study of the species composition and distribution of hematophagous flies was conducted between dry and wet seasons, at Ikara and Kubau Local Government Areas, Kaduna State, using Biconical (Charlier and Laviessiere, 1973) and Nzi traps (Omoogun, 1994). Survey for the occurrence of *Trypanosoma* species in cattle in the study area was also conducted during these periods. Twelve traps each was placed for 48hrs in four districts in each of the local government areas and the trap catches were harvested every 24hrs.A total of 232 flies were caught during the study period and their occurrence differ between the two local governments. Kubau Local Government had 105(45.3%) while Ikara Local Government had 127 (54.7%).The specific occurrences of the hematophagous flies caught were: *Stomoxys calcitrans*107 (85.3%), *Tabanus* 9(7.1%) and *Glossina* 0(0.0%). Overall percentage of fly catches per trap was 130 (56.0%) for biconical and 102(44.0%) for Nzi traps respectively. The highest number of flies caught was during wet season {212 (91.4%)} while in dry season only 20(8.6%) flies were caught during the study. There was significant association (P=0.002) between the flies occurrence and the two seasons of the year. While there was no statistically significant association (P=0.07) between the number of flies caught and the type of trap used. The prevalence rate of *Trypanosoma* infection in cattle in the local government areas was 50% prevalence each of infection due to *T. vivax* and *T. brucei* respectively. There was no significant association between the occurrence of the *Trypanosoma* infection and the Local Government Areas under study.

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CHAPTER ONE

1.0 INTRODUCTION

1.1 Background of the Study

Hematophagous flies belong to the families Glossinidae and Tabanidae and to the genus *Stomoxys*. They play an important role in both human and domestic animals health, because many species of these groups are vectors of organisms responsible for several human and animal diseases (Stephen, 1989). Moreover, several vector-transmitted diseases are considered as emergent due to their recent evolution and propagation. The preponderant role of the species belonging to the Glossinidae family in the transmission of African Human Trypanosomosis (AHT), or sleeping sickness and AAT has historically hidden the potential role of other hematophagous flies, like those belonging to the genus *Stomoxys* and the family Tabanidae in trypanosome transmission and the transmission of other pathogens. For example, it is now recognized that several species of the genus *Stomoxys* are mechanical vectors of parasites, such as *Trypanosoma* species (Phelps *et al*, 1978) and various viruses, such as the *Capripox-viruses* responsible for lumpy skin disease in sheep and goats. The Tabanidae are also mechanical or biological vectors of many human and animal pathogens and an analogous pattern of *Trypanosoma* transmission has been documented for several tabanids of the *Atylotus* genus. Species of the *Chrysops* genus are involved in the cyclical transmission of *Loa loa* filariasis (Morlais *et al*.1998).

The feeding action of these hematophagous flies opens a channel for contamination of the host species with disease causing agents. Thus, many animal and human infectious diseases are transmitted by hematophagous species, such as the bubonic plague, chagas disease, dengue fever, eastern equine encephalitis, filariasis, leishmaniasis, Lyme

disease, malaria, rabies, sleeping sickness, St. Louis encephalitis, tularemia, typhus, Rocky Mountain spotted fever, west nile fever and many others. Insects and arachnids of medical and veterinary importance that are hematophagous, at least in some species, include the sand-fly, black fly , tsetse fly, bedbug, assassin bug, mosquito, tick, louse, mite, midge, and flea (Maudlin,1970).

1.2 Statement of Research Problem

Hematophagous flies activity is highly seasonal. For example, most of the *Glossina* species respond to seasonal patterns, and within a region, the populations of the different species increase in the rainy season (Shaba *et al.*, 2012). The *Tabanus* and *Stomoxys* species are caught throughout the year but there is usually a seasonal rise in abundance corresponding to the ends of the rains and the beginning of the dry season (Dede *et al.*, 2005). However, the basic seasonal pattern of the different groups is influenced by local climatic parameters and species exhibit various patterns of population fluctuation related to local climate, vegetation and host blood meal source (Challier, 1982), which also coincides with the incidence of the diseases they transmit. Trypanosomosis is the most economically important disease transmitted by these hematophagous flies most importantly by *Glossina* species (Moiser, 1912).

Trypanosomosis lowers productivity in livestock, reduces cattle density by 70%, sale of meat and milk by 50% and calving rates by 20% and increases calf mortality by 20% (Swallow, 2000). Tsetse flies, through the cyclical transmission of trypanosomosis to both humans and their animals, greatly influence food production, natural-resource utilization and the pattern of human settlement throughout much of sub-Saharan Africa (Kabayo, 2003). It is estimated that the annual direct production losses in cattle alone

amount to between US$6billion and $12billion, while animal deaths may reach 3 million (FAO, 1992).

According to Lehane *et al*. (2003), tsetse flies transmit African trypanosomosis leading to half a million human cases annually and that the disease also known as Nagana in animals remains a massive break on African Agricultural development.

More than a third of the land area across Africa is infested with tsetse flies (8.7 million km^2), where at least 46 million cattle are exposed to the risk of contracting tsetse-borne trypanosomosis, as are millions of sheep, goats, donkeys, camels and horses (Reid *et al*., 1998). African livestock producers are administering an estimated 35 million curative and preventive treatments annually (Geerts and Holmes, 1997).

Apart from disease transmission, biting flies can produce an array of symptoms to animals including pain, itching, urticarial and cellulitis. Allergic response is the most common which may be characterize by hives, and in some cases wheezing (Ripamonti, *et al.,* 2002). Tabanid bites are very painful with some individuals developing severe lesion, fever and general disability. These allergic responses are due to the large amount of saliva injected by the flies to prevent their blood meal from clotting. Stable flies bites are quite painful and they produce small papules that quickly fade but are often itchy (Maldonado, 1910).

The design of management strategies against the different species and of strategies that minimize the risk of fly bites and transmission of diseases such as trypanosomosis requires a full regional understanding of the species' phenology and ecology. However, such information is scarce in Northern part of Kaduna State. Hence, the present study was conducted to improve our understanding of the distribution, the abundance, and the

phenology of hematophagous flies (Glossinidae, Tabanidae Muscidae) in the Ikara and Kubau Local government areas of Kaduna State.

1.3 Justification

Trypanosomosis is one of the major animal health constraints to livestock production in Sub-Saharan Africa (FAO, 2002). In fact, it is also an important blood parasitic disease in humans (Shaba *et al.,* 2012). Animals have a central role in most African societies, and provide milk, meat, manure, hides and skins as well as valuable draught power. Additionally, because livestock represent a means of accumulating and distributing wealth, they have great social significance.

Control of trypansomosis is aimed against either on the tsetsefly or the trypanosome and in the absence of adequate funds for large-scale tsetse control, trypanocides are widely used. At the farmer's level trypanocides provide a way out for the individual to take action to control the disease. With few exceptions throughout Africa, governments have lacked the resources to continue to provide effective Veterinary services to control trypanosomosis, among other diseases.

Survey of *Glossina* (tsetse fly) and other biting flies and the *Trypanosoma* species they transmit is an essential tool for strategic control measures against the vectors and etiologic agent of trypanosomosis. This will go a long way to improving animal production in Nigeria. This study is likely going to help in reducing the economic waste associated with treating the disease by livestock owners and appropriate control and preventive measures will be recommended.

Ikara and Kubau Local Government Areas of Kaduna State are situated in the Northern Guinea savannah area which is tsetse endemic and a favourable settlement area for cattle pastoralist (Ohaeri, 2007). Therefore, there is need to undertake this study to

ascertain the potentials of these areas in terms of animal productivity and suggestive suitable tsetse control.

1.3.1 Aim of the study

To survey for hematophagous flies and *Trypanosoma* species in Ikara and Kubau Local Government Areas, Kaduna State, Nigeria.

1.3.2 Objectives of the study

The objectives of this were to:

- Investigate the distribution of hematophagous flies of veterinary importance in Ikara and Kubau LGAs of Kaduna State using trapping.
- Compare the catching efficiency of Biconical and Nzi traps in hematophagous flies of veterinary importance in Ikara and Kubau LGAs of Kaduna State.
- Determine the occurrence of *Trypanosoma* species in cattle in Ikara and Kubau LGAs of Kaduna State.

1.3.3 Research Questions

- Are there hematophagous flies of veterinary importance in Ikara and Kubau Local Government Areas of Kaduna State?
- Which trap between biconical and Nzi traps is more efficient in catching fly in Ikara and Kubau LGAs of Kaduna State?
- Are there *Trypanosoma* species in cattle in Ikara and Kubau Local Government Areas of Kaduna State?

CHAPTER TWO

2.0 LITERATURE REVIEW

2.1 Historical Perspective

It is believed that hematophagy arose independently at least six times among the arthropods of the Jurassic and Cretaceous periods (145–65 million years ago) (Balashov, 1984; Ribeiro, 1995).The very patchy nature of the insect fossil record means that discussion of the evolution of the bloodsucking habit has until now relied heavily on detective work, with the major clues lying in the diversity of forms and lifestyles seen in modern-day insects, and in some cases in the details of their relationships with vertebrates. From careful interpretation of this evidence quite credible accounts of the likely evolution of the blood-sucking habit can be made. From this starting point, it has been convincingly argued that the evolution of the blood-sucking habit in insects has occurred on several occasions, in each case along one of two main routes (Waage, 1979), and these are discussed below. Insect molecular systematic is beginning to emerge from its 'Tower of Babel' stage (Caterino *et al.*, 2000) and it will make a major contribution in defining the detail of the evolutionary routes taken by hematophagousinsects (Esseghir *et al.*, 1997; Hafner *et al.*, 1994; Lanzaro *et al.*, 1998; Mans *et al.*, 2002; Sallum *et al.*, 2002). The proposed population bottleneck suffered by phlebotomines in the late Pleistocene and the subsequent radiation of the species out from the eastern Mediterranean sub-region is a good example of what we can expect (Esseghir *et al.*, 1997).

Hematophagy can be classified into obligatory and optional practice. Obligatory hematophagous animals do not have any other type of food besides blood; one such species is Rhodnius prolixus (an assassin bug from South America). This contrasts with

optional hematophagous, like the many mosquitoes species, such as *Aedes aegypti*, which may also feed on pollen, fruit juice, and other biological fluids. Sometimes only the female of the species is a hematophous (this is essential for egg production and reproduction).

Hematophagous flies, and among those, species belonging to the families Glossinidae and Tabanidae and to the *Stomoxys* genus.

2.2.0 General description of *Glossina*

Glossina species have been widely studied because of their economic importance as major transmitters of animal and human trypanosomosis (Ndams, 1987).

Tsetse flies are robust insects measuring about 6-15mm in length. They are readily distinguished from other biting flies morphologically by the combinations of useful features such as widely separated compound eyes which are dark brown, and able to detect moving objects at 137meters (150yards) (Pollock, 1982), with forwardly projecting proboscis which sticks out horizontally from the front of the head (Jordan,1993). They differ from most biting flies and non cyclorrhaphous insects because when at rest, the wings are held over the other such that they overlap like the blade of a closed pair of scissors, thus revealing part of the abdomen (Davies, 1977). They also possess a characteristic wing venation where the discal medial cell of the wing is shaped like a butcher's cleaver (referred to as the 'Hatchet cell') and a distinctive row of branched hairs on the arista of the antenna (Service, 1980; Jordan, 1993).

Tsetse flies range in color from yellowish or grayish to dark or almost blackish-brown, sometimes there is a slight pink or sandy red tinge (Davies, 1977). The dorsal surface has a pattern of dark brown stripes and patches making the insect difficult to see when

settled on tree bark, rock and soil. They have seven visible abdominal segments Pollock, 1982).

2.2.1 Habitat and distribution of *Glossina*

The genus *Glossina* occurs over some 11 million $km^2$of Africa. Its northern limit extends across the continent from Senegal in the west to southern Somalia in the east. This limit is at about 14^oN but in Somalia it is only about 4^oN. The northern limit corresponds closely to the southern edges of the Sahara and Somalia Deserts. The southern limit is less well defined. In the south west it ranges between $10^o$and 20^oS, corresponding closely to the northern edges of the Kalahari and Namibian Deserts, whereas in the south east, it is generally at about 20^oS but extends as far as 29^oS along the east African littoral (Jordan, 1986), with the mid pint of infestation at about 7^oS (Davies, 1977). In Nigeria, tsetse flies cover about 75% of the landmass from latitude 4^o to 13^oN to longitude 2^o to 15^oE, an area covering all the five agro-ecological zones of the country (Okam, 1988; Onyiah, 1997).

There are twenty two known species of *Glossina* which can be arranged into three distinct species groups based on habitat preference (Ford, 1970) and morphological differences in the construction of the male genitalia (Pollock,1982). These species groups are summarized below:

The Morsitans species group (Morsitans) these include seven species and subspecies namely:- *G. longipalpis; G. pallidipes; G. morsitans morsitans; G. morsitans submorsitans; G. morsitans centralis; G. swynnertoni; G. austeni (*Bossche *et al.,* 1997).

The Palpalis groups (Nemorhina) consists of a total of nine species and subspecies namely: - *G. palpalis palpalis; G. palpalis gambiensis; G. fuscipes fuscipes; G. fuscipes*

martini; G. fuscipes quanzensis; G. tachinoides; G. pallicera pallicera; G. pallicera newsteadi; G. calliginea (Grev, 2007).

The Fusca species group (Austenia) also consists of fourteen species and subspecies namely: *G. Fusca Fusca; G. Fusca congolensis; G. nigrofusca nigrofusca; G. nigrofusca hopkinsi; G. fuscipleuris; G. haningtoni; G. schwetzi; G. tabaniformis; G. nashi; G. vanhoofi; G. medicorum; G. severini; G.brevipalpis; G. longipennis* (Rogers, *et al.,* 2000).

2.2.2 Habitat and distribution of the morsitans group

The three *Glossina* subspecies of the morsitans group are exceptionally good vectors of trypanosomes; all the seven species are potential vectors of both human and animal trypanosomosis. All species within this group inhabit the savanna woodlands that surround the two major blocks of lowland rainforests in Africa, and as such are referred to as 'game or savanna flies' (Davis, 1977). The distribution of tsetse flies in this group closely follows the distribution of wild animals and water sources. In wetter areas, the flies are observed to disperse more widely over the woodland, but in drier areas their movements are 28 restricted to the mesophytic vegetation of the watercourses, particularly during the severe dry season (Nash, 1937; Jordan, 1986). In Eastern and Southern Africa where *Glossina morstans morsitans* is the primary vector of human and animal trypanosomosis, the 'Miombo' woodland (Brachystegia–Jilbernardia) that extends from Mozambique to Tanzania, as well as the 'Mopane' woodlands (Colophospermum mopane) in Zambia and Zimbabwe are typical habitats. *Glossina morsitans centralis* dominate northwards from Botswana and Angola into southern Uganda.

Glossina morsitans submorsitans has an east to west distribution from Ethiopia to Senegal in 'doka' woodland where the vegetation is dominated by *Isoberlinia doka* species, and can be sporadically found to occur in the southern Guinea savanna vegetation zone as well as the drier Sudan zone (Jordan, 1986).*Glossina swynnertoni* is restricted to a small area between Tanzania (Serengeti) and southern Kenya (Masaimara) where Acacia- commiphora vegetation can be found, with abundant wild life. *Glossina longipalpis* and *Glossina pallidipes* both have a much wider range of possible habitats displaying versatility by existing in different vegetation types. *Glossina longipalpis* occurs in the narrow savanna belt just north of the rainforest in West Africa, from Guinea to Cameroon while *Glossina pallidipes* occurs in East Africa from Mozambique to Ethiopia over a relatively wide range of climatic and vegetation conditions. Finally, *Glossina austeni* occupies secondary shrub, thickets and islands of forests along the East African coast from Mozambique to Somalia, are rarely found at altitudes over 200m or more than 250km inland from the coast (Jordan, 1986). In Nigeria, the two morsitans group species present are:-*Glossina morsitans submorsitans* found in the north where annual rainfall is as low as 635mm (25in) with a dry season of seven months and further south, an annual rainfall of 1400mm (55in) with a dry season of four months. *Glossina longipalpis* is found in the southern guinea and derived savanna zones in the west and a small localities in the east, with annual rainfall not less than 1150mm 45in) or more than 2300mm (90in) (Davies, 1977).

2.2.3 Habitat and distribution of the palpalis group

Out of the nine species in the palpalis subgenera, only five *palpalis* and *fuscipes* subspecies are vectors of both human and animal trypanosomosis. Although flies in this group are continuously found in the lowland rainforest, some are known to extend out of the savanna region particularly along rivers and streams. The habitat of

the palpalis flies occurs mainly in the drainage systems leading to the Atlantic or the Mediterranean ocean, extending from the wet mangrove and rainforests along the coastal regions of West Africa to the drier savanna areas just north of the rainforests. The flies of the palpalis group are less tolerant to the wide range of climatic conditions of the savanna belt, and are therefore restricted to the ecoclimate of the water courses from where they derived their label as the 'riverine species'. Many of the palpalis species, such as the *Glossina palpalis palpalis* in Cote d'lvoire prefer peri-domestic conditions and have been observed to maintain close association with villages (Baldry, 1980). Similarly, it is thought that the advancement of *Glossina tachinoides* in Cote d'lvoire and Togo have been attributed to intense agricultural development and the rapid human population growth around the plantation (Hendrick *et a*l., 1997). In general, most of the palpalis group flies are less suited to desiccating conditions, and therefore survive in thick riverine forests with enough shelter from wind and heat. This is especially the case for the three fuscipes subspecies which are confined to hygrophytic habitats rarely far from open water lacustrine or riverine habitats. *Glossina tachinoides*, although typically a riverine species, were found in Northern Nigeria to extend into human-inhabited savanna woodlands during the wet season, displaying strong adaptation to peri-domestic habitats (Kuzoe, 1985; Ahmed, 2004).

2.2.4 Habitat and distribution of the fusca group

With the exception of *Glossina brevipalpis* and *Glossina longipennis* all the tsetse flies in the Fusca group are found in West African forests. None of the species in the Fucsa group are vectors of human trypanosomosis; however both *Glossina fusca* and *Glossina medicorum* are efficient vectors of trypanosomosis to livestock (mainly *Trypanosoma vivax*) causing considerable economic burden. Distribution of the Fusca group depends primarily on forest vegetation and climatic factors. With the exception of *Glossina*

longipennis, most Fusca group species inhabit moist, evergreen habitats either in riverine forests with savanna (such as *Glossina medicorum*) or in dense and wet rainforests (*Glossina tabaniformis* and *Glossina nigrofusca*).In stark contrast to the rest, the *Glossina longipennis* species lives in one of the driest habitats inhabited by tsetse flies (Jordan, 1986). Due to its pupal adaptation to dry conditions, their primary habitats – consisting of dry deciduous acacia bush- are discontinuously spread throughout East Africa (Glasgow, 1963).

2.3.0 Reproduction of *Glossina*

Tsetse flies exhibit a form of reproduction known as adenotrophic viviparity (Hagan, 1951) because the egg and larva stages develop within the fly. The egg contains sufficient yolk for embryonic development and the larva in the uterus is nourished by special maternal organs. The consequences of viviparity are that only a small number of fully developed larvae can be produced, a free-larval stage is practically eliminated and both adults and immature stages are dependent upon the same source of food (blood) (Saunders, 1960).

2.3.1 Mating

Female tsetse flies mate within a day or so after emergence from their pupal case; mating which usually takes place near or on host animals (Pollock, 1982). Male and female generally meet when the female is about to take the first blood meal or is in the process of doing so (Jordan, 1986). Almost at the end of the period spent in copulo the male jerks vigorously and it is at this time that sperms are ejaculated into the spermatophore (Pollock, 1982). Within the next few hours, the sperms are slowly released from the spermatophore and move into the spermathecae, a paired golden-colored rigid structure connected by ducts to the anterior end of the uterus. Active and

viable sperms can remain in the spermathecae throughout the life of the female fertilizing each egg as it is produced from the ovaries, thus allowing the female to breed throughout her life time (Davies, 1977). Female tsetse flies usually mate only once in their lives but some may mate more than once, males can mate several times. Older males are better able to mate successfully than very young ones (Pollock, 1982).

2.3.2 Egg stage

The female tsetse fly has two ovaries, each of which has two ovarioles; eggs develop sequentially in the four ovarioles (Saunders, 1970) and are ovulated into the uterus at intervals of about 9 – 10 days, the first ovulation occurring when the fly is about 9 days old (Jordan, 1986). Each egg is fertilized immediately it enters the uterus by sperm from the spermathecae coming into contact with and penetrating the anterior part of the egg. The fertilized egg remains lying in the uterus for about four days, while development of the first instar larva takes place inside (Pollock, 1982). The age of wild-caught females can be determined through dissection technique involving ovaries examination 33 and counting the number of ovulations that have occurred (Saunders, 1962; Challier, 1965).

2.3.3 Larval stages

The larva in *Glossina* passes through several stages or instars, as it grows. There are three larval instars in *Glossina*: the first, second and third instars. The larva has a mouth at the anterior end, and two posterior spiracles (Pollock, 1982). The intrauterine larva is supplied with nutrients in the form of ‘milk’ substance secreted from a modified accessory gland (Attardo *et al.*, 2006), grows rapidly and molts twice before larviposition.

2.3.3.1 First instar larvae

The first instar larva develops within the egg and breaks out of the chorion using a sharp larvae tooth. It grows to 1.8mm (*Glossina morsitans*) and lasts for a day (Pollock, 1982).

2.3.3.2 Second Instar Larvae

The second instar larva grows and develops rapidly lasting for two days and can reach the length of 4.5mm (*Glossina morsitans*). Each side of the posterior spiracles swells with small spines in between.

2.3.3.3 Third instar larvae

This also grows and develops rapidly. The third instar larvae is white in color and has two conspicuous black respiratory lobes which are white at first, and later become black. The third instar lava last just over two days and grows to a length of 6.7mm (*Glossina morsitans*) (Pollock, 1982). The larva when fully developed is deposited (larviposited) on the ground at shady sites to prevent desiccation. The larva burrows itself rapidly in the soil to a depth of 1- 5cm, depending on the species, the season and the soil type (Grev, 2002). Within an hour, the larva contracts to form a barrel-shaped puparium, darkens rapidly to black. After about four days, ecdysis occurs within the shell of the puparium and the true pupa is formed (Jordan, 1986).

2.3.4 The pupa

The pupa is dark brown and rounded; at the posterior end are the polypneustic lobes, the shape of which helps to distinguish the tsetse pupa from the pupae of other flies. The pupa is slightly shorter than the larva. The pupal stage usually lasts about four to five weeks, depending on the temperature. Higher temperatures shorten the pupal period,

lower temperatures lengthen the pupal period. Too high or too low temperatures cause death of the pupae (Pollock, 1982).

2.3.5 The adult fly

The adult fly emerges by expanding its ptilinum to burst open the end of the puparium, pushing itself out of the soil after which the ptilinum sink back into the head of the fly (Davies, 1977). At this stage, the body is very soft and the wings are small and crumpled (Pollock, 1982). The wings flatten out into their normal shape within a few minutes of birth by blood being forced into the veins, harden and is then capable of flight. The sex ratio on emergence is normally 1:1 (Jordan, 1986). The young fly before its first blood meal is called 'Teneral fly', the abdomen which appears whitish and semi-transparent with ptilinum everted when squeezed between fingers on the head. The non-teneral fly in contrast, are flies that have taken blood meal. They appear creamier yellow, the thorax is firmer and hard and the ptilinum is not easily everted (Pollock, 1982).

2.4.0 General behavior of *Glossina*

The behavior, distribution pattern and density of a population of flies depends mainly on climate (temperature, humidity, sun, rain etc.); vegetation (shade and shelter); wild animals(food); soil(breeding sites); predators of tsetse and human population (Davies, 1977).

2.4.1 Movement and activity of tsetse flies

The movement and dispersal of tsetse flies are related to the climate, the hunger stage of the fly and the sex of the fly (Grev, 2002). The flies are active during the day spending only about 15 – 30 minutes each day in active flight. A single flight does not last longer than about 1½ - 2 ½ minutes and the speed of flight may be 3 -6 m/sec (Pollock, 2000).

They are usually inactive during low temperatures and dull days; some species have been found flying in the moon light (Service, 1980). Dispersal is higher in the wet period, during these periods they spread all over the savanna and have implications for transmission of African trypanosomosis; such fly movement facilitate both the spread of the disease to new areas and their reintroduction to areas where it was previously under control. In drier periods, where unfavorable conditions prevail, they utilize places with dense vegetation close to water where suitable climatic condition exist (Ndams, 1987; Grev, 2002). The activity of tsetse flies during the daytime is mostly early morning or late afternoon. Females are only active for a few moments a day while mature males can be active up to 30 minutes a day (Grev, 2002).

2.4.2 Resting sites

Resting sites may vary according to the time of the day or night, climate and season, species of tsetse fly, vegetation and resting places of host animals. During the hottest part of the day (usually early to mid-afternoon), the true resting sites are lowest down on tree trunks, and on the underside of shaded, fallen logs. At cooler times of the day, and in cooler seasons, the flies rest 37 higher up tree trunks, and on the underside of branches. At night some flies go up into the canopy of trees and rest on the leaves or twigs (Pollock, 2000).

2.4.3 Response to host animals

Response to host animals is usually by tsetse fly sense of smell and sight (Davies, 1977), from up to one hundred meters away, with larger host animals being more attractive to tsetse fly than smaller host animals (Pollock, 2000). Flies moves up-wind closer to the host animal when it smells the host. Tsetse fly land on a greater variety of host animals; flies that show the most attraction to the host animals are usually the most

hungry flies in the population, while the non-hungry flies, particularly the males make up the 'following swarm'. A male in a following swarm may fly on to a virgin female as she comes for her first meal, and mate with her (Pollock, 2000).

2.5.0 Tsetse fly population dynamics

Tsetse fly populations are influenced mainly by density-independent factors such as temperature and humidity, which in turn depend on vegetation cover. Fly densities are determined by factors such as the availability of hosts or suitable habitats, which in turn are influenced by human activity. According to Pollock, (2000) the density of a tsetse population in a given area is never very accurately known unless all the flies are caught, this could only be done on an island or in a very isolated woodland or thicket; even then the fact that more than half of the total population of tsetse fly in an area are present below ground as pupae, makes the estimation of a tsetse population a difficult exercise. In some parts of Africa the fly population per square mile has been calculated, but in most of Nigeria, flies are not often evenly distributed over any area because of local differences in vegetation and climate, it is therefore difficult to determine the population this way (Davies, 1977). In estimating tsetse population the term apparent density and true density are sometimes used. This does not necessarily give information about the 'true density' (the number of flies per unit area) which may be heavy in a certain place indicating a hungry population rather than a dense one. The apparent density on the other hand may differ from time to time according to the availability of the flies, when conditions for catching are poor, the apparent density will be low even though the true density may be high (Davies, 1977). When traps are used, apparent density is defined as the number of flies per trap per day (F/T/D).

2.6.0 Transmission of trypanosomosis

Tsetse flies transmit the protozoan parasite of the Genus *Trypanosoma,* the agents of human and animal trypanosomosis in sub-Saharan Africa (Hu *et al.*, 2006). In West Africa, the human trypanosomosis caused by T*rypanosoma brucei gambiense* is transmitted by *Glossina palpalis* and *Glossina tachinoides,* the disease is devastating and chronic. It is described as Gambian sleeping sickness (Dutton, 1902; Abenga *et al.*, 2005), whereas animal trypanosomosis caused by *Trypanosoma brucei brucei* is transmitted by *Glossina submorsitans* and *Glossina longipalpis.* In East Africa, both the human and animal trypanosomosis caused by *Trypanosoma brucei rhodesiense and Trypanosoma brucei brucei* respectively are transmitted by *Glossina morsitans, Glossina pallidipes* and to some extent *Glossina palpalis* (Willet, 1970; Grev, 2002). The East African form described as Rhodesian form (Stephen and Fantham, 1910) is a more acute, lasting for weeks or months (Garcia *et al.*, 2006).

Trypanosomes exist as trypomastigote in blood and lymph in infected animals (Pollock, 2000); they are slightly curved elongated protozoan measuring about 10 - 35µm with a single nucleus. Each possesses a single flagellum which originates near the posterior end of the body and extends forward to the body by an undulating membrane. Near the base of the flagellum is the dark-staining kinetoplast. The size and shape of the body, position of the nucleus and kinetoplast, length and form of the undulating membrane and flagellum are the diagnostic characters of various species of Trypanosomes (Jordan, 1986).

The form of trypanosomes in the tsetse fly can be identified to subgenus level on morphological grounds and on their sites of development within the fly. In the subgenus *Duttonella* is the *Trypanosoma (vivax*group) of trypanosomes with large terminal kinetoplast, distinct free flagellum and an inconspicuous undulating

membrane. *Trypanosoma vivax* is a large (18 - 26µm long) monomorphic organism that is very active in wet – mount blood smears. Their development within the tsetse fly is restricted to the proboscis (labrum or hypopharynx). In the subgenus: Nannomonas is the *Trypanosoma congolense* which is a small trypanosome with medium – sized marginal kinetoplast, no free flagellum, and a poorly developed undulating membrane. *Trypanosoma congolense* are hematic trypanosome found only in the blood vessels of the animal they infect. They develop within the proboscis (labrum or hypopharynx) and midgut of the tsetse fly. In the subgenus: Trypanozoon is the *Trypanosoma brucei brucei;* this group is an extremely polymorphic; while some trypanosome occur as short, stumpy organisms without flagella, others are found to be long slender organisms with distinct flagella, and there are intermediate forms that are usually flagellated (Moulton, 1976). These trypanosomes develop within the proboscis, midgut and salivary glands. However, when trypanosomes are found only in the labrum, they are regarded as immature form of all the subgenus mentioned, whereas if they are found in the labrum and midgut, it indicates immature form of the congolense group (Davies, 1977).

When trypanosomes are ingested by tsetse fly, they undergo a cycle of development within the fly. In the gut, they transform into the trypomastigote form and move into the mouthpart (labrum and/or hypopharynx) where they develop into the epimastigote form; they later reproduce by binary fission to produce the metacyclic form which is the infective form. This final binary fission takes place in the proboscis, midgut or salivary gland depending on the species of trypanosome (Jordan, 1986). However, transmission of trypanosomes can also occur from one mammalian host to the other in or on the mouthparts of various species of biting flies e.g. *Tabanus, Stomoxys, Chrysops* etc including tsetse; this process is known as mechanical transmission (Jordan, 1986). This

cycle of development varies in duration depending on the species of trypanosome, species of tsetse fly, temperature, reservoir host, age, sex. For *Trypanosoma vivax, Trypanosoma congolense* and *Trypanosoma brucei brucei,* it varies from 5 – 13 days at 22ºC - 29ºC; 12 -53 days and 11 – 60 days respectively (Davies, 1977; Jordan, 1986). Reports by various authors have showed that *Trypanosoma vivax* have the highest infection rate, followed by *Trypanosoma congolense* and then *Trypanosoma brucei brucei* (Onyiah, 1997; Omotainse *et al.*, 2000).

2.6.1 Control of tsetse flies

Attempts to control tsetse and trypanosomosis date back nearly 100 years, employing a range of methods and approach. This range of methods have been developed and put into practice to keep the disease under control, some of them less good than others (Grev, 2002). The control strategies over the years have been directed against both the parasites and the vectors (Onyiah, 1997). The initial methods of tsetse control comprised of Hand-catching (Glasgow, 1970), Bush clearing (Steiner, 1964; Ford *et* al., 1970), Game destruction, Settlement of people (Davies, 1977) and the use of chemicals as insecticides (Davies, 1964). More recently, are the use of traps and screens (Challier and Laveissiere, 1973) and the Sterile Insect Technique (SIT). Other interventions aimed at eliminating the parasites have been by chemotherapy, chemoprophylaxis and promotion of trypanotolerant breeds of cattle (Grev, 2002).

a. Hand-catching

Hand-catching is the most ancient method of insect control and was first tried in about 1913 against *G. palpalis* in the Portuguese island Principe, and against *G. palpalis* (*G. fuscipes*) by the Germans on an island, Riamugasire, on Lake Victoria. Hand-catching is absolutely specific and has been found effective against some species, however they

are more expensive than chemicals because it always require a large labour force especially if large areas were to be attacked, thus it fell into disuse (Glasgow, 1970).

b. Bush burning/Clearing

This method was performed based on the knowledge of the biology of tsetse fly, by cutting down dense vegetation, thus destroying both the adult fly and pupae due to decrease in humidity (Nagel, 1995). Bush clearing can be total or ruthless when all vegetation is totally cleared and partial when it involves the destruction of only a portion of the vegetation to render the environment unsuitable for tsetse fly. Partial clearing could be discriminate clearing when woody vegetation in known tsetse concentration sites is destroyed or selective where only certain components of the vegetation forming the fly habitat are removed, which may be removal of only the under storey, leaving the tall trees untouched or removing particular species of shrubs and trees (Jordan, 1986).

The history of the development and application of methods of partial clearing for tsetse control have been extensively reviewed by a number of authorities (Buxton, 1955; Ford *et al.*, 1970). In Nigeria, the record of partial clearing was described by Moiser (1912) in Geidam, Borno State when the population of *Glossina tachinoides* was controlled by vegetation clearing. Significant control was also achieved by Nash, (1940) on *Glossina tachinoides* at Gadau and Anchau in Kano State.

The major short-coming of these method lie in the limited size for which they can be economically deployed relative to the total size of tsetse affected area (Agyemang, 2001) since it requires economically unacceptable destruction of vast area of bush and forest. Bush clearing also results in soil degradation and deforestation and as such was no longer in use around 1970 (Nagel, 1995).

c. Game destruction

The concept of game destruction was developed following the great rinderpest epizootic at the end of the nineteenth century, which resulted in the death of many game animals and thus the disappearance of tsetse flies and trypanosomosis (Jordan, 1986). This method was used many years in Zimbabwe Zambia, Mozambique, Botswana and Uganda to eliminate a wide range of the population of savanna species of *Glossina* and Trypanosomosis due to its close association with game animal. McLennan, (1981) reported the ineffectiveness of game destruction on riverine species because they feed on other hosts besides wild game e.g. man, domestic animals, crocodiles and reptiles. In West Africa, savanna species of tsetse population thrives on very low game densities and as such, this method of control against tsetse did not succeed in Nigeria. This method ceased because, animals migrated to area once cleared from larger mammals, making it possible for tsetse to recover, and also because it became unacceptable to public opinion (Grev, 2002).

d. Trapping method

Trapping method for tsetse fly originated from the Island of Principe, where farm workers wore on their back a dark – colored piece of cloth, covered with glue, to reduce biting nuisance of the flies (Maldonado, 1910) alongside with vegetation clearing and pig elimination to control tsetse on the Island (Da Costa *et al.,* 1916). The first real tsetse trap constructed by Harris was used to eliminate *Glossina pallidipes* from Zululand (Harris, 1938), and other various modified versions were introduced such as the Animal trap (Morris and Morris, 1949); Biconical trap (Challier and Laveissiere, 1973); Monoconical trap (Lancien, 1981); Nitse trap (Omoogun, 1994) etc. In Nigeria, Biconical and Nitse traps have been extensively used for tsetse sampling, ecological studies and control (Omoogun *et al.*, 1994; Dede *et al.*, 2005). Traps were initially

made for control purposes but were later used for sampling and ecological studies due to their poor performance (Omoogun *et al.,* 1994).

The interest in controlling tsetse flies by trapping declined rapidly with the introduction of synthetic insecticides, but was renewed in the seventies, mainly due to increased public awareness of environmental pollution by excessive use of insecticide (Grev, 2002). In West Africa, traps were widely used to control riverine species and was suggested to be better for community – based operations because local people can see flies being caught and killed, and can also see the catch decline as the operation progresses. Trapping method is simple, inexpensive, and harmless to the environment and is currently being used as an integrated part of the arsenal of tsetse control method where fly population is suppressed by trap and other conventional methods such as bait and target. However, reinvasion remains a reoccurring problem (Hargrove, 2002).

e. Use of pheromones

Most work undertaken on pheromones (Langley *et al.,* 1975; Huyton *et al.,* 1980; Offor *et al.,* 1981; Carlson *et al.,* 1984) was based on the identification of the sex recognition pheromone, in the cuticle of female *Glossina* species which induces copulatory behavior in males of the same species, but not of other species, upon contact. This method aims at mass producing the compound(s) that constitute the sex recognition pheromones for the purpose of attracting flies to impregnated traps or screens to thus effect control, but because of the lack of volatility of these pheromones, they could not be exploited as attractant (Jordan, 1985).

f. Bait technology

The potential of bait technology for the control of tsetse fly was appreciated in the first half of the 20th century. The strategy of the technology was to improve bait design (live

or artificial) by the careful analysis of basic responses of tsetse to baits, and using the knowledge to improve the design of devices used in the field (Vale and Torr, 2004). Van den Bossche and De Deken, (2004) reported the use of artificial baits to control fly and reduce trypanosomosis at lower cattle density and stressed that insecticide – treated cattle are more effective than stationary bait in area with higher cattle densities.

The work of various authors (Ndams, 1987; Bossche, 1997; Brightwell *et al.,* 2001; Esterhuizen *et al.,* 2006) has suggested the efficiency of this method for the control of tsetse flies.

Even though improved traps and bait technology (targets) rapidly became the standard control method throughout Africa in late 1980s and 1990s, bait technology had pitfalls in its application which include tackling too small an area and the variability in costs and benefits relating to community – based action (Vale and Torr, 2004).

g. Use of insecticides

The use of insecticides as control methods against tsetse fly commenced in the mid-1940s and is still today a major technique used in large scale (Grev, 2002), and almost all method now used depend on insecticide. Two chemical groups have been widely used in tsetse control: The organochlorides (DDT, dieldrin, endosulfan):- DDT was the first chemical insecticides used against tsetse fly, after which dieldrin became popular because of its high lethal characteristics in more humid conditions. These two chemicals were then displaced by endosulfan for its toxicity and better solubility in spray solvent (Vreysen, 1995). DDT is cheap, has low mammalian toxicity, persists long in the environment and is effective against tsetse fly, while dieldrin on the other hand is expensive, has longer persistent rate than DDT (Davies, 1977). However, both fall victim of international ban due to their environmental side effect (Allsopp and

Hursey, 2004). The spray of endosulfan (being an organochlorine insecticide) against tsetse fly was reported to cause significant fish mortality (Douthwaite *et al.,* 1981).

The synthetic pyrethroids (deltamethrin, alpacypermathrin and betadyfluthrin) (Mangwiro *et al.,* 1999), are the most potent insecticides used against tsetse fly. Deltamethrin have been widely used for both spraying and for Impregnating traps and targets. The major disadvantages of the synthetic pyrethroids are their high cost (Vreysen, 1995).

In Nigeria, the use of insecticides to control tsetse fly started in 1955 (Davies, 1964) at Kamadugu Gana river system which later extended to Kiyawa-Jama'are Katagun system, both area lying within Kano, Borno and Bauchi States. Persistent used of insecticides (DDT) were applied from the ground on the tsetse resting sited by means of pneumatic knapsack pressure sprayers (Davies, 1964; Maclennam, 1967). Aerial spraying started in 1971 in the northern guinea vegetation zone, extended to the southern guineas savanna zone (Spielberger *et al.,* 1977). Spraying and re-spraying activities reclaimed large area of land of about 399,551km² in the Sudan and northern guinea vegetation zones from tsetse fly (Bature, 1985). In Botswana, an area of 16,000km² in the Okavango Delta was reclaimed from *Glossina morsitans centralis* using aerial spraying with deltamethrin applied at 0.26–0.3g/ha and 12,000 deltamethrin treated targets (Kgori *et al.,* 2006).

h. Sterile Insect Technique (SIT)

The concept of sterile insect technique involves the mass production, sterilization – by exposure of males to short burst of gamma radiation from a cobalt-60 source (Okhoya, 2003) and sequential release of sterile males to the target species to compete with the wild male population. Mating between released sterile males and the wild females

produces unviable progeny and the population is reduced over several generations to unsustainable levels (Abila *et al.,* 2003).

The application of the sterile male technique received considerable attention in the 1980s and is one method that seems feasible for the eradication of tsetse flies from the continent of Africa. Economic feasibility of which is grater in the area-wide approach (Feldmann, 2004). This approach was applied on an area-wide basis to eradicate the New World Screw worm *Cochiiomyia hominivirax* in the U.S.A, Mexico and Central America. Since then very effective programmes integrating the SIT have been mounted against tropical fruit flies (Klassen and Curtis, 2005) and some species of tsetse flies on pilot trials at Lake Kariba, Zimbabwe (vs. *Glossina morsitans morsitans*), at Tanga, Tanzania vs. *Glossina morsitans morsitans*), in Burkina Faso (vs. *Glossina palpalis gambiense, Glossina tachinoides and Glossina morsitans submorsitans*) in Plateau state, Nigeria (vs. *Glossina palpalis palpalis)* and in Zanzibar, Tanzania (vs. *Glossina austeni*) (Feldmann, 2004). The success in Zanzibar demonstrated the technical feasibility of fighting the disease through the sterile insect technique approach (Kabayo, 2003).

The obvious constraints of sterile insect technique are the high cons associated with mass rearing, low competitiveness of released sterile males (Whitten and Mahon, 2005), low reproductive rate and low rate of re-infestation (Feldmann, 2004). The technique which can only be employed realistically when density of target population is low is Impractical for use against high density population. It is therefore used only in an integrated approach with other control methods such as traps and targets (Jordan, 1986). However, sterile insect technique still remains an exceptionally promising pest control method in terms of efficacy and environmental compatibility (Nagel and Peverling, 2005).

2.7 Tabanidae

These insects commonly known as ‘horseflies’ or breeze flies, they are large, robust flies with wings and large eyes (Soulsby1982).The pain caused by their bites leads to interrupts feeding, and as consequence, flies may feed on succession of hosts and are therefore important in the mechanical transmission of pathogens such as trypanosomes (Urquhart *et al*., 1996).

There are many genera of tabanids, but only three are of veterinary significance, namely Tabanus, Haematopota and Chrysops. Since the three genera are closely related in behavior and pathogenic significance they will be discussed as a group.

2.7.1 Distribution and host

Distribution is worldwide although certain genera are absent from large areas: example, there are no Haematopota species in Australia or North and South America (Urquhart *et al*., 1996). The tabanid flies have been recovered from localities at sea-level and at altitude of up to 10,000 feet (Nnochiri, 1974).

The hosts are generally large domestic or wild animals and man, but some small mammals and birds may also be attached (Urquhart *et al*., 1996).

2.7.2 Morphology

These are medium to large biting flies, up to 25cm in length, with wing spans of up to 6.5cm.They are generally dark coloured, but may have various stripes or patches of colour in the abdomen or thorax which varies from brown and red to yellow.

They have large coloured compound eyes which occupy a wide area of an equally semi lunar head. The eyes are dichoptic in the female and holoptic in the male. It may be coloured, the wings are broad and are characterized by their marginal cells. The

coloration of the wings is useful in differentiating the three major genera: thus Tabanus has clear or brownish wings, while there are often dark bands across the wings in Chrysops. In contrast, Haematopota has ruffled or speckled wings (Urquhart *et al*., 1996) They have a short, stout, interiorly projecting antennae consisting of three markedly differentiate segments (Georgi and Georgi, 1990).The first segment of antennae is small, the second may be expanded, and the third is marked by annulations that make tabanid antennae appear to consist of many more than three units.

The antennae are also useful in generic differentiation. In species of the genus chrysops the first and second segments of the antennae are long: the third (terminal) segment has four annulations. While in species of the genus Haematopota the first segment of the antennae is large and the second segment narrower, while the terminal segment has three annulations (Soulsby, 1982).

The mouthparts, which are adapted for slashing/sponging, are short and strong and always pointed downwards. The labium is also expanded terminally as paired large labella which carry tubes called pseudotrachea through which the blood or fluid from wounds is aspirated, The biting fascicle, which create the wound, consist of six elements, the upper sharp labrum, hypopharynx with its salivary ducts paired rasp like maxillae on pared boad pointed mandibles. Male flies have no mandibles and therefore cannot feed on blood (Urquhart *et al*., 1996).

2.7.3 Life cycle

After a blood meal the female lays batches of several hundred creamy-white or grayish cigar-shaped eggs, 1.0-2.5mm long, on the underside of vegetation or on stones, generally in muddy or marshy areas. The larvae hatch after four to seven days and drop into the water, or mud, into which they disappear. They are maggot-like and the body

has 11 segments, besides the cephalic portion or which is conspicuous. Each segment has weight fleshy tubercle. The mouth parts are prehensile and masticatory; the larvae are carnivorous. There are three jointed antennae and the large lateral tracheae open on the penultimate segment, which also bears a retractile siphoned tube. The larvae feed on small crustaceans, or even on one another, and grow for two or three months, performing several ecdyses. Finally, they pass through a quiescent stage and then pupate. The pupa is brown and sub cylindrical; the abdominal segments are movable and in the anterior part the appendages of the Imago can be distinguished. These stages last about 10-14 days. The whole life-cycle takes four to five months under favorable conditions, but low temperature prolongs development and the larvae may hibernate (Soulsby, 1982).

2.7.4 Feeding and habitat

Unlike the males which feed only on vegetable sugars (nectar), female tabanid flies attack animals and in addition, feed on plant juices. Feeding occurs during day light hours especially early in the morning and late in the afternoon. A blood meal is usually necessary for the development of the ovaries. With the exception of Chrysops silacea which feeds indoors, most tabanids are outdoor feeders (Nnochiri, 1974). Some feed mainly on the underside of the abdomen around the navel or on the legs; others bite also on the neck and withers (Soulsby, 1982).

The flies feed about every three days. After feeding they rest for two hours on the under-side of leaves or on stones or trees.

2.7.5 Pathogenic significance

The bites of the *tabanidae* are painful and may give rise to weals in soft-skinned animals. Horses and cattle are restless when troubled by these flies and may become

unmanageable, these flies also act as an efficient mechanical vectors of the organism responsible for diseases such as anthrax, pasteurellosis, trypanosomosis, anaplasmosis and the human filarial disease, loasis (Urquhart *et al.,* 1996).

2.7.6 Control

This poses a special problem since breeding places are both diffuse and difficult to detect (Urquhart *et al*., 1996). Where drainage is possible the breeding place may be destroyed by these methods. Since the flies have the habit of skimming over water and occasionally dipping their bodies in to it, the practice of pouring kerosene into water which kills the flies when they dip into it can be utilized. Animals should be kept away from places where the flies abound during hot part of the day.

For general flies control insecticidal spray with a residual effect are used in animal houses and on the animals themselves. There is also the possibility of dark panels with sticky adhesive as drapes and there are a number of electrocution grids which may prove useful in animal's houses (Urquhart *et al*., 1996).

2.8 Stomoxys

Stomoxys calcitrans is the commonest species of this genus and is known as the stable fly or biting housefly. It occurs all over the world and the host includes most animals and man.

2.8.1 Morphology

S. calcitrans resemble the housefly *M. domestica*, being similar in size and grey with four longitudinal dark stripes on the thorax. Its abdomen however is shorter and broader than Musca with three dark spots on the second and third abdominal segments. The proboscis is conspicuous and forward projecting which differentiates it from Musca and other genera of non-biting muscid-flies. Stable fly can be distinguished from biting

muscid flies of the genus *Haematobia* by the bigger size and the shorter pulp of stable flies (Urquhart *et al*., 1996).

Larvae of *Musca* and *Stomoxys* can be differentiated by examination of the posterior spiracles.

2.8.2 Life cycle

Stomoxys sometimes lay the eggs in horse manure, but prefers decaying vegetable matter like straw and hay, especially when contaminated with urine. The female lays batches of 25-50 eggs, resembling those of house flies. Eggs hatch in 1-4 days, or longer in cold weather, and the larva are mature in 6-30 days.

After emergence the adult female require several blood meals before the ovaries mature and egg laying can start.

The complete life cycle from egg to adult fly may take 12-60days depending mainly on temperature (Urquhart *et al.*, 1996).

2.8.3 Feeding and habitat

When feeding, the proboscis swings downwards and skin penetration is achieved by the rasping action of fine teeth on the end of the labium. Approximately three minutes is required for a blood meal and feeding is often interrupted, thus allowing mechanical transmission of micro-organisms.

The flies are most abundant in summer and autumn and live about a month under natural conditions. They prefer a fairly strong light; they are not seen in dark stables or houses. They enter buildings only in autumn or during rainy weather. They are swift flies; but do not travel along distances (Soulsby, 1982).

2.8.4 Pathogenic significance

Both males and females are blood-suckers, attacking man, horse, cattle and other mammals, and even birds and reptiles.

Trypanosoma evansi (Surra of equines and dogs) and *T. equinum* (Mal de caderas of equines, cattle, sheep and goats) are transmitted mechanically by stomoxys. The species may also mechanically transmit *T. gambiense* and *T. rhodesience*, the causative agents of human Trypanosomosis in Africa, and *T. brucei* and *T. vivax,* which cause nagana of cattle, sheep, goats and equines of Africa. It also serves as intermediate host of the nematode Habronema majus, a nematode parasite of the stomach of the horse.

The role of *S. calcitran*s in the transmission of equine infections anemia is still under debate (Steelman, 1976). However, the fly is responsible for the mechanical transmission of septicaemia infections such as anthrax. The importance of biting flies in the transmission of disease and economic loss through 'fly worry' has been reviewed by (Stork, 1979).

2.8.5 Control

The fly is most troublesome in localities where suitable breeding places are readily found. Control measures should therefore be directed toward destroying breeding-places by regular removal of most bedding, hay and faeces from stables and yards, and food waste from feeding troughs, and by preventing the accumulation of heaps of weeds, grass cuttings and vegetable refuse.

Regular application of pyrethrins, synergized pyrethins, pyrethroids, coumaphos, stirofos, or dichlorvos is indicated.

Application of insecticides to areas where they habitually rest (Urquhart *et al.*, 1996).

2.9.0 *Sarcophaga*

Sarcophaga haemorrhoidalis, also known as the red-tailed flesh fly, is a fly in the Sarcophagidae family. This fly often breeds in carrion and feces, making it a possible vector for disease. The larvae of this species can cause myiasis, as well as accidental myiasis. It is potentially useful in forensic entomology.

2.9.1 Distribution of *Sarcophaga*

Sarcophaga haemorrhoidalis is a common species of flesh flies that appear worldwide in distribution and is commonly found in the United States. It can be found throughout the year in the southern portion of the United States. The larvae are adaptable and can live in moist semi-aquatic habits that are unsuitable for most other fly species. Overall, *S. haemorrhoidalis* is most likely to be found in climates with higher temperatures and will prefer high temperatures throughout its entire life cycle.

2.9.2 Morphology

Sarcophagidae is the dipteran family commonly known as flesh flies, comprising approximately 2000 species. Many species of Sarcophagidae prefer to breed in carrion over other mediums, but there are several species that breed in dung. A large number of species are parasitoids or cleptoparasitoids and never breed in carrion. It is difficult to identify the *S. haemorrhoidalis* species unless genitalia can be observed. Only males can be identified and classified within the genus. Sarcophagids are rather large in size ranging from 4 to 23 mm, (adults of *S. haemorrhoidalis* vary in size from 7 to 14 mm). Distinguishing characteristics include a checkerboard like pattern on the abdomen, stripes on the thorax and red eyes. Flesh flies are attracted to anything rotting, including feces. Sarcophagidae are unimpeded by rain and fly in any weather. Because of this trait, Sarcophagidae will often be the first flies to colonize a corpse after an extended

period of rain. Flesh flies appear to prefer sunlight over shaded conditions. *Sarcophaga haemorrhoidalis* (*Bercaea cruentata*) is the one of the most common species of Sarcophagidae recovered from indoor crime scenes in the United States.

2.9.2 Distribution

All members of the family Sarcophagidae are larviparous or ovoviviparous. *Sarcophaga haemorrhoidalis* (*Bercaea cruentata*) gives live birth to larvae with the female retaining the egg case in her abdomen. Flesh flies are strongly attracted to carrion or dry flesh. The female has a strong desire to lay larvae on the flesh and have even been noted to larviposit on the sleeve of a garment that has been previously soiled with blood Oldroyd states that the larvae of *Sarcophaga spp* are voracious and will take anything of animal origin be it alive or dead. A larva is forced out of the larvipositor usually head first and soon disappears into the food material. Once larvae are deposited as 1st stage instars, rapid development follows with 3rd instars usually being achieved by three to four days. Larviposition to adulthood generally takes around two weeks.

If the fly is forced to hibernate due to temperate climates, it will do so in the pupal stage.

2.9.3 Importance of *Sarcophaga*

2.9.3.1 Medical importance

Due to its attraction to feces and carrion, *S. haemorrhoidalis* has been accounted for as a dipteran species that may serve as a mechanical vector for disease, especially if it intrudes into homes. The family Sarcophagidae is particularly attracted to human food and filth. Bacteria can be transferred physically from the fly's body, legs, or proboscis, to an animal, human food, or open sores. *S. haemorrhoidalis* has also been found to carry poliovirus. During a 1914 polio epidemic, samples of the virus were collected

from *S. haemorrhoidalis*, among other dipterans. The sample was used to infect a monkey with polio, showing that it was an active virus. However, there is still no conclusive evidence as to whether or not this species actually transmits diseases to humans or animals.

The larvae of *S. haemorrhoidalis* may produce myiasis on necrotic or dead flesh. The first case of auricular myiasis (on the outer ear) on a human was reported in Iran in 1974. Other myiasis cases have been recorded around the world in both humans and animals. Examples range from aural myiasis caused by *S. haemorrhoidalis* in four children in Israel (from 1990 to 1993) that produced symptoms of ear discharge, otalgia and itching, to the infection of a schnauzer in Umbria, Italy in 1994 by *S. haemorrhoidalis* maggots.

Accidental myiasis can also be caused by *S. haemorrhoidalis* larvae. When meat contaminated with live larvae is eaten, the maggots can make their way into the gastrointestinal tract and infest the intestines. The larvae are usually excreted with the feces.

2.9.3.2 Forensic importance

S. haemorrhoidalis is hardly ever used in forensic investigations, due to its global distribution and the fact that little is known about them. Usually, other more researched flies and beetles, if present on the body, take precedence. The fly has a pupation time ranging from 93 hours to 153 hours. Development from larvae to adult can range from 252 to 802 hours. Knowing the pupation and life cycle times of *S. haemorrhoidalis* and taking into consideration that this species is ovoviviparous allows investigators to calculate how long the fly has been on the corpse. If time of colonization of the corpse by maggots is known, it can help determine the PMI, or interval. The larvae of *S.*

haemorrhoidalis occur on carcasses in the early and advanced stages of decomposition. The maggots can live in amphibious habitats in which many other fly species may not be able to thrive or breed, making it possible for them to be the first dipterans on a corpse in wet weather.

CHAPTER THREE

3.0 MATERIALS AND METHODS

3.1 Study Area

The research was conducted in two (2) Local Government Areas – Ikara and Kubau Local Government Areas of Kaduna state. (Fig 1) Four (4) Districts namely Paki, Pala, Furana, Kurmin Kogi for Ikara LGA and Mai tyiga, Angwan Palami, Ruwan Sanuyi and Doka for Kubau LGA were randomly selected as the study sites.

Ikara Local Government area is located some 30 kilometers north-east of the city of Zaria. It has an area of 853km^2 with a population of 194,723 (NPC 2006), its headquarters is in the town of Ikara (geographically located at (11.38 degrees). Kubau Local Government has an area of 2,505km2 and a population of 282,045 during the (NPC 2006). The headquarter is located at Anchau on Longitude 0.80 61'E, also of Kaduna State, Nigeria. The vegetation of these two areas is typical of the Northern Guinea woodland savannah. However, because of the effects of annual bush farming, settlement patterns are mainly hamlets and farm compounds. There are more than ten different ethnic groups in these areas, among them are Kurama, Hausa, Amarwa, Warsa and Fulani and the general spoken language is Hausa. The main occupations of the people are farming, fishing and trading. Mixed farming of crop cultivation and animal production is the usual practice.

These two Local Governments areas are known for their maize production and supply to several parts of Nigeria. (Onah., 1985)During the rainy season, many of the cattle rearers go on transhumance to Plateau and Bauchi States with their animals and return at the end of the rainy season to feed on farm residuals. Infrastructure development is poor with some areas being inaccessible during the rains.

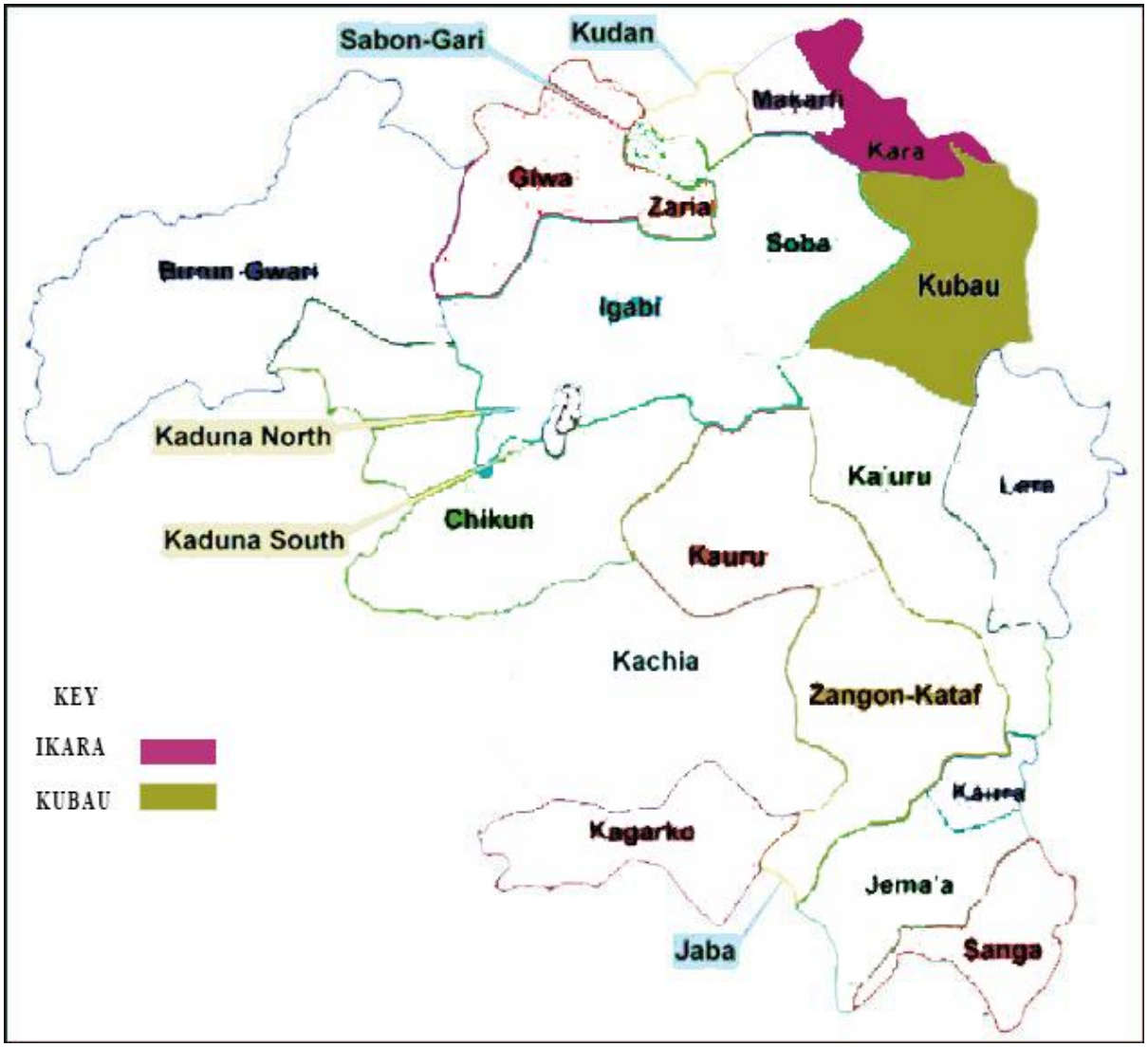

Fig 3.1: Map of Kaduna State Showing the 23 LGAs

Source: www.kadunastate.gov.ng

3.2 Study Design

3.2.1 Survey for hematophagous flies in Ikara and Kubau LGAs of Kaduna State

The study was conducted between the months of March (dry season) and August (rainy season) 2015.

A total of twelve traps in each LGAs, 6 Biconical and 6 Nzi traps were deployed for the purpose of the study. Traps were positioned at about 100 -150 meters apart for 48 hours in the following districts surveyed at Ikara LGA; Paki district - 2 biconical traps; Pala district- 2 biconical traps; Furana district- 2 biconical traps and 2 Nzi traps; Kurmin Kogi district - 4 Nzi traps. In Kubau LGA, Doka district 4 biconical traps: Mai Tgiya district 2 biconical traps: Angwan Palamin district 2 Nzi traps: Ruwan Sanuyi district 4 Nzi traps: were installed.

Flies caught were harvested after 48hrs and placed in a bottle or polythene bags andthe blood samples collected from nomadic white Fulani cattle's in each LGA within March and August 2015 were placed in a cool box and brought to Entomology and protozoology laboratory respectively, Department of Veterinary Parasitology and Entomology, A.B.U, Zaria for Zaria for flies' identification using the guide described by Keith, (1990).

3.2.2 Survey for *Trypanosoma* species in Ikara and Kubau Local Government Areas of Kaduna State

3.2.2.1 Sample size

Sample size was determined using the formula described by Thrustfield (1997).

N=ZP2q/d2

Where

N= Sample size

Z= Standard normal deviation for 95% confidence interval (1.96)

P= Expected prevalence (15.8% by Ahmed, 2010)

D= desired absolute precision (0.05)

Q= 1-p

n= 1.962 x 0.157 x (1-0.157) 0.052 = 200

200 blood samples from 200 cattle were to be collected for this research, 100 from each LGA.

3.2.2.2 Blood sampling

A random sampling was used to obtain a total of 200 blood samples from cattle of different ages in the study areas.

Animals below one year were considered as young calves, and those from one year and above were regarded as adults. Two millilitres of blood was taken from each of the selected animal from the jugular vein into specimen bottles containing ethylene diamine tetra acetic acid (EDTA) dispensed at one milligram powder per milliliter (ml) of blood and conveyed in cold box with ice packs to the protozoology laboratory of the Department of Veterinary Parasitology and Entomology A B U Zaria for analysis.

3.3 Parasitological Analysis of the Blood Samples

The collected blood samples were analyzed for the presence of *Trypanosoma* species using the thick blood smear, thin blood smears as well as HCT as described by Soulsby (1986);

3.3.1 Thick blood smears

A large drop of blood of about 10µl was placed on the centre of a clean microscope slide and swirled with a toothpick or the corner of another slide so that an area of approximately 1.0–1.25 cm in diameter was covered. This was air-dried for 1 hour or longer, while protecting it from flies. The unfixed smear was then stained with Giemsa (one drop of commercial Giemsa + 1 ml of phosphate buffered saline (pH 7.2), for 25 minutes. After rinsing the slides with water, the slides were examined under a light microscope at high magnification (x100) using oil immersion.

3.3.2 Thin blood smears

A drop of blood (3-5 µl) was placed 20 mm from one end of a clean microscope slide and a thin film was drawn out. The film was air-dried briefly, fixed in methyl alcohol for 2 minutes and allowed to dry. The smears were then stained with Giemsa (one drop Giemsa + 1 ml PBS, pH 7.2) for 25 minutes. This preparation was then poured off and the slide washed under running water, dried, and examined under x1000 objective lens using immersion oil.

3.3.3 Haemotocrit centrifugation

Blood was collected (70 µl) into heparinised capillary tubes (75 x 1.5 mm), which were sealed at the dry end and centrifuged, with sealed end down, at 3000 g for 10 minutes. After centrifugation, two pieces of glass (25 x 10 x 1.2 mm) were glued to a slide and the spun capillary tube was placed between them. A drop of oil immersion was placed

on top at the level of the Buffy coat junction where the trypanosomes were concentrated and the Buffy coat area were examined under the microscope at x100.

3.4 Data Analysis

Statistical Package for Social Sciences (SPSS) Version 20.0, IBM USA 2011 was used for data analysis.

Prevalence was calculated and expressed as percentages with respect to LGAs.

Chi-square test was used to test for association between prevalence of flies and difference in prevalence with respect to seasons

Fisher's Exact Test was used to calculate for association between the number of flies in each LGAs and per season.

The level of significance was determined at $p<0.05$ at 95% confidence interval (C.I).

CHAPTER FOUR

4.0 RESULTS

4.1 Survey for Hematophagous Flies in Ikara and Kubau LGAs

A total of 232 hematophagous flies were caught, during the studies in Kubau and Ikara LGAs, with the highest number 127 (54.7%) being caught in Ikara Local Government while 105(45.3) were caught in Kubau Local Government.

Of the total number of flies caught, 212(91.4%) and 20(8.6) were caught during rainy and dry seasons respectively (Table 4.1) .The overall highest number of flies caught in both seasons from Ikara LGA with seasonal distributions were 54.7% and 55% for the rainy and dry seasons respectively (Table 4.1).

The occurrence of flies in Kubau LGA is shown in Table 4.2, Ruwan Sanuyi district had the highest prevalence of flies 35(97.2) caught followed closely by Mai Tgiya district 25(92.6) during the rainy season. However during the dry season, Doka district had the highest prevalence of 3(20.0%) followed by Angwan Palami district with 3(11.1).There was no significant difference ($P>0.05$) between occurrence of the flies and seasons in Kubau LGA.

Occurrence of flies in four district of Ikara LGA. Paki district had the highest prevalence of 51(91.1%) while Furana district had the least prevalence of 7(63.6%) during the wet season similarly in the dry season, Paki district had the highest prevalence of 5(8.9%) while Pala had the least. There was statistically significant different ($P<0.05$) between the occurrences of the flies in the districts (Table 4.3).

Table 4.1: Overall Occurrence of flies in Ikara and Kubau Local Government Areas of Kaduna State, Nigeria

LGA	Total No of flies caught (%)	Seasonal occurrence	
		Wet season (%)	Dry season (%)
Ikara	127(54.7)	116(54.7)	11(55)
Kubau	105(45.3)	96(45.3)	9(45)
TOTAL	232	212(91.4)	20(8.6)

Table 4.2: Occurrence of flies in four districts of Kubau Local Government Area of Kaduna State, Nigeria

District	Total No of flies caught (%)	Seasonal occurrence	
		Wet season (%)	Dry season (%)
Mai Tgiya	27	25(92.6)	2(7.4)
Angwan Palami	27	24(88.9)	3(11.1)
Ruwan Sanuyi	36	35(97.2)	1(2.8)
Doka	15	12(80.0)	3(20.0)
TOTAL	105	96(91.4)	9(8.6)

Fisher's Exact Test=4.267, df =3, P-value=0.203

Table 4.3: Occurrence of flies in four districts of Ikara Local Government Area of Kaduna State, Nigeria

District	Total No of flies caught (%)	Seasonal Occurrence	
		Wet season (%)	Dry season (%)
Paki	56	51(91.1)	5(8.9)
Pala	42	42(100)	0(0.0)
Furana	11	7(63.6)	4(36.4)
Kurmin Kogi	18	16(88.9)	2(11.1)
TOTAL	127	116(91.3)	11(8.7)

Fisher's Exact Test= 12.596,df=3,P-value=0.002

The species specific occurrence of flies in Kubau LGA is shown in Table 4.4, with *Stomoxys calcitrans* having the highest prevalence of 56(95.9) during the rainy season followed by *Musca domestica* with a prevalence of 21(95.2) and Sarcophaga 15(6.7%). However, during dry season, Sarcophaga was more abundant with occurrence rate 6.7%, *Stomoxys calcitrans* (22.2%) and *Musca domestica* (22.2%).

During the rainy season in Ikara LGA, *Musca domestica* had the highest prevalence of 48(92.0) followed by *Stomoxys calcitrans* with 43(87.8) which however were not significantly different (P>0.05) between the species of the flies caught and seasons of the year (Table 4.5).

Similarly the result of determining the efficiency of flies catch by the Biconical and Nzi trap in both LGA during wet and dry season shows that Biconical had the highest trapping efficiency of 118(55.7), 12(60) with Nzi trap having 94(44.3), 8(40) respectively and this difference was statistically significant (P<0.002) between the number of flies caught or trapping efficiency between the traps (Table 4.6).

The specific occurrence of *Trypanosma* infection of cattle in Kubau and Ikara LGA with *T. brucei* having a prevalence of 1(100) and 0(0.0) respectively while *T. vivax* for Kubau and Ikara had 3(60) and 2(40) respectively However, all the two positive samples from Ikara were all *T. vivax* infections (Table 4.7).

Table 4.4: Species-specific occurrence off lies in Kubau Local Government Area of Kaduna State, Nigeria

Species of flies	No of flies caught (%)	Seasonal Occurrence	
		Wet season (%)	Dry season (%)
Stomoxys calcitrans	58(46.7)	56(65.1)	2(22.2)
Musca domestia	32(20.0)	21(24.4)	2(22.2)
Sarcophaga	15(6.7)	9(10.5)	6(6.67)
TOTAL	105	86	9

Table 4.5: Species -specific occurrence of flies in Ikara Local Government Area of Kaduna State

Specie of flies	Total No of flies caught (%)	Seasonal Occurrence	
		Wet season (%)	Dry season (%)
Stomoxys calcitrans	49(38.6)	43(37.1)	6(54.5)
*Tabanus*species	9(7.1)	9(7.76)	0(0.0)
Musca domestica	52(39.4)	48(41.4)	4(36.4)
Sarcophaga	9(3.1)	8(6.9)	1(9.1)
TOTAL	127	116	11

Fisher's Exact Test=1.229, df =1, P=0.576

Table 4.6: **Trapping efficiency of Biconical and Nzi traps on flies in Kubau and Ikara Local Government of Kaduna State during wet and dry season**

LGA	Wet season			Dry season		
	Bioconical	Nzi	Total	Bioconical	Nzi	Total
Kubau	60(62.5)	36(37.5)	96	5(55.6)	4(44.4)	9
Ikara	58(50.0)	58(50.0)	116	7(63.6)	4(36.4)	11
Total	118 (55.7)	94(44.3)	212	12(60)	8(40)	20

Table 4.7: **Occurrence of Trypanosoma infection of cattle and Number positive for *T. vivax* and *T. brucei* in Kubau and Ikara Local Government Areas of Kaduna State**

LGA	No. of Cattle Sampled	No. of positive Samples (%)	No of positive for (%)	
			T. vivax	*T. brucei*
Kubau	100	4(20)	3(60)	1(100)
Ikara	100	2(33.3)	2(40)	0(0.0)
Total	200	6(3.0)	5(2.5)	1(0.5)

Plate 1: Installation of the Biconical trap in the study areas

Plate II: Installation of the Nzi trap in the study areas

Plate III: Stomoxys calcitrans caught at one of the sampled sites

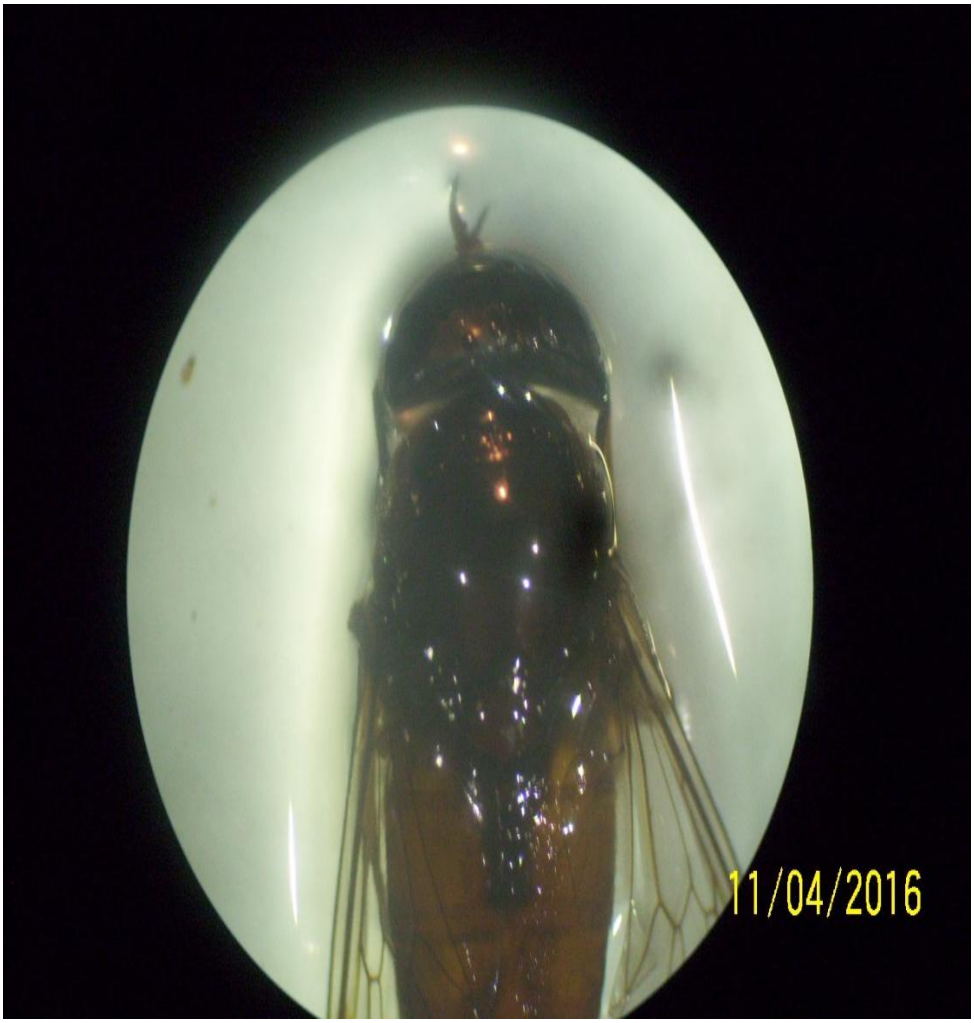

Plate IV: *Tabanus sp* caught at one of the sampled sites

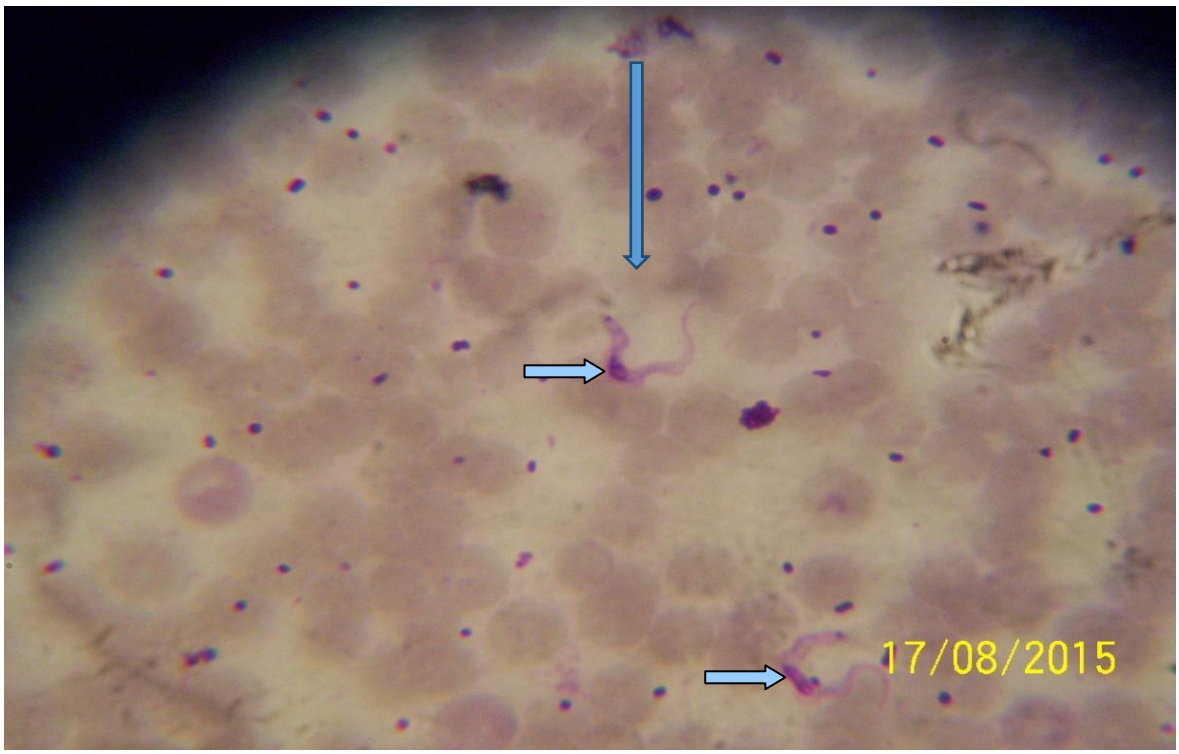

Plate V: *Trypanosoma brucei* in sampled blood smear (arrowed)

CHAPTER FIVE

5.0 DISCUSSION

The present results showed the population of flies with particular reference on *Glossina* species and other flies of Veterinary importance in Ikara and Kubau LGAs. The findings has shown that *Stomoxys calcitrans* and *Tabanus* species are the dominant hematophagous flies of Veterinary importance in the study areas, as also reported by Jegede *et al*., (2015) of the abundance of these flies in Abuja municipal Area council, Nigeria. This perhaps indicates that *Stomoxys* and *Tabanus* are well adapted to the savannah vegetation zones of Nigeria.

The abundance of *Tabanus sp* was far less than that of *Stomoxys* species in the study area, notwithstanding, *Tabanus* sp is of considerable significance, which serving as the mechanical transmitter of various diseases most especially trypanosomosis and causing economic losses in meat and milk industries due to its painful bite (Chvala *et al*.,1972; Foil,1989).

The apparent absence of Glossina species (tsteflies) in the study areas but the detection of *T. brucei* indicate the ability of hematophagous flies (biting flies) notably trypanosome species and as potential vectors of this species of trypanosome.

The vegetation in the study areas was dry and the woodland open in the dry season, except some few residual forests and thickets along streams; this makes the habitat unsuitable for *tsetse flies and* may probably result in the migration of the fly to more suitable habitats. Even though the riverine and woodland vegetation are thick in the wet season, it is possible that dispersal could have been responsible for the absence of the fly due to high humidity. This observation is supported by the findings of Nash, (1937)

who reported that *G. m. submorsitans* exhibits low density in the dry season due to high temperature which has drastic effect on the population.

Other important non hematophagous dipteran flies recorded in the present study were *Musca domestica* and *Sarcophaga* species. The presence and abundance of *Musca domestica* in the study area is of great concern as they are efficient mechanical transmitters of important human and animal diseases such as corynebacterium pseudo tuberculosis (Addo, 1983). In the present study, the Biconical trap caught more flies than the Nzi trap throughout the study. Although the differences in the number of flies caught between the two traps was not statistically significant, the result may still indicate that Biconical trap is a better trap for catching dipteran flies as also reported by Jegede *et al*, (2015) who also made similar observation in their study on fly types and use of these traps in Abuja FCT, Nigeria.

The occurrence of *Trypanosoma* infection of cattle in the study area was very low and this agrees with the reports of Abenga *et al.,* (2009) in northern parts of Kaduna State. This could be attributed to the very low population or absence of *Glossina* species, the biological vectors of African Animal trypanosomoses (AAT) in the study areas.

CHAPTER SIX

6.0 SUMMARY, CONCLUSION AND RECOMMENDATIONS

6.1 Summary

The survey for hematophagous flies and *trypanosoma* species was conducted in Ikara and Kubau LGAs of Kaduna State. The study was conducted between the months of March (a dry season period) and August (a rainy season period) 2015. Twelve traps (Biconical and Nzi) were deployed and positioned at about 100-150 meters apart for 48hours in Ikara and Kubau LGA and the trappings harvested. Blood samples collected from pastoralists raised white Fulani cattle in each LGAs were analysed in the Entomology and Protozoology laboratories respectively in the Department of Veterinary Parasitology and Entomology, ABU Zaria.The study showed that *Stomoxys, calcitrans* and *Tabanus* species were the major hematophagous flies of Veterinary importance in the study areas and were more abundant in the rainy season than in the dry season. The occurrence of *Trypanosoma vivax* and *brucei* species infection among cattle in the study areas was very low probably due to the absence of the major biological vector of the parasites in the study area - *Glossina* species (tsetse flies).

6.2 Conclusions

i. The present study showed that *Stomoxys calcitrans* and *Tabanus* species were the major hematophagous flies of Veterinary importance in the study areas and were more abundant in the rainy season than in the dry season.

ii. *Glossina* species were not captured in the study areas despite being the main targeted fly species.

iii. There was no difference in the flies trapping efficiency of biconical and Nzi traps.

iv. *T. vivax* and *T. brucei* were isolated from blood samples with *T. vivax* being more prevalent.

v. The occurrence of *Trypanosome* species infection among cattle in the study areas was very low probably due to the absence of the major biological vector of the parasite in the study area.

6.3 Recommendations

Based on the survey of hematophagous flies and *trypanosoma* species in Ikara and Kubau LGAs, the following recommendations were made:

i. Further investigation of the LGAs is suggested in order to understand the actual status of the hematophagous lies in the LGAs, since the study area is in the guinea savanna belt where tsetse flies should ordinarily be found.

ii. The presence of hematophagous flies and trypanosome species in the study area may pose a health risk to animals in the LGAs. Therefore, control or eradication of hematophagous flies is suggested.

REFERENCES

Abenga, J. N. and Lawal, I. A. (2005). Implicating role of animal reservoir hosts in the resurgence of Gambian trypanosomosis (88sleeping sickness). *African Journal of Biotechnology***,** 4(2):134 – 137.

Abila, P. P., Kiendrebeogo, M.; Mutika, G. N., Parker, A. G. and Robinson, A. S. (2003). The effect of age on the mating competitiveness of male *Glossina fuscipes fuscipes* and *G. palpalis palpalis*. *Journal of Insect Science*, 3(13): 1 -8.

Agyemang, K., (2001). The role of trypanotolerant livestock's in the content of the methodological approach for tsetse – trypanosomosis control/eradication strategies with particular reference to West Africa. Workshop on strategic planning of area-wide tsetse and trypanosomosis control in West Africa Ouagadougou, Burkina faso. pp.61.

Ahmed, A. B., Omoogun, G. A. and Shaida, S. S. (2000). Trypanosomes infection in *Glossina* species at the Kainji Wild Life Park, Nigeria. *Entomological Society of Nigeria Occasional Publication*, 32: 57 – 62.

Ahmed, A.B. (2004). A peridomestic population of the tsetse fly *Glossina palpalis palpalis* ROBINEAU-DESVOIDY, 1830 (DIPTERA: GLOSSINIDAE) at Kontagora Town, Niger State, Nigeria. *Entomologia Vectorale*, 11(4):599 – 610.

Allsopp, R. and Hursey, B. H. (2004). Insecticidal control of tsetse. In The Trypanosomiasis (eds. I. Maudlin, P. H. Holmes and M. A. Miles) CADL Publishing, 491 -507.

Anavhe, A. O. (1988). Tsetse ecological studies at Ekosodin locality, Benin, Bendel state. *Annual Scientific Report***.** Nigerian Institute for Trypanosomiasis Resaearch, Kaduna, Nigeria. 12 – 20.

Anon., (2002). Report of workshop on harmonization of the Activities of PAAT and PATTEC Held at the FAO Headquarters, Rome, Italy, in *Tsetse and Trypanosomiasis Information Quarterly*, 25(2**):** 29.

Anon. (2004). ADF Seeks to Eradicate Tsetse. *Press Release* (No. SEGL3/F/ / 043).

Askoy, S, Gibson, W.C and Lehane, M.J. (2003). Interaction between Tsetse and Trypanosomes with implications for the control of Trypanosomiasis. *Advances in Parasitology*, 53:1. 83.

Atkins, J. (1978). Sleeping sickness. Tropical Medicine and Parasitology, Classic Investigations. Cornell University Press. Ithaca, Newyork**,** 181-182.

Attardo, G. M.; Guz, N.; Strickler-Dinglasan, P. and Askoy, S. (2006). Molecular aspects of viviparous reproductive biology of the tsetse fly *(Glossinamorsitans morsitans*): Regulation of yolk and milk gland protein synthesis. *Journal of Insect Physiology***,** 52:1128 -1136.

Baldry, D.A.T. (1969). Variation in the ecology of *Glossina* spp with special reference to Nigerian populations of *Glossina tachinoides*. *Bulletin of World Health Organisation,* 40: 859-869.

Baldry, D.A.T. (1980). Local distribution and ecology of *Gossina palpalis* and *Glossina tachinoides* in forest foci of West African human trypanosomiasis, with special reference to associations between peri – domestic tsetse and their hosts. *Insect Science and its Applications***,** 1:85 – 93.

Baldry, D.A.T & Vloet van der, A.M.V., (1982). A modified wing-fray analysis technique Forage estimation of *Glossina*. *Procceedings of Symposiumon Radiation In: Insect Control,* 225/56: 185-190.

Bature, F., (1985). Management problems in tsetse (*Glossina*) control: The Nigerian Experience. Project written up for the advance management course of the Administrative Staff College of Nigeria. Topo, Badagry.

Bossche, P. vanden. (1997). The control of *Glossina morsitans morsitans* (Diptera: Glossinidae) in a settled area in Petauke District (Eastern Province, Zambia) using odour – baited targets. *Onderstepoort Journal of Veterinary Research,* 64 (4): 251 -257.

Boulanger, N; Brun, R; Ehret-sabatier, L; Kunz, C and Bulet, P.(2002) Immunopeptides in the Defense reactions of Glossina morsitans to bacterial and *trypanosoma brucei brucei* infections. *Insect Biochemistry and Molecular Biology***,** 32(4): 369-375.

Brightwell, B. Dransfield, B.; Maudlin, I. Stevenson, P. and Shaw, A. (2001). Reality vs. rhetoric - a survey and evaluation of tsetse control in East Africa. *Agricultural and Human Values,* 18: 219 – 233.

Carlson, D. A. Nelson, D. R. Langley, P. A. Coates, T. W. Davies, T.L. and Leegwater-vander Linden, M. E., (1984). Contact sex pheromones in tsetse fly *Glossina pallidipes* (Austen): identification and synthesis, *Journal of Chemical Ecology***,** 10: 429 – 450.

Challier, A. (1965). A melioration de la méthode de détermination déliage physiologique de glossines. *Bulletin de la Société Pathologie Exotique,* 58: 250.

Challier, A. and Larveissiere, C. (1973). UN nouveau piege pour la capture des Glos (Glossina: Diptera, Muscidae) description *et* essays sur le terrain. Cah. ORSTOM**.** *Series Entomologie Medicale et Parasitologie,* 11:251-262.

Chretien, J. P. L and Smoak, B. L. (2005). African trypanosomiasis. Changing epidemiology and consequences. *Current Infectious Disease Reports,* 7(1)**:** 54 - 60.

Coccossis, H. and Parpairis, A. (1995). "Assessing the Interaction between Heritage, Environment and Tourism: Mykonos", In: Coccossis, H. and Nijkamp, P. (eds) *Sustainable Tourism Development*, Avebury, Hong Kong, pp 127-140.

Conway-Klaassen, J. M; Wyrick-Glatzel, J. M. Neyrinck, N. Belair, P. A. (2002). Sleeping sickness in a young American tourist *Laboratory Medical*, 33: 783-788.

Dagnogo, M. Traore, G. and Souleymane, F. (2004). Determination of sleeping sickness transmission risk areas from trypanosome infection rates of tsetse flies in Daloa, Cote d'Ivoire. *International Journal of Tropical Insect Science,* 24(2):170 –176.

Dankwa, D., Oddoye, E.O.K. and Mzamo, K. B.2000. Efficacy of three trap types for trapping *Glossina palpalis, G. tachinoides and G. morsitans* in the Mole Game Reserve in Ghana. *Ghana Journal of Agricultural Science, 33 (2)*: 177-181.

Dede, P.M., Halid, I., Omoogun, G.A., Uzoigwe, N.R., Njoku, C.I., Daniel, A. D. and Dadah, A.J. (2005). Current tsetse and trypanosomiasis situation on Jos

Dutton, J.E. (1902). 'Preliminary note upon a trypanosome occurring in the blood of man', Thomson Yates Lab. Repts., 4, 455. In.*'The Africa Trypanosomiasis'* eds. Mulligan, H. W. Potts, W. H. Allen & Unwin, London.

FAO, (2002). Fighting tsetse – a scourge to African farmers. www.fao.org/english/newsroom/nes/2002/4620-en-html.

Federal Ministry Agriculture,. (1981). A livestock production plan for Nigeria: A study conducted for the Federal Ministry of Agriculture and the Green Revolution Committee.

Feldmann, U., (2004). The sterile insect technique as a component of area-wide integrated pest management of tsetse. In: *The Trypanosomiasis* (eds. I. Maudlin, P. H. Holmes and M.A. Miles) CABI publishing, 565 – 582pp.

Ford, J., (1970). The geographical distribution of *Glossina.* In: *The African Trypanosomosis*. Eds. Mulligan, H.W and Potts, W.H. 274 – 275. London, Allen and Unwin**.**

Ford, J. (1971). The role of the trypanosomosis in African Ecology; a study of the tsetse problem. Clarendon Press, Oxford 568.

Ford, J., Nash, T.A.M. and Welch, J.R. (1970). Control by clearing of vegetation, chap. 27, 534 – 56 in Mulligan, H.W. (ed.), and *The African Trypanosomiases*. George Allen and Unwin, London.

Garcia, A. Courtin, D. Solano, P. Koffi, M. and Jamonneau, V. (2006). Human African trypanosomiasis: connecting parasite and host genetics. *Trends in Parasitology,* 22(9): 405 -409.

Glasglow, J. P., (1963). The distribution and abundance of tsetse. International Series of Monographs in Pure and Applied Biology, 20, Pergamon Press, Oxford, 241.

Glasglow, J. P., (1970). The *Glossina* community. In: '*The Africa Trypanosomiasis'* eds. Mulligan, H. W., Potts, W. H. Allen & Unwin, London, 348 -381.

Glasson, J., Godfrey, K., and Goodey, B. (1995). Towards Visitor Impact Management: Visitor Impacts, Carrying Capacity and Management Responses in Europe's Historic Towns and Cities. Avebury, England**.**

Grev, H. (2002). The tsetse fly – a disease spreader. SLU**,** *Tropical Ecology and Environmental Management*, B10380, 01-15.

Hadis, M; Endeshaw, T.; Kebede, A.; Asfaw, T. and Tilahun, T., (1995). Decline of *Glossina morstians ugadensis* in Gambella, Ethiopia. *East African Medical Journal*, 72 (6): 363 – 369.

Hargrove, J. W. (1975). Some changes in the flight apparatus of tsetse flies, *Glossina morsitans* and *G. pallidipes*, during maturation. *Journal of Insect Physiology*, 21:1485 – 1489.

Hendrickx, G. Napala, A. (1997). Le contróle dela trypanosome a lecarte: une approche integree basee sur un systeme d'information Geographique. Academis Royale ds Sciences d' outhre Mer. Memoires classe scinces Naturellles and medicales. In 8th Nouvelle serie, Tome 24 – 2 Bruxelles, 100.

Hu, C and Askoy, S. (2006). Innate immune responses regulate trypanosome parasite infection of the tsetse fly *Glossina morsitans morsitans*. *Molecular Microbiology,* 60(5): 1194.

Huyton, P. M. Langley, P. A. Carlson, D.A. and Schwarz, M., (1980). Specificity of contact sex pheromones in tsetse fly *Glossina spp*, *Physiology and Entomology.* 5**:** 253 -264.

Itard, J. 1971. Chromosomes de *Glossina Fusca congolenses* Newstead et Evns, (1921). (Diptera – Muscidae) *c.r.hebd. Séance Acad Sci. Paris 272,* 2561- 2564.

Jackson,C.H.N., (1948). The analysis of a tsetse population III**,** *Annals of Eugen. Camb.* 14: 19-108.

Jaenson, T. G. (1981). Ecology and behaviour of *Glossina pallidipes* Austen (Diptera*: Glossinidae*) in Southern Kenya. *Bulletin Entomological Research,* 71:703 -715.

Jamonneau, V. Ravels, S. Koffi, M. Kaba, D. Zeze, D. G. Ndri, I. Sane, B. Coulibaly, B.Cunn, G. & Solano, P. (2004). Mixed infections of trypanosomes in tsetse and pigs and their epidemiological significance in a sleeping sickness focus of Cote d'Ivoire. *Parasitology,* 129(6): 693 702.

Jelinek, J. Bisoffi, Z. Bonazzi, L. VanTheil, P. Bronne, U. De frey, A. Gundersen, S. G. McWhinney, P. Ripamonti, D. (2002). Cluster of African trypanosomiasis in Travellers to Tanzania National Park. *Emerging Infectious Disease,* 8: 634-635.

Jordan, A.M. (1961). An assessment of the economic importance of the tsetse species of Southern Nigeria and Southern Cameroon based on their trypanosome infection rates and ecology. *Bulletin Entomological Research,* 52:431-441.

Jordan, A. M. (1974). Recent development in the ecology and methods of control of tsetse flies *Glossina* spp (*Diptera: Glossinidae*). *Bulletin Entomological Research,* 63: 361-399.

Jordan, A. M. (1986). Trypanosomiasis Control and African Rural Development. Longman, London and New York, 357.

Jordan, A. M. (1993). Tsetse flies (*Glossinidae*). In: Medical Insects and arachnids (eds. R. P. Lane and R. W. Crowsskey). Chapman and Hall, London, 333 – 388.

Kabayo, J. P. (2002). Aiming to eliminate tsetse from Africa. *Trends in Parasitology,* 18(11**):** 473 – 475.

Kabayo, J. P. (2003). Eradicating tsetse flies from Africa. In: *Africa Recovery***,** Vol. 17.

Kalu, A. U. and Olatunde, O. A. (1989). Tsetse and Trypanosomiasis on the Jos Plateau, Nigeria. *Annual Scientific Report*, Nigerian Institute for Trypanosomiasis Research, Kaduna, Nigeria**.** pp 33 – 35.

Kalu, A. U. (1991). An outbreak of trypanosomiasis on the Jos Plateau, Nigeria. *Tropical Animal Health Production,* 23: 215 -216.

Kennedy, P.G. (2006) Human African Trypanosomiasis – neurological aspects. *Journal of Neurology*, 253(4): 411 -416.

Kgori, P. M. Modo. S. and Torr, S.J. (2006). The use of aerial spraying to eliminate tsetse from the Okawango Delta of Botswana. *Acta Tropica,* 99(2-3**):** 184 -199.

Mahama, C. I. Mohammed, H. A. Abavana, M.; Sidibe, I. Kone, A and Geerts, S. (2003). Tsetse and trypanosomoses in Ghana in the twentieth century: a review. *Revue d'Elevage et de medicine veterinaire des pays Tropicaux,* 56(1-2): 27 – 32.

Maldonado, B. (1910). (English abstract of Portuguese texts of 1906 and 1909), sleeping sickness *Bureau Bulletin,* 2: 26.

Mangwiro, T.N. C. Torr, S.J. Cox, J. R. and Holloway, M. T. P. (1999). The efficacy of various pyrethroid insecticides for use on odour-baited targets to control tsetse. *Medical and Veterinry Entomology,* 13:315 - 323

Maudlin, J. (1970). Preliminary studies on the karyotypes of the species of *Glossina*. *Parasitology,* 6: 71 – 74.

Mellanby, K. 1936. 'Experimental work with the tsetse fly *G. palpalis* in Uganda,' *Parasitology,* 31**,** 193. In: *The African Trypanosomiasis*. (Mulligan, H.W.; Potts, W.H, Ed) ODA, George Allen & Unwin. London, 950.

Mohammed-Ahmed, M. M. Abdulkarim, E. I. and Rahman, A. H. A. (1993). Reproductive status, catch and age composition of a Natural population of *Glossina morsitans submorsitans* in Bahr El Arab fly belt, Sudan. *Insect Science and its Application,* 14(4): 445 -453.

Mohammed-Ahmed, M. M. and Dairri, M.F. (1987). Trypanosome infection rate of *Glossina pallidipes* during wet and dry seasons in Somalia. *Tropical Animal Health Production,* 19:11 – 20.

Mohammed-Ahmed, M.M and Wynholds, Y. (1997). Effects of vegetation and weather on trap catches of *Glossina fudcipes* near Lake Victoria, Kenya. *Entomologia experimentalis et applicata,* 85(3):231-236.

Moiser, B. (1912). Notes on the haunts and habitats of *Glossina tachinoides* near Geidam, Bornu Province, Northern Nigeria. *Bulletin Entomological Research,* 3:195 -202.

Moore, A. C. Ryan, E. T. & Waldron, M. A. (2002). A 37 years old man with fever, hepatosplenomegaly, and a cutaneous foot lesion after a trip to Africa – East African trypanosomiasis (*Trypanosoma brucei rhodesiense* infection). *New England Journal of Medicine,* 346(26): 2069 – 2076.

Morlais, I. Grebaut, P.; Bodo, J.M. Djoha, S. Cuny, G.; Herder, B.(1998). Detection and identification of trypanosomes by polymerase chain reaction in wild tsetse flies in Cameroon. *Acta tropica,* 70(1) 109-117.

Morris, K.R.S and Morris, M.G. (1949). The use of traps against tsetse in West Africa. *Bulletin Entomological Research,* 39:491-528.

Nagel, P. (1995). Environmental monitoring handbook for tsetse control operations. Margraf Verlag**,** Germany, 323.

Nagel, P. and Perverling, R.(2005). Environment and the sterile insect technique. In: Sterile Insect Technique: Principles and practice an Area-wide Integrated Pest Management. Springer, the Netherlands, 499 – 524.

Nash, T.A.M.(1937). Climate, the vital factor in the ecology of *Glossina*. *Bulletin Entomological Research,* 28, 75 – 127.

Nash, T. A. M., (1940). The effect upon *Glossina* of changing the climate in the true habitat by partial clearing of vegetation. *Bulletin Entomological Research,* 31:69 - 84

Nash, T.A.M.(1969)**.** Africa's Bane. The tsetse fly. Collins Press, London, 224.

Ndams, I.S.(1987). Attraction of tsetse flies to experimental targets. M.Sc Thesis. Ahmadu Bello University, Zaria.

Offor, I. I.Carlson, D. A. Gadzama, N. M. Bozimo, H. T.(1981). Sex recognition pheromones in the West African tsetse fly, *Glossina palpalis* palpalis (Robineau-Desvoidy). *Insect Science and its Application,* 1:417- 420.

Okhoya, N.(2003). Eradicating tsetse flies from Africa. *African Recovery***,** 7(1):17.

Omoogun, G.A. Dipeolu, O. O. Akinboade, O. A., (1991). The decline of a *Glossina morsitans submorsitans* belt in the Egbe area of the derived savanna zone, Kwara State, Nigeria**.** *Medical and Veterinary Entomology,* 5: 43 – 50.

Omoogun, G.A.(1994). The design and construction of the Nitse trap. *Insect Science and its Application,* 15: 535- 539.

Omotainse, S. O. Edeghere, H. Omoogun, G. A. Elhassan, E. O. Thompson, G. Igweh, C.A. Ukah, J. A. C. Halid, M. A.(2000). The prevalence of Animal Trypanosomosis in Konshisha Government Area of Benue State, Nigeria**.** *Isreal Journal of Veterinary Medicine,* 55: 4.

Onah, J. A., Onyeka, J.O.A. and Tenabe, S. O. (1985). Studies on trypanosome infection rates of *Glossina* (Diptera: Glossinidae) in Pandam Wildlife Park, Nigeria. *Nigerian Journal of Entomology,* 6:53-59.

Onyiah, J. A.(1995). Tsetse distribution in 30 states and the Federal Capital Territory, *N.I.T.R. publication series*.

Onyiah, J.A.(1997). African Animal Trypanosomiasis: An overview of the current status in Nigeria. *Tropical Veterinary,* 15:111 – 116.

Owaga, M. L. A.(1981). Trypanosome infection rate in the tsetse species, *Glossina pallidipes* Austen, in a rural situation in Kenya. *Insect Science and its Application,* 1(4):411 – 416.

Phelps, R.J. and Vale, G.A.(1978). Studies on populations of *Glossina morsitansmorsitans* and *G. pallidipes* (Diptera: *Glossinidae*) in Rhodesia. *Journal of Applied Ecology,* 15:743-760.

Pieter de Raadt.(2002). **"**The First Millenia". Historical Perspectives on African Trypanosomosis. http//www. Wuhmo.ox.ac.uk/docs/africantrypan abs.html.

Pollock, J. N. 1982. Training manual for tsetse control personnel. Vol. 1. Editor: FAO Publ. No.M/P5178/E.280.

Pollock, J. N.(2000). Training manual for tsetse control personnel. Vol. II. Editor: FAO publ: No M/P 5444/E1.82/1.99.

Potts, W.H.(1970). Miscellaneous techniques used in study of Glossina. In: *The African Trypanosomiasis*. (Mulligan, H.W.; Potts, W.H, Ed) ODA, George Allen & Unwin. London**,** 950.

Potts, W. H.(1973)**.** *Glossinidae* (Tsetse flies). In: Insects and other arthropods of medical importance eds by Smith, K. G. U. 209 – 248.

Rogers, D. J. and Randolph, S. E., (2002). A response to the aim of eradicating tsetse from Africa. *Trends in Parasitology*, 18(12): 534 – 536.

Randolf, S. E. and Rogers, D. J., (1981). Physiological correlates of the availability of *Glossina morsitans centralis* Machado to different sampling methods. *Ecology and Entomology*, 6:63 – 77.

Sabbah, P. Brosset, P. Imbert, G. Bonardel, P. Jean del, J. Briant, F.(1997). Human African Trypanosomiasis: *Neuroradiology*, 39:708-710.

Saunders, D.S.(1962). Age determination for female tsetse flies and the age composition of samples of *Glossina pallidipes* Aust., *G. palpalis fuscipes* Newst.and *G. brevipalis* Newst. *Bulletin Entomological Research,* 53:579-595.

Service, M.W.(1980). A Guide to Medical Etomology. Macmillan International College Edition. The Macmillan Press Ltd. 95 – 101.

Spielberger, U., Na'isa, B.K. and Abdurrrahim, U.(1977). Tsetse (Diptera: Glossinidae) eradication by aerial (helicopter) spraying of persistent insecticides in Nigeria**,** *Bulletin Entomological Research,* 67: 589 -598.

Statistical Package for Social Sciences (SPSS) (2011).Version 20.0, IBM USA

Stephen, A.A.(1989). Intraspecific morphological variation in *Glossina pallidipes* (Diptera: Glossinidae) over its Geographical Range. Postgraduate Thesis.

Steiner, J. O.(1964). Tsetse control in *Rural Health Report*, 1963, Min. of Health, Northern Nigeria, Kaduna.

Turner, D. A.(1987). The populationecology of *Glossina pallidipes* Austen (Diptera: Glossinidae) in the Lambwe valley, Kenya. I. feeding behaviour and activity patterns. *Bulletin Entomological Research,* 77:317 -333.

Truc, P.(2003). About trypanosome brucei, gambiense, the causative agent of the chronic form of Human African Trypanosomiasis: some findings and proposal. *African Journal of Biotechnology,* 2(12):657 – 661.

Vale, G. A. and Torr, S. J.(2004). Development of bait technology to control tsetse. In the Trypanosomiasis (eds. I. Maudlin, P. H. Holmes & M. A. Miles) CABL Publishing, 509 – 523.

Van den Bossche, P. and De Deken, R.(2004). The application of bait technology to control tsetse. In the Trypanosomiasis (eds I. Maudlin, P. H. Holmes & M. A. Miles) CABL Publishing**,** 525 -532.

Van Etten, J.(1981). Comparative studies on the relative efficiency of two trap types for two allopatric populations of *Glossina pallidipes* in Kenya. *Entomologia Experimentalis Applicata,* 29:209 – 217.

Vreysen Mark, J. B.(1995). Radiation induced sterility to control tsetse flies. Grafisch bedrijf ponsen and Looijen B. V. the Netherlands, 282.

Wahab S., and Pigram, J.J. (1997). Tourism, Development and Growth: The Challenge of sustainability, *Routledge*, London**.**

Whitten, M. and Mahon, R. (2005). Misconceptions and constraints. In: Sterile Insect technique: Principles and Practice in Area-wide Integrated Pest management**.** *Springer*, the Netherlands. 601.

APPENDIX

I: Researchers on site during the collection of sample

II:. Educating the villagers on the mission about the samplings

III: Deworming of cattle as part of the incentives

IV: Species-specific occurrence of other flies caught in Kubau Local Government Area of Kaduna State

Species of flies	Total No of flies caught	Season Occurrence Wet season (%)	Dry season (%)
Butterfly	11(10.5)	1(100)	0(0.0)
Bee	4(3.8)	1(25.0)	3(75.0)
Grasshopper	9(8.6)	9(100)	0(0.0)
Wasp	4(3.8)	2(50.0)	2(50.0)
TOTAL	24	13	5

V: Species-specific occurrence of other flies caught in Ikara Local Government Area of Kaduna State

Species of flies	Total No of flies caught	Season occurrence Wet season (%)	Dry season (%)
Culex mosquitoes	3(2.4)	3(100)	0(0.0)
W asp	5(3.9)	5(100)	0(0.0)
Grasshopper	2(1.8)	2(100)	0(0.0)
Butterfly	3(2.4)	3(100)	0(0.0)
Bee	2(1.8)	2(100)	0(0.0)
TOTAL	15	15	0

VI. Percentage rate of other blood parasites in the samples collected in both LGA

Kubau LGA			Ikara LGA		
Parasite Prevalence	No. Positive	%Prevalence	Parasite Prevalence	No. Positive	%Prevalence
T. vivax	1	0.5	*T. vivax*	2	1
T. brucei	3	1.5	*T. brucei*	-	-
Total Samples Collected	100	5%	Total Samples Collected	100	3.5%

VII: The result of thin and thick blood smear examination of blood samples collected from 200 cattle in Ikara and Kubau LGAs

IKARA LGA BLOOD SAMPLE COLLECTION 17 MAY 15

S/N	TAG NO	THIN BLOOD SMEAR	THICK BLOOD SMEAR
1	C2	NPF	NPF
2	C24	NPF	NPF
3	C1	NPF	NPF
4	C26	NPF	NPF
5	C27	*T.vivax*	NPF
6	C13	NPF	NPF
7	C12	NPF	NPF
8	C22	NPF	NPF
9	C14	NPF	NPF
10	C5	NPF	NPF
11	C6	NPF	NPF
12	C9	NPF	NPF
13	C18	*T.vivax*	NPF
14	C7	NPF	NPF
15	C11	NPF	NPF
16	C31	NPF	NPF
17	C16	NPF	NPF
18	C17	NPF	NPF
19	C35	NPF	NPF
20	17	NPF	NPF

21	C16	NPF	NPF
22	C31	NPF	NPF
23	C11	NPF	NPF
25	C7	NPF	NPF
26	C8	NPF	NPF
27	C18	NPF	NPF
28	C9	NPF	NPF
29	C6	NPF	NPF
30	C5	NPF	NPF
31	C14	NPF	NPF
32	C22	NPF	NPF
33	C12	NPF	NPF
34	C13	NPF	NPF
35	C27	NPF	NPF
36	C26	NPF	NPF

VIII: KUBAU LGA BLOOD SAMPLE COLLECTION 09 AUG 15 (WET SEASON)

S/N	TAG NO	PCV (%)	WET MOUNT	THIN BLOOD SMEAR	THICK BLOOD SMEAR
1	C90	38	NPF	NPF	NPF
2	C60	42	NPF	NPF	NPF
3	C78	29	NPF	NPF	NPF
4	C82	32	NPF	NPF	NPF
5	C21	41	NPF	NPF	NPF
6	C92	39	NPF	*Theilera mutans*	NPF
7	C22	27	NPF	NPF	NPF
8	C86	38	NPF	NPF	NPF
9	C56	39	NPF	NPF	NPF
10	C97	43	NPF	NPF	NPF
11	C61	51	NPF	NPF	NPF
12	C83	47	NPF	NPF	NPF
13	C50	36	NPF	NPF	NPF
14	C89	39	NPF	NPF	NPF
15	C49	46	NPF	NPF	NPF
16	C84	41	NPF	NPF	NPF
17	C88	50	NPF	NPF	NPF
18	C35	49	NPF	NPF	NPF
19	C52	36	NPF	NPF	NPF
20	C79	38	Positive	*T.brucei*	NPF
21	C18	43	Positive	*T.brucei*	NPF
22	C76	36	NPF	NPF	NPF

IX KUBAU LGA BLOOD SAMPLING (WET SEASON)

S/N	TAG NO	PCV (%)	WET MOUNT	THIN BLOOD SMEAR	THICK BLOOD SMEAR
23	C8	38	NPF	*Anaplasma bovis*	NPF
24	C99	31	NPF	NPF	NPF
25	C11	29	NPF	NPF	NPF
26	C68	40	NPF	NPF	NPF
27	C12	43	NPF	NPF	NPF
28	C71	46	NPF	NPF	NPF
29	C75	57	NPF	NPF	NPF
30	C23	45	Positive	*T.brucei*	NPF
31	C1	29	NPF	NPF	NPF
32	C47	42	NPF	NPF	NPF
33	C85	28	Positive	*T.vivax*	*T.vivax*
34	C25	41	NPF	NPF	NPF
35	C28	37	NPF	NPF	NPF
36	C58	47	NPF	NPF	NPF
37	C58	45	NPF	NPF	NPF
38	C40	40	NPF	NPF	NPF
39	C17	38	NPF	NPF	NPF
40	C77	57	NPF	NPF	NPF
41	C87	39	NPF	NPF	NPF
42	C2	30	NPF	NPF	NPF
43	C27	37	NPF	NPF	NPF
44	C69	40	NPF	NPF	NPF

X IKARA LGA BLOOD SAMPLING 23 AUG 15 (WET SEASON)

S/N	TAG NO	PCV (%)	WET MOUNT	THIN BLOOD SMEAR	THICK BLOOD SMEAR
45	C96	46	NPF	*Theilera mutans*	NPF
46	C3	41	NPF	NPF	NPF
47	39	41	NPF	NPF	NPF
48	C95	50	NPF	NPF	NPF
49	C98	46	NPF	NPF	NPF
50	C65	43	NPF	NPF	NPF
51	C54	39	NPF	NPF	NPF
52	C74	40	NPF	NPF	NPF
53	C26	51	NPF	NPF	NPF
54	C64	46	NPF	NPF	NPF
55	C14	38	NPF	NPF	NPF
56	C91	36	NPF	NPF	NPF
57	C57	40	NPF	NPF	NPF
58	C39	46	NPF	NPF	NPF
59	C93	50	NPF	NPF	NPF
60	C36	37	NPF	NPF	NPF
61	C55	31	NPF	NPF	NPF
62	C53	36	NPF	NPF	NPF
63	C94	44	NPF	NPF	NPF
64	C5	40	NPF	NPF	NPF
65	C80	36	NPF	NPF	NPF
66	C63	41	NPF	*Anaplasma bovis*	NPF

67	C70	39	NPF	*Anaplasma bovis*	NPF
68	C13	42	NPF	*Theileria mutans*	NPF
69	C100	48	NPF	NPF	NPF
70	C81	38	NPF	NPF	NPF
71	C44	39	NPF	NPF	NPF
72	C32	43	NPF	NPF	NPF
73	C27	52	NPF	NPF	NPF
74	C30	57	NPF	NPF	NPF
75	C37	41	NPF	NPF	NPF
76	C45	31	NPF	NPF	NPF
77	C41	40	NPF	NPF	NPF
78	C15	50	NPF	NPF	NPF
79	C46	46	NPF	NPF	NPF
80	C16	33	NPF	*Anaplasma bovis*	NPF
81	C57	43	NPF	NPF	NPF
82	C31	48	NPF	NPF	NPF
83	C38	32	NPF	NPF	NPF
84	C10	34	NPF	NPF	NPF
85	C42	45	NPF	NPF	NPF
86	C34	36	NPF	NPF	NPF
87	C7	31	NPF	NPF	NPF
88	C6	49	NPF	NPF	NPF
89	C4	40	NPF	NPF	NPF
90	C72	32	NPF	NPF	NPF
91	C48	30	NPF	NPF	NPF

92	C73	41	NPF	NPF	NPF
93	C33	52	NPF	NPF	NPF
94	C68	38	NPF	NPF	NPF
95	C9	42	NPF	NPF	NPF
96	C82	46	NPF	NPF	NPF
97	C20	50	NPF	NPF	NPF
98	C43	38	NPF	NPF	NPF
99	C67	36	NPF	NPF	NPF
100	C24	31	NPF	NPF	NPF

S/NO	DATE	TAG NO	THIN BLOOD SMEAR	THICK BLOOD SMEAR
1	15 May 15	C35	NPF	NPF
2		C54	NPF	NPF
3		C46	NPF	NPF
4		C58	NPF	NPF
5		C43	NPF	NPF
6		C32	NPF	NPF
7		C33	NPF	NPF
8		C27	NPF	NPF
9		C16	NPF	NPF
10		C52	NPF	NPF
11		C11	*Babesia bigemina*	NPF
12		C30	NPF	NPF
13		C41	NPF	NPF
14		C22	NPF	NPF
15		C6	NPF	NPF
16		C15	NPF	NPF
17		C13	NPF	NPF
18		C31	NPF	NPF
19		C38	NPF	NPF
20		C42	NPF	NPF
21		C57	NPF	NPF

22	C18	NPF	NPF
23	C4	NPF	NPF
24	C14	*Theleria mutans*	NPF
25	C53	NPF	NPF
26	C31	NPF	NPF
27	C8	NPF	NPF
28	C7	NPF	NPF
29	C5	NPF	NPF
30	C34	NPF	NPF
31	C48	NPF	NPF
32	C50	NPF	NPF
33	C29	NPF	NPF
34	C21	NPF	NPF
35	C45	NPF	NPF
36	C40	*Theilera mutans*	NPF
37	C31	NPF	NPF
38	C19	NPF	NPF
39	C20	NPF	NPF
40	C2	NPF	NPF
41	C9	NPF	NPF
42	C12	NPF	NPF
43	C25	NPF	NPF
44	C26	NPF	NPF
45	C3	NPF	NPF
46	C36	NPF	NPF

47	C56	NPF	NPF
48	C55	NPF	NPF
49	C39	*Theleria mutans*	NPF
50	C44	NPF	NPF
51	C11	NPF	NPF
52	C47	NPF	NPF
53	C23	NPF	NPF
54	C24	NPF	NPF
55	C10	NPF	NPF
56	C49	NPF	NPF
57	C28	NPF	NPF

S/N	TAG NO	PVC %	HCT	WET MOUNT
1	C1	28%	NPF	NPF
2	C2	31%	NPF	NPF
3	C3	26%	NPF	NPF
4	C4	34%	NPF	NPF
5	C5	25%	NPF	NPF
6	C6	38%	NPF	NPF
7	C7	31%	NPF	NPF
8	C8	24%	NPF	NPF
9	C9	37%	NPF	NPF
10	C10	22%	NPF	NPF
11	C11	29%	NPF	NPF
12	C12	23%	NPF	NPF
13	C13	32%	NPF	NPF
14	C14	40%	NPF	NPF
15	C15	28%	NPF	NPF
16	C16	30%	NPF	NPF
17	C17	23%	NPF	NPF
18	C18	27%	NPF	NPF
19	C19	30%	NPF	NPF
20	C20	21%	NPF	NPF
21	C21	33%	NPF	NPF
22	C22	42%	NPF	NPF
23	C23	41%	NPF	NPF

24	C24	32%	NPF	NPF
25	C25	29%	NPF	NPF
26	C26	36%	NPF	NPF
27	C27	26%	NPF	NPF
28	C28	30%	NPF	NPF
29	C29	34%	NPF	NPF
30	C30	39%	NPF	NPF
31	C31	24%	NPF	NPF
32	C32	39%	NPF	NPF
33	C33	32%	NPF	NPF
34	C34	41%	NPF	NPF
35	C35	28%	NPF	NPF
36	C36	30%	NPF	NPF