EVALUATION OF SELECTED BOTANICALS AS INSECTICIDES AGAINST CABBAGE INSECT PESTS IN TANZANIA

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ABSTRACT

Synthetic pesticides are frequently and unwisely used to control cabbage insect pests by smallholder farmers despite the environmental pollution and insect pests' resistance development. This work assessed the insecticidal efficacy of botanicals from *Tephrosia vogelii, Croton dichogamus* and *Syzygium aromaticum* against cabbage insect pests in Northern Tanzania. Firstly, larvicidal action of extracts against *Crocidolomia binotalis* and *Plutella xylostella* larvae was assessed in the laboratory. Secondly, insecticidal and synergistic actions of aqueous extracts against cabbage insect pests were assessed in field experiment. Lastly, chemical compounds in *S. aromaticum* and in *C. dichogamus* were determined. The larvicidal activities of extracts were assessed for mortality of ten larvae into 9 cm Petri-dishes for 24 hours of exposure. Chlorpyrifos and acetone were used as a positive and negative control, respectively. The insecticidal efficacy of 10%, 5% and 1% w/v of *T. vogelii, C. dichogamus* and *S. aromaticum* aqueous extracts and their mixture (2.5% and 5%) was assessed against cabbage insect pests in the field. Chlorpyrifos was used as a positive control, and water and water plus soap were used as negative controls. The GC-MS was used for compounds identification in *C. dichogamus* and in *S. aromaticum.* Results from the study revealed that *S. aromaticum* extract (16, 24 and 32 mg/mL), *T. vogelii* (24 and 32 mg/mL) and *C. dichogamus* (32 mg/mL) gave 100 ± 0.0% mortality of *C. binotalis* larvae after 24 hours of exposure. Moreover, *S. aromaticum* extract (8, 16, 24 and 32 mg/mL), *T. vogelii* (16, 24 and 32 mg/mL) and *C. dichogamus* (32 mg/mL) gave 100 ± 0.0% mortality of *P. xylostella* larvae after 24 hours of exposure. The aqueous extracts from those plants significantly ($P \leq 0.05$) lowered the population of cabbage insect pests compared with negative controls. The 5% of aqueous extract from mixed plants possessed significantly ($P \leq 0.01$) lower population of cabbage insect pests in both wet seasons compared with other concentrations. Then, it was followed by 10% of *S. aromaticum, C. dichogamus* and *T. vogelii* aqueous extracts and 1% and 5% of aqueous extracts of *S. aromaticum, C. dichogamus* and *T. vogelii* and 2.5% of aqueous extracts from the mixed plants significantly lowered the population of insect pests compared with negative controls in both seasons. The compounds identified in *S. aromaticum,* at higher percent were Eugenol (52.66%); Eugenol acetate (20.46%) and β-caryophyllene (7.52%). Moreover, the compounds identified in *C. dichogamus* at higher percent were 4,6-Bis (4-chloro-3-(trifluoromethyl) phenoxy)-2-pyrimidinol (25.08%); Cholestan-6-en-3-ol (18.63%) and 1-Heptadecene (7.34%). These compounds could be responsible for larvicidal and insecticidal activities against cabbage

insect pests. Therefore, these plants can be recommended to be used by smallholder farmers for cabbage insect pest control at higher concentrations and development of insecticides.

DECLARATION

I, Nelson Mpumi, do hereby declare to the Senate of the Nelson Mandela African Institution of Science and Technology that this thesis is my own original work and that it has neither been submitted nor being concurrently submitted for degree award in any other institution.

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CERTIFICATION

The undersigned certify that they have read and hereby recommend for acceptance by the Nelson Mandela African Institution of Science and Technology a Thesis entitled: "Evaluation of selected botanicals as insecticides against cabbage insect pests in Tanzania" and recommend for examination in fulfillment of the requirements for the degree of Doctor of Philosophy in Environmental Science and Engineering of the Nelson Mandela African Institution of Science and Technology.

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DEDICATION

Dedication goes to my lovely father, the late Mr. Simon Mpumi Hayuda and My lovely Mother the Late Elizabeth Juma Pyuza. May their soul rest in peace.

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LIST OF ABBREVIATIONS AND SYMBOLS

ANOVA	Analysis of Variance
ATP	Adenosine Triphosphate
DCM	Dichloromethane
DDD	Dichlorodiphenyldichloroethane
DDT	Dichlorodiphenyltrichloroethane
DNA	Deoxyribonucleic Acid
GC	Gas Chromatography
GC-MS	Gas Chromatography Mass Spectroscopy
GPS	Global Positioning System
HCB	Hexachlorobenzene
HPLC	High Performance Liquid Chromatography
IPM	Integrated Pest Management
LC_{50}	Lethal Concentrations which Kills 50% of the Exposed Larvae
LC_{90}	Lethal Concentrations which Kills 90% of the Exposed Larvae
LCL	Lower Confidence Limit
LD_{50}	Median Lethal Dose which Kills 50% of Tested Organisms in a Population
LSD	Fisher's Least Significant Difference
MeOH	Methanol
mg/kg	Milligram Per Kilogram
NIST	National Institute of Standards and Technology
NM-AIST	Nelson Mandela African Institution of Science and Technology
PD	Percentage of Damage
POPs	Persistent Organic Pollutants
RCBD	Randomized Complete Block Design
RH	Relative Humidity
SEM	Standard Error of the Mean
TPRI	Tanzania Pesticides Research Institute
UCL	Upper Confidence Limit
UV	Ultraviolet Radiations
WHO	World Health Organization

CHAPTER ONE

INTRODUCTION

1.1 Background of the problem

Globally, there are increasing reports of hunger in various parts of the world due to increasing human population (Ramankutty *et al.*, 2018). This trend resulted in an increase in efforts of production of agricultural products towards the achievement of Goal number 2- zero hunger of the Sustainable Development Goals (SDGs) (Assembly, 2015). Pest damage during crop active vegetative growth, harvest and storage is one of the constraints limiting enhanced crop production. To address this constraint, various strategies for pest control are being developed (Gill & Garg, 2014). The main strategy is the use of synthetic pesticides, either in field or storage of crops (Were *et al.*, 2016). Synthetic pesticides have been used intensively for a long time to control insect pests (Obopile *et al.*, 2008). They significantly reduce the crop losses, improve the yield of crops, improve leafy vegetables, potatoes, cotton, increase the economic benefits of the crops (Ntow *et al.*, 2006), work quickly, are easy to apply and are less labour intensive (Macharia *et al.*, 2005). However, besides their beneficial effects, synthetic pesticides have potential environmental and public health impacts (Ntow *et al.*, 2006).

In the environment, the use of synthetic pesticides results into water contamination and soil pollution. Synthetic pesticides cause insect pest resistance and threaten the human health (Chikukura *et al.*, 2011; Ntow, 2008; Obopile *et al.*, 2008). To overcome insect pest resistance, smallholder farmers have decided to increase the frequency and dosages of pesticide applications (Macharia *et al.*, 2005) which intensify the water contaminations and soil pollution and threaten the aquatic and soil ecosystems (Ondieki, 1996). Synthetic pesticides can stay in the soil or water bodies for many days, even years and can cause bioaccumulation and biomagnifications in the bodies of the organisms (Mpumi *et al.*, 2020). Thus, most of the organisms in the aquatic environment can be killed and the whole ecosystem can be destroyed (Mpumi *et al.*, 2020). This phenomenon, affects the quality of water chemically, biologically and physically. The contamination of soil affects the quality of soil and the soil macro and microorganisms which are decomposers of organic matter (Mpumi *et al.*, 2021). Synthetic pesticides, also, affect the health of farmers during preparation, application in the farms and the consumption of vegetables (Kapeleka *et al*., 2019). Due to these constraints, scientists and smallholder farmers are looking for alternatives to synthetic pesticides (Mkindi *et al*., 2017;

Grzywacz *et al.*, 2014; Mochiah *et al.*, 2011). Phytochemicals or botanical pesticides can serve as promising alternatives.

Botanical pesticides also called botanicals or phytochemicals are naturally occurring chemicals extracted from plants (Henn & Weinzierl, 1989; Mudzingwa *et al.*, 2013b). Botanical pesticides have been used extensively in the protection of cereal crops from insect pests in the field and during storage. This is because they are affordable (de Seffrin *et al.*, 2010), easy to prepare and use (Amoabeng *et al.*, 2014), environmentally friendly (Mochiah *et al.*, 2011), degraded rapidly in sunlight, air, and moisture and are readily broken down by detoxification enzymes and have reduced risks of toxicity to human and to nontarget organisms (Amoabeng *et al.*, 2014; Isman, 2006). However, there is little information on the use of botanicals from plants to control insect pests in cabbage crops in the field.

Cabbage (*Brassica oleracea* var. capitata), is an essential leafy plant grown as an annual mainly for use as a vegetable crop (Baidoo & Mochiah, 2016; Gyanoba, 2018). Cabbage is a leafy vegetable of *Brassica* family and is round, oblate and pointed shapes. *Brassica* family vegetables are very important and are intensively grown for resource poor African smallholder farmers for subsistence and a source of income (Mudzingwa *et al.*, 2013). Cabbage is a water loving plants. Thus, it is grown in the areas with enough supply of water. Cabbage has soft, light green or whitish inner leaves covered with harder and dark green outer leaves. Cabbages are full of vitamins such as vitamin K and C and the dietary fibers and full of potassium and manganese (Gyanoba, 2018) and it has antioxidant and anti-inflammatory properties in the body of human being. Also, it has detoxifying effect due to its high sulphur and vitamin C contents (Baidoo & Adam, 2012). Cabbage is commonly used all over the world and can be prepared in a number of ways for eating and most frequently, it is included as either a cooked or raw part of many salads (Baidoo & Adam, 2012; Baidoo & Mochiah, 2016).

However, cabbage is susceptible to insect pest infestations in the field, which causes huge loses to the growers (Baidoo & Mochiah, 2016). The insect pests affecting cabbage (*B. oleracea*) production worldwide, Africa and Tanzania particularly include diamondback month (*Plutella xylostella*), dipterous leafy miner (*Liriomyza trifolii*), aphids (*Brevicoryne brassicae*), cabbage webworms (*Hellula undalis*), cutworms (*Agrotis spp.*), armyworm (*Spodoptera exempta*), cucumber beetles (*Diabrotia spp.*), stem borer (*Chilo partellus*), thrips (*Thrips tabaci*), whiteflies (*Bemisia tabaci*), sawflies (*Neodiprion spp.*) and cabbage looper (*Trichoplusia ni*) (Ntow *et al.*, 2006; Nuessly & Webb, 2003). These insect pests cause huge damage to the

leaves, stems and the heads of cabbage plants at different stages during growth and development (Baidoo & Adam, 2012). The infestations on cabbage crops affect the quality and the quantity at the market (Furlong *et al.*, 2013). Baidoo and Adam (2012) reported that, diamondback moth is the most dangerous insect pest affecting cabbage crops whose larvae eat the leaves and the center of cabbages. Damage occurs when the first-instar larvae mine leaf tissues, while the later instars consume the leaf tissues from the underside, chewing irregular patches and often leave the top epidermal layer and leaf veins with a window-like appearance (Baidoo & Adam, 2012). The cabbages (*Brassica species*) which are commonly cultivated in Africa and infested by these insect pests include cabbage (*B. oleracea*) and Chinese cabbage (*Brassica campestris*) (Ntow, 2008). These green vegetables are commonly used all over the world and normally are included as either cooked or raw parts and generates substantial income for the producers and other agents involved in the marketing system (Macharia *et al.*, 2005).

Most of cabbage gardeners in African countries in which the use of modern technology is limited rely on traditional practices like site selection, crop rotation and seed selection, sowing date, row spacing, plant density and weeds control to reduce insect pests' infestation in cabbage crops (Baidoo & Adam, 2012). However, these cultural practices are less effective to protect cabbage crops from insect pests although are safe to the environment and cheap (Mpumi *et al.*, 2020). Natural enemies of insect pests like predators, parasitoids and pathogens called biological controls (Flint *et al.*, 1998) help to reduce insect pest infestations. Pathogens like *Bacillus thuringiensis*, control certain caterpillars, beetles and flies (Kareru *et al.*, 2013). Biological control is safe and eco-friendly but requires intensive study to know the efficacy to control insect pests in cabbage crops in the field.

In the present study, the efficacy of botanicals from *T. vogelii, C. dichogamus* and *Syzygium aromaticum* were assessed to control insect pests in cabbage (*B. oleracea*) crops in the field. Normally, *T. vogelii* is known as the "fish bean", "fish-poison bean", or "vogel's tephrosia" (Orwa *et al.*, 2009). Smallholder farmers in many countries in Africa use *T. vogelii* as an organic pesticide to control pests on livestock, in cultivated fields (Mkindi *et al*., 2019) and as medicine for skin diseases (Munthali *et al.*, 2014). The other pesticidal plant, *S. aromaticum* contains a spicy chemical compound known as eugenol (Kamatou *et al.*, 2012). This chemical compound in *S. aromaticum* is used as an insect repellent (Araujo *et al.*, 2016). This property of insect repellent has been reported to be potential in agriculture to protect foods from micro-organisms during storage (Tian *et al.*, 2015). Moreover, *S. aromaticum* has a variety of

pharmacological activities including antimicrobial, anti-inflammatory, analgesic, anti-oxidant and anticancer activities (Araujo *et al.*, 2016). Globally, *Croton species* are usually used in folk medicines for the treatment of various health problems such as cancer, constipation and diabetes (Silva *et al.*, 2018). The phytochemical investigations of the *Croton species* revealed the presence of various secondary metabolites including alkaloids, phenolics and terpenoids in all plant's parts (Aldhaher *et al.*, 2017).

However, there is limited information on the applications of *S. aromaticum, T. vogelii* and *C. dichogamus* aqueous extracts to control insect pests of cabbage (*B. oleracea*) crops in the field. Therefore, this study focused on assessment of insecticidal efficacy of botanicals extracted from *T. vogelii, C. dichogamus* and *S. aromaticum* to control insect pests on cabbage in the field in Northern part of Tanzania.

1.2 Problem statement

Several studies on the efficacy of botanical pesticides for controlling insect pests are reported extensively in cereal crops such as maize, beans, rice, cowpeas both in the field and during storage. However, few studies on the use of botanical pesticides for controlling insect pests on cabbage leafy green vegetable are reported. Specifically, little information exists on insecticidal actions and synergistic effects of various botanical pesticides including their derivatives on insect pests infesting cabbage leafy vegetable crops found specifically in Tanzania. Therefore, this study intended to assess the insecticidal actions and synergistic effects of *T. vogelii, C. dichogamus* and *S. aromaticum* including their derivatives against insect pests affecting cabbage (*B. oleracea*) in the field.

1.3 Rationale of the study

This work was proposed to study the efficacy and potentials of the botanicals/ phytochemicals extracted from *T. vogelii, C. dichogamus* and *S. aromaticum* for the control and management insect pests affecting cabbage (*B. oleracea*) in Tanzania. The leaf extracts from *T. vogelii* and *C. dichogamus* and also the bud's extracts were tested for their effectiveness against insect pests affecting cabbage crops. In addition, the plant extracts were analyzed by GC-MS and confirmed for the presence of important compounds with insecticidal properties which could help to manage insect pests. All plant extracts effectively controlled and managed the cabbage insect pests. Therefore, the application of these important pesticidal plants for the control of

cabbage insect pests were the best option to the smallholder cabbage (*B. oleracea*) farmers in Tanzania.

1.4. The objectives of the study

1.4.1 General objective

Assessment of insecticidal actions exhibited by botanicals (*T. vogelii, C. dichogamus* and *S. aromaticum*) against common insect pests affecting cabbage in Northern Tanzania.

1.4.2 Specific objectives

(i) To assess the larvicidal action of extracts from *T. vogelii, C. dichogamus* and *S. aromaticum* against selected common cabbage insect pests.

(ii) To evaluate the insecticidal and synergistic actions of aqueous extracts from *T. vogelii, C. dichogamus* and *S. aromaticum* against common cabbage insect pests.

(iii) To determine the bioactive chemical compounds, present in the two selected pesticidal plants, *S. aromaticum* and *C. dichogamus.*

1.5 Research questions

(i) To what extent does extracts from three selected plants exhibit larvicidal action against common insect pests affecting cabbage crops.

(ii) What is the insecticidal and synergistic action of aqueous extracts from selected plants against common cabbage insect pests?

(iii) What are the bioactive chemical compounds present in *S. aromaticum* and *C. dichogamus* plants?

1.6 Significance of the study

The results of this study will provide knowledge to the smallholder farmers and the society in general about the easily available, effective and environmentally friendly control technology of the insect pests affecting the growing of cabbage (*B. oleracea*) in the field. Also, the findings of this study will provide knowledge, awareness and positive attitudes to the authorities and policy makers about the usefulness of the botanical pesticides for controlling insect pests of

cabbage for the purpose of saving the environment from pollutants as well as enhancing food production and raise household income.

1.7 Delineation of the study

The insecticidal activity of the botanicals extracted from *T. vogelii, C. dichogamus* and *S. aromaticum* was tested against cabbage insect pests in laboratory and in the field. In the laboratory bioassay, only *P. xylostella* and *C. binotalis* were tested for larvicidal activity because these insect pests were easy to collect and rear in the laboratory and get large colony for larvae production. Also, the aqueous extracts of these plants were tested against cabbage insect pests in field conditions to assess their efficacies. The efficacy of these plants was only tested against cabbage insect pests. However, they can be tested against other insect pests in the other crops due to differences in the ecologies of the insect pests.

CHAPTER TWO

LITERATURE REVIEW

2.1 Literature review

Various works reviewed the biological cycle of *Brassica species* and their common insect pests, common practices used by cabbage (*B. oleracea*) growers to control the insect pests of cabbage crops, effectiveness of various botanicals from pesticidal plants and the synergistic effects of botanicals on controlling insect pests of cabbage crops.

2.2 The biological cycle of *Brassica species* and their common insect pests in Africa

2.2.1 Propagation and biological cycle of selected *Brassica species*

The propagation and regeneration of *Brassica species* has been successful using seeds and different explants like petioles, cotyledons, stems and shoot tips (Basak *et al.*, 2012) as presented in Table 1. Shoot regeneration and rooting of *Brassica species* are successfully obtained from cotyledons and hypocotyl explants (Basak *et al.*, 2012). The shoot tip explants of *Brassica* species are reported to be effective for initiating shoots and roots (Zhang *et al.*, 2002). Table 1 shows the *Brassica species* propagation and biological cycle length to maturity.

Table 1: Propagation and biological cycle length of *Brassica species*

Name of Brassica Species	Propagation	Biological Cycle Length	References
Brassica oleracea L.	Conventional propagation is through seed, with seedlings being raised in seedbeds or pots and then transplanted to field sites. However, some *B. oleracea* subspecies, such as tronchuda, can be propagated through vegetative from stem and side shoot cuttings whereby the stem and side shoot cuttings are obtained from 5-week-old plants, which is rooted, and transplanted as normal cuttings	Seed germinates within 5 days after sowing at 20–25 ℃. The *species* takes 80 to 180 days to maturity	Msikita *et al.* (1992)
Brassica juncea L.	Conventional propagation is through seeds also, it has been successfully by using petioles, cotyledons, stems and shoot tips as explants.	Seed germinates within 5 days after sowing at 20–25 ℃. It takes 30 to 50 days to maturity.	Basak *et al.* (2012); Eapen *et al.* (1989)
Brassica napus L.	Conventional propagation is through seeds. The seedlings are raised in seedling trays or in a seedbed. Also, it is propagated successful by using stems, cotyledons, nodal stems and hypocotyl as explants in vitro.	The seeds take 3–5 days to emerge and about 90 days to mature at 20–25 ℃.	Basak *et al.* (2012); Dubey and Gupta (2014); Sharma and Gupta (2012)
Brassica rapa L.	Conventional propagation is done using seeds but also, the propagation is successful through petioles, stems, cotyledons, stems and shoot tips as explants in vitro.	The seeds require 3–5 days to germinate at 20–25 ℃. It takes 30 to 35 days to mature	Basak *et al.* (2012)
Brassica campestris L.	Conventional propagation is through seeds. Also, petiole and cotyledons can be used in the development of plants in vitro culture. Four-day seedlings are enough to give a viable *Brassica campestris* plants	The seeds require 3–5 days to germinate and 50 to 60 days to mature at 20–25 ℃	Basak *et al.* (2012); Dubey and Gupta (2014); Hachey *et al.* (1991)
Brassica nigra	The propagation is done using seeds. The small seeds require a level and a well-prepared seedbed.	The first leaves are usually visible within 48 hours and 60 to 90 days to mature	Smartt and Immonds (1995)

2.2.2 Common insect pests affecting cabbage (*B. oleracea*) in Africa

Many insect pests such as diamondback moth (*Plutella xylostella*), cabbage webworm (*Helula undalis*), cabbage white butterfly (*Pieris brassicae*), the cabbage aphids (*Brevicoryne brassicae*), green peach aphids (*Myzus persicae*) and cabbage loopers (*Trichoplusia ni*) (Baidoo & Adam, 2012; Baidoo & Mochiah, 2016) (Table 2) hinder the proper cabbage crop production on the field in Africa and Tanzania particularly. The insect pests named in Table 2

infest the cabbage crops at different stages of growth, causing significant damage to the crops (Labou *et al.*, 2017) and resulting into huge cabbage yield losses. Krishnamoorthy (2004) showed that cabbage insect pests all together can cause 52% yield loss on cabbage. Severe infestation by *Plutella xylostella* usually causes huge economic crop losses and may result into 100% yield loss of the cabbage (*B. oleracea*) (Waiganjo *et al.*, 2011). Due to heavy infestations which result into huge losses, the African smallholder farmers of cabbage crops spray four or more than four times in a month and two or more than two mixed insecticides into the field for strongly and effectively control of the cabbage insect pests (Ahouangninou *et al.*, 2011; Ngowi *et al.*, 2007). The consequence of that scenario is environmental pollution especially water and soil, detrimental effects to non-target organisms and endanger the health of human being (Ondieki, 1996). This section reviews the major insect pests of economic importance infesting cabbage crops at different stages of growth in African countries and how the control measures are potential water and soil pollution threat.

Table 2: Common insect pests infesting cabbage (*B. oleracea*) crops

Common Name	Scientific Name	Parts of Cabbages Damaged	References
Dimondback moth	*Plutella xylostella*	Cabbage heads and foliar tissues	Baidoo and Adam (2012)
Cabbage webworm	*Hellula undalis*	Leaves, petioles and heads of cabbages	Weinberger and Srinivasan (2009)
Cabbage white butterfly	*Pieris brassicae*	Head of cabbage and leaves	Baidoo and Adam (2012)
Cabbage aphids	*Brevicoryne brassicae*	Tips, flowers and leaves	Baidoo and Mochiah (2016)
Green peach aphids	*Myzus persicae*	Tips, flowers, developing pods and leaves	Baidoo and Mochiah (2016)
Cabbage looper	*Trichoplusia ni*	Leaves, stems and veins of leaves	Baidoo and Adam (2012)
Cabbage leaf webber (CLW)	*Crocidolomia binotalis*	Leaves, stems and veins of leaves	Usui *et al*. (1987)

(i) Cabbage looper (*Trichoplusia ni*)

The cabbage looper (Plate 1A) (*Trichoplusia ni*) is a moth found in the family noctuidae, a family which is commonly referred to as owlet moths (Lingren & Green, 1984). Its common name comes from its preferred host plants and distinctive crawling behavior. The members of noctuidae are brown or gray night-flying moths whereby the larvae infest the growing cruciferous vegetables (Chomchalow, 2003). Cruciferous vegetables like cabbages and broccoli are the main host plants to cabbage lopper and hence, the reference to cabbage in its

common name (Capinera, 2001). The larva is called a looper since it arches its back into a loop when it crawls (Lingren & Green, 1984). While crucifers are preferred, however, over 160 plants can serve as hosts of cabbage looper larvae (Lingren & Green, 1984). The adult cabbage looper is a migratory moth and its migratory behavior can be found in a wide range of host plants and this contributes to its wide range of distribution (Chomchalow, 2003).

The cabbage looper larvae is a vegetable pest for crucifers and has been reported to damage broccoli, cabbages, cauliflowers, Chinese cabbages, collards, kale, mustards, radish, turnip and watercress (Chomchalow, 2003). The cabbage looper larvae interfere with plant growth and marketability by making irregular holes of variable shapes (Plate 1A and 1B) while feeding on the leaves of the host cabbage plants (Fening *et al.*, 2013). Although it is not extremely destructive, but it is becoming difficult to control and manage due to its broad distribution and resistance to many insecticides (Capinera, 2001; Fening *et al.*, 2013). Therefore, African smallholder farmers rely intensively on the application of synthetic pesticides to control the cabbage insect pests. However, synthetic pesticides result into environmental pollution, insect pest resistance and contaminate the foods which consequently threaten human health (Bolor *et al.*, 2018). Therefore, environmental benign, the botanical pesticides from *T. vogelii, S. aromaticum* and *C. dichogamus* can be utilized to control the insect pests in the field instead of synthetic pesticides. Although the potentialities are ignored, but botanical pesticides have been in use for centuries by smallholder farmers in developing countries to control insect pests of both field and stored products (Begna & Damtew, 2015; Isman, 2006). Therefore, they could be used to control cabbage insect pests in the field to minimize the infestation.

Plate 1: (A) Mature larvae of cabbage looper, *Trichoplusia ni*. (B) The cabbage plant damaged by Cabbage looper larva. Photograph by Nelson Mpumi, NM-AIST-Arusha, Tanzania

(ii) Cabbage webworm *(Hellula undalis)*

Among the most destructive insect pests which attack cruciferous vegetables is the cabbage webworm (Plate 2A) (*Hellula undalis*) (Lepidoptera: Pyralidae) (Sivapragasam & Aziz, 1992). The cabbage webworm (*H. undalis*) is a major pest of cruciferous crops in the tropics and subtropics (Pérez-Lucas *et al.*, 2018; Shine *et al.*, 2003). It is a widespread species in the world especially in Europe across Asia to the Pacific and also, in African countries (Ebenebe *et al.*, 2011; Waterhouse & Sands, 2001). Shine *et al.* (2003) reported that *H. undalis* is distributed mostly in tropical and subtropical regions but can similarly be found in countries with moderate climates.

Ebenebe *et al*. (2011) reported that, *H. undalis* larva causes serious and severe damage to the leaves and the heads of cabbages (Plate 2B) in the field. According to Waterhouse and Norris (1987), *H. undalis* feeds on a variety of plants, especially the *Brassicaceae* family members. Waterhouse and Norris (1987) revealed that *H. undalis* larva can cause a huge yield loss of up to 100% to crucifers crops in the field and if its management is not well considered. The larvae feed on leaves, petioles, growing points and stems (Waterhouse & Sands, 2001). According to Sivapragasam and Aziz (1992) and Waterhouse and Sands (2001), the plants in which *H. undalis* larvae feed include broccoli, head cabbage, Chinese cabbage, spoon cabbage, daikon radish, horseradish, mustard, radish and turnip. Shine *et al.* (2003) revealed that *H. undalis* is

a very serious agricultural pest to crucifer crops grown by the African smallholder famers. The incidence of *H. undalis* did not depend on the number of insecticide applications, but depend highly on host crop abundance and the temperature of the area (Labou *et al.*, 2017). The larvae make mines in the leaves and bore into the stem and later, they tunnel into the heart of the plant, destroying the bud causing the leaves to become distorted and stunted (Baidoo & Mochiah, 2016). A study done by Sivapragasam and Aziz (1992) indicated that a single larva of cabbage webworm, can either cause a number of deaths to the young plant or lead to the formation of unmarketable multiple heads on relatively older plants. On the field, a low population of larvae can cause very huge significant losses to the cabbage crop and in untreated cabbages, losses could go as high as 99% (Ebenebe *et al.*, 2011). Although, the larva can be present throughout the cropping season, it is severe only during the period between transplanting and the heading stage of cabbage crops (Li *et al.*, 2016).

Currently, African smallholder farmers rely intensively on the application of synthetic pesticides as the only effective control method to the cabbage webworm on the field (Sivapragasam & Aziz, 1992). The effective insecticides which are used to control cabbage webworm worldwide and Africa particularly, include permethrin, abamectin, teflubenzuron, chlorfluazuron, triflumuron, phenthoate, exthofenprox and Lambda-cyhalothrin and among those insecticides, abamectin is found to be the most effective of the other insecticides (Sivapragasam & Aziz, 1992). However, some are reported hazardous and therefore unwise and overuse of those insecticides can result into severe environmental pollution especially water and soil, development of insect resistance to some of insecticides and health problems to human beings. Therefore, there is a need of searching and using benign and environmentally friendly botanicals from pesticidal plants as a cabbage insect pest control strategy.

Plate 2: (A) Mature larva of the cabbage webworm, *H. undalis*. Photograph by Lyle Buss, Entomology and Nematology Department, University of Florida (March, 2016). (B) The head of cabbage crop damaged by larva of *H. undalis*. Photograph by Nelson Mpumi, NM-AIST Arusha, Tanzania

(iii) Diamondback month *(Plutella xylostella)*

The diamondback moth (Plate 3A) (*Plutella xylostella*), sometimes called the cabbage moth, is a moth species belonging to the family Plutellidae and genus *Plutella* (Li *et al.*, 2016). Badenes-Perez *et al.* (2006) reported that *P. xylostella* is believed to have originated in Europe, South Africa, or the Mediterranean region, but it has now spread worldwide. The diamondback moth is the dominant and most destructive insect pest of cruciferous crops worldwide (Begna & Damtew, 2015). Justus and Mitchell (1996) reported that, *P. xylostella* larvae feed on the leaves between the large veins and midribs of cruciferous crops and the plants which produce glucosinolates. *Plutella xylostella* larvae prefers to feed on the lower leaf surface, leaving the upper epidermis intact creating a "window-paning" effect (Plate 3B) (Begna & Damtew, 2015). Timbilla and Nyarko (2004) showed that, severe feeding damage (Plate 3B) stunts and destroys the cabbage heads and can cause heads to abort leading to huge yield depression and total crop loss. The most cabbage plant damage is caused by larval feeding resulting in a complete removal of foliar tissues and disrupt head formation in cabbages, broccoli and cauliflower (Begna & Damtew, 2015). The destruction of cruciferous crops by diamondback moth larva reduces the quality and the marketability of the cabbage crops and hence yield losses due to *P. xylostella* can go up to 100% (Waiganjo *et al.*, 2011; Weinberger & Srinivasan, 2009) and vary widely depending on the season and severity of pest infestation (Ayalew, 2006).

Generally, it is estimated that diamondback moths cause an annual loss of about 16 million dollars on the basis of 2.5 per cent damage even on the protected crops (Mohan & Gujar, 2003). Also, in the tropics, diamondback moths cause threat of great loss of 90% and above to crucifer production crops (Charleston *et al.*, 2006). Therefore, there is a need to conduct research to determine the cabbage losses due to infestation of diamondback moths in various parts of Africa.

The diamondback moth and its larvae control in cabbage by African smallholder farmers is still deeply dependent on chemical insecticides although their use is connected with many adverse and lethal consequences. Inappropriate and excessive application of chemical insecticides result into environmental pollution especially water and soil pollution (Dalvie *et al.*, 2003; Schulz *et al.*, 2001). Pedigo and Rice (2014) indicated that extreme use of insecticides also induces resistance development in target pests as well as killing beneficial organisms like pollinators such as bees and other natural enemies such as spiders, lacewings and ladybird beetles. Therefore, the benign, environmentally friendly botanicals have to be searched to control this pest instead of relying on synthetic pesticides which have many negative impacts and problems to the environment. The benign and environmentally friendly control measures with broad spectrum of the activities are the botanicals, the chemicals from pesticidal plants (Amoabeng *et al.*, 2013). Those alternatives with antifeedant, repellency and insect growth regulators of their natural origin having non-neurotoxic modes of action to human beings and low environmental persistence can be applied.

Botanical pesticides are not only effective against crop pests but remain safe to the environment and to natural enemies (Mkenda *et al*., 2015). In developing countries, botanicals have been in use for centuries by smallholder farmers to control insect pests both in field and storage (Begna & Damtew, 2015). For instance, nicotine, rotenone and pyrethrum were famous and among the botanical insecticides used in those days (Isman, 2006). Those chemicals from pesticidal plants possess one or more useful properties like repellency, anti-feeding, fast knock down, flushing action, biodegradability, broad spectrum of activity and ability to reduce insect resistance (Isman, 2006; Mochiah *et al.*, 2011). Therefore, there is a need to use environmentally friendly products, for instance, the botanicals/phytochemicals from *T. vogelii*, *S. aromaticum* and *C. dichogamus* to control cabbage insect pests in the field.

Plate 3: (A) Mature larva of Diamondback moth, *P. xylostella*. (B) The cabbage plant damaged by larva of *P. xylostella*. Photograph by Nelson Mpumi. NM-AIST, Arusha, Tanzania

(iv) Cabbage aphids (*Brevicoryne brassicae*)

Cabbage aphid (Plate 4A) (*Brevicoryne brassicae*) belongs to the family Aphididae of the order Hemiptera (Mersha *et al.*, 2014) and the genus *Brevicoryne* (Gill *et al*., 2013). The name is derived from two Latin words "brevi" and "coryne" and which means "small pipes" (Mersha *et al.*, 2014). These aphids have two small pipes called cornicles or siphunculi at the posterior end which can be observed when using a hand lens during the observations (Carter & Sorensen, 2013b). The cornicles of the cabbage aphid are comparatively shorter than the cornicles of other aphids except those of the turnip aphid, *Lipaphis erysimi* (Carter & Sorensen, 2013b). The short cornicles and the waxy coating present on cabbage aphids differentiate cabbage aphids from other aphids which can attack the same host plants (Carter & Sorensen, 2013b; Opfer and McGranth, 2013). The cabbage aphid is native to Europe, but now has a worldwide distribution (Opfer & McGranth, 2013) and can be found in Asia, Canada, Australia (Ahmad & Akhtar, 2013), America, India, China and Netherland (Gill *et al.*, 2013) and also in African countries.

Jahan *et al*. (2013) and Moharramipour *et al.* (2003) indicated that, cabbage aphids are serious plant sap sucking pests worldwide. Cabbage aphids are the most common damaging species causing significant yield loss to many crops of Brassicaceae, like the mustards and crucifers (Blackman & Eastop, 2000; Mudzingwa *et al.*, 2013). Blackman and Eastop (2000) insisted that, cabbage aphids mostly attack growing parts of the host plants such as tips, flowers,

developing pods, leaves and eventually cover the whole plants (Plate 4B) at high population. According to Elwakil and Mossler (2016) and Lashkari *et al.* (2007), cabbage aphids (Plate 4A) have direct and indirect damaging effects to cabbage crops. The direct damage caused by this pest is by sucking cell sap, secrete honeydew which result into sooty mold formation on leaves and shoots and indirect damaging effect is as a vector of 20 plant viral diseases in a wide range of plants. According to Valenzuela and Hoffmann (2015) and Zaker and Mosallanejad (2010), the damaging viruses transmitted by cabbage aphids are such as potato leafroll virus, potyviruses, beet western yellows, beet yellows, cauliflower mosaic, cucumber mosaic, lettuce mosaic, turnip mosaic and watermelon mosaic. High population and feeding of cabbage aphids result into curling, distortion and yellowing of leaves, stunting plant growth, deforming developing heads, damaging of flowers and green pods and discoloration of any growth stage and part of plants (Ahmad & Akhtar, 2013; Carter & Sorensen, 2013b). Feeding by cabbage aphids can stop terminal growth resulting into reduced plant size and yield (Liu & Sparks, 2001).

Eliminating weeds in Brassicaceae field borders is one of the cultural methods which may help to reduce the population and damage of the cabbage aphids (Lashkari *et al.*, 2007; Liu *et al.*, 2014). However, cultural methods alone are less effective to completely control the cabbage aphids from the farmers' field (Mersha *et al.*, 2014). So, biological control can play a major role in the natural suppression of aphids. Among the biological controls which can be applied to control the aphids are the natural enemies such as ladybird beetles adult and larvae, lacewing larvae, syrphid fly larvae, predatory bugs and lacewing adults (Lashkari *et al.*, 2007; Liu *et al.*, 2014). Other biological control agents are entomopathogenic fungi, which particularly can be applied during the periods of high humidity and precipitation (Liu *et al.*, 2014). However, natural enemies alone and other biological controls are also insufficient to prevent economic damage by a rapidly increasing population of cabbage aphids (Zaki, 2008).

Due to high pest pressure and damage caused by those aphids on cabbages in African countries, growers resort to excessive and intensive chemical pesticides application for aphids and other insect pest management (Mersha *et al.*, 2014). Chemical pesticides are intensively, excessively and doubly rated for insect pest management (Hines & Hutchison, 2013). However, intensive and heavy reliance on the application of synthetic pesticides results into extreme soil and water pollution and pose serious threats to the non-target organisms including human beings (Mersha *et al.*, 2014; Razaq *et al.*, 2012). For instance, Bami (1997) reported that, every year, one

million people are suffering from pesticide poisoning in India. The pesticide poisoning threatens the health of human beings and natural enemies. Also, the soil pollution threatened the soil ecosystems. Decomposers are also in danger due to soil pollution through excessive and intensive application of synthetic pesticides (Ondieki, 1996).

Due to those problems associated with the application of synthetic pesticides, there is a need of assessing the potential of botanical pesticides from various plants such as *T. vogelli, S. aromaticum* and *C. dichogamus* for cabbage aphid control and management in the field. Botanicals from different pesticidal plants have many advantages over synthetic pesticides such as local availability and inexpensive pest control agents (Mersha *et al.*, 2014; Mkenda *et al.*, 2015).

Plate 4: (A) Cabbage aphids, (*Brevicoryne brassicae*). (B) The damaged cabbage crop by cabbage Aphids. Photograph by Nelson Mpumi. NM-AIST, Arusha, Tanzania

(v) Green peach aphids *(Myzus persicae)*

The green peach aphid (*Myzus persicae*) (Plate 5A), is found throughout the world and can be present at any time throughout the year (Gu *et al.*, 2007). Generally, its colour is pale green, and there are two forms of green peach aphids; winged and wingless forms (Edwards *et al.*, 2008). The green peach aphids have prominent cornicles on the abdomen that are markedly swollen and club like in appearance (Blackman & Eastop, 2000). The frontal tubercles at the base of the antennae are very prominent and are convergent (Edwards *et al.*, 2008). Winged forms of the green peach aphid have a distinct dark patch near the tip of the abdomen; wingless forms lack this dark patch (Umina *et al.*, 2014). The green peach aphid is adapted to high environmental temperatures (Gu *et al.*, 2007).

Blackman and Eastop (2000) and Gu *et al*. (2007) showed that over 40 plant families are hosts of green peach aphids. According to them, those plants include woody and herbaceous plants including vegetable crops in the family Solanaceae, Chenopodiaceae, Compositaceae, Brassicaceae and Cucurbitaceae. Some of the host plants which support the growth and development of green peach aphids include cabbages, spinach, asparagus, bean, beets, broccoli, Brussels sprouts, carrot, cauliflower, cantaloupe, celery, corn, cucumber, fennel, kale, eggplant, lettuce, mustard, okra, parsley, parsnip, pea, pepper, potato, radish, squash, tomato, turnip, watercress and watermelon (Blackman & Eastop, 2000). Moreover, Gu *et al.* (2007) added that many flower crops and ornamental plants are also suitable for growth and development of green peach aphids. Different crops differ in their vulnerability to green peach aphids, but the actively growing plants and plants' parts, or the youngest plant tissues often are affected by large aphid populations (Umina *et al.*, 2014). Broadleaf vegetables are particularly very suitable host plants for green peach aphids. Therefore, the broadleaf vegetables create pest infestation problems in nearby crops (Gu *et al.*, 2007). The green peach aphids can achieve very high densities on young plant tissues, causing water stress, wilting and reduced growth rate of the plant (Edwards *et al.*, 2008).

Anstead *et al*. (2007) and Umina *et al.* (2014) indicated that, adults and nymphs of aphids can damage the crops in three ways:- firstly, they feed directly on young tender plant tissues and causes drying out of shoots, wilting and distortions of the plants' parts (Plate 5B); secondly, they produce honeydew which falls onto foliage and becomes blackened by sooty mould fungi; and thirdly, they spread more than 100 viruses. According to Anstead *et al*. (2007), De Little and Umina (2014), the damaging viruses transmitted by green peach aphids include potato leafroll, potyviruses in pepper, beet western yellows, beet yellows, cauliflower mosaic, cucumber mosaic, lettuce mosaic, papaya ringspot, turnip mosaic and watermelon mosaic. These viruses affect the proper growth and development of the crops and reduce the marketability. The damaging levels caused by green peach aphids are characterized by large numbers of aphids found on the underside of leaves sucking the plant saps (Anstead *et al.*, 2007; De Little & Umina, 2017). In addition to attacking plants in the field, the green peach aphid can readily infest vegetables and ornamental plants grown in glasshouses (Gu *et al.*, 2007). Umina *et al*. (2014) reported that the aphids feed by sucking sap from leaves and flower buds, but the entire crop foliage may be covered when populations are large resulting in reduced or stunted growth of young plants. The extensive feeding of green peach aphids on crops enforces plants to turn yellow and the leaves to curl downward and inward from the edges

resulting into wilting, stunted growth and finally death of the crops (De Little & Umina, 2017). When young plants are infested in glasshouses and then transplanted into the field, the fields will not only be inoculated with aphids but insecticide resistance may be introduced (Gu *et al.*, 2007). De Little and Umina (2017) insisted that the green peach aphid is considered the most important vector of plant viruses in the world. Also, contamination of harvestable plant material with aphids, or aphid honeydew, causes the loss of the food quality and quantity (Stewart *et al.*, 1980). Therefore, prolonged aphid infestation of crops can reduce the yield of crop products.

The green peach aphid is attacked by a number of common predators such as lacewings, lady beetles, syrphid flies and parasites, including the parasitic wasps (*Lysiphlebus testaceipes, Aphidius matricariae*, *Aphelinus semiflavus,* and *Diaeretiella rapae*) and is susceptible to the fungus disease, *Entomophthora spp*. All those natural enemies together with field sanitation helps to control the incidence and spread of viruses transmitted by green peach aphid, but it does little to control the aphid itself. So, the smallholder farmers rely on the application of chemical insecticides to control the green peach aphids in the field. The use of chemical pesticides to control *M. persicae* on the food crops is increasing globally (Sparks &Nauen, 2015; Umar *et al*., 2015). For instance, in African countries like Tanzania, *M. persicae* are now extensively controlled with insecticides in oilseeds, pulses, and vegetable crops (Edwards *et al.*, 2008). However, heavy reliance on insecticides to manage aphid populations result into strong insect pest resistance and *M. persicae* has probably developed resistance to more insecticides than any other insect species (Sparks & Nauen, 2015; Whalon *et al.*, 2008). Therefore, broad spectrum insect pest control strategies are needed to ensure the aphids are controlled.

The severe damage caused by insect pests in various parts of cabbage crops (Table 3; Plate 5) compel the African smallholder farmers to increase the doses of synthetic pesticides during the application.

Plate 5: (A) Green peach aphids, *Myzus persicae*. (B) The cabbage affected by green peach aphids. Photograph by Nelson Mpumi. NM-AIST, Arusha, Tanzania

Table 3: The parts of cabbage (*B. oleracea*) damaged by insect pests, signs and their effects

Insect pests	**Parts of cabbage damaged**	**Signs of the damaged crop**	**Effects on crop**	**References**
Plutella xylostella	Cabbage heads and remove foliar tissues	Stunts and destroys the cabbage heads	Reduces quality and marketability of cabbage crops	Mohan and Gujar (2003)
Hellula undalis	Leaves, petioles and heads of cabbages	Distorted of plant organ and stunted growth	Deaths to young plants and formation of unmarketable multiple heads	Weinberger and Srinivasan (2009)
Pieris brassicae	Head of cabbage and leaves	Deforming developing heads of cabbage and leaves	Interfere with plant growth and marketability of the cabbages	(Baidoo and Mochiah 2016)
Brevicoryne brassicae	Tips, flowers and leaves	Curling, distortion and yellowing of leaves, stunting growth, deforming developing heads	Stop terminal growth leading to reduced plant size and yield	Baidoo and Mochiah (2016)
Myzus persicae	Tips, flowers, developing pods and leaves	Yellowing of leaves, stunting growth, deforming developing heads and curling of leaves	Wilting, stunted growth and finally death of the crops	Baidoo and Angbanyere (2014)
Trichoplusia ni	Leaves, stems and veins of leaves	Large irregular holes of variable shapes on the leaves	Interfere with crop growth and marketability of the cabbages	Baidoo and Angbanyere (2014)

2.2 Cabbage insect pests with insecticides' resistance

Some of the important pests of cabbage (*B. oleracea*) such as the diamondback moth (*P. xylostella*), cabbage webworms (*H. undalis*), whiteflies (*Bemicia tabaci*) and aphids (*B. brassicae* and *M. persicae*) have developed resistance to a wide range of commonly used pesticides (Seif & Nyambo, 2013). For instance, *P. xylostella* is documented to have developed resistance to a number of insecticides (Vuković *et al.*, 2014). The tests done in four regions in New Zeland between 1999 and 2000 reported that *P. xylostella* developed resistance to synthetic pyrethroids (Walker *et al.*, 2001). The resistance of *P. xylostella* to pyrethroids is based on the oxidative detoxification of monooxygenase enzymes (Sun *et al.*, 1992). The level of resistance of *P. xylostella* to cypermethrin can be 1096 folds (Takahashi *et al.*, 1992). However, the resistance of *P. xylostella* to pyrethroids insecticides can be even 2880 folds (Vuković *et al.*, 2014). Verma and Sandhu (1968) reported the resistance of *P. xylostella* to DDT and parathion organochlorine insecticides in India. Also, *P. xylostella* is reported to have developed resistance to fenitrothion and malathion (Joia *et al.*, 1996), cypermethrin, decamethrin and quinalphos (Saxena *et al.*, 1989), cartap hydrochloride, diafenthiuron and flufenoxuron (Joia *et al.*, 1996; Vuković *et al.*, 2014). The major reasons for *P. xylostella* to develop resistance to insecticides includes: the increase in number of sprays, misuse of pesticides, inappropriate dosages used by farmers and frequency of applications (Gibson, 2012; Imran *et al.*, 2017). Apart from the insecticides' resistance developed by *P. xylostella*, also cabbage webworms (*H. undalis*), whiteflies (*B. tabaci*), aphids (*B. brassicae* and *M. persicae*) have developed resistance to cypermethrin, decamethrin, chlorpyrfors, malathion and lamda-cyhalothrine (Walker *et al.*, 2001). Therefore, cabbage (*B. oleracea*) insect pest resistance to synthetic pesticides calls for search of the alternative pesticidal plant products which can effectively control those insect pests in the field.

2.3 The biological life cycle of cabbage insect pests and common practices used to control

2.3.1 The biological life cycle of cabbage insect pests

Table 4 briefly presents the number of generations, the eggs per adult and the biological length of selected common insect pests of cabbage crops (*B. oleracea*). For the proper integrated management and effectively control of the common insect pests of cabbage crops (*B. oleracea*), there is a need to at least understand briefly the biological life cycle of them.

Table 4: Generation number, eggs/adult and the biological length of selected insect pests of cabbage crops

Cabbage insect pests	Generation number	Eggs/adult	Damaging stages	Length of biological cycle	References
Plutella xylostella	It completes 13–14 generations annually.	187 eggs per adult during the lifetime in *Brassica oleracea* var. Capitata	Larval stage	It requires 19.4 days to complete the life cycle	Huaripata and Sánchez (2019)
Hellula undalis	It ranges from 7–8 generations annually.	175 eggs per adult during her lifetime in *Brassica oleracea*	Larval Stage	The total time for the life cycle ranged from 22.75 days at 35 °C to 89.93 days at 20 °C and that depend on the hosts	Harakley (1969)
Trichoplusia ni	At least one generation can be completed per month successfully under favourable weather conditions.	300 to 600 eggs are produced by a female during her lifetime.	Larval stage	It requires 18 to 25 days when they are held at 32 to 21 °C, respectively to complete the life cycle.	Shorey (1963); Toba *et al.* (1973)
Brevicoryne brassicae	An average of 15 generations are completed during the crop season	A female can give birth 30 – 50 nymphs without mating and the colony will consist of females only. When mating occurs, a female can lay 5 – 7 eggs. The colony will consist of males and females	Nymphs and adults	It ranges from 16–50 depending on temperatures. It is shorter at high temperatures and long at low temperatures	Hines and Hutchison (2013); Kessing and Mau (1991)
Myzus persicae	The maximum number of generations is 20 and 21 in a year and it depends on favourable weather conditions	The oviparous female can oviposit four to thirteen eggs. The viviparous female can give birth to a mean fecundity of 75 offsprings.	Nymphs and adults	The mean length of the reproductive period is 20 days.	Van Emden *et al.* (1969)

2.3.2 Common practices used to control cabbage insect pests

This part reviews the common practices (Table 5) used by African smallholder farmers to efficiently and effectively control the insect pests infesting cabbage crops (*B. oleracea*) in the field.

Table 5: Cabbage insect pest control practices done by African smallholder farmers

Practices	Advantages	Disadvantages
Cultural practices	They are cheap and safe to the environment. Affordable by most smallholder farmers.	Those methods are effective when used in Combination with other practices
Biological practices	Have little effect to the populations of beneficial insects. Have low human toxicity and little environmental pollution problems.	Requires enough expertise, enough skills and knowledge in developing them and apply for the control of cabbage insect pests.
Chemical pesticides	Fast effective, reliable against a wide range of insect pests and easily tested for new insect pests	Causes environmental pollution, threatened human health, kills non-target organisms, and destroy the Ecosystems
Botanicals	Less persistence in the environment, harmless to non-target organisms, low mammalian toxicity, rapid in action	It is not easy to standardize the extracts, rapid degradation and affected by weather conditions

(i) Chemical pesticides

Synthetic pesticides have been used intensively for many years to control crop insect pests. Alavanja (2009) reported that about 5.6 billion pounds of synthetic pesticides are used to protect foods and commercial crops. In Africa, the predominant pesticide groups used to control the insect pests of crops and cabbages include insecticides mainly organophosphates, fungicides and herbicides (Kapeleka *et al.*, 2019; Pérez-Lucas *et al.*, 2018). The effective insecticides which are used to control cabbage insect pests worldwide and Africa particularly, include permethrin, abamectin, teflubenzuron, chlorfluazuron, triflumuron, phenthoate, exthofenprox and Lambda-cyhalothrin. Among these insecticides, abamectin was found to be the most effective (Sivapragasam & Aziz, 1992). According to Labou *et al*. (2017), carbaryl, methomyl, permethrin and trichlorfon are effective in controlling larvae of insect pests in the field. Also, Nyirenda *et al*. (2011) reported that synthetic pesticides are effective, reliable against a wide range of insect pests and act quick and easily tested for new insect pests. Moreover, some of synthetic pesticides such as DDT is used in public health programs and commercial applications, for lawn and garden applications and in around the homes (Asante &

Ntow, 2009; Williamson *et al.*, 2008) despite its toxic effect and persistence nature in the environment. Cypermethion, carbaryl and λ-cyhalothrin are used to control the pests in crops (Ntow *et al.*, 2006; Scaife & Turner, 1983). Therefore, the use of synthetic pesticides assisted to significantly reduce crop losses and improve the yield of crops such as grain crops, leafy vegetables and potatoes (Ntow *et al.*, 2006).

However, besides their beneficial effects, most of synthetic pesticides such as endosulfan, lindane and DDT have potential environmental pollution and public health impacts (Kapeleka *et al.*, 2019; Ntow *et al.*, 2006). The reports by De Bon *et al*. (2014) and Weinberger and Srinivasan (2009) indicated that many synthetic pesticides used are persistent in the environment, threaten human health, kill non-target organisms and destroy the ecosystems (Fig. 2). Moreover, in the environment, the application of synthetic pesticides commonly results into water and soil contaminations (Fig. 2), development of insect resistance to the pesticides applied and threatened the food security for human being (Chikukura *et al.*, 2011; Ntow *et al.*, 2006; Obopile *et al.*, 2008).

Apart from that, also, the availability of synthetic pesticides in distant rural areas where cabbage (*B. oleracea*) smallholder farmers are living and practice cabbage cultivation are either unreliable or are expensive. Again, synthetic pesticides are extremely diluted to ineffective concentrations by dishonest traders and they are toxic to non-target organisms (Fig. 2) (Stevenson *et al.*, 2012). When synthetic pesticides contaminate the soil, the soil ecosystems are threatened. Synthetic pesticides have high persistence in the environment (Isman, 2006) which means the pesticides can stay in the soil for many days, even years and can cause bioaccumulation and biomagnifications in the bodies of the organisms in the environment. In soil, organisms are killed and can result into biomagnification and bioaccumulation (Alavanja, 2009; Pérez-Lucas *et al.*, 2018). Biomagnification refers to increase in the concentration of a pollutant such as a pesticide or a toxic chemical in the tissues of tolerant organisms from one trophic level to another trophic level (Landrum & Fisher, 1999). This increase can occur as a result of;- first persistence of that chemical substance whereby the substance cannot be broken down by environmental processes into simple and less harmful substances, secondly food chain energetics in which the substance's concentration increases progressively as it moves up a food chain and lastly, low or non-existent rate of internal degradation or excretion of the substance which is often due to water insolubility (Landrum & Fisher, 1999). Bioaccumulation refers to the increase in the concentration of a substance in certain tissues of organisms' bodies due to

absorption of the chemical substance from food and the environment (Landrum & Fisher, 1999). The contamination of soil affects soil macro and microorganisms which are decomposers. The soil pollution affects the quality of soil chemically, biologically and physically and therefore reduces the soil fertility and productivity. Apart from that, synthetic pesticides also, affect the health of farmers during preparation, application in the farms and the consumption of cabbages. Therefore, botanical pesticides which are affordable and have health benefits to the applicators, consumers and the environment should be used for the control of insect pests of the cabbage crops (Amoabeng *et al.*, 2014).

(ii) Cultural methods

Cultural control practices refer to a broad set of management techniques which are used to minimize or eliminate insect pests by agricultural producers to achieve the crop production goals. There are several cultural practices, which African smallholder farmers use to reduce infestations of the insect pests of the crops in the field (Mpumi *et al.*, 2016). Generally, the cultural practices such as site selection, intercropping practices, crop rotation, seed selection and sowing date can minimize the invasion of insect pests in the crops (Mwanauta *et al.*, 2015). Weinberger and Srinivasan (2009) reported that, when the intercropping or trap crops are grown along with the crucifers in the same field, pest populations are kept at low. Apart from that, cabbage looper, (*Trichoplusia ni*) can be managed by crop rotation when lettuce is introduced into the garden after *Brassica oleracea* to eliminate it. Also, using clean planting materials and transplanting only healthy and vigorous insect-free seedlings, reduce the infestation of *Brassica oleracea* insect pests in the field. Moreover, uprooting and burning of any remains of cabbage and other related plant debris, protecting seedling beds and either using greenhouses with close mesh nets or screens also, reduce infestations by insect pests.

The planting time is important to be observed since proper planting time of *B. oleracea* in the field minimizes the infestation of insect pests. For instance, aphid infestation in *B. oleracea* is reduced by early sowing time (Baidoo & Adam, 2012). Therefore, sowing time can affect the population of aphids and other arthropods attacking cabbages. Moreover, Weinberger and Srinivasan (2009) reported that mechanical means such as weeding, or natural methods can be used to control insect pests of cabbages in the field. In other agronomic studies, row spacing and plant density and weed control are used to control insect pests of cabbage production in the field. Moreover, removing weeds aids to control aphids. Handpicking, minimizes large pests such as slugs, leatherjackets or caterpillars. However, this method is quite efficient

especially in a small garden. Apart from that, cultural control methods are friendly to the environment and to the health of human beings, less costly in terms of money and time, minimizes chances for biotype selection and also not harmful to non-target organisms. However, most of the cultural practices alone are not effective enough to protect cabbage insect pests in the field although they are cheap and safe to the environment (Gyanoba, 2018).

(iii) Biological control

The term "biological control" has been used long time ago to describe either the use of live predatory insects, entomopathogenic nematodes, microbial insecticides and natural enemies or the use of the natural products extracted or fermented from numerous sources to suppress populations of different insect pests (Pal & Gardener, 2006). In cabbage production, biological control is involved in the control of cabbage insect pests. For example, microbial insecticides are involved in cabbage looper management and their potential role has so far been fully realized. Gupta and Dikshit (2010) reported that, the most widely known microbial pesticides are varieties of the bacterium *Bacillus thuringiensis* (Bt) which control certain insects in cabbage, potatoes and other crops. *Bacillus thuringiensis* has been used for a long time to effectively suppress the cabbage looper and has little effect on the populations of beneficial insects (Pal &Gardener, 2006). *Bacillus thuringiensis* produces a protein that is harmful to specific insect pests of cabbage like diamondback moth and cabbage looper which when the protein is ingested by either pest or pest larvae, the midgut of the pest is damaged, eventually killing it (Gupta & Dikshit, 2010). Generally, *B. thuringiensis*, controls certain caterpillars, beetles and flies (Kareru *et al.*, 2013) and has low human toxicity and little environmental pollution problems.

The natural enemies such as predators, parasitoids and pathogens are involved in the management of cabbage insect pests (Mpumi *et al.*, 2016). The predators such as spiders, lacewings, lady beetles, ground beetles, rove beetles, hover flies, and true bugs (Flint *et al.*, 1998; Mkenda *et al.*, 2015) attack, kill and feed on insect pests affecting the production of crops. Also, those organisms can kill and feed on the insect pests which affect the production of leafy vegetables in the field. Ladybird beetles, family *Coccinelidea*, both adults and larvae feed on aphids (Chapman *et al.*, 1981) and as a consequence reduce the populations of aphids in the cabbage field. Ladybird beetles are stronger, larger and normally are more intelligent than the prey (Mpumi *et al.*, 2016) and hence attack several hosts in a short period of time. Parasitoids such as many species of wasps and some flies parasitize and kill other invertebrates

(Mpumi *et al.*, 2016). Some of species of parasitoids, when they are in immature stage, develop on or within a single insect host forming mummies and finally kill the host (Mahr and Ridgway, 1993). Parasitoids are parasitic when they are in immature stage and kill their hosts as they reach maturity (Flint *et al.*, 1998). Biological control is safe and eco-friendly and therefore, more study is required for insect pests' control in cabbages (Tembo *et al.*, 2018). Despite their safety, lower environmental and low toxicity effect of biological control methods, African smallholder farmers have little knowledge and understanding on the application of biological control method for cabbage insect pest management and therefore rely much on the use of synthetic pesticides to control cabbage insect pests. Relying on application of synthetic pesticides for cabbage insect pests control leads to environmental pollution. Therefore, application of safe, environmentally friendly botanicals from pesticidal plants for cabbage insect pests' control should be employed to minimize the environmental pollution from synthetic pesticides (Fig. 1).

(iv) Botanical pesticides

Botanical pesticides have been used as alternatives to synthetic insecticides to control cereal crop insect pests in the field and in the storage because they pose little threat to the environment, to ecosystems and to human health (Amoabeng *et al.*, 2014; Isman, 2006; Secoy & Smith, 1983). In the middle of the 17th century, pyrethrum, nicotine and rotenone were recognized as effective insect control agents (Arannilewa *et al.*, 2006; Henn & Weinzierl, 1989; Isman, 2006). In fact, Arannilewa *et al*. (2006) revealed that many plants with medicinal properties demonstrated potential insect pests control agents. Those plants with medicinal properties comprise numerous active chemicals, which affects the reproductive and digestive process of a number of important pests (Gupta & Dikshit, 2010; Isman, 2006) (Table 6). Le Roy and Wrana (2005) reported that the plants with active chemical compounds of medicinal and pesticidal properties are botanicals. Botanical insecticides like tobacco extracts, neem oil extracts have been found useful for pest control in cereal crops. In addition, the plants like *T. vogelii, Azadirachta indica, Annona squamosa, Capsicum frutescens, Allium sativum* have potentials for controlling cereal crop insect pests (Koona & Dorn, 2005), *Aristolochia ringens* and *A. sativum* displayed antifeedants, contact poisons and repellents properties against *Sitophilus zeamais* (Arannilewa *et al.*, 2006) and *Nicotiana tabacum, Azadirachta indica, Eucalyptus camaldulemsis* and *Swietenia mehagani,* indicated effectiveness against aphids (Arannilewa *et al.*, 2006).

Therefore, those potentials of botanical pesticides as mentioned above can be employed in controlling cabbage insect pests in the field. The mixture of bioactive compounds in plants have potential advantages in terms of efficacy and short life span development of resistance (Kareru *et al.*, 2013; Sola *et al.*, 2014). Those chemicals have little mammalian toxicity, degrade rapidly in sunlight, air, and moisture and therefore, are less persistence in the environment and are rapid in action to the insect pests (Amoabeng *et al.*, 2013; Henn & Weinzierl, 1989; Stevenson *et al.*, 2015). Moreover, botanicals have little effects to non-target organisms and natural enemies of insect pests (Fig. 1), have little or no toxic effect on plant growth and cooking quality of the edible part of the crop and also, are less expensive and easily available in the farmers' natural environment (Fig. 1) (Amoabeng *et al.*, 2014; Mkindi *et al.*, 2017). However, botanical pesticides are extensively used for the protection of cereal crops from insect pests in the field and storage and there is very little information available on the use of botanical pesticides to control cabbage insect pests in the field.

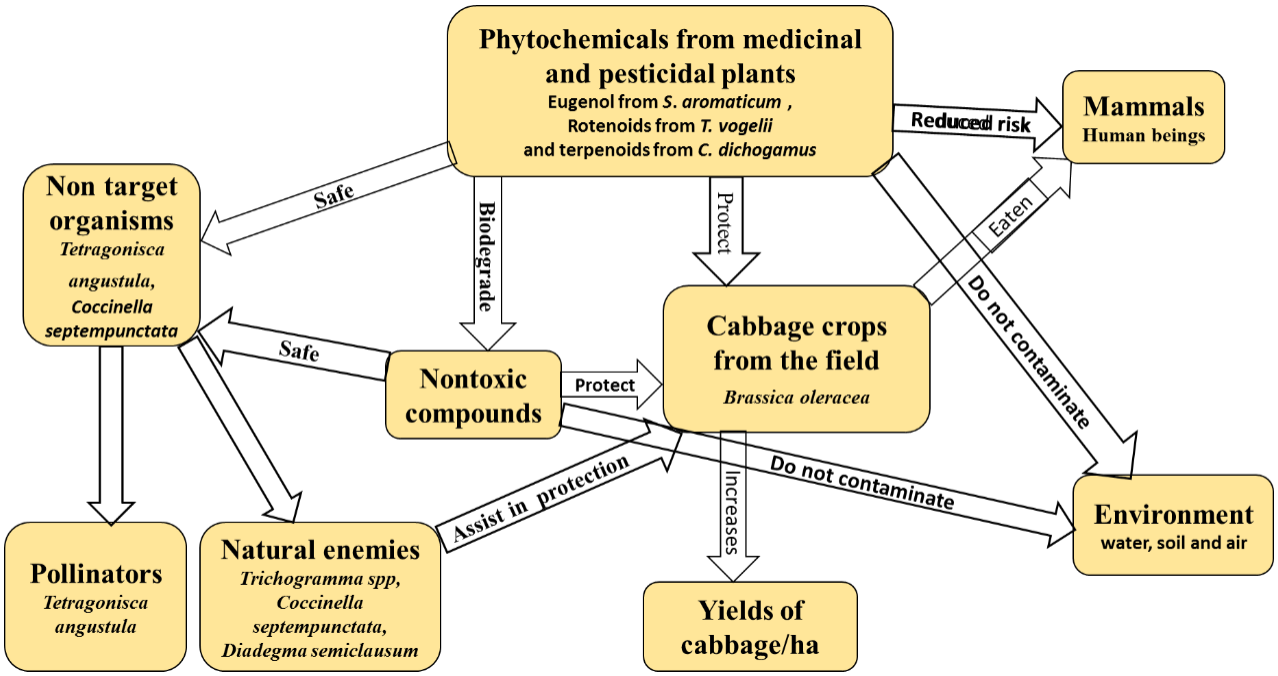


Figure 1: Summary of the advantages of botanicals/phytochemicals when applied on cabbage crops

Table 6: Some of the pesticidal plants reported to control cabbage insect pests in Africa

Chemical and Pesticidal Plant	Insect Pests Controlled	The Area of Study	References
Garlic (*Allium sativum* L.) and Hot Pepper (*Capsicum frutescens* L.)	*Brevicoryne brassicae* (L.), *Plutella xylostella* (L.), *Helula undalis* (Fab.) and *Trichoplusia ni* (Hub)	In a greenhouse nursery	Baidoo and Mochiah (2016); Fening *et al.* (2013)
Lantana camara (L.) and *Azadirachta indica* (A. Juss)	*Plutella xylostella*, *Brevicoryne brassicae* and *Hellula undalis*	In a greenhouse nursery	Baidoo and Adam (2012)
Ageratum conyzoides, Chromolaena odorata Synedrella nodiflora, Capsicum frutescens, Nicotiana tabacum Cassia sophera, Jatropha curcas, Ricinus communis and *Ocimum gratissimum*	Cabbage aphids (*Brevicoryne brassicae*) and diamondback moth (*Plutella xylostella*)	It was a field cage experiment	Amoabeng *et al.* (2013)
Lantana camara (L.), *Azadirachta indica* (A. Juss), *Capsicum annuum* (L.) and *Curcuma longa* (L.)	Diamondback Moth, *Plutella xylostella* L. (Lepidoptera: Plutellidae)	It was conducted at the field	Begna and Damtew (2015)
Plant extract Neem azal - S	*Brevicoryne brassicae* and *Bemesia tabaci*	It was conducted at the field	Zaki (2008)
Tephrosia vogelii, *Allium sativum* and *Solanum incanum*	*Brevicoryne brassicae* in *Brassica napus* done in greenhouse nursery	It was conducted in the greenhouse nursery	Mudzingwa *et al.* (2013)

2.4 The fate of pesticides used to control cabbage insect pests by African smallholder farmers

Generally, pest control in cabbage by smallholder farmers is still heavily dependent on synthetic insecticides although their use is associated with many undesirables and sometimes lethal consequences (Ayalew, 2006). The herbicides such as triazines (atrazine, simazine, terbuthylazine, propazine, cyanazine, terbutryn, prometryn), phenylureas (diuron, linuron, isoproturon, chlortoluron) and anilides (alachlor, acetochlor, metolachlor), insecticides such as organophosphorus (malathion, dimethoate, parathion-methyl, azinphos-ethyl, chlorpyrifos, fenitrothion) and organochlorine (lindane and DDTs) (Kapeleka *et al.*, 2019) and some of their metabolites are the most common pesticides found in the soil, the surface and groundwater bodies (Pérez-Lucas *et al.*, 2018). The pollution of the groundwater and surface water by unwise use of pesticides in agriculture threatened the soil organisms and their ecosystems (Fig. 6) and drinking water resources (Labou *et al.*, 2017; Pérez-Lucas *et al.*, 2018). Moreover, excessive use of insecticides also induces resistance development in target pests as well as

killing beneficial organisms such as pollinators for example bees and natural enemies in the field (Mkenda *et al.*, 2015; Waiganjo *et al.*, 2011).

Due to the severe infestation of cabbage caused by insect pests, most of smallholder farmers in African countries decide to effectively suppress and kill the insect pests of cabbages in the field through the following ways. Firstly, they increase the concentration of the synthetic pesticides during the application in the cabbage field (Ntow *et al.*, 2006). That means, the smallholder farmers use synthetic pesticides beyond the recommended amount by the manufacturers which result into extreme soil pollution.

Secondly, they increase the rate of application of synthetic pesticides to the field. Sometimes pesticide is applied twice in a week to strongly and effectively kill the very stubborn insect pests of cabbages (Orr and Ritchie, 2004). For instance, Ngowi *et al*. (2007) and Ntow *et al.* (2006) reported that, 5 to 16 times pesticide applications per crop is practiced for the whole growing season in African countries, with onion being the most treated crop, followed by tomatoes and cabbages being the last and the frequency on a weekly basis application in many situations. For instance, Orr and Ritchie (2004) reported that, the farmers spray on average of 19 times in tomatoes and 14 times in cabbages in Malawi throughout the growing season. Also, Ahouangninou *et al*. (2011) revealed that 70% of vegetable growers in Southern Benin apply four to five times chemical treatments per month while doubling or tripling the recommended dosage. According to De Bon *et al*. (2014), the smallholder farmers believe that the frequency of pesticides applications certainly prevent the insect pests attack effectively. The improper and overuse of synthetic pesticides magnify water and soil pollution which finally threatened the water and soil ecosystems (Pérez-Lucas *et al.*, 2018).

Thirdly, they mix more than two synthetic pesticides at the same time in order to increase the spectrum of destroying and killing various insect pests in the field. In so doing, the environment is threatened (Pérez-Lucas *et al.*, 2018). Specifically, water bodies and soil pollution occur due to intensive application of synthetic pesticides without considering the recommendation of the manufacturers. The synthetic pesticides kill the organisms in the environment indiscriminately which imply that, both beneficial and harmful organisms are killed indiscriminately (Ambrósio *et al.*, 2008). The decomposers, fishes and other organisms in water for example are usually affected by extreme application of synthetic insecticides meaning that, the decomposition of organic matter to release nutrients into the soil is affected. As a result, the soil becomes infertile and less productive. In water bodies, aquatic organisms such as fishes, anglerfishes, sponges,

shrimps, phytoplankton and zooplanktons are killed and can result into biomagnification and bioaccumulation (Ambrósio *et al.*, 2008; Amoabeng *et al.*, 2013). The increase of concentration of toxic substances along the food chain can occur as a result of first, persistence of that chemical substance in the environmental media whereby the substance cannot be broken down by environmental processes into simple and less harmful substances; secondly the food chain energetics in which the substance's concentration increases progressively as it moves up a food chain, and; lastly, low or non-existent rate of internal degradation or excretion of the substance which is often due to water insolubility (Landrum & Fisher, 1999).

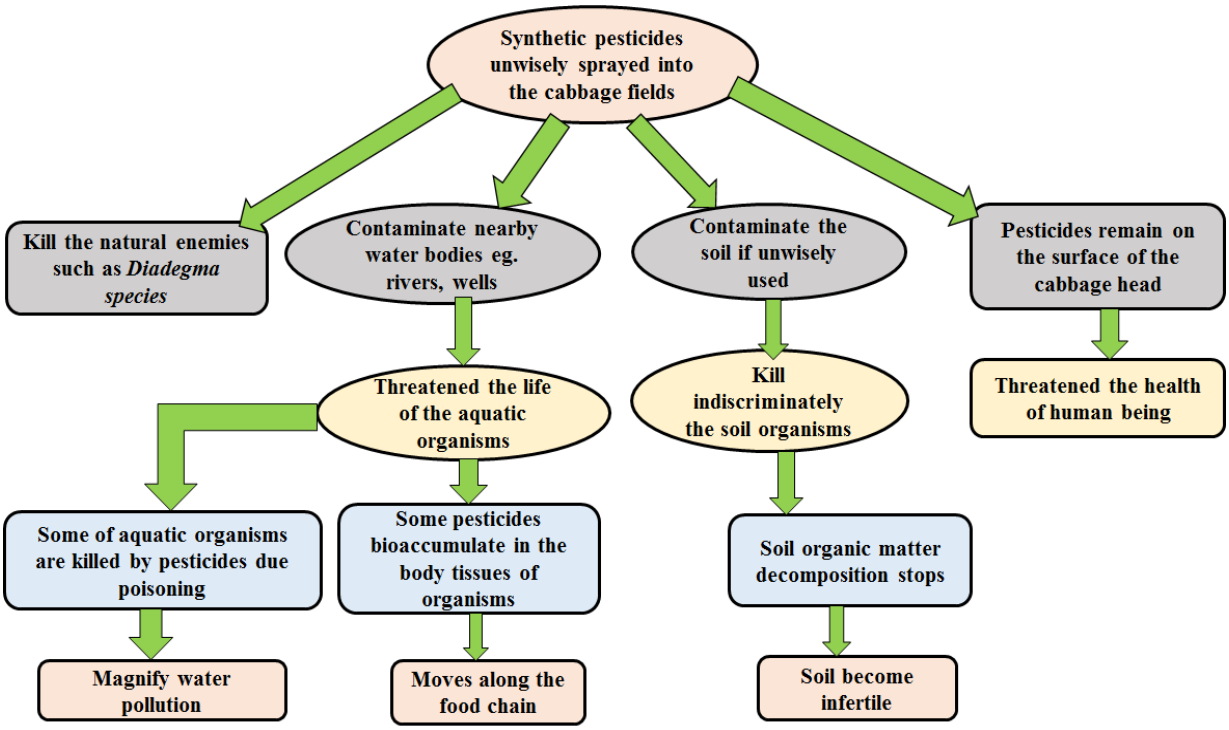


Figure 2: Summary of the fate of synthetic pesticides when applied heavily on *Brassica oleracea*

2.5 Effectiveness of botanical pesticides

Many studies reported the effectiveness of various pesticidal plants for controlling cereal crop insect pests in the field and in storage. For instance, *Alstonia boonei* and *Eugenia aromatic* have reported to reduce the infestation by cowpea beetles (Ileke & Oni, 2011). *Capsicum frutescens, C. annum* fruit and *Citrus sinensis* peel have shown mortality of adult *Dasyses rugosella* in yam tuber (Ashamo, 2010). The effectiveness of plant material for the pesticidal action depends on the time of exposure and the concentration of the extracts and the powder (Ileke & Oni, 2011). However, most of information available about the effectiveness of botanical pesticides are on the protection of cereal crops from insect pests in the field and

during storage. Therefore, this study assessed the insecticidal actions of *T. vogelii*, *C. dichogamus* and *S. aromaticum* to control cabbage insect pests in the northern part of Tanzania.

2.5.1 Clove (*S. aromaticum*)

Cloves are the aromatic flower buds of a tree in the family Myrtaceae, *S. aromaticum.* A major component of clove taste is imparted by the chemical eugenol (Araujo *et al.,* 2016). Because of the bioactive chemicals of clove, the spice nature may be used as a repellent. *Syzygium aromaticum* has a range of pharmacological activities which includes antimicrobial, anti-inflammatory, analgesic, anti-oxidant and anticancer activities, amongst others (Kamatou *et al.,* 2012). In addition, it is used in agricultural applications to protect foods from micro-organisms during storage, which might have an effect on human health and as a pesticide and fumigant (Tian *et al.*, 2015). The essential oils from *S. aromaticum* exhibited larvicidal activity against resistant populations of *Aedes aegypti* (Araujo *et al.*, 2016). However, there is limited information available on the applications of *S. aromaticum* for the control of cabbage insect pests in the field.

2.5.2 *Tephrosia vogelii*

Tephrosi vogelii is either herbs or small tree that is native to tropical Africa and has also been used in tropical America as well as South and Southeast Asia (Orwa *et al.*, 2009). It can attain a height of 2 to 3 m in a growing season of 5 to 7 months (Mkindi *et al.*, 2019). *Tephrosi vogelii* is commonly known as the "fish bean", "fish-poison bean", or "vogel's tephrosia" (Dougnon *et al.*, 2014). It is used by farmers in numerous countries in Africa to get rid of pests on livestock, control pests in cultivated fields as an organic pesticide, improves soil fertility, medicine for skin diseases and internal worms and for storage of crops (Munthali *et al.*, 2014). On livestock keeping, *T. vogelii* has activity against ticks (*Amblyomma variegatum)* in which the leaf extracts of the plants are sprayed directly on the tick infested parts of the bulls and showed 98.5% effectiveness (Dougnon *et al.*, 2014). It contains chemical compounds such as glycosides, rotenoids, flavones, chalcones, flavanones, flavanols, and prenylated flavonoids (Chen *et al.*, 2014). Moreover, according to Belmain *et al*. (2012), HPLC-UV analysis revealed that, *T. vogelii* contains chemotype 1 and chemotype 2 phytoconstituents. Their study reported that, chemotype 1 was characterized by the presence of rotenoids including rotenone (1), tephrosin (2), deguelin (3), α-toxicarol (4), and sarcolobine (5). Rotenone, has a molecular formula of $C_{23}H_{22}O_6$, with a melting point of 165 °C (Mkindi *et al.*, 2019). It is an odorless, colorless, crystalline isoflavone used as a broad-spectrum insecticide, piscicide and pesticide

(El-Wakeil, 2013). Deguelin is a derivative of rotenone with an empirical formula of $C_{23}H_{22}O_6$, crystal and melting point is 171 °C (Dzenda *et al.*, 2007). Tephrosin, a nearly colorless rotenoid with a formula of $C_{23}H_{22}O_7$ and melting point of 198 °C and it is thought to be the oxidation product of deguelin (Murillo *et al.*, 2002). The content of rotenoids in the leaves of *T. vogelii* is higher than that in petals, stems and roots, accounting for 80% to 90% of the total rotenoids (Delfel *et al.*, 1970) meaning that, the leaves of *T. vogelii* are potential for extracting the chemical components for insecticidal activity. Similarly, chemotype 2 was characterized by the absence of rotenoids and the presence of prenylated flavanones including, obovatin-5-methylether (6) and other 6 currently reported flavanones and the flavone tephrostachin (Stevenson *et al.*, 2012) (Fig. 3). However, there is limited information about the efficacy and usefulness of the chemical compounds from *T. vogelii* to control insect pests of cabbage in the field.

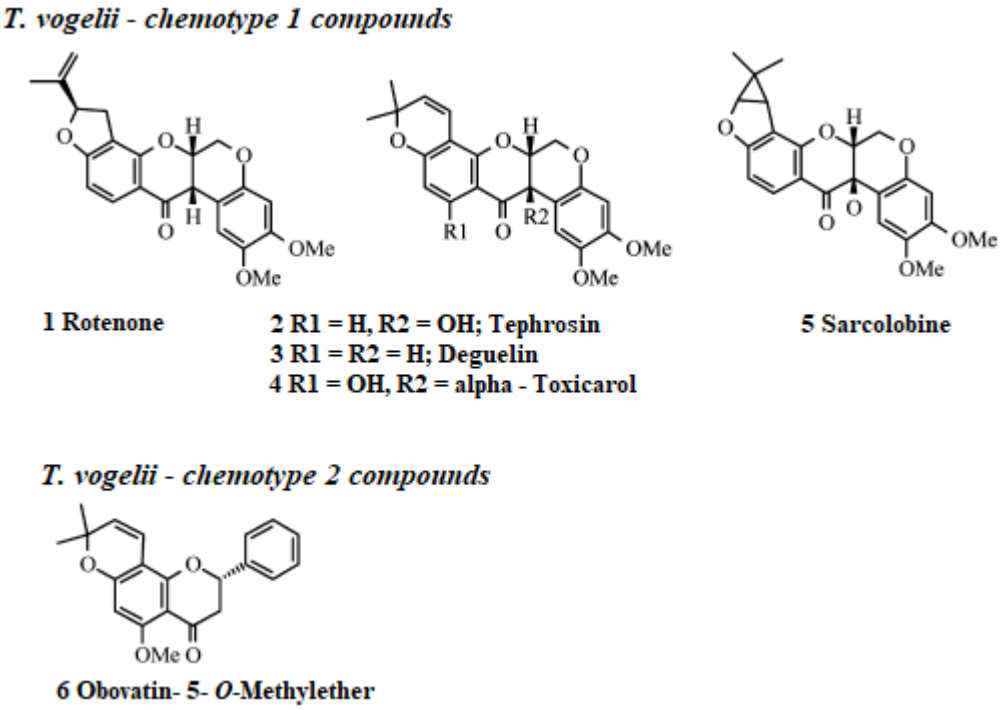


Figure 3: Chemical compound structures identified in *T. vogelii* chemotypes 1 and 2. (Belmain *et al.*, 2012)

2.5.3 *Croton dichogamus*

The genus *Croton* is among the largest group of the Euphorbiaceae family having more than 1200 species occurring in the tropics and subtropics worldwide (Silva *et al*., 2018). *Croton species* are commonly distributed in tropics and subtropics (Xu *et al.*, 2018). *Croton dichogamus* is among the *Croton species* which is widely distributed in Ethiopia, Kenya,

Madagascar, Mozambique, Tanzania and Somalia (Salatino *et al*., 2007). In the northern zone of Tanzania *C. dichogamus* is widely distributed around the roads and mountains. In Africa, America and Asia *Croton species* are commonly used as folk medicines in the treatment of cancer, constipation, diabetes, digestive problems, dysentery, external wounds, fever, hypercholesterolemia, hypertension, inflammation, intestinal worms, malaria, pain, ulcers and weight-loss (Silva *et al*., 2018). *Croton dichogamus* is used as a dietary additive to milk or meat soup by the Maasai and Batemi of Kenya and Tanzania (Salatino *et al*., 2007). The phytochemical studies of the *Croton species* have revealed the presence of alkaloids, phenolics, terpenoids including monoterpenes, sesquiterpenes and diterpenes in all plant parts (Aldhaher *et al*., 2017; Silva *et al*., 2018). These compounds deter insects (Saviranta *et al.*, 2010). But the chemical compounds in plants may differ in types and concentrations due variation of vegetative state, the genetic, growing season and the places of origin (Fu *et al.*, 2007; Samarasekera *et al.*, 2008; Srivastava *et al.*, 2005). However, there is limited information about the efficacy and insecticidal effects of the chemical compounds from *C. dichogmus* to control insect pests of cabbage in the field.

2.6 Classes of botanicals in pesticidal plants and their potentialities

Plants possess plenty of secondary metabolites which prevent herbivores and pathogens from attacking them (Mazid *et al.*, 2011). Those secondary metabolites are called phytochemicals or botanicals (Geyid *et al.*, 2005). Phytochemicals are chemical substances which are derived from plants (Mehta *et al.*, 2015). The secondary metabolic compounds are responsible for protecting the plant against insect herbivore infestation or microbial infections (Mazid *et al.*, 2011). The phytochemicals are located in various parts of plants such as leaves, stems, roots, barks, rhizomes, flowers, fruits, grains, buds or seeds (Bhatt *et al.*, 2014; Sasidharan *et al.*, 2011). The main classes of botanicals which can be extracted from plants include alkaloids, flavonoids, terpenoids, glycosides (Cushnie & Lamb, 2005). Others include phenolics, saponins and tannins (Ramawat & Mérillon, 2013), essential oils and steroids (Patra, 2012; Wangchuk *et al.*, 2011)

2.6.1 Alkaloids

Alkaloids are nitrogen-containing compounds and they are the largest group of secondary metabolites (Fig. 4) (Gul & Hamann, 2005). Despite the chemical similarity, the structures and functions of alkaloids vary so widely (McNaught & Wilkinson, 1997). Discovery and isolation

of alkaloids revealed about 25 000 alkaloids which are as stipulated by the dictionary of natural products (Keasling, 2008). Alkaloids are readily soluble in alcohol; however, in water are sparingly soluble. They are basic with alkaline reactions due to one or more nitrogen atoms (Fig. 4). The major groups of alkaloids are as follows; Tropane (or Pyrrolidine), Isoquinoline, Pyridine, Pyrrolizidine, Quinoline, Indole, Purine (Gul & Hamann, 2005). The bitterness of alkaloids has a potentiality for use as good natural substances for protection of plants against insect pests and pathogens in crops. Furocoumarin and quinolone alkaloids extracted from *Ruta chalepensis* leaves showed larvicidal and antifeedant activities against *Spodoptera littoralis* larvae (Emam *et al.*, 2009). Moreover, alkaloids extracted from *Pergularia tomentosa* exhibited antifeedant and larvicidal effects into various larvae of insect pests (Acheuk & Doumandji-Mitiche, 2013). Alkaloids interfere with the nervous system especially in the chemical transmitter and membrane transport (Ndakidemi & Dakora, 2003). Alkaloids also protect plant seeds against pathogen attack (Acheuk & Doumandji-Mitiche, 2013). Therefore, these antifeedant effects, larvicidal effects and bitterness are potential properties in management and control of insect pests in leafy green vegetables.

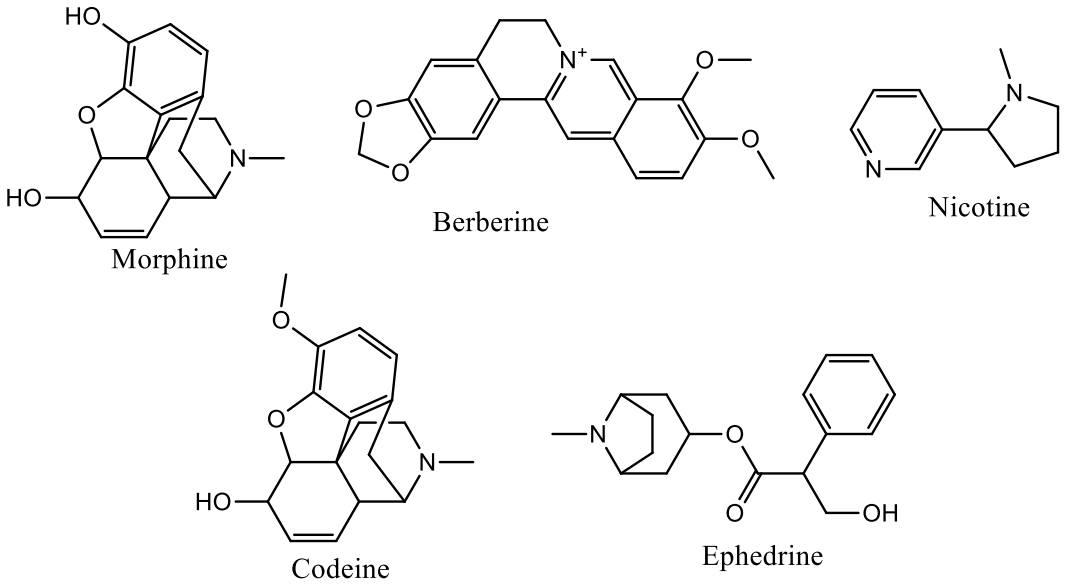


Figure 4: Structures of some plant-derived alkaloids (Alamgir, 2018)

2.6.2 Flavonoids

The name flavonoid comes from the latin word *flavus* meaning yellow color in nature. Those are classes of plants' and fungus secondary metabolites. Flavonoids are polyphenolic secondary metabolites of plants (Sonneberg *et al.*, 2013; Venkateswara Rao *et al.*, 2017) which contain 15 carbon atoms and are soluble in water (Fig. 5). Flavonoids consist of two benzene

rings connected by a short three carbon chain (Fig. 5) (McNaught & Wilkinson, 1997). One of the carbons in the chain is connected to a carbon in the benzene rings, either through an oxygen bridge or directly, which gives a third middle ring (Fig. 5). Flavonoids are widely distributed in plants and perform many functions (Makoi *et al.*, 2010; Ndakidemi & Dakora, 2003). The flavonoids can be divided into six major groups including chalcones, flavones, isoflavonoids, flavanones, anthoxanthins and anthocyanins (Ballard & Junior, 2019; Lorent *et al.*, 2015). Some of the flavonoids can be used in a pest-management approach (Acheuk & Doumandji-Mitiche, 2013). Many flavonoids are involved in defense against herbivory insects and mammalian herbivory. For instance, isoflavones, flavones and flavanones are recognized as antifungal plant agents (Makoi *et al.*, 2010; Mierziak *et al.*, 2014). Flavonoids such as anthocyanin chemicals from the seeds of cowpea, marama bean and Bambara groundnut are deterrents to attack by insect pests and pathogens (Ndakidemi & Dakora, 2003). The seeds of *Mucuna* spp. for example, is very rich in L-dopa (3,4-dihydroxyphenyl), which is toxic to pathogens and protect seeds against pathogenic pests (Rajaram & Janardhanan, 1991).

Moreover, Makoi *et al*. (2010) reported that flavonoids glycosides isolated from *Tephrosia purpuria* inhibited insecticidal properties on *C. maculatus grubs*. Flavonoids play an important role in the protection of plants against plant feeding insects and herbivores (Harborne & Williams, 2000). Their presence can alter the delectableness of the plants and reduce their nutritive value, decrease digestibility or even act as toxins (Mierziak, 2014). In rice, three flavone glucosides which inhibit digestion in insects can function as deterrent agents towards *Nilaparvata lugens* (Harborne & Williams, 2000). Isoflavonoids and proanthocyanidins are responsible for plant protection against insects (Mierziak, 2014). For instance, naringenin procyanidin reduce the development of *Aphis craccivora* (Harborne & Williams, 2000). Flavonoids such as kaempferol, quercetin and myricetin (flavonols) act as deterrents against *Radopholus similis* and *Meloidogyne incognita*, while genistein and daidzein (isoflavones) are active against *Radopholus similis* (Mierziak, 2014). Flavonoids can also prevent insects from laying eggs, for instance, quercetin-3-*O*-rutinoside acts as a stimulant to *Danaus plexippus*, but as a deterrent to *Pieris rapae* (War *et al*., 2012). Flavonoids, like flavones, flavanone and isoflavonoids also play important role as feeding deterrents (Mierziak, 2014). Flavonoids are cytotoxic and interact with different enzymes through complexation (Mierziak, 2014).

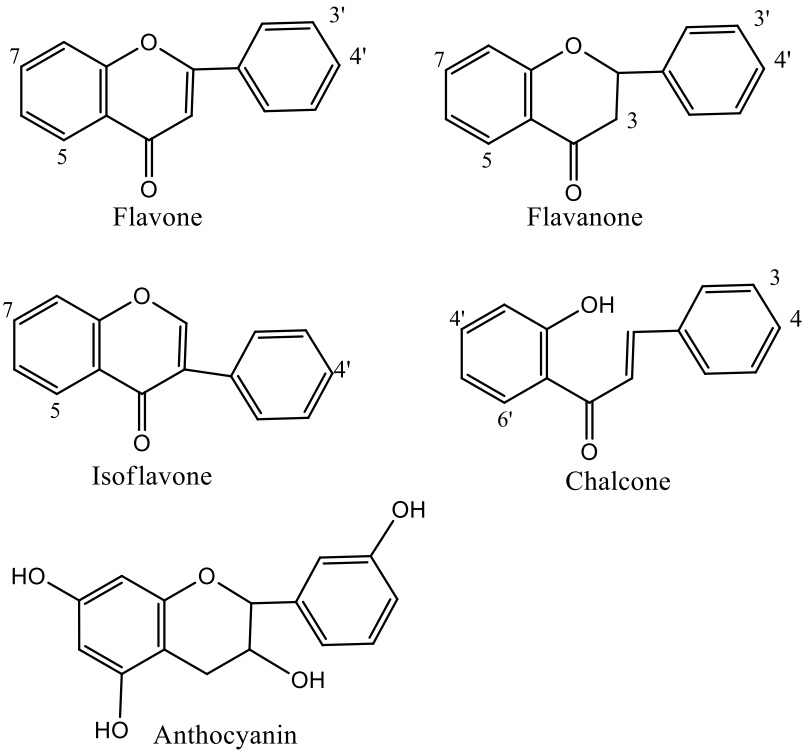


Figure 5: Basic structures of common flavonoid compounds (Ndakidemi & Dakora, 2003)

2.6.3 Terpenoids

The terpenoids are also called isoprenoids or terpenes. Terpenoids are formed by joining together the isoprene unit represented by five carbon atoms (Cho *et al.*, 2017; Kandi *et al.*, 2015). The five-carbon isoprene units assembled and modified in thousands of ways forming a vast number of compounds, the terpenoids (Fig. 6) (Aharoni *et al.*, 2005; Kandi *et al.*, 2015). Most are multicyclic structures of compounds which differ from one another in their functional groups and the basic carbon skeletons. Terpenes are classified into hemiterpene (5C), monoterpenes (10C), sesquiterpenes (15C), diterpenes (20), sesterterpenes (25C), triterpenes (30C), tetraterpenes (40C) according to the number of carbon atoms present in them (Reynolds & Enriquez, 2016). Around 60% of natural products are known as terpenoids. The presence of terpenes in plants gives them an aromatic odour and recognized to be extremely distributed in spices such as *S. aromaticum, Cuminum cyminum, Zingiber officinale, Coriandrum sativum, A. sativum, Elettaria cardamomum, Curcuma longa*. The most commonly known terpenoids include citral, camphor, menthol, and salvinorin A in the plant *Salvia divinorum*, the cannabinoids in cannabis, ginkgolides and bilobalide in *Ginkgo biloba*, and the curcuminoids.

Due to aromatic characteristics of terpenoids, they are broadly used for traditional herbal remedies and in pest management approaches (Bhatt *et al.*, 2014). For instance, monoterpenes,

citral, citronellal and L-carvone exhibited a potent fungicidal activity against phytopathogenic fungi (Garcia *et al.*, 2008). According to Garcia *et al*. (2008) citral at a concentration of 0.6% or above, completely prevented mycelium growth of *C. musae* and *C. gloeosporioides*. Comparing with the monoterpenes tested, concentrations of 1% of citronellal and L-carvone completely inhibited mycelium growth of *C. musae and C. gloeosporioides* and showed an inhibition of approximately 80% of Fungal growth. Moreover, terpenoids like pyrethrins which occur in the leaves and flowers of pyrethrum show strong neurotoxins against beetles, wasps, moths and other insect pests (Mazid *et al.*, 2011). Sesquiterpenes are reported for their role of feeding repellence to many insect pests (Tak *et al*., 2016). Volatile terpenoids are used as repellents, by preventing or decreasing plant-insect contact and transmission of (viral) disease. Moreover, Khanh *et al*. (2007) reported that terpenoids are used as insect pests and infection control chemicals. Generally, terpenoids act against fungal infections in sorghum, tomatoes, potatoes, wheat, barley, and millet crops (Ribera & Zuñiga, 2012). Thus, these compounds in plants and in plants' parts can resist pathogens and insect pests' attack.

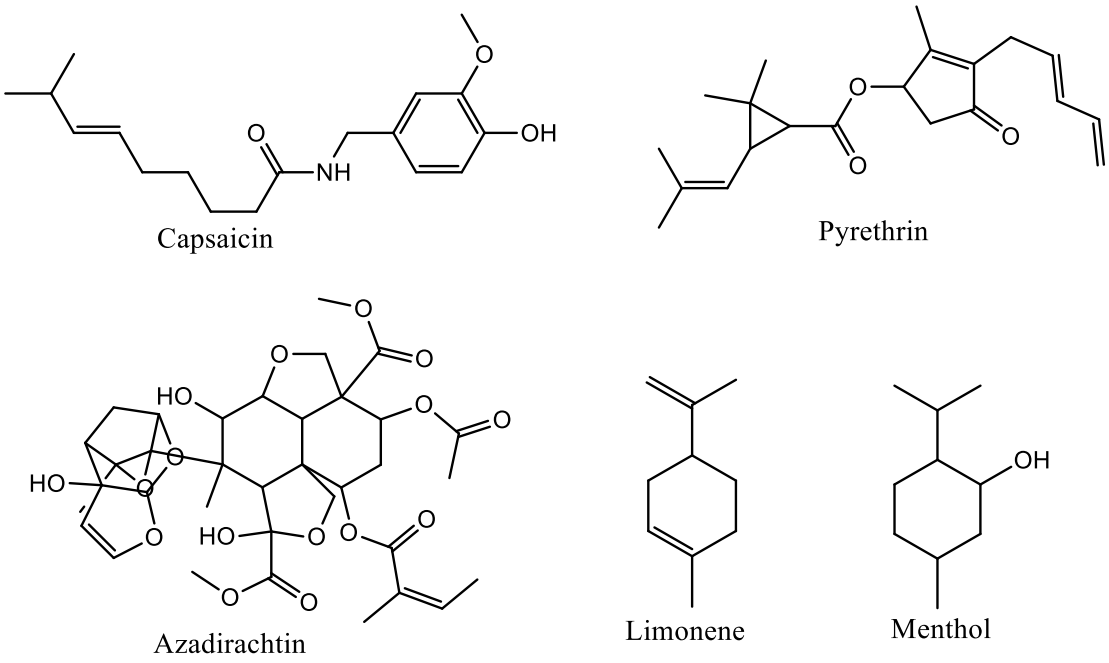


Figure 6: Structures of some plant-derived terpenes (Ndakidemi & Dakora, 2003)

2.6.4 Essential oils

Essential oils are compounds which are composed of mostly terpenes and phenylpropanoids (El Asbahani *et al.*, 2015). Essential oils are also known as volatile oils, ethereal oils, aetheroleum, or simply as the oil extracted from the plant, such as oil of clove (Lee *et al.*,

2012). The essential oils may be found in the roots, stems, leaves, flowers, or fruits of the plants (El Asbahani *et al.*, 2015). The most important characteristic of essential oils, which also gives the special economic value, is the specific smell "fragrance" (Butnariu & Sarac, 2018). This characteristic is potential for their use in perfumery, cosmetics and the food industry. Many essential oils have special therapeutic qualities, for instance have been known and used since ancient times (Butnariu & Sarac, 2018). Essential oils are often used for aromatherapy, a form of alternative medicine for healing purposes (Posadzki *et al.*, 2012). Research has shown that essential oils have potential use as natural pesticides (Butnariu & Sarac, 2018). Studies of certain oils have been shown to have a variety of deterring effects on pests, specifically insects and arthropods (Nerio *et al.*, 2010). These effects may include repelling, inhibiting digestion, stunting growth (Regnault-Roger *et al.*, 2012) decreasing rate of reproduction, or death of pests which consume the oil or inhale the smell of the oils. However, the molecules within the oils that cause these effects are usually non-toxic for mammals (De Toledo *et al.*, 2016).

Therefore, extraction of essential oils as insecticides from aromatic plants have increased progressively, due to their insecticidal properties and used by organic growers (Do Ngoc Dai *et al.*, 2015; Righi *et al.*, 2018). The essential oils have repellent, insecticidal, antifeedants, growth inhibitors, oviposition inhibitors, ovicides and growth-regulator effects on various insect pests (Regnault-Roger *et al.*, 2012). The organic growers extract and use organic essential oils from pesticidal plants because they are less toxic to human beings and friendly to the environment. De Toledo *et al*. (2016) revealed that essential oils of various plants exhibited insecticidal activities against the larvae of moth insect pests. For instance, the essential oils extracted from *Mentha piperita* repels ants, flies, lice, moths and is effective against *Callosobruchus maculatus* and *Tribolium castaneum* (Kordali *et al.*, 2005). Moreover, essential oils from *Zingiber officinale* rhizomes exhibited insecticidal and antifeeding activities against *T. castaneum* and *Sitophilus oryzae* (Chaubey, 2012). *Melaleuca alternifolia* essential oils possess the fumigant toxicity against *S. zeamais* (Yang *et al.*, 2020). Also, *Laurus nobilis* essential oil exhibited toxicities when tested against *Rhyzopertha dominica* and *T. castaneum* (Chaubey, 2019). *Rosmarinus officinalis* and *Eucalyptus globulus* essential oils showed insecticidal activities against *Acanthoscelides obtectus* adults (Papachristos *et al.*, 2004). The pesticidal and antifeedant activities of eucalyptus oils are due to the presence of 1,8-cineole, citronellal, citronellol, citronellyl acetate, p-cymene, limonene, linalool, α-pinene, ϒ-terpinene, α-terpineol, alloocimene, and aromadendrene components (Hikal *et al.*, 2017). Hikal *et al*. (2017) concluded that, the fumigant toxicity/repellent activity of essential oils from *Eucalyptus*

cinerea, *Eucalyptus viminalis*, and *Eucalyptus saligna*, against permethrin-resistant human head lice. Therefore, these examples indicate the potentialities of the essential oils against various insect pests.

2.7 Synergistic effects of botanicals

The mixture of chemicals in botanical pesticides have synergistic effects (Tak & Isman, 2017a). Synergistic effect occurs when the mixture of two or more chemical compounds interacts and produces combined effects on the biological system which is greater than the algebraic sum of the effects of those chemical compounds when they act individually. Normally, plants produce secondary metabolites for defense either as distress signals to lure predators, or to directly deter or repel herbivores (Tak & Isman, 2017a) when act synergistically. Several combinations of plants' active compounds exhibited synergistic insecticidal activities when topically applied (Tak *et al.*, 2016). When these active chemical compounds used in combination as insecticides in crop protections against insect pests produce superior effects. For example, a binary mixture of 1,8-cineole and camphor showed enhanced insecticidal activity with a synergy ratio of 1.72 against the larva of *Trichoplusia ni* (Tak *et al.*, 2016). However, there is little information about synergistic action of phytochemicals from *T. vogelii*, *C. dichogamus* and *S. aromaticum* against cabbage insect pests in Tanzania.

CHAPTER THREE

MATERIALS AND METHODS

3.1 Plant materials collection, drying and grinding

The fresh plant materials were collected from different locations in Manyara, Arusha, Tanga and Kilimanjaro regions in Tanzania. Leaves of *C. dichogamus* and *T. vogelii* were collected from Manyara, Arusha and Kilimajaro regions of Tanzania. Flower buds of *S. aromaticum* plants were collected in Tanga Region. The leaves of plants were separately washed thoroughly with water and air dried under shade and at room temperature for 7 days. The dried leaves of *C. dichogamus* and *T. vogelii,* and flower buds of *S. aromaticum* were pulverized into fine particles (powder) using an electric blender. Then the powder obtained were stored in a cool and dark place for laboratory and field experimental procedures.

3.2 Larvicidal activity of dichloromethane – methanol extracts of *T. vogelii, C. dichogamus* and *S. aromaticum* plants against *C. binotalis* larvae and *P. xylostella* larvae

This experiment was conducted at the Laboratory of life sciences of the Nelson Mandela African Institution of Science and Technology to determine the larvicidal efficacy of dichloromethane – methanol (1:1) extracts of *T. vogelii*, *C. dichogamus* and *S. aromaticum* plants against *C. binotalis* larvae and *P. xylostella* larvae.

3.2.1 Preparation of extracts from plant materials for larvicidal bioassay

The powder of pesticidal plants obtained (Section 3.1) was sieved to get fine powder from which the extracts for larvicidal activity were prepared. The pesticidal plant powders were extracted in a separate container using dichloromethane – methanol (1:1) in equal ratio using 2 L Erlenmeyer flasks for 18 hours. Then the solution of the mixture was filtered through Whatman no. 40 filter paper and the filtrate was collected. This procedure was repeated two times with fresh volume of solvents for extraction again. The filtrates of the extracts were concentrated using a rotary-evaporator below 45 °C under low pressure and over water bath until the solvent completely evaporated. The extract of each plant was labelled and placed in the freezer at 4 °C until testing for larvicidal activity. Stock extract solutions (4%) were prepared by weighing 0.8 g of pesticidal plant extracts from each plant and dissolved into 20

mL of acetone and stirred for 45 min at 25 °C with a magnetic stirrer. Afterwards, the mixture was filtered through filter paper (Whatman No. 1). The stock aqueous solution (4%) from each plant were labelled and refrigerated at 4 °C until the subsequent larvicidal bioassay. The stock solution of each plant was further diluted with distilled water to prepare five concentrations (1.6, 8, 16, 24 and 32 mg/mL).

3.2.2 Collard green seedlings planting

Three weeks collard green (*Brassica oleracea* var. viridis) seedlings' leaves were used for the larval rearing and extract solution bioassay tests throughout the experiments. Collard green seeds were obtained from Kibo Seed Company Limited, Arusha, Tanzania and were sown in clear plastic cups (175 mL, one seed/ cup) containing sterilized soil (Plate 6A). A week after germination of collard green, seedlings were transferred to 200 mL plastic cups to allow the collard green plants to grow up to three weeks at least to possess large leaves for insect pests to lay eggs. No pesticides were used throughout the collard green plants preparation process.

3.2.3 *Plutella xylostella* and *C. binotalis* larvae insect pest rearing

Plutella xylostella and *C. binotalis* larvae (Plate 6C and 7C, respectively) were selected for laboratory bioassay experiments. Eggs were obtained from adult colony maintained at the glass cages of Tanzania Agricultural Research Institute (Tengeru Centre) TARI (latitude 3° 23′ 4.5″ S and longitude 36° 48′ 26.7″ E at an elevation of 1262 m above sea level) and optimum conditions (25 - 27 °C temperature and 76 - 82% relative humidity) which had been reared according to Prijono (1998) with little modifications (Plate 6A and 7A, respectively). The colonies were established and maintained at the glass cages (60 cm × 60 cm × 60 cm) at TARI (Plate 6A and 7A, respectively). The adults were collected from the Collard green garden farms of TARI, by using sweep nets. The collected adults (Plate 6B and 7B, respectively) were kept and maintained in the three cages and fed with 10% honey solution soaked in cotton pads (Plate 6 and 7). In each cage 40 Collard green (*Brassica oleracea* var. viridis) seedlings were placed for adults of the two insect pests to lay eggs into the leaves of Collard green seedlings. The leaves of collard green with eggs were then harvested into the lunch box for the eggs to hatch into larvae after about 4 days. The 3-4th instar larvae were obtained and collected for laboratory experiments and others were left into the lunch box and allowed for adult emergence for the purpose of increasing the number of adult colonies. By doing so, 3-4th instar larvae were continuously available for different bioassay tests. The whole setup of the rearing experiment

was maintained at 28 ± 2 ºC and 70-80% relative humidity under the light conditions approximated L12: D12 agro-light.

Plate 6: A - *P. xylostella* colony in cages, B - *P. xylostella* adult, C- *P. xylostella* larvae

Colony of *P. xylostella* reared in glass cages at 25-29 °C and 70-80% RH. The insects were supplied with 10% honey for feeding adults and collard green leaves to lay eggs and to feed larvae. Larvae that emerged were collected for bioassay.

Plate 7: A - *C. binotalis* colony in cages, B - *C. binotalis* adult, C- *C. binotalis* larvae

Colony of *C. binotalis* reared in glass cages at 25-29 °C and 70-80% RH. The insects were supplied with 10% honey for feeding adults and collard green leaves to lay eggs and to feed larvae. Larvae that emerged were collected for bioassay.

3.2.4 Bioassay test for larvicidal activity

The larval bioassay tests for the larvicidal effect of dichloromethane - methanolic (1:1) extract derived from *T. vogelii, C. dichogamu*s and *S. aromaticum* against *C. binotalis* and *P. xylostella* larvae were carried out in accordance with Prijono and Hassan (1993) with little modifications. During the bioassays, the reared 3-4th instar larvae of *P. xylostella* and *C. binotalis* were used (Plate 8 and 9). The 3-4th instar larvae of both insects were visually detected by relatively bigger size. Larvicidal activity (percentage of mortality) and LC_{50} and LC_{90} values were determined using WHO (2005) bioassay protocol with slight modifications. The tested larvae were free from any exposure to insecticides or chemicals. A series of five concentrations (1.6, 8, 16, 24 and 32 mg/mL) from the stock solutions was prepared. Ten larvae were released by means of a small fine camel's soft hair brush from the container into 9 cm petri dishes containing sliced leaves of Collard green for feeding larvae (Plate 9). Then, the treatment of larvae using the extracts diluted with distilled water with the required concentrations of solution was applied. A negative control (acetone diluted with distilled water only) and positive control (chlorpyrifos) was also included in the experiments (Plate 9). For each treatment, three replicates were used in order to check the mortality in a completely randomized design. Larvae were considered dead if they showed no sign of movement even after being probed with a fine camel's hair brush (Tian *et al*., 2015). The number of larval mortalities was recorded after every 6 hours interval for 24 hours and the percentage mortality was calculated using equation (1) and the % mortality was corrected using Abbott's formula (Abbott, 1925) equation (2).

$$\text{Percentage mortality} = \frac{\text{Number of dead larvae}}{\text{Number of tested larvae}} \times 100\% \quad \ldots\ldots\ldots\ldots\ldots\ldots\ldots\ldots\ldots\ldots \quad (1)$$

$$\text{Corrected \% mortality} = \frac{\text{\% Mortality in treated} - \text{\% Mortality in control}}{100 - \text{\% } Mortality \text{ in control}} \times 100 \ldots \quad (2)$$

Plate 8: A – sliced collard green leaves in 9 cm petri dishes ready for introducing larvae and treatments, B – *P. xylostella* larvae in infected collard green leaves ready for experiment (Prepared collard green leaves and insect pest larvae for experiment)

Plate 9: A-Prepared petri-dishes for larvicidal test, B & C - Treatments applications

3.3 Methodologies and materials for field experiments

The experiment from this sub-section was conducted to assess the insecticidal and synergistic actions of aqueous extracts from *S. aromaticum, C. dichogamus* and *T. vogelii* against common cabbages insect pests.

3.3.1 Study location for field experiments

The study for field experiments was conducted in the Northern part of Tanzania. The sites of the study were located in Arusha region and in Kilimanjaro region in Tanzania. In Kilimanjaro region, the experiment was set in Boro site located at Latitude 3°17′31.5′′S and Longitude 37°17′49.1′′E and an elevation of 1078 m above sea level in both two wet seasons (Plate 10). In Arusha region, the experiment was set in Tengeru site located at latitude 3°23′4.5′′S and longitude 36°48′26.7′′E at an elevation of 1262 m above sea level in both 2019 and 2020 wet seasons (Plate 10). Moreover, the meteorological data of rainfall precipitations and temperature was also observed from the experimental sites to observe the current weather situations.

3.3.2 Land preparation and transplanting

The land was cleared and prepared prior to transplanting of seedlings. Ploughing and harrowing was performed on the land before transplanting of the seedlings at both experimental sites using hand hoes and a plough. The Cabbage (*B. oleracea*) seeds were sown near the experimental plots on March 2019 and 2020 wet seasons, then after 5 weeks, the seedlings were transferred and transplanted into the experimental plots from the mid of April to August 2019 and 2020 wet seasons at both experimental sites (Plate 10). The seedlings of cabbage were planted at spacing of 50 cm between the rows and 45 cm in the rows within the plots that was measured 2.0 m × 2.5 m at both experimental sites (Plate 10). The distance from one plot and another plot was 0.5 m. Watering was done two times a day in the morning and the evening for one week after transplanting, then it was done once a day throughout the growing of the crop.

Plate 10: Cabbage (*B. oleracea*) crop on field experimental sites in 2019 and 2020 wet seasons

3.3.3 Experimental design and treatments

(i) Procedures and plant extracts preparation for field experiments

The experiment was laid out in a Randomized Complete Block Design (RCBD) with 12 treatments replicated four times. The treatments consisted aqueous plant extracts of three pesticidal plants (*T. vogelii, C. dichogamus* and *S. aromaticum*), two negative controls (water only and water plus soap) and one positive insecticide control (chlorpyrifos) were also included in the treatments.

From each individual plant, three concentrations 1%, 5% and 10% w/v of aqueous plant extracts were prepared to spray on the cabbage (*B. oleracea*) crop field. The concentrations of aqueous plant extracts were prepared in water containing 0.1% soap from dry powder of *T. vogelii, C. dichogamus* and *S. aromaticum.* Thus, 1%, 5% and 10% of aqueous plant extracts was prepared by dissolving 10 g, 50 g and 100 g of powder into one litre of water respectively. The extraction experiment was left to stand for 24 hours at room temperature (Mkenda *et al*., 2015). There were 12 treatments in each experimental site with 4 plot replicates making a total of 48 plots.

Apart from that, the aqueous plant extracts with combination of the pesticidal plants were prepared by mixing 25 g and 50 g of powder from each plant (*T. vogelii, C. dichogamus* and *S.*

aromaticum) in equal ratio and dissolved in one litre of water (w/v) separately to make 2.5% and 5% concentrations respectively. So, similarly there were 2 treatments with combination (2.5% and 5%) in each experimental site, replicated four times making a total of 8 treatments plots. Therefore, 8 plots of combination treatments and 48 plots made a grand total of 56 treatment plots in each experimental site. Mixing of the pesticidal plants was done to evaluate the synergistic insecticidal action of the aqueous extracts from *S. aromaticum, C. dichogamus* and *T. vogelii* against insect pests on cabbage crop (*B. oleracea*) in the field. Soap was used during extraction experimental procedure because firstly, it helps to extract compounds which are not water soluble from plant materials and secondly, it helps to spread the extract onto the plant leaves more effectively during applications.

(ii) Treatments applications

The treatments were sprayed into the cabbage (*B. oleracea*) crops in the field at the interval of 7 days throughout the growing of the *B. oleracea* crop in the two seasons. The concentration of synthetic insecticide (Chlorpyrifos) was applied as per manufacturers' recommendations. The treatments were sprayed, on top and under the leaves of *B. oleracea* crop by using a 2 L knapsack sprayer in the evening during the growing of the crops in both 2019 and 2020 wet seasons. The spraying was done during the evening hours in order to avoid direct sunlight which may cause the decomposition of bioactive compounds of the botanicals. Each plot required approximately 250 mL of the aqueous plant extracts at both sites. The sprayer was thoroughly cleaned with water and soap before re-filling it again with another formulation for application.

3.3.4 Insect assessment and cabbage crop damage assessment

Insect evaluation was done one day before spraying of pesticide and extracts by randomly selecting 5 inner cabbage crops inside the 2.0 m x 2.5 m plots each week. Small insect pests such as aphids were scored using a modified method (Afun *et al.*, 1991) as 0 = absent, 1 = a few scattered individuals, 2 = a few isolated small colonies, 3 = several isolated small colonies, 4 = large isolated colonies, 5 = large continuous colonies. Large insects like *P. xylostella, T. ni, H. undalis, C. binotalis* together with beneficial insects like spider, lady birds etc, were just counted. Assessment of damage severity of crops caused by insect pests was done through counting the number of damaged leaves of cabbage crops and their heads. Five cabbage crops were sampled from each plot randomly for counting the number of damaged plant parts. The

assessment of damaged parts was differentiated into four scale; 0% damage, up to 25% damage, up to 50% damage, up to 75% damage and up to 100 % depending on the number of leaves damaged (Mkenda *et al*., 2015). The incidence level was assessed by observing the infested cabbage crops per plot divided by the total number of cabbage crops per plot times 100%.

3.3.5 Measurement of weight, canopy spread and estimation of cabbage with heads

The measurement of canopy spread and estimation of cabbage with heads and without heads was done. Canopy spread was measured with a ruler at the time of harvest as the horizontal distance from one end of the plant to the other end. The cabbage with heads was recorded during harvesting time by recording the total number of cabbages with heads and without heads in a plot and then calculating the percentages of cabbage with heads and without heads in the plot. Then, 5 cabbage crops with marketable heads were randomly selected for measuring the weight from each plot at the centre and were recorded. The percent of cabbage with heads was determined by the following formula.

$$\text{Percent of cabbage with/without heads} = \frac{\text{Cabbage with/without heads in a plot}}{\text{Total cabbages in a plot}} \text{ x } 100\% \ldots (3)$$

3.4 Determining the chemical compounds in *C. dichogamus* and *S. aromaticum*

3.4.1 Procedures and plant material extract (crude extract) preparation

The prepared plant powder (300 g) from *C. dichogamus* and *S. aromaticum* plants was macerated overnight in 400 mL:400 mL of dichloromethane - methanol (DCM-MeOH) in equal ratio at room temperature for 18 hours with occasional shaking. Then the resulting extract was filtered using filter paper (Whatman No 1.5). The extraction of plant powder materials was performed using dichloromethane and methanol solvents mixed in equal ratio to see the chemical compounds which are present in the total extraction in 2000 mL of Erlenmeyer flask. The extract was filtered and then evaporated using a rotary-evaporator at 45 °C for complete dryness and complete freezing at - 20 °C followed by lyophilization. The supernatant was collected and filtered using filter paper and was dried by freezing to eliminate water by sublimation. The extract from two pesticidal plants were stored in a deep freezer at 4 °C for further activities. But, due to enough literatures even here in Tanzania for instance Mkindi *et al*. (2019) about the chemical compounds in *T. vogelii*, the literatures were reviewed to spot

out the chemical compounds which mighty be responsible for the larvicidal and insecticidal efficacy against *P. xylostella* and *C. binotalis* larvae and cabbage insect pests in the field.

3.4.2 Gas chromatography-Mass spectrometry (GC-MS) analysis

The GC-MS analysis was carried out using Agilent technologies 7890A GC connected to Agilent 5975 MSD (Agilent technology, USA). The inert gas helium (99.999%) was used as carrier gas with a flow rate of 1.2 mL/min. The GC was equipped with capillary column (HP 5) length of 30 m, film of 0.25 µm and internal diameter of 0.25 mm and temperature limit of 80 °C to 300 °C was used. The oven temperature rose from 80 °C up to 300 °C with the rate of 4 °C/min rise in temperature. The sample size of 1µl was injected through the injector port. The mass spectrometer operated in electron ionization mode with an ionizing energy of 70 eV. The inlet temperature was 250 °C and the total GC-MS running time was 35 minutes. The compounds found were recorded and reported.

3.5 Data analysis

3.5.1 Larvicidal data analysis

The data on larvae mortality counts were collected after 24 hours exposure, whereby mortality between 10 and 100% was considered. Bioassay tests showing more than 20% in the control mortality were discarded and the test was repeated. However, when the control mortality ranged from 5% to 20%, the corrected mortality was calculated using Abbott's formula (Abbott, 1925). Mean percent mortalities of the larvae that were treated with leaf extracts were determined by one-way Analysis of Variance (ANOVA) using the STATISTICA software. The Fisher's Least Significant Difference (LSD) was used to compare treatment means at $P = 0.05$ level of significance. Results with $P < 0.05$ were considered to be statistically significant. The concentration in mg/mL lethal to 50% (LC_{50}) and 90% (LC_{90}) for larvicidal activity with its 95% confidence intervals of Upper Confidence Limit (UCL) and Lower Confidence Limit (LCL), chi-square values, the slope, were estimated using probit analysis (Finney, 1971) with LdP Line software (http://www.ehabsoft.com/ldpline/) and SPSS software.

3.5.2 Field data analysis

The collected data from field experiments were analyzed using Statistica 8.0 software package version 7 program and the graphs were drawn by using excel software. Two-way ANOVA

statistical analyses were performed to compare plots' locations and the treatments. Three-way ANOVA statistical analyses were performed to compare plots' locations, season variations and the treatments. The Fisher's Least Significant Difference (LSD) was used to compare the treatment means at level of significance.

CHAPTER FOUR

RESULTS AND DISCUSSION

4.1 Results

The results of the larvae mortality in response to various concentrations of the three pesticidal plants used in this study addressed specific objective 1, the effectiveness of aqueous extracts from *S. aromaticum, T. vogelii* and *C. dichogamus* against cabbage (*B. oleracea*) insect pests at Tengeru experimental site and Boro experimental site in both 2019 and 2020 wet seasons addressed specific objective 2 and lastly the chemical compounds present in *S. aromaticum* and *C. dichogamus* addressed specific objective 3.

4.1.1 Larvicidal Efficacy of Dichloromethane-Methanol extracts from *S. aromaticum, T. vogelii* and *C. dichogamus* against *P. xylostella* and *C. binotalis* larvae

(i) Larvicidal efficacy of plant extracts against *P. xylostella*

The effect of plant extracts on mortality of P. xylostella larvae

Highest mortality percent of *P. xylostella* larvae was obtained in *S. aromaticum* at all concentrations after 24 hours of exposure (Table 7). The percent mortality of *P. xylostella* larvae in response to *S. aromaticum, C. dichogamus* and *T. vogelii* DCM-MeOH extracts differed significantly ($P \leq 0.05$) from the larvae treated with negative control (Table 7). In case of 1.6 mg/mL of extract solution of the three pesticidal plant, *S. aromaticum* gave the highest percent mortality (70.0 ± 0.0%) while the minimum percent mortality (30.0 ± 0.0) was recorded in *C. dichogamus* after 6 hours of exposure. The percent mortality was found to increase continuously from 6 to 24 hours of exposure and reached 93.3 ± 3.3% in *S. aromaticum* extract solution. In the case of 8 mg/mL, the percent mortality of *P. xylostella* larvae was found to be increased compared with 1.6 mg/mL of extract solution (Table 7; Fig. 7). The same trend of percent mortality of *P. xylostella* larvae was observed from short time exposure to long time exposure whereby the highest percent mortality (100.0 ± 0.0%) was observed in *S. aromaticum* and the lowest percent of larvae mortality (86.7 ± 3.3%) was observed in *C. dichogamus* from 6 to 24 hours of exposure. In general, it was found that the three pesticidal plant extracts at 16 mg/mL and above exhibited *P. xylostella* larvae mortality ranging from 80.0 ± 0.00 to 100.0 ± 0.00% after 12 hours of exposure (Table 7). Apart from that, the extract solutions of *S.*

aromaticum and *T. vogelii* both at 16, 24 and 32 mg/mL gave 100.0 ± 0.0% mortality of *P. xylostella* larvae after 18 hours of exposure. But the extract solution of *C. dichogamus* at 32 mg/mL gave 100.0 ± 0.0% larvae mortality after 18 hours of exposure (Table 7). It was revealed that, there was significantly ($P \leq 0.05$) lower percent mortality in synthetic pesticide (chlorpyrifos) which was the positive control on *P. xylostella* larvae compared with pesticidal plant extract solutions from 16 mg/mL and above (Table 7). Moreover, *T. vogelii, S. aromaticum* and *C. dichogamus* DCM-MeOH extract solutions used in this study at 16, 24 and 32 mg/mL caused 80.0 ± 0.0% and above after 6 hours of exposure (Table 7). The percent mortality of *P. xylostella* larvae was found to be increased with an increase in concentrations of the extracts and time of exposure (Table 7; Fig. 7). Therefore, the results from this study show the potentiality of the extract solutions from *T. vogelii, S. aromaticum and C. dichogamus* at high concentrations for the control *P. xylostella* larvae.

Table 7: Percent mortality of *P. xylostella* larvae after 24 hours exposure

Plant extracts and controls	**Mortality ± SE**			
Treatments	**6 hours**	**12 hours**	**18 hours**	**24 hours**
Acetone (- control)	3.3 ± 3.3h	3.3 ± 3.3f	3.3 ± 3.3g	3.3 ± 3.3f
Chlorpyrifos (+ control)	56.7 ± 3.3e	56.7 ± 3.3d	70.0 ± 5.8e	86.7 ± 3.3cd
T. vogelii (1.6 mg/mL)	46.7 ± 3.3ef	53.3 ± 8.8d	60.0 ± 5.8f	70.0 ± 5.8e
T. vogelii (8 mg/mL)	56.7 ± 3.3e	76.7 ± 3.3c	86.7 ± 3.3bc	90.0 ± 0.0bcd
T. vogelii (16 mg/mL)	80.0 ± 5.8cd	90.0 ± 0.0b	100.0 ± 0.0a	100.0 ± 0.0a
T. vogelii (24 mg/mL)	93.3 ± 3.3ab	100.0 ± 0.0a	100.0 ± 0.0a	100.0 ± 0.0a
T. vogelii (32 mg/mL)	96.7 ± 3.3a	100.0 ± 0.0a	100.0 ± 0.0a	100.0 ± 0.0a
S. aromaticum (1.6 mg/mL)	70.0 ± 0.0d	76.7 ± 3.3c	80.0 ± 0.0cd	93.3 ± 3.3abc
S. aromaticum (8 mg/mL)	80.0 ± 0.0cd	83.3 ± 3.3bc	93.3 ± 3.3ab	100.0 ± 0.0a
S. aromaticum (16 mg/mL)	90.0 ± 0.0abc	90.0 ± 5.7b	100.0 ± 0.0a	100.0 ± 0.0a
S. aromaticum (24 mg/mL)	93.3 ± 3.3ab	100.0 ± 0.0a	100.0 ± 0.0a	100.0 ± 0.0a
S. aromaticum (32 mg/mL)	96.7 ± 0.33a	100.0 ± 0.0a	100.0 ± 0.0a	100.0 ± 0.0a
C. dichogamus (1.6 mg/mL)	30.0 ± 0.0g	36.7 ± 5.8e	73.3 ± 3.3de	83.3 ± 3.3d
C. dichogamus (8 mg/mL)	43.3 ± 3.3f	50.0 ± 0.0d	80.0 ± 0.0cd	86.7 ± 3.3cd
C. dichogamus (16 mg/mL)	83.3 ± 3.3bc	83.3 ± 3.3bc	83.3 ± 3.3c	90.0 ± 0.0bcd
C. dichogamus (24 mg/mL)	83.3 ± 6.7bc	90.0 ± 0.0b	93.3 ± 3.3ab	96.7 ± 3.3ab
C. dichogamus (32 mg/mL)	96.7 ± 3.3a	100.0 ± 0.0a	100.0 ± 0.0a	100.0 ± 0.0a
1-Way ANOVA (F-statistics)	53.86***	67.32***	74.89***	93.29***

Each value is a mean ± standard error of three replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fisher's Least Significant Difference (LSD) test.

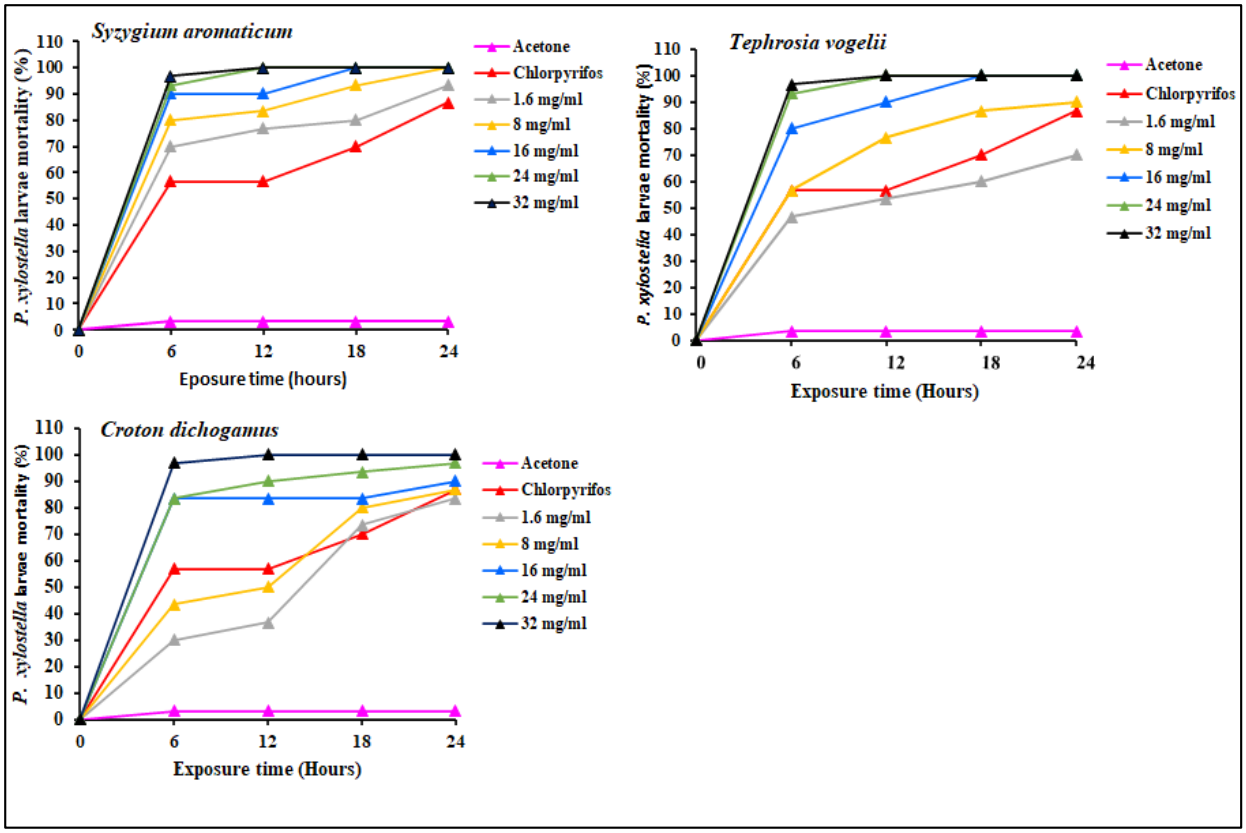


Figure 7: Variation of % mortality of *P. xylostella* larvae in DCM-MeOH extracts at different time intervals

Larvicidal activity of plant extracts against P. xylostella larvae

The LC_{50} and LC_{90} values of three pesticidal plant (*S. aromaticum, C. dichogamus* and *T. vogelii*) DCM-MeOH (1:1) extracts against the 4th instar larvae of *P. xylostella* were determined (Table 8). Among the three pesticidal plants, the DCM-MeOH (1:1) extracts of *S. aromaticum* displayed the highest larvicidal activity, with the lowest LC_{50} value of 0.081 mg/mL after 24 hours of exposure (Table 8; Fig. 8). Then, it was followed by *C. dichogamus* with the LC_{50} value of 0.105 mg/mL. The extracts of *T. vogelii* exhibited the LC_{50} value of 0.865 mg/mL in this study (Table 8). The LC_{90} values for the extracts of *S. aromaticum, T. vogelii* and *C. dichogamus* after 24 hours of exposure were 1.168, 4.876 and 6.569 mg/mL, respectively (Table 8). Moreover, the 95% Confidence limits (LCL – UCL) were also determined (Table 8). The LC_{50} and LC_{90} values of plant extracts on *P. xylostella* larvae decreased as the time of exposure increased (Fig. 8). Therefore, this study provides encouraging results which could possibly lead to recommendation of use of *S. aromaticum, C. dichogamus* and *T. vogelii* extracts as an ecofriendly larvicides and insecticide for the control of *P. xylostella* larvae.

Table 8: The LC_{50} and LC_{90} of the pesticidal plant extracts against *P. xylostella* larvae in 24 hours exposure

Plant extracts	95% Confidence limit		95% Confidence limit			Regression Equation
	LC_{50} (mg/mL)	LCL - UCL	LC_{90} (mg/mL)	LCL - UCL	$\chi 2$ (df=12)	
T. vogelii	0.865	0.206 -1.604	4.876	2.985 - 9.378	3.393	Y = 1.706X + 0.108
S. aromaticum	0.081	0.000 - 0.235	1.168	0.285 - 2.985	7.104	Y = 1.107X + 1.207
C. dichogamus	0.105	0.000 - 0.823	6.569	0.872 - 29.880	5.817	Y = 0.713X + 0.699

Larvicidal activity of dichloromethane-methanol (1:1) extract of *S. aromaticum, T. vogelii* and *C. dichogamus* against fourth instar larvae of *P. xylostella* with its respective lethal concentrations, confidence intervals (95%), chi-square, and standard error after 24 hours of exposure. LC_{50}: lethal concentration which kills 50% of the exposed larvae; LC_{90}: lethal concentration which kills 90% of the exposed larvae; LCL: Lower Confidence Limit; UCL: Upper Confidence Limit; χ^2, Chi-square; df: degree of freedom; ± SE: Standard Error. *Significant at P <0.05 level.

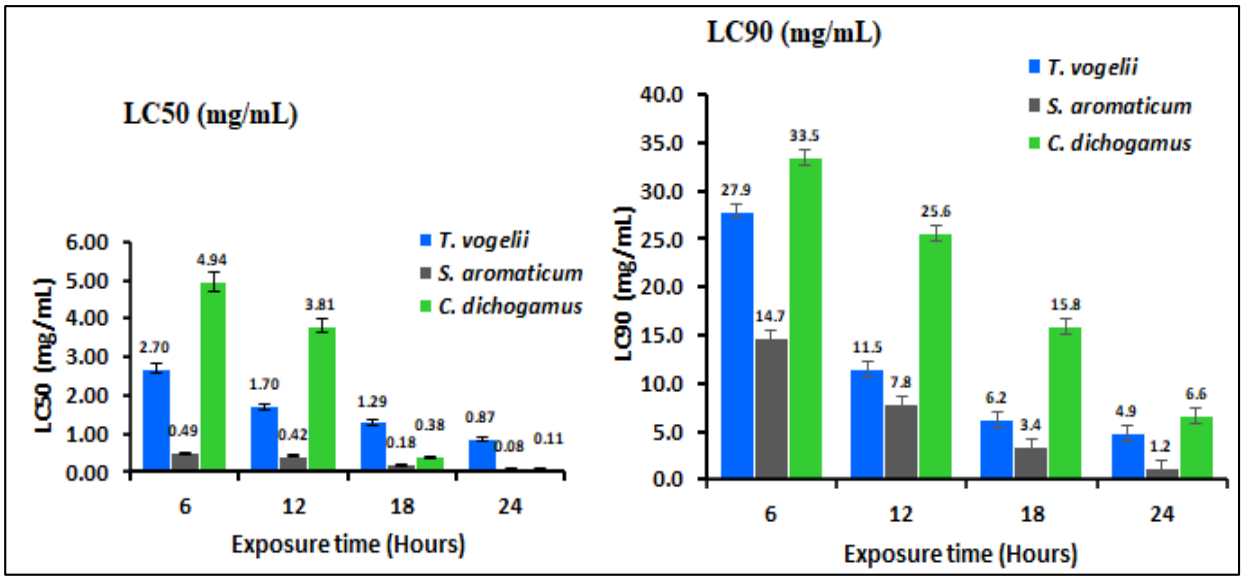


Figure 8: LC_{50} and LC_{90} values of plant extracts on *P. xylostella* at different time of exposure

Correlation of % mortality of P. xylostella larvae with concentrations of the extracts

The association of *S. aromaticum*, *C. dichogamus* and *T. vogelii* DCM-MeOH (1:1) concentrations of the extract and the percent mortality of *P. xylostella* larvae after 24 hours of exposure is shown in Fig. 9. It was clearly shown that, the percent mortality of *P. xylostella* larvae and the concentrations of *S. aromaticum*, *C. dichogamus* and *T. vogelii* DCM-MeOH (1:1) extracts have positive and very strong correlation (r = 0.938, r = 0.979, r = 0.920), respectively) (Fig. 9). Therefore, this correlation revealed that, the percentage mortality of *P. xylostella* larvae was found to be increased with an increase in concentrations of the *S.*

aromaticum, *C. dichogamus* and *T. vogelii* DCM-MeOH (1:1) extracts and time of exposure (Fig. 9).

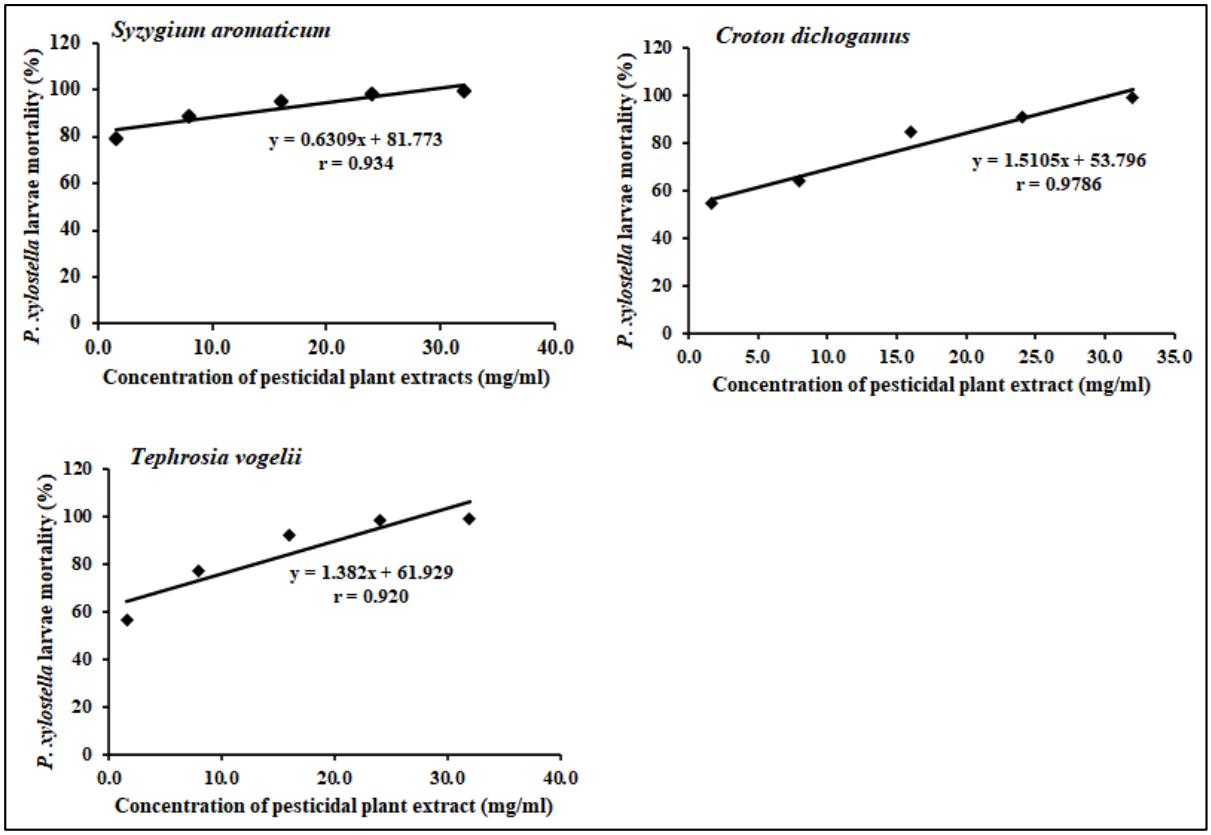


Figure 9: Linear regression equation (Y) and correlation coefficient (r) between plant extract concentration and larval mortality (%) of *P. xylostella* after 24 h exposure. The mortality percentage increased with increasing concentrations at 24 h and showed a positive correlation

(ii) Larvicidal activity of plant extracts against *C. binotalis* larvae

The effect of plant extracts on mortality of C. binotalis larvae

The effect on *Crocidolomia binotalis* larvae mortality due to *S. aromaticum*, *C. dichogamus* and *T. vogelii* DCM-MeOH extracts differed significantly ($P \leq 0.05$) from the *C. binotalis* larvae mortality treated with control (Table 9). It was found that all extracts at 8 mg/mL and above exhibited *C. binotalis* larvae mortality ranging from 80.0 ± 0.00 to 100.0 ± 0.00% after 18 hours of exposure (Table 9; Fig. 10). Moreover, *S. aromaticum* extracts from 8 mg/mL and above were significantly ($P \leq 0.05$) effective as synthetic pesticide (chlorpyrifos) after 12 hours of exposure (Table 9). The extracts of *S. aromaticum* at 24 and 32 mg/mL gave 100.0 ± 0.0% mortality of *C. binotalis* larvae after 12 hours of exposure and in this scenario was the most effective and caused 100% mortality of *C. binotalis* larvae in just 12 hours of exposure.

Moreover, it was found that, from 18 hours of exposure and above, *S. aromaticum* extracts at 16, 24 and 32 mg/mL gave 100.0 ± 0.0% mortality of *C. binotalis* larvae. Also, *T. vogelii* and *C. dichogamus* at 32 mg/mL gave 100% mortality of *C. binotalis* larvae (Table 9). In general, *T. vogelii, S. aromaticum and C. dichogamus* extracts used in this study at 32 mg/mL caused 90.0 ± 0.0%, 96.7 ± 3.3% and 96.7 ± 3.3% mortality of *C. binotalis* larvae respectively after 6 hours of exposure (Table 9; Fig. 10). The percent mortality of *C. binotalis* larvae increased with increase in concentrations of the extracts (Table 9) and increase in time of exposure (Fig. 10). It was revealed that, the DCM-MeOH (1:1) extracts of *T. vogelii, S. aromaticum* and *C. dichogamus* can be used to control *C. binotalis* larvae which is the most destructive stage of this insect pests to cabbage (*B. oleracea*) in the field.

Table 9: Percent mortality of *C. binotalis* larvae after 24 hours exposure

Treatments	Time in hours			
	6	12	18	24
Acetone	10.0 ± 0.0g	13.3 ± 3.3i	16.7 ± 3.3g	20.0 ± 0.0d
Chlorpyrifos	76.7 ± 3.3b	86.7 ± 3.3bc	90.0 ± 0.0bcd	100.0 ± 0.0a
T. vogelii (1.6 mg/mL)	33.3 ± 3.3f	53.3 ± 3.3h	63.3 ± 3.3f	80.0 ± 0.0c
T. vogelii (8 mg/mL)	43.3 ± 3.3ef	56.7 ± 3.3gh	83.3 ± 6.7de	93.3 ± 3.3ab
T. vogelii (16 mg/mL)	56.7 ± 3.3cd	63.3 ± 6.7fgh	86.7 ± 6.7cde	96.7 ± 3.3ab
T. vogelii (24 mg/mL)	66.7 ± 3.3bc	80.0 ± 5.8cd	90.0 ± 5.8bcd	100.0 ± 0.0a
T. vogelii (32 mg/mL)	90.0 ± 0.0a	96.7 ± 3.3ab	100.0 ± 0.0a	100.0 ± 0.0a
S. aromaticum (1.6 mg/mL)	66.7 ± 3.3bc	76.7 ± 3.3bc	86.7 ± 3.3b	93.3 ± 3.3ab
S. aromaticum (8 mg/mL)	76.7 ± 3.3b	76.7 ± 3.3bc	90.0 ± 0.0ab	96.7 ± 3.3ab
S. aromaticum (16 mg/mL)	76.7 ± 8.8b	93.3 ± 3.3a	100.0 ± 0.0a	100.0 ± 0.0a
S. aromaticum (24 mg/mL)	93.3 ± 3.3a	100.0 ± 0.0a	100.0 ± 0.0a	100.0 ± 0.0a
S. aromaticum (32 mg/mL)	96.7 ± 3.3a	100.0 ± 0.0a	100.0 ± 0.0a	100.0 ± 0.0a
C. dichogamus (1.6 mg/mL)	43.3 ± 3.3ef	53.3 ± 3.3h	70.0 ± 0.0f	80.0 ± 0.0c
C. dichogamus (8 mg/mL)	53.3 ± 3.3de	66.7 ± 3.3efg	80.0 ± 0.0cd	90.0 ± 0.0b
C. dichogamus (16 mg/mL)	63.3 ± 3.3cd	66.7 ± 3.3efg	83.3 ± 3.3de	90.0 ± 0.0b
C. dichogamus (24 mg/mL)	63.3 ± 3.3cd	73.3 ± 3.3def	90.0 ± 0.0bcd	93.3 ± 3.3ab
C. dichogamus (32 mg/mL)	96.7 ± 3.3a	96.7 ± 3.3ab	100.0 ± 0.0a	100.0 ± 0.0a
1-Way ANOVA (F-statistics)	42.64***	39.62***	35.07***	69.83***

Each value is a mean ± standard error of three replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fisher's Least Significant Difference (LSD) test.

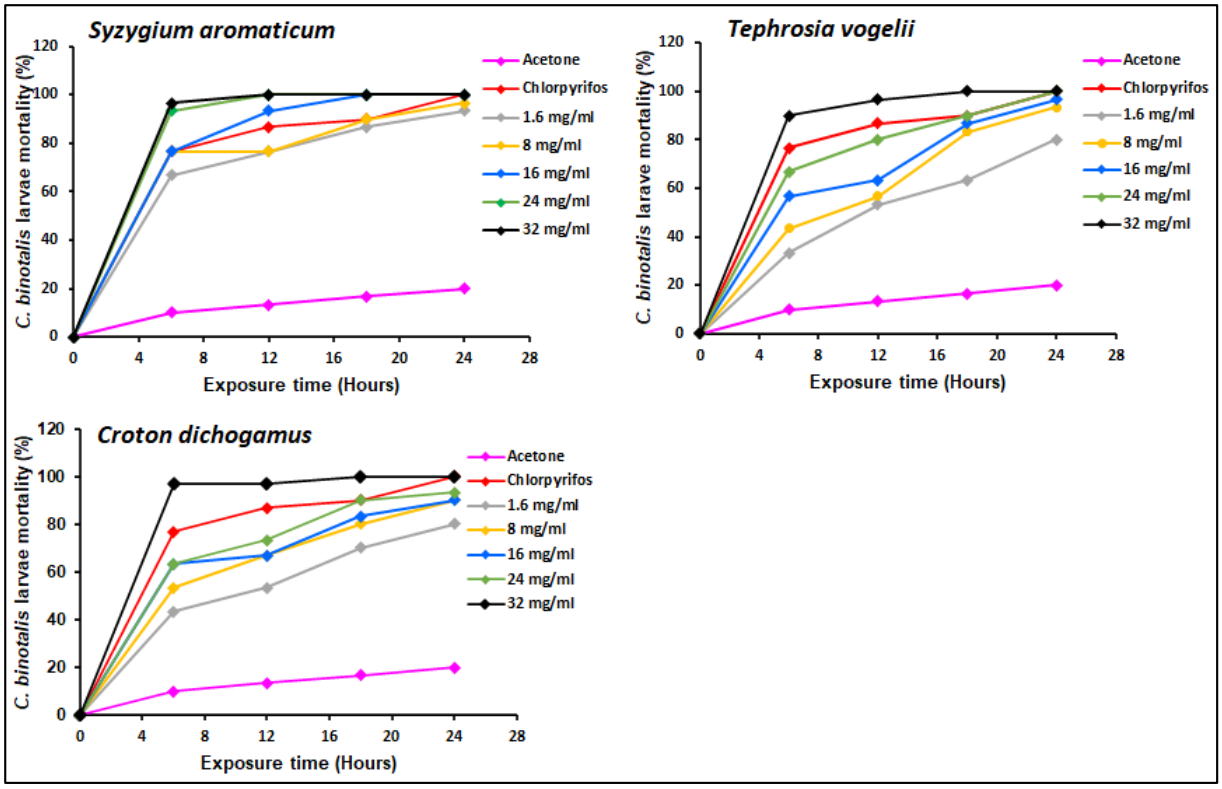


Figure 10: Variation of % mortality of *C. binotalis* larvae in different concentrations of extracts at different time intervals

In addition, the percent mortality of the *C. binotalis* larvae increased significantly ($P \leq 0.05$) with time of exposure to a particular concentration of the pesticidal plant extracts (Fig. 10). The percent mortality of *C. binotalis* larvae due to pesticidal plant extracts used in this study, differed significantly ($P \leq 0.05$) from 6 to 24 hours of exposure (Fig. 10). After 6 hours of exposure to pesticidal plant extracts, less than 55% of the *C. binotalis* larvae mortality was observed in *T. vogelii* (1.6 mg/mL and 8 mg/mL) and *C. dichogamus* (1.6 mg/mL and 8 mg/mL) extracts (Table 9). But at 1.6, 8, 16 and 24 mg/mL concentrations of *S. aromaticum* extracts used in this study exhibited highest percent of mortality *C. binotalis* larvae even at short time exposure (Fig. 10). It was found that, the higher the exposure time of the *C. binotalis* larvae to the extracts, the higher the percent mortality was observed (Fig. 10).

Larvicidal activity of the plant extracts against C. binotalis fourth instar larvae

The LC_{50} and LC_{90} values of three pesticidal plant (*S. aromaticum, C. dichogamus* and *T. vogelii*) DCM-MeOH (1:1) extracts against the 4th instar larvae of *C. binotalis* were determined (Table 10) after 24 hours of exposure. Among the three pesticidal plants, the DCM-MeOH (1:1) extracts of *S. aromaticum* exhibited the highest larvicidal activity, with the lowest LC_{50} value of 0.081 mg/mL after 24 hours of exposure (Table 10). It was followed by *C. dichogamus*

with the LC_{50} value of 0.127 mg/mL and lastly *T. vogelii* with LC_{50} value of 0.377 mg/mL in this study (Table 10). The LC_{90} values for the three pesticidal plant extracts (*S. aromaticum, C. dichogamus* and *T. vogelii*) after 24 hours of exposure were 1.168, 7.558 and 3.911 mg/mL respectively (Table 10). The 95% Confidence limits (LCL – UCL) were also determined (Table 10). The LC_{50} and LC_{90} values on *C. binotalis* decreases as the time of exposure increased (Fig. 11). Therefore, the present study recommends the use of *S. aromaticum, C. dichogamus* and *T. vogelii* extracts as an ecofriendly larvicide and insecticide for the control of *C. binotalis* at the larval stage (Table 10).

Table 10: The LC_{50} and LC_{90} of the pesticidal plant extracts against *C. binotalis* in 24 hours exposure

Plant extracts	95% Confidence limit		95% Confidence limit			Regression Equation
	LC_{50} (mg/mL	LCL - UCL	LC_{90} (mg/mL	LCL - UCL	χ^2 (df=12)	
T. vogelii	0.377	0.009 -1.094	3.911	1.583 - 8.320	7.205	Y = 1.261X + 0.535
S. aromaticum	0.081	0.000 - 0.212	1.168	0.275 - 2.768	7.104	Y = 1.106X + 1.207
C. dichogamus	0.127	0.000 - 0.883	7.558	1.672 - 36.337	3.527	Y = 0.722X + 0.647

Larvicidal activity of dichloromethane-methanol (1:1) extract of *S. aromaticum, T. vogelii* and *C. dichogamus* against fourth instar larvae of *C. binotalis* with its respective lethal concentrations, confidence intervals (95%), chi-square, slope and standard error after 24 hours of exposure. LC_{50}: lethal concentration which kills 50% of the exposed larvae; LC_{90}: lethal concentration which kills 90% of the exposed larvae; LCL: Lower Confidence Limit; UCL: Upper Confidence Limit; χ^2, Chi-square; df: degree of freedom; ± SE: Standard Error. *Significant at P <0.05 level.

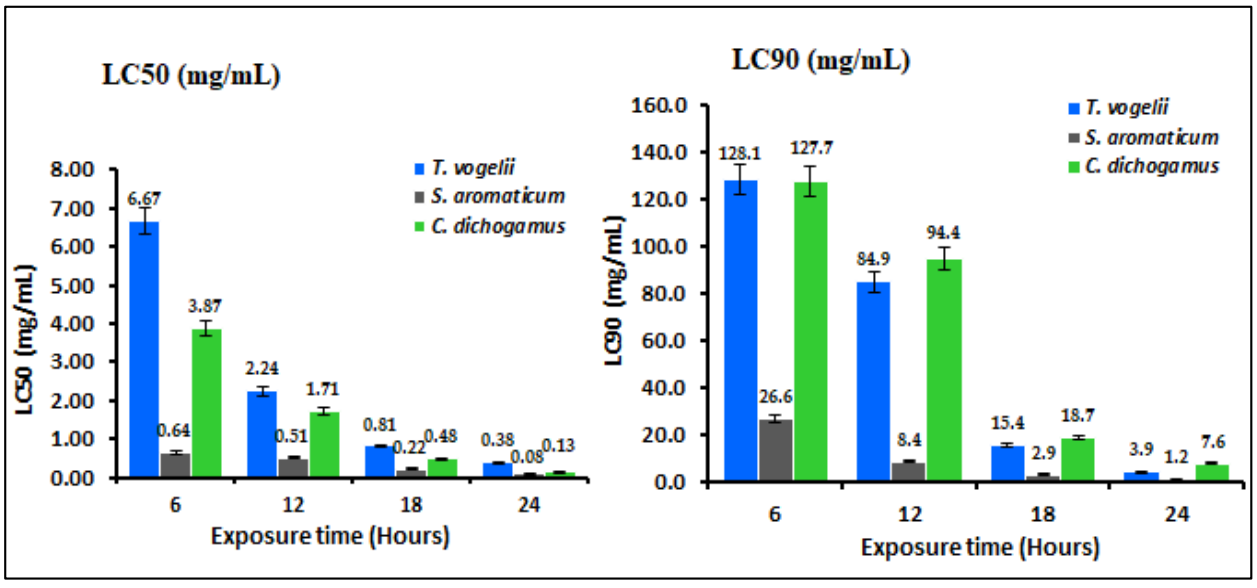


Figure 11: LC_{50} and LC_{90} values of plant extracts on *C. binotalis* at different time of exposure

Correlation of % mortality of C. binotalis larvae with concentrations of the extracts

Figure 12, shows the association of *S. aromaticum*, *C. dichogamus* and *T. vogelii* DCM-MeOH (1:1) concentrations of the extracts and the percent mortality of *C. binotalis* larvae after 24 hours of exposure. It was clearly revealed that, the concentrations of *S. aromaticum*, *C. dichogamus* and *T. vogelii* DCM-MeOH (1:1) extracts and the percent mortality of *C. binotalis* larvae have positive and very strong correlation (r = 0.9702, r = 0.9485, r = 0.9926, respectively) (Fig. 12). This correlation suggests that, the percent mortality of *C. binotalis* larvae increased proportionally with concentrations of the *S. aromaticum*, *C. dichogamus* and *T. vogelii* DCM-MeOH (1:1) extracts after 24 hours of exposure and showed a positive and very strong association (Fig. 12).

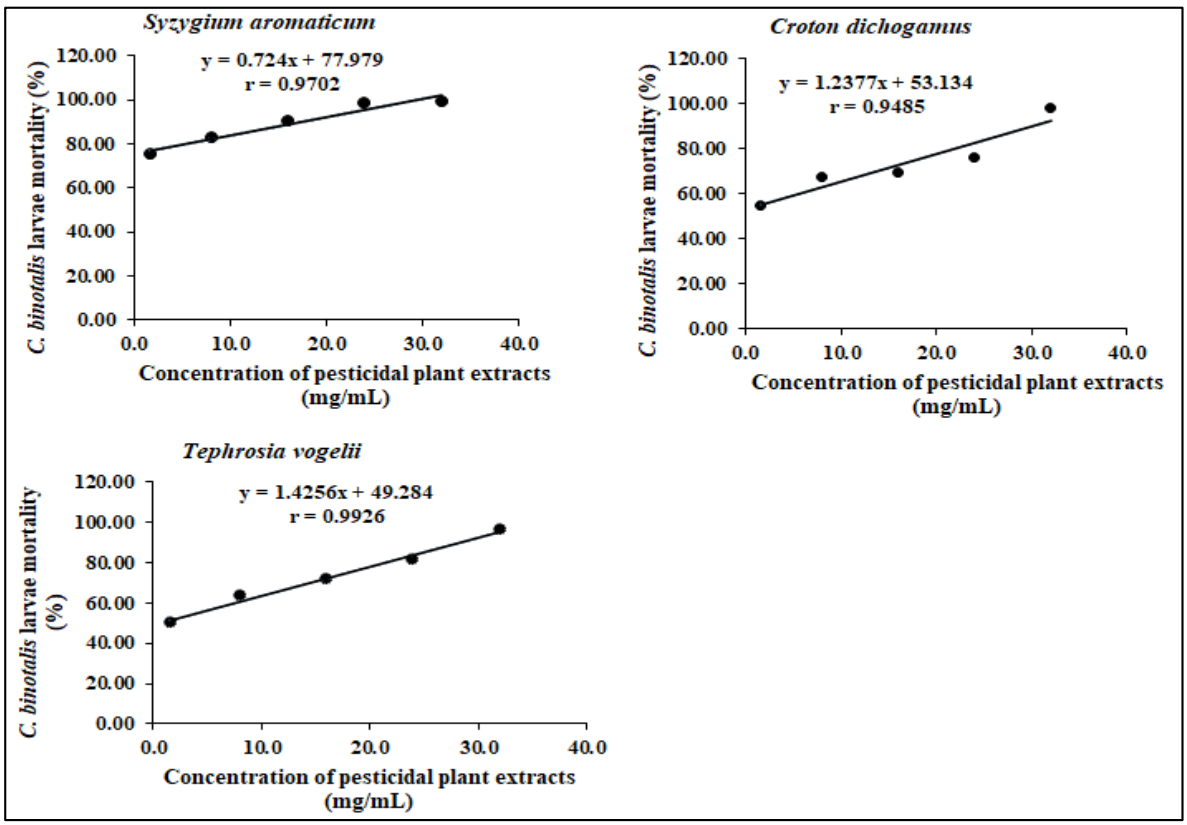


Figure 12: Linear regression equation (y) and correlation coefficient (r) between plant extract concentrations and larval mortality (%) after 24 hours of exposure

4.1.2 Weather conditions of experimental sites, insecticidal and synergistic action of extracts from selected plants against common cabbage insect pests

This section reports the results of the maximum and minimum temperatures together with the rainfall precipitations of experimental sites between the two seasons and the insecticidal and

synergistic action of extracts from *S. aromaticum, C. dichogamus* and *T. vogelii* and the synthetic pesticide used as a positive control against cabbage (*B. oleracea*) insect pests (*B. brassicae, M. persicae, P. xylostella*, *T. ni, H. undalis* and *C. binotalis*). The negative controls used in this study were water and water plus soap.

(i) Temperatures and rainfall precipitation of the experimental sites in 2019 and 2020 seasons

Figure 13 (A, B, C and D) shows the temperatures and precipitation of the two experimental sites in 2019 season and 2020 season, respectively. It was observed that the maximum temperatures of Tengeru experimental site were higher relative to those of Boro experimental site in both seasons throughout the study periods (Fig. 13A and 13C). However, the minimum temperatures varied relatively between the two experimental sites in the two seasons. Boro experimental site was cooler than Tengeru experimental site throughout the study periods 2019 and 2020 seasons. Moreover, the rainfall precipitations of Boro experimental site were higher than that of Tengeru experimental site in 2019 season and the difference was observed in April-June at the two experimental study sites. But, in 2020 season, the rainfall precipitations were higher in March, July, August and September at Tengeru experimental site than at Boro experimental site. In February, April and June the rainfall precipitations were relatively the same in both experimental sites in 2020 season. The mean maximum and minimum temperatures of Boro experimental site were 25.16 and 16.11 ℃, respectively while the mean maximum and minimum temperatures of Tengeru experimental site were 29.54 and 16.91 ℃, respectively in 2019 season, which were higher than those of Boro experimental site. Also, the mean rainfall precipitations of Boro and Tengeru experimental sites were 148.05 and 70.81 mm respectively in 2019 season. In 2020 season, the mean maximum and minimum temperatures of Boro experimental site were 24.3 and 15.7 ℃, respectively while at Tengeru experimental site, the mean maximum and minimum temperatures were 28.0 and 17.1 ℃, respectively (Fig. 13). The mean rainfall precipitations at Tengeru and Boro experimental sites were 253.7 and 209.4 mm, respectively (Fig. 13). It was found that the mean rainfall precipitations in 2020 season was higher than in 2019 season. These variations in rainfall precipitations and temperatures could have affected either by lowering or increasing the population abundance of *B. brassicae, M. persicae, P. xylostella, T. ni, C. binotalis* and *H. undalis* pests between 2019 and 2020 seasons and between the two experimental sites.

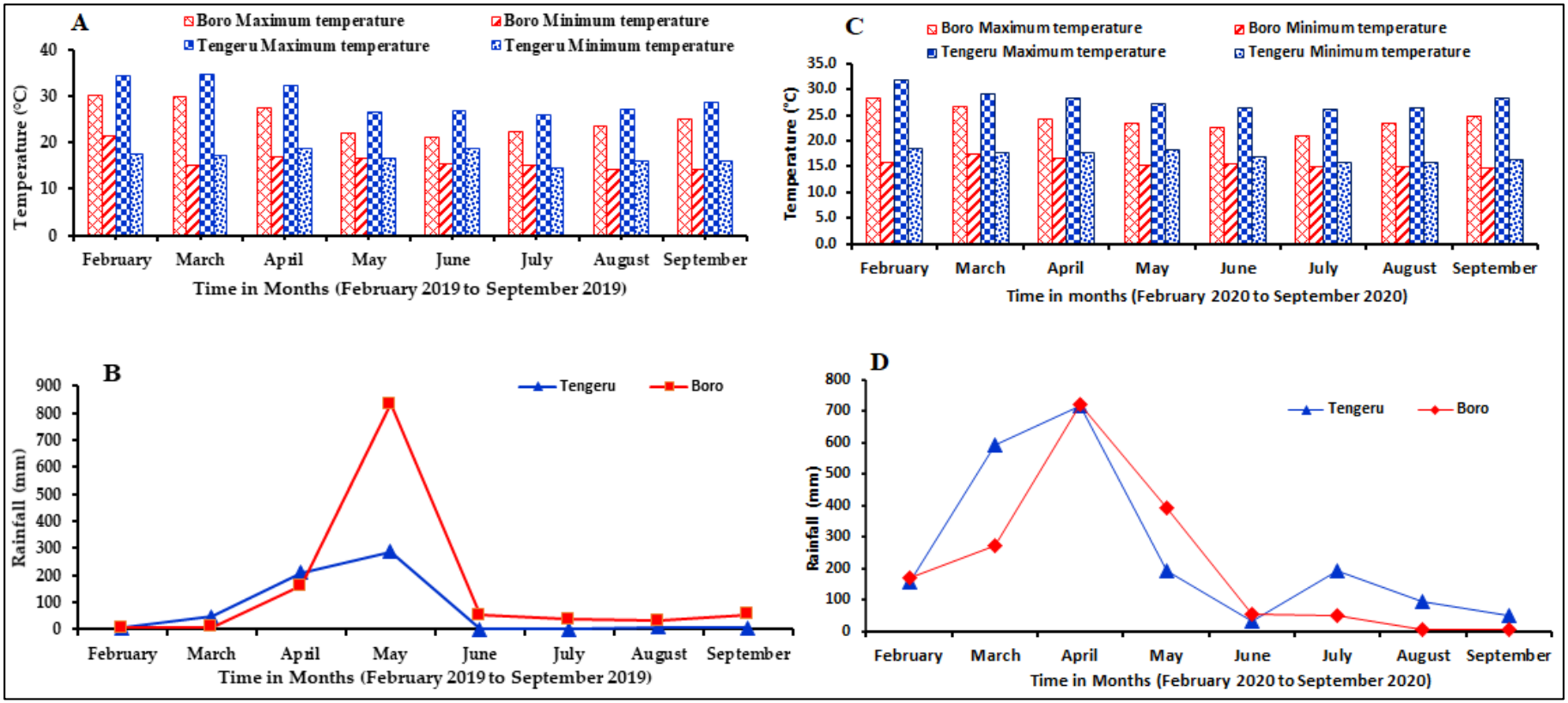


Figure 13: Temperatures (A & C) and Rainfall precipitations (B & D) of Boro experimental site and Tengeru experimental site during the field experiments 2019 and 2020

(ii) Insecticidal and synergistic action of extracts from the selected plants against cabbage insect pests

This part reports the insecticidal efficacy of the aqueous extracts of *T. vogelii*, *C. dichogamus* and *S. aromaticum* against common insect pests recorded infesting *B. oleracea* in the field in Northern Tanzania. Six common insect pests damaging *B. oleracea* were observed at the field sites whereby, two were aphids, the green peach aphids (*M. persicae*) (Plate 5) and cabbage aphids (*B. brassicae*) (Plate 4). Others were larvae of various moths including *P. xylostella* larvae (Plate 3 and 6), *H. undalis* larvae (Plate 2), *T. ni* larvae (Plate 1) and *C. binotalis* larvae (Plate 7). Therefore, the efficacy of aqueous plant extracts used in this study against these mentioned insect pests are reported in this part.

Population dynamics of B. brassicae in response to the treatments

Cabbage aphids (*B. brassicae*) were found in the field plots just the 3rd week after transplanting of the seedlings in both wet seasons (2019 and 2020 seasons). However, on the 3rd week, they were very scattered on the *B. oleracea* crops' leaves, stems and the shoots hence the infestation due to their presence was low. On the 4th week, the application of the treatments began. Generally, it was observed that, the population abundance of *B. brassicae* was significantly ($p \leq 0.01$) lower (0.35 ± 0.04) at Boro experimental site than (0.56 ± 0.08) at Tengeru experimental site in 2019 wet season. But in 2020 wet season, the population abundance of *B. brassicae* was insignificant (Table 11). Moreover, it was found that the population abundance of *B. brassicae* was significantly ($P \leq 0.05$) lower in the aqueous plant extracts and synthetic pesticide treated plots compared with negative controls (Table 11). The 5% concentration of the aqueous extracts from the mixed plants, had significantly ($P \leq 0.01$) lower (0.07 ± 0.02 and 0.13 ± 0.03) population abundance of *B. brassicae* as synthetic pesticide (chlorpyrifos) (0.09 ± 0.02 and 0.15 ± 0.03) in 2019 and 2020 wet seasons, respectively (Table 11). Thus, the 5% concentration of the aqueous extracts from the mixed plants was effective as synthetic pesticide (chlorpyrifos) for the reduction of population abundance of *B. brassicae* in the field in 2019 and 2020 wet seasons (Table 11). That was followed by the 10% concentration of the aqueous extracts from *C. dichogamus, T. vogelii* and *S. aromaticum.* The other concentrations (1% and 5%) of the aqueous extracts from *C. dichogamus, T. vogelii* and *S. aromaticum* and 2.5% of the aqueous extracts from the mixed plants significantly reduced the population abundance of *B. brassicae* compared with negative controls for both seasons (Table 11).

In addition, the weekly observations of the two wet seasons and two experimental sites revealed that, the mean population abundance of *B. brassicae* differed significantly between 2019 and 2020 wet seasons. It was found that, the mean population abundance of *B. brassicae* was significantly ($p \leq 0.01$) higher (1.16 ± 0.05, 0.53 ± 0.04, 0.63 ± 0.06, 0.82 ± 0.08, 0.74 ± 0.08, 0.75 ± 0.09) in 2020 wet season compared with 2019 wet season (0.41 ± 0.03, 0.45 ± 0.04, 0.38 ± 0.04, 0.45 ± 0.05, 0.47 ± 0.05, 0.51 ± 0.07) from week 1 before application of the treatments to weeks 1, 2, 3, 4 and 5 after application of the treatments, respectively (Table 12). Moreover, it was found that, the mean population of *B. brassicae* was significantly higher (0.86 ± 0.06, 0.55 ± 0.04, 0.72 ± 0.09) at Tengeru experimental site compared with Boro experimental site (0.70 ± 0.05, 0.43 ± 0.03, 0.54 ± 0.07) on week 1 before applications of the treatments and week 1 and 5 after applications of the treatments, respectively (Table 12). But, on weeks 2, 3 and 4 after application of treatments, the population abundance of *B. brassicae* was insignificant between the two experimental sites (Table 12).

The insecticidal efficacy of weekly observation results showed that, before treatment application, the mean population abundance of *B. brassicae* on different plots of the two experimental sites and in the two wet seasons was random and was insignificant ($p > 0.05$) among the plots in the field. However, on weeks 1, 2, 3, 4 and 5 of the treatment applications, the population abundance of *B. brassicae* was significantly different ($P \leq 0.05$) in the plots (Table 12). The study revealed that, the 5% concentration of the aqueous extracts from the mixed plants, possessed significantly ($p \leq 0.01$) lower (0.10 ± 0.04, 0.14 ± 0.04, 0.10 ± 0.03, 0.10 ± 0.03 and 0.09 ± 0.03) population of *B. brassicae* compared with other concentrations of plant extracts used in this study from weeks 1, 2, 3, 4 and 5 after treatment applications, respectively (Table 12). It was significantly ($P \leq 0.05$) lower as in synthetic pesticide (chlorpyrifos) (0.23 ± 0.05, 0.10 ± 0.03, 0.11 ± 0.03, 0.10 ± 0.03 and 0.05 ± 0.02) from weeks 1, 2, 3, 4 and 5 after treatment applications, respectively. Then, this was followed by 10% concentrations of the aqueous extracts of *C. dichogamus, T. vogelii* and *S. aromaticum.* However, the other concentrations (1% and 5 %) of the aqueous extracts of *C. dichogamus, T. vogelii* and *S. aromaticum* and 2.5% of the aqueous extracts from the mixed plants significantly reduced the population abundance of *B. brassicae* compared with negative controls for both wet seasons (Table 12).

When each season is considered separately, the results of weekly observations revealed that, the population abundance of *B. brassicae* was significantly ($p \leq 0.01$) lower (0.33 ± 0.04, 0.36

± 0.05, 0.38 ± 0.05, 0.35 ± 0.06) at Boro experimental site than (0.58 ± 0.06, 0.54 ± 0.08, 0.58 ± 0.10, 0.68 ± 0.11) at Tengeru experimental site in 2019 season on weeks 1, 3, 4 and 5, after treatment application, respectively (Appendix 1). But on the 1st week before application of the treatments and the 2nd week after application of the treatments, the population abundance of *B. brassicae* was insignificant (Appendix 1). In 2020 wet season, the population abundance of *B. brassicae* was insignificant between the two experimental sites except in the first week before applications of the treatments and the 2nd week after application of the treatments (Appendix 1). Therefore, the aqueous extracts of *C. dichogamus, T. vogelii* and *S. aromaticum* possessed significantly ($P \leq 0.05$) lower population abundance of *B. brassicae* compared with negative controls (water and water plus soap) in the field for both wet seasons (Table 11). Moreover, in the negative controls (water and water plus soap) treated plots, *B. brassicae* persisted from week one before and the 1st, 2nd, 3rd, 4th and 5th weeks after treatments applications and the population increased continuously in both negative controls (water and water plus soap) (Table 11 and 12). Moreover, it was observed that the effectiveness of the aqueous plant extracts depended on mixing of plant materials during preparations and the concentration of the aqueous extracts used (Table 11 and 12).

Table 11: Mean population of *B. brassicae* per cabbage crop in response to the treatments

Location and Treatments	**Seasons**	
	2019 season	**2020 season**
Site		
Tengeru	0.56 ± 0.08a	0.66 ± 0.09a
Boro	0.35 ± 0.04b	0.72. ± 0.10a
Treatments		
Water	1.38 ± 0.24a	2.36 ± 0.16a
water + soap	1.22 ± 0.26a	2.23 ± 0.09a
Synthetic pesticide	**0.09 ± 0.02d**	**0.15 ± 0.03g**
C. dichogamus (1%)	0.61 ± 0.06b	0.66 ± 0.07bc
C. dichogamus (5%)	0.40 ± 0.04bc	0.50 ± 0.04cd
C. dichogamus (10%)	0.19 ± 0.04cd	0.28 ± 0.03efg
S. aromaticum (1%)	0.56 ± 0.06b	0.81 ± 0.07b
S. aromaticum (5%)	0.37 ± 0.06bcd	0.51 ± 0.07cd
S. aromaticum (10%)	0.20 ± 0.04cd	0.30 ± 0.05efg
T. vogelii (1%)	0.55 ± 0.07b	0.72 ± 0.07b
T. vogelii (5%)	0.32 ± 0.03bcd	0.46 ± 0.03de
T. vogelii (10%)	0.16 ± 0.04cd	0.26 ± 0.03fg
Mixed plants (2.5%)	0.25 ± 0.04cd	0.35 ± 0.04def
Mixed plants (5%)	**0.07 ± 0.02d**	**0.13 ± 0.03g**
2 - way ANOVA	**(F- Statistics)**	
Site	18.30***	3.36ns
Treatments	18.65***	113.88***
Location*treatments	1.74ns	0.90ns

Each value is a mean ± standard error of eight replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fisher's Least Significant Difference (LSD) test.

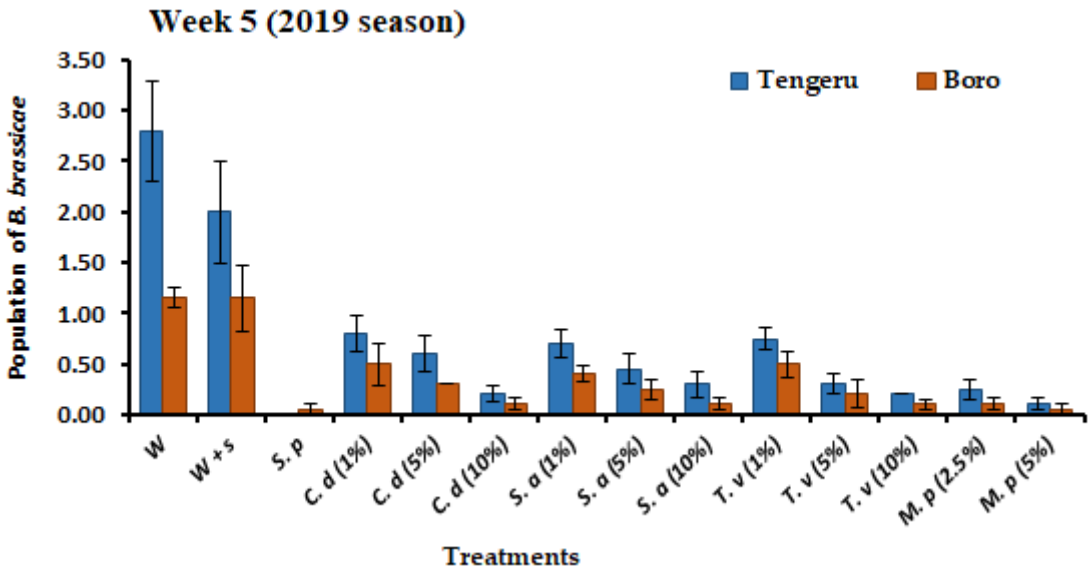
Table 12: Mean population of *B. brassicae* per crop in response to weekly application of treatments

Location and Treatments	Week1 before Treatment	Weeks after treatments				
		1	2	3	4	5
Seasons						
Wet season 1 (2019)	0.41 ± 0.03b	0.45 ± 0.04b	0.38 ± 0.04b	0.45 ± 0.05b	0.47 ± 0.05b	0.51 ± 0.07b
Wet season 2 (2020)	1.16 ± 0.05a	0.53. ± 0.04a	0.63 ± 0.06a	0.82 ± 0.08a	0.74 ± 0.08a	0.75 ± 0.09a
Locations						
Tengeru	0.86 ± 0.06a	0.55 ± 0.04a	0.48 ± 0.05a	0.66 ± 0.07a	0.64 ± 0.07a	0.72 ± 0.09a
Boro	0.70 ± 0.05b	0.43. ± 0.03b	0.53 ± 0.05a	0.62 ± 0.07a	0.57 ± 0.06a	0.54 ± 0.07b
Treatments						
Water	1.16 ± 0.14a	1.25 ± 0.11a	1.51 ± 0.19a	1.95 ± 0.27a	2.13 ± 0.25a	2.49 ± 0.25a
water + soap	1.05 ± 0.13a	1.19 ± 0.14a	1.26 ± 0.16ab	1.90 ± 0.27a	1.98 ± 0.23a	2.29 ± 0.26a
Synthetic pesticide	**0.89 ± 0.18a**	**0.23 ± 0.05fg**	**0.10 ± 0.03g**	**0.11 ± 0.03f**	**0.10 ± 0.03f**	**0.05 ± 0.02f**
C. dichogamus (1%)	0.51 ± 0.09a	0.65 ± 0.08b	0.63 ± 0.09bc	0.64 ± 0.08bcd	0.60 ± 0.05bc	0.64 ± 0.08bc
C. dichogamus (5%)	0.74 ± 0.16a	0.45 ± 0.05cde	0.45 ± 0.05cde	0.49 ± 0.06cde	0.44 ± 0.05bcde	0.41 ± 0.07bcd
C. dichogamus (10%)	0.60 ± 0.11a	0.20 ± 0.04fg	0.26 ± 0.05efg	0.23 ± 0.05ef	0.28 ± 0.04ef	0.20 ± 0.04def
S. aromaticum (1%)	0.94 ± 0.18a	0.64 ± 0.07bc	0.53 ± 0.09cd	0.91 ± 0.12b	0.71 ± 0.07b	0.64 ± 0.08bc
S. aromaticum (5%)	0.66 ± 0.14a	0.46 ± 0.09bcde	0.44 ± 0.06cde	0.50 ± 0.09cde	0.43 ± 0.05cde	0.36 ± 0.05cde
S. aromaticum (10%)	0.65 ± 0.13a	0.28 ± 0.09efg	0.26 ± 0.07efg	0.29 ± 0.05ef	0.21 ± 0.05ef	0.20 ± 0.04def
T. vogelii (1%)	0.84 ± 0.17a	0.53 ± 0.06bcd	0.60 ± 0.08cd	0.80 ± 0.08bc	0.58 ± 0.08bcd	0.68 ± 0.06b
T. vogelii (5%)	0.68 ± 0.11a	0.36 ± 0.03def	0.38 ± 0.08def	0.40 ± 0.04def	0.44 ± 0.06bcde	0.35 ± 0.06cdef
T. vogelii (10%)	0.71 ± 0.13a	0.23 ± 0.04fg	0.25 ± 0.05efg	0.19 ± 0.02ef	0.20 ± 0.05ef	0.18 ± 0.03def
Mixed plants (2.5%)	0.74 ± 0.15a	0.30 ± 0.06ef	0.25 ± 0.04fg	0.39 ± 0.07def	0.30 ± 0.05def	0.25 ± 0.04def
Mixed plants (5%)	**0.79 ± 0.18a**	**0.10 ± 0.04g**	**0.14 ± 0.04fg**	**0.10 ± 0.03f**	**0.10 ± 0.03f**	**0.09 ± 0.03ef**
3 - way ANOVA	**(F- Statistics)**					
Season (S)	179.27***	4.58*	35.57***	59.32***	36.10***	27.57***
Location (L)	8.68***	10.62**	1.73ns	0.87ns	2.32ns	17.78***
Treatments (T)	2.92ns	28.44***	28.95***	44.26***	58.81***	84.51***
S*L	7.59***	15.79***	15.58***	6.98**	7.86**	9.14**
S*T	1.16ns	0.92ns	3.13***	6.89***	6.32***	6.64***
L*T	1.01ns	1.11ns	0.38ns	1.02ns	0.93ns	1.74ns
S*L*T	1.53ns	1.50ns	1.67ns	2.70**	0.72ns	1.80ns

Each value is a mean ± standard error of sixteen replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fisher's Least Significant Difference (LSD) test.

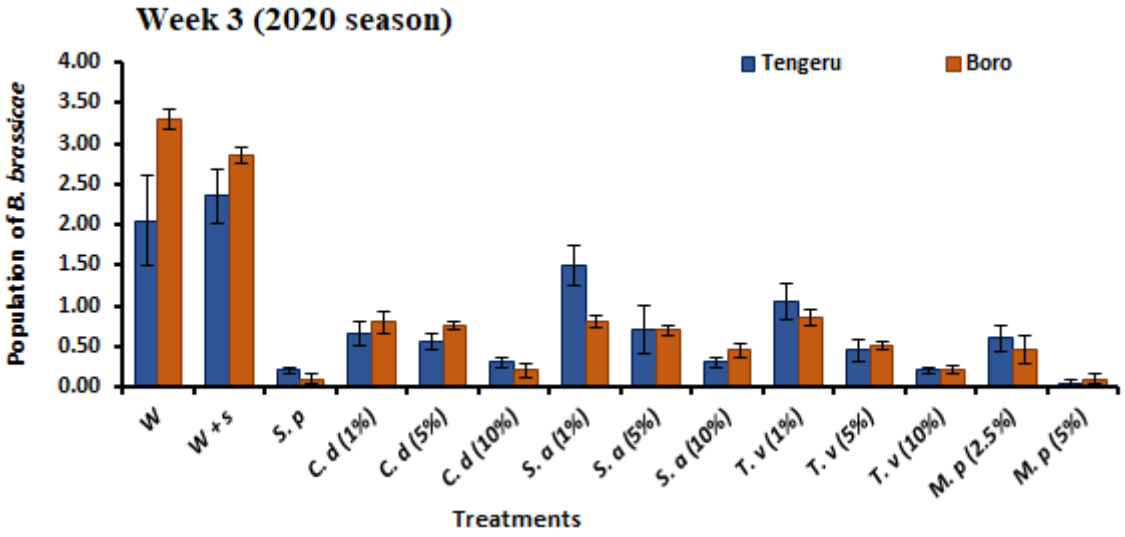
The interactions among the weather conditions (temperatures and rainfall precipitations) of experimental sites, treatments and seasons significantly ($P \leq 0.05$) affected the population abundance of the *B. brassicae* compared with the negative controls in the plots (Table 11 and 12; Fig. 14, 15 and 16). The interaction (Fig. 14) of the weather conditions (rainfall and temperatures) of the experimental sites in 2019 wet season and in 2020 wet season (Fig. 15) were observed on week 5 and week 3 after treatment applications (Appendix 1), respectively. It was observed that, the population abundance of *B. brassicae* in 2019 wet season decreased in the treated plots relatively to the negative controls (water and water plus soap). At Tengeru experimental site, the population abundance of *B. brassicae* was relatively higher compared with Boro experimental site in 2019 wet season (Fig. 14). In 2020 wet season the population of *B. brassicae* was dynamic compared with 2019 wet season (Fig. 15). The study revealed that, in 2019 season, the population of *B. brassicae* was lower compared with 2020 season (Fig.

16). Moreover, Table 12 indicates trends of the dynamic and the interactions of the population of *B. brassicae* (Fig. 16) in the two wet seasons (2019 and 2020) in the 2nd, 3rd, 4th and 5th weeks after the treatments. Generally, it was observed that, the population of *B. brassicae* was significantly lower in 2019 season relative to 2020 season at both experimental sites (Fig. 16) except in the 2nd and the 6th week of the experiment, whereby the population abundance of *B. brassicae* at both wet seasons at Tengeru experimental site had no significant variations (Fig. 16). Also, there was higher population abundance of *B. brassicae* in 2020 season compared with 2019 season (Fig. 17).



W - water, w + s - Water plus soap, S. p - Synthetic pesticide, *C. d – Croton dichogamus*, *S. a – Syzygium aromaticum*, *T. v – Tephrosia vogelii* and M. p – Mixed plants

Figure 14: Interaction of weather conditions of the study sites and the treatments on lowering the population of *B. brassicae* 2019 (Week 5)



W - water, w + s - Water plus soap, S. p - Synthetic pesticide, *C. d – Croton dichogamus*, *S. a – Syzygium aromaticum*, *T. v – Tephrosia vogelii* and M. p – Mixed plants

Figure 15: Interactions of weather conditions of the study sites and the treatments on the population of *B. brassicae* 2020 (Week 3)

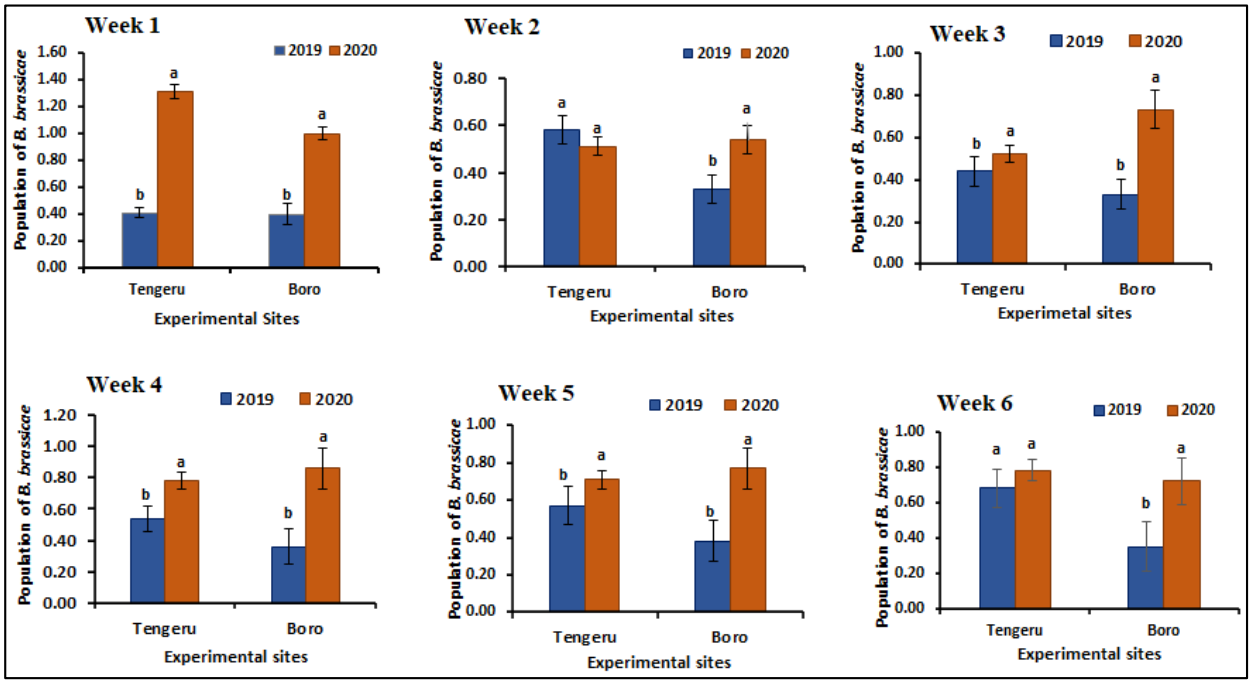
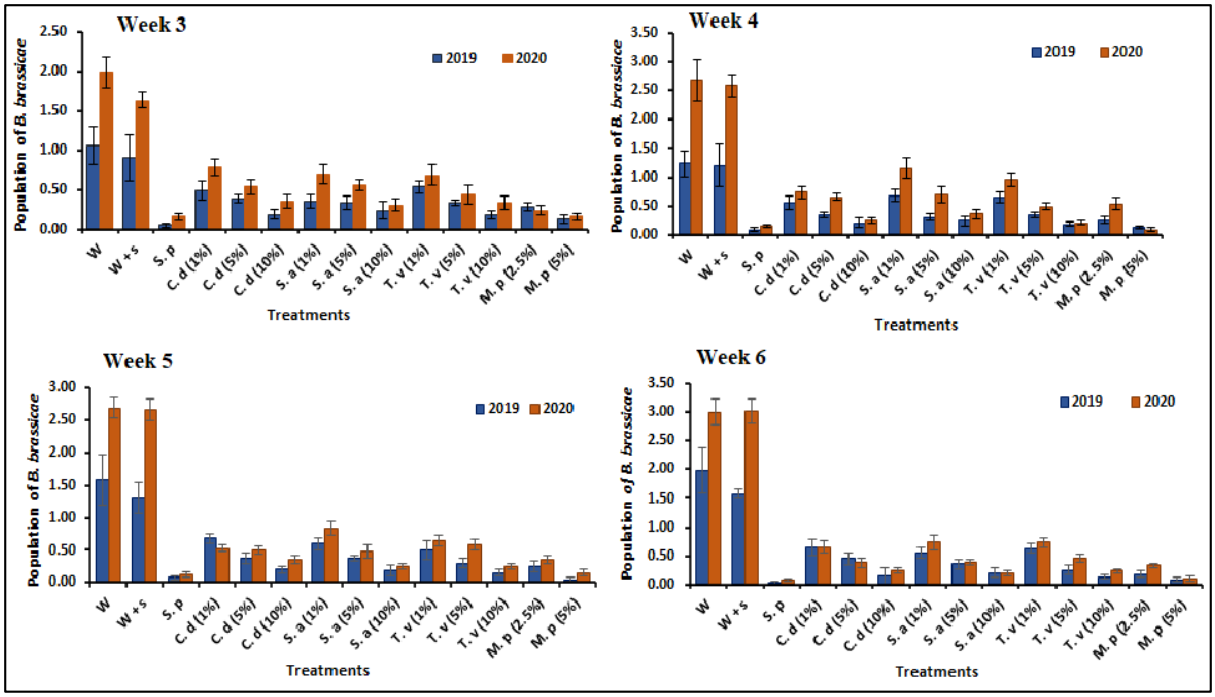


Figure 16: Interaction of weather conditions of the experimental sites and seasons on the population abundance of *B. brassicae* in 2019 and 2020 wet seasons



W - water, w + s - Water plus soap, S. p - Synthetic pesticide, *C. d – Croton dichogamus*, *S. a – Syzygium aromaticum*, *T. v – Tephrosia vogelii* and M. p – Mixed plants

Figure 17: Interaction of seasons and treatments on the population abundance of *B. brassicae* (Weeks 3, 4, 5 and 6)

Population dynamics of M. persicae in response to the treatments

M. persicae was identified infesting the cabbage (*B. oleracea*) two weeks after transplanting and the mean population increased progressively week after week (Table 14). Table 13, indicates the abundance of *M. persicae* in response to the treatments used in the two experimental sites in 2019 and 2020 wet seasons per cabbage crop. It was observed that the mean population abundance of *M. persicae* was significantly ($P \leq 0.05$) the same in the two experimental sites (Table 13) in both 2019 and 2020 wet seasons.

Moreover, the study revealed that, the treatments differed significantly ($P \leq 0.05$) in reduction of the population of *M. persicae* in the field plots (Table 13) in both wet seasons. The 5% concentration of the aqueous extract from the mixed plants possessed significantly ($P \leq 0.01$) lower (0.12 ± 0.03 and 0.16 ± 0.04) population abundance of *M. persicae* as in synthetic pesticide (chlorpyrifos) (0.21 ± 0.10 and 0.11 ± 0.03) treated plots in 2019 and 2020 wet seasons, respectively (Table 13). Then, it was followed by the 10% of the aqueous extracts from *S. aromaticum* and *T. vogelii* in 2019 and 2020 wet seasons for both experimental sites. However, the 10% concentration of *C. dichogamus* and 1% and 5% of the aqueous extracts of *C. dichogamus, T. vogelii* and *S. aromaticum* of individual plants and 2.5% concentration of

the aqueous extracts from the mixed plants significantly lowered the population abundance of *M. persicae* compared with negative controls for both 2019 and 2020 wet seasons (Table 13).

In addition, Table 14 indicates weekly observations of the population abundance of *M. persicae* at the two experimental sites in both wet seasons of 2019 and 2020 years. The results revealed that, the mean population abundance of *M. persicae* was significantly ($p \leq 0.01$) higher (1.05 ± 0.05, 0.67 ± 0.07, 0.76 ± 0.07, 0.76 ± 0.08) in 2020 wet season compared with 2019 wet season (0.87 ± 0.05, 0.49 ± 0.06, 0.51 ± 0.07, 0.57 ± 0.09) in week 1 before application of treatments and weeks 3, 4 and 5 after application of treatments, respectively. However, in week 1 after application of the treatments, the population was significantly ($p \leq 0.01$) higher (0.66 ± 0.06) in 2019 wet season compared with 2020 wet season (0.52 ± 0.04). In week 2 after application of the treatments, the mean population of *M. persicae* was significantly the same between the two seasons. In addition, the mean population of *M. persicae* was significantly ($P \leq 0.01$) higher (1.15 ± 0.06 and 0.74 ± 0.04) at Tengeru experimental site than at Boro experimental site (0.76 ± 0.09 and 0.59 ± 0.04) in week 1 before treatment and week 5 after treatment applications in both wet seasons, respectively (Table 14). However, on weeks 1, 2, 3 and 4 after treatment applications, the population of *M. persicae* was statistically the same at the two experimental sites in both wet seasons (Table 14). The present study revealed that the aqueous extracts applied were effective against *M. persicae* in both seasons. On week 1 before application of the treatments, the mean population of *M. persicae* on different plots was random and was insignificant ($p > 0.05$) among the plots in the field. However, at the 1^{st}, 2^{nd}, 3^{rd}, 4^{th}, and 5^{th} week of application of the treatments, the population of *M. persicae* differed significantly (≤ 0.05) among the treated plots (Table 14).

Moreover, the study revealed that the 5% concentration of the aqueous extracts from the mixed plants possessed significantly ($p \leq 0.01$) lower mean population of *M. persicae* (0.20 ± 0.04, 0.10 ± 0.03, 0.12 ± 0.03, 0.14 ± 0.04, 0.13 ± 0.04) compared with other treatments on weeks 1, 2, 3, 4 and 5 of the treatments, respectively. It was significantly effective as synthetic pesticide (chlorpyrifos) (0.14 ± 0.04, 0.18 ± 0.04, 0.18 ± 0.07, 0.21 ± 0.07, 0.18 ± 0.05) on weeks 1, 2, 3, 4 and 5 of the treatments, respectively. Then it was followed by 10% concentrations of the aqueous extracts of the three pesticidal plants used in this study. The 1% and 5% of the aqueous extracts of *C. dichogamus, T. vogelii and S. aromaticum* and 2.5% concentration of the aqueous extracts from the mixed plants significantly lowered the

population of *M. persicae* relative to water alone and water plus soap for both seasons (Table 14).

Moreover, the results of weekly observations when each season is considered separately, revealed that, the population abundance of the *M. persicae* was significantly ($p \leq 0.01$) higher (1.04 ± 0.07, 0.74 ± 0.09, 0.64 ± 0.11) at Tengeru experimental site than (0.64 ± 0.06, 0.57 ± 0.08, 0.47 ± 0.47) at Boro experimental site on week 1 before treatment applications, week 1 and 2 after treatment application in 2019 wet season (Appendix 2). However, on weeks 3, 4 and 5 after treatment applications, the population abundance of *M. persicae* was significantly ($P \leq 0.01$) the same in 2019 wet season between the two experimental sites (Appendix 2). In 2020 wet season, the population abundance of *M. persicae*, was significantly ($P \leq 0.05$) the same on week 1 before treatment applications, week 1 and week 3 after treatment applications (Appendix 2). On weeks 2, 4 and 5 the population abundance of *M. persicae* varies significantly ($P \leq 0.05$) from one experimental site to another (Appendix 2).

Table 13: Mean number of *M. persicae* per cabbage crop in response to application of treatments

Location and	**Seasons**	
Treatments	**2019 season**	**2020 season**
Location		
Tengeru	0.59 ± 0.11a	0.67 ± 0.08a
Boro	0.51 ± 0.08a	0.63. ± 0.09a
Treatments		
Water	2.27 ± 0.25a	2.07 ± 0.12a
water + soap	2.90 ± 0.24a	2.05 ± 0.09a
Synthetic pesticide	0.21 ± 0.10de	0.11 ± 0.03g
C. dichogamus (1%)	0.64 ± 0.08c	0.57 ± 0.05bc
C. dichogamus (5%)	0.35 ± 0.03de	0.48 ± 0.03cd
C. dichogamus (10%)	0.39 ± 0.08cde	0.33 ± 0.03efg
S. aromaticum (1%)	0.44 ± 0.04cd	0.79 ± 0.09b
S. aromaticum (5%)	0.32 ± 0.05de	0.51 ± 0.04cd
S. aromaticum (10%)	0.23 ± 0.03de	0.33 ± 0.04efg
T. vogelii (1%)	0.32 ± 0.02de	0.72 ± 0.05b
T. vogelii (5%)	0.21 ± 0.04de	0.44 ± 0.04de
T. vogelii (10%)	0.15 ± 0.02de	0.23 ± 0.02fg
Mixed plants (2.5%)	0.20 ± 0.05de	0.36 ± 0.06def
Mixed plants (5%)	0.12 ± 0.03e	0.16 ± 0.04g
2 - way ANOVA	**(F- Statistics)**	
Locations	2.51ns	2.47ns
Treatments	42.19***	132.71***
Location*treatments	1.18ns	2.18ns

Each value is a mean ± standard error of eight replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fisher's Least Significant Difference (LSD) test.

Table 14: Mean number of *M. persicae* per crop in response to weekly application of treatments

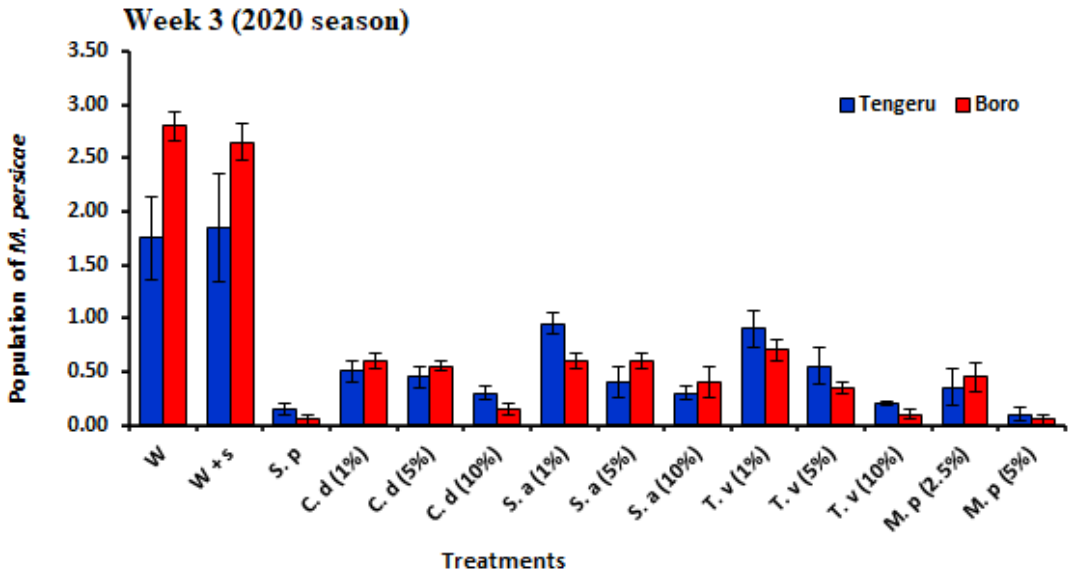
Location and Treatments	Week 1 before Treatment	Weeks after treatments				
		1	2	3	4	5
Seasons						
Wet Season 1 (2019)	0.87 ± 0.05b	0.66 ± 0.06a	0.55 ± 0.06a	0.49 ± 0.06b	0.51 ± 0.08b	0.57 ± 0.09b
Wet Season 2 (2020)	1.05 ± 0.05a	0.52. ± 0.04b	0.54 ± 0.05a	0.67 ± 0.07a	0.76 ± 0.07a	0.76 ± 0.08a
Locations						
Tengeru	1.15 ± 0.05a	0.62 ± 0.05a	0.55 ± 0.06a	0.58 ± 0.07a	0.68 ± 0.08a	0.74 ± 0.09a
Boro	0.76 ± 0.04b	0.55. ± 0.05a	0.54 ± 0.05a	0.57 ± 0.07a	0.59 ± 0.07a	0.59 ± 0.08b
Treatments						
W	1.165± 0.09a	1.71 ± 0.17a	1.86 ± 0.19a	2.15 ± 0.16a	2.44 ± 0.21a	2.67 ± 0.18a
W + s	0.89 ± 0.11a	1.54 ± 0.15a	1.42 ± 0.14ab	1.93 ± 0.23a	2.21 ± 0.16a	2.76 ± 0.14a
S. p	1.08 ± 0.15a	0.14 ± 0.04f	0.18 ± 0.09fgh	0.18 ± 0.07ef	0.21 ± 0.07cde	0.18 ± 0.05ef
C. d (1%)	1.13 ± 0.15a	0.71 ± 0.08b	0.64 ± 0.12cd	0.54 ± 0.07bcd	0.60 ± 0.07b	0.54 ± 0.06bc
C. d (5%)	0.83 ± 0.13a	0.45 ± 0.04cd	0.47 ± 0.05cde	0.39 ± 0.06cde	0.39 ± 0.05bcde	0.36 ± 0.05cde
C. d (10%)	0.96 ± 0.13a	0.46 ± 0.04cd	0.40 ± 0.08cdef	0.20 ± 0.05ef	0.38 ± 0.07bcde	0.35 ± 0.05cdef
S. a (1%)	0.98 ± 0.10a	0.60 ± 0.08bc	0.56 ± 0.07cd	0.61 ± 0.07b	0.63 ± 0.12b	0.66 ± 0.09b
S. a (5%)	0.81 ± 0.11a	0.45 ± 0.06cd	0.45 ± 0.06cde	0.39 ± 0.06bcde	0.46 ± 0.10bcd	0.31 ± 0.05cdef
S. a (10%)	0.94 ± 0.12a	0.36 ± 0.04def	0.31 ± 0.06efgh	0.28 ± 0.05def	0.20 ± 0.05de	0.24 ± 0.04ef
T. v (1%)	0.88 ± 0.15a	0.50 ± 0.07bcd	0.58 ± 0.06cd	0.55 ± 0.09bc	0.49 ± 0.09bc	0.48 ± 0.10bcd
T. v (5%)	0.93 ± 0.16a	0.41 ± 0.07cde	0.35 ± 0.05defg	0.33 ± 0.07cdef	0.29 ± 0.06bcde	0.24 ± 0.05ef
T. v (10%)	1.04 ± 0.16a	0.29 ± 0.04def	0.15 ± 0.02fgh	0.16 ± 0.03ef	0.16 ± 0.04f	0.17 ± 0.04ef
M. p. (2.5%)	0.88 ± 0.12a	0.38 ± 0.08cde	0.20 ± 0.03fgh	0.30 ± 0.07cdef	0.26 ± 0.07cde	0.25 ± 0.07def
M. p. (5%)	0.96 ± 0.10a	0.20 ± 0.04ef	0.10 ± 0.03h	0.12 ± 0.03f	0.14 ± 0.04f	0.13 ± 0.04f
3 - way ANOVA	**(F- Statistics)**					
Season (S)	8.40**	10.86**	0.03ns	15.59***	26.84***	24.98***
Location (L)	36.90***	3.15ns	0.00ns	0.06ns	3.33ns	15.91***
Treatments (T)	0.77ns	34.63***	37.92***	54.88***	64.37***	146.59***
S * L	0.07ns	4.83***	14.93***	4.77*	9.39**	7.82**
S * T	0.31ns	2.46**	1.32ns	1.61ns	1.42ns	1.98ns
L * T	0.66ns	0.34ns	0.74ns	0.89ns	0.74ns	1.01ns
S * L * T	0.86ns	0.90ns	2.67**	2.37ns	0.79ns	1.76ns

Each value is a mean ± standard error of sixteen replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fisher's Least Significant Difference (LSD) test.

W - water, w + s - Water plus soap, S. p - Synthetic pesticide, *C. d – Croton dichogamus, S. a – Syzygium aromaticum, T. v – Tephrosia vogelii,* M. p – Mixed plants

The interactions among the experimental sites' weather conditions, treatments and seasons significantly ($P \leq 0.05$) affected the population abundance of *M. persicae* compared with the negative controls in the plots (Table 14; Appendix 2; Fig. 18, 19 and 20). The interaction between treatments and experimental sites was only significant ($P \leq 0.05$) in the 4^{th} week of experiment in 2020 season (Appendix 2; Fig. 18) but the other weeks of the study was insignificant ($p > 0.05$) in both seasons 2019 and 2020. Figure 18 shows that, the interactions of Synthetic pesticides, *C. dichogamus* (10%), *S. aromaticum* (1%), *T. vogelii* (at all tested concentrations) and 5% of the mixed plants and the experimental sites' weather conditions (rainfall precipitations and temperatures) and the natural enemies' proliferations significantly enhanced the reduction of the population of *M. persicae* in the plots on week 3 in 2020 season (Fig. 18).

Moreover, the interactions of weather conditions of the experimental sites and seasons enhanced the reduction of *M. persicae* at the field (Fig. 19). On weeks 2 and 3, *M. persicae* was lower in 2020 season compared with 2019 season at Tengeru experimental site while on weeks 4, 5 and 6 the population of *M. persicae* was lower in 2019 season at Tengeru experimental site. But, at Boro experimental site, *M. persicae* was lower in 2019 season compared with 2020 season on weeks 3, 4, 5 and 6, except week 2 during the experiments. *M. persicae* mighty have been favoured by low rainfall precipitation and temperatures in 2020 wet season at Boro experimental site. Similarly, the three-way ANOVA comparisons of the population abundance of *M. persicae* was significantly different ($P \leq 0.05$) on weeks 2, 3, 4, 5 and 6 of the experiment in 2019 and 2020 wet season (Appendix 2; Fig. 19). The interactions among experimental sites' weather conditions, treatments and seasons significantly ($P \leq 0.05$) affected the population abundance of the *M. persicae* compared with the negative controls (water and water plus soap) in the plots on week 2 after treatments applications (Table 14; Fig. 20).



W - water, w + s - Water plus soap, S. p - Synthetic pesticide, *C. d – Croton dichogamus*, *S. a – Syzygium aromaticum*, *T. v – Tephrosia vogelii*, M. p – Mixed plants

Figure 18: Interactions of weather conditions of the study sites and the treatments on lowering of population of *M. persicae* 2020 (Week 3)

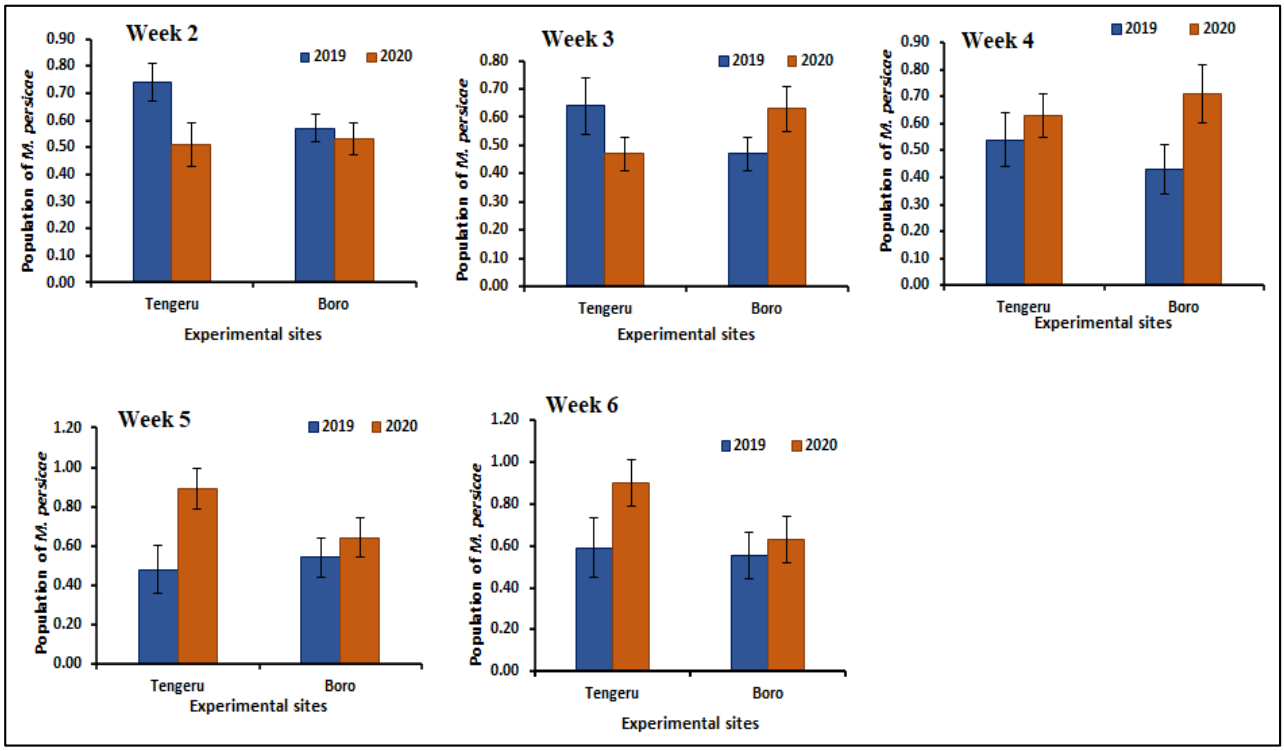
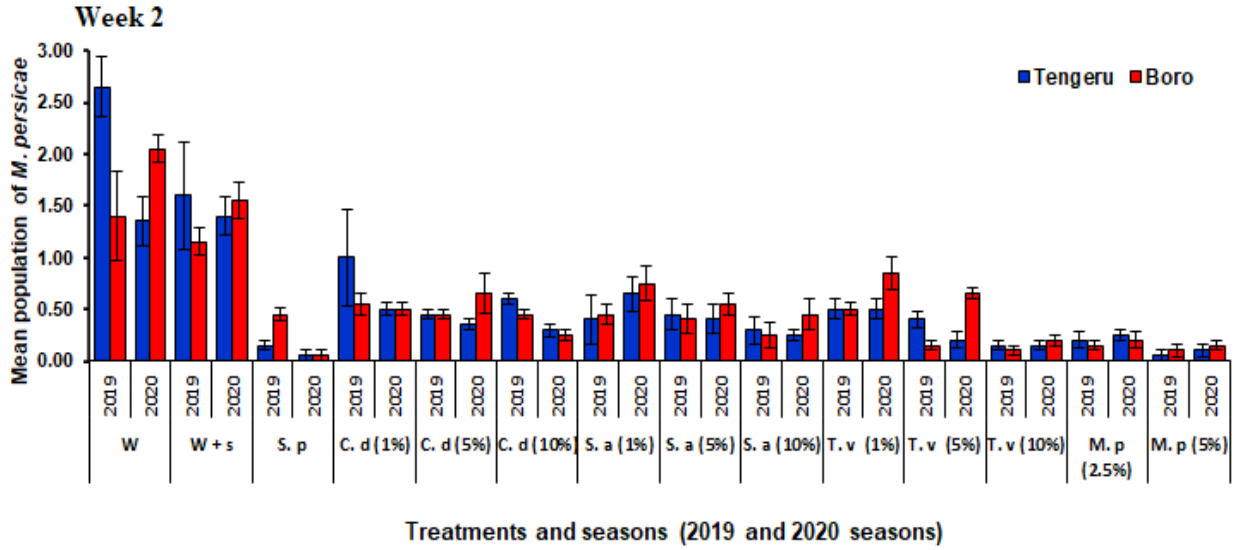


Figure 19: Interactions of weather conditions of the experimental sites and the seasons on the mean population abundance of *M. persicae* for 2019 and 2020 seasons (Week 3)



W - water, w + s - Water plus soap, S. p - Synthetic pesticide, *C. d – Croton dichogamus, S. a – Syzygium aromaticum, T. v – Tephrosia vogelii,* M. p – Mixed plants

Figure 20: Interactions of weather conditions of the experimental sites, seasons and the treatments on the mean population of *M. persicae* for 2019 and 2020 wet seasons (Week 2)

Population dynamics of P. xylostella in response to the treatments

The mean population abundance of *P. xylostella* for two wet seasons 2019 and 2020 is presented in Table 15. In general, there was significant difference ($P \leq 0.05$) in population abundances between the two experimental sites Tengeru and Boro in the 2019 wet season, while in 2020 wet season, the population abundance of *P. xylostella* was insignificant (Table 15).

The insecticidal efficacy of the treatments differed significantly ($P \leq 0.05$) from one treatment to another in the field. It was observed that the aqueous extracts of the pesticidal plants used in this study were significantly ($P \leq 0.05$) effective as synthetic pesticide in the first wet season 2019. But in 2020 wet season, the 5% of aqueous extract from the mixed plants was significantly ($P \leq 0.05$) effective as synthetic pesticide in lowering the population abundance of *P. xylostella* in the field (Table 15). It was revealed that, the mean population abundance of *P. xylostella* in 5% concentration of aqueous extract of the mixed plants had significantly ($P \leq 0.05$) lower (0.07 ± 0.02 and 0.20 ± 0.01) population of *P. xylostella* as synthetic pesticide (0.10 ± 0.04 and 0.20 ± 0.03) in 2019 and 2020 wet seasons, respectively. This was followed by the 10% concentrations of the aqueous extracts of *C. dichogamus, T. vogelii* and *S. aromaticum.* But, the other concentrations (1% and 5 %) of the aqueous extracts of *C.*

dichogamus, T. vogelii and *S. aromaticum* and 2.5% of the aqueous extracts from the mixed plants significantly lowered the population abundance of *P. xylostella* compared with negative controls for both 2019 and 2020 seasons (Table 15).

Moreover, the weekly observations found that, the mean population abundance of *P. xylostella* was significantly ($P \leq 0.05$) lower (0.08 ± 0.01, 0.23 ± 0.05, 0.41 ± 0.08) in 2019 wet season compared with 2020 wet season (0.45 ± 0.03, 0.37 ± 0.03, 0.56 ± 0.06) on week 1 before treatment applications, week 1 and 4 after treatment applications, respectively (Table 16). But the mean population abundance of *P. xylostella* was insignificant on weeks 2, 3 and 5 after treatment applications in both wet seasons (Table 16). It was also, observed that, the population abundance of *P. xylostella* was significantly higher (0.37 ± 0.05, 0.49 ± 0.04, 0.67 ± 0.08, 0.58 ± 0.08 and 0.67 ± 0.11) at Boro experimental site compared with Tengeru experimental site (0.23 ± 0.03, 0.25 ± 0.04, 0.34 ± 0.05, 0.39 ± 0.06, and 0.54 ± 0.07) from week 1 to week 5 after the treatment applications, respectively (Table 16).

Similarly, the insecticidal efficacy of the treatments differed significantly ($P \leq 0.05$) from one treatment plot to another in the field. It was discovered that the aqueous extracts of *C. dichogamus, T. vogelii,* and *S. aromaticum* were significantly ($P \leq 0.05$) effective as synthetic pesticide in 2019 wet seasons. But, in 2020 wet season, the 5% concentration of aqueous extract from the mixed plants was as effective as synthetic pesticide in controlling *P. xylostella* (Table 16). It was revealed that, the mean population abundance of *P. xylostella* in 5% concentration of aqueous extract of the mixed plants possessed significantly ($P \leq 0.05$) lower (0.10 ± 0.04, 0.08 ± 0.03, 0.09 ± 0.03, 0.06 ± 0.02 and 0.10 ± 0.03) population abundance of *P. xylostella* compared with other treatments on weeks 1, 2, 3, 4 and 5 after treatment applications, respectively (Table 16). The population abundance of *P. xylostella* in 5% concentration of aqueous extract of the mixed plants was as lower as in chlorpyrifos (0.10 ± 0.03, 0.07 ± 0.04, 0.21 ± 0.08, 0.11 ± 0.03 and 0.13 ± 0.04) on weeks 1, 2, 3, 4 and 5 after treatment applications, respectively (Table 16). It was followed by 10% concentrations of the aqueous extracts of *C. dichogamus, T. vogelii* and *S. aromaticum.* But, the other concentrations (1% and 5%) of the aqueous extracts of *C. dichogamus, T. vogelii* and *S. aromaticum* and 2.5% of the aqueous extracts from the mixed plants had significantly lower population abundance of *P. xylostella* compared with negative controls (Table 16). Moreover, the weekly trends observations in each wet season are indicated in Appendix 3.

Table 15: Mean number of *P. xylostella* per cabbage in response to treatments application

Location and Treatments	Mean ± SE	
	2019 season	2020 season
Location		
Tengeru	0.23 ± 0.06b	0.46 ± 0.05a
Boro	0.62 ± 0.13a	0.49. ± 0.05a
Treatments		
Water	2.11 ± 0.43a	1.32 ± 0.10a
water + soap	2.01 ± 0.27a	1.29 ± 0.12a
Synthetic pesticide	0.10 ± 0.04b	0.20 ± 0.03f
C. dichogamus (1%)	0.28 ± 0.08b	0.50 ± 0.04bc
C. dichogamus (5%)	0.22 ± 0.06b	0.38 ± 0.04cde
C. dichogamus (10%)	0.14 ± 0.03b	0.25 ± 0.03ef
S. aromaticum (1%)	0.23 ± 0.06b	0.52 ± 0.04b
S. aromaticum (5%)	0.17 ± 0.06b	0.37 ± 0.03cde
S. aromaticum (10%)	0.11 ± 0.03b	0.28 ± 0.02def
T. vogelii (1%)	0.17 ± 0.03b	0.41 ± 0.02bcd
T. vogelii (5%)	0.14 ± 0.04b	0.35 ± 0.02de
T. vogelii (10%)	0.10 ± 0.03b	0.24 ± 0.02ef
Mixed plants (2.5%)	0.12 ± 0.04b	0.32 ± 0.03def
Mixed plants (5%)	0.07 ± 0.02b	0.20 ± 0.01f
2 - way ANOVA	**(F- Statistics)**	
Locations	97.46***	1.32ns
Treatments	84.79***	50.78***
Location*treatments	13.28***	0.43ns

Each value is a mean ± standard error of eight replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fisher's Least Significant Difference (LSD) test.

Table 16: Mean of *P. xylostella* per cabbage crop in response to weekly application of treatments in two seasons

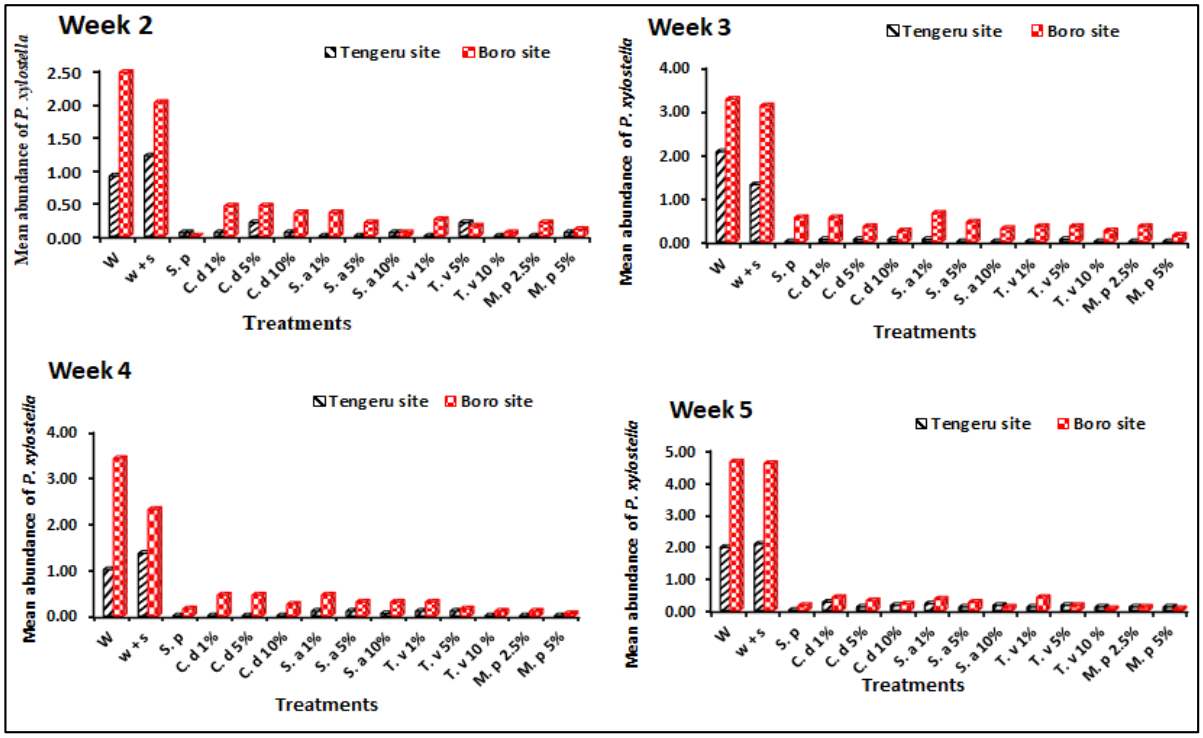
Location and Treatments	Week 1 before Treatment	Weeks after treatments				
		1	2	3	4	5
Seasons						
Wet season 1 (2019)	0.08 ± 0.01b	0.23 ± 0.05b	0.35 ± 0.06a	0.52 ± 0.09a	0.41 ± 0.08b	0.61 ± 0.12a
Wet season 2 (2020)	0.45 ± 0.03a	0.37. ± 0.03a	0.39 ± 0.03a	0.49 ± 0.05a	0.56 ± 0.06a	0.59 ± 0.07a
Locations						
Tengeru	0.23 ± 0.03b	0.23 ± 0.03b	0.25 ± 0.04b	0.34 ± 0.05b	0.39 ± 0.06b	0.54 ± 0.07b
Boro	0.29 ± 0.03a	0.37. ± 0.05a	0.49 ± 0.04a	0.67 ± 0.08a	0.58 ± 0.08a	0.67 ± 0.11a
Treatments						
W	0.30± 0.06a	1.03 ± 0.27a	1.41 ± 0.22a	2.04 ± 0.23a	2.06 ± 0.29a	2.60 ± 0.37a
W + s	0.28 ± 0.10a	0.90 ± 0.10a	1.26 ± 0.18a	1.91 ± 0.25a	1.71 ± 0.20b	2.74 ± 0.32a
S. p	0.24 ± 0.08a	0.10 ± 0.03c	0.07 ± 0.04e	0.21 ± 0.08bcd	0.11 ± 0.03cd	0.13 ± 0.04c
C. d (1%)	0.26 ± 0.06a	0.35 ± 0.08b	0.36 ± 0.06b	0.41 ± 0.07bc	0.41 ± 0.09c	0.45 ± 0.07b
C. d (5%)	0.28 ± 0.07a	0.23 ± 0.06bc	0.35 ± 0.05bc	029 ± 0.06bcd	0.34 ± 0.04cd	0.25 ± 0.05bc
C. d (10%)	0.18 ± 0.06a	0.16 ± 0.04bc	0.18 ± 0.05bcde	0.20 ± 0.03bcd	0.18 ± 0.04cd	0.25 ± 0.05bc
S. a (1%)	0.36 ± 0.09a	0.25 ± 0.06bc	0.33 ± 0.07bcd	0.46 ± 0.08b	0.38 ± 0.06c	0.46 ± 0.06b
S. a (5%)	0.23 ± 0.07a	0.23 ± 0.05bc	0.21 ± 0.04bcde	0.29 ± 0.05bcd	0.35 ± 0.05cd	0.26 ± 0.04bc
S. a (10%)	0.28 ± 0.08a	0.13 ± 0.04bc	0.15 ± 0.03bcde	0.18 ± 0.04bcd	0.19 ± 0.04cd	0.20 ± 0.04bc
T. v (1%)	0.15 ± 0.06a	0.20 ± 0.04bc	0.23 ± 0.04bcde	0.33 ± 0.06bcd	0.39 ± 0.09c	0.38 ± 0.07bc
T. v (5%)	0.25 ± 0.08a	0.15 ± 0.04bc	0.24 ± 0.05bcde	0.28 ± 0.05bcd	0.29 ± 0.06cd	0.25 ± 0.04bc
T. v (10%)	0.28 ± 0.08a	0.16 ± 0.05bc	0.11 ± 0.03cde	0.15 ± 0.05cd	0.11 ± 0.03cd	0.14 ± 0.03c
M. p. (2.5%)	0.30 ± 0.06a	0.22 ± 0.05bc	0.19 ± 0.04bcde	0.23 ± 0.05bcd	0.19 ± 0.05cd	0.23 ± 0.05bc
M. p. (5%)	0.31 ± 0.08a	0.10 ± 0.04c	0.08 ± 0.03de	0.09 ± 0.03d	0.06 ± 0.02d	0.10 ± 0.03c
3 - way ANOVA	**(F- Statistics)**					
Season (S)	158.25***	10.19**	1.09ns	0.52ns	10.00***	0.15ns
Location (L)	4.15*	10.74**	42.51***	67.60***	17.34***	8.10**
Treatments (T)	0.95ns	13.12***	39.19***	72.95***	49.81***	103.31***
S * L	6.33*	13.67***	3.77ns	24.69***	25.90***	44.79**
S * T	0.50ns	0.53ns	3.52ns	6.93***	1.33ns	9.11***
L * T	1.07ns	1.70ns	5.10**	4.35***	3.47***	5.60***
S * L * T	1.01ns	2.50ns	1.13ns	1.41ns	3.19ns	8.43***

Each value is a mean ± standard error of sixteen replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fisher's Least Significant Difference (LSD) test.

W - water, w + s - Water plus soap, S. p - Synthetic pesticide, *C. d – Croton dichogamus*, *S. a – Syzygium aromaticum*, *T. v – Tephrosia vogelii*, M. p – Mixed plants

The interactions among experimental sites' weather conditions, treatments and seasons significantly ($P \leq 0.05$) affected the population abundance of *P. xylostella* compared with the negative controls (water and water plus soap) in the plots (Table 16; Appendix 3; Fig. 21, 22 and 23). The interaction between treatments and experimental sites was significant ($P \leq 0.05$) in weeks 2, 3, 4, and 5 of the study periods in 2019 wet season (Appendix 3; Fig. 21 and 22) but the other weeks of the study was insignificant ($P > 0.05$) in both wet seasons 2019 and 2020. It was observed that the population abundance of *P. xylostella* in 2019 wet season significantly decreased in the treated plots relative to the negative controls (water and water plus soap) (Appendix 3; Fig. 21 and 23).

There was significant ($P \leq 0.05$) interaction of experimental sites' weather conditions and seasons on weeks 1, 2, 4, 5 and 6 of the treatment experiments (Table 16; Fig. 22). It was observed that, the population abundance of the *P. xylostella* was significantly affected by weather conditions and seasons on weeks 1, 2, 4, 5 and 6 compared with the negative controls in the plots (Table 16; Fig. 22). The interactions of treatments and seasons significantly ($P \leq 0.05$) affected the population abundance of *P. xylostella* compared with the negative controls (water and water plus soap) in the plots (Table 16; Fig. 23) in weeks 3 and 5 after treatments (Table 16; Fig. 23). Moreover, the interactions of experimental sites' weather conditions, treatments and seasons significantly ($P \leq 0.05$) affected the population abundance of *P. xylostella* compared with the negative controls (water and water plus soap) in the plots on week 6 of the experimental treatments (Table 19; Fig. 23). The proliferation and high density of natural enemies and weather conditions mighty have contributed to the higher insect pests at one season and experimental sites compared with the other season and the other experimental site. For instance, in 2019 wet season, there was high rain precipitation at Boro experimental site compared with Tengeru experimental site which mighty have contributed to the higher population of *P. xylostella* at Boro compared to Tengeru experimental site.



W - water, w + s - Water plus soap, S. p - Synthetic pesticide, *C. d – Croton dichogamus*, *S. a – Syzygium aromaticum*, *T. v – Tephrosia vogelii*, M. p – Mixed plants

Figure 21: Interactions of weather conditions of experimental sites and the treatments on reduction of abundance of *P. xylostella* larvae (Week 2, 3, 4 and 5 after treatments) in 2019 season

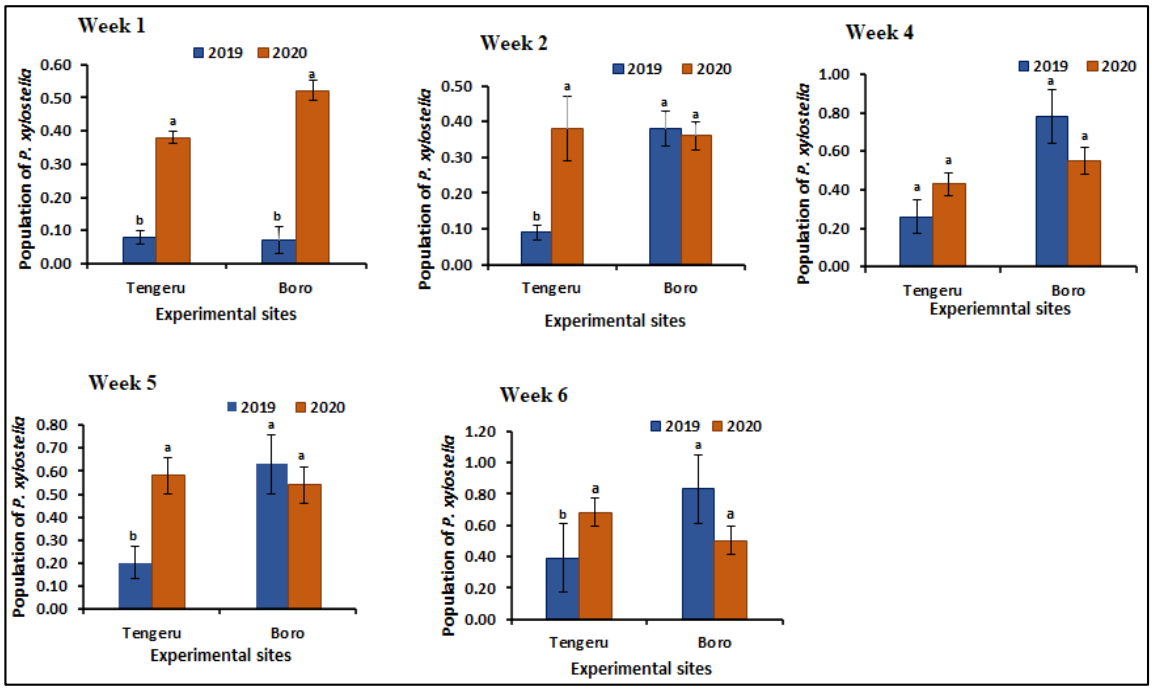
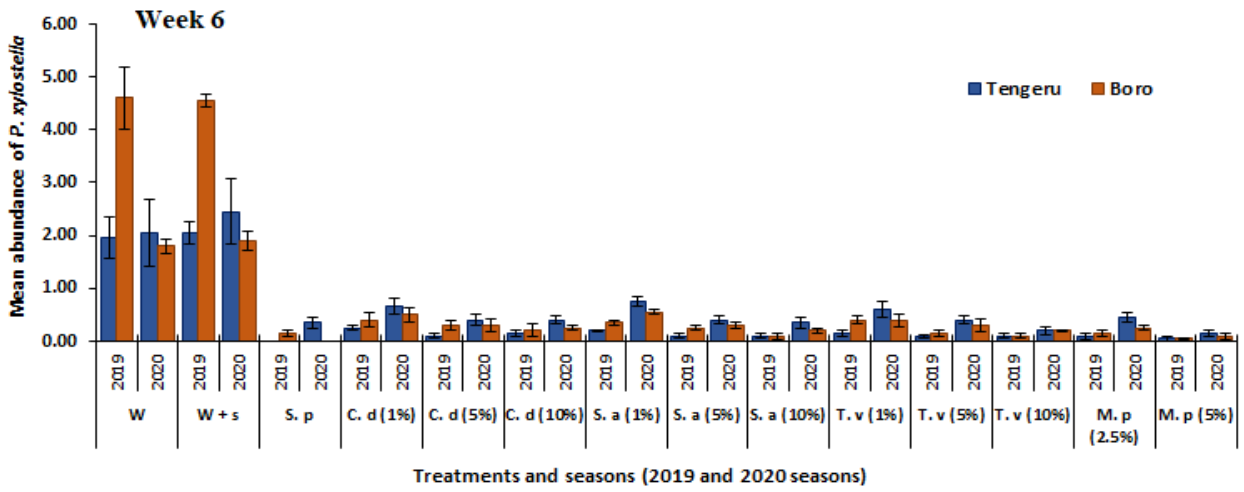


Figure 22: Interactions of weather conditions of experimental sites and the seasons for reduction of abundance of *P. xylostella* larvae (Week 1, 2, 4, 5 and 6) in 2019 and 2020 season



W - water, w + s - Water plus soap, S. p - Synthetic pesticide, *C. d – Croton dichogamus*, *S. a – Syzygium aromaticum*, *T. v – Tephrosia vogelii*, M. p – Mixed plants

Figure 23: Interactions of experimental sites, seasons and the treatments for reduction of abundance of *P. xylostella* larvae in 2019 and 2020 season

Population dynamics of H. undalis in response to the treatments

Generally, the population abundance of *H. undalis* was lower compared with other pests observed in this study. It was found that the population abundance of cabbage webworm larvae (*H. undalis*) differed significantly ($P \leq 0.05$) between the two wet seasons (2019 and 2020) in both experimental sites (Table 17). It was found that, in 2019 season, the population abundance of *H. undalis* was significantly ($P \leq 0.05$) higher (0.39 ± 0.06 and 0.35 ± 0.04) compared with 2020 wet season (0.24 ± 0.02 and 0.23 ± 0.02) at Tengeru and Boro experimental sites, respectively (Table 17).

After application of the treatments, the insecticidal actions of the aqueous extracts used and the pesticide differed significantly ($P \leq 0.05$) from one treated plot to another in the field (Table 17). It was revealed that the 5% concentration of aqueous extract from the mixed plants was significantly ($P \leq 0.05$) effective as synthetic pesticide in lowering population abundance of *H. undalis* in the field at both experimental sites and in both wet seasons (Table 17). In 5% concentration of aqueous extract from the mixed plant treated plots, the mean population of *H. undalis* was significantly ($P \leq 0.05$) as lower (0.06 ± 0.03 and 0.08 ± 0.02) as synthetic pesticide (0.03 ± 0.02 and 0.07 ± 0.02) in 2019 and 2020 wet season, respectively (Table 17). The effectiveness of the 10% concentrations from *C. dichogamus*, *T. vogelii*, and *S. aromaticum* followed the 5% concentration of aqueous extract from the mixed plants and synthetic pesticide (Table 17). The other concentrations (1% and 5%) from *C. dichogamus*, *T. vogelii*, and *S.*

aromaticum and 2.5% concentration of aqueous extract from the mixed plants significantly ($P \leq 0.05$) lowered *H. undalis* compared with negative controls (water and water plus soap) (Table 17).

Moreover, the weekly observations, indicated that, the population abundance of *H. undalis*, was significantly ($P \leq 0.05$) higher (0.24 ± 0.02, 0.30 ± 0.04, 0.33 ± 0.04, 0.38 ± 0.05, 0.41 ± 0.05, 0.43 ± 0.05) in 2019 wet season compared with 2020 wet season (0.19 ± 0.02, 0.24 ± 0.02, 0.24 ± 0.02, 0.24 ± 0.02, 0.25 ± 0.02, 0.27 ± 0.02) from week 1 before treatment applications to week 5 after applications of the treatments (Table 18). The population abundance of *H. undalis* varied significantly on week 1 before treatment, week 1 after treatment applications (Table 18) between the two experimental sites. On the other weeks (weeks 2, 3, 4 and 5 after treatment applications) the population abundance of *H. undalis* was significantly ($P \leq 0.05$) the same in both experimental sites (Table 18).

Also, the weekly observations revealed that, the insecticidal actions of the aqueous extracts from *C. dichogamus, T. vogelii,* and *S. aromaticum* differed significantly ($P \leq 0.05$) when compared with negative controls in both experimental sites (Table 18). Similarly, the 5% concentration of aqueous extract from the mixed plants was significantly ($P \leq 0.05$) as effective as synthetic pesticide because they hosted less (0.06 ± 0.03, 0.05 ± 0.02, 0.04 ± 0.02, 0.08 ± 0.02, 0.06 ± 0.03) population abundance of *H. undalis* compared with other concentrations reported in this study. Then, it was followed by the 10% concentrations of the aqueous extracts of *C. dichogamus, T. vogelii* and *S. aromaticum* (Table 18). But the other concentrations (1% and 5%) and 2.5% of the extracts from the mixed plants significantly lowered the population abundance of *H. undalis* compared with the negative controls (Table 18). Moreover, appendix 4 showed the weekly trends of population abundance of *H. undalis* in the two wet seasons separately in both experimental sites (Tengeru and Boro) in the entire period of the study.

Table 17: Mean population of *H. undalis* per cabbage crop in response to treatments application

Location and Treatments	Seasons	
	2019 season	**2020 season**
Location		
Tengeru	0.39 ± 0.06a	0.24 ± 0.02a
Boro	0.35 ± 0.04a	0.23 ± 0.02a
Treatments		
Water	1.15 ± 0.20a	0.55 ± 0.04a
water + soap	1.14 ± 0.13a	0.53 ± 0.04a
Synthetic pesticide	0.03 ± 0.02f	0.07 ± 0.02f
C. dichogamus (1%)	0.41 ± 0.07bc	0.28 ± 0.04bc
C. dichogamus (5%)	0.27 ± 0.03cd	0.22 ± 0.03cde
C. dichogamus (10%)	0.17 ± 0.01def	0.14± 0.03efg
S. aromaticum (1%)	0.36 ± 0.04bcd	0.34 ± 0.03b
S. aromaticum (5%)	0.25 ± 0.03cde	0.20 ± 0.01cde
S. aromaticum (10%)	0.16 ± 0.02def	0.16 ± 0.03def
T. vogelii (1%)	0.49 ± 0.04b	0.23 ± 0.02cd
T. vogelii (5%)	0.31 ± 0.07bcd	0.20 ± 0.03cde
T. vogelii (10%)	0.17 ± 0.02def	0.14 ± 0.03efg
Mixed plants (2.5%)	0.24 ± 0.05cde	0.18 ± 0.02de
Mixed plants (5%)	0.06 ± 0.03ef	0.08 ± 0.02f
2 - way ANOVA	**(F- Statistics)**	
Locations	0.69ns	0.80ns
Treatments	21.70***	24.03***
Location*treatments	0.84ns	0.73ns

Each value is a mean ± standard error of eight replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fisher's Least Significant Difference (LSD) test.

Table 18: Mean population of *H. undalis* larvae per cabbage crop in response to weekly treatments application in two seasons

Location and Treatments	Week 1 before Treatment	Weeks after treatments				
		1	2	3	4	5
Seasons						
Season 1 (2019)	0.24 ± 0.02a	0.30 ± 0.04a	0.33 ± 0.04a	0.38 ± 0.05a	0.41 ± 0.05a	0.43 ± 0.05a
Season 2 (2020)	0.19 ± 0.02b	0.24. ± 0.02a	0.24 ± 0.02b	0.24 ± 0.02b	0.25 ± 0.02b	0.27 ± 0.02b
Locations						
Tengeru	0.17 ± 0.02b	0.32 ± 0.04a	0.30 ± 0.04a	0.32 ± 0.05a	0.34 ± 0.04a	0.35 ± 0.03a
Boro	0.26 ± 0.03a	0.21 ± 0.02b	0.28 ± 0.02a	0.30 ± 0.03a	0.31 ± 0.03a	0.35 ± 0.04a
Treatments						
W	0.24± 0.05a	0.83 ± 0.19a	0.76 ± 0.12a	0.84 ± 0.18a	0.95 ± 0.18a	1.03 ± 0.20a
W + s	0.25 ± 0.05a	0.74 ± 0.10a	0.78 ± 0.14a	0.95 ± 0.23a	0.94 ± 0.12a	0.94 ± 0.10a
S. p	0.13 ± 0.04a	0.00 ± 0.00f	0.03 ± 0.02f	0.05 ± 0.03de	0.08 ± 0.03d	0.06 ± 0.02e
C. d (1%)	0.20 ± 0.06a	0.31 ± 0.07bc	0.36 ± 0.06b	0.36 ± 0.05bc	0.36 ± 0.07bc	0.38 ± 0.07bc
C. d (5%)	0.26 ± 0.04a	0.21 ± 0.05bcde	0.24 ± 0.04bcd	0.25 ± 0.06bcde	0.24 ± 0.06bcd	0.26 ± 0.04bcde
C. d (10%)	0.23 ± 0.04a	0.11 ± 0.03def	0.16 ± 0.04cdef	0.15 ± 0.04cde	0.16 ± 0.03cd	0.18 ± 0.04cde
S. a (1%)	0.24 ± 0.03a	0.33 ± 0.06b	0.35 ± 0.06b	0.30 ± 0.05bcd	0.36 ± 0.04bc	0.45 ± 0.05b
S. a (5%)	0.26 ± 0.05a	0.21 ± 0.04bcde	0.23 ± 0.04cde	0.19 ± 0.05bcde	0.21 ± 0.05cd	0.26 ± 0.05bcde
S. a (10%)	0.28 ± 0.05a	0.16 ± 0.05cdef	0.14 ± 0.03def	0.14 ± 0.04cde	0.14 ± 0.03cd	0.16 ± 0.02de
T. v (1%)	0.15 ± 0.04a	0.29 ± 0.07bcd	0.34 ± 0.08bc	0.41 ± 0.08b	0.43 ± 0.06b	0.41 ± 0.05b
T. v (5%)	0.20 ± 0.05a	0.18 ± 0.04cdef	0.24 ± 0.04bcd	0.34 ± 0.09bc	0.25 ± 0.05bcd	0.29 ± 0.04bcd
T. v (10%)	0.24 ± 0.05a	0.13 ± 0.03def	0.14 ± 0.05def	0.11 ± 0.04cde	0.19 ± 0.04cd	0.15 ± 0.03de
M. p. (2.5%)	0.18 ± 0.04a	0.20 ± 0.04bcde	0.21 ± 0.05cde	0.21 ± 0.03bcde	0.20 ± 0.03cd	0.25 ± 0.05bcde
M. p. (5%)	0.23 ± 0.05a	0.06± 0.03ef	0.05 ± 0.02ef	0.04 ± 0.02e	0.08 ± 0.02d	0.06 ± 0.03e
3 - way ANOVA	**(F- Statistics)**					
Season (S)	4.34*	2.75ns	7.22**	8.07**	19.10***	18.83***
Location (L)	14.45***	7.64**	0.43ns	0.16ns	0.73ns	0.01ns
Treatments (T)	0.82ns	10.61***	11.97***	9.06***	18.10***	18.69***
S * L	0.62ns	0.00ns	2.80ns	1.14ns	0.57ns	7.62**
S * T	0.47ns	0.69ns	1.89ns	1.52ns	3.01***	3.32***
L * T	0.45ns	1.11ns	0.53ns	0.56ns	0.68ns	0.18ns
S * L * T	0.31ns	0.19ns	0.26ns	0.56ns	0.31ns	0.35ns

Each value is a mean ± standard error of sixteen replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fisher's Least Significant Difference (LSD) test.

Note: W - water, w + s - Water plus soap, S. p - Synthetic pesticide, *C. d – Croton dichogamus*, *S. a – Syzygium aromaticum*, *T. v – Tephrosia vogelii*, M. p – Mixed plants

The interactions of experimental sites' weather conditions, treatments and seasons significantly ($P \leq 0.05$) affected the population abundance of *H. undalis* compared with the negative controls (water and water plus soap) in the plots (Table 18; Fig. 24 and 25). The interaction between seasons and treatments was significant ($P \leq 0.05$) on week 5 and 6 of the whole period of the study in two wet seasons (Table 18; Fig. 25). But the other weeks of the study period were insignificant ($P > 0.05$) in both wet seasons 2019 and 2020 (Table 18). It was observed that the population abundance of *H. undalis* in 2019 season significantly decreased in the treated plots relative to the negative controls (water and water plus soap).

The interactive efficacies of the treatments and the seasons was observed on week 5 and 6 from the 1st before treatment to 5th week after treatment applications (Fig. 25). The combinations of the treatments and the seasons significantly ($P \leq 0.05$) enhanced the reduction of the population of *H. undalis* compared with the negative controls (water and water plus soap) (Table 18; Fig. 25). Seasonal variations facilitated by weather condition variations mighty have affected the population abundance of *H. undalis* at the study sites. Higher temperatures and lower rainfall precipitations in 2019 wet season could be the reason of the higher population of *H. undalis* compared with 2020 season in which the temperatures and rainfall precipitation were relatively high.

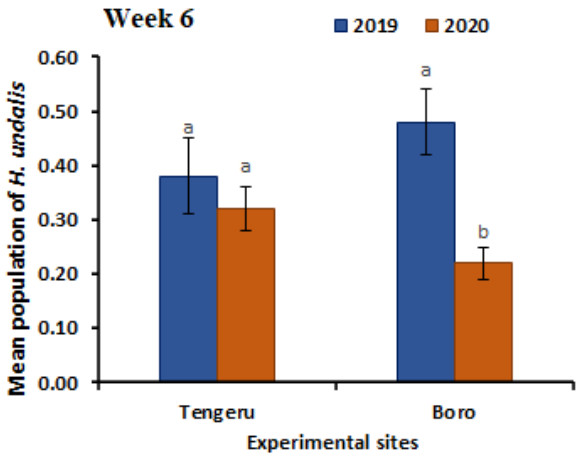
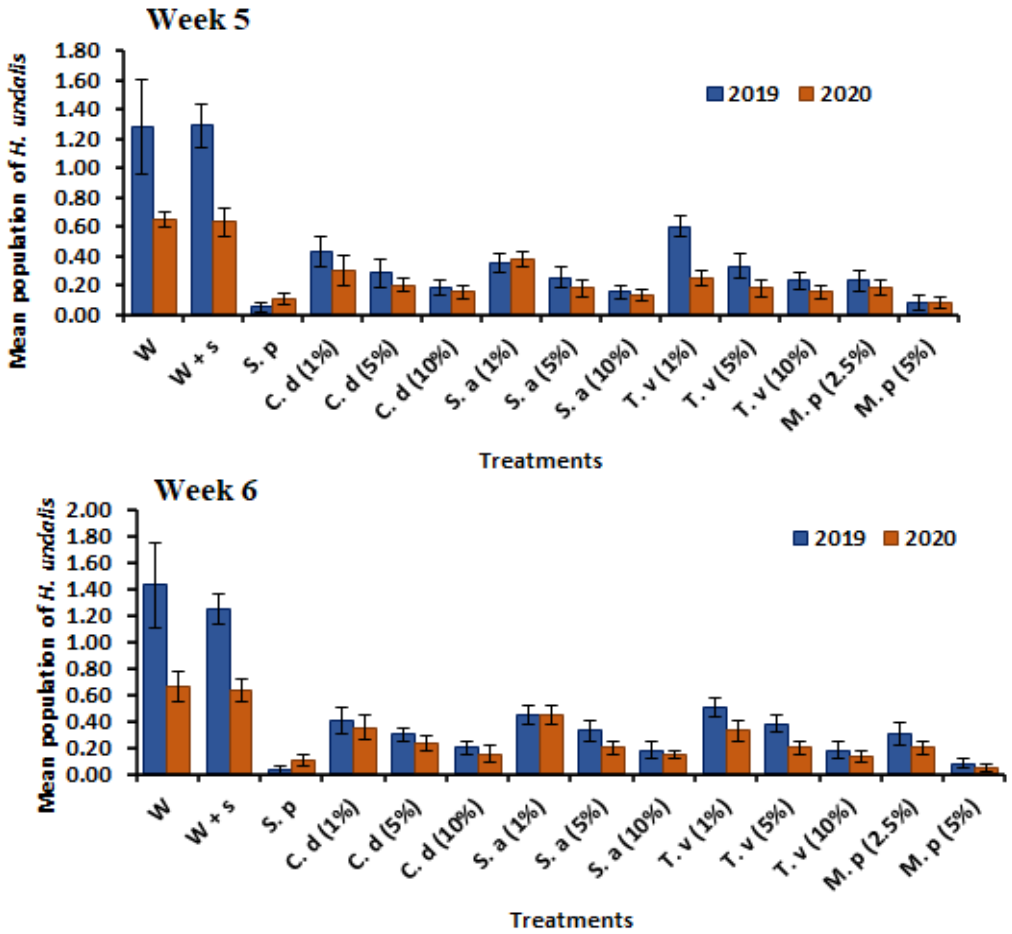


Figure 24: Interaction of the experimental sites and wet seasons on population lowering of *H. undalis* (Week 6)



W - water, w + s - Water plus soap, S. p - Synthetic pesticide, *C. d – Croton dichogamus, S. a – Syzygium aromaticum, T. v – Tephrosia vogelii*

Figure 25: Interaction of treatments and wet seasons on lowering the population of *H. undalis* (Week 5 and 6)

Population dynamics of T. ni in response to the treatments

Generally, the mean abundance of *T. ni*, for 2019 and 2020 wet seasons at the two experimental sites is presented in Table 19. It was observed that the population abundance of *T. ni* between the two experimental sites in 2019 wet season differed significantly ($P \leq 0.05$). It was found that, in 2019 season, the population abundance of *T. ni* was lower (0.09 ± 0.02) at Boro experimental site compared with Tengeru experimental site (0.18 ± 0.04) (Table 19). But in 2020 wet season, the population abundance of *T. ni* was significantly the same between the two experimental sites (Table 19).

Apart from differences in population abundance between the two experimental sites, the insecticidal actions of the treatments differed significantly ($P \leq 0.05$) from one treatment plot to another in the field. It was found that, the aqueous extracts of *C. dichogamus*, *T. vogelii*, and

S. aromaticum were significantly ($P \leq 0.05$) effective as synthetic pesticides in the 2019 wet season (Table 19). But in 2020 season, the 5% concentration of aqueous extract from the mixed plants was significantly ($P \leq 0.05$) effective as synthetic pesticide in lowering population abundance of *T. ni* in the field (Table 19). The mean population abundance of *T. ni* in 5% concentration of aqueous extract of the mixed plants had significantly ($P \leq 0.05$) lower (0.05 ± 0.01) as synthetic pesticide (0.04 ± 0.01) in 2020 wet season. Then, it was followed by the 10% concentrations of the aqueous extracts of *C. dichogamus, T. vogelii* and *S. aromaticum.* But, the other concentrations (1% and 5%) of the aqueous extracts of *C. dichogamus, T. vogelii* and *S. aromaticum* and 2.5% of the aqueous extracts from the mixed plants significantly lowered the population abundance of *T. ni* compared with negative controls in 2020 wet season (Table 19).

Moreover, the weekly observations revealed that mean population abundance of *T. ni* was significantly ($P \leq 0.05$) lower (0.13 ± 0.01, 0.11 ± 0.02, 0.13 ± 0.03, 0.14 ± 0.03, 0.14 ± 0.03, 0.15 ± 0.03) in 2019 wet season compared with 2020 wet season (0.18 ± 0.01, 0.20 ± 0.02, 0.21 ± 0.02, 0.25 ± 0.02, 0.23 ± 0.02, 0.24 ± 0.02) from week 1 before application of the treatments, week 1 to week 5 after application of the treatments, respectively (Table 20). Similarly, the mean population abundance of *T. ni* was significantly ($P \leq 0.05$) lower (0.14 ± 0.01, 0.11 ± 0.02, 0.13 ± 0.03, 0.14 ± 0.03, 0.14 ± 0.03) at Boro experimental site compared with Tengeru experimental site (0.17 ± 0.01, 0.18 ± 0.03, 0.19 ± 0.03, 0.22 ± 0.03, 0.23 ± 0.03) in week 1 before treatment applications, weeks 1, 2, 3 and 5 after treatments applications except week 4 which the mean population abundance of *T. ni* was significantly the same between the two experimental sites (Table 20).

Also, the insecticidal actions of the aqueous extracts from *C. dichogamus, T. vogelii* and *S. aromaticum* differed significantly ($P \leq 0.05$) from one plot to another when compared with the negative control plots (water and water plus soap) (Table 20). It was revealed that, 5% concentration of aqueous extracts of the mixed plants possessed significantly ($p \leq 0.05$ lower mean number of *T. ni* (0.03 ± 0.02, 0.05 ± 0.02, 0.00 ± 0.00, 0.03 ± 0.02, 0.01 ± 0.01) as in chlorpyrifos (0.04 ± 0.03, 0.01 ± 0.01, 0.03 ± 0.02, 0.01 ± 0.01, 0.04 ± 0.02) on weeks 1, 2, 3, 4 and 5 after treatments, respectively (Table 20). Therefore, 5% concentration of aqueous extract from the mixed plants was significantly ($P \leq 0.05$) as effective as synthetic pesticide. Then, it was followed by the 10% concentrations of the aqueous extracts of *C. dichogamus, T.*

vogelii and *S. aromaticum* (Table 20). The other concentrations were more effective compared with the negative controls (water and water plus soap) (Table 20).

Table 19: Mean number of *T. ni* per cabbage crop in response to the treatments for two wet seasons

Location and Treatments	Mean ± SE	
	2019 season	2020 season
Location		
Tengeru	0.18 ± 0.04a	0.28 ± 0.04a
Boro	0.09 ± 0.02b	0.19 ± 0.02a
Treatments		
Water	0.60 ± 0.14a	0.49 ± 0.03a
water + soap	0.58 ± 0.17a	0.48 ± 0.04a
Synthetic pesticide	0.03 ± 0.01b	0.04 ± 0.01f
C. dichogamus (1%)	0.11 ± 0.04b	0.23 ± 0.03bcd
C. dichogamus (5%)	0.09 ± 0.02b	0.20 ± 0.02cde
C. dichogamus (10%)	0.06 ± 0.02b	0.14± 0.02e
S. aromaticum (1%)	0.09 ± 0.03b	0.27 ± 0.04bc
S. aromaticum (5%)	0.04 ± 0.01b	0.20 ± 0.04cde
S. aromaticum (10%)	0.03 ± 0.02b	0.15 ± 0.01de
T. vogelii (1%)	0.13 ± 0.02b	0.28 ± 0.02b
T. vogelii (5%)	0.07 ± 0.02b	0.20 ± 0.04cde
T. vogelii (10%)	0.04 ± 0.03b	0.13 ± 0.03e
Mixed plants (2.5%)	0.02 ± 0.01b	0.18 ± 0.02de
Mixed plants (5%)	0.00 ± 0.00b	0.05 ± 0.01f
2 - way ANOVA	**(F- Statistics)**	
Locations	7.89**	0.39ns
Treatments	11.46***	21.92***
Location*treatments	1.46ns	0.61ns

Each value is a mean ± standard error of eight replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fisher's Least Significant Difference (LSD) test.

Table 20: Mean number of *T. ni* per cabbage crop in response to weekly application of treatments

Location and Treatments	Week 1 before Treatment	Weeks after treatments				
		1	2	3	4	5
Seasons						
Wet season 1 (2019)	0.13 ± 0.01b	0.11 ± 0.02b	0.13 ± 0.03b	0.14 ± 0.03b	0.14 ± 0.03b	0.15 ± 0.03b
Wet season 2 (2020)	0.18 ± 0.01a	0.20. ± 0.02a	0.21 ± 0.02a	0.25 ± 0.02a	0.23 ± 0.02a	0.24 ± 0.02a
Locations						
Tengeru	0.17 ± 0.01a	0.18 ± 0.03a	0.19 ± 0.03a	0.22 ± 0.03a	0.20 ± 0.03a	0.23 ± 0.03a
Boro	0.14 ± 0.01b	0.13 ± 0.02b	0.14 ± 0.02b	0.17 ± 0.02b	0.17 ± 0.02a	0.16 ± 0.02b
Treatments						
W	0.21± 0.06a	0.46 ± 0.07a	0.58 ± 0.14a	0.63 ± 0.12a	0.51 ± 0.08a	0.66 ± 0.09a
W + s	0.16 ± 0.04a	0.35 ± 0.05ab	0.45 ± 0.10a	0.65 ± 0.13a	0.61 ± 0.10a	0.71 ± 0.11a
S. p	0.13 ± 0.03a	0.04 ± 0.03fg	0.01 ± 0.01c	0.03 ± 0.02de	0.01 ± 0.01e	0.04 ± 0.02cd
C. d (1%)	0.15 ± 0.03a	0.15 ± 0.03cde	0.13 ± 0.04bc	0.21 ± 0.04bc	0.24 ± 0.06b	0.15 ± 0.04bcd
C. d (5%)	0.11 ± 0.04a	0.14 ± 0.04cde	0.15 ± 0.03bc	016 ± 0.04bcd	0.15 ± 0.03bcd	0.16 ± 0.04bc
C. d (10%)	0.18 ± 0.03a	0.09 ± 0.03efg	0.10 ± 0.03bc	0.10 ± 0.03cde	0.10 ± 0.03de	0.09 ± 0.03bcd
S. a (1%)	0.11 ± 0.02a	0.20 ± 0.04bc	0.20 ± 0.05b	0.19 ± 0.05bc	0.19 ± 0.06bc	0.19 ± 0.05b
S. a (5%)	0.18 ± 0.03a	0.13 ± 0.05cdef	0.15 ± 0.04bc	0.10 ± 0.04cde	0.11 ± 0.03cde	0.13 ± 0.04bcd
S. a (10%)	0.16 ± 0.03a	0.10 ± 0.03defg	0.09 ± 0.03bc	0.05 ± 0.02cde	0.13 ± 0.04cde	0.06 ± 0.02bcd
T. v (1%)	0.18 ± 0.04a	0.19 ± 0.04cd	0.20 ± 0.04b	0.24 ± 0.04bcd	0.24 ± 0.04b	0.19 ± 0.04b
T. v (5%)	0.18 ± 0.05a	0.10 ± 0.02defg	0.11 ± 0.03bc	0.16 ± 0.05bcd	0.10 ± 0.04de	0.14 ± 0.04bcd
T. v (10%)	0.15 ± 0.02a	0.06 ± 0.03efg	0.06 ± 0.02bc	0.09 ± 0.03cde	0.08 ± 0.03de	0.06 ± 0.03bcd
M. p. (2.5%)	0.13 ± 0.03a	0.10 ± 0.04defg	0.09 ± 0.03bc	0.11 ± 0.04cde	0.10 ± 0.03de	0.11 ± 0.05bcd
M. p. (5%)	0.16 ± 0.04a	0.03 ± 0.02g	0.05 ± 0.02bc	0.00 ± 0.00e	0.03 ± 0.02e	0.01 ± 0.01d
3 - way ANOVA	**(F- Statistics)**					
Season (S)	7.20**	25.86***	8.33**	15.23***	12.79***	15.46***
Location (L)	5.03*	8.11**	3.46*	3.93*	0.77ns	9.35**
Treatments (T)	0.91ns	13.52***	9.07***	14.68***	12.39***	22.26***
S * L	11.25***	8.11**	14.33***	3.44ns	0.04ns	0.76ns
S * T	0.77ns	1.24ns	0.80ns	0.76ns	0.93ns	2.56**
L * T	1.31ns	0.51ns	1.67ns	1.74ns	0.39ns	1.45ns
S * L * T	1.42ns	0.82ns	1.72ns	2.20*	0.38ns	0.47ns

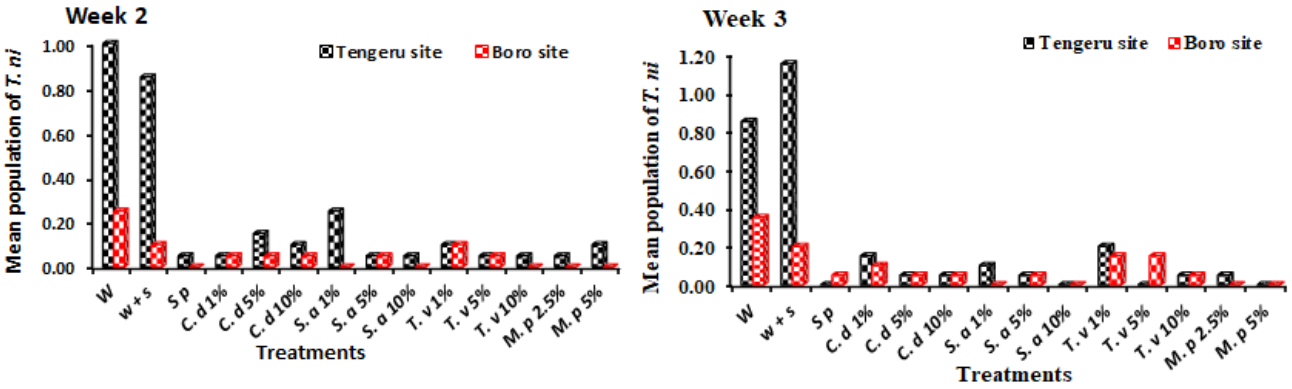
Each value is a mean ± standard error of sixteen replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fisher's Least Significant Difference (LSD) test.

W - water, w + s - Water plus soap, S. p - Synthetic pesticide, *C. d – Croton dichogamus*, *S. a – Syzygium aromaticum*, *T. v – Tephrosia vogelii*, M. p – Mixed plants.

The interactions among experimental sites' weather conditions, treatments and seasons significantly ($P \leq 0.05$) affected the population abundance of the *T. ni* compared with the negative controls (water and water plus soap) in the plots (Table 19 and 20; Appendix 5; Fig. 26, 27 and 28). The interaction between treatments and experimental sites was significant ($P \leq 0.05$) on weeks 2, 3 after treatments in 2019 wet season (Appendix 5; Fig. 26) while the other weeks of the interaction was insignificant ($P > 0.05$). Also, in 2020 wet season the interaction of treatments and experimental sites was insignificant (Appendix 5). It was observed that the population abundance of *T. ni* in 2019 wet season significantly decreased in the treated plots relative to the negative controls (water and water plus soap).

The interactions of experimental sites' weather conditions and seasons significantly ($P \leq 0.05$) affected the population abundance of the *T. ni* compared with the negative controls (water and water plus soap) in the plots (Table 20; Fig. 27) on weeks 1, 2, 3 during the application of the treatments. Moreover, it was revealed that, the interactions of treatments and seasons significantly ($P \leq 0.05$) affected the population abundance of the *T. ni* compared with the negative controls (water and water plus soap) in the plots (Table 20; Fig. 27) on weeks 1, 2 and 3 during the study period (Table 20; Fig. 28).

The interactions among the experimental sites' weather conditions, treatments and seasons significantly ($P \leq 0.05$) affected the population abundance of *T. ni* compared with the negative controls (water and water plus soap) in the plots in week 4 of the study period (Table 20; Fig. 28).



W - water, w + s - Water plus soap, S. p - Synthetic pesticide, *C. d – Croton dichogamus*, *S. a – Syzygium aromaticum*, *T. v – Tephrosia vogelii*, M. p – Mixed plants

Figure 26: Interaction of the study sites and treatments on population abundance of *T. ni* (Week 2 and 3)

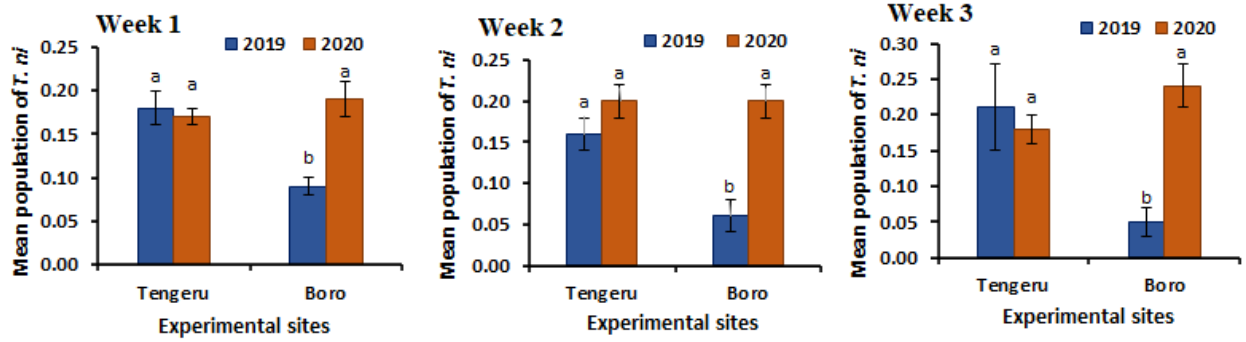
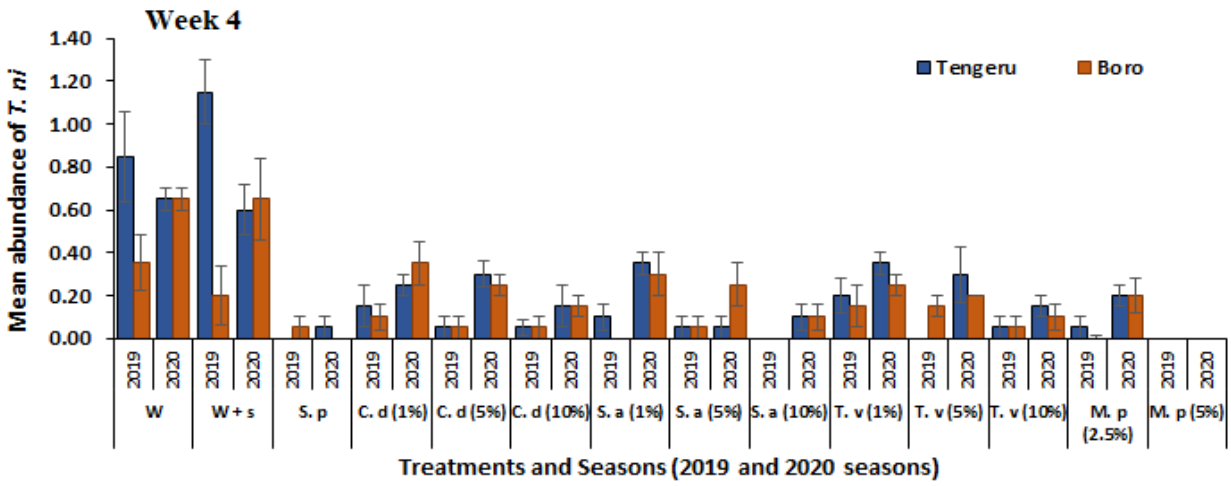


Figure 27: Interaction of the experimental sites and the seasons on reduction of population abundance of *T. ni* (Week 1, 2 and 3 during the study)



W - water, w + s - Water plus soap, S. p - Synthetic pesticide, *C. d – Croton dichogamus*, *S. a – Syzygium aromaticum*, *T. v – Tephrosia vogelii*, M. p – Mixed plants.

Figure 28: Interaction of the study sites, seasons and treatments on reduction of population abundance of *T. ni* (Week 4 during the experimental study period)

Population dynamics of C. binotalis in response to the treatments

This pest was identified on the plants four weeks after transplanting of the seedlings on the plots and the mean population increased rapidly during the head formation and was found hidden within the wrapper leaves, completely shielded from the sun. In 2019 wet season, it was revealed that, the population abundance of *C. binotalis* was significantly ($P \leq 0.05$) lower (0.24 ± 0.05) at Boro experimental site compared with Tengeru experimental site (0.40 ± 0.09) (Table 21) while in 2020 wet season the population abundance of *C. binotalis* was significantly lower (0.31 ± 0.03) at Tengeru experimental site compared with Boro experimental site (0.52 ± 0.09) (Table 21).

The insecticidal action of aqueous extracts used to control the *C. binotalis* in the plots differed significantly ($P \leq 0.05$) compared with negative controls (water and water plus soap) in the field. The results showed that there was significant different ($P \leq 0.05$) among the treatments in the population abundance of *C. binotalis* (Table 21) in both seasons, 2019 wet season and 2020 wet season. It was found that the mean population of *C. binotalis* was significantly ($P \leq 0.05$) lower in the aqueous extract treated plots as in synthetic pesticide (Table 21) in 2019 wet season. But in 2020 wet season, the 5% concentration of aqueous extract from the mixed plants hosted significantly ($P \leq 0.05$) lower population abundance (0.13 ± 0.02) of *C. binotalis* compared with other concentrations of the aqueous extracts (Table 21). The 5% concentration of aqueous extract from the mixed plants was as effective as synthetic pesticide (Table 21).

The other concentrations (1%, 5% and 10%) from *C. dichogamus, T. vogelii,* and *S. aromaticum* and 2.5% concentration of aqueous extract from the mixed plants significantly ($P \leq 0.05$) controlled population abundance of *C. binotalis* compared with negative controls (Water and water plus soap) (Table 21).

Moreover, Table 22 indicates the weekly observations of population abundance of *C. binotalis* between the two wet seasons and at the two experimental sites. It was found that, the mean population of *C. binotalis* was significantly ($P \leq 0.05$) lower (0.37 ± 0.07, 0.34 ± 0.06, 0.36 ± 0.07) in 2019 wet season compared with 2020 wet season (0.48 ± 0.08, 0.52 ± 0.08, 0.59 ± 0.09) on weeks 3, 4 and 5 after treatment applications, respectively (Table 22). But on weeks 1 before treatment applications and weeks 1 and 2 after treatment applications, the population abundance of *C. binotalis* was significantly the same (Table 22). In addition, the mean population abundance of *C. binotalis* varied significantly ($P \leq 0.05$) between Tengeru experimental site and Boro experimental site from week 1 to week 6 during the study period (Table 22).

It was found that, the insecticidal action of the aqueous extracts from *C. dichogamus, T. vogelii* and *S. aromaticum* differed significantly ($P \leq 0.05$) among the plots in the field compared with negative controls in both experimental sites in the two seasons (Table 22). It was found that the aqueous extracts from *C. dichogamus, T. vogelii* and *S. aromaticum* were significantly ($P \leq 0.05$) as effective as synthetic pesticide on weeks 1 and 2 after treatment applications (Table 22). But the 5% concentration of the extract from the mixed plants, possessed significantly ($P \leq 0.05$) lower (0.03 ± 0.02, 0.04 ± 0.02, 0.05 ± 0.03) mean number of *C. binotalis* compared with other concentrations of aqueous extracts on weeks 3, 4 and 5, respectively (Table 22). Similarly, 1%, 5% and 10% of the aqueous extracts from *C. dichogamus, T. vogelii,* and *S. aromaticum* and 2.5% of the aqueous extracts from the mixed plants, significantly lowered the population abundance of *C. binotalis* compared with the negative controls (Table 22). Appendix 6 indicates the trends of the mean population of *C. binotalis* in response to treatments in 2019 and 2020 wet seasons separately.

Table 21: Mean number of *C. binotalis* per crop in response to application of treatments in the two seasons

Location and Treatments	Seasons	
	2019 season	**2020 season**
Location		
Tengeru	0.40 ± 0.09a	0.31 ± 0.03b
Boro	0.24 ± 0.05b	0.52 ± 0.09a
Treatments		
Water	1.51 ± 0.22a	1.48 ± 0.27a
water + soap	1.50 ± 0.18a	1.39 ± 0.25a
Synthetic pesticide	0.08 ± 0.03b	0.09 ± 0.02c
C. dichogamus (1%)	0.23 ± 0.04b	0.32 ± 0.02bc
C. dichogamus (5%)	0.13 ± 0.02b	0.30 ± 0.04bc
C. dichogamus (10%)	0.13 ± 0.03b	0.20± 0.02bc
S. aromaticum (1%)	0.21 ± 0.04b	0.40 ± 0.04b
S. aromaticum (5%)	0.12 ± 0.02b	0.28 ± 0.05bc
S. aromaticum (10%)	0.11 ± 0.02b	0.21 ± 0.02bc
T. vogelii (1%)	0.15± 0.03b	0.36 ± 0.05bc
T. vogelii (5%)	0.09 ± 0.03b	0.26 ± 0.03bc
T. vogelii (10%)	0.08 ± 0.02b	0.17 ± 0.03bc
Mixed plants (2.5%)	0.10 ± 0.02b	0.21 ± 0.03bc
Mixed plants (5%)	0.06 ± 0.01b	0.13 ± 0.02c
2 - way ANOVA	**(F- Statistics)**	
Locations	25.73***	50.75***
Treatments	74.03***	60.51***
Location*treatments	5.57***	13.73***

Each value is a mean ± standard error of eight replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fisher's Least Significant Difference (LSD) test.

Table 22: Mean population of *C. binotalis* per cabbage crop in response to weekly application of treatments

Location and Treatments	Week 1 before Treatment	Weeks after treatments (Mean ± SE)				
		1	2	3	4	5
Seasons						
Season 1 (2019)	0.28 ± 0.02a	0.26 ± 0.05a	0.32 ± 0.06a	0.37 ± 0.07b	0.34 ± 0.06b	0.36 ± 0.07b
Season 2 (2020)	0.31 ± 0.02a	0.25 ± 0.03a	0.34 ± 0.04a	0.48 ± 0.08a	0.52 ± 0.08a	0.59 ± 0.09a
Locations						
Tengeru	0.26 ± 0.02b	0.29 ± 0.05a	0.33 ± 0.06a	0.32 ± 0.06b	0.39 ± 0.06a	0.54 ± 0.03a
Boro	0.33 ± 0.03a	0.22 ± 0.03b	0.33 ± 0.05a	0.53 ± 0.09a	0.47 ± 0.08a	0.41 ± 0.07b
Treatments						
W	0.45± 0.07a	1.01 ± 0.17a	1.44 ± 0.26a	1.91 ± 0.33a	1.86 ± 0.33a	2.29 ± 0.16a
W + s	0.33 ± 0.05a	0.99 ± 0.21a	1.29 ± 0.18a	2.03 ± 0.29a	1.73 ± 0.12a	2.34 ± 0.24a
S. p	0.19 ± 0.06a	0.05 ± 0.02b	0.06 ± 0.02b	0.05 ± 0.02c	0.08 ± 0.03cd	0.06 ± 0.03d
C. d (1%)	0.25 ± 0.05a	0.21 ± 0.05b	0.23 ± 0.04b	0.33 ± 0.05b	0.31 ± 0.05b	0.30 ± 0.05bc
C. d (5%)	0.31 ± 0.06a	0.11 ± 0.03b	0.20 ± 0.05b	0.20 ± 0.06bc	0.28 ± 0.05bc	0.20 ± 0.04bcd
C. d (10%)	0.30 ± 0.06a	0.09 ± 0.03b	0.11 ± 0.03b	0.11 ± 0.04bc	0.20 ± 0.04bcd	0.15 ± 0.05bcd
S. a (1%)	0.33 ± 0.05a	0.28 ± 0.06b	0.24 ± 0.05b	0.34 ± 0.07b	0.35 ± 0.07b	0.31 ± 0.08b
S. a (5%)	0.28 ± 0.05a	0.18 ± 0.06b	0.19 ± 0.05b	0.25 ± 0.07bc	0.18 ± 0.05bcd	0.16 ± 0.04bcd
S. a (10%)	0.33 ± 0.04a	0.13 ± 0.04b	0.14 ± 0.04b	0.13 ± 0.03bc	0.13 ± 0.03bcd	0.13 ± 0.04bcd
T. v (1%)	0.25 ± 0.06a	0.16 ± 0.05b	0.23 ± 0.07b	0.24 ± 0.08bc	0.34 ± 0.07b	0.33 ± 0.04b
T. v (5%)	0.26 ± 0.06a	0.11 ± 0.05b	0.18 ± 0.04b	0.15 ± 0.06bc	0.21 ± 0.05bcd	0.13 ± 0.04bcd
T. v (10%)	0.30 ± 0.06a	0.08 ± 0.03b	0.11 ± 0.03b	0.08 ± 0.03bc	0.11 ± 0.04cd	0.09 ± 0.03cd
M. p. (2.5%)	0.25 ± 0.03a	0.14 ± 0.04b	0.11 ± 0.04b	0.11 ± 0.04bc	0.19 ± 0.04bcd	0.13 ± 0.04bcde
M. p. (5%)	0.34 ± 0.05a	0.05± 0.03b	0.08 ± 0.03b	0.03 ± 0.02c	0.04 ± 0.02d	0.05 ± 0.03d
3 - way ANOVA	**(F- Statistics)**					
Season (S)	0.80ns	0.16ns	0.44ns	6.93**	16.89***	30.21***
Location (L)	7.21**	2.68*	0.02ns	21.69***	3.49ns	9.27***
Treatments (T)	1.35ns	18.12***	45.35***	64.33***	53.20***	98.34***
S * L	31.65***	15.48***	70.23***	46.85***	23.59***	0.01ns
S * T	0.75ns	2.63**	4.54***	1.44ns	0.56ns	1.31ns
L * T	0.98ns	0.44ns	0.25ns	4.22**	3.47***	1.06ns
S * L * T	0.80ns	1.80ns	9.28***	13.07***	17.35***	1.94ns

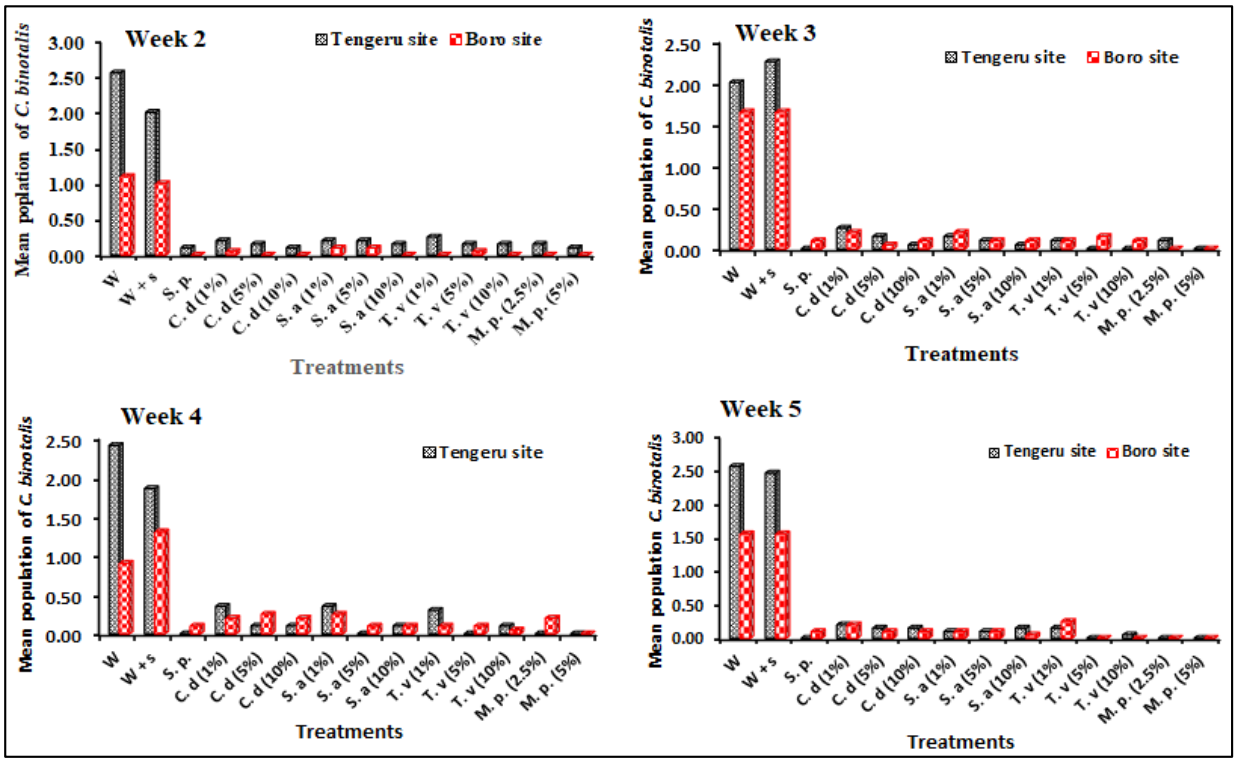
Each value is a mean ± standard error of sixteen replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fisher's Least Significant Difference (LSD) test.

W - water, w + s - Water plus soap, S. p - Synthetic pesticide, *C. d – Croton dichogamus*, *S. a – Syzygium aromaticum*, *T. v – Tephrosia vogelii*, M. p – Mixed plants.

The interactive effects among experimental sites' weather conditions, treatments and seasons significantly ($P \leq 0.05$) affected the population abundance of *C. binotalis* compared with the negative controls (water and water plus soap) in the plots (Table 21 and 22; Appendix 6). The interaction between treatments and experimental sites was significant ($P \leq 0.05$) in weeks 2, 3, 4 and 5 after treatments application in 2019 wet season (Table 22; Appendix 6; Fig. 29) while in 2020 wet season the interactive effects was observed on weeks 2, 3 and 4 (Fig. 30).

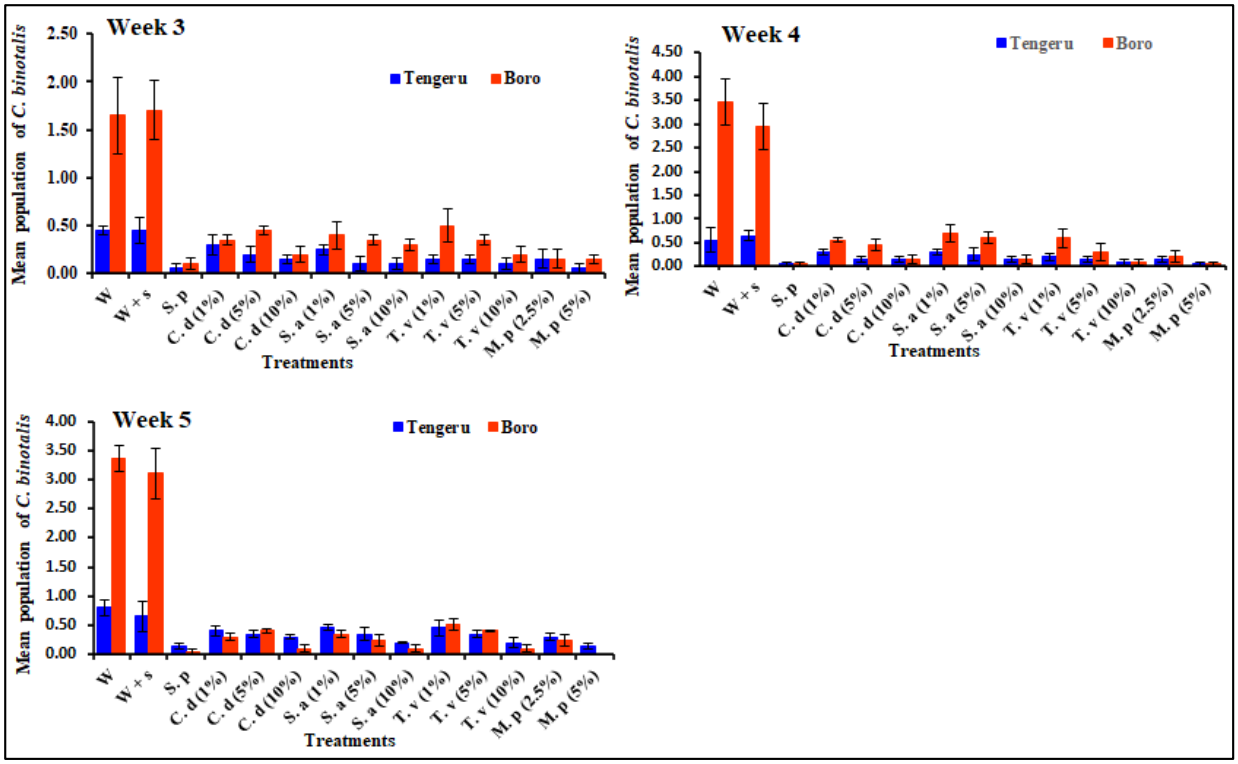
There was interactions of experimental sites' weather conditions and seasons which significantly ($P \leq 0.05$) affected the population abundance of *C. binotalis* compared with the

negative controls (water and water plus soap) in the plots (Table 22; Fig. 31) on weeks 1, 2, 3, 4 and 5 in the treated experimental plots). Similarly, there was interaction of treatments and seasons which significantly ($P \leq 0.05$) reduced the population abundance of *C. binotalis* compared with the negative controls (water and water plus soap) in the plots (Appendix 6; Fig. 29). The interactions among the experimental sites' weather conditions, treatments and seasons significantly ($P \leq 0.05$) affected the population abundance of *C. binotalis* compared with the negative controls (water and water plus soap) in the plots on weeks 3 and 4 of the experimental treatments (Table 22; Fig. 32).



W - water, w + s - Water plus soap, S. p - Synthetic pesticide, *C. d – Croton dichogamus*, *S. a – Syzygium aromaticum*, *T. v – Tephrosia vogelii*, M. p – Mixed plants.

Figure 29: Interactions of experimental sites with the treatments for reduction of the *C. binotalis* (Week 2,3,4 and 5) in 2019 wet season



W - water, w + s - Water plus soap, S. p - Synthetic pesticide, *C. d – Croton dichogamus, S. a – Syzygium aromaticum, T. v – Tephrosia vogelii*, M. p – Mixed plants.

Figure 30: Interaction of experimental sites with the treatments for reduction of the *C. binotalis* (Week 3,4 and 5) in 2020 wet season

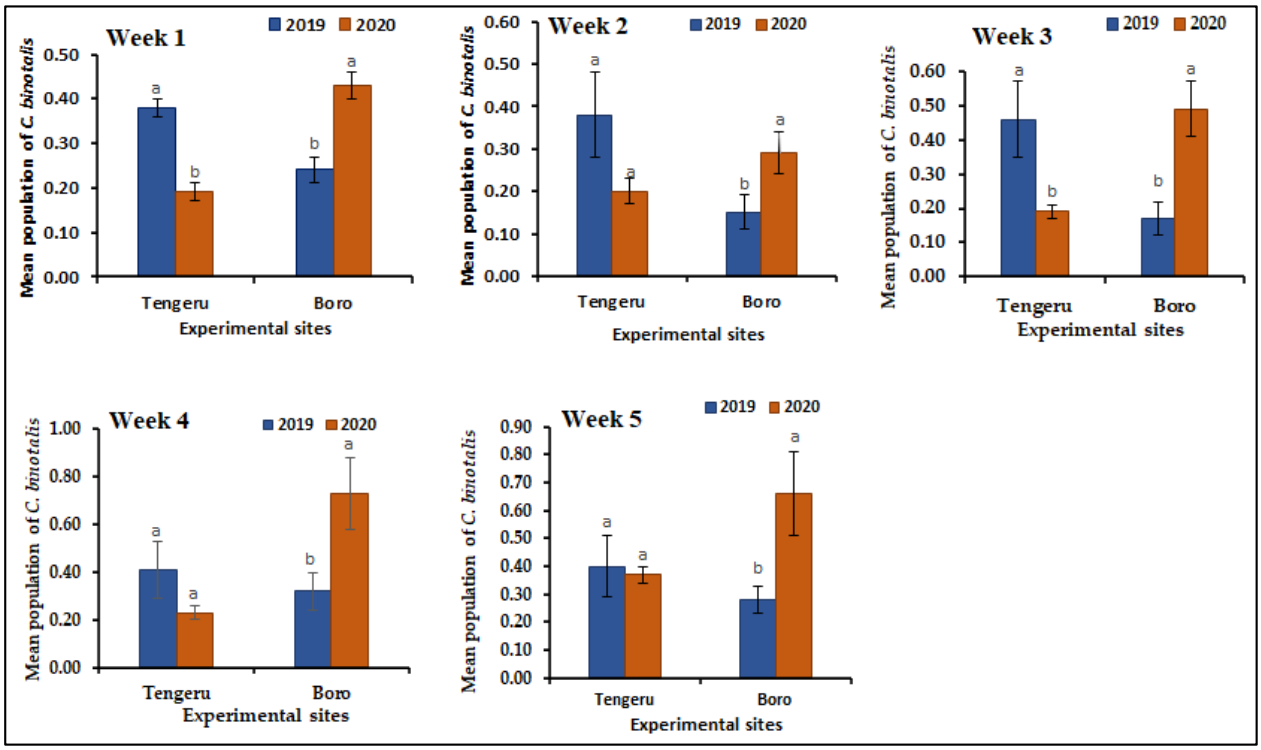
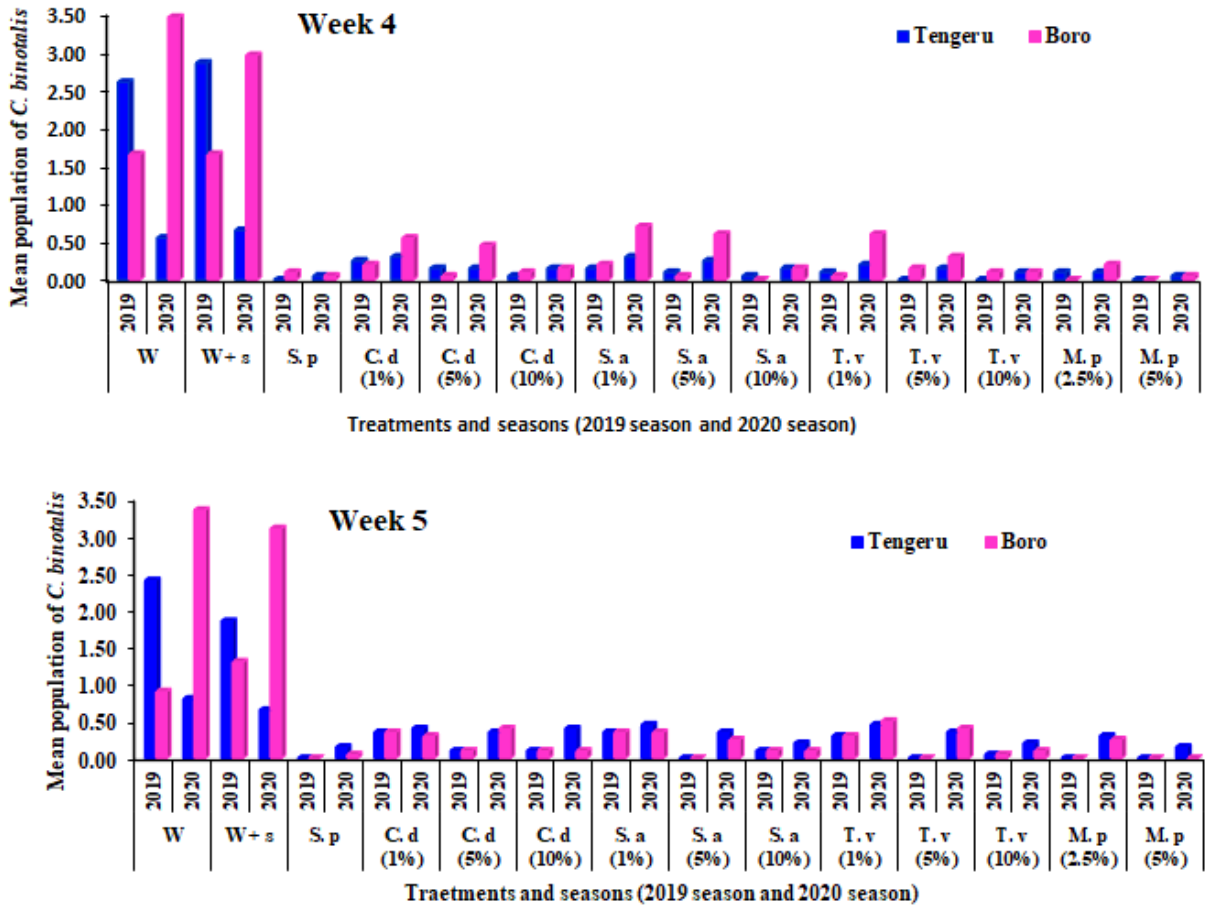


Figure 31: Interactions of experimental sites with the seasons for reduction of the *C. binotalis* (Week 1, 2, 3, 4 and 5) in 2019 and 2020 wet seasons



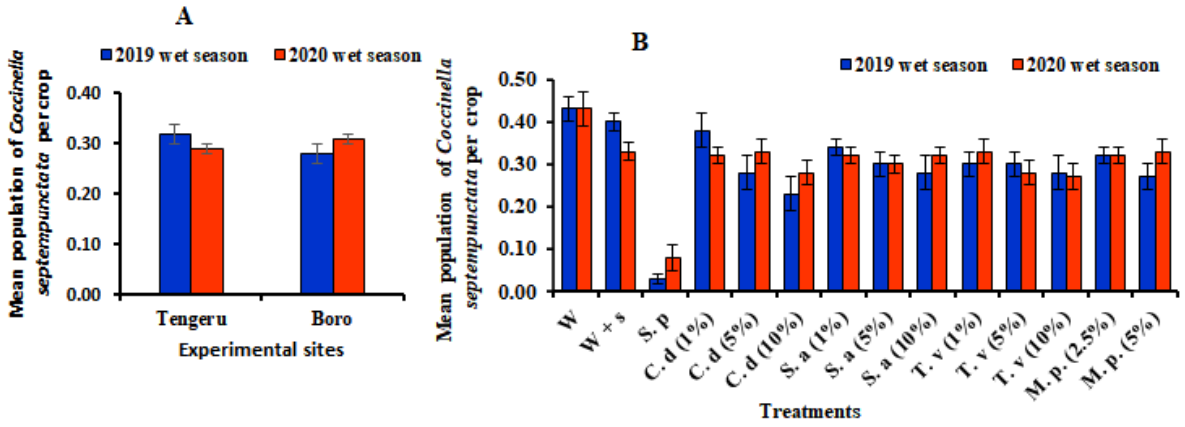
W - water, w + s - Water plus soap, S. p - Synthetic pesticide, *C. d – Croton dichogamus*, *S. a – Syzygium aromaticum*, *T. v – Tephrosia vogelii*, M. p – Mixed plants.

Figure 32: Interactions of treatments, sites and seasons for reduction of *C. binotalis* (Week 4, 5)

Population and abundance and response of Cocinella septempunctata to the treatments

Among the natural enemies of *M. persicae* observed during the study period from March, 2019 and 2020 wet season to August, 2019 and 2020 wet seasons *Cocinella septempunctata* was present in higher population relative to others. Others such as *Cotesia plutellae* were present in very low population. Figure 33 indicates the population of *C. septempunctata* in response to the treatments used in the two experimental sites in two wet seasons, 2019 and 2020. It was observed that the aqueous plant extracts possessed higher population abundance of *C. septempunctata* compared with synthetic pesticide at the two experimental sites in both wet seasons of 2019 and 2020 (Fig. 33). The population of *C. septempunctata* was higher at Tengeru experimental site compared with Boro experimental site in 2019 wet season (Fig. 33A) while in 2020 wet season the population of *C. septempunctata* was slightly higher at Boro experimental site compared with Tengeru experimental site (Fig. 33A). In both 2019 and 2020

wet seasons, the aqueous plant extracts possessed higher *C. septempunctata* compared with synthetic pesticide which possessed very low *C. septempunctata* (Fig. 33B).



water, w + s - Water plus soap, S. p - Synthetic pesticide, *C. d – Croton dichogamus*, *S. a – Syzygium aromaticum*, *T. v – Tephrosia vogelii*, M. p – Mixed plants.

Figure 33: A-Population of *C. septempunctata* per crop in the two wet seasons. B-Population of *C. septempunctata* in response to treatments

(iii) Percentage damage of cabbage (*B. oleracea*) crop caused by studied insect pests

Before and at the beginning of the applications of the treatments the damage of the crops was generally, low. Therefore, percent damages of cabbage (*B. oleracea*) were insignificant ($p > 0.05$) in the plots between the two experimental sites because the *B. oleracea* crops were still young plants (Table 24). However, percent damage increased progressively, as the time went on particularly in the plots which were treated with negative controls reaching 65 ± 3.2% and 61.6 ± 2.7% in water and water plus soap treated plots, respectively (Table 24) at week 5 after applications of the treatments. It was found that in 2019 wet season, the percentage damage of *B. oleracea* crop caused by the insect pests observed in this study, was significantly ($P \leq 0.05$) higher (16.2 ± 1.8%) at Tengeru experimental site compared with Boro experimental site (14.1 ± 1.5%) (Table 23). But in 2020 wet season, the percentage damage of cabbage crops (*B. oleracea*) was significantly the same in the two experimental sites (Table 23).

In both seasons, 2019 wet season and 2020 wet season the aqueous extracts and the chlorpyrifos used in this experiment significantly ($P \leq 0.05$) reduced the percentage damage of *B. oleracea* compared with the negative controls (water and water plus soap) at both experimental sites (Tengeru and Boro) (Table 23). In both wet seasons, the 5% concentration of aqueous extract from the mixed plants significantly ($P \leq 0.05$) reduced the percentage damage of *B. oleracea* compared with the other concentrations (1%, 5% and 10%) of the individual plants and the

2.5% concentration of aqueous extract from the mixed plants (Table 23) at both experimental sites. It was found that, percentage damage of *B. oleracea* in 5% concentration of aqueous extract from the mixed plants' treated plots was significantly ($P \leq 0.05$) lower (5.9 ± 1.1% and 5.0 ± 1.0%) as in synthetic pesticide (6.8 ± 0.9% and 4.5 ± 1.1%) in 2019 wet season and 2020 wet season, respectively (Table 23). It was followed by 2.5% concentration of aqueous extract from the mixed plants and the 10% concentrations of *C. dichogamus, T. vogelii,* and *S. aromaticum* of aqueous extract (Table 23). However, 1% and 5% concentrations of *C. dichogamus, T. vogelii* and *S. aromaticum* significantly performed better in reducing the percentage damage of *B. oleracea* crops compared with negative controls (Table 23).

Moreover, the weekly observations revealed that, the percentage damage of *B. oleracea* was significantly ($P \leq 0.05$) higher (15.3 ± 1.2, 17.5 ± 1.5, 18.8 ± 1.7) in 2020 wet season compared with 2019 wet season (13.7 ± 1.0, 14.6 ± 1.1, 15.0 ± 1.3) from week 2 to 4 after treatment applications, respectively (Table 24). But the percentage damage was the same on week 1 and 5 after treatment applications in both wet seasons (Table 24) which might be contributed by higher population of insect pests observed in 2020 wet season compared with 2019 wet season. In addition, percent damage varied significantly between Tengeru experimental site and Boro experimental site (Table 24).

The treatments applied reduced the percentage damage of *B. oleracea* significantly ($P \leq 0.05$) effectively. It was found that, the percentage damage of *B. oleracea* in the 5% concentration of aqueous extract from the mixed plant treated plots was significantly ($P \leq 0.05$) lower (8.8 ± 1.7, 5.3 ± 1.0, 4.4 ± 0.9, 4.4 ± 1.0, 4.4 ± 1.4) as in chlorpyrifos treated plots (9.1 ± 1.7, 4.7 ± 1.1, 4.7 ± 1.1, 5.3 ± 1.2, 4.4 ± 1.1) from week 1 to 5 after applications of the treatments, respectively (Table 24). Then, it was followed by the 10% concentration of aqueous extracts from *C. dichogamus, T. vogelii,* and *S. aromaticum* of aqueous extracts. However, the other concentrations (1% and 5%) and the 2.5% concentration of aqueous extract from the mixed plants) significantly reduced the percentage damage of *B. oleracea* compared with the negative controls (water and water plus soap) (Table 24).

Table 23: Mean percent damage per cabbage crop in the two seasons

Experimental sites and Treatments	Seasons	
	2019 season	2020 season
Experimental sites		
Tengeru	16.2 ± 1.8a	17.1 ± 1.4a
Boro	14.1 ± 1.5b	17.3 ± 2.2a
Treatments		
Water	44.3 ± 2.4a	46.6 ± 4.4a
water + soap	40.3 ± 2.2a	47.8 ± 3.4a
Synthetic pesticide	6.8 ± 0.9ef	4.5 ± 1.1f
C. dichogamus (1%)	17.6 ± 0.6b	18.1 ± 1.0b
C. dichogamus (5%)	14.5 ± 1.8bc	14.4 ± 1.4bcd
C. dichogamus (10%)	11.5 ± 1.2cd	8.4 ± 1.2ef
S. aromaticum (1%)	16.0 ± 1.0b	19.3 ± 1.3b
S. aromaticum (5%)	9.6 ± 0.7de	15.1 ± 1.5bcd
S. aromaticum (10%)	8.3 ± 0.5def	11.6 ± 2.1de
T. vogelii (1%)	15.0 ± 1.7bc	17.5 ± 1.1bc
T. vogelii (5%)	8.5 ± 1.0def	12.3 ± 0.9cde
T. vogelii (10%)	7.4 ± 0.7ef	7.8 ± 1.2ef
Mixed plants (2.5%)	6.9 ± 0.7ef	12.3 ± 1.2cde
Mixed plants (5%)	5.9 ± 1.1f	5.0 ± 1.0f
2 - way ANOVA	**(F- Statistics)**	
Experimental sites (L)	9.29**	0.09ns
Treatments (T)	93.54***	73.44***
L*T	1.13ns	4.59***

Each value is a mean ± standard error of eight replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fisher's Least Significant Difference (LSD) test.

Table 24: Mean damage (%) per cabbage crop on weekly basis for two seasons

Location and Treatments	Week 1 before Treatment	Weeks after treatments				
		1	2	3	4	5
Seasons						
Wet season 1 (2019)	15.1 ± 0.8b	14.1 ± 1.0a	13.7 ± 1.0b	14.6 ± 1.1b	15.0 ± 1.3b	18.4 ± 1.9a
Wet season 2 (2020)	19.6 ± 0.5a	14.5 ± 0.8a	15.3 ± 1.2a	17.5 ± 1.5a	18.8 ± 1.7a	19.8 ± 1.9a
Experimental sites						
Tengeru	20.1 ± 0.7a	16.4 ± 0.9a	15.6 ± 1.1a	15.5 ± 1.3a	16.0 ± 1.2b	19.6 ±1.6a
Boro	14.7 ± 0.6b	12.2 ± 0.8b	13.3 ± 1.1b	16.6 ± 1.5a	17.8 ± 1.7a	18.7 ± 2.1a
Treatments						
W	20.6 ± 1.9a	32.8 ± 3.1a	37.2 ± 3.0a	44.4 ± 3.8a	47.8 ± 3.5a	65.0 ± 3.2a
W + s	19.4 ± 2.5a	27.8 ± 2.9a	35.0 ± 2.5a	45.3 ± 2.9a	50.0 ± 4.4a	61.6 ± 2.7a
S. p	17.8 ± 1.9a	9.1 ± 1.7d	4.7 ± 1.1f	4.7 ± 1.1f	5.3 ± 1.2fg	4.4 ± 1.1g
C. d (1%)	17.8 ± 1.4a	16.3 ± 1.8b	16.9 ± 1.6b	17.8 ± 1.4b	18.4 ± 1.3b	20.0 ± 1.4b
C. d (5%)	18.1 ± 2.0a	11.6 ± 0.9bcd	15.0 ± 2.0b	15.6 ± 1.9bcd	15.0 ± 1.3bcd	15.0 ± 1.7cd
C. d (10%)	15.6 ± 1.5a	10.6 ± 1.0cd	9.1 ± 1.1cdef	9.4 ± 1.1ef	10.6 ± 1.6def	10.0 ± 1.8ef
S. a (1%)	16.3 ± 1.5a	14.7 ± 1.7bc	16.3 ± 1.3b	17.8 ± 1.7b	19.4 ± 1.3b	20.0 ± 1.6b
S. a (5%)	15.0 ± 1.5a	12.5 ± 1.5bcd	13.1 ± 0.9bc	11.6 ± 1.6cde	12.2 ± 1.4cde	12.5 ± 1.5de
S. a (10%)	16.6 ± 1.6a	10.0 ± 1.4cd	10.0 ± 1.6cd	9.4 ± 1.7ef	10.3 ± 1.4def	10.0 ± 1.4ef
T. v (1%)	14.4 ± 1.8a	13.4 ± 1.2bcd	15.9 ± 2.3b	16.6 ± 1.4bc	16.6 ± 0.9bc	18.8 ± 1.5bc
T. v (5%)	17.8 ± 1.9a	11.6 ± 0.9bcd	9.4 ± 1.0cde	10.6 ± 1.1de	9.7 ± 1.2efg	10.6 ± 1.6def
T. v (10%)	15.6 ± 1.4a	10.6 ± 1.5cd	6.3 ± 1.1def	6.9 ± 0.9ef	7.2 ± 1.1efg	6.9 ± 1.1fg
M. p. (2.5%)	19.4 ± 2.2a	10.6 ± 1.0cd	8.4 ± 1.0def	10.3 ± 1.5e	9.7 ± 1.6efg	8.8 ± 1.7efg
M. p. (5%)	19.1 ± 2.3a	8.8 ± 1.7d	5.3 ± 1.0ef	4.4 ± 0.9f	4.4 ± 1.0g	4.4 ± 1.4g
3 - way ANOVA						
Season (S)	31.91***	0.25ns	4.10*	14.07***	17.86***	3.18ns
Experimental sites (L)	45.80***	26.45***	8. 56**	2.00ns	3.94*	1.19ns
Treatments (T)	1.56ns	22.44***	45.70***	75.59***	76.00***	178.72***
S * L	34.49***	35.01***	18.28***	2.64ns	2.49ns	9.29**
S * T	0.58ns	0.66ns	1.61ns	2.37ns	2.71ns	4.36***
L * T	0.98ns	1.00ns	0.59ns	1.76ns	2.62ns	3.43**
S * L * T	1.08ns	1.61ns	2.85ns	5.37***	1.94ns	1.63ns

Each value is a mean ± standard error of eight replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.0 from each other using Fisher's Least Significant Difference (LSD) test.

W - water, w + s - Water plus soap, S. p - Synthetic pesticide, *C. d – Croton dichogamus, S. a – Syzygium aromaticum, T. v – Tephrosia vogelii,* M. p – Mixed plants.

The interactive effects among experimental sites' weather conditions, treatments and seasons significantly ($P \leq 0.05$) enhanced the reduction of the damage of *B. oleracea* compared with the negative controls (water and water plus soap) in the plots (Table 24; Appendix 7). The interaction between treatments and experimental sites was significant ($P \leq 0.05$) in weeks 3, 4, 5 and 6 after treatments application in 2020 season (Appendix 7; Fig. 35) while in 2019 season there was no interactive effects of the treatments and experimental sites' weather conditions.

The interactions of experimental sites' weather conditions and seasons significantly ($P \leq 0.05$) enhanced the lowering of the damage of *B. oleracea* compared with the negative controls (water and water plus soap) in the plots in the weeks 1, 2, 3 and 6 of the treatment experiments (Table 24; Fig. 34). Moreover, the interactions among the experimental sites' weather conditions, treatments and seasons significantly ($P \leq 0.05$) enhanced the reduction of the damage of *B. oleracea* compared with the negative controls (water and water plus soap) in the plots in week 4 of the experimental treatments (Table 24; Fig. 36).

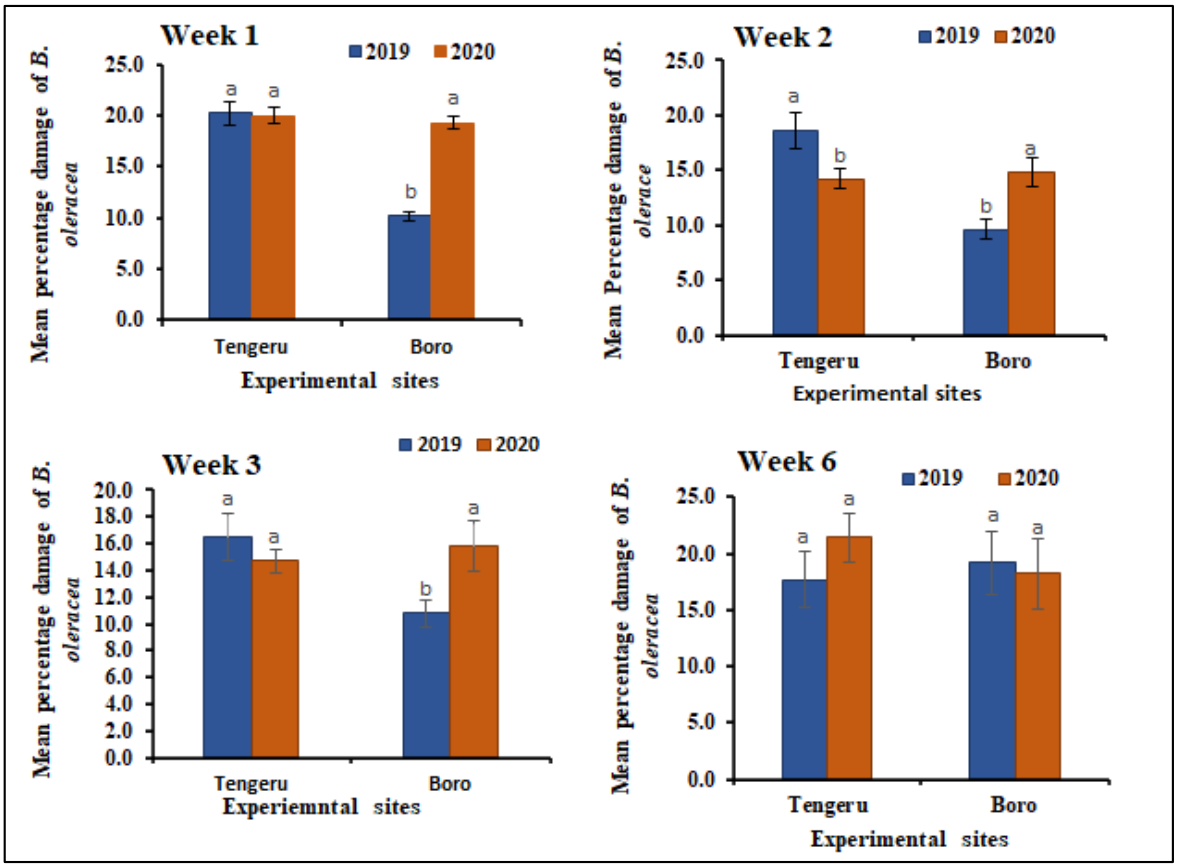
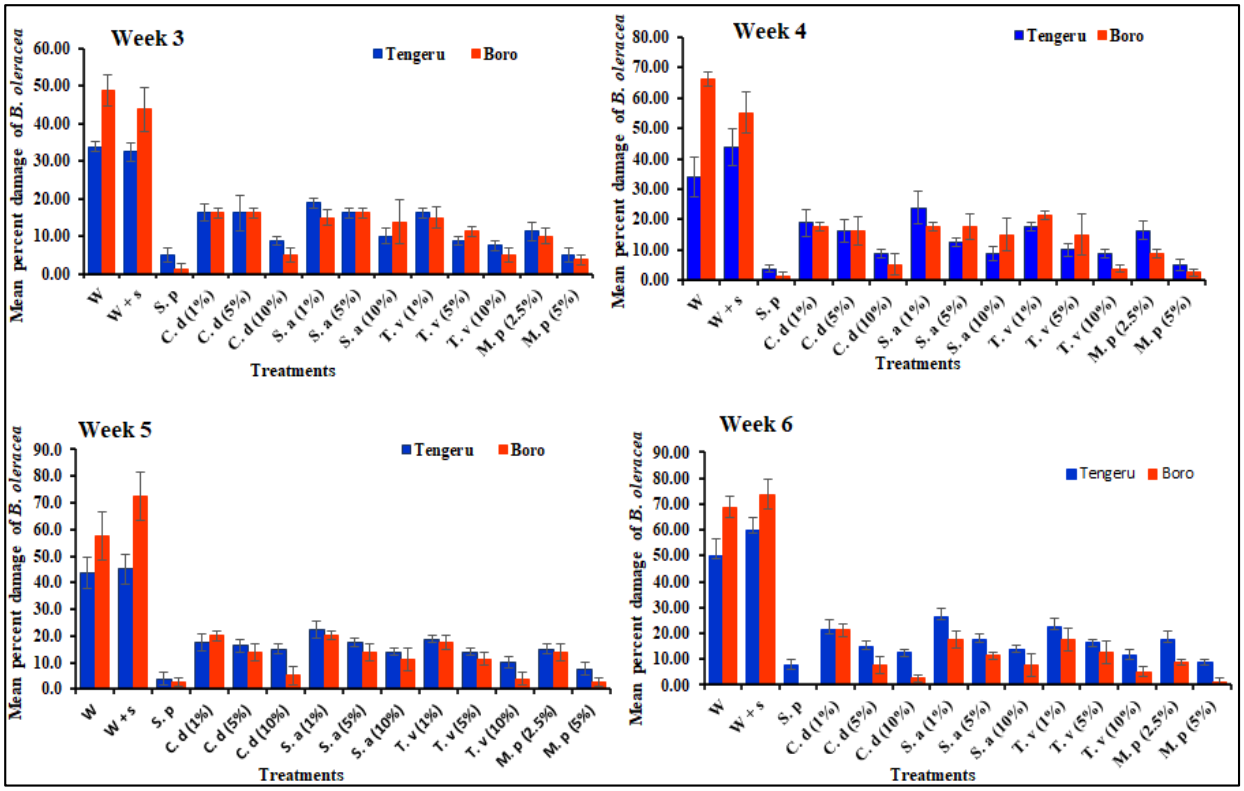
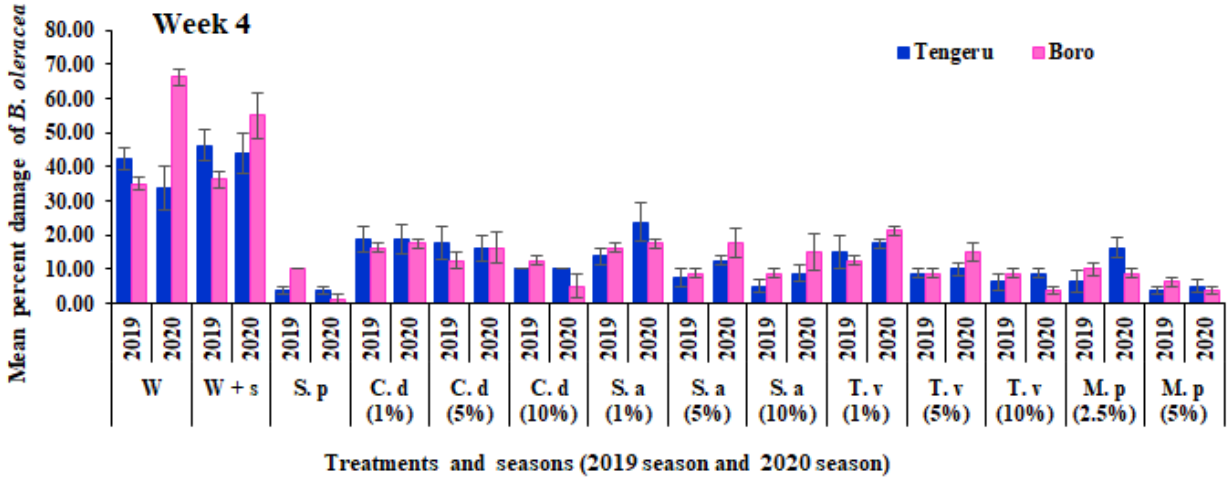


Figure 34: Interaction of experimental sites with the seasons for reducing the damage of *B. oleracea* (Week 1, 2, 3 and 6) in 2019 and 2020 seasons



W - water, w + s - Water plus soap, S. p - Synthetic pesticide, *C. d – Croton dichogamus*, *S. a – Syzygium aromaticum*, *T. v – Tephrosia vogelii*

Figure 35: Interactions of experimental sites with the treatments for reduction of the damage of *B. oleracea* (Week 2,3,4 and 5) in 2020 season



W - water, w + s - Water plus soap, S. p - Synthetic pesticide, *C. d – Croton dichogamus*, *S. a – Syzygium aromaticum*, *T. v – Tephrosia vogelii*, M. p – Mixed plants.

Figure 36: Interactions of experimental sites, seasons and treatments for reduction of the damage of *B. oleracea* (Week 4) in 2020 season

(iv) The level of incidence of infestations of *B. oleracea* caused by insect pests

In general, the level of incidence of infestations of *B. oleracea* in the plots differed significantly ($p \leq 0.001$) at the two experimental sites in the two wet seasons (Table 25). It was found that the incidence level differed significantly between the two experimental sites. So, the incidence level was significantly ($P \leq 0.05$) higher (22.2 ± 2.4%) at Boro experimental site compared with Tengeru experimental site (19.3 ± 2.4%) (Table 25) in 2019 wet season. But, in 2020 wet season the level of incidences was vice versa. It was significantly ($P \leq 0.001$) higher (21.9 ± 1.7%) at Tengeru experimental site compared with Boro experimental site (19.1 ± 2.3%) (Table 25).

Additionally, the level of incidences differed significantly ($P \leq 0.001$) among the plots applied with treatments in the field in the two wet seasons (Table 25). In both wet seasons, the 5% concentration of aqueous extract from the mixed plants had significantly ($P \leq 0.05$) lower (4.8 ± 0.8% and 3.8 ± 1.0%) level of incidences as in synthetic pesticide (6.1 ± 1.3% and 5.2 ± 1.4%) in 2019 wet season and 2020 wet season, respectively (Table 25). Moreover, the effectiveness of the aqueous extracts in reducing the infestation level in *B. oleracea* depended on the concentrations of the aqueous extracts used. Thus, the 10% concentrations from *C. dichogamus, T. vogelii,* and *S. aromaticum* of aqueous extract followed the 5% concentration of aqueous extract from the mixed plants (Table 25) in reducing the incidences in *B. oleracea* crops. The other concentrations (1% and 5%) and 2.5% of aqueous extract from the mixed plants of *C. dichogamus, T. vogelii* and *S. aromaticum* significantly reduced the level of incidences of infestations of *B. oleracea* compared with negative controls (water and water plus soap) (Table 25) in both experimental sites in the two seasons (2019 and 2020 wet seasons).

Table 26 indicates the weekly observations of the level of incidences in the two experimental sites for the two wet seasons. It was revealed that, between the two wet seasons, the level of incidences of *B. oleracea* was significantly ($P \leq 0.05$) higher (16.7 ± 1.1, 17.3 ± 1.3) in 2020 wet season compared with 2019 wet season (7.0 ± 0.9, 15.8 ± 1.3) on weeks 1 and 2 after applications of the treatments (Table 26). However, on weeks 3, 4 and 5 after applications of the treatments, the level of incidence of *B. oleracea* was significantly ($P \leq 0.05$) higher (26.4 ± 2.0, 27.4 ± 2.3, 27.2 ± 2.4) in 2019 wet season compared with 2020 wet season (19.5 ± 1.6, 23.9 ± 1.7, 25.0 ± 2.0).

Also, the weekly observations revealed that, the level of incidences varied significantly ($P \leq 0.05$) between the two experimental sites (Table 26). On week 1 before treatment applications, the level of incidences was significantly higher (12.9 ± 1.1) at Tengeru experimental site compared with Boro experimental site (10.8 ± 1.0) (Table 26). On week 3 after application of the treatments, the level of incidences was significantly higher (24.4 ± 1.9) at Boro experimental site compared with Tengeru experimental site (21.4 ± 1.7) (Table 26). But on weeks 2, 4 and 5, the level of incidences was significantly the same at both experimental sites (Table 26). The proliferation of natural enemies and changes in weather conditions could be the reasons for the variations of the level of incidences between the two experimental sites and the two wet seasons.

The treatments applied reduced significantly the level of incidences of *B. oleracea* between the two experimental sites in the plots and between the two wet seasons. It was found that, the 5% concentration of aqueous extract from the mixed plants had significantly ($P \leq 0.05$) lower (2.6 ± 1.0, 3.6 ± 1.1, 4.7 ± 1.4, 4.4 ± 1.0, 6.1 ± 1.2) the level of incidences compared with other concentrations of aqueous extracts from *C. dichogamus, T. vogelii* and *S. aromaticum* on weeks 1, 2, 3, 4 and 5 after the treatment applications, respectively (Table 26). The 5% concentration of aqueous extract from the mixed plants possessed as lower level of incidences as in synthetic pesticide (5.7 ± 2.0, 3.4 ± 1.0, 7.3 ± 2.3, 6.0 ± 1.4, 5.9 ± 1.2) on weeks 1, 2, 3, 4 and 5 after the treatment applications, respectively (Table 26). Then, it was followed by the 10% concentration of *C. dichogamus, T. vogelii,* and *S. aromaticum* of aqueous extracts. However, the other concentrations (1% and 5%), and the 2.5% concentration of aqueous extract from the mixed plants significantly reduced the level of incidences of *B. oleracea* compared with the negative controls in the plots (water and water plus soap) (Table 26). Moreover, the appendix 8 indicates the weekly observations of the level of incidences in the two experimental sites for the two wet seasons separately.

Table 25: Level of incidences (%) per plot in the field in the two wet seasons

Location and Treatments	Seasons	
	2019 season	**2020 season**
Experimental sites		
Tengeru	19.3 ± 2.4b	21.9 ± 1.7a
Boro	22.2 ± 2.4a	19.1 ± 2.3b
Treatments		
Water	60.0 ± 1.8a	50.8 ± 2.7a
water + soap	57.3 ± 0.8a	51.9 ± 2.6a
Synthetic pesticide	**6.1 ± 1.3fg**	**5.2 ± 1.4h**
C. dichogamus (1%)	28.1 ± 2.2b	24.6 ± 1.4b
C. dichogamus (5%)	17.2 ± 1.4d	18.8 ± 1.0cd
C. dichogamus (10%)	13.9 ± 1.7de	11.9 ± 2.4fg
S. aromaticum (1%)	26.5 ± 1.7bc	25.2 ± 1.3b
S. aromaticum (5%)	15.4 ± 1.7d	19.0 ± 2.3cd
S. aromaticum (10%)	9.8 ± 2.5ef	12.9 ± 1.8efg
T. vogelii (1%)	23.0 ± 1.9c	22.5 ± 1.3bc
T. vogelii (5%)	10.2 ± 1.7ef	15.0 ± 0.5def
T. vogelii (10%)	7.7 ± 1.5fg	8.1 ± 1.2ef
Mixed plants (2.5%)	10.3 ± 1.3ef	17.3 ± 1.8de
Mixed plants (5%)	**4.8 ± 0.8g**	**3.8 ± 1.0h**
2 - way ANOVA	**(F- Statistics)**	
Locations	10.63**	14.39***
Treatments	115.01***	115.30***
Location*treatments	0.36ns	4.91***

Each value is a mean ± standard error of eight replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fisher's Least Significant Difference (LSD) test.

Table 26: Weekly level of incidences (%) per plot in the field in the two seasons (2019 and 2020)

Location and	Weeks after treatments in 2019 and 2020 seasons				
Treatments	**1**	**2**	**3**	**4**	**5**
Seasons					
Season 1 (2019)	7.0 ± 0.9b	15.8 ± 1.3a	26.4 ± 2.0a	27.4 ± 2.3a	27.2 ± 2.4a
Season 2 (2020)	16.7 ± 1.1a	17.3 ± 1.3a	19.5 ± 1.6b	23.9 ± 1.7b	25.0 ± 2.0b
Experimental sites					
Tengeru	12.9 ± 1.1a	16.9 ± 1.2a	21.4 ± 1.7b	25.1 ± 1.9a	26.6 ± 2.0a
Boro	10.8 ± 1.0b	16.2 ± 1.4a	24.4 ± 1.9a	26.2 ± 2.1a	25.6 ± 2.3a
Treatments					
W	30.5 ± 2.8a	42.4 ± 2.4a	62.8 ± 3.4a	66.8 ± 3.9a	74.6 ± 2.8a
W + s	26.8 ± 2.7a	41.1 ± 1.4a	59.4 ± 3.2a	71.0 ± 3.4a	75.1 ± 2.2a
S. p	**5.7 ± 2.0ef**	**3.4 ± 1.0i**	**7.3 ± 2.3fg**	**6.0 ± 1.4f**	**5.9 ± 1.2h**
C. d (1%)	17.2 ± 1.8b	24.9 ± 1.2b	26.2 ± 2.5b	30.1 ± 2.1b	33.5 ± 1.8b
C. d (5%)	12.1 ± 2.3bcd	14.7 ± 1.7ef	19.0 ± 1.9cd	22.1 ± 1.4c	22.0 ± 1.9de
C. d (10%)	6.8 ± 2.4def	9.6 ± 1.8g	14.1 ± 1.5cde	17.6 ± 2.2cd	16.3 ± 3.0ef
S. a (1%)	14.5 ± 1.9bc	22.7 ± 1.9bc	29.3 ± 2.1b	32.8.4 ± 1.5b	30.1 ± 1.9bc
S. a (5%)	9.1 ± 2.0cde	16.5 ± 1.7de	19.3 ± 1.8c	20.7 ± 1.7cd	20.3 ± 1.7de
S. a (10%)	8.1 ± 2.2def	7.9 ± 1.7gh	13.2 ± 2.3def	15.4 ± 2.2d	12.4 ± 2.9fg
T. v (1%)	13.9 ± 2.2bc	19.3 ± 1.3cd	25.4 ± 1.7b	28.9 ± 1.5b	26.2 ± 2.3cd
T. v (5%)	6.1 ± 2.1ef	9.0 ± 1.6g	15.1 ± 1.7cde	15.2 ± 2.2de	17.4 ± 1.9ef
T. v (10%)	5.6 ± 1.2ef	6.1 ± 1.4ghi	9.6 ± 1.3efg	9.4 ± 1.7ef	8.7 ± 1.4gh
M. p. (2.5%)	7.3 ± 2.0def	10.3 ± 1.5fg	15.8 ± 1.8cd	18.8 ± 1.4cd	16.9 ± 2.3ef
M. p. (5%)	**2.6 ± 1.0f**	**3.6 ± 1.1hi**	**4.7 ± 1.4g**	**4.4 ± 1.0f**	**6.1 ± 1.2h**
3 - way ANOVA					
Season (S)	123.38***	3.53ns	54.47***	16.74***	5.15*
Experimental sites (L)	5. 78*	0. 60ns	10.11**	1.55ns	1.10ns
Treatments (T)	24.39***	63.92***	102.70***	155.33***	148.73***
S * L	20.43***	3.53ns	3.61ns	7.81**	19.56***
S * T	0.83ns	1.06ns	3.01ns	7.16***	3.53***
L * T	0.38ns	1.41ns	1.43ns	2.05ns	1.14ns
S * L * T	0.58ns	0.58ns	2.07ns	3.62***	1.87ns

Each value is a mean ± standard error of sixteen replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.0 from each other using Fisher's Least Significant Difference (LSD) test.

W - water, w + s - Water plus soap, S. p - Synthetic pesticide, *C. d – Croton dichogamus, S. a – Syzygium aromaticum, T. v – Tephrosia vogelii,* M. p – Mixed plants.

The interactions among experimental sites' weather conditions, treatments and seasons significantly ($P \leq 0.05$) reduced the level of incidences of *B. oleracea* compared with the negative controls (water and water plus soap) in the plots (Table 25 and 26; Fig. 37, 38 and 39). The interaction between treatments and experimental sites was significant ($P \leq 0.05$) in weeks 3, 4 and 5 after treatment application in 2020 season (Appendix 8; Fig. 38) while in 2019 season there was no significant interaction of the treatments and experimental sites weather conditions. The interactions of experimental sites' weather conditions and seasons was significant ($P \leq 0.05$) in weeks 1, 4 and 5 after the treatment applications of *B. oleracea* compared with the negative controls (water and water plus soap) in the plots (Table 26; Fig. 37) of the treatment experiments. Moreover, the interactions among the experimental sites'

weather conditions, treatments and seasons significantly ($P \leq 0.05$) reduced the level of incidences of *B. oleracea* compared with the negative controls (water and water plus soap) in the plots in week 4 of the experimental treatments (Table 26; Fig. 39).

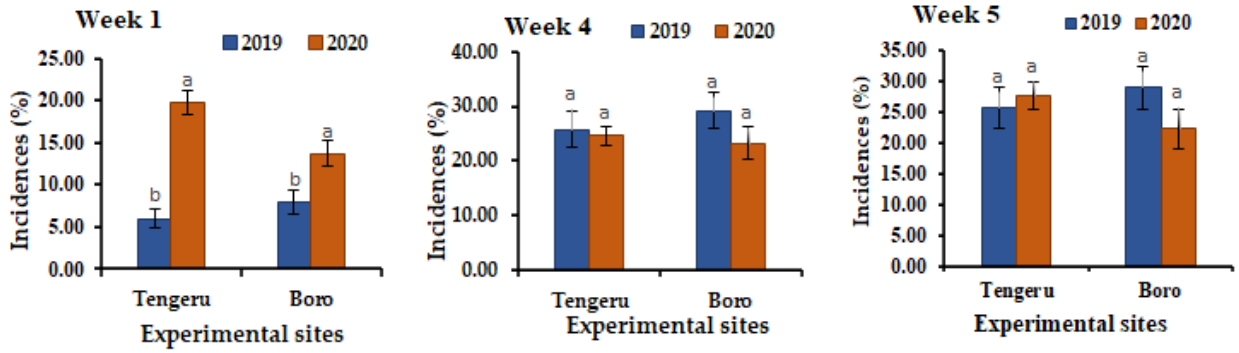
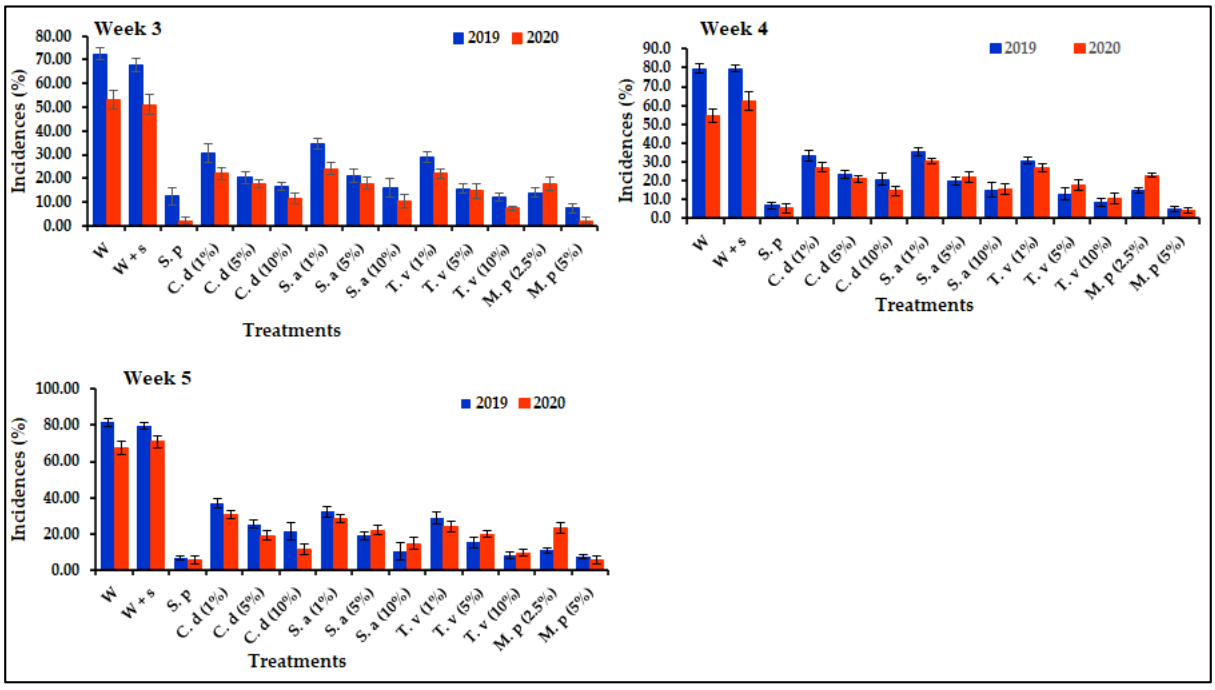
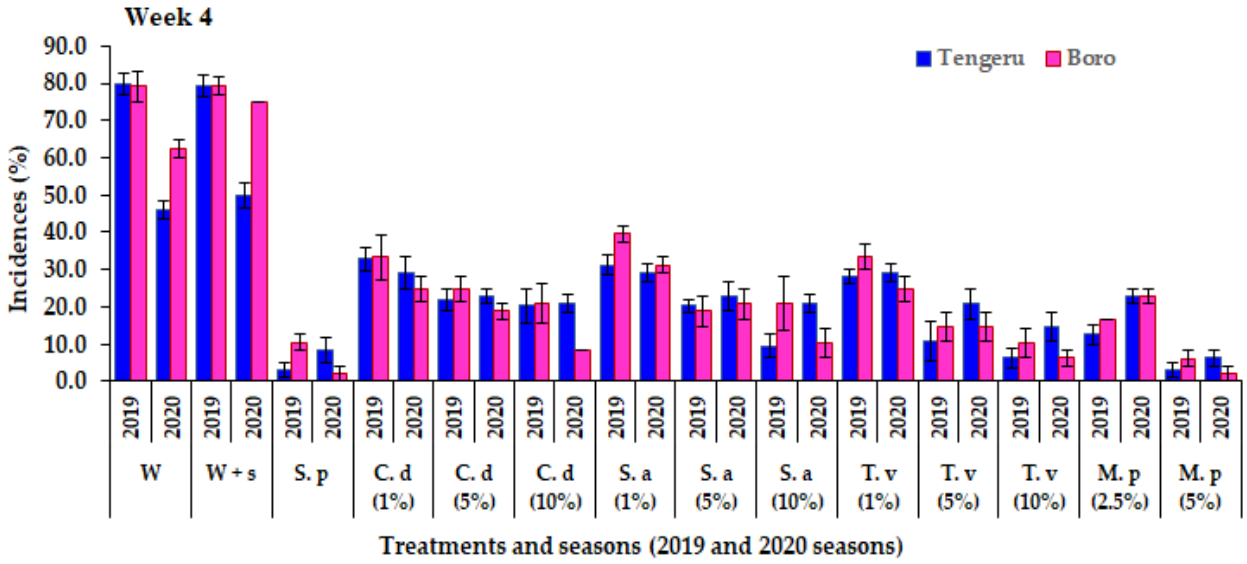


Figure 37: Interaction of experimental sites and the seasons for reduction of the level of incidences of *B. oleracea* (Week 1, 4 and 5) in the two seasons



W - water, w + s - Water plus soap, S. p - Synthetic pesticide, *C. d – Croton dichogamus*, *S. a – Syzygium aromaticum*, *T. v – Tephrosia vogelii*, M. p – Mixed plants.

Figure 38: Interaction of treatments and the seasons for reduction of the level of incidences of *B. oleracea* (Week 3, 4 and 5) in the two seasons

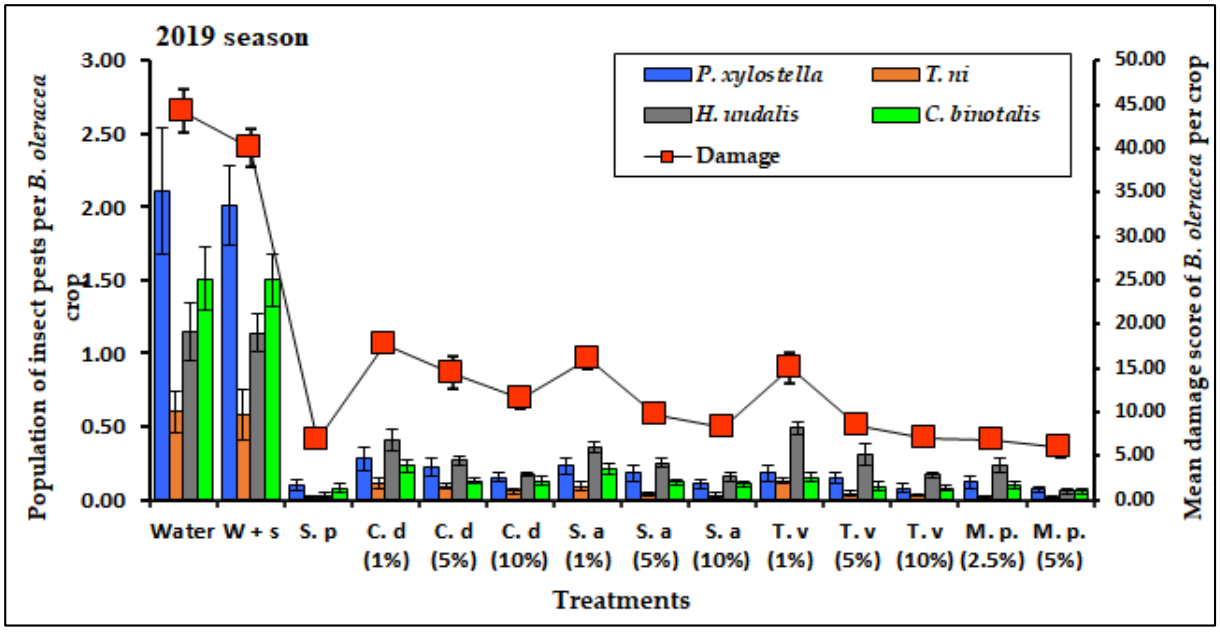


W - water, w + s - Water plus soap, S. p - Synthetic pesticide, *C. d – Croton dichogamus*, *S. a – Syzygium aromaticum*, *T. v – Tephrosia vogelii*, M. p – Mixed plants.

Figure 39: Interaction of treatments, experimental sites and the seasons for reduction of the level of incidences of *B. oleracea* (Week 4) in the two seasons

(v) Relationship between insect pest population and the damage (%) of cabbage crop

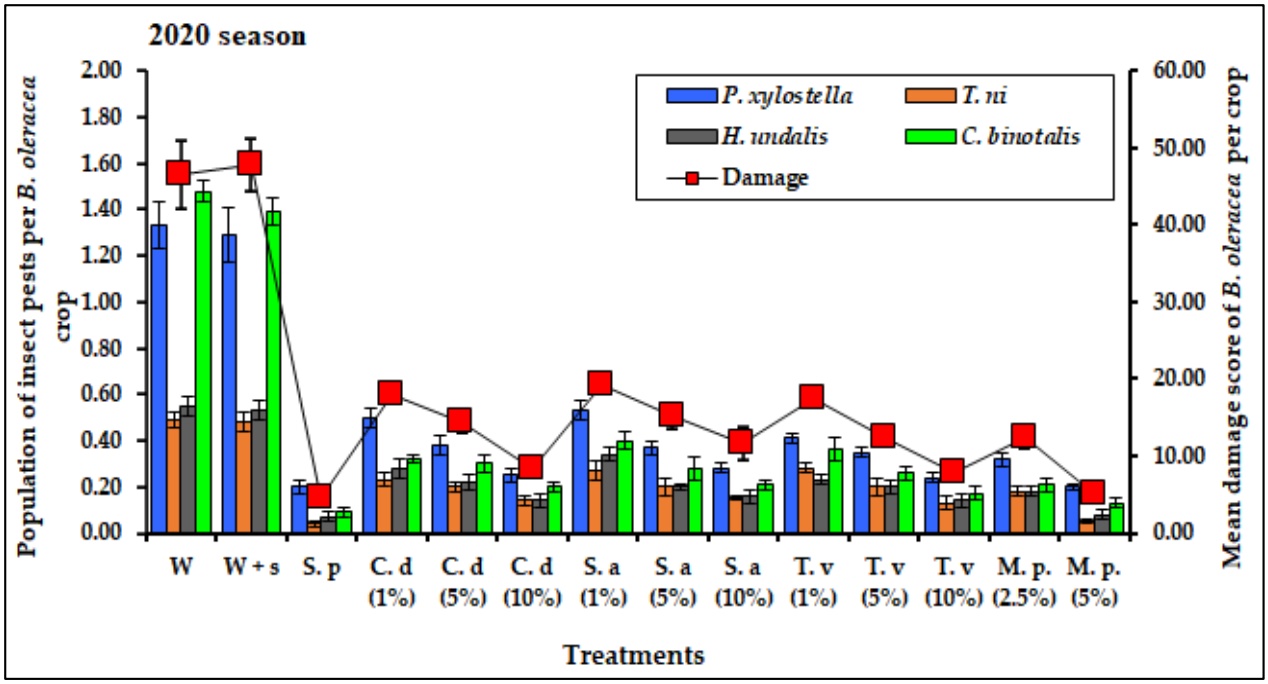
Figure 40 and 41 indicate the relationship between the population abundance of *P. xylostella*, *T. ni*, *H. undalis* and *C. binotalis* and the percentage damage of cabbage (*B. oleracea*) incurred in the negative controls and in the plots sprayed with the treatments in 2019 wet season and 2020 wet season, respectively. The percentage damage of *B. oleracea* was highest in the negative controls (water and water plus soap) due to higher population abundance of *P. xylostella*, *T. ni*, *H. undalis* and *C. binotalis* which caused higher damaging effect compared with the plots which were treated with synthetic pesticide (chlorpyrifos) and aqueous extracts from *T. vogelli* and *S. aromaticum* and *C. dichogamus* in the two wet seasons. Also, there was a relationship between the population abundance of insect pests in this study and the concentrations of the aqueous extracts applied at the field for the protection of *B. oleracea* crop. In case high concentrations of the aqueous extracts were used, the lower percentage damage of *B. oleracea* crop was observed because the insect pests were also lower in higher concentrations of aqueous extracts of plants. Similarly, the concentrations of aqueous extracts from the mixed plant exhibited lower population abundance of *P. xylostella*, *T. ni*, *H. undalis* and *C. binotalis* and finally lower percentage damage of *B. oleracea* in both seasons.



W—water, w + s—Water plus soap, S. p—Synthetic pesticide, *C. d—Croton dichogamus*, *S. a—Syzygium aromaticum*, *T. v—Tephrosia vogelii*, M. p – Mixed plants.

Figure 40: Relationship of the studied insect pests' population and damage (%) of *B. oleracea* in 2019 season

Thus, in 2019 wet season, the 5% concentration of extracts from the mixed plants and synthetic pesticide hosted lowest population abundance of *P. xylostella*, *T. ni*, *H. undalis* and *C. binotalis* which implies that, the damage percent of *B. oleracea* crops caused by those insect pests was also, lower (Fig. 40) compared with other concentrations of the aqueous extracts from *T. vogelli* and *S. aromaticum* and *C. dichogamus*. Mixing of the plant materials during extract preparations might have enhanced the reduction of the insect pests which eventually reduced the damage of the *B. oleracea* crops compared with individual plants at higher concentrations. However, the extracts of the individual plants at higher concentrations (*T. vogelli* (10%) *S. aromaticum* (10%) and *C. dichogamus* (10%)) hosted lower population abundance of *P. xylostella*, *T. ni*, *H. undalis* and *C. binotalis* larvae implying lower damaging percent compared with lower concentrations (1%, 5% and 2.5%) of the plants (Fig. 40). Moreover, *T. vogelii* (5%), *S. aromaticum* (5%) and *C. dichogamus* (5%) aqueous extracts possessed lower percentage damage of *B. oleracea* crop compared with 1% of extracts from each plant due to lower population of insect pests observed (Fig. 40). However, lower percentage damage was also, observed in 1% concentrations from *T. vogelli* and *S. aromaticum* and *C. dichogamus* because it hosted lower population of *P. xylostella*, *T. ni*, *H. undalis* and *C. binotalis* and compared with the negative controls.



W—water, w + s—Water plus soap, S. p—Synthetic pesticide, *C. d—Croton dichogamus, S. a—Syzygium aromaticum, T. v—Tephrosia vogelii*

Figure 41: Relationship of the population of studied insect pests and damage (%) of *B. oleracea* in 2020 season

However, in 2020 wet season, the population abundance of *P. xylostella*, *T. ni*, *H. undalis* and *C. binotalis* was slightly higher compared with 2019 wet season (Fig. 41). Thus, the population abundance of *P. xylostella*, *T. ni*, *H. undalis* and *C. binotalis* in 2020 wet season was also highly related with the damaging effect of *B. oleracea* caused by them. Also, as in 2020 wet season, the population abundance of *P. xylostella*, *T. ni*, *H. undalis* and *C. binotalis* larvae was lowest in Synthetic pesticide and in 5% concentration of aqueous extracts from the mixed plants treated plots implying lowest damaging effect of *B. oleracea* in these two treatments in the field. But it was noted that there were more insect pests in 2020 wet season compared with 2019 season (Fig. 40 and 41) which implied a higher damaging effect compared with 2019 wet season. The heavy rainfall in 2020 wet season could be a reason for higher density of insect pests even in the treated plots compared with the 2019 wet season. That is because heavy rainfall precipitations which tends to wash out the treatments and reduces its efficacy which could have allowed the proliferation of the insect pests at the field. The 10% concentration of aqueous extracts from *S. aromaticum*, *T. vogelii*, *C. dichogamus* and the 2.5% concentration of aqueous extracts from the mixed plants possessed lower population of insect pests implying lower damaging percent compared to lower concentrations of these plants. Moreover, 1% and 5% concentration of *S. aromaticum* and *T. vogelii* possessed lower population abundance of *P.*

xylostella, T. ni, H. undalis and *C. binotalis* which implies lower percentage damage of *B. oleracea* compared with the negative controls in the field (Fig. 41). Therefore, it can be concluded that, the population abundance of *P. xylostella, T. ni, H. undalis* and *C. binotalis* was highly related with percentage damage of *B. oleracea* in the field in the two seasons.

(vi) Correlation matrix of insect pests and the damage (%) of cabbage crop (*B. oleracea*)

The correlation matrix of population abundance of the insect pests and the damage of *B. oleracea* is presented in Table 27, 28, 29 and 30 at Tengeru experimental site and at Boro experimental site in the two wet seasons. The correlation matrix analysis (Table 27, 28, 29 and 30), clearly showed that, the population of *P. xylostella, T. ni, H. undalis* and *C. binotalis* had positive and significantly ($P < 0.001$) very strong relationship with the damage score of the *B. oleracea* crops. In general, the insect pests had strong and positive association between each other meaning that, insect pests do not live in isolation. They live in association with one another forming ecosystems. The living together of insect pests in the field causes huge damaging effect to the crop when control management strategies are not well considered and implemented in time.

Table 27: Correlation matrix of insect pests and damage of cabbage crops at Tengeru site in 2019 wet season

	B. brassicae	*M. persicae*	*P. xylostella*	*T. ni*	*H. undalis*	*C. binotalis*	Damage (%)
B. brassicae	1.000						
M. persicae	0.911***	1.000					
P. xylostella	0.797***	0.875***	1.000				
T. ni	0.797***	0.836***	0.896***	1.000			
H. undalis	0.787***	0.785***	0.771***	0.733***	1.000		
C. binotalis	0.776***	0.859***	0.848***	0.767***	0.923***	1.000	
Damage (%)	0.825***	0.903***	0.881***	0.827***	0.831***	0.896***	1.000

*, **, *** means correlations are significant at n = 56, $p \leq 0.05$, $p \leq 0.01$, $p \leq 0.001$, respectively

Table 28: Correlation matrix of insect pests and damage of cabbage crops at Boro site in 2019 wet season

	B. brassicae	*M. persicae*	*P. xylostella*	*T. ni*	*H. undalis*	*C. binotalis*	Damage (%)
B. brassicae	1.000						
M. persicae	0.779***	1.000					
P. xylostella	0.834***	0.942***	1.000				
T. ni	0.706***	0.863***	0.874***	1.000			
H. undalis	0.877***	0.816***	0.884***	0.760***	1.000		
C. binotalis	0.826***	0.912***	0.964***	0.790***	0.870***	1.000	
Damage (%)	0.861***	0.933***	0.943***	0.813***	0.876***	0.934***	1.000

*, **, *** means correlations are significant at n = 56, $p \leq 0.05$, $p \leq 0.01$, $p \leq 0.001$, respectively

Table 27 indicates the association of the insect pests to damage (%) at Tengeru experimental site in 2019 wet season. All insect pests reported in this study, exhibited positive and very strong association between one insect pest and another and the insect pest and the damage (%) of cabbage (*B. oleracea*) in 2019 wet season at Tengeru and Boro experimental sites. The results of association of the insect pests to the damage of *B. oleracea* obtained at Tengeru experimental site did not differ significantly with those obtained at Boro experimental site (Table 28). So, *B. brassicae* were strongly and positively (0.825 and 0.861) associated with damage (%) at Tengeru and Boro experimental sites, respectively in 2019 wet season. *M. persicae* was strongly and positively (0.903 and 0.933) associated with damage at Tengeru and Boro experimental sites, respectively in 2019 wet season. *P. xylostella* was strongly and positively (0.881 and 0.943) associated with damage at Tengeru and Boro experimental sites, respectively in 2019 wet season. *T. ni* was strongly and positively (0.827 and 0.813) associated with damage at Tengeru and Boro experimental sites, respectively in 2019 wet season. *H. undalis* was strongly and positively (0.831 and 0.876) associated with damage at Tengeru and Boro experimental sites, respectively in 2019 wet season. *Crocidolomia binotalis* was strongly and positively (0.896 and 0.934) associated with damage at Tengeru and Boro experimental sites, respectively in 2019 wet season. Also, some insect pests exhibited higher Pearson's correlation coefficient at Boro experimental site compared with others at the site and others at Tengeru experimental site meaning that, their population abundances and the type of insect mighty have enhanced the damaging effect to cabbage (*B. oleracea*) crop in the field.

Table 29: Correlation matrix of insect pests and damage (%) of *B. oleracea* at Tengeru site in 2020 season

	B. brassicae	*M. persicae*	*P. xylostella*	*T. ni*	*H. undalis*	*C. binotalis*	Damage (%)
B. brassicae	1.000						
M. persicae	0.972***	1.000					
P. xylostella	0.893***	0.863***	1.000				
T. ni	0.802***	0.817***	0.764***	1.000			
H. undalis	0.880***	0.894***	0. 842***	0.820***	1.000		
C. binotalis	0.879***	0.892***	0.827***	0.808***	0.848***	1.000	
Damage (%)	0.937***	0.951***	0.857***	0.829***	0.902***	0.892***	1.000

*, **, *** means correlations are significant at n = 56, $p \leq 0.05$, $p \leq 0.01$, $p \leq 0.001$, respectively

Table 30: Correlation matrix of insect pests and damage of *B. oleracea* at Boro site in 2020 season

	B. brassicae	*M. persicae*	*P. xylostella*	*T. ni*	*H. undalis*	*C. binotalis*	Damage (%)
B. brassicae	1.000						
M. persicae	0.990***	1.000					
P. xylostella	0.948***	0.958***	1.000				
T. ni	0.824***	0.826***	0.811***	1.000			
H. undalis	0.812***	0.815***	0. 843***	0.792***	1.000		
C. binotalis	0.957***	0.970***	0.967***	0.764***	0.780***	1.000	
Damage (%)	0.974***	0.972***	0.953***	0.808***	0.809***	0.958***	1.000

*, **, *** means correlations are significant at n = 56, $p \leq 0.05$, $p \leq 0.01$, $p \leq 0.001$, respectively

The association of the insect pests to damage (%) at Tengeru and Boro experimental sites in 2020 wet season is presented in Table 29 and 30. Similarly, all insect pests reported in the present study, showed positive and very strong association between each other and between insect pest and the damage (%) of *B. oleracea* in 2020 wet season at Tengeru and Boro experimental sites. Moreover, the results of association of the insect pests to damage of *B. oleracea* obtained at Tengeru experimental site did not differ significantly with those obtained at Boro experimental site in 2020 wet season (Table 29 and 30). Thus, *B. brassicae* were strongly and positively (0.937 and 0.974) associated with damage at Tengeru and Boro experimental sites, respectively in 2020 wet season. *M. persicae* was strongly and positively (0.951 and 0.972) associated with damage at Tengeru and Boro experimental sites, respectively in 2020 wet season. *P. xylostella* was strongly and positively (0.857 and 0.953) associated with damage at Tengeru and Boro experimental sites, respectively in 2020 wet season. *T. ni* was strongly and positively (0.829 and 0.808) associated with damage at Tengeru and Boro experimental sites, respectively in 2020 wet season. *H. undalis* was strongly and positively (0.902 and 0.809) associated with damage at Tengeru and Boro experimental sites, respectively in 2020 wet season. *Crocidolomia binotalis* was strongly and positively (0.892 and 0.954) associated at Tengeru and Boro experimental sites, respectively in 2020 wet season.

(vii) Effect of treatments on yield and yield components of cabbage (*B. oleracea*) crop

Canopy spread, cabbage with heads, harvestable cabbages and the weight of cabbage was assessed and recorded to see the effect of the population abundance of the insect pests in relation to the treatments applied at Tengeru and Boro experimental sites in 2019 and 2020 wet seasons and the yields. It was found that, in both seasons (2019 wet season and 2020 wet season), the canopy spread, percent of cabbage with head, percent of harvestable cabbage and weight of cabbage head were insignificant ($p > 0.05$) in the two experimental sites (Table 31).

Moreover, there was significant difference ($P \leq 0.05$) among the treatments applied in the assessed yield components (Table 31) at the two experimental sites and in the two wet seasons (2019 season and 2020 season). The 5% concentration of aqueous extract from the mixed plants and synthetic pesticide treated plots possessed significantly ($P \leq 0.05$) higher (97.1% and 95% in 2019 wet season; 94.8% and 92.7% in 2020 season) percentage of cabbage with head compared with other treatments used, respectively (Table 31). But, the lowest percent of cabbage with head was observed in water and water plus soap in both wet seasons (Table 31). Also, the 5% concentration of aqueous extract from the mixed plants and synthetic pesticide treated plots possessed significantly ($P \leq 0.05$) higher (96.0% and 90.6% in 2019 wet season; 92.7% and 90.6% in 2020 wet season) percentage of harvestable cabbage head compared with other treatments used, respectively (Table 31). Also, the lowest percent of harvestable cabbage was observed in water and water plus soap treated plots (Table 31). Moreover, 5% concentration of aqueous extract from the mixed plants treated plots possessed significantly ($P \leq 0.05$) higher (1.81 kg in 2019 wet season and 1.65 kg in 2020 season) weight of *B. oleracea* head compared with other treatments used (Table 31). Water and water plus soap treated plots possessed cabbage (*B. oleracea*) head with lowest weight (Table 31).

The three- way ANOVA analysis results were presented in Table 32. Between the two wet seasons, it was found that, the canopy spread, percentage of cabbage with head, percentage of harvestable cabbage and the weight of *B. oleracea* head were significantly ($P \leq 0.05$) higher in 2019 wet season (53.3 cm, 81.0%, 76.3% and 1.45 kg) compared with 2020 wet season (47.5 cm, 77.4%, 72.6% and 1.30 kg), respectively (Table 32). Moreover, between the two experimental sites, the percentage of cabbage (*B. oleracea*) with head, percent of harvestable *B. oleracea* and weight of cabbage head were significantly ($P \leq 0.05$) lower at Tengeru experimental site (77.8%, 71.9% and 1.35 kg) compared with Boro experimental site (80.6%, 77.0% and 1.41 kg) (Table 32). The variation of rainfall conditions and infestation of insect pests could be the reason for differences of the weight of cabbage and other yield components in this study. In this study higher rainfall precipitations were recorded in 2020 wet season when compared with 2019 wet season which affected negatively the yield components of *B. oleracea* crop. This could be contributed by the proliferation of a higher number of almost all insect pests observed in 2020 wet season in this study which reduced the proper growth and head formation of the *B. oleracea* crop resulting into low weight of cabbage crops.

Table 31: Assessed yield parameters of cabbage crops in 2019 season and 2020 season

Location and Treatments	2019 season				2020 season			
	Canopy spread (cm)	% Cabbage with heads	% Harvestable cabbage	Weight of cabbage head	Canopy spread (cm)	% Cabbage with heads	% Harvestable cabbage	Weight of cabbage head
Location								
Tengeru	53.1 ± 0.8a	79.8 ± 2.5a	74.9 ± 2.7a	1.40 ± 0.04a	47.3 ± 0.6a	75.8 ± 2.9a	68.9 ± 3.1a	1.29 ± 0.05a
Boro	53.5 ± 0.8a	82.2 ± 2.3a	77.7 ± 2.5a	1.50 ± 0.06a	47.6 ± 0.6a	79.0 ± 2.6a	76.3 ± 2.8a	1.31 ± 0.04a
Treatments								
Water	47.4 ± 1.1f	43.8 ± 4.1f	37.5 ± 3.5f	1.0 ± 0.04fg	40.4 ± 1.3h	36.5 ± 4.7e	30.2 ± 4.4f	0.73 ± 0.04h
W + s	44.6 ± 1.7f	50.0 ± 4.7f	37.5 ± 4.5f	0.94 ± 0.03g	39.5 ± 0.9h	35.4 ± 4.4e	24.0 ± 4.0f	0.62 ± 0.07h
S. p	**52.5 ± 0.7cd**	**95.0 ± 2.9ab**	**90.6 ± 2.9ab**	**1.54 ± 0.04bcd**	**49.8 ± 1.0bcd**	**92.7 ± 2.5a**	**90.6 ± 1.9a**	**1.51 ± 0.04bc**
C. d (1%)	49.1 ± 2.7def	71.9 ± 4.1e	67.7 ± 2.9ef	1.30 ± 0.14def	46.6 ± 1.3efg	70.8 ± 2.7d	67.7 ± 2.7e	1.15 ± 0.06g
C. d (5%)	55.4 ± 1.7bc	91.7 ± 2.2abc	84.4 ± 2.9bc	1.50 ± 0.12bcd	49.3 ± 0.6bcde	86.5 ± 3.5ab	79.2 ± 3.9bcd	1.36 ± 0.03de
C. d (10%)	53.2 ± 1.7cd	87.5 ± 3.9bcd	85.4 ± 3.8bc	1.60 ± 0.16abcd	48.4 ± 0.8cdef	86.5 ± 2.7ab	85.4 ± 2.6abc	1.45 ± 0.05cd
S. a (1%)	52.1 ± 1.0cd	72.2 ± 3.9e	69.4 ± 3.4e	1.27 ± 0.09ef	46.1 ± 1.0fg	75.2 ± 3.2cd	71.5 ± 3.6de	1.20 ± 0.05fg
S. a (5%)	52.7 ± 0.8cd	81.7 ± 3.1de	78.3 ± 2.0de	1.32 ± 0.07bcde	47.2 ± 1.3defg	80.6 ± 2.8bc	76.4 ± 2.6cde	1.31 ± 0.05ef
S. a (10%)	55.3 ± 0.6bc	89.2 ± 2.2bcd	83.3 ± 3.5bc	1.61 ± 0.08abc	51.2 ± 1.3abc	88.9 ± 2.0ab	87.0 ± 1.5ab	1.52 ± 0.03bc
T. v (1%)	53.8 ± 1.8c	82.3 ± 2.9d	78.1 ± 2.7de	1.29 ± 0.09def	45.0 ± 1.0g	76.0 ± 4.6cd	68.8 ± 4.9e	1.22 ± 0.06fg
T. v (5%)	55.4 ± 1.7bc	89.6 ± 2.1bcd	85.4 ± 2.6bc	1.53 ± 0.02bcd	48.4 ± 0.8cdef	86.5 ± 2.7ab	79.2 ± 2.2bcd	1.47 ± 0.03cd
T. v (10%)	60.5 ± 2.5a	91.7 ± 2.2abc	90.6 ± 1.9ab	1.79 ± 0.20ab	52.0 ± 1.40ab	91.7 ± 2.2a	89.6 ± 2.1a	1.61 ± 0.05ab
M. p. (2.5%)	54.0 ± 0.7c	89.2 ± 2.2bcd	83.3 ± 3.5bc	1.62 ± 0.09abc	47.8 ± 1.0defg	81.3 ± 4.7bc	74.0 ± 5.0de	1.45 ± 0.07cd
M. p. (5%)	**59.1 ± 1.7a**	**97.1 ± 2.4a**	**96.0 ± 1.9a**	**1.81 ± 0.17a**	**53.0 ± 1.1a**	**94.8 ± 2.2a**	**92.7 ± 1.9a**	**1.65 ± 0.04a**
2 - way ANOVA	**(F- Statistics)**							
Location	0.28ns	2.62ns	3.60ns	3.78ns	0.08ns	2.96ns	20.98***	0.31ns
Treatments	5.62***	26.68***	34.55***	5.68***	12.65***	33.00***	50.32***	38.83***
Location*treatments	0.11ns	0.99ns	0.69ns	0.68ns	0.57ns	1.10ns	1.38ns	0.56ns

Each value is a mean ± standard error of eight replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fisher's Least Significant Difference (LSD) test.

W—water, w + s—Water plus soap, S. p—Synthetic pesticide, *C. d—Croton dichogamus*, *S. a—Syzygium aromaticum*, *T. v—Tephrosia vogelii*, M. p – Mixed plants.

Table 32: The yield parameters of cabbage crops in the two wet seasons

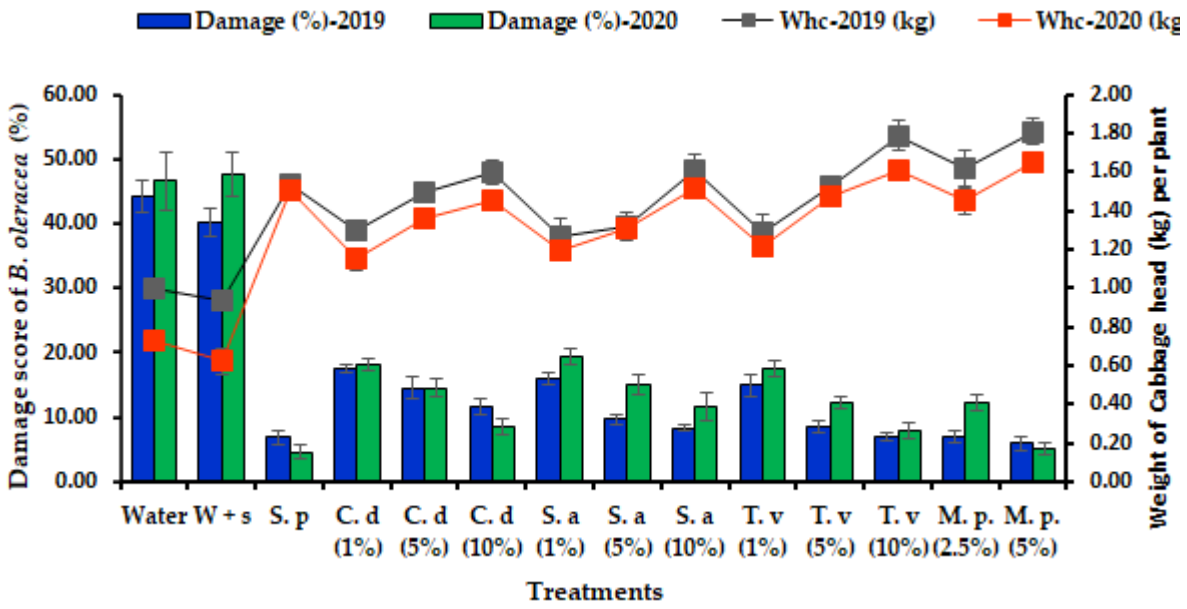
Location and treatments	Canopy spread (cm)	% Cabbage with heads	% Harvestable cabbage	Weight of cabbage head (kg)
Season				
Season 1 (2019)	53.3 ± 0.6a	81.0 ± 1.7a	76.3 ± 1.8a	1.45 ± 0.04a
Season 2 (2020)	47.5 ± 0.4b	77.4 ± 1.9b	72.6 ± 2.1b	1.30 ± 0.03b
Location				
Tengeru	50.2 ± 0.6a	77.8 ± 1.9b	71.9 ± 2.0b	1.35 ± 0.03b
Boro	50.6 ± 0.6a	80.6 ± 1.7a	77.0 ± 1.9a	1.41 ± 0.04a
Treatments				
Water	43.9 ± 1.2f	40.1 ± 3.2h	33.9 ± 2.9h	0.87 ± 0.05g
W + s	42.1 ± 1.2f	42.7 ± 3.6h	30.7 ± 3.4h	0.78 ± 0.06g
S. p	51.1 ± 0.7bcde	93.9 ± 1.8ab	90.6 ± 1.3ab	1.53 ± 0.02c
C. d (1%)	47.8 ± 1.5e	71.4 ± 2.4g	67.7 ± 1.8g	1.22 ± 0.07f
C. d (5%)	52.5 ± 1.2bc	89.1 ± 2.1bc	81.8 ± 2.4cd	1.43 ± 0.07cde
C. d (10%)	50.8 ± 1.1bcde	87.0 ± 2.3cd	85.4 ± 2.2bc	1.52 ± 0.08c
S. a (1%)	49.1 ± 1.0de	73.7 ± 2.5fg	70.5 ± 2.4fg	1.23 ± 0.05f
S. a (5%)	49.7 ± 1.2bcde	81.1 ± 2.0de	77.3 ± 1.6def	1.32 ± 0.04def
S. a (10%)	53.3 ± 0.8ab	89.0 ± 1.4bc	85.1 ± 1.5bc	1.57 ± 0.05abc
T. v (1%)	49.4 ± 1.5cde	79.2 ± 2.7ef	73.4 ± 3.0efg	1.26 ± 0.05ef
T. v (5%)	51.9 ± 1.3bcd	88.0 ± 1.7bcd	82.3 ± 1.8cd	1.50 ± 0.02cd
T. v (10%)	56.2 ± 1.8a	91.7 ± 1.7abc	90.1 ± 1.4ab	1.71 ± 0.11ab
M. p. (2.5%)	50.9 ± 1.3bcde	85.4 ± 2.7cde	78.6 ± 3.2de	1.53 ± 0.06c
M. p. (5%)	56.1 ± 1.3a	95.9 ± 1.6a	94.4 ± 1.7a	1.73 ± 0.09a
3 - way ANOVA				
Season (S)	10.16***	8.75**	10.43**	16.31***
Experimental sites (L)	0. 36ns	5. 58*	20.51***	4.00*
Treatments (T)	14.39***	59.02***	82.85***	21.28***
S * L	0.09ns	0.01ns	3.15ns	2.44ns
S * T	0.79ns	0.92ns	1.38ns	0.58ns
L * T	0.23ns	1.40ns	0.93ns	0.65ns
S * L * T	0.24ns	0.70ns	1.11ns	0.67ns

Each value is a mean ± standard error of sixteen replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fisher's Least Significant Difference (LSD) test.

Whc – weight of cabbage head, W—water, w + s—Water plus soap, S. p—Synthetic pesticide, *C. d—Croton dichogamus, S. a—Syzygium aromaticum, T. v—Tephrosia vogelii*, M. p – Mixed plants.

Figure 42 indicates the relationship between the population of insect pests, damage (%) and the weight of cabbage heads in 2019 wet season and 2020 wet season in the negative controls and the treated plots. It was observed that there was a significant and strong relationship between the percentage damage and the weight of cabbage in both wet seasons (Fig. 42). It was observed that the weight of *B. oleracea* head in different plots was highly related to the damage (%) depending on the treatments applied in the plots and the concentrations of aqueous extracts used. It was found that, the higher weight of cabbage head was recorded in 5% concentration of aqueous extract from the mixed plants, the 10% concentrations of aqueous extract and synthetic pesticide treated plots which was directly related with the lower damage (%) observed in the plots compared with other concentrations of aqueous extracts used (Fig. 42). However,

the other concentrations of the aqueous extracts of *T. vogelii*, *S. aromaticum* and *C. dichogamus* possessed better weight of cabbage compared with weight of cabbage in the negative controls (water and water plus soap) due to higher populations of assessed insect pests which increased the damaging effect of cabbage (*B. oleracea*) crops (Fig. 42). Similarly, the weight of cabbage heads was slightly higher in 2019 wet season compared with 2020 wet season as the damage (%) was also recorded to be slightly lower in 2019 wet season compared with 2020 wet season (Fig. 42).



Whc – weight of cabbage head, W—water, w + s—Water plus soap, S. p—Synthetic pesticide, *C. d—Croton dichogamus, S. a—Syzygium aromaticum, T. v—Tephrosia vogelii*, M. p – Mixed plants.

Figure 42: Relationship of weight of cabbage head with damage score (%)

(viii) Correlation matrix of insect pests and yield components of *B. oleracea*

Correlation matrix of insect pests, damage of *B. oleracea*, canopy spread, % cabbage with heads, % harvestable cabbage and weight of cabbage head in 2019 and 2020 wet seasons are presented in Table 33 and 34, respectively. Generally, in 2019 and 2020 wet season, the population abundance of insect pests and damage score had strong and negative correlation with canopy spread, percent of cabbage with head, percent of harvestable cabbage and weight of cabbage heads (Table 33 and 34). Thus, *P. xylostella* had negative (r = -0.4766, -0.6085, -0.7028, -0.4707 in 2019 wet season and r = -0.6919, -0.8377, -0.8413, -0.7178 in 2020 season) and significantly ($P < 0.01$) strong correlation with canopy spread, percent of cabbage with head, percent of harvestable cabbage and weight of cabbage heads, respectively (Table 33 and 34). *T. ni* had negative (r = -0.4547, -0.6903, -0.7117, -0.4522 in 2019 wet season and r = -

0.6647, -0.7372, -0.7788, -0.6997 in 2020 wet season) and significantly ($P < 0.01$) strong correlation with canopy spread, percent of cabbage with head, percent of harvestable cabbage and weight of cabbage heads, respectively (Table 33 and 34). *Helulla undalis* had negative (r = -0.5565, -0.7433, -0.7927, -0.4976 in 2019 season and r = -0.6633, -0.7731, -0.8194, -0.7173 in 2020 wet season) and significantly ($P < 0.01$) strong correlation with canopy spread, percent of cabbage with heads, percent of harvestable cabbages and weight of cabbage heads, respectively (Table 33 and 34). *Croton binotalis* had negative (r = -0.5412, -0.7889, -0.8227, -0.5086 in 2019 wet season and r = -0.6243, -0.7074, -0.6861, -0.5893 in 2020) and significantly ($P < 0.01$) strong correlation with canopy spread, percent of cabbage with head, percent of harvestable cabbages and weight of cabbage heads, respectively (Table 33 and 34). Damage score (%) had negative (r = -0.5320, -0.8282, -0.8632, -0.5791 in 2019 and r = -0.7100, -0.8423, -0.8408, -0.7357 in 2020 season) and significantly ($P < 0.01$) strong correlation with canopy spread, percent of cabbage with head, percent of harvestable cabbage and weight of cabbage heads, respectively (Table 33 and 34). Also, as reported previously, the insect pests correlated positively and strongly to one another. This scenario significantly increased the damage (%) of cabbage (*B. oleracea*) crops in the field. Moreover, each assessed yield parameter positively and strongly correlated to one another (Table 33 and 34). It is clear from the large negative Pearson's correlation coefficient (r) values observed in association of some insect pests, the damage (%) and the yield components that higher population of insect pests caused intense damaging effect in 2020 wet season relative to 2019 wet season.

Table 33: Correlation matrix of insect pests, damage of *B. oleracea* and assessed yield parameters in 2019 season

	P. xylostella	***T. ni***	***H. undalis***	***C. binotalis***	**Damage of *B. oleracea* (%)**	**Canopy spread (cm)**	**% Cabbage with heads**	**% Harvestable cabbage**	**Weight of cabbage head (kg)**
P. xylostella	1.0000								
T. ni	0.6203***	1.0000							
H. undalis	0.6813***	0.7299***	1.0000						
C. binotalis	0.6748***	0.7769***	0.9020***	1.0000					
Damage of *B. oleracea* (%)	0.7871***	0.7990***	0.8411***	0.8912***	1.0000				
Canopy spread (cm)	-0.4766***	-0.4547***	-0.5565***	-0.5412***	-0.5320***	1.0000			
% Cabbage with heads	-0.6085***	-0.6903***	-0.7433***	-0.7889***	-0.8282***	0.5436***	1.0000		
% Harvestable cabbage	-0.7028***	-0.7117***	-0.7927***	-0.8227***	-0.8632***	0.6035***	0.9261***	1.0000	
Weight of cabbage head (kg)	-0.4707***	-0.4522***	-0.4976***	-0.5086***	-0.5791***	0.5645***	0.5765***	0.6575***	1.0000

*, **, *** means correlations are significant at n = 112, $p \leq 0.05$, $p \leq 0.01$, $p \leq 0.001$, respectively.

Table 34: Correlation matrix of insect pests, damage of *B. oleracea* and assessed yield parameters in 2020 season

	P. xylostella	***T. ni***	***H. undalis***	***C. binotalis***	**Damage of *B. oleracea (%)***	**Canopy spread (cm)**	**% Cabbage with heads**	**% Harvestable cabbage**	**Weight of cabbage head (kg)**
P. xylostella	1.0000								
T. ni	0.7862***	1.0000							
H. undalis	0.8319***	0.8009***	1.0000						
C. binotalis	0.8509***	0.6953***	0.6734***	1.0000					
Damage of *B. oleracea* (%)	0.8998***	0.7994***	0.8045***	0.9080***	1.0000				
Canopy spread (cm)	-0.6919***	-0.6647***	-0.6633***	-0.6243***	-0.7100***	1.0000			
% Cabbage with heads	-0.8377***	-0.7372***	-0.7731***	-0.7074***	-0.8423***	0.7083***	1.0000		
% Harvestable cabbage	-0.8413***	-0.7788***	-0.8194***	-0.6861***	-0.8408***	0.7410***	0.9158***	1.0000	
Weight of cabbage head (kg)	-0.7178***	-0.6997***	-0.7173***	-0.5893***	-0.7357***	0.7071***	0.7365***	0.7781***	1.0000

*, **, *** means correlations are significant at n = 112, $p \leq 0.05$, $p \leq 0.01$, $p \leq 0.001$, respectively

4.1.3 The chemical compounds present in *S. aromaticum* and *C. dichogamus*

The GC-MS analysis results revealed the presence of different phytochemical compounds from DCM-MeOH extracts of *Croton dichogamus* and *Syzygium aromaticum.* The mass spectra of the detected compounds from the leaf and bud extracts of *Croton dichogamus* and *Syzygium aromaticum* were compared with the spectra of the known compounds stored in the NIST library, respectively. The name of the compound, retention time, molecular weight and molecular formular of the compounds contained in these extracts are presented in Table 35 and 36.

(i) *Syzygium aromaticum*

In this study, the following phytochemicals were obtained from DCM-MeOH extract of *S. aromaticum* by GC-MS analysis;- Phenol, 2-methoxy-3-(2-propenyl)-; Phenol, 4-allyl-2-methoxy- (Eugenol); (1S,4E,9R)-4,11,11-trimethyl-8-methylidenebicyclo [7.2.0] undec-4-ene (β-caryophyllene); α-Caryophyllene; α-Farnesene-(E); (1S,8aR)-1-Isopropyl-4,7-dimethyl-1,2,3,5,6,8a-hexahydronaphthalene; Phenol, 2-methoxy-4-propenyl-, acetate (Eugenol acetate); Caryophyllene epoxide; Caryophylla-4(12),8(13)-dien-5.beta.-ol; cyclopropa[c,d]pentalene-1,3-dione, hexahydro-4-(2-methyl-2-propenyl)-2,2,4-trimethyl; cis-9, cis-12-octadecadienoic acid, picolinyl ester; Limonene oxide, cis-(-)-; 2,3,4-Trimethoxyacetophenone; Benzyl Benzoate and Cyclobutanecarboxylic acid, 2-methyloct-5-yn-4-yl ester. Among those phytochemical compounds obtained, the compounds with higher percent in brackets identified were; - Eugenol (4-Allyl-2-methoxyphenol) (52.66%); Phenol, 2-methoxy-4-propenyl-, acetate (Eugenol acetate) (20.46%); (1S,4E,9R)-4,11,11-trimethyl-8-methylidenebicyclo [7.2.0] undec-4-ene (β-caryophyllene) (7.52%); Phenol, 2-methoxy-3-(2-propenyl)- (4.17%); α-Caryophyllene (1.36%) and Caryophyllene epoxide (1.04%) (Table 35). Similarly, previous studies reported the presence of tannins, saponins, flavonoids, terpenoids, alkaloids and phenolic compounds. For instance, Tian *et al*. (2015), reported five constituents in the essential oil extracted from *S. aromaticum* by GC-MS, accounting to 99.89% by which the major components were eugenol (88.61%), eugenol acetate (8.89%) and β-caryophyllene (1.89%). The difference in concentrations between the previous studies and the present study is caused by the variation of vegetative state, growing season and the places of origin (Fu *et al.*, 2007; Samarasekera *et al.*, 2008; Srivastava *et al.*, 2005). Therefore, these chemical compounds obtained in *S. aromaticum* mighty be responsible for insecticidal efficacy against

cabbage (*B. oleracea*) insect pests in the field and when larvicidal action was tested against *P. xylostella* and *C. binotalis* larvae.

Table 35: Chemical constituents from *S. aromaticum* identified by GC-MS

RT (min)	Peak area (%)	Compound name	Molecular formula	Molecular weight (g/mol)	References
17.569	4.17	Phenol, 2-methoxy-3-(2-propenyl)-	$C_{10}H_{12}O_2$	164.20	Kiran and Prakash (2015)
18.930	52.66	Phenol, 4-allyl-2-methoxy- (Eugenol)	$C_{10}H_{12}O_2$	164.20	Tian *et al.* (2015)
19.434	7.52	(1S,4E,9R)-4,11,11-trimethyl-8-methylidenebicyclo [7.2.0] undec-4-ene (β-caryophyllene)	$C_{15}H_{24}$	204.35	Tian *et al.* (2015)
20.304	1.36	α-Caryophyllene	$C_{15}H_{24}$	204.35	Da Silva *et al.* 2015)
21.271	0.49	α-Farnesene-(E)	$C_{15}H_{24}$	204.35	Boncan *et al.* (2020)
21.843	0.32	(1S,8aR)-1-Isopropyl-4,7-dimethyl-1,2,3,5,6,8a-hexahydronaphthalene	$C_{15}H_{24}$	204.35	Boncan *et al.* (2020)
22.804	20.46	Phenol, 2-methoxy-4-propenyl-, acetate (Eugenol acetate)	$C_{12}H_{14}O_3$	206.24	Boncan *et al.* (2020); Tian *et al.* (2015)
23.931	1.04	Caryophyllene epoxide	$C_{15}H_{24}$	220.35	Tian *et al.* (2015)
25.198	0.70	Caryophylla-4(12),8(13)-dien-5.beta.-ol	$C_{15}H_{24}O$	220.35	Da Silva *et al.* (2015); Tian *et al.* (2015)
25.665	0.36	cyclopropa[c,d]pentalene-1,3-dione, hexahydro-4-(2-methyl-2-propenyl)-2,2,4-trimethyl	$C_{15}H_{20}$	232.32	Shooshtari *et al.* (2013)
25.848	0.72	cis-9, cis-12-octadecadienoic acid, picolinyl ester	$C_{24}H_{37}NO_2$	371.56	Zafari-Shayan *et al.* (2016)
		Limonene oxide, cis-(-)-	$C_{10}H_{16}O$	152.23	Sessini *et al.* (2020)
26.878	0.50	2,3,4-rimethoxyacetophenone	$C_{11}H_{14}O_4$	210.23	Shooshtari *et al.* (2013)
28.320	0.36	Benzyl Benzoate	$C_{14}H_{12}O_2$	212.24	Johnson *et al.* (2017)
28.801	0.39	Cyclobutanecarboxylic acid, 2-methyloct-5-yn-4-yl ester	$C_{14}H_{22}O_2$	222.32	Zafari-Shayan *et al.* (2016)

(ii) *Croton dichogamus*

Previous phytochemical study analysis of *Croton species* revealed the presence of alkaloids, phenolics, terpenoids and volatile oils, with a wide range of diterpenoid classes predominating (Salatino *et al.*, 2007). In addition, previous investigation of the leaves of *C. dichogamus*, collected in Kenya, led to the identification of two main crotofolane diterpenoids, crotoxide A and B (Jogia *et al.*, 1989). Similarly; the present investigation revealed the following phytoconstituents from DCM-MeOH extract of *C. dichogamus*;- Distannoxane, hexabutyl-; Silane, (5β-pregnane-3α,11β,17,20α,21-pentaylpentaoxy)pentakis[trimethyl; 3-Bromo-4-hydroxy-2, 3'-dimethyl-5,5',8,8'-tetramethoxy-1,2'-binaphthalene-1',4'-dione; Cyclohexasiloxane, dodecamethyl; 2-Tetradecene (E) -; 7-Hexadecene (Z); 1-Tetradecene; Cycloheptasiloxane, tetradecamethyl-; Pentasiloxane, dodecamethyl; 2H-1, 4-Benzodiazepin-2-one 7-bromo-1,3-dihydro-5-phenyl-1-(4-(4-phenylpiperain-1-l) butyl)-; Phenol, 2,4-bis (1,1-dimethylethyl); 1-Heptadecene; 3-hexadecene, (Z) -; 2-Tetradecene, (E)-; 1-Nonadecene; 9-Tricocene, (Z); E-14-Hexadecenal; 1-Nonadecene; 1-Dococene; Trichloroacetic acid, tetradecyl ester; 9-Tricocene, (Z); Cyclotetracosane; Cyclotrisiloxane, hexamethyl; Benzo(h)quinoline, 2,4-dimethyl; 1,2-Benzenediol, 3,5-bis(1,1-dimethylethyl)-4,6-Bis (4-chloro-3-(trifluoromethyl)phenoxy)-2-pyrimidinol; N-(2,4,6-Trichlorobenzoyl)-4-chlorophenyl-3-morpholinopyrrol-2-carboxylic acid, methyl ester; Cholestan-6-en-3-ol, O-acetyl-24-methyl-5,8-(tetrahydrofuran-2,5-dione-3,4-diyl); eta-Pentamethylcyclopentadienyl-ethylisonitril-(N,N,N',N'-tetramethylethin-1,2-diamin) molybdaeniodi. Among those chemical compounds obtained from the extracts of *C. dichogamus*, these compounds;- 4,6-Bis (4-chloro-3-(trifluoromethyl) phenoxy)-2-pyrimidinol (25.08%); Cholestan-6-en-3-ol (18.63%); 1-Heptadecene (7.34%); 1-Tetradecene (6.30%); Distannoxane, hexabutyl- (5.93%); 2,4-Di-tert-butylphenol (5.26%); (Z)-9-Tricosene and Trichloroacetic acid, tetradecyl ester (5.23%) were present in larger concentrations. Similarly, the chemical compounds obtained in *C. dichogamus* by using GC-MS analysis could be resiponsible for insecticidal efficacy and larvicidal activities tested against the studied insect pests.

Table 36: Chemical compounds from *C. dichogamus* identified by GC-MS

RT (min)	Peak area (%)	Compound name	Molecular formula	Molecular weight (g/mol)	References
8.277	5.93	Distannoxane, hexabutyl-	$C_{24}H_{54}OSn_2$	596.10	Buchweitz *et al.* (2013)
9.450	1.69	Cyclohexasiloxane, dodecamethyl-	$C_{12}H_{36}O_6Si_6$	444.92	Mary and Giri (2017)
11.275	6.30	1-Tetradecene	$C_{14}H_{28}$	196.37	Buchweitz *et al.* (2013)
11.767	0.96	Cycloheptasiloxane, tetradecamethyl	$C_{14}H_{42}O_7Si_7$	519.07	Mary and Giri (2017)
13.009	5.26	**2**,**4**-Di-tert-butylphenol	$C_{14}H_{22}O$	206.32	Ren *et al.* (2019)
13.970	7.34	1-Heptadecene	$C_{17}H_{34}$	238.50	Adebisi *et al.* (2019)
16.402	5.07	1-Nonadecene	$C_{19}H_{38}$	266.50	Tonisi *et al.* (2020)
16.402	5.07	(Z)-9-Tricosene	$C_{23}H_{46}$	322.60	Buchweitz *et al.* (2013)
18.611	5.23	Trichloroacetic acid, tetradecyl ester	$C_{16}H_{29}C_{l3}O$	359.76	Mary and Giri (2017)
20.636	1.34	Cyclotetracosane	$C_{24}H_{48}$	336.6	Vahedi *et al.* (2013)
30.701	0.83	Cyclotrisiloxane, hexamethyl	$C_6H_{18}O_3Si_3$	222.46	Alhazmi *et al.* (2019)
31.148	25.80	4,6-Bis (4-chloro-3-(trifluoromethyl) phenoxy)-2-pyrimidinol	$C_{21}H_{16}Cl_2F_6N_2O_3Si$	557.3	MacLachlan and Hamilton (2010)
31.199	4.44	N-(2,4,6-Trichlorobenzoyl)-4-chlorophenyl-3-morpholinopyrrol-2-carboxylic acid, methyl ester	$C_{23}H_{18}Cl_4N_2O_4$	528.2	MacLachlan and Hamilton (2010)
31.308	18.63	Cholestan-6-en-3-ol	$C_{27}H_{48}O$	388.7	Ibraheam *et al.* (2017)

4.2 Discussion

4.2.1 Larvicidal actions of extracts against *P. xylostella* and *C. binotalis* larvae

This study investigated the larvicidal actions of extracts of *S. aromaticum*, *T. vogelii* and *C. dichogamus* against *P. xylostella* and *C. binotalis* larvae. The DCM-MeOH (1:1) extracts of *S. aromaticum*, *T. vogelii* and *C. dichogamus* with LC_{50} value of 0.081 mg/mL, 0.377 mg/mL and 0.127 mg/mL showed larvicidal activity against the third and the fourth instar larvae of *C. binotalis* respectively after 24 hours of exposure. Also, the extracts of *S. aromaticum*, *T. vogelii* and *C. dichogamus* exhibited larvicidal activity against *P. xylostella* larvae with the LC_{50} value of 0.081 mg/mL, 0.865 mg/mL and 0.105 mg/mL, respectively. These results agree with other

studies (Akhtar *et al.*, 2007; Atshan *et al*., 2017; Huang & Renwick, 1995; Rathi & Gopalakrishnan, 2010). Atshan *et al*. (2017) reported that, pesticidal plants possess chemical compounds which exhibit larvicidal actions against many larvae of insects. For instance, according to Atshan *et al*. (2017), the seed extract of *Lantana camara*, *Sapindus trifoliatus*, *Solanum trilobatum* and *Ceiba pentandra* exhibited larvicidal activity of 25 to 100 mg/kg against *Trichoplusia ni* and *Pieris brassicae* larvae. Similarly, according to Huang and Renwick (1995) and others, plant extracts act as good source of antifeedant, repellent and growth regulator agents to insect pest larvae. Therefore, clearly, the pesticidal plant extracts used in this study were highly effective in killing *C. binotalis* and *P. xylostella* larvae and hence, they showed the insecticidal toxicity property against the targeted pests at early stages of growth and development.

The results of this study also showed that the mortality percentage of *C. binotalis* and *P. xylostella* larvae increased with increase in concentration of the extracts and time of exposure. The larvicidal activities of the pesticidal plants used in this study is contributed by the complex mixtures of phytochemicals of the bioactive chemical compounds in plants such as alkaloids, flavonoids, terpenoids and essential oils found in them. These results also concur with Samatha *et al.* (2012) who reported that the insecticidal activities of pesticidal plants are contributed by the presence of different types of botanicals like alkaloids, terpenoids and phenolic compounds. In this study, the extraction of extracts containing chemical compounds was achieved by using total extraction solvents DCM – MeOH (1:1) which extracted both the polar and non-polar compounds from the pesticidal plants used. Hence the extracts obtained from *S. aromaticum*, *T. vogelii* and *C. dichogamus* mighty have possessed both polar and non-polar compounds which could had caused larvicidal and toxicity effects against *C. binotalis* and *P. xylostella* larvae. Belmain *et al*. (2012) and Grzywacz *et al*. (2014) reported the presence of deguelin, rotenone, sarcolobine, α-toxicarol and tephrosin in *T. vogelii* which mighty be responsible for the larvicidal actions against *C. binotalis* and *P. xylostella* larvae whereby rotenone is the most toxic compound to insect pests at all stages of their growth.

Moreover, the GC-MS analysis of extracts from *S. aromaticum* revealed the presence of eugenol, β-caryophyllene, α-humulene and eugenol acetate in higher concentrations. According to Araujo *et al*. (2016), among these chemical compounds, eugenol was found to be the most toxic bioactive compound which could be responsible for larvicidal activities on *C. binotalis* and *P. xylostella* larvae. Also, *Croton dichogamus* extracts comprise alkaloids,

phenolics and terpenoids which have toxicity, repellent and deterrent effects (Aldhaher *et al*., 2017; Silva *et al*., 2018) toward insect pests. Therefore, those compounds could be responsible for the larvicidal efficacy of *Croton dichogamus* against *C. binotalis* and *P. xylostella* larvae.

4.2.2 Population dynamics of cabbage insect pests in response to weather conditions

The population abundance of insect pests observed in this study differed significantly from one experimental location to another and from 2019 wet season to 2020 wet season. Those results could be attributed to the variations of weather conditions of the experimental sites and the seasons which enhanced either the building up or lowering down of population of insect pests on cabbage crops (*B. oleracea*) at the field. The variation of weather conditions such as heavy rainfall, variation of temperatures, and high humidity have strong influences on the population abundance of the insects and their ecosystems (Patra *et al.*, 2013). From this study, in 2019 wet season, the mean maximum and minimum temperatures of Boro experimental site were 25.16 and 16.11 ℃, respectively while those of Tengeru experimental site were 29.54 and 16.91 ℃ respectively (Mpumi *et al.*, 2021). The mean rainfall precipitation of Boro and Tengeru experimental sites were 148.05 and 70.81 mm respectively (Mpumi *et al.*, 2021). Similarly, in 2020 wet season, the mean maximum and minimum temperatures of Boro experimental site were 24.3 and 15.7 ℃, respectively, while at Tengeru experimental site the mean maximum and minimum temperatures were 28.0 and 17.1 ℃, respectively. The mean rainfall precipitation at Tengeru and Boro experimental sites were 253.7 and 209.4 mm, respectively in 2020 wet season. It was clear that higher rainfall precipitation was observed during the study period in 2020 wet season compared with 2019 wet season in both experimental sites.

The variations in rainfall precipitations and temperatures in the two experimental sites affected significantly ($P \leq 0.05$) the population abundance of the insect pests in the two wet seasons. For instance, *M. persicae* and *B. brassicae,* were significantly higher in 2020 wet season compared with 2019 wet season. Moreover, it was noted that, *M. persicae* and *B. brassicae,* were significantly higher at Tengeru experimental site compared with Boro experimental site which mighty have been contributed by differences in weather conditions between the two experimental sites. Aphids' population build up vary from season to season and from location to location whereas the population abundances of *M. persicae* were significantly ($P \leq 0.05$) higher in 2020 wet season compared with 2019 wet season. These variations in temperatures and rainfall precipitations could have affected significantly ($P \leq 0.05$) the population abundance of *B. oleracea* insect pests in the two wet seasons. The results concur with Patra *et*

al. (2013) who reported that cabbage insect pests are affected either positively or negatively with variations in seasons due to variations in rain precipitations and temperatures.

In addition, *P. xylostella* larvae were significantly higher at Boro experimental site relative to Tengeru experimental site and varied significantly from season to season. These results agree with Patra *et al*. (2013) and Tanyi *et al*. (2018) who reported that, the incidence and infestations of *P. xylostella* on cabbage crops vary from region to region. The rainfall precipitations and temperatures of Boro experimental site might have contributed the higher abundances of *P. xylostella* larvae compared with Tengeru study site whereas the abundance was low. The results concur with Tanyi *et al*. (2018) and Patra *et al*. (2013) who reported that, population building up of *P. xylostella* is favoured by warm conditions and rain precipitations of the region and the season. Moreover, Ayalew (2006) reported that rain precipitations and temperatures ranging from 25 to 33 °C have significant influences on the population of *P. xylostella*. According to them, the increase in population of *P. xylostella* was reported to be positively and strongly correlated with high rain precipitations and the temperatures which ranges from 23 to 30 °C. Similarly, *C. binotalis* were significantly higher at Tengeru experimental site in 2019 season and lower in 2020 season compared with Boro experimental site.

Moreover, *H. undalis* were significantly higher in 2019 season when compared with 2020 season. Those population abundance variations of *C. binotalis* and *H. undalis* larvae mighty have been contributed by differences in weather conditions between the two experimental sites and the two wet seasons. There was high rain precipitations and maximum temperatures ranged from 15 °C to 28 °C in 2020 wet season which could have influenced positively the population of *C. binotalis* larvae. Those observations are in line with the results of Patait *et al.* (2008) and Usui *et al*. (1987) who reported that the population of *C. binotalis* larvae was positively and strongly influenced by maximum temperatures and rain precipitations. Therefore, the variations of weather conditions, like rain precipitations and temperatures could have affected the population of *C. binotalis* larvae. Also, the low rain precipitations of the experimental site could have contributed to the proliferation of *H. undalis*. Those results concur with Sivapragasam and Aziz (1999) who reported that warmer temperatures and low rain precipitations of the region favour the proliferation of *H. undalis* larvae. Seasonal variations facilitated by weather condition variations mighty have affected the population abundance of *H. undalis* at the experimental sites. Because, the increase in larval population of *H. undalis*, is favoured by high temperatures and low rainfall precipitations (Yamada, 1981). Thus, higher

temperatures and lower rainfall precipitations in 2019 season could be the reason for higher population of *H. undalis* compared with 2020 season in which the temperatures and rainfall precipitations were relatively high. In general, it was clear from this study that the population abundance of insect pests infesting *B. oleracea* varies from location to location and from season to season due to weather conditions variability, species diversity and distribution of natural enemies. Moreover, those results concur with Ayalew (2006) who reported that the importance of a particular insect pest varies from location to location due to variations in weather conditions (rainfall and temperatures).

4.2.3 Insecticidal actions of aqueous extracts against cabbage (*B. oleracea*) insect pests

Moreover, the study revealed that the distribution of insect pests on the 1st week before application of the pesticidal plant extracts and the chlorpyrifos was low and was not significantly different and the infestation was less intense because the cabbage (*B. oleracea*) crops were still young. In the negative control (water and water plus soap) plots the population abundance of insect pests persisted from week 1 up to week 6 and the infestation of the *B. oleracea* crops increased progressively week after week. After application of the treatments, it was clearly observed that, the aqueous extracts from *T. vogelii, C. dichogamus* and *S. aromaticum* significantly ($P \leq 0.05$) lowered the population abundance of *H. undalis, C. binotalis, T. ni* and *P. xylostella* larvae and *B. brassicae* and *M. persicae* in *B. oleracea* crops and also, reduced the damage of the crops while in the negative control plots, the infestation and damage increased progressively. It was found that the extracts were as effective as synthetic pesticide (Chlorpyrifos) for controlling *H. undalis, C. binotalis, T. ni* and *P. xylostella* larvae in the *B. oleracea* crops in the field.

The efficacy of the treatments of the aqueous extracts of *C. dichogamus, T. vogelii,* and *S. aromaticum* against those insect pests at higher concentrations (5% concentration of aqueous extracts from mixed plants and 10% concentration from individual plants) was found to be significantly as effective as the chlorpyrifos used in this study as a positive control in managing the insect pests on the cabbage crop in the field. Poor control of these insect pests was observed in the negative control (water and water plus soap) plots and the population abundance of them continued to grow from week one up to week six during the experiments in both seasons. Moreover, the infestations of cabbage (*B. oleracea*) crops in negative controls increased gradually at both experimental sites in 2019 and 2020 wet seasons till harvesting. After applying the treatments, it was found that, the aqueous extracts from *C. dichogamus, T. vogelii*

and *S. aromaticum* pesticidal plants reduced significantly ($P \leq 0.05$) the population of *H. undalis, C. binotalis, T. ni* and *P. xylostella* larvae and *B. brassicae* and *M. persicae* and the infestations on cabbage crops in the field. Particularly, the 5% concentrations of the aqueous extracts from the mixed plants were as effective as synthetic pesticide (Chlorpyrifos) for reducing the population abundance of *H. undalis, C. binotalis, T. ni* and *P. xylostella* larvae and *B. brassicae* and *M. persicae* on the cabbage crops in the field. Notwithstanding, the effectiveness of the synthetic pesticide is contributed by the presence of the active ingredients in it which are environmentally stable. Nevertheless, the effectiveness of aqueous extracts from *C. dichogamus, T. vogelii,* and *S. aromaticum* used for controlling these insect pests in the field could be attributed by the presence of different chemical compounds in each plant with their potency properties like antifeedant, toxicity and repellence against the insect pests studied in this study.

Moreover, it was observed that, among the aqueous extracts used, the 5% concentration of the mixed plants was significantly effective for controlling the *H. undalis, C. binotalis, T. ni* and *P. xylostella* larvae and aphids (*B. brassicae* and *M. persicae*) in the field. The efficacy of the 5% concentration of the aqueous extracts from the mixed plants might be attributed by the synergistic effects of the active chemical compounds present in the extracts of the pesticidal plants used in this study. These results of synergistic effects of the aqueous extracts agree with Tak and Isman (2017b) who reported that the mixture of chemicals in plants have synergistic effects. Synergistic effect occurs when the mixture of two or more chemical compounds interacts and produce combined effects on the biological system which is greater than the algebraic sum of the effects of those chemical compounds when they act individually. Usually, plants produce secondary metabolites for defense either as a distress signal to lure predators, or to directly deter or repel herbivores (Tak & Isman, 2017a). For instance, Tak *et al*. (2016) revealed that, binary mixture of 1,8-cineole and camphor extracted from *Rosmarinus officinali* exhibited enhanced insecticidal activity with a synergy ratio of 1.72 against *T. ni* larvae.

The 10% concentration of the plant extracts was more effective in reduction of the abundance of *H. undalis, C. binotalis, T. ni* and *P. xylostella* larvae and *B. brassicae* and *M. persicae* and the damage of *B. oleracea* compared with 1% and 5% of the aqueous extracts. Similarly, the 10% concentration of *T. vogelii, C. dichogamus,* and *S. aromaticum* extracts was more effective in reduction of the *H. undalis, C. binotalis, T. ni* and *P. xylostella* larvae and *B. brassicae* and *M. persicae* and the damage of *B. oleracea* compared with 1% and 5%. Statistically, it was the

second, after the 5% concentration of the extracts from the mixed plants and the chlorpyrifos in reduction of the population abundance of these insect pests in the fields from week 1 to week 6. The efficacy of the aqueous extracts from the pesticidal plants concur with the results of other researchers (Belmain *et al.*, 2013; Belmain & Stevenson, 2001; Do Ngoc Dai *et al.*, 2015; Grzywacz *et al.*, 2014; Ileke & Oni, 2011; Kamanula *et al.*, 2010; Mkenda *et al.*, 2014; Mwanauta *et al.*, 2014). Kamanula *et al*. (2010) and Grzywacz *et al*. (2014) reported that the control of insect pests using pesticidal plant extracts could be contributed by the presence of insecticidal bioactive compounds in those pesticidal plants. Belmain *et al*. (2012) reported that, in *T. vogelii*, there are two separate chemotypes, the chemotype I and chemotype II. Chemotype I contain rotenoids. The rotenoids contain deguelin, rotenone, sarcolobine, α-toxicarol and tephrosin. Those chemicals in *T. vogelii* could be responsible for *H. undalis, C. binotalis, T. ni* and *P. xylostella* larvae and *B. brassicae* and *M. persicae* control efficacy. Among these chemicals, rotenone is the most toxic compound to insect pests. Chemotype II contains obovatin-5-methyl ether which is not rotenoids and is none poison. Belmain *et al*. (2012) reported that rotenone (Fig. 43) works by hindering the electron transport chain in mitochondria. Khater (2012) indicated that, rotenone is a contact and stomach poison which limits the electron transport chain in the Mitochondria. It inhibits the transfer of electrons from iron-sulfur centers in complex I to ubiquinone. Therefore, rotenone (Fig. 43) interferes cellular energy production (ATP) in the cell of the insect pests. In that phenomenon, complex I is unable to pass through its electron to complex Q, creating a back-up of electrons within the mitochondrial matrix. During this limiting process, cellular oxygen is reduced to the radical which is a reactive species. This reactive species can damage DNA and other components of the mitochondria.

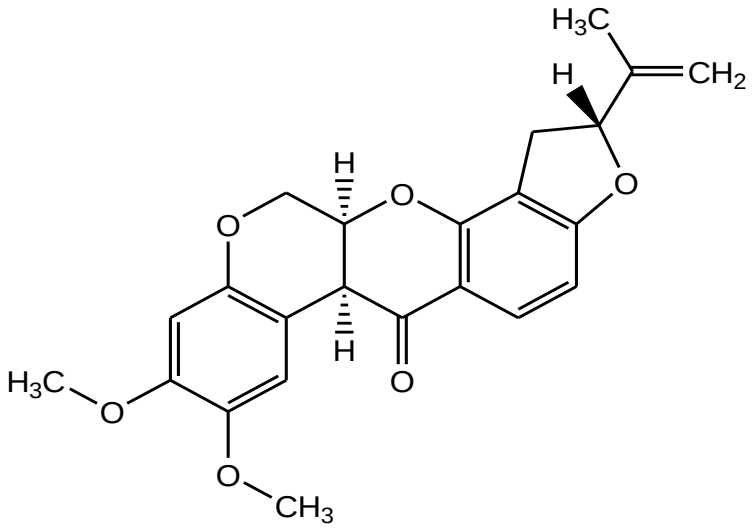


Figure 43: Chemical structure of rotenone $C_{23}H_{22}O_6$. (Mpumi *et al.*, 2016)

Apart from that, this study revealed that, all concentrations of the extracts used from *Syzygium aromaticum* significantly ($P \leq 0.05$) reduced the population abundance of *H. undalis*, *C. binotalis*, *T. ni* and *P. xylostella* larvae and *B. brassicae* and *M. persicae* when compared with negative controls (water and water plus soap) at the field. The GC-MS analysis results from this study shows the presence of different phytochemical compounds from DCM-MeOH extracts of *S. aromaticum*. Among those botanical compounds obtained, the major compounds identified were; - Eugenol (52.66%); Eugenol acetate (20.46%); (β-caryophyllene) (7.52%); Phenol, 2-methoxy-3-(2-propenyl)- (4.17%); α-Caryophyllene (1.36%) and Caryophyllene epoxide (1.04%) (Table 35). These findings agree with Kamatou *et al.* (2012); Araujo *et al.* (2016) and Tian *et al*. (2015).

Kamatou *et al.* (2012) and Araujo *et al*. (2016) reported the presence of eugenol, β-caryophyllene, α-humulene and eugenol acetate and eugenol being the most active compound responsible for imparting the taste of the *S. aromaticum* whereby the concentrations of each chemical compound differ from this study to others. Although, the concentrations of the chemical compounds from *S. aromaticum* essential oils differ significantly from one country to another, but there is no doubt that eugenol is the major constituent in all places (Fu *et al.*, 2007). The difference in concentrations of the chemical compounds in *S. aromaticum* may be caused by the variation of vegetative state, growing season and the places of origin (Fu *et al.*, 2007; Samarasekera *et al.*, 2008; Srivastava *et al.*, 2005). Moreover, the concentrations of the chemical compounds in *S. aromaticum* can vary depending on plant part and the condition in which the extraction is performed, such as sun-dried leaves, peduncle and dried flower buds (Oliveira *et al.*, 2009).

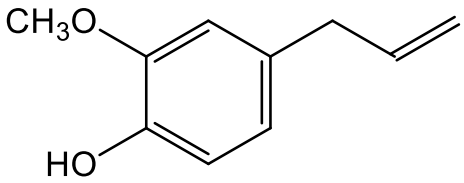


Figure 44: Chemical structure of Eugenol $C_{10}H_{12}O_2$ (Kamatou *et al.*, 2012)

Therefore, eugenol (Fig. 44) and other chemicals compounds reported in this study together with the reports of other researchers from *S. aromaticum* mighty be responsible for the larvicidal and insecticidal actions against *H. undalis*, *C. binotalis*, *T. ni* and *P. xylostella* larvae and *B. brassicae* and *M. persicae*. For instance, Tian *et al.* (2015) reported that, *S. aromaticum* has a range of pharmacological activities which includes antimicrobial, anti-inflammatory, analgesic, anti-oxidant and anticancer activities, amongst others and therefore can be used in agriculture to protect crops and foods from micro-organisms during storage. In addition, *S aromaticum* has an effect on insect pests as a pesticide and fumigant (Kamatou *et al.*, 2012; Tian *et al.*, 2015).

Also, this study reported that, all concentrations (1%, 5% and 10%) of the extracts used from *Croton dichogamus* significantly ($P \leq 0.05$) reduced the population abundance of *H. undalis*, *C. dichogamus*, *T. ni* and *P. xylostella* larvae and *B. brassicae* and *M. persicae* when compared with negative controls. Likewise, the GC-MS analysis results from this study report the presence of different phytochemical compounds from DCM-MeOH extracts of *Croton dichogamus*. Among those phytochemicals obtained from the extracts of *C. dichogamus*, they include compounds; - 4,6-Bis (4-chloro-3-(trifluoromethyl) phenoxy)-2-pyrimidinol (25.08%); Cholestan-6-en-3-ol (18.63%); 1-Heptadecene (7.34%); 1-Tetradecene (6.30%); Distannoxane, hexabutyl- (5.93%); 2,4-Di-tert-butylphenol (5.26%); (Z)-9-Tricosene and Trichloroacetic acid, tetradecyl ester (5.23%) were present in larger concentrations. Most of the organic compounds obtained here in this study were alkaloids, phenolics and terpenoids. These results agree with Aldhaher *et al*. (2017) and Silva *et al*. (2018) who reported that, the *Croton species* possesses alkaloids, phenolics, terpenoids including monoterpenes, sesquiterpenes and diterpenes in all plant parts. Those compounds could be responsible for the insecticidal, repellents and deterrent effects to insect pests observed in this study. In Africa, America and Asia *Croton species* are commonly used in folk medicines in the treatment of cancer, constipation, diabetes, digestive problems, dysentery, external wounds, fever, hypercholesterolemia, hypertension, inflammation, intestinal worms, malaria, pain, ulcers and weight-loss (Silva *et al*., 2018) due to the present of those chemicals. Moreover, these results

agree with Silva *et al*. (2018) who reported that *Croton* species contain chemicals which are responsible for insecticidal activity.

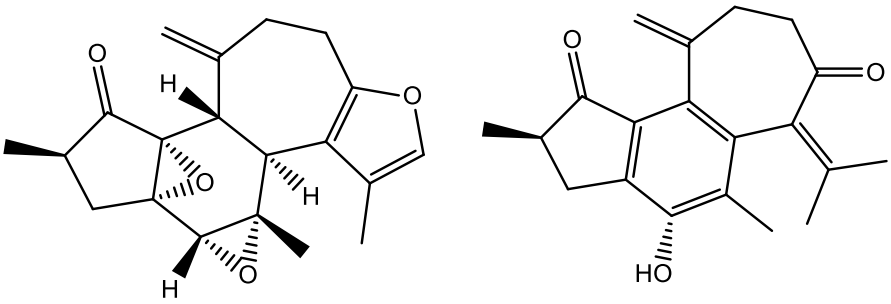


Figure 45: Chemical compounds in the genus *Croton species.* (Xu *et al.*, 2018)

4.2.4 Interactions of weather conditions, seasons and treatments for lowering of *B. oleracea* crop insect pests

The interactions of the site's weather condition, seasons and treatments was also observed in some weeks during the application of pesticidal plant extracts and pesticide. The interaction of weather conditions with the treatments significantly ($p \leq 0.05$) enhanced the lowering of the population of *H. undalis, C. dichogamus, T. ni* and *P. xylostella* larvae and *B. brassicae* and *M. persicae* compared with the negative controls (water and water plus soap). Moreover, the interactions of experimental sites' weather conditions and seasons was also seen in some weeks during the experiments. It was seen that, the population abundance of insect pests in this study was significantly affected by weather conditions and seasons compared with the negative controls in the plots. Lastly there was interaction of weather conditions of the experimental sites, treatments and seasons which significantly affected the population abundance of the insect pests compared with the negative controls.

The proliferation and high density of natural enemies and changes of weather conditions mighty have contributed to the higher insect pests at one season and experimental site compared with the other season and the other experimental site. For instance, in 2019 season, there was high rain precipitations at Boro experimental site compared with Tengeru experimental site which mighty have contributed to the higher population of *P. xylostella* at Boro compared to Tengeru experimental site. This indicates that weather conditions, vegetation density near the study plots and predation pressure, mighty have enhanced the growth and development of a particular insect pests and the culturing of the natural enemies which together with treatments and seasons suppress the growth and proliferation of insect pests. Therefore,

cabbage smallholder farmers should be advised to observe the weather conditions when managing the cabbage insect pests in different locations using the botanicals where synthetic pesticides are not affordable.

CHAPTER FIVE

CONCLUSION AND RECOMMENDATIONS

5.1 Conclusion

This study assessed the larvicidal action of the extracts from *S. aromaticum, T. vogelii* and *C. dichogamus* dichloromethane-methanol (1:1) extracts against *P. xylostella* and *C. binotalis* larvae and insecticidal efficacy of aqueous extracts of 1%, 5% and 10% concentrations of *S. aromaticum, T. vogelii* and *C. dichogamus* and the mixture of these plants (2.5% and 5%) for the control of cabbage (*B. oleracea*) insect pests in the field in general. The extracts from these plants used in this study exhibited the larvicidal activities against *P. xylostella* and *C. binotalis* larvae. Due to larvicidal activities of these plants against *P. xylostella* and *C. binotalis* larvae, the extracts from *S. aromaticum, T. vogelii* and *C. dichogamus* exhibited the larvicidal actions hence can be used to suppress the growth of the larvae into other stages. The extracts from these plants are therefore promising alternative for use in the control of *P. xylostella* and *C. binotalis* larvae in place of synthetic pesticides.

Similarly, the mixtures of the aqueous plant extracts at 5% concentration and the individual plants at higher concentrations (10%) reduced the population abundance of *C. binotalis, T. ni, P. xylostella, H. undalis* larvae and aphids (*M. persicae* and *B. brassicae*) and therefore significantly lowered the damage of cabbage (*B. oleracea*) in the field and increased the cabbage heads. The decrease in population abundance of *C. binotalis, T. ni* and *P. xylostella, H. undalis* larvae and aphids (*M. persicae* and *B. brassicae*) and decrease in damage (%) of cabbage (*B. oleracea*) on the treated plots indicates the efficacies of the aqueous plant extracts from *S. aromaticum, T. vogelii* and *C. dichogamus* against *C. binotalis, T. ni* and *P. xylostella, H. undalis* larvae and aphids (*M. persicae* and *B. brassicae*) in the field. Therefore, the individual plants at higher concentrations (10%) and the aqueous extracts of the mixture of these plants at 5% concentrations of *S. aromaticum, T. vogelii* and *C. dichogamus* can be used to control the common cabbage insect pests in the field in place of synthetic pesticides. This study revealed the potentialities of mixing of the plant materials during extractions which enhanced the insecticidal activity and broadened the spectrum of efficacies in reduction of the damage of cabbage crops (*B. oleracea*) in the fields compared with the negative controls and individual plant concentrations.

The GC-MS analysis of extracts from *S. aromaticum* revelaed the presence of eugenol; eugenol acetate; beta-caryophyllene; phenol, 2-methoxy-3-(2-propenyl) and alpha-caryophyllene in higher concentrations and the analysis of extracts from *C. dichogamus* discovered the presence of 4,6-Bis (4-chloro-3-(trifluoromethyl) phenoxy)-2-pyrimidinol; Cholestan-6-en-3-ol; 1-Heptadecene and 1-Tetradecene in higher concentrations. These chemical compounds could be responsible for insecticidal efficacy and larvicidal actions against cabbage insect pests in this study. Therefore, the present study recommends the use of *T. vogelii*, *S. aromaticum* and *C. dichogamus* extracts as cheap and eco-friendly insecticide for the control of *C. binotalis, T. ni, P. xylostella, H. undalis* larvae and aphids (*M. persicae* and *B. brassicae*) in cabbage crops at the field at higher concentrations and development of insecticides.

5.2 Recommendations

The role of botanicals in insect pest management and crop protection is great, but is less explored due to limited scientific information about their effectiveness, toxicity, commercialization and costs of instruments for identifications of the phytochemicals in pesticidal plants.

(i) Thus, detailed study is required to investigate the toxicity of botanicals from *S. aromaticum, T. vogelii* and *C. dichogamus* to ensure the safety to human health which can facilitate the commercialization of chemicals from these plants for crop protection.

(ii) Apart from that, the investigation of the half-lives of the bioactive compounds which are present in these pesticidal plants is required to be sure with their persistence in the environment and in the cabbage crops when used for the control of insect pests.

(iii) Moreover, the study of the degradation rate of the bioactive compounds present in these botanicals when applied to the vegetables' garden is required to be aware of the persistence of them while in the leaf surfaces, in the soil and in water.

(iv) The results of this study suggest more study into a bioassay, fractionation, isolations and purifications of compounds from the crude extracts of the leaves of *C. dichogamus* and the buds of *S. aromaticum.*

(v) Simple, costless and manageable tools should be designed to identify the chemicals which are present in these pesticidal plants to increase the potentialities and use for the protection of the cabbage crops from insect pests damage and infestations

(vi) Also, the environmental regulatory agents and public health laws should create opportunities for the use of botanicals from these plants which are environmentally benign, safe to humans and ecosystems for the control of cabbage insect pests in the fields in place of synthetic pesticides.

REFERENCES

Abbott, W. S. (1925). A Method of computing the effectiveness of an insecticide. *Journal of Economic Entomology*, *18*(2), 265–267. https://doi.org/10.1093/jee/18.2.265a

Acheuk, F., & Doumandji-Mitiche, B. (2013). Insecticidal activity of alkaloids extract of *Pergularia tomentosa* (Asclepiadaceae) against fifth instar larvae of *Locusta migratoria* cinerascens (Fabricius 1781)(Orthoptera: Acrididae). *International Journal of Science and Advanced Technology*, *3*(6), 8-13.

Adebisi, O., Dolma, S. K., Verma, P. K., Singh, B., & Reddy, S. E. (2019). Volatile, non-volatile composition and insecticidal activity of *Eupatorium adenophorum* Spreng against diamondback moth, *Plutella xylostella* (L.), and aphid, *Aphis craccivora* Koch. *Toxin Reviews*, *38*(2), 143-150. https://doi.org/10.1080/15569543.2018.1434795

Afun, J. V. K., Jackai, L. E. N., & Hodgson, C. J. (1991). Calendar and Monitored insecticide application for the control of cowpeas pests. *Crop Protection*, *10*(5), 363 – 370. https://doi.org/10.1016/S0261-2194(06)80025-6.

Aharoni, A., Jongsma, M. A., & Bouwmeester, H. J. (2005). Volatile science? Metabolic engineering of terpenoids in plants. *Trends in Plant Science*, *10*(12), 594-602. https://doi.org/10.1016/j.tplants.2005.10.005

Ahmad, M., & Akhtar, S. (2013). Development of insecticide resistance in field populations of *Brevicoryne brassicae* (Hemiptera: Aphididae) in Pakistan. *Journal of Economic Entomology*, *106*(2), 954-958. http://dx.doi.org/10.1603/EC12233

Ahouangninou, C., Fayomi, B. E., & Martin, T. (2011). Assessing health and environmental risks as regards pesticide practices of vegetable growers in the rural city of Tori-Bossito in southern Benin. *Cahiers Agricultures*, *20*(3), 216-222. http://dx.doi.org/10.4236/jep.2012.33030

Akhtar, Y., Isman, M. B., Paduraru, P. M., Nagabandi, S., Nair, R., & Plettner, E. (2007). Screening of dialkoxybenzenes and disubstituted cyclopentene derivatives against the cabbage looper, *Trichoplusia ni*, for the discovery of new feeding and oviposition deterrents. *Journal of Agricultural and Food Chemistry*, *55*(25), 10323-10330. https://doi.org/10.1021/jf071636d

Alamgir, A. (2018). Secondary metabolites: Secondary metabolic products consisting of C and H; C, H, and O; N, S, and P elements; and O/N heterocycles *Therapeutic Use of Medicinal Plants and their Extracts*: Springer. https://doi.org/10.1007/978-3-319-92387-1_3

Alavanja, M. C. (2009). Introduction: Pesticides Use and Exposure, Extensive Worldwide. *Reviews on Environmental Health*, *24*(4), 303-310. http://dx.doi.org/10.1515/REVEH.2009.24.4.303.

Aldhaher, A., Langat, M., Ndunda, B., Chirchir, D., Midiwo, J. O., Njue, A., Schwikkard, S., Carew, M., & Mulholland, D. (2017). Diterpenoids from the roots of *Croton dichogamus* Pax. *Phytochemistry*, *144*, 1-8. https://doi.org/10.1016/j.phytochem.2017.08.014

Alhazmi, H. A., Khalid, A., Sultana, S., Abdelwahab, S. I., Ahsan, W., Oraiby, M. E., & Bratty, M. A. (2019). Determination of phytocomponents of twenty-one varieties of smokeless tobacco using gas chromatography-mass spectroscopy (GC-MS). *South African Journal of Chemistry*, *72*, 47-54. http://dx.doi.org/10.17159/0379-4350/2019/ v72a7.

Ambrósio, S. R., Oki, Y., Heleno, V. C. G., Chaves, J. S., Nascimento, P. G. B. D., Lichston, J. E., Constantino, M. G., Varanda, E. M., & Da Costa, F. B. (2008). Constituents of glandular trichomes of *Tithonia diversifolia*: Relationships to herbivory and antifeedant activity. *Phytochemistry*, *69*(10), 2052-2060. http://dx.doi.org/10.1016/j.phytochem.2008.03.019.

Amoabeng, B. W., Gurr, G. M., Gitau, C. W., Nicol, H. I., Munyakazi, L., & Stevenson, P. C. (2013). Tri-trophic insecticidal effects of African plants against cabbage pests. *PLoS One*, *8*(10), 1-10. https://doi.org/10.1371/journal.pone.0078651

Amoabeng, B. W., Gurr, G. M., Gitau, C. W., & Stevenson, P. C. (2014). Cost: Benefit analysis of botanical insecticide use in cabbage: Implications for smallholder farmers in developing countries. *Crop Protection*, *57*, 71-76. http://dx.doi.org/10.1016/j.cropro.2013.11.019.

Anstead, J., Mallet, J., & Denholm, I. (2007). Temporal and spatial incidence of alleles conferring knockdown resistance to pyrethroids in the peach–potato aphid, *Myzus persicae* (Hemiptera: Aphididae), and their association with other insecticide resistance mechanisms. *Bulletin of Entomological Research, 97*(3), 243-252. http://dx.doi.org/10.1017/S0007485307004889

Arannilewa, S., Ekrakene, T., & Akinneye, J. (2006). Laboratory evaluation of four medicinal plants as protectants against the maize weevil, *Sitophilus zeamais* (Mots). *African Journal of Biotechnology*, *5*(21), 2032-2036.

Araujo, A. F. D. O., Ribeiro-Paes, J. T., Deus, J. T. D., Cavalcanti, S. C. D. H., Nunes, R. D. S., Alves, P. B., & Macoris, M. D. L. D. G. (2016). Larvicidal activity of *Syzygium aromaticum* (L.) Merr and *Citrus sinensis* (L.) Osbeck essential oils and their antagonistic effects with temephos in resistant populations of *Aedes aegypti*. *Memórias do Instituto Oswaldo Cruz, 111*(7), 443-449. http://dx.doi.org/10.1590/0074-02760160075

Asante, K. A., & Ntow, W. J. (2009). Status of environmental contamination in Ghana, the perspective of a research scientist. *Interdisciplinary Studies on Environmental Chemistry*, *2*, 253-260.

Ashamo, M. (2010). *Management of the yam moth, Dasyses rugosella, a pest of stored yam tubers using plant products.* Paper presented at the 10th International Working Conference on Store Product Protection, Federal University of Technology, Akure, Nigeria. https://doi.org/10.5073/jka.2010.425.071

Assembly, U. G. (2015). *Transforming our world: The 2030 agenda for sustainable development*. http://www.un.org/ga/search/view_doc.asp?symbol=A.*RES/70/1& Lang = E*.

Atshan, K. A., & Rao, Y. T. (2017). Effect of seed extracts as insecticides against lepidopteran pests on cauliflower. *Asian Journal of Agricultural Research*, *11*(3), 78-85. http: //dx.doi.org /10.3923 /ajar.2017.78.85

Ayalew, G. (2006). Comparison of yield loss on cabbage from Diamondback moth, *Plutella xylostella* L. (Lepidoptera: Plutellidae) using two insecticides. *Crop Protection*, *25*(9), 915-919. http://dx.doi.org/10.1016/j.cropro.2005.12.001

Badenes-Perez, F. R., Nault, B. A., & Shelton, A. M. (2006). Dynamics of diamondback moth oviposition in the presence of a highly preferred non-suitable host. *Entomologia Experimentalis et Applicata*, *120*(1), 23-31. https://doi.org/10.1111/j.1570-7458.2006.00416.x

Baidoo, P., & Adam, J. (2012). The effects of extracts of *Lantana camara* (L.) and *Azadirachta indica* (A. Juss) on the population dynamics of *Plutella xylostella*, *Brevicoryne brassicae* and *Hellula undalis* on cabbage. *Sustainable Agriculture Research*, *1*(2), 229-234. https://doi.org/10.5539/sar.v1n2p229

Baidoo, P., & Angbanyere, M. (2014). The effect of pollinators and pollination on fruit set and fruit yield of okra (*Abelmoschus esculentus* (L.) Moench) in the forest region of Ghana. *American Journal of Experimental Agriculture, 4*(9), 985-995. https://doi.org/10.9734/AJEA/2014/7472

Baidoo, P., & Mochiah, M. (2016). Comparing the effectiveness of garlic (*Allium sativum* L.) and hot pepper (*Capsicum frutescens* L.) in the management of the major pests of cabbage *Brassica oleracea* (L.). *Sustainable Agriculture Research*, *5*(2), 83-91. https://doi.org/ 10.22004/ag.econ.234995.

Ballard, C. R., & Junior, M. R. M. (2019). Health benefits of flavonoids *Bioactive compounds* (pp. 185-201): https://doi.org/10.1016/B978-0-12-814774-0.00010-4

Bami, H. (1997). Pesticide use in India-Ten questions. *Chemical Weekly-Bombay*, *42*(4), 137-146. https://doi.org/10.4236/gep.2021.96009

Basak, H., Biswas, B., Azad, M., Arifuzzaman, M., & Sharmeen, F. (2012). Micropropagation of mustard (*Brassica spp*.) from leaf explants. *Thai Journal of Agricultural Science*, *45*(2), 75-81.

Begna, F., & Damtew, T. (2015). Evaluation of four botanical insecticides against Diamondback Moth, *Plutella Xylostella* L. (Lepidoptera: Plutellidae) on head cabbage in the central rift valley of Ethiopia. *Sky Journal of Agricultural Research*, *4*(5), 97-105.

Belmain, S., Haggar, J., Holt, J., & Stevenson, P. (2013). *Managing legume pests in sub-Saharan Africa: Challenges and prospects for improving food security and nutrition through agro-ecological intensification*. Chatham Maritime (United Kingdom): Natural Resources Institute, University of Greenwich. http://projects.nri.org/adappt /docs/ Managing_legume _pests_in_subSaharan_Africa.pdf

Belmain, S., & Stevenson, P. (2001). Ethnobotanicals in Ghana: reviving and modernising age-old farmer practice. *Pesticide Outlook*, *12*(6), 233-238. https://doi.org/10.1039/B110542F

Belmain, S. R., Amoah, B. A., Nyirenda, S. P., Kamanula, J. F., & Stevenson, P. C. (2012). Highly variable insect control efficacy of *Tephrosia vogelii* chemotypes. *Journal of Agricultural and Food Chemistry*, *60*(40), 10055-10063. http://dx.doi.org/10.1021/jf3032217.

Bhatt, P., Thodsare, N., & Srivastava, R. (2014). Toxicity of some bioactive medicinal plant extracts to Asian army worm, *Spodoptera litura*. *Journal of Applied and Natural Science*, *6*(1), 139-143.

Blackman, R. L. & Eastop, V. F. (2000). *Aphids on the world's crops: An identification and information guide*: John Wiley & Sons Ltd, Hoboken, NJ, USA. https://www.Wiley.com/en-us/Aphids+on+the+World%27s+Crops%3A+An+Identification+and+Information+Guide%2C+2nd+Edition-p-9780471851912

Bolor, V. K., Boadi, N. O., Borquaye, L. S., & Afful, S. (2018). Human risk assessment of organochlorine pesticide residues in vegetables from Kumasi, Ghana. *Journal of Chemistry*, *2018*, 1-11. https://doi.org/10.1155/2018/3269065

Boncan, D. A. T., Tsang, S. S., Li, C., Lee, I. H., Lam, H. M., Chan, T. F., & Hui, J. H. (2020). Terpenes and terpenoids in plants: Interactions with environment and insects. *International Journal of Molecular Sciences*, *21*(19), 1-19.

Buchweitz, J. P., Bokhart, M., Johnson, M., & Lehner, A. (2013). Determination of methomyl in the stomach contents of baited wildlife by gas chromatography–mass spectrometry. *Journal of Veterinary Diagnostic Investigation*, *25*(6), 744-749. https://doi.org/10.1177/1040638713505564

Butnariu, M., & Sarac, I. (2018). Essential oils from plants. *Journal of Biotechnology and Biomedical Science*, *1*(4), 35-43. https://doi.org/10.14302/issn.2576-6694.jbbs-18-2489

Capinera, J. (2001). *Handbook of vegetable pests*: Academic Press, Cambridge, MA, USA. https://www.elsevier.com/books/handbook-of-vegetable-pests/capinera/978-0-12-158861-8

Carter, C., & Sorensen, K. (2013). *Insect and related pests of vegetables. In Cabbage and turnip aphid. Center for Integrated Pest Management*. North Carolina State University, Raleigh, NC, USA. http://entomology.ifas.ufl.edu/creatures

Chapman, R., Bernays, E., & Simpson, S. (1981). Attraction and repulsion of the aphid, *Cavariella aegopodii*, by plant odors. *Journal of Chemical Ecology*, *7*(5), 881-888. http://dx.doi.org/10.1007/BF00992385.

Charleston, D. S., Gols, R., Hordijk, K. A., Kfir, R., Vet, L. E., & Dicke, M. (2006). Impact of botanical pesticides derived from *Melia azedarach* and *Azadirachta indica* plants on the emission of volatiles that attract parasitoids of the diamondback moth to cabbage plants. *Journal of Chemical Ecology*, *39*(1), 105-114. http://dx.doi.org/10.1007/s10886-005-9004-9

Chaubey, M. K. (2012). Responses of *Tribolium castaneum* (Coleoptera: Tenebrionidae) and *Sitophilus oryzae* (Coleoptera: Curculionidae) against essential oils and pure compounds. *Herba Polonica*, *58*(3), 33-45.

Chaubey, M. K. (2019). Essential oils as green pesticides of stored grain insects. *European Journal of Biological Research*, *9*(4), 202-244. http://dx.doi.org/10.5281/zenodo.3528366

Chen, Y., Yan, T., Gao, C., Cao, W., & Huang, R. (2014). Natural products from the genus Tephrosia. *Molecules, 19*(2), 1432-1458. http://dx.doi.org/10.3390/molecules19021432

Chikukura, L., Mvumi, B., Chikonzo, R., & Chenzara, C. (2011). *Evaluation of selected indigenous pesticidal plant powders against stored maize and cowpeas insect pests. Paper presented at the African Crop Science Conference Proceedings.* https://www.researchgate.net/publication/283047411_Evaluation_of_selected_indigenous_pesticidal_plant_powders_against_stored_maize_and_cowpeas_insect_pests

Cho, K. S., Lim, Y. R., Lee, K., Lee, J., Lee, J. H., & Lee, I. S. (2017). Terpenes from forests and human health. *Toxicological Research, 33*(2), 97-106. http://dx.doi.org/10.5487/TR.2017.33.2.097

Chomchalow, N. (2003). Protection of stored products with special reference to Thailand. *Assumption University Journal of Technology, 7*(1), 34-47. http://dx.doi.org/10.1.1.550.3377

Cushnie, T. T., & Lamb, A. J. (2005). Antimicrobial activity of flavonoids. *International Journal of Antimicrobial Agents, 26*(5), 343-356. http://dx.doi.org/10.2174/092986732166614091611344 3

Da Silva, R. C. S., Milet-Pinheiro, P., Bezerra Da Silva, P. C., Da Silva, A. G., Da Silva, M. V., Navarro, D. M. D. A., & Da Silva, N. H. (2015). (E)-caryophyllene and α-humulene: *Aedes aegypti* oviposition deterrents elucidated by gas chromatography-electrophysiological assay of *Commiphora leptophloeos* leaf oil. *PLoS One, 10*(12), 1-14. https://doi.org/10.1371/journal.pone.0144586

Dalvie, M. A., Cairncross, E., Solomon, A., & London, L. (2003). Contamination of rural surface and ground water by endosulfan in farming areas of the Western Cape, South Africa. *Environmental Health, 2*(1), 1-15. https://doi.org/10.1186/1476-069X-2-1

De Bon, H., Huat, J., Parrot, L., Sinzogan, A., Martin, T., Malezieux, E., & Vayssieres, J. F. (2014). Pesticide risks from fruit and vegetable pest management by small farmers in sub-Saharan Africa. A review. *Agronomy for Sustainable Development, 34*(4), 723-736. https://doi.org/10.1007/s13593-014-0216-7

De Cássia Seffrin, R., Shikano, I., Akhtar, Y., & Isman, M. B. (2010). Effects of crude seed extracts of *Annona atemoya* and *Annona squamosa* L. against the cabbage looper, *Trichoplusia ni* in the laboratory and greenhouse. *Crop Protection*, *29*(1), 20-24. https://doi.org/10.1016/j.cropro.2009.09.003

De Little, S. C., & Umina, P. A. (2017). Susceptibility of Australian *Myzus persicae* (Hemiptera: Aphididae) to three recently registered insecticides: Spirotetramat, cyantraniliprole, and sulfoxaflor. *Journal of Economic Entomology*, *110*(4), 1764-1769. https://doi.org/10.1093/jee/tox132

De Toledo, L. G., Ramos, M. A. D. S., Spósito, L., Castilho, E. M., Pavan, F. R., Lopes, É. D. O., Zocolo, G. J., Silva, F. A. N., Soares, T. H., & Dos Santos, A. G. (2016). Essential oil of *Cymbopogon nardus* (L.) Rendle: A strategy to combat fungal infections caused by *Candida species*. *International Journal of Molecular Sciences*, *17*(8), 1-16. https://doi.org/10.3390/ijms17081252

Delfel, N. E., Tallent, W. H., Carlson, D. G., & Wolff, I. A. (1970). Distribution of rotenone and deguelin in *Tephrosia vogelii* and separation of rotenoid-rich fractions. *Journal of Agricultural and Food Chemistry*, *18*(3), 385-390. https://doi.org/10.1021/jf60169a053

Do Ngoc Dai, T. D. T., Ogunmoye, A., Eresanya, O. I., & Ogunwande, I. A. (2015). Chemical constituents of essential oils from the leaves of *Tithonia diversifolia*, *Houttuynia cordata* and *Asarum glabrum* grown in Vietnam. *American Journal of Essential Oils and Natural Products*, *2*(4), 17-21.

Dougnon, T., Farougou, S., Kpodékon, T., Hounmanou, G., & Hounnnonkpè, D. (2014). Comparative study of the effect of *Tephrosia vogelii*'s leaves ethanolic extract and Alfapor on *Amblyomma variegatum* in Borgou cattle. *International Journal of Biosciences (IJB)*, *5*(5), 1-7. https://doi.org/10.12692/ijb/ 5.5.1-7

Dubey, S. K., & Gupta, A. K. (2014). Callus induction and shoot proliferation from seedling explants of different mustard genotypes. *International Journal of Current Microbiology and Applied Science*, *3*(7), 858-864.

Dzenda, T., Ayo, J., Adelaiye, A., & Adaudi, A. (2007). Ethno-medical and veterinary uses of *Tephrosia vogelii* hook. F.: A review. *Nigerian Veterinary Journal*, *28*(3), 24-39. https://doi.org/10.4314/nvj.v28i3.3564

Eapen, S., Abraham, V., Gerdemann, M., & Schieder, O. (1989). Direct somatic embryogenesis, plant regeneration and evaluation of plants obtained from Comparative study of the effect of *Tephrosia vogelii*'s leaves ethanolic extract and Alfapor®(Alpha-cypermethrin) on *Amblyomma variegatum* in Borgou cattle mesophyll protoplasts of *Brassica juncea*. *Annals of Botany*, *63*(3), 369-372. https://doi.org/10.1093/oxfordjournals.aob.a087753

Ebenebe, A. A., Achari, S. R., Chand, N., Krishna, A. A., & Baleisuva, S. (2011). The cabbage webworm (*Hellula undalis*) on tickweed (*Cleome viscosa*) in Samoa. *The South Pacific Journal of Natural and Applied Sciences*, *29*(1), 1-6. https://doi.org/10.1071/SP11001

Edwards, O. R., Franzmann, B., Thackray, D., & Micic, S. (2008). Insecticide resistance and implications for future aphid management in Australian grains and pastures: A review. *Australian Journal of Experimental Agriculture*, *48*(12), 1523-1530. https://doi.org/10.1071/EA07426

El-Wakeil, N. E. (2013). Botanical pesticides and their mode of action. *Gesunde Pflanzen*, *65*(4), 125-149. http://dx.doi.org/10.1007/s10343-013-0308-3.

El Asbahani, A., Miladi, K., Badri, W., Sala, M., Addi, E. A., Casabianca, H., El Mousadik, A., Hartmann, D., Jilale, A., & Renaud, F. (2015). Essential oils: from extraction to encapsulation. *International Journal of Pharmaceutics*, *483*(1-2), 220-243.

Elwakil, W. M., & Mossler, M. (2016). Florida crop/pest management profile: Cabbage. *University of Florida, Gainesville, FL, USA*, *1256*(4), 1-18. https://doi.org/10.32473/edis-pi042-2013

Emam, A. M., Swelam, E. S., & Megally, N. Y. (2009). Furocoumarin and quinolone alkaloid with larvicidal and antifeedant activities isolated from Ruta chalepensis leaves. *Journal of Natatural Products*, *2*, 10-22.

Fening, K., Amoabeng, B., Adama, I., Mochiah, M., Braimah, H., Owusu-Akyaw, M., Narveh, E., & Ekyem, S. (2013). Sustainable management of two key pests of cabbage, *Brassica oleracea* var. capitata L.(Brassicaceae), using homemade extracts from garlic and hot pepper. *Organic Agriculture*, *3*(4), 163-173. https://doi.org/10.1007/s13165-014-0058-2

Finney, D. J. (1971). *Probit Analysis*. (3rd Edn.), Cambridge University Press, Cambridge, https://doi.org/10.1002/jps.2600600940

Flint, M. L., Dreistadt, S. H., & Clark, J. K. (1998). *Natural enemies handbook: The illustrated guide to biological pest control*, University of California Press, University of California Press, Berkeley, CA, USA. http://ipm.ucanr.edu/IPMPROJECT/ADS/manual_ naturalenemies.html

Fu, Y., Zu, Y., Chen, L., Shi, X., Wang, Z., Sun, S., & Efferth, T. (2007). Antimicrobial activity of clove and rosemary essential oils alone and in combination. *Phytotherapy Research*, *21*(10), 989-994. https://doi.org/10.1002/ptr.2179

Furlong, M. J., Wright, D. J., & Dosdall, L. M. (2013). Diamondback moth ecology and management: Problems, progress, and prospects. *Annual Review of Entomology*, *58*(2013), 517-541. https://doi.org/10.1146/annurev-ento-120811-153605

Garcia, R., Alves, E. S., Santos, M. P., Aquije, G. M., Fernandes, A. A. R., Santos, R. B. D., Ventura, J. A., & Fernandes, P. (2008). Antimicrobial activity and potential use of monoterpenes as tropical fruits preservatives. *Brazilian Journal of Microbiology*, *39*(1), 163-168. https://doi.org/10.1590/S1517-838220080001000032

Geyid, A., Abebe, D., Debella, A., Makonnen, Z., Aberra, F., Teka, F., Kebede, T., Urga, K., Yersaw, K., & Biza, T. (2005). Screening of some medicinal plants of Ethiopia for their anti-microbial properties and chemical profiles. *Journal of Ethnopharmacology*, *97*(3), 421-427. https://doi.org/10.1016/j.jep.2004.08.021

Gibson, A. (2012). *Colewart and the cole crops*. University of California Los Angeles, Los Angeles. https://www.gov.nl.ca/ffa/files/agrifoods-plants-pdf-cole-crops.pdf

Gill, H. K., & Garg, H. (2014). Pesticide: Environmental impacts and management strategies. *Pesticides-toxic Aspects*, *8*(2014), 187-231. https://doi.org/10.5772/57399

Gill, H. K., Garg, H., & Gillett-Kaufman, J. L. (2013). *Cabbage aphid Brevicoryne brassicae Linnaeus* (Insecta: *Hemiptera*: *Aphididae*). IFAS Extension University of Florida: Gainesville, FL, USA. https://edis.ifas.ufl.edu/pdf/IN/IN101400.pdf

Grzywacz, D., Stevenson, P., Mushobozi, W. L., Belmain, S., & Wilson, K. (2014). The use of indigenous ecological resources for pest control in Africa. *Food Security*, *6*(1), 71-86. https://doi.org/10.1007/s12571-013-0313-5

Gu, H., Fitt, G. P., & Baker, G. H. (2007). Invertebrate pests of canola and their management in Australia: A review. *Australian Journal of Entomology*, *46*(3), 231-243. https://doi.org/10.1111/j.1440-6055.2007.00594.x

Gul, W., & Hamann, M. T. (2005). Indole alkaloid marine natural products: An established source of cancer drug leads with considerable promise for the control of parasitic, neurological and other diseases. *Life Sciences*, *78*(5), 442-453. https://doi.org/10.1016/j.lfs.2005.09.007

Gupta, S., & Dikshit, A. (2010). Biopesticides: An ecofriendly approach for pest control. *Journal of Biopesticides*, *3*, 186-188.

Gyanoba, M. S. C. (2018). *Bio-efficacy of newer insecticides against Diamondback moth (Plutella xylostella L.) and their residues in cabbage* [PhD Thesis, Mahatma Phule Krishi Vidyapeeth], Maharashtra, India. https://doi.org/10.20546/ijcmas.2018.707.349

Hachey, J. E., Sharma, K. K., & Moloney, M. M. (1991). Efficient shoot regeneration of *Brassica campestris* using cotyledon explants cultured in *vitro*. *Plant Cell Reports*, *9*(10), 549-554. https://doi.org/10.1007/BF00232329.

Harakley, F. (1969). Biological studies on the cabbage web-wrom, *Hellula undalis* Fabr. (Lepidoptera: Crambidae-Pyraustinae). Bulletin de la Societe Entomologique d'Egypte. https://agris.fao.org/agris-search/search.do?recordID=US201301190344

Harborne, J. B., & Williams, C. A. (2000). Advances in flavonoid research since 1992. *Phytochemistry*, *55*(6), 481-504. https://doi.org/10.1016/s0031-9422(00)00235-1.

Henn, T., & Weinzierl, R. (1989). *Botanical insecticides and insecticidal soaps*: University of Illinois at Urbana-Champaign, College of Agriculture, Cooperative Extension Service. https://www.ideals.illinois.edu/bitstream/handle/2142/28854/botanicalinsecti1296henn.pdf?sequence=1

Hikal, W. M., Baeshen, R. S., & Said-Al Ahl, H. A. (2017). Botanical insecticide as simple extractives for pest control. *Cogent Biology*, *3*(1), 1-16. https://doi.org/10.1080/23312025.2017.1404274

Hines, R., & Hutchison, W. (2013). Cabbage aphids. VegEdge, vegetable IPM resource for the Midwest. *University of Minnesota, Minneapolis, MN*. https://edis.ifas.ufl.edu/pdf/IN/IN101400.pdf

Huang, X. P., & Renwick, J. (1995). Cross habituation to feeding deterrents and acceptance of a marginal host plant by *Pieris rapae* larvae. *Entomologia Experimentalis et Applicata*, *76*(3), 295-302. https://doi.org/10.1111/j.1570-7458.1995.tb01973.x

Huaripata, C., & Sánchez, G. (2019). Life cycle of the Diamondback moth *Plutella xylostella* L.(Lepidoptera: Plutellidae), in broccoli and cauliflower under laboratory conditions. *Peruvian Journal of Agronomy*, *3*(1), 1-5. https://doi.org/10.21704/pja.v3i1.1132

Ibraheam, I. A., Hadi, M. Y., & Hameed, I. H. (2017). Analysis of Bioactive Compounds of Methanolic Leaves extract of *Mentha pulegium* Using Gas Chromatography-Mass Spectrometry (GC-MS) Technique. *International Journal of Pharmaceutical and Clinical Research*, *8*(04), 174-182.

Ileke, K., & Oni, M. (2011). Toxicity of some plant powders to maize weevil, *Sitophilus zeamais* (motschulsky)[Coleoptera: Curculiondae] on stored wheat grains (*Triticum aestivum*). *African Journal of Biotechnology*, *6*(13), 3043-3048. https://doi.org/10.5897/AJAR11.622

Imran, M., Kanwal Hanif, M. A., Nasir, M., & Sheikh, U. A. A. (2017). Comparative toxicity of insecticides against two important insect pests of cauliflower crop. *Asian Journal of Agriculture and Biology*, *5*(2), 88-98.

Isman, M. B. (2006). Botanical insecticides, deterrents, and repellents in modern agriculture and an increasingly regulated world. *Annual Review of Entomology*, *51*(1), 45-66. http://dx.doi.org/10.1146/annurev.ento.51.110104.151146.

Jahan, F., Abbasipour, H., Askarianzade, A., Hasanshahi, G., & Saeedizadeh, A. (2013). Effect of eight cauliflower cultivars on biological parameters of the cabbage aphid, *Brevicoryne brassicae* (L.)(Hem: Aphididae) in laboratory conditions. *Archives of Phytopathology and Plant Protection*, *46*(6), 636-642. https://doi.org/10.1080/03235408.2012.749697

Jogia, M. K., Andersen, R. J., Parkanyi, L., Clardy, J., Dublin, H. T., & Sinclair, A. (1989). Crotofolane diterpenoids from the African shrub *Croton dichogamus* Pax. *The Journal of Organic Chemistry*, *54*(7), 1654-1657. https://doi.org/10.1021/J000268A029

Johnson, W., Bergfeld, W. F., Belsito, D. V., Hill, R. A., Klaassen, C. D., Liebler, D. C., Marks, J. G., Shank, R. C., Slaga, T. J., & Snyder, P. W. (2017). Safety assessment of benzyl alcohol, benzoic acid and its salts, and benzyl benzoate. *International Journal of Toxicology*, *36*(3), 5-30. https://doi.org/10.1177/1091581817728996

Joia, B. S., Chawla, R. P., & Udeaan, A. S. (1996, August). *Present insecticide use practices on cole crops in Punjab and strategies for managing multiple insecticide resistance in diamondback moth*. In *Proceedings of the XX. International Congress of Entomology*, Florence, Italy. (pp. 19-106). https://www.cabi.org/isc/datasheet/42318

Justus, K., & Mitchell, B. (1996). Oviposition site selection by the Diamondback moth, *Plutella xylostella* (L.) (Lepidoptera: Plutellidae). *Journal of Insect Behavior*, *9*(6), 887-898. https://doi.org/10.1007/BF02208976

Kamanula, J., Sileshi, G. W., Belmain, S. R., Sola, P., Mvumi, B. M., Nyirenda, G. K., Nyirenda, S. P., & Stevenson, P. C. (2010). Farmers' insect pest management practices and pesticidal plant use in the protection of stored maize and beans in Southern Africa. *International Journal of Pest Management*, *57*(1), 41-49. https://doi.org/10.1080/09670874.2010.522264

Kamatou, G. P., Vermaak, I., & Viljoen, A. M. (2012). Eugenol—from the remote Maluku Islands to the international market place: A review of a remarkable and versatile molecule. *Molecules*, *17*(6), 6953-6981. https://doi.org/10.3390/molecules17066953

Kandi, S., Godishala, V., Rao, P., & Ramana, K. (2015). Biomedical significance of terpenes: an insight. *Biomedicine*, *3*(1), 8-10. https://doi.org/10.12691/BB-3-1-2

Kapeleka, J. A., Sauli, E., Sadik, O., & Ndakidemi, P. A. (2019). Biomonitoring of acetylcholinesterase (AChE) activity among smallholder horticultural farmers occupationally exposed to mixtures of pesticides in Tanzania. *Journal of Environmental and Public Health, 2019,* 1–11. https://doi.org/10.1155/2019/3084501

Kareru, P., Rotich, Z. K., & Maina, E. W. (2013). Use of botanicals and safer insecticides designed in controlling insects: The African case. *Insecticides–Development of Safer and More Effective Technologies*, *10*, 297-309.

Keasling, J. (2008). From yeast to alkaloids. *Nature Chemical Biology*, *4*(9), 524-525. https://doi.org/10.1038/nchembio0908-524.

Kessing, J., & Mau, R. (1991). Cabbage aphid, *Brevicoryne brassicae* (Linnaeus). *Crop Knowledge Master.* Department of Entomology, Honolulu, Hawaii. http://entomology.ifas.ufl.edu/creatures.

Khanh, T. D., Xuan, T. D., & Chung, I. M. (2007). Rice allelopathy and the possibility for weed management. *Annals of Applied Biology, 151,* 325 - 339. https://doi.org/10.1111/j.1744-7348.2007.00183.x

Khater, H. F. (2012). Prospects of botanical biopesticides in insect pest management. *Pharmacologia*, *3*(12), 641-656. https://doi.org/10.5567/pharmacologia.2012.641.656

Kiran, S., & Prakash, B. (2015). Toxicity and biochemical efficacy of chemically characterized *Rosmarinus officinalis* essential oil against *Sitophilus oryzae* and *Oryzaephilus surinamensis*. *Industrial Crops and Products*, *74*(2015), 817-823. https://doi.org/10.1016/j.indcrop.2015.05.073

Koona, P., & Dorn, S. (2005). Extracts from *Tephrosia vogelii* for the protection of stored legume seeds against damage by three bruchid species. *Annals of Applied Biology*, *147*(1), 43-48. http://dx.doi.org/10.1111/j.1744-7348.2005.00006.x.

Kordali, S., Cakir, A., Mavi, A., Kilic, H., & Yildirim, A. (2005). Screening of chemical composition and antifungal and antioxidant activities of the essential oils from three Turkish *Artemisia species*. *Journal of Agricultural and Food Chemistry*, *53*(5), 1408-1416. http://dx.doi.org/10.1021/jf048429n

Krishnamoorthy, A. (2004). Biological control of Diamondback moth *Plutella xylostella* (L.), an Indian scenario with reference to past and future strategies. In *A. A. Kirk, & D. Bordat* (eds). Improving biocontrol of *Plutella xylostella*. *Proceedings of the International Symposium*, Montpellier, CIRAD. https://agritrop.cirad.fr/511169/

Labou, B., Brévault, T., Sylla, S., Diatte, M., Bordat, D., & Diarra, K. (2017). Spatial and temporal incidence of insect pests in farmers' cabbage fields in Senegal. *International Journal of Tropical Insect Science*, *37*(4), 225-233. https://doi.org/10.1017/S1742758417000200

Landrum, P. F., & Fisher, S. W. (1999). Influence of lipids on the bioaccumulation and trophic transfer of organic contaminants in aquatic organisms. *Lipids in Freshwater Ecosystems*: Springer. https://link.springer.com/chapter/10.1007/978-1-4612-0547-0_10

Lashkari, M. R., Sahragard, A., & Ghadamyari, M. (2007). Sublethal effects of imidacloprid and pymetrozine on population growth parameters of cabbage aphid, *Brevicoryne brassicae* on rapeseed, *Brassica napus* L. *Insect Science*, *14*(3), 207-212. https://doi.org/10.1111/j.1744-7917.2007.00145.x

Lee, M. S., Choi, J., Posadzki, P., & Ernst, E. (2012). Aromatherapy for health care: An overview of systematic reviews. *Maturitas*, *71*(3), 257-260. https://doi.org/10.1016/j.maturitas.2011.12.018

Le Roy, C., & Wrana, J. L. (2005). Signaling and endocytosis: A team effort for cell migration. *Developmental Cell*, *9*(2), 167-168. https://doi.org/10.1016/j.devcel.2005.07.007

Li, Z., Feng, X., Liu, S. S., You, M., & Furlong, M. J. (2016). Biology, ecology, and management of the diamondback moth in China. *Annual Review of Entomology*, *61*(2016), 277-296. https://doi.org/10.1146/annurev-ento-010715-023622

Li, Z., Zalucki, M., Yonow, T., Kriticos, D., Bao, H., Chen, H., Hu, Z., Feng, X., & Furlong, M. (2016). Population dynamics and management of diamondback moth (*Plutella xylostella*) in China: the relative contributions of climate, natural enemies and cropping patterns. *Bulletin of Entomological Research*, *106*(2), 197-214. https://doi.org/10.1017/S0007485315001017

Lingren, P., & Green, G. (1984). *Suppression and management of cabbage looper populations*. U.S. Dept. of Agriculture, Agricultural Research Service and U.S. G.P.O. Washington, DC, USA. https://handle.nal.usda.gov/10113/CAT85819524

Liu, T. X., & Sparks Jr, A. N. (2001). *Aphids on cruciferous crops: Identification and management*. Texas FARMER Collection, Texas A&M University, USA. https://cdn-ext.agnet.tamu.edu/wp-content/uploads/2018/10/EB-6109-aphids-on-cruciferous-crops-identification-and-management.pdf

Liu, Y. Q., Shi, Z. H., Zalucki, M. P., & Liu, S. S. (2014). Conservation biological control and IPM practices in Brassica vegetable crops in China. *Biological Control*, *68*(2014), 37-46. https://doi.org/10.1016/j.biocontrol.2013.06.008

Lorent, K., Gong, W., Koo, K. A., Waisbourd-Zinman, O., Karjoo, S., Zhao, X., Sealy, I., Kettleborough, R. N., Stemple, D. L., & Windsor, P. A. (2015). Identification of a plant isoflavonoid that causes biliary atresia. *Science Translational Medicine*, *7*(286), 267-286. https://doi.org/10.1126/scitranslmed.aaa1652.

Macharia, I., Löhr, B., & De Groote, H. (2005). Assessing the potential impact of biological control of *Plutella xylostella* (diamondback moth) in cabbage production in Kenya. *Crop Protection*, *24*(11), 981-989. https://doi.org/10.1016/j.cropro.2005.02.005

MacLachlan, D. J., & Hamilton, D. (2010). Estimation methods for maximum residue limits for pesticides. *Regulatory Toxicology and Pharmacology*, *58*(2), 208-218. https://doi.org/10.1016/j.yrtph.2010.05.012

Mahr, D., & Ridgway, N. (1993). *Biological control of insects and mites* (Vol. 481). North Central Regional Publication. Greenwood, USA. ttps://link.springer.com/book/10. 1007/ 978-3-030-63990-7? noAccess=true

Makoi, J. H., Belane, A. K., Chimphango, S. B., & Dakora, F. D. (2010). Seed flavonoids and anthocyanins as markers of enhanced plant defence in nodulated cowpea (*Vigna unguiculata* L. Walp.). *Field Crops Research*, *118*(1), 21-27. https://doi.org/10.1016/j. fcr.2010.03.012

Mary, A., & Giri, R. (2017). GC-MS analysis of bioactive compounds of *Achyranthes aspera*. *World Journal of Pharamaceutical Research*, *7*(1), 1045-1056.

Mazid, M., Khan, T., & Mohammad, F. (2011). Role of secondary metabolites in defense mechanisms of plants. *Biology and Medicine*, *3*(2), 232-249.

McNaught, A. D., & Wilkinson, A., (1997). *"IUPAC Compendium of Chemical Terminology"*. (2nd ed.), Blackwell Scientific, Oxford. https://doi.org/10.1351/goldbook

Mehta, P., Shah, R., Lohidasan, S., & Mahadik, K. (2015). Pharmacokinetic profile of phytoconstituent (s) isolated from medicinal plants a comprehensive review. *Journal of Traditional and Complementary Medicine*, *5*(4), 207-227. https://doi.org/10.1016/j. jtcme.2014.11.041

Mersha, W., Ayele, N., Fentahun, G., Getinet, M., Kassu, K., & Nagappan, R. (2014). Repellent and insecticidal activity of *Mentha piperita* (L.) plant extracts against cabbage aphid [*Brevicoryne brassicae* Linn.(Homoptera: Aphididae)]. *American-Eurasian Journal of Scientific Research*, *9*(6), 150-156. https://doi.org/10.5829/idosi. aejsr.2014.9.6.1141

Mierziak, J., Kostyn, K., & Kulma, A. (2014). Flavonoids as important molecules of plant interactions with the environment. *Molecules*, *19*(10), 16240-16265. https://doi.org/10. 3390/molecules191016240

Mkenda, P., Mwanauta, R., Stevenson, P. C., Ndakidemi, P., Mtei, K., & Belmain, S. R. (2015). Extracts from field margin weeds provide economically viable and environmentally benign pest control compared to synthetic pesticides. *PLoS One*, *10*(11), 1-14. https://doi.org/10.1371/journal.pone.0143530

Mkenda, P. A., Mtei, K., & Ndakidemi, P. A. (2014). Pesticidal efficacy of *Tephrosia vogelii* and *Tithonia diversifolia* against field insect pests of common beans [*Phaseolus vulgaris* L.] within African farming communities. *African Journal of Applied Agricultural Sciences and Technologies*, *2*(1), 9-26.

Mkindi, A., Mpumi, N., Tembo, Y., Stevenson, P. C., Ndakidemi, P. A., Mtei, K., Machunda, R., & Belmain, S. R. (2017). Invasive weeds with pesticidal properties as potential new crops. *Industrial Crops and Products*, *110*(2017), 113-122. https://doi.org/10.1016/j.indcrop.2017.06.002

Mkindi, A. G., Tembo, Y., Mbega, E. R., Medvecky, B., Kendal-Smith, A., Farrell, I. W., Ndakidemi, P. A., Belmain, S. R., & Stevenson, P. C. (2019). Phytochemical analysis of *Tephrosia vogelii* across East Africa reveals three chemotypes that influence its use as a pesticidal plant. *Plants*, *8*(12), 1-11. https://doi.org/10.3390/plants8120597.

Mochiah, M., Banful, B., Fening, K., Amoabeng, B., Ekyem, S., Braimah, H., & Owusu-Akyaw, M. (2011). Botanicals for the management of insect pests in organic vegetable production. *Journal of Entomology and Nematology*, *3*(6), 85-97.

Mohan, M., & Gujar, G. (2003). Local variation in susceptibility of the diamondback moth, *Plutella xylostella* (Linnaeus) to insecticides and role of detoxification enzymes. *Crop Protection*, *22*(3), 495-504. https://doi.org/10.1016/S0261-2194(02)00201-6

Moharramipour, S., Monfared, A., & Fathipour, Y. (2003). Comparison of intrinsic rate of increase and mean relative growth rate of cabbage aphid (*Brevicoryne brassicae* L.) on four rapeseed (*Brassica napus* L.) varieties in growth room. *Agricultural Science (Tabriz)*, *13*(3), 79-86.

Mpumi, N., Machunda, R. L., Mtei, K. M., & Ndakidemi, P. A. (2021). Insecticidal Efficacy of *Syzygium aromaticum*, *Tephrosia vogelii* and *Croton dichogamus* Extracts against *Plutella xylostella* and *Trichoplusia ni* on *Brassica oleracea* crop in Northern Tanzania. *AIMS Agriculture and Food*, *6*(1), 185-202. https://doi.org/10.3934/agrfood.2021012

Mpumi, N., Machunda, R. L., Mtei, K. M., & Ndakidemi, P. A. (2020). Selected insect pests of economic importance to *Brassica oleracea*, their control strategies and the potential threat to environmental pollution in Africa. *Sustainability*, *12*(9), 1-22. https://doi.org/10.3390/su12093824

Mpumi, N., Mtei, K., Machunda, R., & Ndakidemi, P. A. (2016). The toxicity, persistence and mode of actions of selected botanical pesticides in Africa against insect pests in common beans, *P. vulgaris*: A review. *American Journal of Plant Sciences*, *2016*(7), 138-151. https://doi.org/10.4236/ajps.2016.71015

Msikita, W., Wilkinson, H., & Skirvin, R. (1992). Propagation of Tronchuda (*Brassica oleracea* var. tronchuda Bailey) from Cuttings. *HortScience*, *27*(9), 1036-1038. https://doi.org/10.21273/HORTSCI.27.9.1036

Mudzingwa, S., Muzemu, S., & Chitamba, J. (2013). Pesticidal efficacy of crude aqueous extracts of *Tephrosia vogelii* L., *Allium sativum* L. and *Solanum incanum* L. in controlling aphids (*Brevicoryne brassicae* L.) in rape (*Brassica napus* L.) n controlling aphids (*Brevicoryne brassicae* L.) in rape (*Brassica napus* L.). *Journal of Research in Ariculture*, *2*(1). 157–163. http://hdl.handle.net/11408/991

Munthali, M. G., Gachene, C. K., Sileshi, G. W., & Karanja, N. K. (2014). Amendment of Tephrosia Improved Fallows with Inorganic Fertilizers Improves Soil Chemical Properties, N Uptake, and Maize Yield in Malawi. *International Journal of Agronomy*, *2014*, 1-10. https://doi.org/10.1155/2014/902820

Murillo, G., Salti, G., Kosmeder Ii, J., Pezzuto, J., & Mehta, R. (2002). Deguelin inhibits the growth of colon cancer cells through the induction of apoptosis and cell cycle arrest. *European Journal of Cancer*, *38*(18), 2446-2454. https://doi.org/10.1016/S0959-8049(02)00192-2

Mwanauta, R. W., Mtei, K. A., & Ndakidemi, P. A. (2014). Prospective Bioactive Compounds from *Vernonia amygdalina*, *Lippia javanica*, *Dysphania ambrosioides* and *Tithonia diversifolia* in Controlling Legume Insect Pests. *Agricultural Sciences*, *5*(12), 1129-1139. http://dx.doi.org/10.4236/as.2014.512123.

Mwanauta, R. W., Mtei, K. M., & Ndakidemi, P. A. (2015). Potential of controlling common bean insect pests (Bean Stem Maggot (*Ophiomyia phaseoli*), Ootheca (*Ootheca bennigseni*) and Aphids (*Aphis fabae*)) using agronomic, biological and botanical practices in field. *Agricultural Sciences*, *6*(05), 489-497. http://dx.doi.org/10.4236/as.2015.65048.

Ndakidemi, P. A., & Dakora, F. D. (2003). Legume seed flavonoids and nitrogenous metabolites as signals and protectants in early seedling development. *Functional Plant Biology*, *30*(7), 729-745. http://dx.doi.org/10.1071/FP03042

Nerio, L. S., Olivero-Verbel, J., & Stashenko, E. (2010). Repellent activity of essential oils: a review. *Bioresource Technology*, *101*(1), 372-378. http://dx.doi.org/10.1016/j.biortech.2009.07.048.

Ngowi, A., Mbise, T., Ijani, A., London, L., & Ajayi, O. (2007). Smallholder vegetable farmers in Northern Tanzania: Pesticides use practices, perceptions, cost and health effects. *Crop Protection*, *26*(11), 1617-1624. http://dx.doi.org/10.1016/j.cropro.2007.01.008.

Ntow, W. J. (2008). *The use and fate of pesticides in vegetable-based agro-ecosystems in Ghana* [PhD Dissertation, Wageningen University]. CRC Press. https: //library. wur.nl /WebQuery/wurpubs/360668

Ntow, W. J., Gijzen, H. J., Kelderman, P., & Drechsel, P. (2006). Farmer perceptions and pesticide use practices in vegetable production in Ghana. *Pest Management Science*, *62*(4), 356-365. http://dx.doi.org/10.1002/ps.1178

Nuessly, G., & Webb, S. (2003). Insect management for leafy vegetables (Lettuce, Endive, and Escarole). *Entomology & Nematology Department document ENY-475. Florida Cooperative Extension Service, Institute of Food and Agricultural Sciences, University of Florida*. https://edis.ifas.ufl.edu/pdf/IG/IG16100.pdf

Nyirenda, S. P., Sileshi, G. W., Belmain, S. R., Kamanula, J. F., Mvumi, B. M., Sola, P., Nyirenda, G. K., & Stevenson, P. C. (2011). Farmers' ethno-ecological knowledge of vegetable pests and pesticidal plant use in Malawi and Zambia. *African Journal of Agricultural Research*, *6*(6), 1525-1537.

Obopile, M., Munthali, D., & Matilo, B. (2008). Farmers' knowledge, perceptions and management of vegetable pests and diseases in Botswana. *Crop Protection*, *27*(8), 1220-1224. https://doi.org/10.1016/j.cropro.2008.03.003

Oliveira, R. A. D., Reis, T. V., Sacramento, C. K. D., Duarte, L. P., & Oliveira, F. F. D. (2009). Constituintes químicos voláteis de especiarias ricas em eugenol. *Revista Brasileira de Farmacognosia*, *19(*3), 771-775. https://doi.org/10.1590/S0102-695X2009000500020

Ondieki, J. J. (1996). The current state of pesticide management in Sub-Saharan Africa. *Science of the Total Environment*, *188*(1), 30-34. https://doi.org/10.1177/1070496520974407

Opfer, P., & McGranth, D. (2013). *Oregon vegetables, cabbage aphid and green peach aphid. Department of Horticulture*. Oregon State University Corvallis, Corvallis, Disponível, USA. https://edis.ifas.ufl.edu/pdf/IN/IN101400.pdf

Orr, A., & Ritchie, J. M. (2004). Learning from failure: Smallholder farming systems and IPM in Malawi. *Agricultural Systems*, *79*(1), 31-54. https://doi.org/10.1016/S0308-521X (03)00044-1

Orwa, C., Mutua, A., Kindt, R., Jamnadass, R., & Simons, A. (2009). Agroforestree database: A tree species reference and selection guide version 4.0. World Agroforestry Centre ICRAF, *Nairobi,* Kenya. http://www.worldagroforestry.org/af/treedb/

Pal, K. K., & Gardener, B. M. (2006). *Biological control of plant pathogens*. Ohio State University, OARDC, Wooster. https: //www.apsnet.org /edcenter/disimpactmngmnt /topc/ Documents/PHI-BiologicalControl.pdf

Papachristos, D. P., Karamanoli, K. I., Stamopoulos, D. C., & Menkissoglu-Spiroudi, U. (2004). The relationship between the chemical composition of three essential oils and their insecticidal activity against *Acanthoscelides obtectus* (Say). *Pest Management Science: Formerly Pesticide Science*, *60*(5), 514-520. https://doi.org/10.1002/ps.798.

Patait, D., Shetgar, S., Subhan, S., Badgujar, A., & Dhurgude, S. (2008). Seasonal abundance of Lepidopteran pests infesting cabbage in relation to weather parameters. *Indian Journal of Entomology*, *70*(3), 255-258.

Patra, A. K. (2012). *An overview of antimicrobial properties of different classes of phytochemicals: Dietary phytochemicals and microbes*. Springer Netherlands. https: //doi.org /10.1007 /978-94-007-3926-0_1

Patra, S., Dhote, V., Alam, S., Das, B., Chatterjee, M., & Samanta, A. (2013). Population dynamics of major insect pests and their natural enemies on cabbage under new alluvial zone of West Bengal. *The Journal of Plant Protection Sciences, 5*(1), 42-49.

Pedigo, L. P., & Rice, M. E. (2014). *Entomology and pest management*: Waveland Press, Inc. https://www.worldcat.org/oclc/214285952?referer=di&ht=edition

Pérez-Lucas, G., Vela, N., El Aatik, A., & Navarro, S. (2018). Environmental risk of groundwater pollution by pesticide leaching through the soil profile. *Pesticides, Anthropogenic Activities and the Health of our Environment*. IntechOpen. https://www.intechopen.com/chapters/64602

Posadzki, P., Alotaibi, A., & Ernst, E. (2012). Adverse effects of aromatherapy: A systematic review of case reports and case series. *International Journal of Risk & Safety in Medicine*, *24*(3), 147-161. https://doi.org/10.3233/JRS-2012-0568

Prijono, D. 1998). Insecticdal activity of Meliaceous seed extracts against *Crocidolomia binotalis* Zeller (Lepidoptera: Pyralidae). *Bulletin of Plant Pests and Diseases*, *10*(1), 1-7. https://doi.org/10.22146/jpti.10069

Prijono, D., & Hassan, E. (1993). Laboratory and field efficacy of neem (*Azadirachta indica* A. Juss) extracts against two broccoli pests. *Journal of Plant Diseases and Protection*, *100*(4), 354-370.

Rajaram, N., & Janardhanan, K. (1991). The biochemical composition and nutritional potential of the tribal pulse, *Mucuna gigantea* (Willd.) DC. *Plant Foods for Human Nutrition, 44*, 45–51. https://doi.org/10.1007/BF02196381

Ramankutty, N., Mehrabi, Z., Waha, K., Jarvis, L., Kremen, C., Herrero, M., & Rieseberg, L. H. (2018). Trends in global agricultural land use: Implications for environmental health and food security. *Annual Review of Plant Biology*, *69*(2018), 789-815. https://doi.org/ 10.1146/annurev-arplant-042817-040256

Ramawat, K. G., & Mérillon, J. M. (2013). *Natural products: Phytochemistry, botany and metabolism of alkaloids, phenolics and terpenes*: Springer. Nature Switzerland AG. https://doi.org/10.1007/978-3-642-22144-6

Rathi, J. M., & Gopalakrishnan, S. (2010). Insecticidal activity of methanolic pooled fractions of *Lantana wightiana* Wall. *Journal of Biopesticides*, *3*(1), 282-285.

Razaq, M., Maqsood, S., Aslam, M., Shad, S. A., & Afzal, M. (2012). Effect of plant spacing on aphid population, yield components and oil contents of late sown canola, *Brassica napus* L. (Brassicaceae). *Pakistan Journal of Zoology*, *44*(4), 991-995.

Regnault-Roger, C., Vincent, C., & Arnason, J. T. (2012). Essential oils in insect control: low-risk products in a high-stakes world. *Annual Review of Entomology*, *57*(2012), 405-424. https://doi.org/10.1146/annurev-ento-120710-100554.

Ren, J., Wang, J., Karthikeyan, S., Liu, H., & Cai, J. (2019). Natural anti-phytopathogenic fungi compound phenol, 2, 4-bis (1, 1-dimethylethyl) from *Pseudomonas fluorescens* TL-1. *Indian Journal of Biochemistry and Biophysics*, *56*(2), 162-168.

Reynolds, W. F., & Enriquez, R. G. (2016). *Terpenes: Mono-, sesqui-, and higher Terpenes* Vol. 2. *Modern NMR Approaches to the Structure Elucidation of Natural Products: Volume 2: Data Acquisition and Applications to Compound Classes*. https://doi.org/ 10.1039/9781849734684-00251.

Ribera, A. E., & Zuñiga, G. (2012). Induced plant secondary metabolites for phytopatogenic fungi control: A review. *Journal of Soil Science and Plant Nutrition*, *12*(4), 893-911. http://dx.doi.org/10.4067/S0718-95162012005000040

Righi, K., Assia Righi, F., Boubkeur, A., Boungab, K., Elouissi, A., & Djendara, A. C. (2018). Toxicity and repellency of three Algerian medicinal plants against pests of stored product: *Ryzopertha dominica* (Fabricius)(Coleoptera: Bostrichidae). *Banat's Journal of Biotechnology*, *9*(17), 50-59. http://dx.doi.org/10.7904/2068-4738-IX(17)-50

Salatino, A., Salatino, M. L. F., & Negri, G. (2007). Traditional uses, chemistry and pharmacology of *Croton species* (Euphorbiaceae). *Journal of the Brazilian Chemical Society*, *18*(1), 11-33. https://doi.org/10.1590/S0103-50532007000100002

Samarasekera, R., Weerasinghe, I. S., & Hemalal, K. P. (2008). Insecticidal activity of menthol derivatives against mosquitoes. *Pest Management Science: Formerly Pesticide Science*, *64*(3), 290-295. https://doi.org/10.1002/ps.1516

Samatha, T., Srinivas, P., Shyamsundarachary, R., Rajinikanth, M., & Rama Swamy, N. (2012). Phytochemical analysis of seeds, stem bark and root of an endangered medicinal forest tree *Oroxylum indicum* (L) Kurz. *International Journal of Pharma and Bio Sciences*, *3*(3), 1063-1075.

Sasidharan, S., Chen, Y., Saravanan, D., Sundram, K., & Latha, L. Y. (2011). Extraction, isolation and characterization of bioactive compounds from plants' extracts. *African Journal of Traditional, Complementary and Alternative Medicines*, *8*(1), 1–10. https://doi.org/10.4314/ajtcam.v8i1.60483

Saviranta, N. M., Julkunen-Tiitto, R., Oksanen, E., & Karjalainen, R. O. (2010). Leaf phenolic compounds in red clover (*Trifolium pratense* L.) induced by exposure to moderately elevated ozone. *Environmental Pollution*, *158*(2), 440-446. https://doi.org/10.1016/j.envpol.2009.08.029

Saxena, J., Rai, S., Srivastava, K., & Sinha, S. (1989). Resistance in the field populations of the diamondback moth to some commonly used synthetic pyrethroids. *Indian Journal of Entomology*, *51*(3), 265-268.

Scaife, A., & Turner, M. (1983). *Diagnosis of mineral disorders in plants*: *Vegetables* (Vol. 2). Her Majesty's Stationery Office. https://www. amazon.com /Diagnosis – Mineral - Disorders - Plants – Vegetables /dp /0112408044

Schulz, R., Peall, S. K., Dabrowski, J. M., & Reinecke, A. J. (2001). Current-use insecticides, phosphates and suspended solids in the Lourens River, Western Cape, during the first rainfall event of the wet season. *Water Sa*, *27*(1), 65-70. https://doi.org/10.4314/wsa.v27i1.5012

Secoy, D., & Smith, A. (1983). Use of plants in control of agricultural and domestic pests. *Economic Botany*, *37*(1), 28-57. https://doi.org/10.1007/BF02859304

Seif, A., & Nyambo, B. (2013). *Integrated Pest Management for Brassica Production in East Africa*. ICIPE Science Press. http://hdl.handle.net/123456789/670

Sessini, V., Palenzuela, M., Damián, J., & Mosquera, M. E. (2020). Bio-based polyether from limonene oxide catalytic ROP as green polymeric plasticizer for PLA. *Polymer, 210,* 1-7. https://doi.org/10.1016/j.polymer.2020.123003

Sharma, M., & Gupta, S. (2012). Effective Callus Induction and Plant Regeneration in *Brassica napus* (L.) Var Dgs-1. *Journal of Cell and Tissue Research*, *12*(2), 3229-3234.

Shine, C., Reaser, J., & Gutierrez, A. (2003). *Invasive alien species in the Austral Pacific Region*: National Reports & Directory of Resources. The Global Invasive Species Programme. https://www.cabi.org/isc/20043073110

Shooshtari, M. B., Kashani, H. H., Heidari, S., & Ghal, R. (2013). Comparative mosquito repellent efficacy of alcoholic extracts and essential oils of different plants against Anopheles Stephensi. *African Journal of Pharmacy and Pharmacology, 7*(6), 310-314. https://doi.org/10.5897/AJPP12.1144

Shorey, H. H. (1963). A simple artificial rearing medium for the cabbage looper. *Journal of Economic Entomology*, *56*(4), 536-537. https://doi.org/10.1093/jee/56.4.536a

Silva, C. G., Oliveira, J., & Da Camara, C. A. (2018). Insecticidal activity of the ethanolic extract from *Croton species* against *Plutella xylostella* L. (Lepidoptera: Plutellidae). *Revista Facultad Nacional de Agronomía Medellín*, *71*(2), 8543-8551.

Sivapragasam, A., & Aziz, A. (1992, December 10-14). *Cabbage webworm on crucifers in Malaysia* [Second International Workshop]. Diamondback moth and other crucifer pests, Tainan, Taiwan. https://www.cabdirect.org/abstract/20207201931

Smartt, J., & Simmonds, N. W. (1995). *Evolution of crop plants* (2nd Ed.). Wiley-Blackwell. https://www.wiley.com/en-us/Evolution+of+Crop+Plants%2C+2nd+Edition-p-9780582086432

Sola, P., Mvumi, B., Ogendo, J., Mponda, O., Kamanula, J., Nyirenda, S., Belmain, S., & Stevenson, P. (2014). Botanical pesticide production, trade and regulatory mechanisms in sub-Saharan Africa: Making a case for plant-based pesticidal products. *Food Security*, *6*(3), 369-384. doi: http://dx.doi.org/10.1007/s12571-014-0343-7.

Sonneberg, D., Ndakidemi, P. A., & Laubscher, C. P. (2013). The effects of various drip fertigated water quantities on flavonoid and anthocyanin content on hydroponically cultivated *Cucumis sativa* L. *International Journal of Physical Sciences*, *8*(19), 012-1016. https://doi.org/10.5897/IJPS2013.3845

Sparks, T. C., & Nauen, R. (2015). IRAC: Mode of action classification and insecticide resistance management. *Pesticide Biochemistry and Physiology*, *121*, 122-128. https://doi.org/10.1016/j.pestbp.2014.11.014

Srivastava, A., Srivastava, S., & Syamsundar, K. (2005). Bud and leaf essential oil composition of *Syzygium aromaticum* from India and Madagascar. *Flavour and Fragrance Journal*, *20*(1), 51-53. https://doi.org/10.1002/ffj.1364

Stevenson, P. C., Kite, G. C., Lewis, G. P., Forest, F., Nyirenda, S. P., Belmain, S. R., Sileshi, G. W., & Veitch, N. C. (2012). Distinct chemotypes of *Tephrosia vogelii* and implications for their use in pest control and soil enrichment. *Phytochemistry*, *78*, 135-146. http://dx.doi.org/10.1016/j.phytochem.2012.02.025.

Stevenson, P. C., Sola, P., Brighton, M. M., Gurr, G., Kamanula, J. F., Anjarwalla, P., & Belmain, S. R. (2015). Optimising pesticidal plants in Africa: Technology innovation, outreach and networks. *Revista de Protección Vegetal*, *30*, 33-36.

Stewart, J. K., Aharoni, Y., Hartsell, P. L., & Young, D. K. (1980). Acetaldehyde fumigation at reduced pressures to control the green peach aphid on wrapped and packed head lettuce. *Journal of Economic Entomology*, *73*(1), 149-152. https://doi.org/10.1093/jee/73.1.149

Sun, C., Tsai, Y., & Chiang, F. (1992). *Resistance in the diamondback moth to pyrethroids and benzoylphenylureas*: ACS Publications, Washington, DC, USA. https://doi=10.1.1.1058.8030

Tak, J. H., & Isman, M. B. (2017a). Enhanced cuticular penetration as the mechanism of synergy for the major constituents of thyme essential oil in the cabbage looper, *Trichoplusia ni*. *Industrial Crops and Products*, *101*, 29-35. https://doi.org/10.1016/j.indcrop.2017.03.003

Tak, J. H., & Isman, M. B. (2017b). Penetration-enhancement underlies synergy of plant essential oil terpenoids as insecticides in the cabbage looper, *Trichoplusia ni*. *Scientific Reports, 7*, 1-9. https://doi.org/10.1038/srep42432.

Tak, J. H., Jovel, E., & Isman, M. B. (2016). Comparative and synergistic activity of *Rosmarinus officinalis* L. essential oil constituents against the larvae and an ovarian cell line of the cabbage looper, *Trichoplusia ni* (Lepidoptera: Noctuidae). *Pest Management Science*, *72*(3), 474-480. https://doi.org/10.1002/ps.4010

Takahashi, H., Mitsui, J., Takakusa, N., Matsuda, M., Yoneda, H., Suzuki, J., Ishimitsu, K., & Kishimoto, T. (1992, November 23-26). *NI-25, a new type of systemic and broad spectrum insecticide.* Brighton Crop Protection Conference, Pests and Diseases, Brighton, UK. https://www.cabi.org/isc/abstract/19931167697

Tanyi, C. B., Ngosong, C., & Ntonifor, N. N. (2018). Effects of climate variability on insect pests of cabbage: Adapting alternative planting dates and cropping pattern as control measures. *Chemical and Biological Technologies in Agriculture, 5*(1), 1–11. https://doi.org/10.1186/s40538-018-0140-1

Tembo, Y., Mkindi, A. G., Mkenda, P. A., Mpumi, N., Mwanauta, R., Stevenson, P. C., Ndakidemi, P. A., & Belmain, S. R. (2018). Pesticidal plant extracts improve yield and reduce insect pests on legume crops without harming beneficial arthropods. *Frontiers in Plant Science*, *9*(1425), 1-10. https://doi.org/10.3389/fpls.2018.01425

Tian, B. L., Liu, Q. Z., Liu, Z. L., Li, P., & Wang, J. W. (2015). Insecticidal potential of clove essential oil and its constituents on *Cacopsylla chinensis* (Hemiptera: Psyllidae) in laboratory and field. *Journal of Economic Entomology*, *108*(3), 957-961. https://doi.org/10.1093/jee/tov075

Timbilla, J., & Nyarko, K. (2004). A survey of cabbage production and constraints in Ghana. *Ghana Journal of Agricultural Science*, *37*(1), 93-101. https://doi.org/10.4314/gjas.v37i1.2084

Toba, H., Kishaba, A., Pangaldan, R., & Vail, P. (1973). Temperature and the development of the cabbage looper. *Annals of the Entomological Society of America*, *66*(5), 965-974. https://doi.org/10.1093/aesa/66.5.965

Tonisi, S., Okaiyeto, K., Hoppe, H., Mabinya, L. V., Nwodo, U. U., & Okoh, A. I. (2020). Chemical constituents, antioxidant and cytotoxicity properties of *Leonotis leonurus* used in the folklore management of neurological disorders in the Eastern Cape, South Africa. *3 Biotech*, *10*(3), 1-14. https://doi.org/10.1007/s13205-020-2126-5

Umar, A., Piero, N., Mgutu, A., Ann, N., Maina, G., & Maina, M. (2015). Bio efficacy of aqueous crude fruit sap extract of *Solanum incanum* against green peach aphids *Myzus persicae* Sulzer (Homoptera: Aphididae). *Entomology, Ornithology and Herpetology*, *5*(169), 1-5. https://doi.org/10.4172/2161-0983.1000169

Umina, P. A., Edwards, O., Carson, P., Van Rooyen, A., & Anderson, A. (2014). High levels of resistance to carbamate and pyrethroid chemicals widespread in Australian *Myzus persicae* (Hemiptera: Aphididae) populations. *Journal of Economic Entomology*, *107*(4), 1626-1638. https://doi.org/10.1603/ec14063

Usui, K., Kurihara, M., Uchiumi, K., Fukami, J., & Tatsuki, S. (1987). Sex pheromone of the female large cabbage-heart caterpillar, *Crocidolomia binotalis* Zeller (Lepidoptera: Pyralidae). *Agricultural and Biological Chemistry*, *51*(8), 2191-2195. https://doi.org/10.1080/00021369.1987.10868350

Vahedi, H., Lari, J., Nasrabadi, M., & Halimi, M. (2013). Chemical constituents and antimicrobial activity of *Crucianella sintenisii* growing wild in Iran. *Chemistry of Natural Compounds*, *49*(5), 955-957. https://doi.org/10.1007/s10600-013-0792-9

Valenzuela, I., & Hoffmann, A. A. (2015). Effects of aphid feeding and associated virus injury on grain crops in A ustralia. *Austral Entomology*, *54*(3), 292-305. https://doi.org/10.1111/aen.12122

Van Emden, H., Eastop, V., Hughes, R., & Way, M. (1969). The ecology of *Myzus persicae*. *Annual Review of Entomology*, *14*(1), 197-270. https://doi.org/10.1146/annurev.en.14.010169.001213

Venkateswara Rao, P., Kiran, S., Rohini, P., & Bhagyasree, P. (2017). Flavonoid: A review on Naringenin. *Journal of Pharmacognosy and Phytochemistry*, *6*(5), 2778-2783.

Verma, A., & Sandhu, G. (1968). Chemical control of diamondback moth, *Plutella maculipennis* (Curtis). *Journal of Research, Punjab Agricultural University, 5,* 420-423.

Vuković, S., Inđić, D., Gvozdenac, S., & Červenski, J. (2014). Efficacy of Insecticides in the Control of Cabbage Pests. *Research Journal of Agricultural Science*, *46*(2), 421-425.

Waiganjo, M., Waturu, C., Mureithi, J., Muriuki, J., Kamau, J., & Munene, R. (2011). Use of entomopathogenic fungi and neem bio-pesticides for Brassica pests control and conservation of their natural enemies. *East African Agricultural Forestry Journal*, *77,* 545-549. https://doi.org/10.9734/IJPSS/2017/38666

Walker, G., Cameron, P., & Berry, N. (2001, November 26-29). *Implementing an IPM programme for vegetable brassicas in New Zealand* [Proceedings of the Fourth International Workshop]. The management of diamondback moth and other crucifer pests, Melbourne, Australia. https://www.cabdirect.org/abstract/20093098370

Wangchuk, P., Keller, P. A., Pyne, S. G., Taweechotipatr, M., Tonsomboon, A., Rattanajak, R., & Kamchonwongpaisan, S. (2011). Evaluation of an ethnopharmacologically selected Bhutanese medicinal plants for their major classes of phytochemicals and biological activities. *Journal of Ethnopharmacology*, *137*(1), 730-742. https://doi.org/ 10.1016/j. jep.2011.06.032

War, A. R., Paulraj, M. G., Ahmad, T., Buhroo, A. A., Hussain, B., Ignacimuthu, S., & Sharma, H. C. (2012). Mechanisms of plant defense against insect herbivores. *Plant Signaling & Behavior*, *7*(10), 1306-1320. https://doi.org/10.1093/aobpla/ply037

Waterhouse, D. F., & Norris, K. R. (1987). *Biological control: Pacific prospects*. Inkata Press. https://www.cabi.org/isc/abstract/19881108201

Waterhouse, D. F., & Sands, D. P. A. (2001). *Classical biological control of arthropods in Australia*. Australian Centre for International Agricultural Research (ACIAR). https://www.cabi.org/ISC/abstract/20013072548

Weinberger, K., & Srinivasan, R. (2009). Farmers' management of cabbage and cauliflower pests in India and their approaches to crop protection. *Journal of Asia-Pacific Entomology*, *12*(4), 253-259. http://dx.doi.org/10.1016/j.aspen.2009.08.003.

Were, K., Gelaw, A. M., & Singh, B. R. (2016). Smart strategies for enhanced agricultural resilience and food security under a changing climate in sub-Saharan Africa. In R. Lal, D. Kraybill, D. O. Hansen, B. R. Singh, T. Mosogoya, L. O. Eik (eds.), *Climate Change and Multi-Dimensional Sustainability in African Agriculture*. Springer International Publishing AG 2016. http://dx.doi.org/10.1007/978-3-319-41238-2_23

Whalon, M., Mota-Sanchez, D., & Hollingworth, R. (2008). *Analysis of global pesticide resistance in arthropods*. CABI Publishing. Wallingford, UK. http://dx.doi.org/10.1079/9781845933531.0005

WHO (2005). *Guidelines for laboratory and field testing of Mosquito larvicides. World Health Organization*. Geneva, Switzerland. https://apps.who.int/iris/handle/10665/69101

Williamson, S., Ball, A., & Pretty, J. (2008). Trends in pesticide use and drivers for safer pest management in four African countries. *Crop Protection*, *27*(10), 1327-1334. http://dx.doi.org/10.1016/j.cropro.2008.04.006.

Xu, W. H., Liu, W. Y., & Liang, Q. (2018). Chemical constituents from *Croton species* and their biological activities. *Molecules*, *23*(9), 1-38. http://dx.doi.org/10.3390/molecules 23092333

Yamada, H. (1981). Seasonal life history of cabbage webworm, *Hellula undalis* Fabricius. *Bulletin of the Vegetable and Ornamental Crops Research Station, Japan, A*, *8*(1981), 131-141.

Yang, Y., Isman, M. B., & Tak, J. H. (2020). Insecticidal activity of 28 essential oils and a commercial product containing *Cinnamomum cassia* bark essential oil against *Sitophilus zeamais* Motschulsky. *Insects*, *11*(8), 1-18. http://dx.doi.org/10.3390/insects 11080474

Zafari-Shayan, S., Moradkhani, S., & Dastan, D. (2016). Analysis of fatty acid composition of two selected *Phlomis species*. *Journal of HerbMed Pharmacology*, *5*(4), 153-156.

Zaker, M., & Mosallanejad, H. (2010). Antifungal activity of some plant extracts on *Alternaria alternata*, the causal agent of alternaria leaf spot of potato. *Pakistan Journal of Biological Sciences*, *13*(21), 1023-1029. http://dx.doi.org/10.3923/pjbs.2010.1023.1029

Zaki, F. (2008). Field application of plant extracts against the aphid, *B. brassicae* and the whitefly, *B. tabaci* and their side effects on their predators and parasites. *Archives of Phytopathology and Plant Protection*, *41*(6), 462-466. https://doi.org/10.1080/03235400600881679

Zhang, F., Takahata, Y., & Xu, J. (2002). Plant regeneration from cotyledonary explants of Chinese cabbage cultured in vitro. *Acta Horticulturae Sinica*, *29*(4), 348-352. https://doi.org/10.3923/ajps.2013.262.270

APPENDICES

Appendix 1: Mean population of *B. brassicae* per crop in response to weekly application of treatments for 2019 and 2020 wet seasons

Location and Treatments	Week1 before Treatment	Weeks after treatments -2019 wet season					Week 1 before Treatment	Weeks after treatments -2020 season				
		1	2	3	4	5		1	2	3	4	5
Location												
Tengeru	0.41 ± 0.03a	0.58 ± 0.06a	0.44 ± 0.07a	0.54 ± 0.08a	0.58 ± 0.10a	0.68 ± 0.11a	1.31 ± 0.08a	0.51 ± 0.06a	0.52 ± 0.07b	0.78 ± 0.11a	0.71 ± 0.11a	0.78 ± 0.13a
Boro	0.40 ± 0.04a	0.33. ± 0.04b	0.33 ± 0.04a	0.36 ± 0.05b	0.38 ± 0.05b	0.35 ± 0.06b	1.00 ± 0.05b	0.54. ± 0.06a	0.73 ± 0.09a	0.86 ± 0.13a	0.76 ± 0.03a	0.72 ± 0.13a
Treatments												
Water	0.83 ± 0.16a	1.05 ± 0.14a	1.05 ± 0.24a	1.23 ± 0.22a	1.58 ± 0.39a	1.98 ± 0.39a	1.50 ± 0.16a	1.45 ± 0.15a	1.98 ± 0.20a	2.68 ± 0.35a	2.68 ± 0.16a	3.00 ± 0.22a
water + soap	0.73 ± 0.18a	1.13 ± 0.25a	0.90 ± 0.29a	1.20 ± 0.36a	1.30 ± 0.24a	1.58 ± 0.09a	1.28 ± 0.19a	1.25 ± 0.14a	1.63 ± 0.10b	2.60 ± 0.19a	2.65 ± 0.17a	3.01 ± 0.21a
Synthetic pesticide	**0.53 ± 0.15a**	**0.23 ± 0.06defg**	**0.05 ± 0.03d**	**0.08 ± 0.04e**	**0.08 ± 0.03e**	**0.03 ± 0.03e**	**1.25 ± 0.27a**	**0.23 ± 0.08de**	**0.15 ± 0.05f**	**0.15 ± 0.03ef**	**0.13 ± 0.04f**	**0.08 ± 0.03f**
C. dichogamus (1%)	0.38 ± 0.08a	0.68 ± 0.13b	0.48 ± 0.12bc	0.55 ± 0.11bcd	0.68 ± 0.08b	0.65 ± 0.14b	0.65 ± 0.15a	0.63 ± 0.10b	0.78 ± 0.11c	0.73 ± 0.10cd	0.53 ± 0.06bcd	0.65 ± 0.11bc
C. dichogamus (5%)	0.33 ± 0.06a	0.45 ± 0.06bce	0.38 ± 0.05bcd	0.33 ± 0.05bcde	0.38 ± 0.08bcde	0.45 ± 0.11bcde	1.15 ± 0.24a	0.45 ± 0.05bcd	0.53 ± 0.09cde	0.65 ± 0.06cd	0.50 ± 0.07cde	0.38 ± 0.08cde
C. dichogamus (10%)	0.28 ± 0.08a	0.20 ± 0.05defg	0.18 ± 0.06cd	0.20 ± 0.08de	0.20 ± 0.05cde	0.15 ± 0.14de	0.95 ± 0.12a	0.20 ± 0.05de	0.35 ± 0.08ef	0.25 ± 0.05ef	0.35 ± 0.06def	0.25 ± 0.05def
S. aromaticum (1%)	0.43 ± 0.10a	0.63 ± 0.09bc	0.35 ± 0.09bcd	0.68 ± 0.11b	0.60 ± 0.09bc	0.55 ± 0.09bcd	1.45 ± 0.25a	0.65 ± 0.12b	0.70 ± 0.12cd	1.15 ± 0.18b	0.83 ± 0.10b	0.73 ± 0.13b
S. aromaticum (5%)	0.20 ± 0.08a	0.48 ± 0.12bcd	0.33 ± 0.08bcd	0.30 ± 0.07cde	0.37 ± 0.04bcde	0.35 ± 0.09bcde	1.13 ± 0.14a	0.45 ± 0.07bcd	0.55 ± 0.07cde	0.70 ± 0.14cd	0.48 ± 0.10cde	0.38 ± 0.05cde
S. aromaticum (10%)	0.20 ± 0.08a	0.15 ± 0.05fg	0.23 ± 0.11bcd	0.23 ± 0.08de	0.18 ± 0.08de	0.20 ± 0.08cde	1.05 ± 0.15a	0.40 ± 0.07bcde	0.30 ± 0.08ef	0.35 ± 0.08def	0.25 ± 0.05ef	0.20 ± 0.05def
T. vogelii (1%)	0.50 ± 0.14a	0.45 ± 0.06bcde	0.53 ± 0.08b	0.65 ± 0.09bc	0.50 ± 0.14bcd	0.63 ± 0.09bc	1.19 ± 0.28a	0.60 ± 0.10bc	0.68 ± 0.13cd	0.95 ± 0.12bc	0.65 ± 0.09bc	0.73 ± 0.09b
T. vogelii (5%)	0.43 ± 0.13a	0.38 ± 0.03cdef	0.33 ± 0.04bcd	0.33 ± 0.05bcde	0.30 ± 0.08bcde	0.25 ± 0.08bcde	0.93 ± 0.13a	0.35 ± 0.05cde	0.43 ± 0.12def	0.48 ± 0.06de	0.58 ± 0.09bcd	0.45 ± 0.07bcd
T. vogelii (10%)	0.28 ± 0.08a	0.18 ± 0.06efg	0.18 ± 0.05cd	0.18 ± 0.04e	0.15 ± 0.05de	0.13 ± 0.04de	1.18 ± 0.10a	0.28 ± 0.05de	0.33 ± 0.08ef	0.20 ± 0.04ef	0.25 ± 0.05ef	0.23 ± 0.04def
Mixed plants (2.5%)	0.30 ± 0.07a	0.30 ± 0.08defg	0.28 ± 0.05bcd	0.25 ± 0.06de	0.25 ± 0.08cde	0.18 ± 0.06de	1.18 ± 0.18a	0.30 ± 0.09de	0.23 ± 0.06f	0.53 ± 0.11de	0.35 ± 0.06cef	0.33 ± 0.04def
Mixed plants (5%)	**0.28 ± 0.08a**	**0.05 ± 0.03g**	**0.13 ± 0.05d**	**0.12 ± 0.03e**	**0.05 ± 0.03e**	**0.08 ± 0.04e**	**1.33 ± 0.21a**	**0.15 ± 0.06e**	**0.15 ± 0.05f**	**0.08 ± 0.04f**	**0.15 ± 0.05f**	**0.10 ± 0.05ef**
2 - way ANOVA	**(F- Statistics)**											
Locations	0.04 ns	29.14***	2.72ns	5.92*	6.78**	23.11***	10.43**	0.23ns	19.08***	1.59ns	1.32ns	0.82ns
Treatments	2.91ns	13.59***	5.46***	7.93***	10.66***	20.78***	1.41ns	15.58***	34.63***	48.20***	81.28***	78.04***
Location*treatments	2.81ns	1.69ns	0.64ns	0.80ns	1.03ns	2.94***	0.82ns	0.99ns	1.66ns	3.10***	0.38ns	0.23ns

Each value is a mean ± standard error of eight replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fishers Least significant Difference (LSD) test.

Appendix 2: Mean number population of *M. persicae* per plant in response to weekly application of treatments

Location and Treatments	Week 1 before Treatment	Weeks after treatments -2019 wet season					Week 1 before Treatment	Weeks after treatments -2020 wet season				
		1	2	3	4	5		1	2	3	4	5
Location												
Tengeru	1.04 ± 0.07a	0.74 ± 0.09a	0.64 ± 0.11a	0.54 ± 0.11a	0.54 ± 0.10a	0.59 ± 0.11a	1.26 ± 0.07a	0.51 ± 0.05a	0.46 ± 0.06b	0.63 ± 0.09a	0.88 ± 0.10a	0.90 ± 0.11a
Boro	0.64 ± 0.06b	0.57 ± 0.08b	0.47 ± 0.06b	0.43 ± 0.08b	0.51 ± 0.08a	0.55 ± 0.15a	0.84 ± 0.05a	0.53 ± 0.06a	0.63 ± 0.08a	0.71 ± 0.12a	0.64 ± 0.10b	0.63 ± 0.11b
Treatments												
Water	1.28 ± 0.15a	2.05 ± 0.27a	2.03 ± 0.34a	2.03 ± 0.16a	2.45 ± 0.42a	2.78 ± 0.35a	1.03 ± 0.11a	1.38 ± 0.12a	1.70 ± 0.18a	2.28 ± 0.28a	2.43 ± 0.14a	2.58 ± 0.12a
W + s	0.80 ± 0.19a	1.80 ± 0.27a	1.38 ± 0.26b	1.73 ± 0.34ab	2.03 ± 0.26a	2.70 ± 0.25a	0.98 ± 0.14a	1.28 ± 0.13a	1.48 ± 0.12b	2.25 ± 0.29ab	2.40 ± 0.18a	2.83 ± 0.13a
S. p	1.00 ± 0.24a	0.20 ± 0.07d	0.30 ± 0.16de	0.25 ± 0.13cd	0.28 ± 0.13bc	0.20 ± 0.08bc	1.15 ± 0.20a	0.08 ± 0.05f	0.05 ± 0.03h	0.10 ± 0.04ef	0.15 ± 0.06g	0.15 ± 0.06e
C. d (1%)	1.08 ± 0.22a	0.90 ± 0.11b	0.78 ± 0.24c	0.53 ± 0.13c	0.55 ± 0.12b	0.45 ± 0.07b	1.18 ± 0.21a	0.53 ± 0.05bcd	0.50 ± 0.04bcd	0.55 ± 0.06bc	0.65 ± 0.07bcd	0.63 ± 0.08bc
C. d (5%)	0.58 ± 0.13a	0.45 ± 0.03cd	0.45 ± 0.03cde	0.28 ± 0.08cd	0.23 ± 0.09bc	0.33 ± 0.05bc	1.08 ± 0.18a	0.45 ± 0.07bcde	0.50 ± 0.11bcd	0.50 ± 0.05bcd	0.55 ± 0.07cde	0.40 ± 0.08cde
C. d (10%)	0.85 ± 0.21a	0.63 ± 0.13bc	0.53 ± 0.14cd	0.18 ± 0.10cd	0.33 ± 0.11bc	0.30 ± 0.08bc	0.93 ± 0.14a	0.30 ± 0.04defg	0.28 ± 0.04defgh	0.23 ± 0.05cdef	0.43 ± 0.11defg	0.40 ± 0.12cde
S. a (1%)	0.78 ± 0.08a	0.55 ± 0.11bcd	0.43 ± 0.06cde	0.45 ± 0.07cd	0.33 ± 0.13bc	0.45 ± 0.07b	1.18 ± 0.21a	0.65 ± 0.11b	0.70 ± 0.11b	0.78 ± 0.09b	0.93 ± 0.15b	0.88 ± 0.13b
S. a (5%)	0.73 ± 0.08a	0.43 ± 0.07cd	0.43 ± 0.10cde	0.28 ± 0.08cd	0.23 ± 0.06bc	0.23 ± 0.07bc	0.90 ± 0.20a	0.48 ± 0.09bcde	0.48 ± 0.11bcde	0.50 ± 0.08bcd	0.70 ± 0.15bcd	0.40 ± 0.05cde
S. a (10%)	0.90 ± 0.18a	0.38 ± 0.06cd	0.28 ± 0.08de	0.20 ± 0.07cd	0.10 ± 0.05c	0.18 ± 0.05bc	1.00 ± 0.15a	0.35 ± 0.05cdef	0.35 ± 0.08defg	0.35 ± 0.07cdef	0.30 ± 0.07efg	0.30 ± 0.07de
T. v (1%)	0.73 ± 0.25a	0.43 ± 0.10cd	0.50 ± 0.05cd	0.30 ± 0.06cd	0.20 ± 0.05bc	0.18 ± 0.03bc	1.03 ± 0.11a	0.58 ± 0.10bc	0.65 ± 0.11bc	0.80 ± 0.10b	0.78 ± 0.07bc	0.78 ± 0.14b
T. v (5%)	0.80 ± 0.20a	0.40 ± 0.11cd	0.28 ± 0.06de	0.20 ± 0.07cd	0.13 ± 0.05bc	0.05 ± 0.03c	1.05 ± 0.26a	0.43 ± 0.10bcdef	0.43 ± 0.10cdef	0.45 ± 0.10bcde	0.45 ± 0.07def	0.43 ± 0.03cd
T. v (10%)	0.98 ± 0.23a	0.30 ± 0.07cd	0.13 ± 0.04de	0.18 ± 0.05cd	0.08 ± 0.04c	0.05 ± 0.03c	1.28 ± 0.22a	0.28 ± 0.06efg	0.18 ± 0.03fgh	0.15 ± 0.03def	0.25 ± 0.06fg	0.30 ± 0.05de
M. p. (2.5%)	0.93 ± 0.20a	0.48 ± 0.12cd	0.18 ± 0.05de	0.23 ± 0.10cd	0.08 ± 0.04c	0.05 ± 0.05c	0.83 ± 0.16a	0.28 ± 0.06efg	0.23 ± 0.04efgh	0.38 ± 0.10cdef	0.45 ± 0.09def	0.45 ± 0.11cd
M. p. (5%)	0.73 ± 0.10a	0.20 ± 0.05d	0.08 ± 0.04e	0.16 ± 0.05d	0.10 ± 0.10c	0.05 ± 0.05c	1.15 ± 0.15a	0.20 ± 0.07fg	0.13 ± 0.04gh	0.08 ± 0.04f	0.18 ± 0.06fg	0.20 ± 0.05de
2 - way ANOVA	**(F- Statistics)**											
Locations (L)	10.43**	5.51*	4.96*	2.55*	0.553ns	0.53ns	22.93**	0.16ns	15.10***	2.22ns	21.69***	35.41***
Treatments (T)	1.41ns	18.02***	14.35***	19.47***	23.12***	60.17***	0.56ns	19.88***	36.67***	40.21***	58.60***	103.56***
L * T	0.82ns	0.46ns	1.70ns	0.63ns	0.73ns	1.77ns	0.55ns	1.02ns	1.71ns	2.99**	0.96ns	0.58ns

Each value is a mean ± standard error of eight replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fishers Least significant Difference (LSD) test.

Appendix 3: Mean population of *P. xylostella* per *B. oleracea* in response to weekly application of treatments for 2019 and 2020 seasons

Location and Treatments	Week 1 before Treatment	Weeks after treatments -2019 wet season					Week 1 before Treatment	Weeks after treatments -2020 wet season				
		1	2	3	4	5		1	2	3	4	5
Location												
Tengeru	0.08 ± 0.02a	0.09 ± 0.02b	0.20 ± 0.07b	0.26 ± 0.09b	0.20 ± 0.07b	0.39 ± 0.09b	0.38 ± 0.04b	0.38 ± 0.05a	0.31 ± 0.03b	0.43 ± 0.06b	0.58 ± 0.08a	0.68 ± 0.11a
Boro	0.07 ± 0.02a	0.38 ± 0.09a	0.50 ± 0.10a	0.78 ± 0.14a	0.63 ± 0.13a	0.83 ± 0.21a	0.52 ± 0.03a	0.36 ± 0.04a	0.47 ± 0.06a	0.55 ± 0.07a	0.54 ± 0.08a	0.50 ± 0.08b
Treatments												
Water	0.13 ± 0.05a	1.13 ± 0.53a	1.68 ± 0.41a	2.28 ± 0.44a	2.20 ± 0.53a	3.28 ± 0.60a	0.48 ± 0.07a	0.93 ± 0.16a	1.15 ± 0.14a	1.55 ± 0.21a	1.93 ± 0.28a	1.94 ± 0.31a
W + s	0.10 ± 0.04a	0.75 ± 0.15a	1.60 ± 0.31a	2.58 ± 0.36a	1.83 ± 0.35a	3.30 ± 0.49a	0.45 ± 0.18a	1.05 ± 0.13a	0.93 ± 0.13b	1.50 ± 0.16a	1.63 ± 0.20a	2.18 ± 0.32a
S. p	0.00 ± 0.00a	0.05 ± 0.10b	0.03 ± 0.03b	0.28 ± 0.15b	0.08 ± 0.04b	0.08 ± 0.04b	0.47 ± 0.12a	0.15 ± 0.03c	0.13 ± 0.07e	0.15 ± 0.05ef	0.15 ± 0.05ef	0.18 ± 0.08d
C. d (1%)	0.13 ± 0.05a	0.28 ± 0.12b	0.25 ± 0.09b	0.30 ± 0.10b	0.23 ± 0.10b	0.33 ± 0.08b	0.40 ± 0.09a	0.43 ± 0.11b	0.48 ± 0.05c	0.53 ± 0.07bc	0.60 ± 0.11b	0.58 ± 0.10bc
C. d (5%)	0.08 ± 0.04a	0.15 ± 0.10b	0.33 ± 0.06b	0.20 ± 0.08b	0.23 ± 0.10b	0.20 ± 0.07b	0.47 ± 0.08a	0.30 ± 0.05bc	0.38 ± 0.07cd	0.38 ± 0.08bcde	0.45 ± 0.03bcde	0.30 ± 0.08bcd
C. d (10%)	0.03 ± 0.03a	0.08 ± 0.04b	0.20 ± 0.08b	0.15 ± 0.05b	0.13 ± 0.08b	0.18 ± 0.06b	0.33 ± 0.08a	0.25 ± 0.06bc	0.15 ± 0.05e	0.25 ± 0.03def	0.23 ± 0.03cdef	0.32 ± 0.06bcd
S. a (1%)	0.18 ± 0.05a	0.13 ± 0.05b	0.18 ± 0.10b	0.35 ± 0.12b	0.23 ± 0.09b	0.28 ± 0.04b	0.55 ± 0.12a	0.38 ± 0.08bc	0.48 ± 0.09c	0.58 ± 0.08b	0.53 ± 0.05bc	0.65 ± 0.06b
S. a (5%)	0.03 ± 0.03a	0.15 ± 0.07b	0.10 ± 0.05b	0.23 ± 0.09b	0.23 ± 0.08b	0.18 ± 0.06b	0.43 ± 0.10a	0.30 ± 0.08bc	0.33 ± 0.07cde	0.36 ± 0.06cdef	0.48 ± 0.13bcd	0.35 ± 0.05bcd
S. a (10%)	0.03 ± 0.03a	0.03 ± 0.03b	0.05 ± 0.05b	0.15 ± 0.08b	0.18 ± 0.06b	0.13 ± 0.04b	0.53 ± 0.07a	0.23 ± 0.06bc	0.25 ± 0.03de	0.20 ± 0.04ef	0.20 ± 0.03def	0.28 ± 0.05cd
T. v (1%)	0.00 ± 0.00a	0.10 ± 0.05b	0.15 ± 0.05b	0.18 ± 0.07b	0.20 ± 0.07b	0.25 ± 0.07b	0.30 ± 0.08a	0.30 ± 0.04bc	0.30 ± 0.05cde	0.48 ± 0.05bcd	0.58 ± 0.08b	0.50 ± 0.09bcd
T. v (5%)	0.08 ± 0.05a	0.08 ± 0.05b	0.18 ± 0.10b	0.20 ± 0.08b	0.13 ± 0.04b	0.15 ± 0.03b	0.43 ± 0.12a	0.23 ± 0.05bc	0.30 ± 0.04cde	0.35 ± 0.05cdef	0.45 ± 0.08bcde	0.35 ± 0.05bcd
T. v (10%)	0.05 ± 0.05a	0.08 ± 0.04b	0.15 ± 0.05b	0.13 ± 0.08b	0.05 ± 0.03b	0.08 ± 0.04b	0.50 ± 0.09a	0.20 ± 0.04bc	0.20 ± 0.04de	0.18 ± 0.06ef	0.18 ± 0.08def	0.20 ± 0.04d
M. p. (2.5%)	0.18 ± 0.05a	0.15 ± 0.04b	0.10 ± 0.05b	0.18 ± 0.07b	0.05 ± 0.03b	0.10 ± 0.04b	0.43 ± 0.08a	0.30 ± 0.07bc	0.28 ± 0.07cde	0.28 ± 0.06cdef	0.33 ± 0.07bcdef	0.35 ± 0.06bcd
M. p. (5%)	0.08 ± 0.04a	0.08 ± 0.04b	0.08 ± 0.04b	0.08 ± 0.04b	0.03 ± 0.03b	0.05 ± 0.03b	0.55 ± 0.11a	0.15 ± 0.05c	0.13 ± 0.04e	0.10 ± 0.04f	0.10 ± 0.04f	0.15 ± 0.03d
2 - way ANOVA	**(F- Statistics)**											
Locations (L)	0.42ns	17.04***	21.74***	66.47***	39.78***	58.69***	6.00*	0.15ns	29.66***	7.65**	0.46ns	6.04*
Treatments (T)	2.04ns	5.42***	20.50***	46.20***	29.15***	110.49***	0.52ns	10.35***	28.16***	28.08***	21.39***	21.85***
L * T	1.95ns	2.83ns	3.00**	3.83***	5.87***	17.68***	0.90ns	0.29ns	3.64ns	1.08ns	0.37ns	0.27ns

Each value is a mean ± standard error of eight replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fishers Least significant Difference (LSD) test.

Appendix 4: Mean population of *H. undalis* larvae per *B. oleracea* crop in response to weekly application of treatments for 2019 and 2020 seasons

Location and Treatments	Week 1 before Treatment	Weeks after treatments 2019 wet season					Week 1 before Treatment	Weeks after treatments 2020 wet season				
		1	2	3	4	5		1	2	3	4	5
Location												
Tengeru	0.20 ± 0.02b	0.35 ± 0.08a	0.38 ± 0.07a	0.41 ± 0.10a	0.41 ± 0.08a	0.38 ± 0.07a	0.13 ± 0.02b	0.29 ± 0.04a	0.22 ± 0.03a	0.23 ± 0.03a	0.28 ± 0.03a	0.32 ± 0.04a
Boro	0.28 ± 0.03a	0.25 ± 0.04a	0.29 ± 0.04a	0.34 ± 0.05a	0.40 ± 0.05a	0.48 ± 0.06a	0.25 ± 0.02a	0.18 ± 0.02b	0.26 ± 0.02a	0.26 ± 0.03a	0.22 ± 0.03a	0.22 ± 0.03b
Treatments												
Water	0.28 ± 0.08a	1.03 ± 0.37a	0.93 ± 0.23a	1.10 ± 0.33a	1.28 ± 0.32a	1.43 ± 0.33a	0.20 ± 0.07a	0.63 ± 0.08a	0.60 ± 0.05a	0.58 ± 0.08a	0.62 ± 0.05a	0.65 ± 0.12a
W + s	0.28 ± 0.08a	0.85 ± 0.17a	1.25 ± 0.25a	1.30 ± 0.42a	1.25 ± 0.15a	1.25 ± 0.11a	0.23 ± 0.07a	0.63 ± 0.10a	0.55 ± 0.08a	0.60 ± 0.05a	0.65 ± 0.10a	0.60 ± 0.09a
S. p	0.13 ± 0.06a	0.00 ± 0.00c	0.00 ± 0.00c	0.05 ± 0.05c	0.05 ± 0.03d	0.03 ± 0.03e	0.13 ± 0.04a	0.00 ± 0.00e	0.05 ± 0.03f	0.05 ± 0.03e	0.10 ± 0.04de	0.10 ± 0.04e
C. d (1%)	0.28 ± 0.11a	0.38 ± 0.08b	0.43 ± 0.11b	0.40 ± 0.10bc	0.43 ± 0.10bc	0.40 ± 0.10bc	0.13 ± 0.05a	0.25 ± 0.10bc	0.30 ± 0.04b	0.33 ± 0.04b	0.30 ± 0.10bc	0.35 ± 0.09bc
C. d (5%)	0.25 ± 0.03a	0.23 ± 0.08bc	0.28 ± 0.06bc	0.29 ± 0.11bc	0.28 ± 0.10cd	0.30 ± 0.05cde	0.28 ± 0.08a	0.20 ± 0.05bcd	0.20 ± 0.04bcde	0.23 ± 0.07bc	0.20 ± 0.05cde	0.23 ± 0.06cde
C. d (10%)	0.28 ± 0.06a	0.13 ± 0.05bc	0.18 ± 0.05bc	0.17 ± 0.07bc	0.18 ± 0.05cd	0.20 ± 0.04cde	0.18 ± 0.05a	0.10 ± 0.04cde	0.15 ± 0.06cdef	0.13 ± 0.05de	0.15 ± 0.05cde	0.15 ± 0.06cde
S. a (1%)	0.23 ± 0.06a	0.30 ± 0.05bc	0.43 ± 0.11b	0.28 ± 0.10bc	0.35 ± 0.07bcd	0.45 ± 0.07bc	0.25 ± 0.06a	0.35 ± 0.09b	0.28 ± 0.06bc	0.33 ± 0.05b	0.38 ± 0.05b	0.45 ± 0.07ab
S. a (5%)	0.20 ± 0.05a	0.18 ± 0.06bc	0.25 ± 0.06bc	0.23 ± 0.11bc	0.25 ± 0.07cd	0.33 ± 0.08cde	0.23 ± 0.08a	0.25 ± 0.05bc	0.20 ± 0.04bcde	0.15 ± 0.05cde	0.18 ± 0.06cde	0.20 ± 0.05cde
S. a (10%)	0.30 ± 0.08a	0.18 ± 0.08bc	0.15 ± 0.05bc	0.15 ± 0.06bc	0.15 ± 0.05cd	0.18 ± 0.07de	0.25 ± 0.05a	0.15 ± 0.05cde	0.15 ± 0.05cdef	0.13 ± 0.04de	0.13 ± 0.04de	0.15 ± 0.05de
T. v (1%)	0.23 ± 0.06a	0.33 ± 0.14bc	0.45 ± 0.014b	0.58 ± 0.13b	0.60 ± 0.06b	0.50 ± 0.05b	0.08 ± 0.04a	0.25 ± 0.10bc	0.25 ± 0.06bcd	0.25 ± 0.03bc	0.25 ± 0.05bcd	0.33 ± 0.08bcd
T. v (5%)	0.23 ± 0.05a	0.20 ± 0.08bc	0.25 ± 0.06bc	0.40 ± 0.10bc	0.33 ± 0.05bcd	0.38 ± 0.08bcd	0.18 ± 0.06a	0.15 ± 0.05cde	0.25 ± 0.06bcd	0.28 ± 0.04bc	0.18 ± 0.06cde	0.20 ± 0.07cde
T. v (10%)	0.22 ± 0.02a	0.13 ± 0.04bc	0.15 ± 0.05bc	0.15 ± 0.06bc	0.23 ± 0.07cd	0.18 ± 0.06de	0.25 ± 0.06a	0.13 ± 0.05cde	0.13 ± 0.05def	0.08 ± 0.04de	0.15 ± 0.05cde	0.13 ± 0.04de
M. p. (2.5%)	0.23 ± 0.07a	0.23 ± 0.07bc	0.23 ± 0.08bc	0.20 ± 0.08bc	0.23 ± 0.06cd	0.30 ± 0.09cde	0.13 ± 0.05a	0.18 ± 0.05bcde	0.20 ± 0.05bcde	0.23 ± 0.05bc	0.18 ± 0.05cde	0.20 ± 0.05cde
M. p. (5%)	0.28 ± 0.08a	0.08 ± 0.05bc	0.03 ± 0.03c	0.03 ± 0.03c	0.08 ± 0.04d	0.08 ± 0.04e	0.18 ± 0.06a	0.05 ± 0.03de	0.08 ± 0.04ef	0.05 ± 0.03e	0.08 ± 0.04e	0.05 ± 0.03e
2 - way ANOVA	**(F- Statistics)**											
Location (L)	3.57*	2.31ns	1.65ns	0.59ns	0.00ns	2.36ns	14.49***	10.98***	1.47ns	1.47ns	3.31ns	8.22**
Treatments (T)	0.36ns	4.82***	6.21***	4.73***	10.91***	12.21***	1.15ns	9.61***	8.32***	12.28***	9.08***	7.37***
L*T	0.17ns	0.62ns	0.43ns	0.59ns	0.42ns	0.14ns	0.74ns	0.78ns	0.27ns	0.61ns	0.80ns	0.64ns

Each value is a mean ± standard error of eight replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fishers Least significant Difference (LSD) test.

Appendix 5: Mean abundance of *T. ni* per *B. oleracea* plant in response to weekly application of the treatments for 2019 and 2020

Location and Treatments	Week 1 before Treatment	Weeks after treatments 2019 wet season					Week 1 before Treatment	Weeks after treatments 2020 wet season				
		1	2	3	4	5		1	2	3	4	5
Location												
Tengeru	0.18 ± 0.02a	0.18 ± 0.03a	0.23 ± 0.0a	0.22 ± 0.07a	0.17 ± 0.05a	0.20 ± 0.05a	0.17 ± 0.01a	0.20 ± 0.02a	0.18 ± 0.02b	0.25 ± 0.03a	0.25 ± 0.03a	0.28 ± 0.04a
Boro	0.09 ± 0.02b	0.06 ± 0.03b	0.06 ± 0.02b	0.10 ± 0.02b	0.15 ± 0.04a	0.14 ± 0.04a	0.19 ± 0.02a	0.19 ± 0.02a	0.24 ± 0.03a	0.23 ± 0.03a	0.22 ± 0.03a	0.19 ± 0.02b
Treatments												
Water	0.18 ± 0.07a	0.50 ± 0.09a	0.63 ± 0.30a	0.60 ± 0.24a	0.48 ± 0.15a	0.78 ± 0.16a	0.25 ± 0.09a	0.43 ± 0.10a	0.53 ± 0.04a	0.65 ± 0.03a	0.55 ± 0.07a	0.55 ± 0.07a
W + s	0.10 ± 0.05a	0.30 ± 0.08b	0.48 ± 0.20a	0.68 ± 0.26a	0.65 ± 0.20a	0.80 ± 0.18a	0.23 ± 0.05a	0.40 ± 0.05a	0.43 ± 0.08ab	0.63 ± 0.10a	0.58 ± 0.06a	0.63 ± 0.14a
S. p	0.13 ± 0.05a	0.05 ± 0.03c	0.03 ± 0.03b	0.03 ± 0.03b	0.03 ± 0.03bc	0.00 ± 0.00b	0.13 ± 0.04a	0.03 ± 0.03e	0.00 ± 0.00h	0.03 ± 0.03fg	0.00 ± 0.00e	0.08 ± 0.04de
C. d (1%)	0.15 ± 0.05a	0.08 ± 0.04c	0.05 ± 0.03b	0.13 ± 0.05b	0.23 ± 0.10b	0.08 ± 0.04b	0.15 ± 0.03a	0.23 ± 0.03cd	0.20 ± 0.05cdef	0.30 ± 0.05b	0.25 ± 0.06bc	0.23 ± 0.06bcd
C. d (5%)	0.08 ± 0.04a	0.10 ± 0.04c	0.10 ± 0.05b	0.05 ± 0.03b	0.13 ± 0.04bc	0.08 ± 0.04b	0.15 ± 0.03a	0.18 ± 0.05cd	0.20 ± 0.04cdef	0.28 ± 0.04bc	0.18 ± 0.06bcd	0.23 ± 0.05bcd
C. d (10%)	0.15 ± 0.05a	0.03 ± 0.03c	0.08 ± 0.04b	0.05 ± 0.03b	0.08 ± 0.05bc	0.08 ± 0.04b	0.18 ± 0.03a	0.15 ± 0.03cde	0.13 ± 0.04efgh	0.15 ± 0.05cdef	0.13 ± 0.04cde	0.10± 0.04cde
S. a (1%)	0.10 ± 0.04a	0.13 ± 0.05c	0.13 ± 0.08b	0.05 ± 0.03b	0.08 ± 0.04bc	0.08 ± 0.04b	0.13 ± 0.04a	0.28 ± 0.05bc	0.28 ± 0.08cd	0.33 ± 0.05b	0.30 ± 0.09b	0.30 ± 0.08b
S. a (5%)	0.18 ± 0.03a	0.05 ± 0.03c	0.05 ± 0.03b	0.05 ± 0.03b	0.00 ± 0.00c	0.03 ± 0.03b	0.18 ± 0.06a	0.20 ± 0.05cd	0.25 ± 0.05cde	0.15 ± 0.06cdef	0.23 ± 0.05bc	0.23 ± 0.06bcd
S. a (10%)	0.10 ± 0.01a	0.05 ± 0.03c	0.03 ± 0.03b	0.00 ± 0.00b	0.05 ± 0.05bc	0.03 ± 0.03b	0.23 ± 0.03a	0.15 ± 0.03cde	015 ± 0.03defg	0.10 ± 0.04efg	0.20 ± 0.04bc	0.10 ± 0.04cde
T. v (1%)	0.13 ± 0.05a	0.10 ± 0.04c	0.10 ± 0.05b	0.18 ± 0.06b	0.15 ± 0.03bc	0.10 ± 0.04b	0.23 ± 0.03a	0.28 ± 0.05bc	0.30 ± 0.05bc	0.30 ± 0.04b	0.33 ± 0.06b	0.28 ± 0.05bc
T. v (5%)	0.20 ± 0.04a	0.05 ± 0.03c	0.05 ± 0.03b	0.08 ± 0.04b	0.03 ± 0.03bc	0.00 ± 0.00b	0.15 ± 0.05a	0.15 ± 0.03cde	0.18 ± 0.05defg	0.25 ± 0.06bcd	0.18 ± 0.06bcd	0.28 ± 0.06bc
T. v (10%)	0.13 ± 0.04a	0.00 ± 0.00c	0.03 ± 0.03b	0.05 ± 0.03b	0.03 ± 0.03bc	0.00 ± 0.00b	0.18 ± 0.03a	0.13 ± 0.04de	0.10 ± 0.04fgh	0.13 ± 0.04defg	0.13 ± 0.04cde	0.13 ± 0.05cde
M. p. (2.5%)	0.10 ± 0.04a	0.05 ± 0.03c	0.03 ± 0.02b	0.03 ± 0.03b	0.00 ± 0.00bc	0.00 ± 0.00b	0.15 ± 0.03a	0.15 ± 0.06cde	0.15 ± 0.03defg	0.20 ± 0.05bcde	0.20 ± 0.05bc	0.23 ± 0.05bcd
M. p. (5%)	0.15 ± 0.05a	0.00 ± 0.00c	0.05 ± 0.03b	0.00 ± 0.00b	0.03 ± 0.03bc	0.00 ± 0.00b	0.18 ± 0.06a	0.03 ± 0.03e	0.05 ± 0.03gh	0.00 ± 0.00g	0.03 ± 0.03de	0.03 ± 0.03e
2 - way ANOVA	**(F- Statistics)**											
Location	13.89***	20.28***	10.01**	4.96*	0.17ns	2.21ns	0.71ns	0.00ns	4.53*	0.02ns	0.87ns	8.38**
Treatments	0.70ns	10.44***	3.91***	5.79***	5.75***	16.63***	1.01ns	5.34***	8.92***	13.28***	8.47***	7.47***
Location*treatments	0.34ns	1.14ns	1.95*	2.46**	0.38ns	1.18ns	2.68ns	0.37ns	0.71ns	0.56ns	0.40ns	0.70ns

Each value is a mean ± standard error of eight replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fishers Least significant Difference (LSD) test.

Appendix 6: Mean population abundance of *C. binotalis* per *B. oleracea* crop in response to weekly application of treatments for 2019 and 2020

Location and Treatments	Week 1 before Treatment	Weeks after treatments 2019 wet season					Week 1 before Treatment	Weeks after treatments 2020 wet season				
		1	2	3	4	5		1	2	3	4	5
Location												
Tengeru	0.33 ± 0.02a	0.38 ± 0.09a	0.46 ± 0.11a	0.41 ± 0.12a	0.40 ± 0.11a	0.43 ± 0.12a	0.19 ± 0.02b	0.20 ± 0.03b	0.19 ± 0.02b	0.23 ± 0.03b	0.37 ± 0.03b	0.65 ± 0.13a
Boro	0.24 ± 0.03b	0.15 ± 0.05b	0.17 ± 0.05b	0.31 ± 0.08a	0.28 ± 0.05b	0.29 ± 0.07b	0.43 ± 0.03a	0.29 ± 0.05a	0.49 ± 0.08a	0.73 ± 0.15a	0.66 ± 0.15a	0.53 ± 0.12a
Treatments												
Water	0.36 ± 0.07a	1.33 ± 0.36a	1.83 ± 0.41a	2.25 ± 0.31a	1.65 ± 0.47a	2.05 ± 0.23a	0.45 ± 0.14a	0.78 ± 0.17a	1.05 ± 0.29a	2.00 ± 0.59a	2.08 ± 0.50a	2.53 ± 0.22a
W + s	0.45 ± 0.06a	1.25 ± 0.29a	1.50 ± 0.20a	1.82 ± 0.31b	1.58 ± 0.21a	2.00 ± 0.23a	0.28 ± 0.06a	0.65 ± 0.16a	1.08 ± 0.28a	1.80 ± 0.50a	1.88 ± 0.52a	2.68 ± 0.41a
S. p	0.20 ± 0.10a	0.05 ± 0.03b	0.05 ± 0.03b	0.05 ± 0.03c	0.05 ± 0.03b	0.05 ± 0.03b	0.18 ± 0.06a	0.05 ± 0.03c	0.08 ± 0.04b	0.05 ± 0.03c	0.10 ± 0.04b	0.08 ± 0.05c
C. d (1%)	0.33 ± 0.08a	0.20 ± 0.07b	0.13 ± 0.05b	0.23 ± 0.06c	0.28 ± 0.08b	0.20 ± 0.04b	0.18 ± 0.05a	0.23 ± 0.08bc	0.33 ± 0.05b	0.43 ± 0.06bc	0.35 ± 0.05b	0.40 ± 0.08bc
C. d (5%)	0.25 ± 0.06a	0.08 ± 0.04b	0.08 ± 0.04b	0.10 ± 0.08c	0.18 ± 0.08b	0.13 ± 0.04b	0.38 ± 0.10a	0.15 ± 0.06bc	0.33 ± 0.06b	0.30 ± 0.08bc	0.38 ± 0.03b	0.28 ± 0.08bc
C. d (10%)	0.30 ± 0.09a	0.05 ± 0.03b	0.05 ± 0.03b	0.08 ± 0.05c	0.15 ± 0.05b	0.13 ± 0.04b	0.30 ± 0.09a	0.13 ± 0.04bc	0.18 ± 0.05b	0.15 ± 0.05bc	0.25 ± 0.06b	0.18 ± 0.09bc
S. a (1%)	0.33 ± 0.06a	0.23 ± 0.08b	0.15 ± 0.05b	0.18 ± 0.03c	0.30 ± 0.14b	0.10 ± 0.04b	0.33 ± 0.08a	0.33 ± 0.09b	0.33 ± 0.08b	0.50 ± 0.12b	0.40 ± 0.04b	0.53 ± 0.10bc
S. a (5%)	0.30 ± 0.07a	0.05 ± 0.03b	0.15 ± 0.07b	0.08 ± 0.04c	0.05 ± 0.03b	0.10 ± 0.04b	0.25 ± 0.09a	0.28 ± 0.11bc	0.23 ± 0.06b	0.43 ± 0.10bc	0.30 ± 0.07b	0.23 ± 0.06bc
S. a (10%)	0.25 ± 0.05a	0.10 ± 0.04b	0.10 ± 0.04b	0.10 ± 0.04c	0.10 ± 0.05b	0.05 ± 0.03b	0.40 ± 0.07a	0.15 ± 0.07bc	0.20 ± 0.05b	0.15 ± 0.03bc	0.15 ± 0.03b	0.20 ± 0.05bc
T. v (1%)	0.20 ± 0.10a	0.13 ± 0.08b	0.13 ± 0.06b	0.08 ± 0.04c	0.20 ± 0.09b	0.20 ± 0.04b	0.30 ± 0.08a	0.20 ± 0.07bc	0.33 ± 0.11b	0.40 ± 0.12bc	0.48 ± 0.08b	0.45 ± 0.05bc
T. v (5%)	0.20 ± 0.09a	0.08 ± 0.05b	0.10 ± 0.04b	0.08 ± 0.03c	0.05 ± 0.03b	0.00 ± 0.00b	0.30 ± 0.07a	0.15 ± 0.03bc	0.25 ± 0.05b	0.23 ± 0.10bc	0.38 ± 0.03b	0.25 ± 0.06bc
T. v (10%)	0.25 ± 0.10a	0.03 ± 0.03b	0.08 ± 0.04b	0.05 ± 0.08c	0.08 ± 0.05b	0.03 ± 0.03b	0.35 ± 0.08a	0.13 ± 0.05bc	0.15 ± 0.05b	0.10 ± 0.04bc	0.15 ± 0.05b	0.15 ± 0.05bc
M. p. (2.5%)	0.25 ± 0.05a	0.10 ± 0.04b	0.08 ± 0.04b	0.05 ± 0.03c	0.10 ± 0.04b	0.00 ± 0.00b	0.25 ± 0.05a	0.18 ± 0.06bc	0.15 ± 0.06b	0.18 ± 0.06bc	0.28 ± 0.05b	0.25 ± 0.06bc
M. p. (5%)	**0.28 ± 0.06a**	**0.03 ± 0.03b**	**0.05 ± 0.03b**	**0.00 ± 0.00c**	**0.00 ± 0.00b**	**0.00 ± 0.00b**	**0.40 ± 0.08a**	**0.08 ± 0.04c**	**0.10 ± 0.04b**	**0.05 ± 0.04c**	**0.08 ± 0.04e**	**0.10 ± 0.05c**
2 - way ANOVA	**(F- Statistics)**											
Locations (L)	3.88*	11.55***	29.87***	2.48ns	3.19*	14.02***	39.00***	4.02*	42.00***	69.22***	37.63***	2.65ns
Treatments (T)	0.78ns	12.53***	33.72***	38.94***	16.78***	115.31***	1.38ns	5.97***	13.47***	27.37***	51.11***	35.97***
L*T	0.75ns	1.33ns	4.25***	2.24*	2.65**	7.03***	1.07ns	0.69ns	5.44***	14.71***	28.46***	0.33ns

Each value is a mean ± standard error of eight replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fishers Least significant Difference (LSD) test.

Appendix 7: Mean Percent damage of *B. oleracea* per crop by insect pests for 2019 and 2020 seasons

Location and Treatments	Week 1 before Treatment	Weeks after treatments 2019 wet season					Week 1 before Treatment	Weeks after treatments 2020 wet season				
		1	2	3	4	5		1	2	3	4	5
Experimental sites												
Tengeru	20.6 ± 1.1a	18.6 ± 1.6a	16.5 ± 1.8a	14.6 ± 1.9a	13.5 ± 1.8b	17.7 ±2.5a	20.0 ± 0.8a	14.2 ± 0.9a	14.7 ± 1.3a	16.3 ± 1.7a	18.6 ± 1.7a	21.4 ±2.1a
Boro	10.1 ± 0.5b	9.6 ± 0.9b	10.8 ± 1.0b	14.5 ± 1.3a	16.6 ± 1.8a	19.2 ± 2.8a	19.3 ± 0.7a	14.8 ± 1.3a	15.8 ± 1.9a	18.8 ± 2.6a	18.9 ± 2.9a	18.2 ± 3.1b
Treatments												
Water	18.8 ± 3.1a	33.8 ± 5.6a	33.1 ± 4.5a	38.8 ± 2.3a	45.0 ± 4.3a	70.6 ± 3.1a	22.5 ± 2.1a	31.9 ± 3.3a	41.3 ± 3.5a	50.0 ± 6.9a	50.6 ± 5.7a	59.4 ± 5.0a
W + s	15.6 ± 4.4a	30.0 ± 5.7a	31.9 ± 3.7a	41.3 ± 3.1a	41.3 ± 3.0a	56.3 ± 2.3b	23.1 ± 1.6a	25.6 ± 1.5b	38.1 ± 3.4a	49.4 ± 4.7a	58.8 ± 7.2a	66.9 ± 4.3b
S. p	16.3 ± 2.6a	8.1 ± 1.6c	6.3 ± 1.6f	6.9 ± 1.9ef	7.5 ± 1.6de	5.0 ± 1.3gh	19.4 ± 2.4a	10.0 ± 3.0de	3.1 ± 1.3f	2.5 ± 0.9g	3.1 ± 1.3g	3.8 ± 1.8ge
C. d (1%)	16.9 ± 2.8a	16.3 ± 3.2b	17.5 ± 3.0b	17.5 ± 1.9b	18.1 ± 1.9b	18.8 ± 1.8c	18.8 ± 0.8a	16.3 ± 1.8c	16.3 ± 1.3b	18.1 ± 2.1bc	18.8 ± 1.8bc	21.3 ± 2.1b
C. d (5%)	15.0 ± 2.7a	10.0 ± 1.3bc	13.8 ± 3.5bcde	15.0 ± 2.7bc	15.0 ± 2.1bc	18.8 ± 1.8c	21.3 ± 2.6a	13.1 ± 0.9cd	16.3 ± 2.3b	16.3 ± 2.8bc	15.0 ± 1.6bcde	11.3 ± 2.3de
C. d (10%)	13.8 ± 1.8a	11.3 ± 1.6bc	11.3 ± 1.6bcdef	11.3 ± 0.8cde	11.3 ± 2.1cd	12.5 ± 2.8de	17.5 ± 2.3a	10.0 ± 1.3e	6.9 ± 1.3ef	7.5 ± 1.9defg	10.0 ± 2.7defg	7.5 ± 2.1de
S. a (1%)	14.4 ± 2.4a	13.8 ± 2.5bc	15.6 ± 2.4bcd	15.0 ± 1.3bc	17.5 ± 1.9b	18.1 ± 1.6c	18.1 ± 1.6a	15.6 ± 2.6cd	16.9 ± 1.3b	20.6 ± 2.9b	21.3 ± 1.6b	21.9 ± 2.7b
S. a (5%)	12.5 ± 1.3a	10.6 ± 1.1bc	10.0 ± 0.0cdef	8.1 ± 1.3ef	8.8 ± 1.3de	10.6 ± 1.5ef	17.5 ± 2.5a	14.4 ± 2.7cd	16.3 ± 0.8b	15.0 ± 2.3bcd	15.6 ± 1.8bcd	14.4 ± 1.8bcd
S. a (10%)	14.4 ± 2.2a	8.8 ± 1.8bc	8.1 ± 0.9ef	06.9 ± 1.3ef	8.1 ± 1.6de	9.4 ± 1.5efg	18.8 ± 2.1a	11.3 ± 2.1cde	11.9 ± 3.0bcd	11.9 ± 3.0cdef	12.5 ± 2.1cdef	10.6 ± 2.4de
T. v (1%)	11.9 ± 3.3a	12.5 ± 1.9bc	16.3 ± 4.6bc	13.8 ± 2.3bcd	15.0 ± 0.9bc	17.5 ± 1.3cd	16.9 ± 1.3a	14.4 ± 1.5cd	15.6 ± 1.5bc	19.4 ± 1.1bc	18.1 ± 1.3bcd	20.0 ± 2.7bc
T. v (5%)	16.3 ± 3.0a	11.3 ± 1.6bc	8.8 ± 1.8def	8.8 ± 0.8def	6.9 ± 1.6de	6.9 ± 1.6fgh	19.4 ± 2.4a	11.9 ± 0.9cde	10.0 ± 0.9de	12.5 ± 1.9bcde	12.5 ± 1.3cdef	14.4 ± 2.2bcd
T. v (10%)	11.9 ± 1.6a	10.0 ± 1.9bc	6.3 ± 1.6f	7.5 ± 1.3ef	7.5 ± 1.3de	5.6 ± 1.1fgh	19.4 ± 1.5a	11.3 ± 2.5cde	6.3 ± 1.3f	6.3 ± 2.8efg	6.9 ± 1.9efg	8.1 ± 1.9de
M. p. (2.5%)	19.4 ± 4.3a	10.6 ± 1.8bc	6.3 ± 1.6f	8.1 ± 1.9ef	5.0 ± 1.3e	4.4 ± 1.1gh	19.4 ± 1.5a	10.6 ± 1.1cde	10.6 ± 1.5cde	12.5 ± 2.1bcde	14.4 ± 1.8bcde	13.8 ± 1.8cd
M. p. (5%)	18.1 ± 4.0a	10.6 ± 3.2bc	6.3 ± 1.6f	5.0 ± 0.8f	3.8 ± 1.3e	3.8 ± 1.3h	23.1 ± 1.9a	6.9 ± 0.9e	4.4 ± 1.1f	3.8 ± 1.6fg	5.0 ± 1.6fg	5.0 ± 1.6e
2 - way ANOVA	**(F- Statistics)**											
Location	82.55***	57.25***	20.28***	0.04ns	8.41**	2.39ns	0.46ns	0.32ns	1.26ns	3.43ns	0.07ns	7.15**
Treatments	1.25ns	12.57***	14.05***	43.79***	39.94***	119.05***	1.11ns	10.38***	40.67***	36.56***	38.99***	73.18***
L*T	1.24ns	1.74ns	1.30ns	1.86ns	0.56ns	0.77ns	0.89ns	0.77ns	2.47**	4.43***	3.32**	3.70**

Each value is a mean ± standard error of eight replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fishers Least significant Difference (LSD) test.

Appendix 8: The weekly level of incidences (%) per plot in the field in 2019 and 2020 wet seasons

Location and Treatments	Weeks after treatments - 2019 season					Weeks after treatments - 2019 season				
	Week 1	Week2 after	Week3 after	Week4 after	Week5 after	Week 1	Week2 after	Week3 after	Week4 after	Week5 after
Location										
Tengeru	6.0 ± 1.1a	15.3 ± 1.7a	24.0 ± 2.8b	25.7 ± 3.3a	25.6 ± 3.3b	19.8 ± 1.4a	18.5 ± 1.7a	18.9 ± 2.0a	24.6 ± 1.7a	27.7 ± 2.3a
Boro	7.9 ± 1.4a	16.2 ± 2.0a	28.7 ± 2.7a	29.2 ± 3.2b	28.9 ± 3.4a	13.7 ± 1.5b	16.2 ± 1.9b	20.1 ± 2.4a	23.2 ± 2.9b	22.3 ± 3.2b
Treatments										
Water	23.4 ± 3.7a	43.2 ± 4.3a	72.4 ±2.6a	79.4 ± 2.4a	81.5 ± 2.5a	37.5 ± 2.2a	41.7 ± 2.7a	53.1 ± 4.1a	54.2 ± 3.5a	67.7 ± 3.7a
W + s	20.6 ± 3.6ab	39.6 ± 2.1b	67.7 ± 2.6a	79.4 ± 1.8a	79.4 ± 1.8a	32.3 ± 2.9a	42.7 ± 1.9a	51.0 ± 4.3a	62.5 ± 5.0a	70.8 ± 3.5a
S. p	**1.0 ± 1.0ef**	**3.6 ± 1.4e**	**12.5 ± 3.6de**	**6.8 ± 1.9gh**	**6.5 ± 1.0i**	**10.4 ± 3.0de**	**3.1 ± 1.5f**	**2.1 ± 1.4f**	**5.2 ± 2.2g**	**5.2 ± 2.2h**
C. d (1%)	14.6 ±2.7bc	25.8 ± 2.1b	30.5 ± 3.8b	33.1 ± 3.1b	36.7 ± 2.5b	19.8 ±2.2b	24.0 ± 1.0b	21.9 ± 2.7bc	27.1 ± 2.6bc	30.2 ± 2.2b
C. d (5%)	5.5 ± 2.0def	11.7 ± 2.4cd	20.3 ± 2.5c	23.4 ± 2.2c	25.3 ± 2.4cde	18.8 ± 2.6bc	17.7 ± 1.9c	17.7 ± 1.9bcd	20.8 ± 1.6cde	18.8 ± 2.6def
C. d (10%)	2.1 ± 2.1ef	8.9 ± 2.1cde	16.7 ± 1.7cd	20.6 ± 3.3cd	21.1 ± 4.7def	11.5 ± 3.8cde	10.4 ± 3.0de	11.5 ± 2.2de	14.6 ± 2.6ef	11.5 ± 3.1fgh
S. a (1%)	9.1 ± 2.4cd	21.4 ± 2.6b	34.6 ± 2.2b	35.4 ± 2.2b	32.0 ± 3.0bc	19.8 ± 1.5b	24.0 ± 2.9b	24.0 ± 2.5b	30.2 ± 1.5b	28.1 ± 2.2bc
S. a (5%)	4.7 ± 2.1def	13.3 ± 1.4c	20.8 ± 2.8c	19.5 ± 2.0cde	18.8 ± 2.2efg	13.5 ± 2.7bcd	19.8 ± 2.7bc	17.7 ± 2.5bcd	21.9 ± 2.7cde	21.9 ± 2.7cde
S. a (10%)	2.6 ± 1.3ef	5.5 ± 2.0de	15.9 ± 3.8cd	15.1 ± 4.2def	10.2 ± 5.0hi	13.5 ± 3.1bcd	10.4 ± 2.6de	10.4 ± 2.6de	15.6 ± 2.9def	14.6 ± 3.0efg
T. v (1%)	7.0 ± 1.2de	19.8 ± 2.3b	28.9 ± 2.2b	30.7 ± 2.0b	28.4 ± 3.4cd	20.8 ± 2.2b	18.8 ± 1.4bc	21.9 ± 2.2bc	27.1 ± 2.1bc	24.0 ± 2.9bcd
T. v (5%)	0.8 ± 0.8ef	6.5 ± 2.2de	15.6 ± 1.9cd	12.8 ± 3.2efg	15.1 ± 3.1fgh	11.5 ± 3.1ce	11.5 ± 2.2d	14.6 ± 3.0cde	17.7 ± 2.9def	19.8 ± 2.2de
T. v (10%)	2.9 ± 1.4ef	7.0 ± 2.3cde	12.0 ± 2.3de	8.3 ± 2.3fgh	8.1 ± 2.1hi	8.3 ± 1.6de	5.2 ± 1.5ef	7.3 ± 1.0ef	10.4 ± 2.6fg	9.4 ± 1.9gh
M. p. (2.5%)	3.1 ± 2.2def	9.1 ± 1.3cde	13.8 ± 2.0cde	14.6 ± 1.4def	10.9 ± 1.5ghi	11.5 ± 2.7cde	11.5 ± 2.7d	17.7 ± 2.9bcd	22.9 ± 1.4bcd	22.9 ± 3.0bcd
M. p. (5%)	**0.0 ± 0.0f**	**5.2 ± 1.7e**	**7.3 ± 2.0e**	**4.7 ± 1.4h**	**7.0 ± 1.2i**	**5.2 ± 1.5e**	**2.1 ± 1.4f**	**2.1 ± 1.4f**	**4.2 ± 1.6g**	**5.2 ± 2.2h**
2 - way ANOVA	**(F- Statistics)**									
Location	2.43ns	0.53ns	12.16***	6.82*	4.62*	22.22***	4.19*	0.87ns	1.49ns	20.04***
Treatments	11.10***	28.20***	62.58***	91.70***	72.40***	13.90***	38.43***	41.87***	65.66***	82.30***
Location*treatments	0.28ns	0.26ns	0.88ns	0.56ns	0.47ns	0.58ns	2.03ns	2.72**	6.25***	3.22***

Each value is a mean ± standard error of eight replicates, *, **, and *** significant at P≤0.05, P≤0.01 and P≤0.001 respectively and ns means not significant. Means within the same column followed by the same letter(s) are not significantly different at P=0.05 from each other using Fishers Least significant Difference (LSD) test.

RESEARCH OUTPUTS

(i) Publications

Mpumi, N., Machunda, R., Mtei, K. M., & Ndakidemi, P. A. (2020). Selected Insect Pests of Economic Importance to *Brassica oleracea*, Their Control Strategies and the Potential Threat to Environmental Pollution in Africa. *Sustainability, 12*(9), 1-22.

Mpumi, N., Machunda, R. L., Mtei, K. M., & Ndakidemi, P. A. (2020). Insecticidal Efficacy of *Syzygium aromaticum*, *Tephrosia vogelii* and *Croton dichogamus* Extracts against *Plutella xylostella* and *Trichoplusia ni* on *Brassica oleracea* crop in Northern Tanzania. *AIMS Agriculture and Food, 6*(1), 185-202. https://doi: 10.3934/agrfood.2021012.

Mpumi, N., Mtei, K. M., Machunda, R. L., & Ndakidemi, P. A. (2021). Efficacy of aqueous extracts from *Syzygium aromaticum*, *Tephrosia vogelii*, and *Croton dichogamus* against *Myzus persicae* on *Brassica oleracea* in Northern Tanzania. *Psyche: A Journal of Entomology*, *2021,* 1-11. https://doi.org/10.1155/2021/2525328.

Mpumi, N., Machunda, R. L., Mtei, K. M., & Ndakidemi, P. A. (2021). Aqueous extracts from *Syzygium aromaticum*, *Tephrosia vogelii* and *Croton dichogamus* provide environmentally benign control of *Crocidolomia binotalis* on *Brassica oleracea* crop in the fields in Northern Tanzania. *Journal of Biodiversity and Environmental Sciences, 19*(5), 65 -77.

(ii) Poster presentation

Mpumi, N., Mtei, K. M., Machunda, R. L., & Ndakidemi, P. A. (2021). Evaluation selected botanicals as insecticides against cabbage insect pests in Tanzania.