UNIVERSITY OF CAPE COAST

THE EFFECTS OF PRE-HARVEST FOLIAR SPRAY AND POST-HARVEST DIP OF ETHEPHON ON THE PHYSIOLOGY AND QUALITY OF ORANGE-FLESHED SWEET POTATO *APOMUDEN* DURING STORAGE

BY

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Thesis submitted to the Department of Agricultural engineering, School of Agriculture of the College of Agriculture and Natural Sciences, University of Cape Coast, in partial fulfilment of the requirements for the award of Master of Philosophy degree in Post-Harvest Technology.

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DECLARATION

Candidate's Declaration

I hereby declare that this thesis is the result of my own original work and that no part of it has been presented for another degree in this University or elsewhere.

Candidate's Name: Weldon Kipkoech Sitienei

Signature: …… Date: …….……..……...

Supervisors' Declaration

We hereby declare that the preparation and presentation of this thesis were supervised in accordance with the guidelines on supervision of thesis laid down by the University of Cape Coast.

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ABSTRACT

The postharvest storage of sweet potato roots in the tropics is a major challenge in the crop's value chain as it deteriorates readily in the hot climate. Although prolonged storage in the cold chain has been shown to be feasible, technical and economic constraints in the tropics makes cold storage inaccessible to growers and retailers of sweet potato. It has been demonstrated in several studies that the plant hormone ethylene greatly influences metabolic changes in many types of horticultural produce postharvest and that controlled application can improve storage. In this research, the ethylene analogue ethephon was applied at four different concentrations as foliar spray on the Orange-Fleshed Sweet Potato *Apomuden* seven days before harvest and stored in the ambient. Untreated roots were also dipped in the same ethephon concentrations postharvest. Changes in the physiological and biochemical quality (preharvest treatment only) *viz*. weight loss, sprouting, decay, shrinkage, dry matter content, starch, sucrose, glucose, fructose and minerals concentration were studied. Preharvest foliar application of ethephon resulted in superior storage performance than postharvest root dip as it better reduced sprouting, improved resistance to weevil damage and shrinkage. Preharvest application of 500 ppm ethephon significantly reduced the dry matter loss compared to the control. Furthermore, carbohydrates, proteins and minerals were significantly more abundant in the ethephon treated roots.

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DEDICATION

To my caring, loving and supportive wife Mrs. Sylvia Sitienei Mosonik and son Jayden Kiprotich Sitienei for their constant support and ample time.

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LIST OF ACRONYMS

ACC	Aminocyclopropanecarboxylate
ANOVA	Analysis of Variance
AVG	Aminoethoxyvinylglycine
CRD	Completely Randomized Design
CIP	International Patato, Lima, Peru
CRI	Crops Research Institute, Ghana
CIPC	Chlorophenylcarbamate
C_2H_4	Ethylene
DW	Dry Weight
DM	Dry Matter
EAC	East African Community
FAO	Food and Agriculture Organization
FAOSTAT	Food and Agriculture Organization Statistics
FW	Fresh Weight
GI	Glycemic Index
HPLC	High Performance Liquid Chromatography
LSD	Least Significant Difference
MENA	Methyl Naphthalene-acetic Acid
1-MCP	1-Methylcyclopropene
MOFA	Ministry of Food and Agriculture
MH	Maleic Hydrazide
nm	nanometer
n	replicates
NIRS	Near Infrared Spectroscopy

OFSP	Orange Fleshed Sweet Potato
PDA	Potato Dextrose Agar
p	probability
USDA	United Stated Department of Agriculture
viz.	namely
WHO	World Health Organization
$\mu l\ L^{-1}$	microliter per liter
ppm	parts per million

CHAPTER ONE

INTRODUCTION

Background to the Study

Sweet potato (*Ipomoea batatas* (L.) Lam) is a member of the Convolvulaceae family which is grown for its fleshy storage roots. Sweet potato is reported to have originated in northwestern South America, arising from a hybrid cross or through karyotypic changes from an unidentified plant of the genus *Ipomoea*. It was introduced to Europe by the Spanish and spread to China, Japan, Malaysia and the Moluccas area. The Portuguese introduced it to India, Africa and Indonesia (O'Brien, 1972).

The root crop is grown annually though it is a perennial crop. It confers a wide range of health benefits that have recently enhanced its popularity. Sweet potato is one of the important food crops especially in developing countries (Woolfe, 1992). The white fleshed variety is the most cultivated in many developing countries and it is ranked among the five most important food crops (CIP, 2016). Sweet potato is among the world's most nutritious, yet under-exploited food crops (Srinivas, 2009).

With annual global production of more than 133 million tonnes, sweet potato is currently positioned as the fifth most essential food crop in developing countries after rice, wheat, maize, and cassava (Oke & Workneh, 2013). Sweet potato is among the widely grown root crops in Sub-Sahara Africa. It is specifically cultivated in countries surrounding the Great Lakes in Eastern, Central and parts of West Africa (Shonga, Gemu, Tadesse, & Urage, 2013). The cultivated area in Africa is about 3.2 million hectares with a projected output of 13.4 million tonnes of roots in 2005. It is mainly produced

in marginal soils in low-input subsistence farming systems of developing countries where it is a major food crop (Woolfe, 1992).

Sweet Potato Cultivation and Storage Challenges

Despite its reputation as a food security crop, sweet potato roots are highly perishable if not well treated and properly stored. Particularly, damage by diseases and pests can lead to severe economic losses. The most common sweet potato pests include sweet potato weevil, white fly, wire worm and cricket (Theberge, 1985). These pests attack the roots both in the field and during storage. Some of the diseases that affect sweet potato include fusarium wilt, soil rot, black rot, root knot-nematode and other viral diseases (Jackson & Bohac, 2006). They all lead to decay and loss of economic value of sweet potato roots. Furthermore, temperatures above 15°C lead to more rapid sprouting and weight loss. The weight loss is due to increased respiration which uses up the stored food reserves in the roots. This restricts their shelf life and the distance over which sweet potato can be economically transported (Rees *et al*., 1998).

Maximum quality of sweet potatoes is mainly determined by the type of pretreatment that they are subjected to immediately after harvesting. This can also be determined by the growing conditions of the roots. Pre-harvest cultural practices during the growing season will later affect postharvest quality. Some factors such as the weather are impossible to control when the crop is planted in the open fields. Other growing factors like fertilizer application can be manipulated by a grower to ensure that a quality product goes into storage. The weather during the growing season, especially just before and during harvest, has a major effect on the postharvest quality. An

extended drought, followed by heavy rain frequently accelerates growth, which often produces roots with delicate skin that are prone to growth cracks and attack by soil-borne pests.

Several studies have been conducted in the past to investigate the effective storage methods to prolong the shelf life and foster availability of the crop year round. Research on various methods of extending the shelf life of sweet potato has been ongoing. These include cold storage, modified atmosphere (MA) storage, pit storage and processing to chips. Other preservation methods involve the application of botanical extracts in the field prior to harvesting.

Some plant hormones like ethylene have been used to preserve both climacteric and non-climacteric produce. The effects of ethylene can extend or reduce the shelf life of agricultural produce depending on the growth stage, species, cultivar, type of tissue and the ethylene application regime (Abeles, 1969). Ethylene effects on the quality of climacteric fruits have been well documented and it is an important commercial hormone used for extending their shelf-life. Ongoing research on the effects of exogenous ethylene on low ethylene producing vegetables is largely focused on understanding its role in regulating dormancy, sprout growth and senescence in potatoes and onions (Amoah, 2014). The use of supplemental ethylene can help in quality retention of stored sweet potato and other agricultural produce (Cheema, Rees, Westby, & Taylor, 2008). In this research ethephon, as an ethylene precursor, was used to treat orange-fleshed sweet potatoes and its potential to preserve quality and shelf life of the roots was investigated on sprout control, storage decay, weight

loss and shrinkage. In addition, the effects on the abundance of some nutritionally beneficial phytochemicals were studied.

Problem Statement

Sweet potato roots once harvested deteriorate rapidly mainly due to physiological, biochemical and microbial changes, which are accelerated by mechanical damage during harvesting, transportation and handling. In tropical weather conditions, the tuberous roots endure just 1-2 weeks and not more than 5 weeks under favorable storage conditions (Rees *et al*., 2003). This significantly reduces the economic and health prospects that could be derived from the production and utilization of sweet potato. The roots exhibit short dormancy and are susceptible to sprouting, decay and insect infestation during storage. The very short storage life after harvest has been recognized as a major hindrance to the cultivation of the crop. As a result, processing the roots into storable forms such as chips and flours are done to prolong the shelf life (Ayinde & Dinrifo, 2001). However, most consumers prefer fresh sweet potato roots to the processed ones. This has encouraged the production and marketing of fresh sweet potatoes but the fresh roots require suitable pretreatment and storage innovations to make it available over the entire year.

In many rural communities in the Central Region of Ghana, sweet potato production is a main income earner for some small-scale farmers. However, these farmers are hindered by the high perishability caused by sprouting and decay of the roots (Duku, 2005). The lack of good storage facilities compels the farmers to sell their sweet potatoes at the most reduced prices when there is surplus during the harvesting season. This has led to food insecurity and high economic losses from the monetary investments in sweet

potato cultivation. This discourages more famers from taking up sweet potato farming (Birago, 2005).

Purpose of the Research

Because of its numerous agronomic advantages including high yield, high calorific value and its adaptation to different climatic environments, sweet potato has great potential for alleviating food insecurity especially in developing countries (Amoah, 2014). Sweet potato is gaining acceptance in both the developing and developed countries as one of the staple crops. Orange-fleshed sweet potatoes (OFSP) may be one of nature's unsurpassed sources of beta-carotene. Several recent studies have shown the superior ability of the OFSP to raise blood levels of vitamin A. This benefit may be particularly true for children (FAO/WHO, 2002).

Storage of sweet potatoes, however, could be associated with a number of physico-chemical changes which may reduce its nutritional and health value. Consequently, there is little involvement in sweet potato farming by farmers in developing countries. Limited storage of sweet potato roots is practiced by retailers during the main harvesting season between June and September. There is an abundant supply which lowers the demand; hence the surplus deteriorates drastically (Agbemafle, Owusu-Sekyere, Diabor, & Essien, 2013). This is largely due to the rapid deterioration of the roots in tropical and sub-tropical climates. Extending the shelf life of the fresh roots is thus a major setback in its role as a stable food crop. Prevalent storage problems include sprouting, microbial decay and weevil infestation.

Research shows that there are pretreatments which farmers and processors use to improve the shelf life of sweet potatoes after harvest. These

include storing in sawdust-ash mix and blanching (Babajide, Obadina, Oyewole & Ugbaka, 2006; Sanadeera, Bhandari, Young & Wijesinghe, 2000). In a research by Teye, Amoah and Tetteh (2011), the use of the botanical extract of *Lantana camara*, ash and brine reduced weevil damage, weight loss, shrinkage, decay and sprouting in two varieties (Ukerewe and TIS2) stored in two different storage structures.

In their findings, TIS 2 variety pre-treated with *Lantana camara* extract was suggested for storage while the evaporative cooling barn was the recommended storage structure for sweet potato roots. However, more research needs to be conducted since only a few of these storage methods have proven to be satisfactory. To date, there is no universally appropriate pre-treatment approved for both subsistence and commercial sweet potato storage. The effects of existing pre-treatment methods on the physiological and biochemical changes during the storage of sweet potato have not been fully researched into. Continuous application of exogenous ethylene gas postharvest to improve the storage of related crops such as potato and onion has been studied with the potential of reducing sprouting and other storage disorders (Daniels-Lakes, Prange, Nowak, Asiedu, & Walsh, 2005, Buffler, 2009).

Similar studies have been done on sweet potato by Cheema, Rees, Colgan, Taylor & Westby (2013) and Amoah (2014). In conclusion, the authors recommended that appropriate ethylene application regime could enhance storage, particularly inhibition of sprout growth. As a natural plant hormone, ethylene may be expected to gain consumer acceptance (Amoah, 2014). The application of ethylene gas, however, may pose difficulties for farmers in developing countries. It is in this regard that this research was

conducted to elucidate the effect of the ethylene precursor, ethephon, on sweet potato shelf life and quality.

Objectives of the Study

Main Objective

To investigate the effects of ethephon treatments applied pre- and post-harvest on the physiology, biochemistry and quality of sweet potatoes during storage.

Specific Objectives

To determine the effects of preharvest foliar application and postharvest root dip of ethephon on:

1. The root physiological weight loss, sprouting and shrinkage.
2. The incidence and severity of root decay.
3. The mode of infection and proliferation of root decay by microorganisms.
4. The concentration of starch, fructose, sucrose, glucose, minerals (Zn, Fe) and proteins as affected by the treatments.

Statement of Hypotheses

Null Hypothesis

There is no significant effect of ethephon treatment on the physiology, biochemistry and quality of orange-fleshed sweet potato.

Alternative Hypothesis

There is a significant effect of ethephon on the physiology, biochemistry and quality of orange-fleshed sweet potato.

Significance of the Study

The study conducted is meant to elucidate the latest sweet potato preservation method whereby ethylene hormone is used to curb storage losses. Ethephon is one of the ethylene precursors being used generate ethylene which regulates the growth and development of different plants and crops. Preharvest ethephon treatments (100 ppm and 500 ppm) performed better than the other treatments in most determinations conducted. The knowledge on the effects of ethephon on *Apomuden* sweet potatoes roots provides the much needed postharvest data and information on pretreatments to improve the shelf life of OFSP.

Delimitations

The study was conducted mainly in Cape Coast, Central Region of Ghana. The biochemical tests, however, were conducted in Crop Research Institute sweet potato quality laboratory in Kumasi.

Limitations

The post-harvest experiment was carried out as a separate research from the preharvest, thus the experiments are separate experiments and comparison between the two applications regimes may have a minor variation in describing the behavior of the OFSP roots. However the roots were sourced from the same farmer and were from the same variety.

Definition of Terms

Ethylene: Ethylene is plant growth hormone that occurs naturally and has various effects on the development, growth and storage life of numerous fruit, vegetables and flower yields at low concentrations.

Ethephon: Is scientifically known as di-chloroethylphosphonic acid. It is a systemic plant growth and development regulator.

Dormancy: a period in a plant's life cycle when growth and development are temporarily stopped.

Organisation of Study

Chapter One introduces the study with the background, statement of problem, objectives and significance of study. Chapter Two reviewed literature on sweet potato botany, economic importance, production, nutritional facts, root structure, storage practices, postharvest losses, diseases, pests and biochemical composition. It also highlight on its storage requirements and use of ethylene to curb deterioration of the roots. In Chapter Three the methodology used in the study is discussed with the fourth chapter presenting the results and their discussion. The last chapter, five, contains the summary of findings, conclusions and recommendations.

CHAPTER TWO

LITERATURE REVIEW

Botany and Origin of Sweet Potato

Sweet potato (*Ipomoea batatas* (L.) lam), a dicotyledonous plant, belongs to the family *Convolvulaceae.* The family comprises of 45 genera and 1000 species with only *Ipomoea batatas* having economic value as food crop (Woolfe, 1992). Sweet potato is reported to have originated in northwestern South America, arising from conceivably as a hybrid cross or through karyotypic changes from an unidentified plant of the genus *Ipomoea*. A sweet potato genetic bank at the International Potato Centre (CIP) conserves 5,526 of the growing *I. batatas* cultivars in 57 countries (Zhang, Cervantes, Huaman, Carey & Ghislain, 2000). Information regarding the genetic make-up of this group is crucial for germplasm preservation and use. Most research findings support the suggestion that Central America is the initial center of diversity and utmost probable source of sweet potato.

Peru-Ecuador is believed to be the secondary source of sweet potato diversification (Zhang, Rossel, Kriegner, & Hijmans, 2004). It was introduced to Europe by the Spanish and it spread to China, Japan, Malaysia and the Moluccas area. The Portuguese introduced it to India, Africa and Indonesia (O'Brien, 1972). Sweet potato is the third most important food crop in seven Eastern and Central African countries ahead of maize and cassava. It is ranked fourth in six Southern African countries and is number eight in four West African countries (CIP, 2016).

Economic Importance of Sweet Potato

Sweet potato has many agronomic benefits such as high yield, early maturity, superior nutritional value and adaptability to different climatic environments. This gives the sweet potato a great potential for reducing hunger, specifically, in the low income households in developing countries (Amoah, 2014). Several varieties of sweet potato have nutritionally beneficial compounds (Islam, 2006). Sweet potato is commonly grown as a staple food in many parts of the tropics and subtropics, which includes several developing countries. Besides its use as a food crop the roots have a variety of other uses such as livestock feed and the production of industrial starch. They are also processed into flour and canned; the flour is further used in preparing confectioneries like pies, puddings, cakes and biscuits (Woolfe, 1992). Sweet potato contributes significantly to the annual food production and utilization in the coastal savannah and north-eastern areas of Ghana (Abano et al., 2011). Improving sweet potato cultivation is subsequently recognized as a means to achieve viable food security and alleviating poverty among the low income households in developing countries (Amoah, 2014).

Production and Consumption of Sweet Potato

Sweet potato is grown in over 100 countries in the warm temperate and tropical regions as staple and export food produce (Lebot, 2009). Among other valuable crops, sweet potato is ranked as the seventh most cultivated food crop in the world and fifth most significant food crop contributing to over 95 % of global production in developing countries (CIP, 2016). When compared with other main staple food crops, sweet potato has a varied range of desirable qualities: high yield, nutritional benefits including vitamins, low glycemic

index and high in dietary fibre. They also do well in most geographical zones with short production cycle and resistance to production stresses like high temperature, water shortage, insect and disease stress. For the past four decades, global sweet potato production has remained static (105 million metric tonnes) despite the world population doubling (Kays, 2004). This has seen the consumption per capita gradually declining. Developing countries consumes over 98 % of the globally produced of sweet potato (Kearney, 2010). Over the last decade, sweet potato has been the center of rigorous, worldwide research in breeding and value addition in the quest to exploit its prospects as a food source. For instance, CIP and its international partners have implemented the sweet potato bio-fortification platform in Sub-Saharan Africa with the emphasis on the development of the OFSP varieties (CIP, 2016). Asia contributes approximately 85 % of the total world production while Africa produces 10 % and about 5 % is produced by the other countries around the globe. China leads in production of more than 80 % of the overall world production (FAOSTAT, 2015).

The cultivated area of sweet potato in Ghana is about 73,400 ha (FAO, 2010) which comes after cassava and yam in order of importance among root crops. Ghana has an estimated production of 135,000 metric tonnes and ranks as the 35^{th} among the sweet potato producing countries (FAO, 2013). Considering the subsistent production, the Crops Research Institute of Ghana approximates the production to be 200,000 tonnes (Asafu-Agyei, 2010).

Production zones, including Ghana, which produce surpluses are localized but scattered, and prompts an absence of well linked markets. There is minimal processing into chips or flour, which could be kept longer

(Andrade *et al*., 2009). Limited crop diversity, coupled with little knowledge of sweet potato's potential nutritional and economic benefits might be the reason for its low productivity and consumption in most parts of Ghana. In addition, poor storage facilities for the farmers have led to slow integration of sweet potatoes into the commercial trade.

According to Abano *et al*. (2011), in the Central Region of Ghana, sweet potato is one of the major income generating crops for most farmers from Moree, Jukwa and Koforidua communities. However, the root tuber undergoes fast deterioration; thus farmers are compelled to sell their harvested crop at low prices.

Nutritional and Health Benefits

Sweet potato is rich in carbohydrates, dietary fibre, iron, calcium and vitamins. Most importantly, the OFSP roots have a great potential to supply high levels of beta-carotene, which is a precursor to vitamin A (Andrade *et al*., 2009). If the orange fleshed variety is incorporated into diets in Sub-Saharan Africa, it could significantly reduce the prevalence of vitamin A deficiency (Meenakshi *et al*., 2010). Research in Africa has shown that high levels of β-carotene in sweet potato could enable diets with low Vitamin A content to be enriched and thus decrease the deficiencies (Hagenimana, Carey, Gichuki, Oyunga, & Imungi, 1998). Moreover, it is acknowledged that carotenoids, particularly carotene provide antioxidant activity to the human food (Akhtar & Bryan, 2008). Carotenoids are among a group of phytochemicals that promote well-being and boost the immunity (De Nardo, Shiroma-Kian, Halim, Francis & Rodriguez-Saona, 2009; El-Sohemy *et al*., 2002). Among all carotenoids, β-carotene is generally found in fruit and vegetables, for example, sweet

potatoes, carrots, tomatoes and sweet potato based foods (Baranska, Schutze & Schulz, 2006).

According to Zuraida (2003) sweet potato roots can be boiled, fried, baked or processed to make starch for ethanol production. It can also be utilized as a partial flour substitute for cake or bread production. Some food products such as tomato sauce, ketchup, dried-cake, spongy cake and biscuit use sweet potato as one of their ingredients. New uses such as for the production of noodles, cookies, doughnuts could increase the demand for sweet potatoes.

Growth of Sweet Potato Roots

Sweet potato can be propagated from vines and roots slip. The vines establish very quickly and can provide a ground cover only a few weeks after establishing (Woolfe, 1992). Sweet potato growth cycle can be categorized into three stages. These include extensive growth of fibrous roots with moderate growth of the vines, extensive growth of the vines with remarkable proliferation in leaf expanse and start of storage root growth and finally, storage root expansion with little growth of fibrous roots and vines. The extent of the stages varies with environmental conditions and cultivars. The duration of growth is between 3-7 months depending on the cultivar and the genetic and environmental conditions which also affect leaf area, number of leaves and abscission, storage root formation and development and total dry matter (Ravi & Indira, 1999). In the tropics, most cultivars are harvested when the roots achieve salable size normally in 3-8 months once planted (Lebot, 2009). The roots should not stay in the ground for too long as they will be susceptible

to attack by pest and various rots. In addition, the storage roots might reach the rejuvenation phase which triggers sprouting (Woolfe, 1992).

Structure of Sweet Potato Roots

The roots are joined to the parent plant by means of stalks at the proximal end where adventitious buds are found and from where sprouts normally originate. The root tapers at the distal end. The buds that are at the central and distal end sprout later than the proximal buds. The surfaces of the roots are covered with layers of cork which may be smooth or unevenly grooved with prominent veins and longitudinal grooves. Lenticels can also be found on the surface in some varieties which can be protruding due to excess water in the soil. This is the part where the adventitious buds are found and thus sprouting starts here (Hayward, 1938). A transverse section of the root has protective periderm and the cortex which varies in thickness depending on the cultivar. It also has the cambium rings and the central parenchyma. The roots vary in shape and sizes depending on the cultivar and type of soil where the roots are growing among other factors. They can be elliptical, round-elliptic, ovate, oblong or long and curved. The root colour can be white, yellow, red, purple, brown-orange, pink and dark purple. The flesh colour can be orange, yellow, cream, white or purple depending on the cultivar (Huaman, 1992).

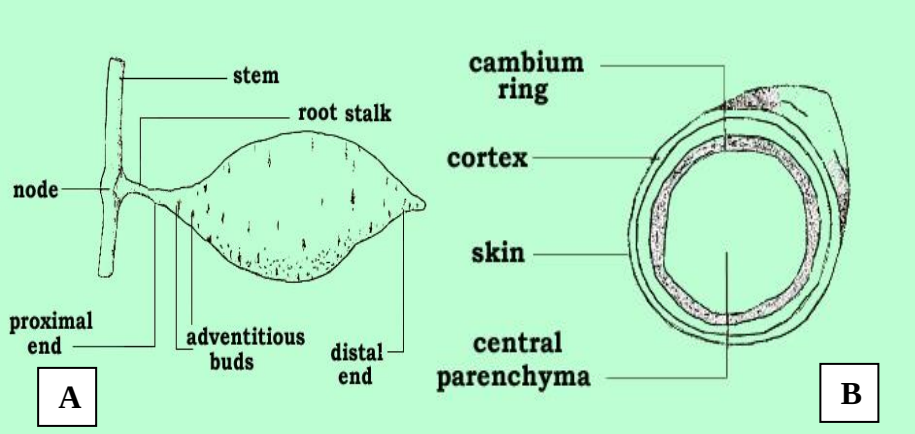

Figure 1: Sweet potato root external structure (A) and transverse section (B) Source: (Huaman, 1992)

Dormancy

Various cultivars and crops have varied levels of the dormancy stages. This is the main feature which explains the differences in their shelf life. Dormancy is elicited in many plant species in response to unfavorable climatic conditions towards the end of the growth regime (Chokchaichamnankit *et al*., 2009). It is the most vital postharvest physiological stage as it determines the duration which the produce can be stored without intrinsic growth. Potatoes, onions and yams also undergo dormancy.

Sweet potato roots, once harvested, can stay dormant for a short time and sprout early if relative humidity and temperature are favourable. It is reported that sweet potato do not undergo dormancy as they can sprout instantly after harvest if stored in conditions that favours sprouting (Cheema, 2010). During dormancy, the roots have reduced intrinsic metabolism but the root maintains the potential for future development. Viola *et al*. (2007) observed that dormancy in potato retards only meristematic growth of the tuber buds but the rest of the tuber remains metabolically active. Various researches propose three types of dormancy which include endodormancy,

ecodormancy and paradormancy. Regulation of dormancy in vegetative buds is an intricate process necessary for plant survival and development. In many cases, dormancy break results in increased cell division and development. Internal activities such as hormones and sugar as well as external signals such as temperature act through specific pathways to regulate endodormancy, ecodormancy and paradormancy (Horvath, Anderson, Chao & Foley, 2003).

Endodormancy is the principal innate dormant period which is reliant on the genetic characteristics of the plant. Ecodormancy involves a prolonged rest period caused by external conditions like humidity and temperature and may be induced by varying the environmental conditions (Schoot, 1996). Paradormancy in sweet potato occurs when the proximal sprouts suppress distal buds from sprouting (Onwueme & Charles, 1994). Plant hormones have been associated with dormancy control whereby growth retardants such as abscisic acid and ethylene induce dormancy while dormancy break is through the biosynthesis of growth stimulators like gibberellins and cytokinnins (Arteca, 2013). Moreover, environmental conditions such as temperature and light play crucial roles in regulating dormancy induction and release of bud dormancy (Chao, Foley, Horvath & Anderson, 2007).

Low temperatures are known to boost the initiation of endodormancy. However, prolonged periods of cold cause a break of endodormancy (Rohde & Bhalerao, 2007). Under temperate field conditions, bud growth does not happen immediately after endodormancy release because ecodormancy is sustained by low temperatures. Mechanisms involved in ecodormancy maintenance are unidentified; however the role of abscicic acid (ABA) is suspected (Horvath *et al*., 2003).

Curing

Proper curing of sweet potatoes after harvest improves the skin toughness, cooking attributes and reduces postharvest losses initiated by diseases. Curing is done at a temperature of 27-30 °C and a relative humidity of 85-95 % for 4-7days before storage (Onwueme, 1994). Curing improves storage life (Wills, Mcglasson, Graham, Lee & Hall, 1989). Curing allows the wounds that occur during harvesting and handling to heal by being coated with layers of lignified and suberized cells. Suberization and lignification protect the roots against water loss and attack from pathogens (McClure, 1960; Snowdon, 1990). Proper curing is an important step to supply the market with high-quality sweet potato all year-round. It also increases sweetness which enhances the culinary characteristics of the roots (Edmunds *et al*., 2008).

Different sweet potato varieties vary in their root dry matter content. Low root dry matter content is attributed to high curing efficiency. Curing efficiency of varieties also differ in response to the curing days. At the end of the curing treatments, samples of healthy storage roots were removed from the center of bins and stored at 15 °C for 1-2 months and evaluated for end rot, soft rot and tip rot. In all locations, soft rot incidence decreased as temperature increased ($r = -0.48$). Soft rot was higher in the control conditions at all locations ranging from 1.4 to 7.5 %. The average soft rot incidence in cured roots was 1.3 %, 0.7 %, and 0.3 % for the Edmonson, Morgan, and Bailey locations, respectively (Ray & Ravi, 2005).

The benefits of curing sweet potatoes and their effects on diseases and culinary characteristics were studied in a bid to encourage growers to adopt proper curing practices of sweet potatoes. Cured and non-cured storage roots

were evaluated for the tip and end rot incidence after 6 weeks. Curing reduced tip rot incidence from 2.2 % to 0.6 % and end rot incidence from 6.4 % to 1.2 % (Manning & Arancibia, 2009).

van Oirschot, Rees, Aked, & Kihurani (2006) conducted a study on the process of wound healing in sweet potatoes subjected to tropical conditions at RH of 50–73 % and temperature between 25–30 °C and noted that at RH of 65% ± 10, the thickness of the lignified cell layers was influenced by both humidity and cultivar. Some of the cultivars steadily failed to generate a lignified cover while others had a layer which was not continuous. The effectiveness of wound healing depended on the continuity of the lignified layer. Reduced predisposition to microbial attack and reduced weight loss was attributed to wound lignification.

Postharvest Losses in Sweet Potato

Sweet potato can be stored under controlled environments for several months. It is reported that temperature range of 13-15 °C and high relative humidity (90%) can keep the roots up to a year (Picha, 1986). However, in tropical countries it is reported that storage for 3-4 months is possible where tubers are carefully sorted and kept in traditional pits or clamps where high humidity is maintained (Linus, 2014). On the other hand, Rees *et al*. (2001) reported that sweet potato roots stored in tropical ambient environmental conditions have a much shorter shelf life of 2-3 weeks. The losses are attributed to sprouting, storage pests and diseases and mechanical injuries during harvesting and transportation. Transportation via road exposes the tubers to mechanical shocks which result in skin injury (Tomlins, Ndunguru, Rwiza & Westby, 2000).

Sprouting

Sprouting is an important economic feature in commercial sweet potato production. During planting time, vigorous and plentiful sprouts production is required for propagation material (Hall, 1993). Conversely, sprout growth decreases the quality and value of roots for fresh market sales (Lewthwaite & Triggs, 1995). Sprouting is one of the main features used to assess the physiological quality of the harvested sweet potato roots. Root sprout is a flaw, and a portion is considered sprouted when 10 % of the roots tubers contain sprouts longer than 19 mm (USDA, 2005). Sweet potato roots sprout easily in the tropics. They have a short period of dormancy of about two weeks when kept in the ambient storage. The roots have high moisture content and thus active metabolism occurs. Subsequently, sprout growth is initiated following a period of endodormancy (Sonnewald, 2001).

The main biochemical mechanism which initiates dormancy break is yet to be exhaustively explained. It has been reported that sprout growth factors and dormancy are controlled by endogenous growth hormones including abscisic acid, giberillins and cytokinins. The action of these hormones can be controlled by manipulating the relative humidity and temperature of the stored roots (Cheema, 2010). At the start of sprouting, almost all the sprouts arise at the proximal section of the root as the buds in the distal and middle segments go through paradormancy. This indicates a difference in spatial metabolism of growth allied factors at the proximal section in relation to the distal and middle sections. Nevertheless, sprouting can also initiate from other segments of the roots as they stay longer in storage (Onwueme & Charles, 1994).

Controlling sprout growth under ambient storage conditions of temperature and humidity is a valuable preservation tool for farmers who do not typically have environmentally controlled stores. Hence, controlling the extent of the dormancy period is of significant economic importance. Other new strategies are needed because the current cold storage techniques or treatment with sprout inhibitors are often not available in resource-poor countries. Cold storage, nevertheless, prompts the starch conversion to sugar if the roots are kept below 12 °C. Gamma irradiation of sweet potatoes is found to be an effective sprouting suppressing technique, but the required quantities are quite high. This can lead to undesirable nutritional and biochemical alterations (Ajlouni & Hamdy, 1988).

Other researches have used chemical sprout suppressants such as isopropyl-3-chlorophenylcarbamate (CIPC) and tetrachloronitrobenzene maleic hydrazide (MH). CIPC is typically applied to the tubers to be stored using thermal fogging techniques. The aerosol is spread through potato piles with the use of fans. Conversely, their use is being discouraged world-wide due to the need to reduce the application of synthetic chemicals to vegetables and fruits during growth and storage. Despite the use of CIPC to inhibit sprouting in tubers, its toxicology and residue levels are still of concern (Forsythe & Forsythe, 1999). Moreover, there is declining market acceptance of all types of agrochemical residues in food (Daniels-Lake, Pruski & Prange, 2011). Ethylene gas has also been used as a sprout inhibitor. Its toxicity to humans and other animals is very low for instance the LC50 for mice in air is 950,000 μl L^{-1}. The mode of action is a simple asphyxiant which displaces oxygen (O'Neil, Smith, Heckelman, & Budavari, 2001). Ethylene is produced

by all plants as a growth regulator. It has been registered as a potato sprout suppressant in Canada since 2002 and is also approved for use in the United Kingom and several other nations (Daniels-Lake *et al*., 2011). Nevertheless, ethylene can have negative effects on the fry colour of processed potato products (Prange *et al*., 1998). The response to ethylene varies among cultivars, and the concentration needed for sufficient sprout inhibition and the influence on sugar metabolism and fry color also varies (Jeong, Prange, & Daniels-Lake, 2002). In addition, ethylene requires sophisticated instruments for its application and hinders its use in the developing countries where farmers cannot afford the machines.

Recent researches on other feasible sources of ethylene have identified some ethylene precursors viz. ethephon and calcium carbide which can be used to supply ethylene (Tripathi, Sayre, Kaul & Narang, 2003; Wang Arancibia, Main, Shankle & LaBonte, 2013). From research, it is evident that, depending on the concentration and time of exposure, ethephon has inhibitory growth effect on plants and this effect was evaluated to find out if it could reduce sprouting of sweet potato during storage. Lately, an alternative method has been developed which involves the production of genetically modified potatoes with improved metabolic pathways to lengthen the dormancy of the harvested tubers (Sonnewald, 2001).

Physiological Loss of Weight

Sweet potatoes weight losses occur mainly from water loss. This is the main means of deterioration of the roots and causes rotting. Physiological weight losses are attributed primarily to water loss through respiration and transformation of dry matter to energy. Substantial loss of dry matter can

cause pithiness which results in the formation of several small holes (Rees *et al*., 1998). Transpiration also causes water losses due to vapor pressure difference between the interior of the root and the outside environment. All these will lead to loss in weight of the roots during storage (Kushman & Pope, 1972; Picha, 1986).

Varied causes of weight loss in sweet potato depending on the variety and storage systems have been reported. Ray, Chowdhury and Balagopalan (1994) conducted an experiment to find out the improvement of shelf life of sweet potato by storing them in locally available materials including sand, soil and saw dust, and to evaluate the percentage of weight loss, rotting and weevil infestation associated with storage. Roots (Variety Pusa Safed and No. 8516) of medium size (75-140 g) were mixed and placed in plastic baskets in storage room having temperature 21°C–30°C and R.H. 70-85 %.

Half the lots were enclosed within polythene sheet so as to create a constant relative humidity of approximately 90 %. Sequential weighing of the roots indicated a continued decrease in roots fresh weight. After 45 days in store, a mean weight loss of 20 % had resulted, and there was no significant difference between cured and uncured tubers. After 15 and 30 days, however, the percentage loss was significantly greater in untreated tubers ($p < 0.01$). Additionally, more than 90 % of the root tubers were free from fungal attack at the end of 45 days storage. Approximately 8.5 and 6.4 % of uncured and cured tubers respectively, exhibited fungal rotting. On the other hand, there were no substantial differences in weevil infestation. The outcome of this experiment showed clearly that the losses in fresh weight noted were mainly due to moisture loss and respiration.

In a comparative storage performance study of two varieties of sweet potato in an evaporative cooling barn and pit structure, Amoah, Teye, Abano, and Tetteh (2011) reported 5 % weight loss after two weeks of storage. Further 12 weeks in store resulted in 32.5 % weight loss.

Postharvest Diseases of Sweet Potato

A number of microorganisms, commonly fungi, are established to cause decay in stored sweet potatoes. The most significant fungal pathogens are *Botryodiplodia theobromae*, *Fusarium* spp., and *Rhizopus oryzae* and*Ceratocystis fimbriata* (Clark & Hoy, 1994). The new but rarely detected decay microorganisms include *Plenodomus destruens,Cochliobolus lunatus, Macrophomina phaseolina and Rhizoctonia solani* (Ray & Misra, 1995).

Fungal and bacterial diseases affecting the stored roots are significant because they reduce the yield, aesthetic value, shelf life and nutritive value of the stored roots. The pathogens cause local disruption of tissues and discoloration of infested roots (Snowdon, 1990). This results in changes in appearance, deterioration of texture and flavor. Microbial rot of sweet potato results in the decrease in total sugar, starch, ascorbic acid and oxalic acid content. However, the levels of polyphenols, ethylene and in some cases phytoalexins increases as a result of the pathogen attack (Ray & Ravi, 2005).

The actions of these pathogens lead to postharvest losses, fall in the market value and low income to farmers. The time of infection differs with the causal organism, location of harvest and store conditions (Moyer, 1982). Black rot, fusarium rot, scurf and bacterial soft rot can occur before harvest, during harvest and after harvest. On the other hand, soft rot infection tends to

occur at harvest and after harvest but root rot and surface rot occur during harvest (Kays, 1991).

Black Rot

This is caused by *Ceratocytis fimbriata* and is among the most destructive diseases of sweet potatoes. The symptoms include black, dry spots on the stored roots and which are often found mainly at the uppermost cortex (Fig. 2). Perithecia of the fungus are at times produced on the surface of infested tissues and they have a distinctive fruity smell. Within few weeks of storage the spots reach a diameter of an inch or more under favorable conditions. The spores are spread in the store on the bodies of insects and possibly by air currents and by other means, during handling in preparation for sale (Woolfe, 1992). Further spread and losses are stopped by removal of all noticeable diseased roots. Roots from soils identified to be infested should be avoided (Snowdon, 2010).

***Figure 2*:** Black root rots in sweet potato roots

Charcoal Rot

Charcoal rot is a predominant postharvest disease which is caused by *Macrophomina phaseolina*. It thrives well in warm weather with extensive host range. It causes a firm decay on the stored roots. The tissues initially

become reddish-brown and then turn black as sclerotia of the pathogen are made in the tissue (Bartz & Brecht, 2002).

As the decay continues, the pathogen advances towards the center of the root. Two distinctive zones can be identified within the infested tissue. These include the leading part that progress as a red-brown decay and a black zone develops behind the active decay portion (Fig. 3) (Ames, Smit, Braun, O'Sullivan & Skoglund, 1996).

Figure 3: Roots infested with charcoal rot

Fusarium Rot

Fusarium surface rot is a storage disease which affects sweet potatoes. It is caused by fungal strains of *Fusarium oxysporum*, which is a soil-borne pathogen carried by wounded roots. The disease typically occurs after harvesting and its symptoms include brown, dry rot confined to the cortex of the roots (Fig. 4). The roots infections majorly depend on the field conditions especially in dry conditions when the roots develop growth cracks (Loebenstein, Thottappilly, Fuentes & Cohen, 2009).

***Figure 4*:** Roots showing Fusarium surface rot

Fusarium root rot is caused by *Fusarium solani* and it is a common field and storage rot. The firm and dark decay extends deep into the root. Internally, oval voids form in which a white mold grows (Fig. 5). It is a soil-borne disease which can be spread through infected planting materials.

Figure 5: Root infected by Fusarium root rot

Rhizopus Soft Rot

The disease is caused by *Rhizopus oryzae* and *Rhizopus nigricans* which are main species in the tropical and subtropical zones. Storage roots become wet, soft and decays with little alteration in skin colour (Fig. 6). Decay occurs mainly in wounds that have their adjacent tissues killed (Holmes & Stange, 2002). Harvested roots are the only one affected and exposing the roots to direct sunlight before storage predisposes them to the disease (Ames *et al*., 1996).

Figure 6: Rhizopus soft rot infected root

Java Black Rot

This is caused by *Diplodia tubericola* and it is among the most destructive postharvest diseases. Decay starts at one or both ends of the root and it initially appears brown, but turns black later. This decay is firm and moist early on, but in short time storage roots become completely black and shriveled (Fig. 7). The disease is fully known by the black stromatic lesions which erupt from the periderm of the roots (Skoglund & Smit, 1994).

***Figure 7*:** Root infected by java black rot disease

Control Measures

A number of methods are used to curb microbial decay. Curing to stimulate wound healing is found as the best appropriate technique to reduce microbial rot. Fungicide usage, irradiation, bio-control and storage in ash and saw dust have been found to have moderate effects in controlling decay and improving shelf life of sweet potato roots. Fungicides are used to protect the

roots from *rhizopus* soft rot attack. However, the use of synthetic fungicides is a potential danger to both the farmer and environment (Amienyo & Ataga, 2007). It has also disrupted biological control by natural enemies, leading to the outbreak of other species of pests which are resistant to the pesticide (Salunke, Kotkar, Mendki, Upasani & Maheshwari, 2005).

Postharvest Pests of Sweet Potato

Sweet potato is known to be affected by *Cylas sp.* both in the field and during storage. Harvested roots that have been affected by weevils should not be stored because weevils multiply and increase in number, which consequently destroys wholesome roots within a short period of time.

Delate, Brecht & Coffelt (1990) used a controlled atmosphere made up of low oxygen (O_2) and increased levels of carbon dioxide (CO_2) with a stable nigrogen (N_2) for up to 10 days at 25°C and 30°C to control adult sweet potato weevils, *Cylas formicarius elegantulus*. The adult sweet potato weevils died within 4–8 days when exposed to 8 % O_2 plus 40–60 % CO_2 at 30°C. At temperature of 25°C, exposure to 2 or 4 % O_2 plus 40 or 60 % CO_2 at 25°C exterminated all the adult insects within 2-8 days.

Biochemical Composition of Sweet Potato

The biochemical composition of sweet potato roots varies according to cultivar, degree of maturity, climatic conditions and storage time after harvest (Osundahunsi, Fagbemi, Kesselman & Shimoni, 2003). The shelf-life and nutritional value of the roots changes after harvest due to several intrinsic and environmental factors which readily influence the nutritional composition of the roots (Sriroth *et al*., 1999).

Dry Matter Content

Sweet potato has high moisture content but low in the dry matter as in other root crops (Woolfe, 1992). Consumers in Africa have been noted to have a preference for high dry matter varieties of sweet potato. However, sweet potato varieties with high carotenoid contents tend to have lower dry matter contents (Kapinga & Carey, 2003). The International Potato Center (CIP) has been breeding OFSP varieties that contain high levels of carotenoid and dry matter (CIP, 2016).

Generally, the dry matter content of sweet potato is 30 %. However, it varies depending on the cultivar, climate, location, type of soil, gardening practices and the occurrence of diseases and pest (Bradbury & Holloway, 1988). In a research by Tomlins, Owori, Bechoff, Menya and Westby (2012), the sensory attributes of bio fortified sweet potato in Africa were studied over an extensive range of carotenoid (0.4–72.5 μg/g fresh weight) and dry matter contents (26.8–39.4 %) were studied. The logarithm of the total carotenoid content was correlated with the dry matter content declining by 1.2 % with each doubling of the carotenoid content. The dry matter content of the sweet potato dropped with increasing carotenoid content.

According to Sajedi, Madani, Safari-Kamal-Abadi (2009), about 30-40 % reduction in yield and dry matter content of the roots is likely to occur by delaying planting time of sweet potato than standard planting time. Five varieties grown in Ghana had their dry matter content ranging between 34.4 to 37.4 % (Aidoo & Tetteh, 2004). On the other hand, dry weight content ranging from 21 to 40 % was obtained for eight varieties bred by Ghana Crops Research Institute (Asafu-Agyei, 2010).

Total Carbohydrates

Carbohydrates comprise the main portion of the dry matter of sweet potato (approximately 80-90%). They consist of various amounts of soluble sugars, starch, cellulose, pectin and hemicelluloses (Woolfe, 1992). These compounds determine the shelf life of the roots; a high total carbohydrate makes the roots store better. The specific carbohydrates and the relative amount influence the quality and shelf life. The carbohydrates differ with the environment, cultivars, the growth stage of the roots and storage conditions. A minor reduction in the carbohydrate composition in sweet potato in the course of storage has been observed (Reddy & Sistrunk, 1980; Zhang, Wheatley & Corke, 2002). It has also been noted that an extended storage of raw root before processing reduced the root firmness and flour pasting viscosity but sucrose and glucose increased (Zhitian, Wheatley & Corke, 2002).

According to Zhitian *et al*. (2002) when sweet potato roots are stored for more than three months at suitably low temperatures, they will undergo a reduction in starch content and a rise in alpha-amylase. Storage also affects both cooking and sensory properties. The roots lose flavor if they are kept in storage for long without proper pretreatment

Starch

Starch is the major carbohydrate in the sweet potato roots. The process formation of starch from photosynthesis stops immediately the roots are harvested hence the living tissues of the roots rely on the internally stored reserves for their biochemical activities. Hence the quantity of stored starch is vital for longer storage life of the roots. The stored starch goes through enzymatic conversion to glucose (Aiyer, 2005). When cooked, the bulk of the

starch is transformed to maltose, which gives the cooked roots the sugary taste (Onwueme & Charles, 1994).

Sweet potato starch has nutritional and health benefits due to its low glycemic index (GI) grade (48 ± 6). Thus there is gradual breakdown of the starch when consumed leading to a slight increase in blood sugar (Foster-Powell, Holt & Brand-Miller, 2002). Consequently, sweet potato is a good diet for patients with type-2 diabetes. In addition, the crop is also used to curb obesity, as it relieves feelings of hunger for several hours. Recently, Low, Walker and Hijmans (2001) reported that bio-fortified sweet potato varieties have the capacity to reduce vitamin A deficiency in children in the Sub-Sahara Africa.

According to Zuraida (2003) the roots can be boiled, fried, baked or processed to make starch for ethanol production. It is used as a partial flour substitute for cake or bread production. Some food products such as tomato sauce, ketchup, dried-cake, spongy cake and biscuit use sweet potato as one of their ingredients. The roots are frequently boiled, fried or baked. New uses such as for the production of noodles, cookies, doughnuts could increase the demand for sweet potatoes. Notable genetic variations exist in the starch content among varieties and genotypes, which results in differences in their storage behavior and utilization as food (Collado & Corke, 1997). The starch composition additionally varies with growth conditions, maturity, location and season with an average of 18 % (Tian, Rickard & Blanshard, 1991).

According to Noda, Takahata, Sato, Ikoma and Mochida (1997), the effect of planting time on starch properties of sweet potato suggested early planting time to raise gelatin materials and adherence of starch. Huber (1983)

found that the starch content is correlated with the dry weight. Approximately 40 % of China's annual sweet potato produced is used for animal, principally pig feed use (Scott & Suarez, 1992). Roots and leaves can be used as feed in many forms, but the low digestibility of raw starch is one of the limitations to feed efficiency. A substantial difference in starch digestibility has been observed among sweet potato genotypes (Noda, Takahata, Nagata & Monma, 1992). It has been established that digestibility of sweet potato flour is greatly correlated with starch digestibility (Zhang, Collins & Belding, 1993).

Zhang *et al*. (2002) investigated the changes in the level of carbohydrate, α-amylase, digestibility trypsin inhibitor activity and the pasting properties of roots of six genotypes of sweet potato during storage. Most genotypes showed an insignificant decrease in starch content during 0–180 days of storage, but in the genotype Hi-dry, it decreased significantly. Alpha-amylase activity rose during the first 2 months of storage, followed by a reduction from continued storage to a level similar to harvesting time. The decline in starch content was correlated with α-amylase activity in the first 60 days storage (r=0.80, p=0.06).

Sugars

The main forms of sugars in sweet potato are sucrose, glucose and fructose. Their comparative amounts vary amongst genotypes (Zhang *et al*., 2002). Sucrose makes the bulk of sweet potato sugar. When cultivars are analyzed in raw form, the sucrose concentration exceeds the other sugars (Van Den, Biermann & Marlett, 1986). It makes up 68 % of total sugar and the rest account for 22- 49 % subject to the variety (La Bonte, Picha & Johnson, 2000). Cooked sweet potatoes have maltose which results from hydrolysis of

the stored starch (Van Den *et al*., 1986). Moreover, storage raises the sugar content in sweet potato roots (Zhitian *et al.,* 2002). The total sugar composition in the fresh roots of different American varieties determined by Picha (1985) ranged from 2.9 % to 3.2 % whilst Puerto Rico cultivars fluctuated from 6.3 to 23.6 mg/100 g of dry weight (Martin & Deshpande, 1985).

La Bonte *et al*. (2000) studied the quantity and pattern of carbohydrate-related changes during storage root development among six sweet potato cultivars. Sucrose was the main sugar during all stages of development, accounting for over 68 % of total sugars across all cultivars and dates. Pairwise evaluations revealed 'Heart-o-Gold' had the highest content for the 12 weeks of evaluation. Fructose content profiles varied within and among cultivars. 'Beauregard' showed a steady increase in fructose through the development while 'Whitestar' showed a constant decrease. The fructose content profiles in the other cultivars were inconsistent. Glucose content profiles were similar to those for fructose changes during development. Zhang *et al*. (2002) found that glucose and sucrose concentration increased early in storage and then remained fairly constant.

Structural Carbohydrates

These are non-starch polysaccharides found in sweet potato and other plants. They include cellulose, hemicelluloses and pectic substances which constitute the cell wall of the plants. They are also called dietary fibres and are known to give protection against colon cancer, diverticulosis, cardiovascular diseases and diabetes (Woolfe, 1992). The dietary fibre portion of sweet potato roots ranges from 1.2-2.62 %. They play structural support by

strengthening the cell walls giving characteristic textural properties like softness, firmness and dryness of the sweet potato roots (MacDougall & Selvendran, 2001).

Total Proteins

According to Maeshima, Sasaki and Asahi (1985), the tuberous roots of sweet potato contain large quantities of two proteins which account for more than 80 % of the total proteins. Ishida et al. (2006) reported that every 100 g of the fresh roots of sweet potato contain 1.0-1.7 g of protein. The two proteins consist of sporamins A and B in monomeric forms. The two forms were significantly present during the dormant period, but a reduction was noted during sprouting (Chokchaichamnankit *et al.,* 2009). The proteins were separated from each other by electrophoresis on polyacrylamide gels in a non-denaturing buffer. They are found to be similar to each other with respect to amino acid composition, peptide map and immunological properties.

These proteins decrease compared to other proteins during sprouting. The amino acid sequencing of the amino terminal part of sporamin A showed that it is a composition of more than two molecular species with different combinations of a few amino acids. Hattori, Nakagawa, and Nakamura, (1990) found that the distribution of the protein in the root is not even. The proximal end has a higher concentration than the distal end. Significantly higher sprouting has been noted at the proximal end than distal sprouting of sweet potato. This could be attributed to the high protein concentration in the proximal end.

Polyphenols

Polyphenols are abundant micronutrients in our diet mainly known for their starring role in the prevention of cardiovascular diseases and cancer. The health effects of polyphenols are influenced by the amount consumed and on their bioavailability. Bioavailability differs significantly between the different polyphenols and the most abundant polyphenols in the diet may not necessarily have the greatest bioavailability profile to impart protective health effects (Manach, Scalbert, Morand, Remesy & Jimenez, 2004).

Polyphenols exist as secondary metabolites in most plant tissues. They play important functional and nutritional roles like flavour, taste, colour, anti-fungal activity and anti-oxidant activity (Borbalan, Zorro, Guillen & Barroso, 2003). Sweet potato leaves are an outstanding source of antioxidative polyphenolics compared to other commercial vegetables. The antioxidants help prevent molecular harm caused by oxidation in addition to helping to fend off several diseases such as muscular degeneration, cancer and cardiovascular diseases (Islam, 2006). They also lead to polyphenol-oxidase-catalyzed enzymatic browning which affect the appearance and sensory properties of food (Friedman, 1997).

Polyphenols are categorized based on the number of phenol rings that they contain and the structural elements that bind the rings to each other. They are broadly divided into four main classes; flavonoids, phenolic acids, stilbenes and lignans. Flavonoids form the bulk of polyphenols in the human diet and have a basic structure which consists of two aromatic rings bonded together by three carbon atoms (Pandey & Rizvi, 2009). Phenolics in sweet potato include caffeic acid, chlorogenic acid, isochlorogenic acids and

caffeylamide (Truong, McFeeters, Thompson, Dean & Shofran, 2007). Research by Truong, Thompson, McFeeters and Lanier (2004) reported significant differences in phenolic contents between tissue zones and among sweet potato cultivars. The total phenolic content measured in commercial US cultivars ranged from 0.72 to 1.75 and 0.50 to 0.70 mg chlorogenic acid equivalent per gram of the fresh weight for the outer root layers and inner tissue, respectively. Total phenolic content for varieties grown in the Philippines including *Dakol*, *Emelda*, *Haponita*, PSBSP and Violet went from 192.7 to 1159.0 mg gallic acid equivalent /100 g dry sample (Rumbaoa, Cornago & Geronimo, 2009).

Minerals and Vitamins

The abundant minerals in the sweet potato roots are sodium (Na), potassium (K), chloride (Cl), calcium (Ca) and phosphorus (P) (Ravindran *et al*., 1995). Sweet potato mineral content depends on the variety. Phosphorus from 38-64 mg, Potassium from 245-403 mg, Calcium 20-41 mg, Magnesium 13-22 mg were found in 100 g of sweet potato by Picha (1985).

Orange flesh sweet potato root is important source of carotenoids which are sources of vitamins precursor. Among all carotenoids, β-carotene is widely distributed in fruits and vegetables such as carrots, sweet potatoes, carrot and sweet potato based products, tomatoes and tomato-based products (Baranska, Schütze & Schulz, 2006). The yellow and orange fleshed varieties contain high quantities of carotenoids that act as precursors of vitamin A. They also contain reasonable quantities of riboflavin (B_2), thiamine (B_1), pantothenic acid (B_5), folic acid and pyrodoxin (Kidmose, Christensen, Agili & Thilsted, 2007). Orange-fleshed sweet potato has emerged as one of the

richest plant sources of ß-carotene (Hagenimana and Low, 2000; Low *et al*., 2001). A 100 grams serving of boiled roots can supply about 50 % of the daily vitamin A need of a young child. The current varieties of orange-fleshed sweet potatoes contain 20–30 times more ß-carotene than does Golden Rice (Kryder, Kowalski & Krattiger, 2000). The ascorbic acid amounts declines as the roots stays in storage (Woolfe, 1992). Variations in carotenoid pigmentation in the course of curing and storage might also influence the root quality in relations to colour and nutritive value (Picha, 1985). Some of the biochemical content of fresh roots and the leaves of *Apomuden* as determined by CSIR Food Research Institute (personal communication) are shown in Table 1.

Table 1: ***Biochemical Content of the Fresh Roots and Leaves of Apomuden OFSP Sourced from a Sweet Potato Farmer in Cape Coast***

Parameter	Method	Unit	Roots	Leaves
Ash	AOAC 923.03(2000) 17th Edition	g/100g	3.87	7.64
Protein	AOAC 984.13(1999) 15th Edition	g/100g	7.36	14.17
Carbohydrates (including fibre)	By difference	g/100g	83.37	69.79
Energy	Atwater Factor	kcal/100g	374.62	355.64
Vitamin C	2,6-dichlorophenolin-dophenol	mg/100g	4.43	9.53
Copper	Atomic Absorption Spectrophotometric	mg/100g	0.30	0.13
Zinc	Atomic Absorption Spectrophotometric	mg/100g	0.33	0.52
Iron	Atomic Absorption Spectrophotometric	mg/100g	-	32

Source (Food Research Institute, CSIR, Ghana)

Sweet Potato Storage

The storage of sweet potato roots in sub-tropical and tropical countries is the key challenging task to growers and sellers due to lack of appropriate storage facilities and the unfavorable climate. As a result, sweet potato is rarely kept in the market chain. Within the tropical countries, sweet potato is frequently kept in traditionally crafted structures which can hardly keep the roots wholesome for long. Typical shelf-life of less than one week in the East Africa market chain has been reported (Van Oirschot, Rees & Aked, 2003). The longevity of the roots is determined by the storage structure. Efforts to realize long-lasting storage of sweet potato with various storage structures within the tropical areas have been described. A number of the methods have developed through native knowledge; and their scientific base and their effects on the, compositional changes, nutritive value and shelf-life have not been exhaustively assessed (Amoah *et al*., 2011; Agbemafle *et al*., 2013).

In addition, hardly does any of the systems so far satisfies the storage needs, as they can barely create the best environments vital for lasting storage. Approximately four weeks or shorter seems the longest time that can be achieved by some of the storage methods (Amoah, 2014). Innovative storage methods of sweet potato in pit and evaporative barn with pre-treatment of roots with botanical plant extract repellants from neem and *Lantana camara* to avert weevil damage has been tried in parts of Ghana (Abano *et al*., 2011). Chemical spraying with maleic hydrazide and methyl naphthalene-acetic acid (MENA) in acetone has been found to prevent sprouting and prolong storage

from 4 to 8 weeks (Kay, 1987). However, these compounds have negative effect on environment and the health of living organisms.

Storage Requirements to Improve Sweet Potato Quality

Sweet potato storage up to 6 months is possible without temperature control if the roots are selected with care and stored in pits (Mwambene, Mayona & Mwakyembe, 1992). This requires that the harvesting, curing and storage are carefully conducted. The storage room should be clean and able to provide favourable conditions of relative humidity, temperature and gas composition. Care should be taken to minimize bruising and injuries on the roots during digging, collection, grading and placing them in storage containers. Moreover injured roots and diseased ones should be removed from the store. The roots should be properly cured and stored in wooden boxes or bins with good ventilation openings (Hall & Devereau, 2000).

Temperature and Relative Humidity

Fresh produce is highly perishable mainly because of its high moisture content. Proper storage of produce is one of the most important aspects of preservation (Watada, 1999). Temperature is the most important element in maintaining the quality of fresh vegetables after harvest. Low temperature storage retards the following elements of deterioration in perishable crops: senescence due to respiration, softening and colour changes. It also reduces detrimental metabolic changes and respiratory heat generation; moisture loss and the wilting that follow. It also curbs deterioration due to attack by bacteria, fungi in addition to reducing undesirable growth like sprouting of potatoes (Hardenburg, Watada & Yang, 1990). Harvesting should be conducted during the coolest time of the day, which is commonly early in the

morning, and the produce should be kept in a shaded place in the field (Thompson, 1996).

According to Woolfe (1992), sweet potato roots can be stored up to a year if kept under an optimal temperature of 12-15 °C. But temperatures below 12 °C should be avoided as the roots can undergo chilling injury. Low temperature storage can be achieved by use of air-cooled stores, refrigeration and commercial cold storage. However, commercial cold stores are only in operation in the industrialized countries unlike in developing countries where rudimentary storage systems are used. High humidity slows down wilting and Paull, (1999) reported that sweet potato roots require high relative humidity of about 85-90 % in store to reduce moisture loss from the root skin.

Dormancy Control by Plant Growth Regulators

Plant growth regulators are organic substances that may occur endogenously in plant tissues, or they may be exogenously applied as synthetic compounds that mimic endogenous hormones. They influence physiological processes at low concentrations (Davies, 2010). Exogenous hormones are normally applied on the soil in the field or as pre-harvest foliar sprays.

Dormancy initiation process has not been clearly known even though plant growth regulators play a key role in inducing and terminating dormancy. The beginning of dormancy is during tuber initiation where the buds remain dormant and tubers act as sink organs. The existing knowledge about dormancy regulation process in other crops could be applicable to sweet potato (Cheema, 2010). There are five classes of endogenous hormones. They include cytokinins, gibberillins, auxins, abscisic acid and ethylene. The auxins

and gibberillins are classified as regulators of cell division while abscisic acid is an inhibitor. Ethylene, on the other hand, has numerous roles in growth and development processes (Cheema, 2013).

Ethylene

Ethylene is plant growth hormone that occurs naturally and has various effects on the development, growth and storage life of numerous fruit, vegetables and flower yields at low concentrations. It has been used to boost agricultural production through plant growth improvement (Yaseen, Ashraf & Ahmad, 2010). Harvested fruit and vegetables may be purposefully or unexpectedly exposed to natural levels of ethylene; and both endogenous and exogenous sources of ethylene add to its biological action (Saltveit, 1999). It significantly influences almost every growth and development of plants from germination to fruit maturation, ripening and senescence (Arshad & Frankenberger, 2002).

Ethylene has both beneficial and detrimental effects on crops depending on the type of produce, physiological stage of development and ethylene application regime (Abeles, 1969). Abscisic Acid (ABA) is categorized alongside ethylene as a growth inhibitor and is therefore involved in the sprout growth inhibition process. Report by Suttle (2004) indicates that whereas both ABA and ethylene are required to induce dormancy, only ABA is necessary to sustain the dormancy. Reduced ABA concentration leads to dormancy break. The concentration of ABA has been reported to be reliant on storage temperature (Coleman and King, 1984; Chope, Terry & White, 2006).

According to a research by Rylski, Rappaport and Pratt (1974), on potatoes exposed to different ethylene concentrations, it was found that all the

potatoes stored in ethylene experienced sprouting, the rate depending on the ethylene concentration as compared to the untreated control which had very little sprouting. However, according to Daniels-Lake *et al*. (2005), application of continuous ethylene effectively inhibits sprout growth of potatoes during storage, but it also leads to darkening of potato when fried. The ethylene concentrations of 400, 40, and 4 µl L^{-1} suppressed sprout growth successfully, the effects being proportional to the concentration. Sprout mass and greatest sprout length in all ethylene concentrations were essentially lower than the control. A study of the effects of ethylene on onion sprouting by Bufler (2009) showed that exogenous ethylene suppressed the growth of both dormant and already sprouting onion bulbs by inhibiting leaf blade elongation. However, ethylene stimulated CO_2 production of the bulbs by about 2-fold. When sprouting is reduced the produce shelf-life and quality is preserved. This subsequently implies that potato tuber can be kept longer by making use of continuous exposure to ethylene.

In sweet potato, Cheema *et al*. (2013) found that 10 µl L^{-1} ethylene applied on the roots at 25 °C led to a decrease in the amounts of fructose and glucose in two varieties viz. “Bushbuck” and “Ibees” at four weeks in storage compared to the untreated roots. This was associated with higher respiration rates for ethylene treated roots. This is consistent with the activation of some additional process by ethylene which uses energy through sugar metabolism. In his research, Amoah (2014) also found a decline in glucose in roots treated with ethylene compared to the air stored roots. The sucrose concentration was considerably increased in the roots treated with ethylene to about 2-fold in the distal sections after the first two weeks of storage. Following a swap of roots

into ethylene from air, the level of sucrose increased abruptly within two weeks but reduced afterwards.

On the contrary, the level of sucrose reduced in roots swapped into air from ethylene. In contrast, Foukaraki, Chope and Terry (2012) established that exogenous ethylene increases the concentration of sugars in potato. Following 1-Methylcyclopropene (1-MCP) treatment, the tubers were put in trays and stored at 6 °C and continuously supplied with ethylene (10 μl L^{-1}) for 30 weeks. The ethylene-treated tubers that did not get 1-MCP treatment had significantly greater glucose, sucrose and fructose content than those which were treated with 1-MCP. The respiration rates of tubers were significantly higher in both air and ethylene-treated tubers that were treated with 1-MCP.

Exogenous ethylene has recently gained commercial use as a sprouting inhibitor of potato. The sprout inhibition effect of ethylene can provide a viable solution to curb sweet potato sprouting in developing countries (Cheema *et al*., 2013). However, at room temperature ethylene exists as a gas and, therefore, difficult to handle for the treatment of crops. This has limited its use in tropical agriculture. There exists, however, chemical compounds such as ethephon and calcium carbide which serve as precursors to ethylene generation. The role of ethylene in inducing dormancy and sprout inhibition of sweet potato is not fully known. Ethylene is synthesized is through a pathway mediated by amino acid methionine and 1-aminocyclopropane-1-carboxylic acid (Yamamoto *et al*., 1995; Yang & Hoffman, 1984).

The enzymes that catalyze the two reactions in this pathway are ACC synthase and ACC oxidase. The main enzyme in this pathway, ACC synthase, is encoded by multigene families. Environmental and endogenous changes

regulate ethylene biosynthesis mainly through the varied expression of ACC synthase genes (Fluhr, Mattoo & Dilley, 1996). ACC oxidase is coded by small gene families, and there is confirmation of differential regulation of ACC oxidase gene expression (Barry *et al*., 1996; Mita, Kirita, Kato & Hyodo, 1999). Biotechnological alterations of ethylene synthesis and sensitivity of produce to ethylene are promising methods of preventing spoilage of agricultural products such as fruit and vegetables of which ripening and senescence are induced by ethylene (Katz, Lagunes, Riov, Weiss & Goldschmidt, 2004).

Fruit and vegetables are categorized as 'climacteric' or 'non climacteric' based on the incidence or absence of a postharvest upsurge in respiration and the generation of ethylene. The key roles of ethylene in the ripening of climacteric fruit and vegetables have been examined at length for many years (Yang & Hoffman, 1984). The sudden rise in ethylene generation at the initial ripening stage induces changes in flavour, aroma, colour, texture and other biochemical and physiological features. In this research, sweet potatoes were treated with the ethylene precursor, ethephon, at various concentrations and their physiological and biochemical changes were evaluated over the storage time.

Ethephon

Ethephon (2-chloroethylphosphonic acid) is a systemic plant growth and development regulator. It decomposes at a pH of 4 or higher to ethylene, phosphate and chloride particles (Bremer, Norton, Einhorn, Crisosto & Fidelibus, 2008). In a research by Wang, Arancibia, Main, Shankle and LaBonte (2013) progressions in skinning occurrence and content of

lignin/suberin in the skin due to pre-harvest ethephon foliar application was examined. Field-developed "Beauregard" (B-14) sweet potato plots were treated with ethephon at 0.84, 1.68 and 2.52 kg per hectare at 1, 3, and 7 days to harvest. Skinning frequency and severity were reduced with ethephon application in 3 and 7 days before harvesting. The power force needed to skin the storage root was measured at harvest and it increased with ethephon applications at 3 and 7 days before harvest.

Ethephon applications additionally increased cortex phenolics and either diminished or maintained skin phenolic content in the stored roots as compared to defoliated root treatment. Arancibia, Main and Clark (2013) used pre-harvest foliar applications of ethephon to determine the possible relationship of tip rot with ethephon-induced stress. Moreover, the effects of ethephon application rate and duration between application and harvest on tip rot were studied. After 1-2 months in storage, tip rot occurrence was witnessed mostly in stored roots from ethephon-treated foliage. The increase in tip rot incidence was correlated well with ethephon rate.

Research by Crane, Marei and Nelson (1970) on the utilization of 500 ppm ethephon on figs restrained fruit growth and caused abscission of fruits in 5-6 days. Applications of 250-1000 ppm ethephon at an early stage, when breba fruit and leaves were starting to develop were the best treatments in reducing the undesirable breba crop load. The plant growth regulator ethephon has been used to reduce height and lodging of intensively managed wheat (Tripathi *et al*., 2003). In addition, field studies were conducted for two growing seasons of corn to determine the effect of ethephon application on vegetative growth and water use and promote drought stress resistance of corn.

Five ethephon treatments; 0.00, 0.28, 0.56, and 0.84 kg ha^{-1} were applied at the 6 or 8 leaf growth stage. Ethephon application reduced plant height by 10 to 40 % relative to the control and this helped in reducing the lodging of corn (Kasele, Nyirenda, Shanahan, Nielsen & d'Andria, 1994). It also has a strengthening effect on wheat and corn stems; this can also help to reduce firmness loss in sweet potato. From the research, it is evident that, depending on the concentration and time of exposure, ethephon inhibit growth of plants meristematic parts. This effect was studied in this research to determine the possibility of utilizing this effect to reduce sprouting of sweet potato during storage.

Quality Determination Using NIRS

Analysis of biochemical changes in sweet potato was conducted using near infrared spectroscopy (NIRS). NIRS covers the wavelength range adjoining the mid infrared and stretches out to the visible range (Reich, 2005). According to Roggo *et al*. (2007) it covers the transition from the visible spectral range (800-2500 nm) to the mid infrared region (12821-4000 cm^{-1}). NIRS gives a viable method for qualitative and quantitative determination of different properties in numerous fields. This procedure is reliable, quick, accurate and inexpensive in terms of cost-per-test for numerous uses. Additionally, NIRS helps to eliminate sampling errors which arises from manual sample preparation and contaminations by the reagent. Besides, the samples may be reserved for additional analysis. The method can also be used by technically unskilled personnel (Osborne, Fearn & Hindle, 1993). Furthermore, NIR examination can give several results for various constituents concurrently by gathering the NIR spectrum of samples over a range of

wavelengths in a single scan and exploring the spectrum by means equations of calibration. These equations are established through modeling processes, by means of chemometrics which involve the use of sets of samples to train the computer to relate the differences in spectral characteristics to sample constituents (Drennen, Gebhart, Kraemer & Lodder, 1990). It has the ability to determine different compounds in a sample (Pettersson & Aberg, 2003).

Spectra produced by NIR spectroscopy have absorbance bands that are largely from three chemical bonds. They include; C-H bond, typically linked to oil and fats, the N-H bond in protein and the O-H bonds found in water (Cozzolino *et al*., 2008). This means that NIRS can quantify the amount of constituents having diverse molecular structures such as starch, protein or water (Murray and Williams 1990). When the NIR emission is exposed to the sample, it can be absorbed, transmitted or reflected. From Beer-Lambert law, the concentration of the component is directly proportional to the absorbance of the sample (Kandala, Sundaram, Settaluri & Puppala, 2012). Different analysts utilized NIRS to determine other critical compounds, some of which include adulteration in raw milk by Cassoli, Sartori and Machado (2011) and antioxidant action (Chen, Guo, Zhao & Ouyang, 2012).

Research by Lu, Huang & Zhang (2006) indicated that NIRS can be used to test sweet potato physiochemical properties with high accuracy. Outcomes of statistical modeling showed that NIRS was practically accurate in predicting protein content, amylose content, total starch content, phosphorus content among others with high coefficients of determination and reasonably insignificant standard prediction errors.

CHAPTER THREE

METHODOLOGY

Plant Material

Orange-fleshed sweet potatoes locally called *Apomuden* were planted at the University of Cape Coast Research Farm. Cape Coast lies on latitude 05-06 ˚N and longitude 01-15 ˚S at an altitude of 1.1m above sea level. The soil type as classified by Asamoah (1973) is a sandy loam with characteristics as neutral to slightly acid and with a pH of 6.5. This site lies within the coastal savannah vegetation of Ghana. The annual temperature is 30 °C–34 °C during the day and 22 °C–24 °C during the night and a relative humidity of 75–79 %. The sweet potato grown was meant to ensure the providence of samples for the research and enable appropriate control of the treatments.

Standard agronomic practices including weeding were also effectively and timely done. An area of 400 m^2 was ploughed, followed by harrowing and ridging. Ridges were spaced at 70 cm intervals and made to 30 cm high and 45 cm at the base and vine cuttings planted at 30 cm intervals. Disease-free vines were planted at an angle in the rows at a depth of about 5 cm with no less than 2 plant nodes in the ground and leaving at least 2 leaves above the ground.

In the postharvest dip experiment, orange-fleshed sweet potatoes *Apomuden* were bought from a farm under WAPP project as the vine multiplication centre for Cape Coast. The roots purchased were fairly uniform in size and disease-free. Sorting was done to obtain mature and wholesome roots for curing, treatment and storage.

Field Experiment Design

The field experiment was a Randomized Complete Block Design (RCBD). The area of 400 m^2 was divided into four blocks each with four plots (5 m x 4 m). The treatments, ethephon as foliar application at 0 ppm (E0), 100 ppm (E1), 500 ppm (E2) and 1000 ppm (E3), were randomized within each block. The roots were then harvested 7 days later and stored in a dark room.

Weed and Pest Control

Field inspection before planting and after planting was conducted to ascertain that a weed free field was used for planting. First weeding was done three weeks after planting. The field was weeded thrice before harvesting. Field hygiene was maintained to reduce incidence of pests and diseases. This was conducted to coincide with the time when pests and diseases are most likely to attack.

Ethephon Application

Ethephon at the four concentrations as stated above was applied on four randomly selected plots in every block 7 days before harvesting. The ethephon mix was prepared by mixing 0.0, 2.0, 10.0 and 20 ml of 480 SL Chemophon stock solutions in 10 litres of water in a knapsack sprayer and thoroughly mixed. Chemophon is a commercial name of ethephon, a systemic plant growth regulator used to generate ethylene to regulate certain growth processes in various crops like cotton, rubber, pineapples tobacco and crops. It is manufactured by Volcano Agro Sciences limited company, South Africa and distributed by Chemico Limited Company, Ghana. The following dilution equation was used to determine the various volumes of the stock solutions used as shown in Equation 1.

$$C_1VV_1 = C_2V_2 \qquad (1)$$

where C_1 and V_1 were the stock solution concentration and volume, respectively and C_2 and V_2 were the concentration and volume required, respectively. The mixture was then used to uniformly spray the selected plots ensuring that each plant leave canopy was sufficiently soaked. The adjacent plots were covered with a polythene sheet to avoid any spray spills on the other plots. Spraying was done in the morning when the weather was calm and cloudy (Fig. 8).

Figure 8: Foliar ethephon application

Harvesting

Before harvesting, the field was irrigated to soften the soil and reduce surface injuries and cuts. The roots were then carefully harvested using hand hoes. The roots were gently handled to reduce injuries. Harvesting was done early in the morning to reduce field heat. Sorting was done to obtain mature and wholesome roots of nearly uniform tuber sizes for curing and storage.

Curing

Selected wholesome roots were cured to heal cuts and reduce decay and shrinkage of sweet potato in storage because it allows the periderm to

thicken and to reform. Curing was done 2 h after harvesting and continued for 6 days at 29 to 30 ^{0}Cand at 85 % relative humidity with ample ventilation (Edmunds *et al*., 2008). Curing was done by digging shallow pits and laying dry grass on the ground before heaping the roots in a conical shape. The roots were then covered with another layer of dry grass (Fig. 9). The daily temperature and relative humidity of the curing pits were monitored and recorded for the curing period. The relative humidity was raised by sprinkling some water on the grass.

Figure 9: Curing process in a shallow pit

Postharvest Ethephon Application

The roots were dipped in ethephon at levels 0 ppm, 100 ppm, 500 ppm and 1000 ppm for 2 minutes. The ethephon mix was prepared as described for the preharvest foliar spray. This was applied 7 days after harvesting.

Experimental Design for Storage

A total of 480 wholesome roots were selected from the cured roots and used for biochemical destructive tests and another 240 roots for non-destructive physiological tests. There were four replications per treatment. Each replicate contained 15 non-destructive roots and 30 roots for destructive

biochemical analysis. From each replicate, six (6) roots were randomly selected at each sampling event for the destructive assays on weekly periodic intervals until the end of storage. The postharvest ethephon-dipped roots were analyzed for physiological changes only with a total of 120 wholesome roots. There were four (4) treatment levels of ethephon as described before with three 3 replications per treatment. Each replicate contained 10 roots (n=10).

Storage

The roots were stored in sterilized well ventilated baskets in the ambient environment in a dark storage room (Fig. 10). Light was excluded from the store by use of black blinders on the windows. The window louvers were opened for good ventilation and fitted with wire mesh to make it vermin proof. The storage temperature and relative humidity were monitored and recorded. Physiological changes in the roots were monitored over a period of 8 weeks. These included percentage weight loss, sprouting index, number and maximum length of sprouts per root, percentage root decay, shrinkage and pathogen isolation and identification. In addition, roots treated with preharvest ethephon spray were sampled from each replicate for NIRS assays of the nutrients and mineral content.

Figure 10: Stored roots in well ventilated storage baskets

Determination of Percentage Weight Loss

The non-destructive roots were individually numbered and their weights were recorded repeatedly at irregular intervals using an electronic weighing balance (Germany, Model: FX-3000i WP) with accuracy 0.01 g. Weight loss was then determined by calculating the difference between the initial and final weights. This was then expressed as the percentage weight loss as shown in Equation 2.

$$\text{Weight loss } (\%) = \frac{\text{Initial weight} - \text{final weight}}{\text{Initial weight}} \times 100\ \% \qquad (2)$$

Sprouting Index of Roots

Sweet potato sprouting is one of the main features used to assess the physiological quality of harvested produce. In postharvest, a sprout is a defect and a lot is considered as having sprouted if 10 % of the roots have sprouts greater than 19 mm (EAC, 2010). The sweet potato roots were assessed for the number of sprouted roots, the mean number of sprouted buds per root and the maximum sprout length (mm) per root. All these were observed in the 240 non-destructive sub-samples. For the purpose of assessing the physiological activity in the roots, any bud growth up to 1 mm or more was counted as a sprout. The sprouting index was then calculated using the formula by Obetta, Ijabo and Satimehin (2007) as shown in Equation 3.

$$\text{Sprouting Index} = \frac{\text{Number of sprouted roots}}{\text{Total number of roots}} \times 100\ \% \qquad (3)$$

Root Decay Incidence and Mode of Infection of Stored Roots

Percentage root decay was determined through visual observation of roots for decay. Rot severity assessment was done based on a scale of 1–5 being 1-0 %, 2–25 %, 3–50 %, 4–75 %, and 5–100 %, respectively. Roots

showing extensive decay and unmarketable were removed from the store and the weight of the remaining roots was calculated as described by Rees *et al.* (2003). At every assessment, the diseased roots (10 % decayed surface) in the non-destructive roots were counted and removed. The mean number of decayed roots per treatment was then calculated.

Shrinkage

Shrinkage of the roots was determined by measuring the mean diameter of the roots with a Vernier caliper at the start of the study and later at irregular intervals for the entire storage period. The diameter measuring points at the start was marked with a permanent marker and used as the subsequent measuring point (Fig. 11). The difference in the initial and final diameter was used to calculate the percentage root shrinkage as shown in Equation 4.

$$\text{Root shrinkage } (\%) = \frac{D_0 - D_1}{D_0} \times 100\ \% \qquad (4)$$

Where; D_0 is initial diameter and D_1 is the subsequent diameters at different time of storage.

Figure 11: Shrinkage determination by use of a digital Vernier (accuracy 0.01)

Isolation, Culturing and Identification of the Decay Causing Pathogens

The root regions of the initiation of infection by microorganisms were noted. Pieces of the diseased parts were cut from the periphery of rotten sweet potato roots with a sterilized knife. They were then surface-sterilized in 5 % sodium hypochlorite solution for 5 min. The surface sterilized diseased tissues were washed three times in sterilized distilled water. The tissues were then covered and allowed to dry. The dried samples were then plated on potato dextrose agar (PDA) medium (Fig. 12).

Four days after isolation and incubation, mycelia that grew from the plated OFSP parts were sub-cultured onto fresh PDA. Additional sub-culturing was carried out until a pure culture of single species isolates was obtained. The pure cultures were then observed under the microscope (×40). Their characteristics were used to identify the fungal organisms to the species level, as described by Mathur and Kongsdal (2003).

Figure 12: Pathogen isolation (A) and cultured pathogen (B)

Biochemical Changes Determination

The quality of agricultural produce can be determined by conventional chemical tests; however, this method can be time consuming and sometimes results in inaccurate results if not carefully done. Recent advances in quality

determination using NIRS has made it possible for quick and precise quality tests to be performed in many agricultural products. NIRS is a quicker, accurate and non-destructive technique that is currently used for the quality analysis of many crops including cocoa, rice and sweet potato. In this study, the biochemical changes in the sweet potato over the storage period were determined using NIRS at the Sweet Potato Research Laboratory of the Crops Research Institute, Kumasi.

Sample Preparation for NIRS Scanning

The roots were thoroughly cleaned in running tap water and rinsed with deionized water before peeling. This was to ensure that all contaminants were removed. The roots were then dried with soft towel. Peeling was done using sterilized stainless steel knives to avoid contaminations of the samples. The peeled samples were sliced into small chips, weighed (Adventure Pro, USA, Model: AV812,) as fresh weight (FW), immediately put in pre-weighed zip-lock bags, sealed and put in a deep freezer (Model: MFC25V7GW) at -25 °C before freeze drying.

The sweet potato samples were freeze-dried for 72 h in a vacuum freeze dryer (True Ten Industrial Co. Ltd, Taiwan, YK-118) to ensure that all samples were dehydrated before milling. Freeze dried products preserve purity, maximum nutrient and colour retention and low enzymatic activity. In the vacuum freeze dryer, moisture sublimes directly from the solid state to vapour. The process freezes the water in the samples into solid form which is then stored in a vacuum sealed space. After lyophilization, the samples were reweighed (dry weight, DW) and the moisture content (wet basis) calculated as in Equation 5.

$$M_{wb} = \frac{FW - DW}{FW} \times 100\ \% \qquad (5)$$

Milling

The dried samples were milled using an electric powered mill (Thomas Scientific-800-345-2100). The milled samples were then sieved using an impact lab test sieve of pore size 400/425 microns (Model BS410 – 1:2000), bagged in zip-lock bags and stored in a commercial freezer (-25 °C) prior to other laboratory analysis.

NIRS Analysis

The frozen samples were removed from the deep freezer and allowed to attain room temperature before analysis. The powder was scooped into cuvettes and covered, ready for scanning. Scanning was done using NIRS XDS Rapid Content Analyzer machine (Metrohm, Switzerland) calibrated for sweet potato quality test at the Postharvest NIR Laboratory of the Crops Research Institute in Kumasi, Ghana.

Spectral data were collected by measurement of diffuse reflectance from the sample in the NIR region within 1100–2500 nm using a model Foss XM-1000 monochromator equipped with a transport module (FOSS NIRS systems, Silver Springs, MD, USA) (Fig. 13). Each sweet potato sample was scanned in triplicate in a small ring quartz window-clad cylindrical cell (NR-7073, internal diameter 35 mm, depth 9 mm). The room was kept at 25 °C during the NIR analysis. The spectroscopic procedures and data recording were done using Win ISI II software (version 1.04, FOSS NIRS systems Silver Springs, MD, USA).

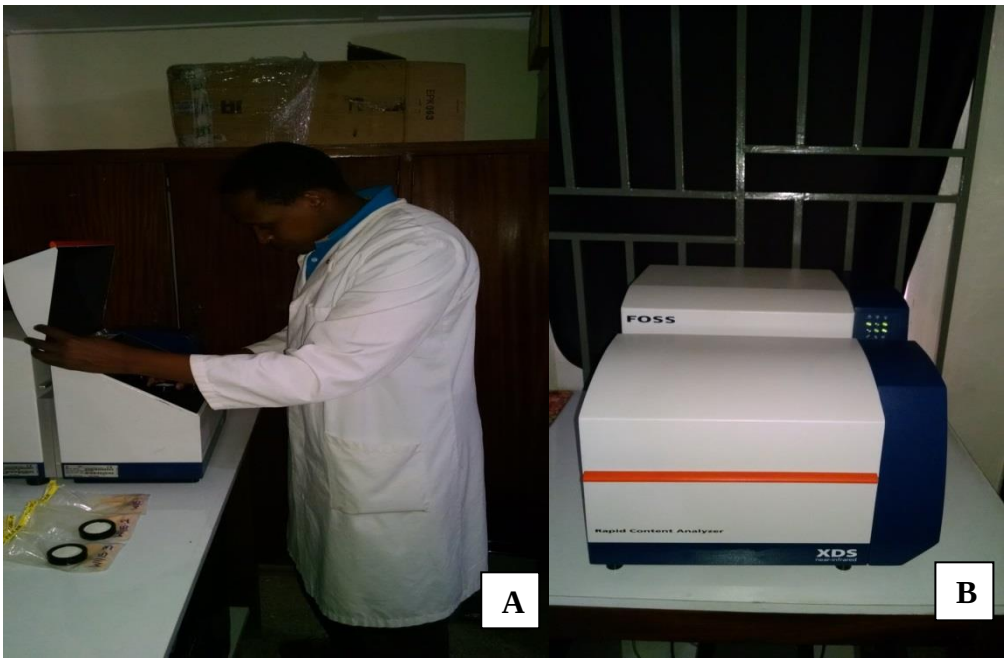

Figure 13: Biochemical analysis (A) and the NIRS Analyzer (B)

The biochemical data was collected over the entire storage period at irregular intervals. Biochemical changes data collected include: starch, sucrose, glucose, fructose, minerals (Zn, Fe) and proteins.

Statistical Analysis

The data was subjected to Shipiro-Wilk normality test and plotted for residuals to attest assumptions for the Analysis of Variance. The ANOVA was generated using GenStat version 10.1 for Windows based on RCBD for preharvest application and CRD for post-harvest application. The treatments exhibiting significant difference were further subjected to Duncan Multiple Range Test (DMRT) for comparison of their means at significance level ($p < 0.05$).

Chapter Summary

All methods used were described appropriately and the concerned sources acknowledged and referenced well. All the materials used in this

study were described and where applicable figures have been provided. The experimental designs used were RCBD for the first experiment while CRD was used in the second experiment. All the statistical analysis tools and data presentation have been clearly explained. The post-harvest experiment was carried out as a separate research from the preharvest, thus the experiments are separate experiments and comparison between the two applications regimes may vary slightly. However, the roots were sourced from the same farmer and were from the same variety.

CHAPTER FOUR

RESULTS AND DISCUSSION

Results

Effects of Pre Harvest Ethephon Application on Sweet potato Morphology

Foliar ethephon application had significant effects on the OFSP both in the field and during storage of the harvested roots. The ethylene precursor had a yellowing effect on the leaves in the field (Figure 14). The yellowing was more pronounced in the highest concentration. Leaves abscission was observed from the third day after ethephon application.

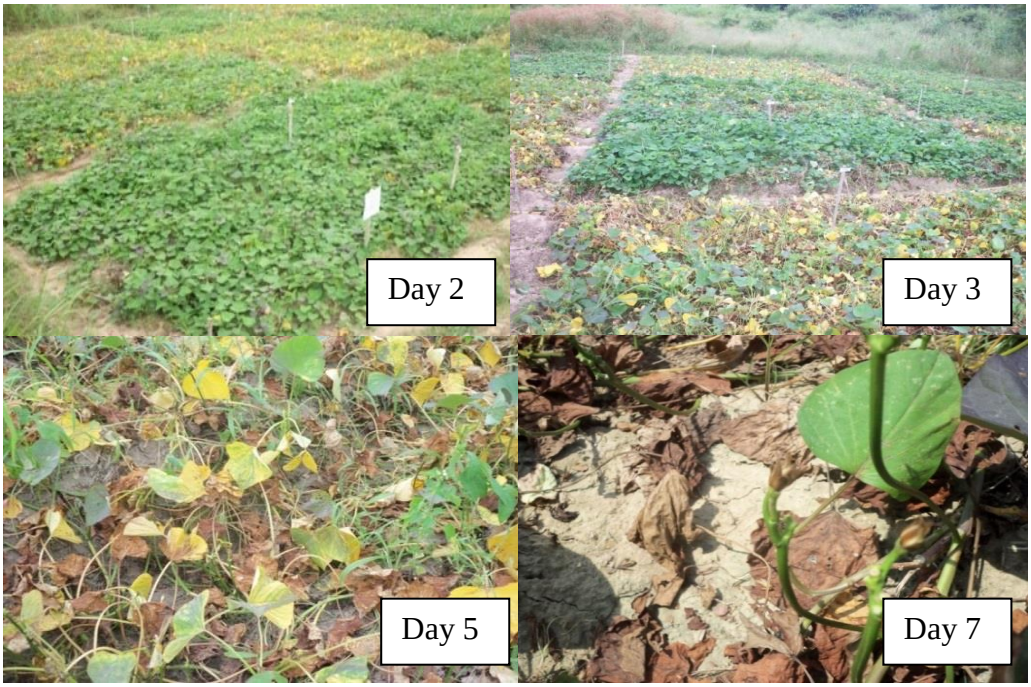

Figure 14: Effect of foliar spray of ethephon on the leaves and vines 2, 3, 5 and 7 days after application

Storage Temperature and Relative Humidity (RH)

The storage conditions recorded throughout the storage period was a mean temperature of 26.6 °C and RH of 89 % (Fig. 15). The conditions were

controlled to mimic market and farm storage conditions to which the roots are currently subjected in the marketing chain in Ghana.

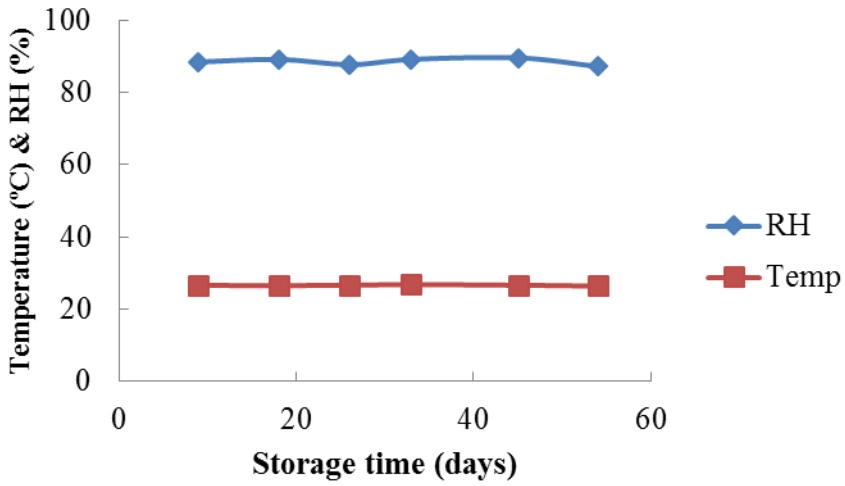

***Figure 15**:* Average temperature (°C) and relative humidity (%) of the store.

Percentage Weight Loss

All levels of ethephon pretreatment significantly increased the weight loss of the roots (p = 0.001) (Table A1). Among the different concentrations of ethephon, roots treated with 1000 ppm (E3) suffered the highest mean weight loss of about 8.3 % (1.6 times the control) followed by E2, E1 and E0 which had mean weight losses of 6.8, 6.2 and 5.2 %, respectively. For all the ethephon levels, the percentage mean weight losses were significantly different from each other (Table 2). The rate of weight loss in all the treatments followed a steady rising trend for the entire storage period (Fig. 16).

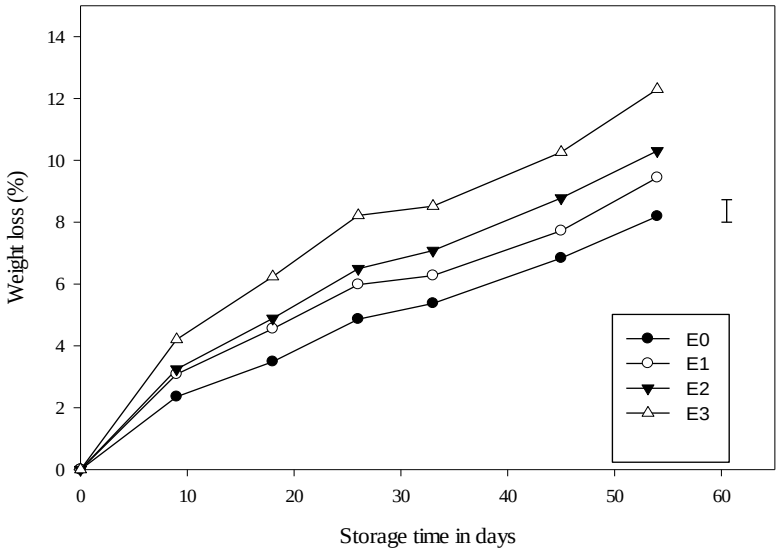

Figure 16: Percentage weight loss OFSP *Apomuden* roots

Treated 7 days before harvesting with ethephon spray and stored at mean temperature and relative humidity of 26.3 °C and 88.7 %, respectively. Every data point is the mean of four treatment baskets containing 15 roots per basket (n=60). The LSD bar shows treatment-days interaction effect. ANOVA Table A1.

Percentage Moisture Content

Except E3, the other ethephon levels significantly reduced the percentage moisture of the roots (p = 0.008) (Table A2). The initial moisture contents were 80.27 % and 80.20 % in the control and treated root samples, respectively. The mean moisture content changes for the various sampling days are shown in Figure 17. The LSD test of interactive effects of different levels ethephon on moisture content of OFSP roots is shown in Table 2. The control had the highest final mean moisture content (78.5 %) followed by E3 with 77.8 % though the difference was not significant. E1 and E2 each had mean moisture content of 77.3 and 77.2 %, respectively.

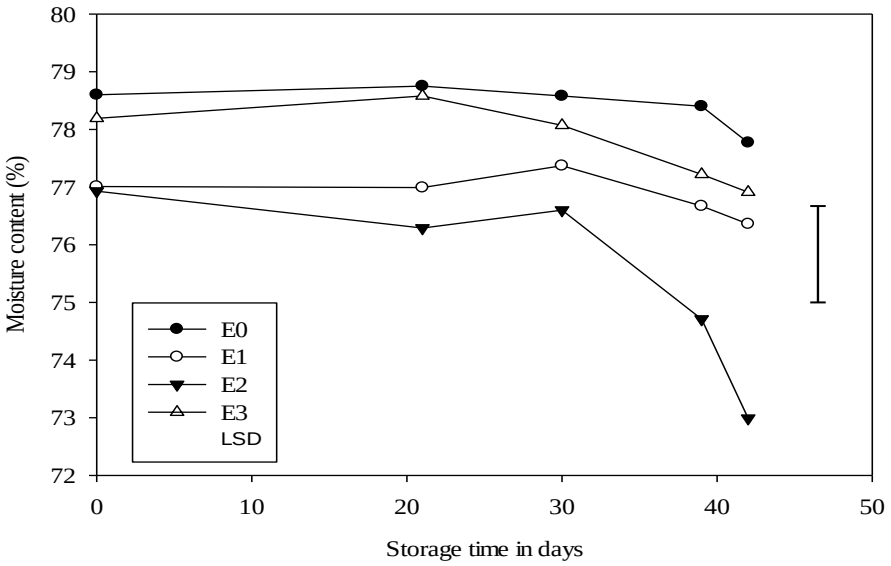

Figure 17: Percentage moisture content of OFSP *Apomuden* roots

Treated 7 days before harvesting with ethephon spray and stored at mean temperature and relative humidity of 26.3 °C and 88.7 %, respectively. Every data point is the mean of four treatment baskets containing 15 roots per basket (n=60). The LSD bar shows treatment-days interaction effect. ANOVA Table A2.

Percentage Shrinkage

All ethephon levels and the control had significantly higher root shrinkage except E1 (p = 0.001) as shown in Table A9. Figure 18 shows the effect of ethephon pre-treatment on the shrinkage of the roots. Highest percentage shrinkage of 3.2 % was observed in E3 while E1 had the lowest shrinkage of 2.35 % at the end of the storage. E0 and E2 each had 2.7 % and 3.1 % shrinkage, respectively. The mean percentage shrinkage for the entire storage time was 1.7 %, 1.4 %, 1.8 %, 1.9 % for E0, E1, E2 and E3, respectively. There was no significant difference in the mean percentage

shrinkage between E3, E2 and E0 while E1was significantly lower than the rest of the treatments (Table 2).

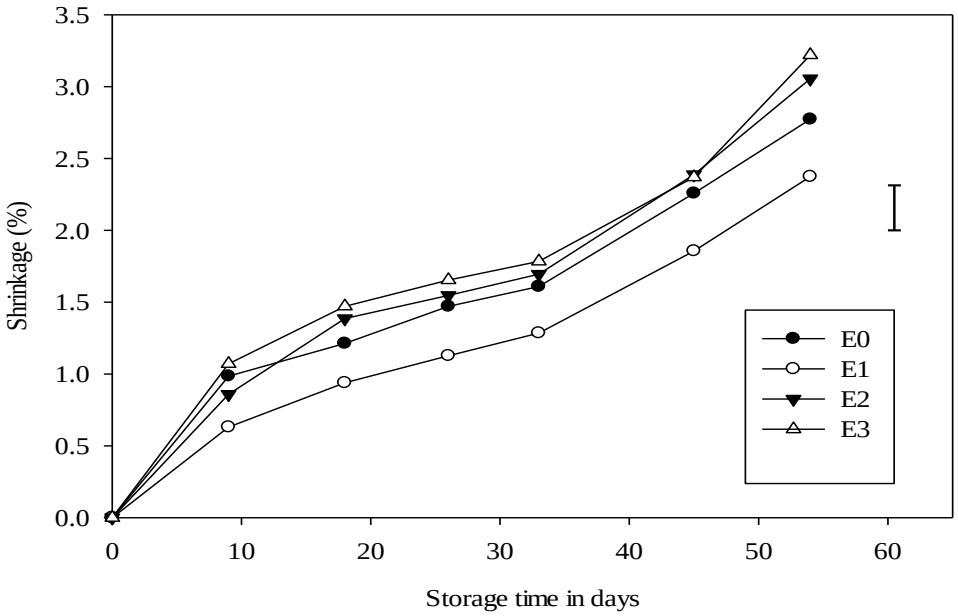

***Figure 18**:* Average percentage shrinkage of OFSP *Apomuden* roots

Treated 7 days before harvesting with ethephon spray and stored at mean temperature and relative humidity of 26.3 °C and 88.7 %, respectively. Every data point is the mean of four treatment baskets containing 15 roots per basket (n=60). The LSD bar shows treatment-days interaction effect. ANOVA Table A8.

Table 2: Treatment Mean Weight Loss, Moisture Content and Shrinkage of OFSP Roots treated 7 days before harvesting

Treatment (ppm)	**Weight loss** (%)	**MC (%)**	**Shrinkage (%)**
E0 (0)	5.18^{d}±1.04	78.42^{a}±13.08	1.72^{a}±0.34
E1 (100)	6.17^{c}±1.18	76.88^{b}±12.86	1.37^{b}±0.30
E2 (500)	6.80^{b}±1.31	75.50^{c}±12.89	1.82^{a}±0.38
E3 (1000)	8.29^{a}±1.54	77.79^{a}±12.97	1.93^{a}±0.38
LSD	0.54	0.782	0.25

Values are means of 240 determinations. Means with same letters within a column are not significantly different ($p < 0.05$).

Sprouting Index, Length and Number of Sprouts

Three sprout growth parameters; percentage of the sprouted roots, the mean number of sprouts per root and the maximum sprout length per root were measured for each treatment. The dormancy break was recorded when 10 % of roots had ≥1 mm sprouted eyes. From data taken from individual roots, dormancy break occurred after approximately day 27, 20, 6 and 11 for E0, E1, E2 and E3, respectively.

Sprouting Index (SI)

There was no significant effect of all levels of ethephon on the sprouting index (SI) of the stored roots (p = 0.09) as shown in Table A4. Throughout the entire storage period, sprouting index for E3 ranged between 21.7-28.3 % while that for E2 ranged between 40.0-53.3 %. The sprouting index for E2 increased steadily up to 51.7 % on the 27^{th} day of storage and later rose to 53.3 % at the end of the storage period. Sprouting index in E3 treated roots declined from 31.7 to 28.3 % at the end of storage period (Fig. 19).

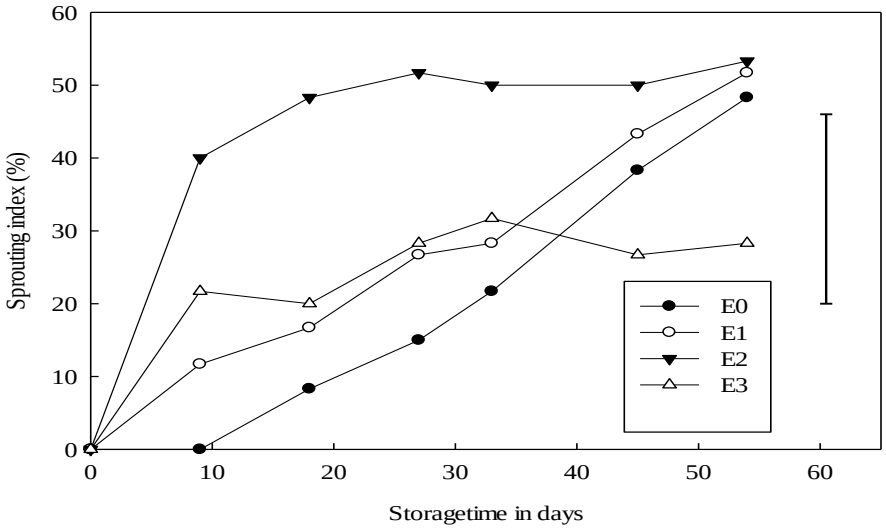

***Figure 19**:* Sprouting index (%) of the roots

Roots were treated 7 days before harvesting. Every data point is the mean of four treatment baskets containing 15 roots per basket (n=60). The LSD bar shows treatment-days interaction effect. ANOVA Table A3. Length of the sprouts Ethephon level E2 significantly increased the length of sprouts (p = 0.006) as shown in Table A5. The mean sprout lengths for the other levels of ethephon and the control were not significantly different from each other (Table 3). The mean maximum sprout lengths were observed in E2 at 18.5 mm followed by E3, E1 and E0 which had mean sprout lengths of 12.9, 11.1, 9.8 mm, respectively. The longest sprouts in individual roots at the end of the study period were 280, 260, 226 and 154 mm and they were recorded in E0, E1, E3 and E2, respectively. The maximum sprout lengths measured at different storage days are plotted in Figure 20.

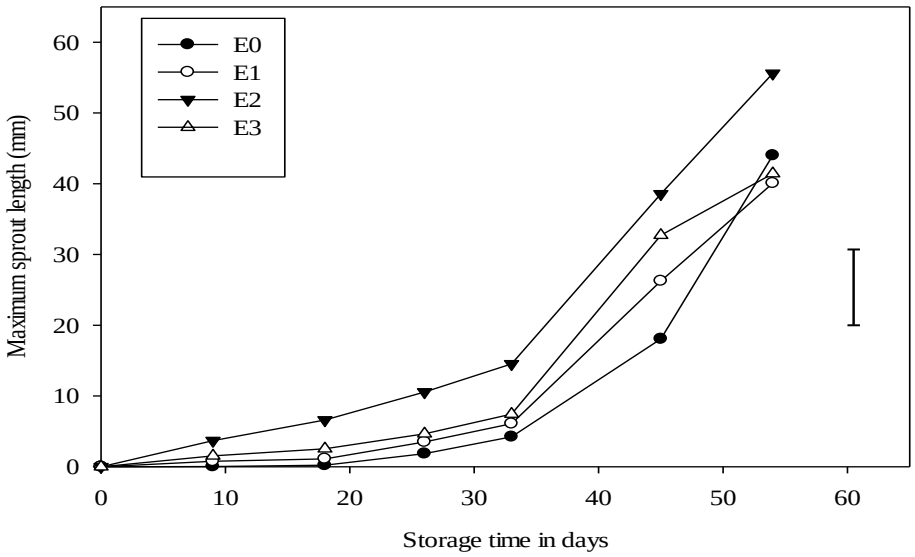

Figure 20: Average maximum sprout lengths (mm) of the OFSP *Apomuden* roots. Every data point is the mean of four treatment baskets containing 15 roots per basket (n=60). The LSD bar shows treatment-days interaction effect. ANOVA Table A4.

Number of Sprouts per Root

All ethephon levels significantly increased the number of sprouts per root except E1 (p = 0.001) as shown in Table A6. The highest mean number of sprouts per root was recorded in E2 at 2.1 followed by E3, E1 and E0 each of which had 1.5, 1.2 and 0.8 sprouts, respectively (Fig. 21). Compared to control, the higher concentrations of ethephon treatment (E2 and E3) increased the number of sprouted eyes per root. The mean number of sprouts for all the levels of ethephon was significantly different from one another except for treatments E1 which was not significantly different from the control (Table 3).

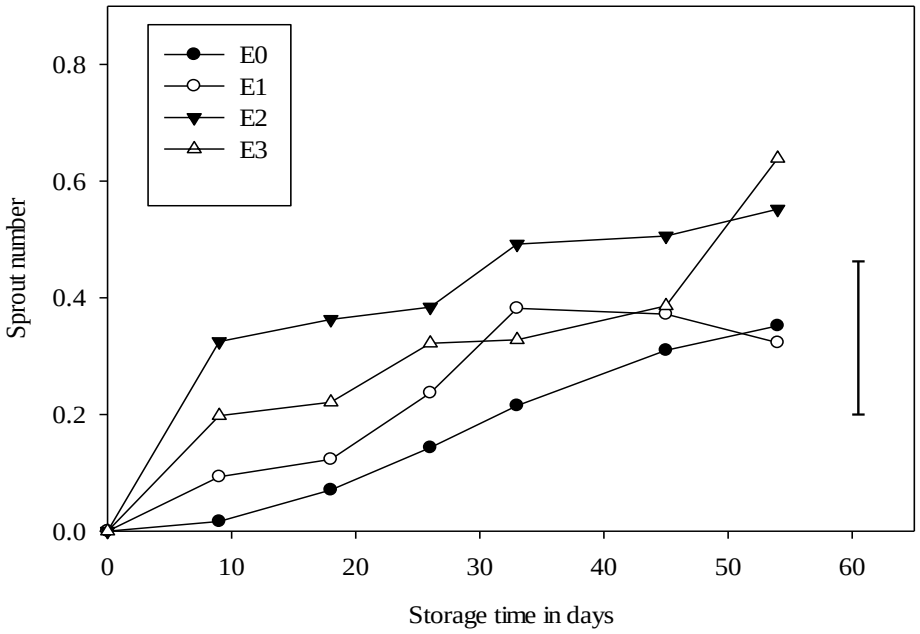

Figure 21*:* Number sprouts of the OFSP *Apomuden* roots. The roots were treated 7 days before harvesting with ethephon spray and stored at mean temperature and relative humidity of 26.3 °C and 88.7 %, respectively. Every data point is the mean of four treatment baskets containing 15 roots per basket (n=60). The LSD bar shows treatment-days interaction effect. ANOVA Table A5.

Incidence of Decay

Figure 22 shows the percentage root decay of the root decay over the storage time. Ethephon level E3 significantly increased root decay (p = 0.01) as shown in Table A7. Incidence of decay was first observed in the second week of storage for E3. The treatment E3 had the highest percentage root decay of 70 % while E0 had the lowest decay of 33 % at the end of storage period. The highest overall mean root decay was observed in E3 with 45.3 % decay while E1, E2 and E0 had 28.1, 21.9 and 14.4 % decay, respectively (Table 3).

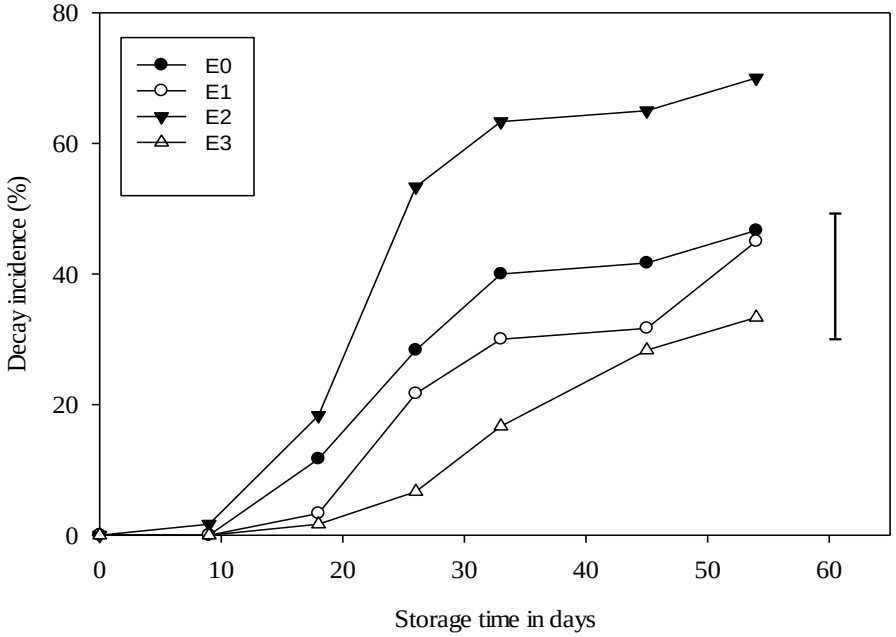

Figure 22: Percentage decay of the OFSP *Apomuden* roots. The roots were treated 7 days before harvesting with ethephon spray and stored at mean temperature and relative humidity of 26.3 °C and 88.7 %, respectively. Every data point is the mean of four treatment baskets containing 15 roots per basket (n=60). The LSD bar shows treatment-days interaction effect. ANOVA Table A6.

Severity of Decay

Ethephon E3 had significantly higher severity of root decay (p = 0.001) as shown in Table A7. The severity trend assessed at different storage time is shown in Figure 23. The highest concentration (E3) of foliar ethephon application increased the severity of root decay compared to the control with a mean severity of decay at 2.3 which was significantly different from E1, E2 and E0 which had 1.7, 1.6 and 1.4, respectively (Table 3). The decay was predominant at the proximal ends for all the decayed roots treated with the three levels of ethephon and the control (Figure 24).

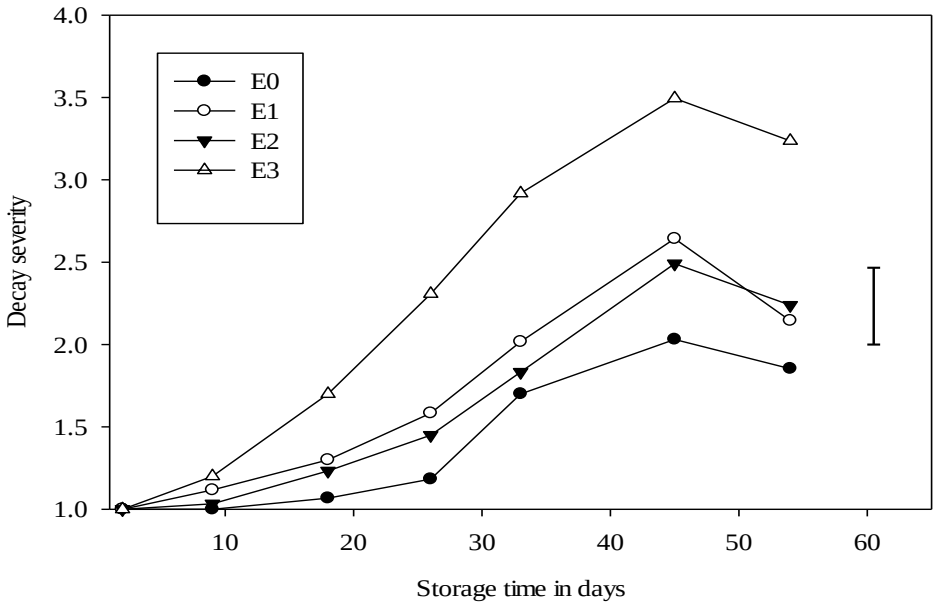

Figure 23*:* Effects of ethephon on severity of decay of OFSP *Apomuden* roots. The roots were treated 7 days before harvesting with ethephon spray and stored at mean temperature and relative humidity of 26.3 °C and 88.7 %, respectively. Every data point is the mean of four treatment baskets containing 15 roots per basket (n=60). The LSD bar shows treatment-days interaction effect. ANOVA Table A7.

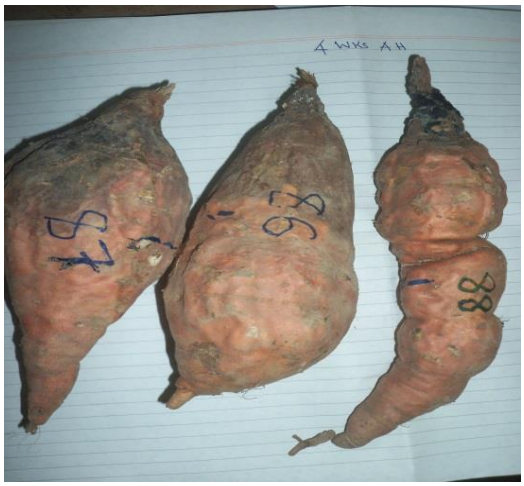

Figure 24: Proximal decay of roots from treatment E1 four weeks after harvesting.

Table 3: Mean Sprout (length, number and index), Decay Incidence and Severity of OFSP Roots

Treatment (ppm)	Sprout length (mm)	Sprout number	Sprouting Index (%)	Decay incidence (%)	Decay severity
E0 (0)	9.77^{b}±6.20	0.81^{c}±0.37	18.8^{a}±7.06	14.44^{b}±5.26	1.41^{c}±0.23
E1 (100)	11.11^{b}±5.94	1.24b^{c}±0.47	25.5^{a}±6.78	28.06^{b}±7.57	1.69^{b}±0.29
E2 (500)	18.50^{a}±7.81	2.07^{a}±0.48	41.9^{a}±7.17	21.94^{b}±6.78	1.61^{bc}±0.28
E3 (1000)	12.90^{b}±6.38	1.48^{b}±0.49	22.4^{a}±4.03	45.28^{a}±11.73	2.27^{a}±0.43
LSD	5.17	0.45	26	15.61	0.27

Values are means of 240 determinations. Means with same letters within a column are not significantly different ($p < 0.05$).

Experiment 2: Postharvest Ethephon Application

Postharvest ethephon application had significant effect only on the shrinkage but no significant effect was noticed on sprouting, decay and weight loss of the OFSP during storage. However, the absolute means were different for the different levels of ethephon and control (Table 4).

Physiological Weight Loss

The postharvest dip in ethephon had no significant effect on the weight loss of the roots ($p = 0.282$) (Table B1). However, at the end of the storage period treatment E3 caused the highest mean weight loss of about 14.6 % followed by E0, E1 and E2 with weight losses of 13.8, 13.2 and 13.1 %, respectively (Table 5). The rate of weight loss was steady for the control roots and treated roots for the entire storage period (Fig. 25).

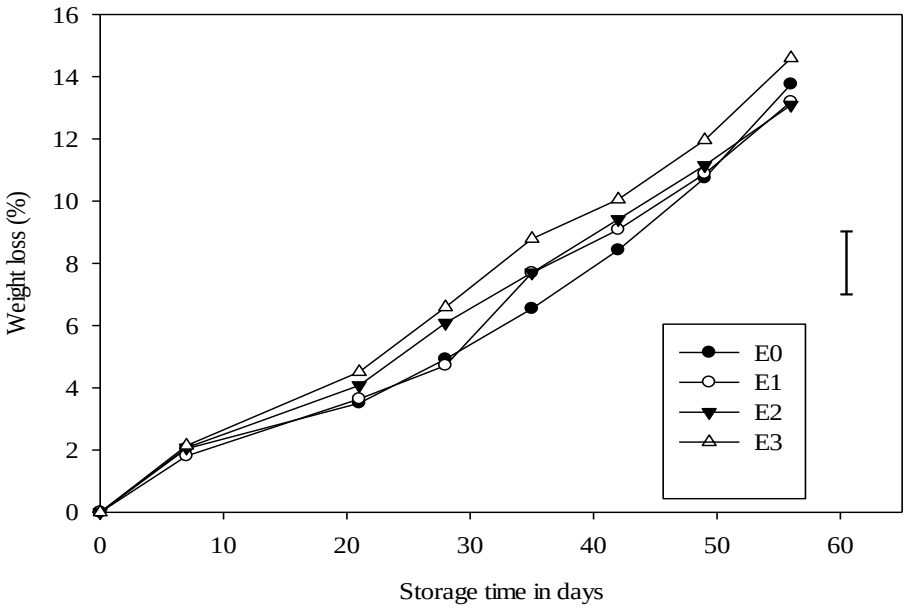

Figure 25: Percentage weight loss of OFSP *Apomuden* roots

Treated 7 days after harvesting with ethephon dip and stored at mean temperature and relative humidity of 26.3 °C and 88.7 %, respectively. Every data point is the mean of three treatment baskets containing 10 roots per basket (n=30). The LSD bar shows treatment-days interaction effect. ANOVA Table B1.

Percentage Shrinkage

Figure 26 shows the effect of postharvest ethephon dip on the shrinkage of the roots. Ethephon had a significant effect on roots shrinkage (p = 0.001) (Table B7). The control (E0) had the highest mean percentage shrinkage of 8.8 % while E2 had the lowest shrinkage of 6.4 % at the end of the storage. Treatment E1 and E3 each had 6.6 and 7.4 % shrinkage, respectively (Table 4).

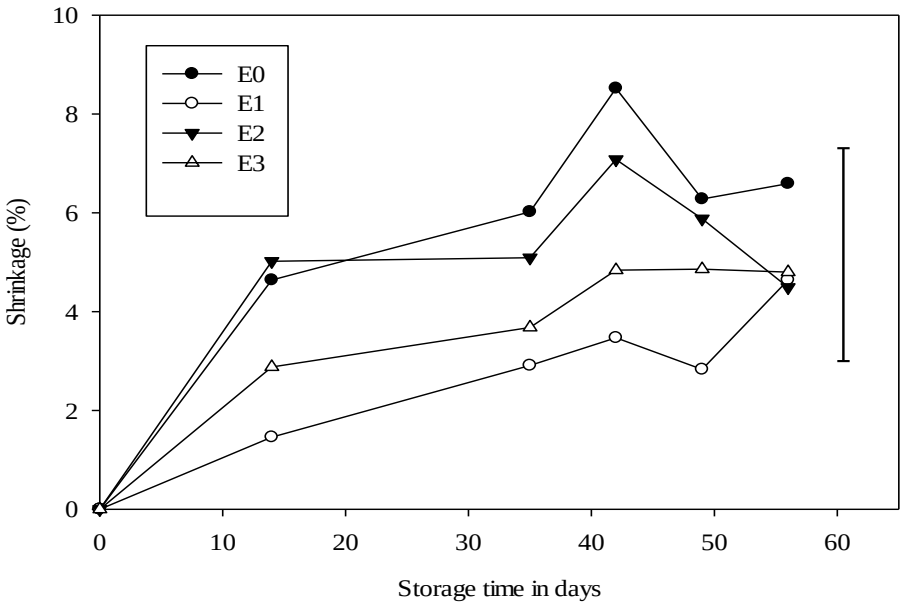

Figure 26: Average percentage shrinkage of OFSP *Apomuden* roots

Treated 7 days after harvesting with ethephon dip and stored at mean temperature and relative humidity of 26.3 °C and 88.7 %, respectively. Every data point is the mean of three treatment baskets containing 10 roots per basket (n=30). The LSD bar shows treatment-days interaction effect. ANOVA Table B7.

Table 4: Mean Shrinkage of OFSP Roots Treated 7 Days after Harvesting

Treatment (ppm)	**Shrinkage (%)**
E0 (0)	8.75^{a}±0.51
E1 (100)	6.63^{b}±0.22
E2 (500)	6.38^{b}±0.39
E3 (1000)	7.44^{a}±0.32
$LSD_{0.05}$	2.14

Values are means of 240 determinations. Means with same letters within a column are not significantly different ($p < 0.05$).

Sprouting Index, Length and Number of Sprouts

Three sprout growth parameters; percentage of the sprouted roots; the mean number of sprouts per root and the maximum sprout length per root were measured for each treatment. The dormancy break was recorded when 10 % of roots had ≥1 mm sprouted eyes.

Sprouting Index

There was no significant effect of ethephon on the sprouting index of the treated roots (p = 0.205) (Table B2). However, from the absolute values E0 had the highest sprouting index of 47.1 % (Table 5). The roots exhibited a steady rise in the sprouting index (Fig. 27). From the absolute values, the highest mean sprouting index was observed in the control (57.5 %) while the lowest was in E2 (47.0 %).

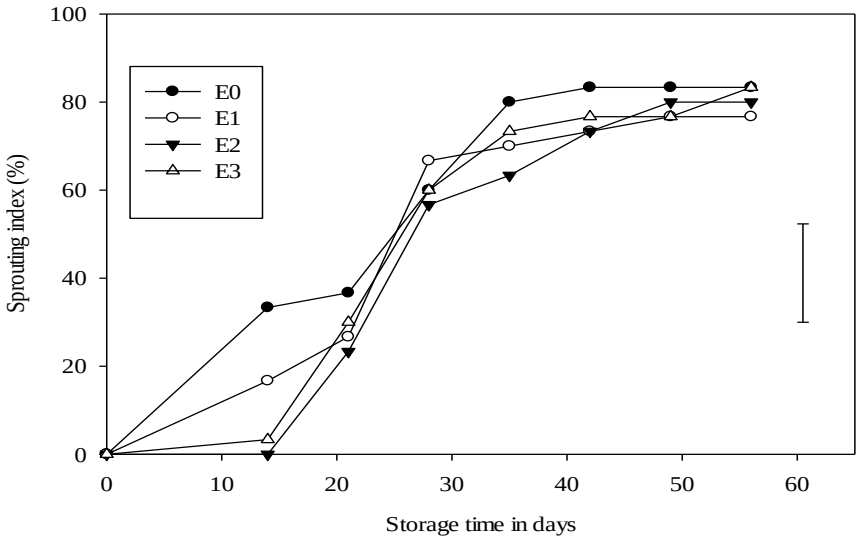

Figure 27: Average sprouting index of OFSP *Apomuden* roots. Treated 7 days after harvesting with ethephon dip and stored at mean temperature and relative humidity of 26.3 °C and 88.7 %, respectively. Every data point is the mean of three treatment baskets containing 10 roots per basket (n=30). The LSD bar shows treatment-days interaction effect. ANOVA Table B2.

Maximum Length of Sprouts

Ethephon had no significant effect (p = 0.613) on the length of sprouts (Table B3). The highest mean maximum sprout lengths were observed in E1 at 18.8 mm followed by E3, E2 and E0 which had 18.0, 16.1, 15.5 mm, respectively (Table 5). In the individual roots, the longest sprouts at different evaluation periods were 210, 165, 90 60, 50, 23 and 20 mm and they were recorded in treatments E1, E1, E3, E0, E0, E1 and E0 on day 56, 49, 42, 35, 28, 21 and 7, respectively. The mean maximum sprout lengths for the various days rose steadily in both the control and treated roots (Fig. 28).

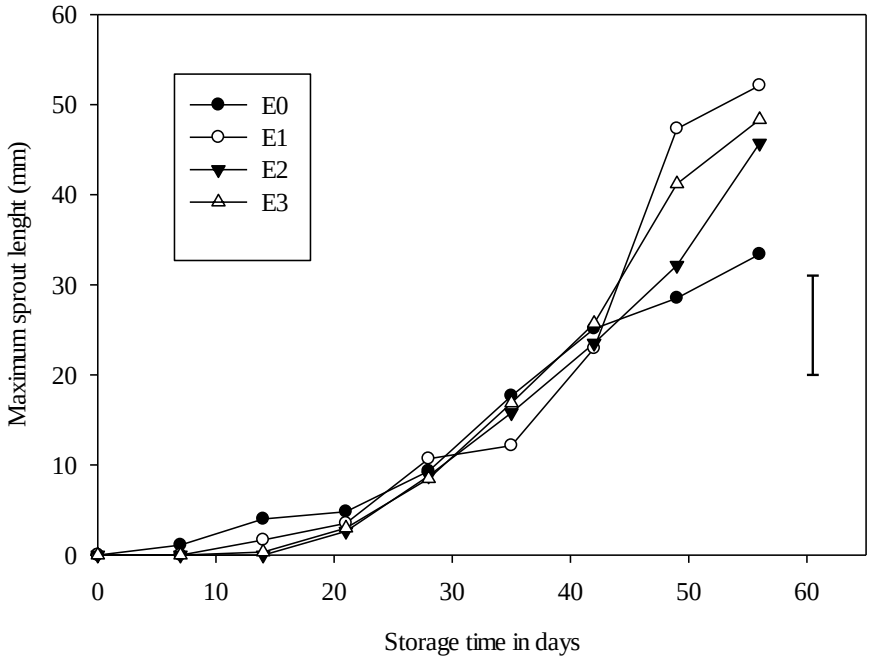

Figure 28: Average maximum sprout lengths of OFSP *Apomude*n roots Treated 7 days after harvesting with ethephon dip and stored at mean temperature and relative humidity of 26.3 °C and 88.7 %, respectively. Every data point is the mean of three treatment baskets containing 10 roots per basket (n=30). The LSD bar shows treatment-days interaction effect. ANOVA Table B3.

Number of Sprouts

Ethephon treatment postharvest did not have a significant effect on the number of sprouts per root (p = 0.545) (Table B4). The highest mean number of sprouts was recorded in both E3 and E2 at 1.8 followed by E0 and E1 which had 1.7 and 1.5 sprouts, respectively (Table 5). The roots sprout number increased consistently both in the control roots and treated roots throughout storage (Fig. 29).

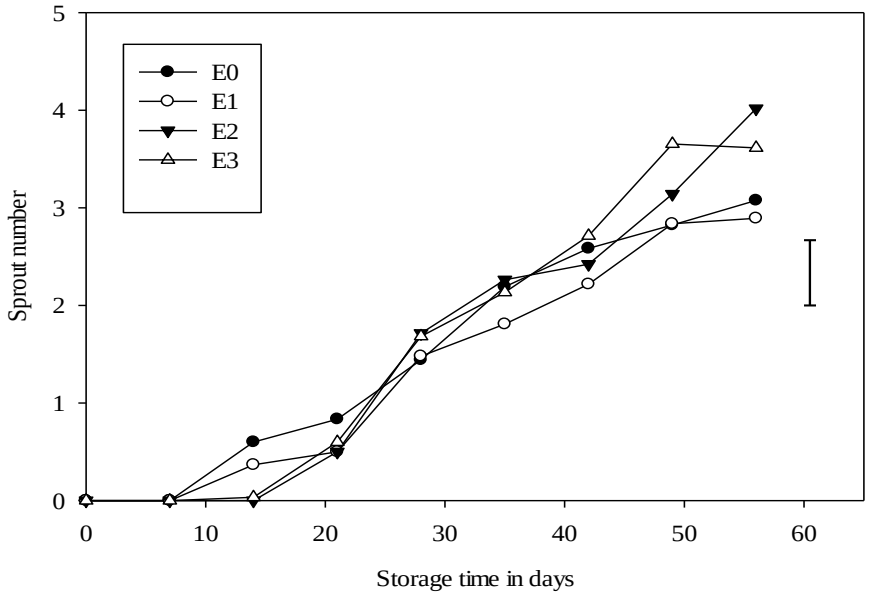

Figure 29: Average number of spouts on OFSP *Apomuden* roots

Treated 7 days after harvesting with ethephon dip and stored at mean temperature and relative humidity of 26.3 °C and 88.7 %, respectively. Every data point is the mean of three treatment baskets containing 10 roots per basket (n=30). The LSD bar shows treatment-days interaction effect. ANOVA Table B4.

Incidence of Decay

Figure 31 shows the percentage incidence of root decay over the storage period. Postharvest dip in ethephon had no significant effect on the

root decay (p = 0.116) (Table B5). The absolute mean root decay was highest in the control (28.3 %) while E3, E2 and E1 had mean values of 17.9 %, 16.2 % and 12.5 % decay, respectively (Table 5). In most of the treated roots decay started at the proximal ends and progressed towards the distal ends (Fig. 30).

Figure 30: Decayed root from treatment E3 after 21days of storage

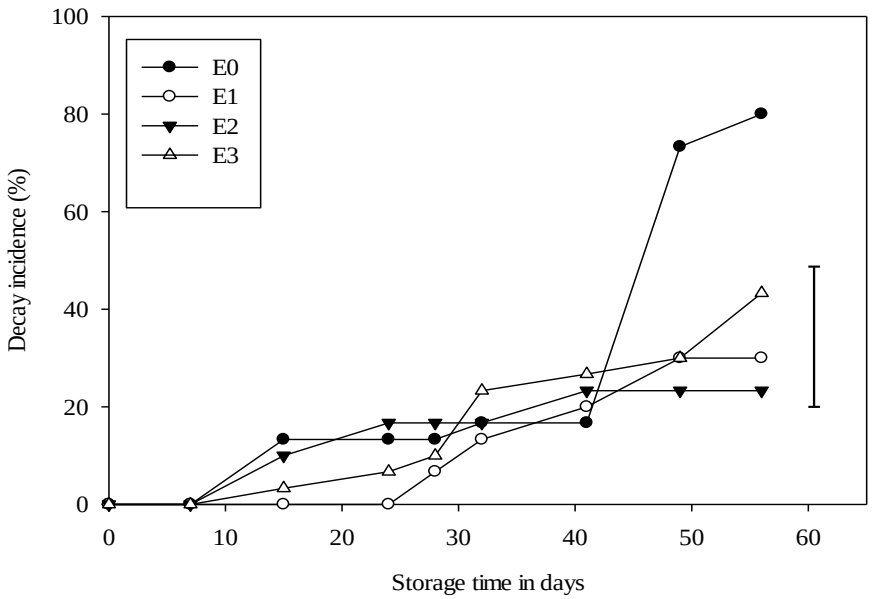

Figure 31*:* Average percentage decay incidence of OFSP *Apomuden* roots Treated 7 days after harvesting with ethephon dip and stored at mean temperature and relative humidity of 26.3 °C and 88.7 %, respectively. Every data point is the mean of three treatment baskets containing 10 roots per basket (n=30). The LSD bar shows treatment-days interaction effect. ANOVA Table B5.

Severity of Decay

Ethephon application postharvest did not have any significant effect on the severity of root decay (p = 0.354) (Table B6). However, E0 and E1 had the highest and lowest mean severity of decay of 1.4 and 1.2, respectively while E2 and E3 both had 1.3 (Table 5). Figure 32 shows the trend of decay severity at different storage time.

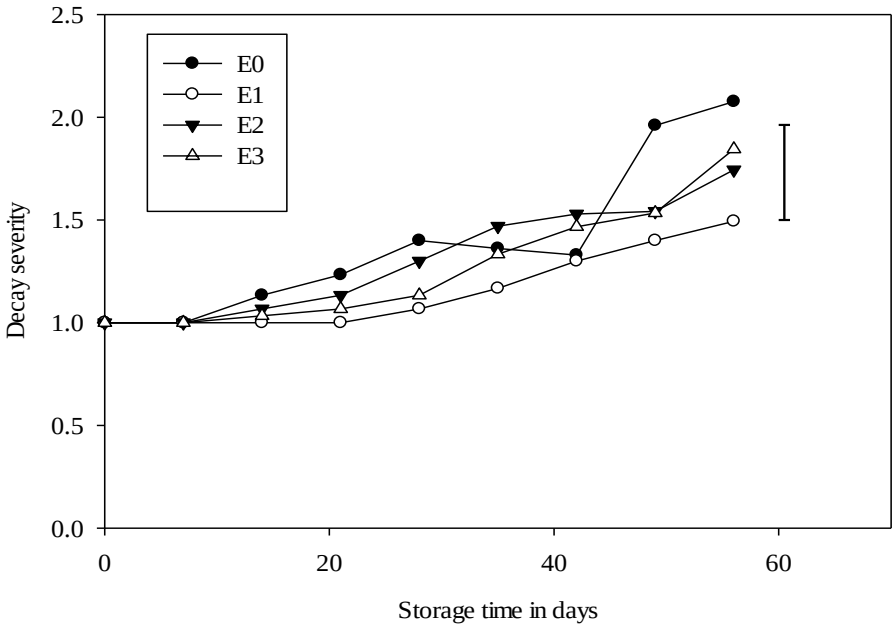

Figure 32: Average decay severity of OFSP *Apomuden* roots

Treated 7 days after harvesting with ethephon dip and stored at mean temperature and relative humidity of 26.3 °C and 88.7 %, respectively. Every data point is the mean of three treatment baskets containing 10 roots per basket (n=30). The LSD bar shows treatment-days interaction effect. ANOVA Table B6.

Table 5: Mean Sprouting and Decay of OFSP Roots Treated 7 Days after Harvesting

Treatment (ppm)	Weight loss (%)	Sprout Length (mm)	Sprout number	Sprouting index (%)	Decay incidence (%)	Decay severity
E0 (0)	7.2±1.6	15.5±4.2	1.7±0.4	57.5±0.4	28.3±1.7	1.4±0.1
E1 (100)	7.8±1.6	18.8±6.7	1.5±0.4	50.8±11.0	12.5±4.6	1.2±0.1
E2 (500)	7.3±1.6	16.1±5.5	1.8±0.5	47.1±12.1	16.2±2.8	1.4±0.1
E3 (1000)	8.4±1.8	17.9±6.2	1.8±0.5	50.4±12.1	17.9±5.4	1.3±0.1
$LSD_{0.05}$	1.4	5.6	0.4	15.9	13.5	0.3

Pathogen Identification

In most instances decay started at the proximal ends of the roots (Fig 30). Decay initially appeared brown, but turned black later. The decay was firm and moist earlier on but in short time storage roots became completely black and shriveled. The pathogens as observed under the electron microscope are as shown in Figure 33. The observed microconidia were long and sparse floccose mycelium. Microconidia were 1-2-celled and ellipsoidal in shape. All these pointed out to *Fusarium solani* as described by Mathur and Kongsdal (2003) Figure 33 A. The disease occurred after harvesting and the symptoms included root rot which penetrated the vascular ring and the lesions had concentric light and dark brown rings. Later on, the lens-shaped cavities developed inside the lesions contained white mould fibres.

In experiment 2 the pathogen as observed in the microscope (×40) showed grouped acervuli with scanty mycelium and absence of sclerotia. The conidia were hyaline and single celled, slightly falcate with rounded ends. All

these pointed out to *Colletotrichum lini* as described by Mathur and Kongsdal (2003) (Figure 33B).

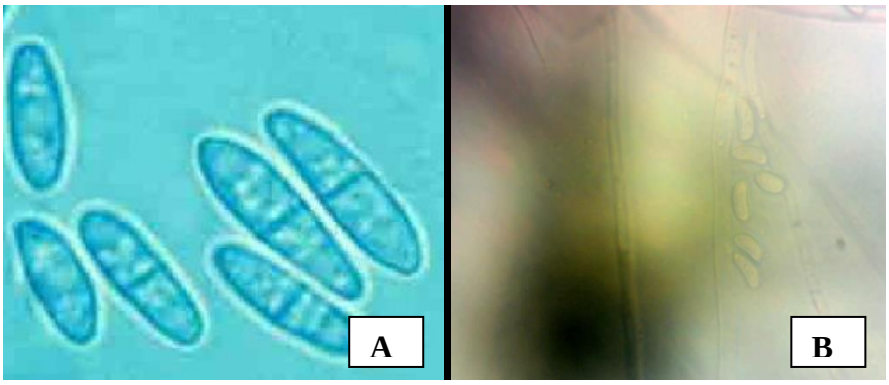

Figure 33: Pathogen as observed in the electron microscope, *Fusarium* sp. (A) and *Colletotrichum* sp. (B) (Magnification × 40)

Biochemical Changes Pre-Harvest Ethephon Application

Pre-harvest ethephon application had significant effects on the various biochemical contents of the OFSP roots during storage. The following are the biochemical constituents studied; dry matter, starch, sucrose, fructose, glucose, proteins and minerals (zinc and iron).

Dry Matter Content

Dry matter weight was obtained as a percentage of the fresh weight (% FW). The mean dry matter weight of samples tested during different time of storage period is as shown in Figure 34. Ethephon had a significant effect on the dry matter content ($p = 0.007$) (Table C1). The mean dry matter proportion of the fresh weight varied throughout storage time within the range 21.5 - 22.8 % (FW). The highest mean dry matter proportion of fresh weight was recorded in E1 as 22.8 % while E2 and E3 had 22.7 and 22.3 %, respectively with no significant difference between the treatments. E0 had the lowest dry matter content of 21.53 % of the fresh weight, which was significantly lower than the treated roots (Table 6).

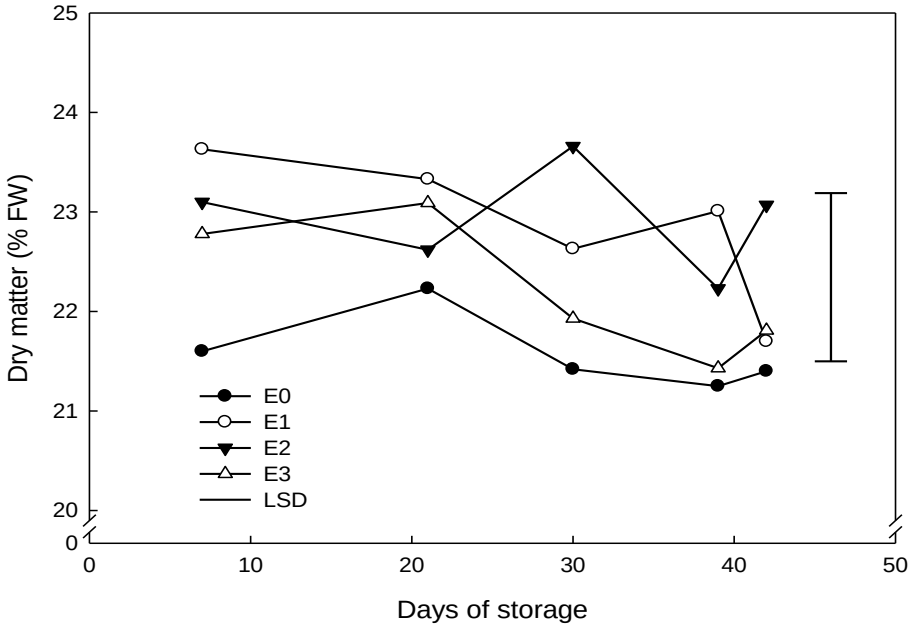

***Figure 34**:* Percentage dry matter weight of OFSP *Apomuden* roots

Treated 7 days before harvesting with ethephon and stored at mean temperature and relative humidity of 26.3 °C and 88.7 %, respectively. Every data point is the mean of four replications per treatment containing 3 roots per replication (n=12). The LSD bar shows treatment-days interaction effect. ANOVA Table C1.

Starch Content

The starch content at different periods of analysis is shown in Figure 35. Ethephon pretreatment had no significant effect on the starch content in the roots (P = 0.062) (Table C2). However, the absolute mean starch concentrations were different among the individual treatments and the control (Table 6). Roots treated with 500 ppm (E2) had the highest mean starch content of 46.44 g/100g DW followed by E1, E3 and E0 which had mean starch contents of 46.2, 45.7 and 44.4 g/100g DW, respectively.

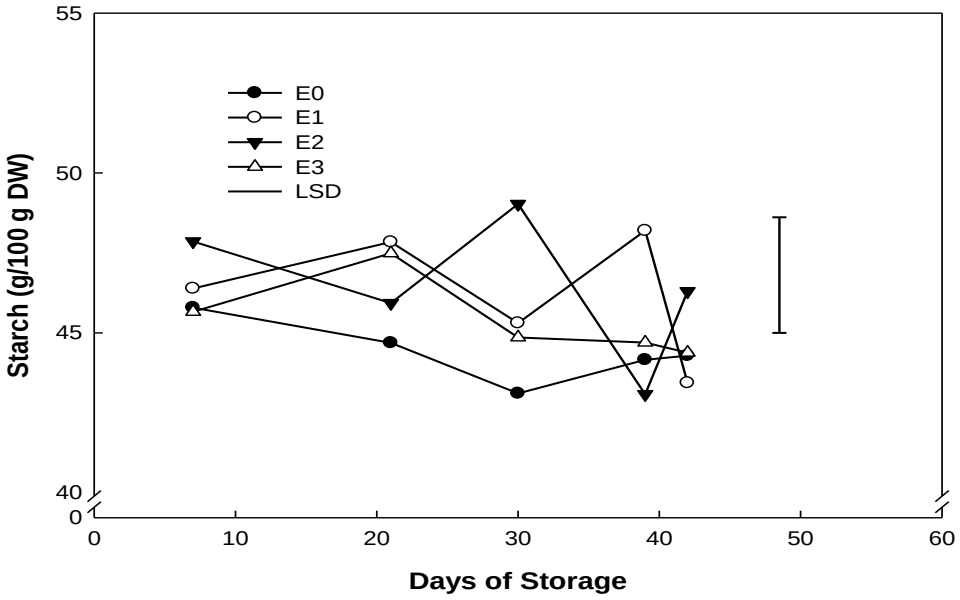

Figure 35: Starch concentration (g/100g DW) of OFSP *Apomuden* roots treated 7 days before harvesting with ethephon and stored at mean temperature and relative humidity of 26.3 °C and 88.7 %, respectively. Every data point is the mean of four replications per treatment containing 3 roots per replication (n=12). The LSD bar shows treatment-days interaction effect. ANOVA Table C2.

Table 6: **Mean Dry Matter and Starch Content of OFSP Roots Treated 7 Days before Harvesting**

Treatment (ppm)	**DM (% FW)**	**Starch (g/100g)**
E0 (0)	21.58^{b}±0.17	44.41±0.43
E1 (100)	22.86^{a}±0.33	46.24±0.87
E2 (500)	22.70^{a}±0.24	46.44±1.01
E3 (1000)	22.21^{ab}±0.31	45.70±0.56
$LSD_{0.05}$	1.69	3.61

Values are means of 240 determinations. Means with same letters within a column are not significantly different ($p < 0.05$).

Sugar Concentration

The main sugars studied were sucrose, fructose and glucose content. Sucrose formed the bulk (20.5g/100g DW) among the three sugars studied followed by glucose (6.7 g/100g DW) and fructose (4.7 g/100g DW) (Table 7). There was significant effect of the days of storage on the sugar concentrations. Glucose and fructose concentrations decreased with time while sucrose remained relatively high.

Sucrose

The treatment had no significant effect on the sucrose concentrations ($p = 0.09$) (Table C4). From the mean absolute values, the control (E0) had the highest mean sucrose concentration (21.4 g/100g DW) followed by E3, E2 and E1 each with 20.5, 20.3 and 19.7 g/100g DW, respectively (Table 7). For all the treatments, the sucrose content fluctuated throughout the storage (Figure 36).

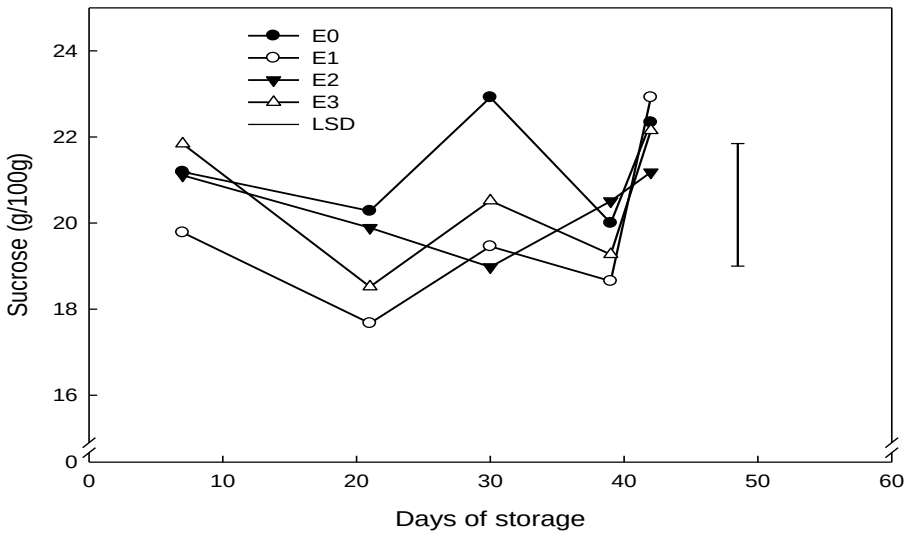

Figure 36*:* Sucrose concentration (g/100g DW) of OFSP *Apomuden* roots treated 7 days before harvesting with ethephon and stored at mean temperature and relative humidity of 26.3 °C and 88.7 %, respectively. The LSD bar shows treatment-days interaction effect. ANOVA Table C3.

Glucose

With the exception of E3 all other levels of ethephon treatment significantly reduced the glucose concentration compared to the control (p = 0.001) (Table C5). Roots treated with E2 had the lowest glucose concentration (5.9 g/100g DW) while the control had the highest glucose content of 7.3 g/100g DW. Treatments E3 and E1each had mean glucose concentrations of 6.8 and 6.7 g/100g DW, respectively (Table7). There was a general decline in glucose content for all the treatments throughout the storage period (Fig. 37).

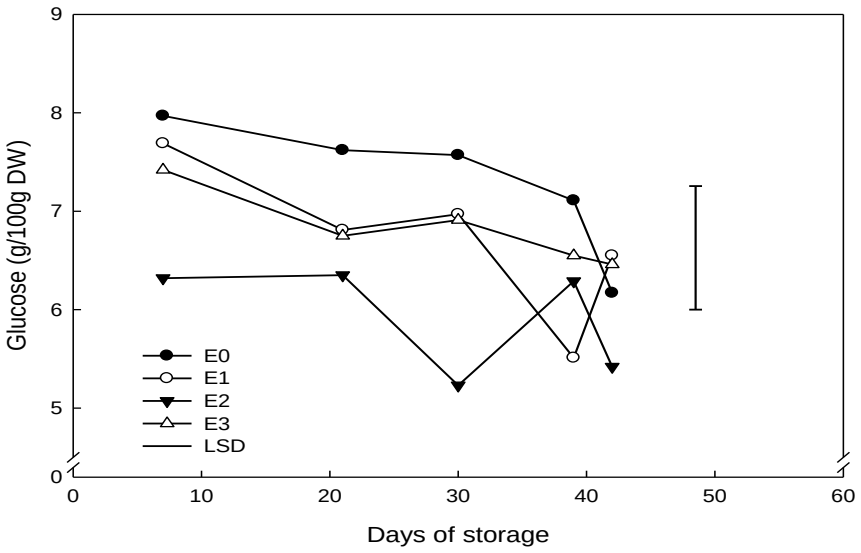

Figure 37: Glucose concentration (g/100g DW) of OFSP *Apomuden* roots Treated 7 days before harvesting with ethephon and stored at mean temperature and relative humidity of 26.3 °C and 88.7 %, respectively. Every data point is the mean of four replications per treatment containing 3 roots per replication (n=12). The LSD bar shows treatment-days interaction effect. ANOVA Table C4.

Fructose

All levels of ethephon treatment significantly suppressed fructose concentration in the roots (p = 0.001) (Table C4). The control had mean

fructose content (5.0 g/100g DW) while E2 had the lowest fructose content (3.9 g/100g DW). This was followed by E3 and E1 with 4.5 and 4.4 g/100g DW, respectively (Table 7). For all the treatments; there was a general decline in fructose content in the first three weeks of storage and a slight increase thereafter in E1 and E3 (Fig. 38).

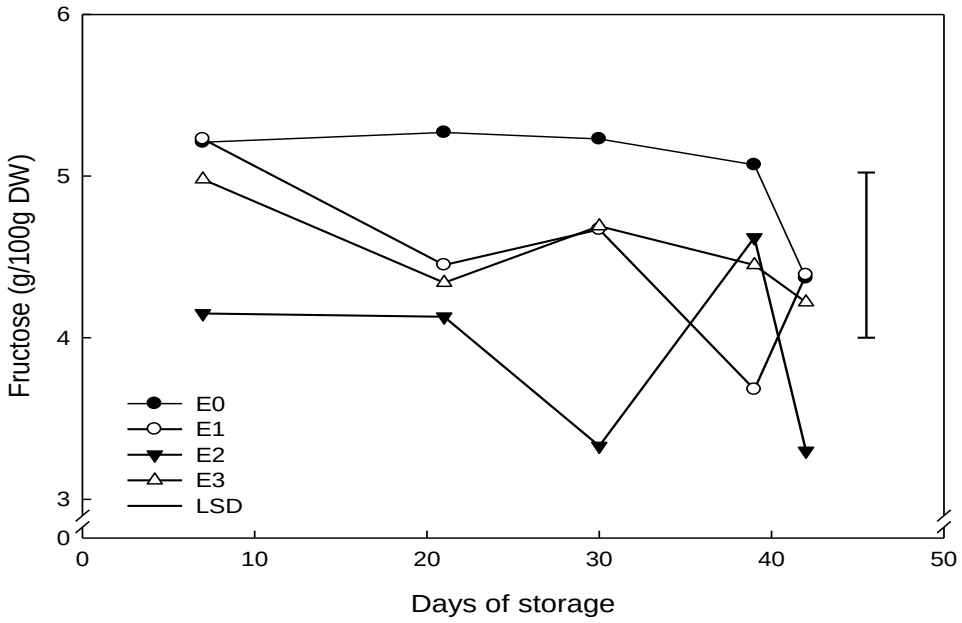

***Figure 38**:* Fructose concentration (g/100g DW) of OFSP *Apomuden* roots Treated 7 days before harvesting with ethephon and stored at mean temperature and relative humidity of 26.3 °C and 88.7 %, respectively. Every data point is the mean of four replications per treatment containing 3 roots per replication (n=12). The LSD bar shows treatment-days interaction effect. ANOVA Table C5.

Table 7: Mean Sucrose, Glucose and Fructose Concentrations of OFSP Roots Treated 7 Days before Harvesting

Treatment (ppm)	Sucrose (g/100g)	Glucose (g/100g)	Fructose (g/100g)
E0 (0)	21.35a±0.57	7.29a±0.31	5.03a±0.17
E1 (100)	19.70a±0.89	6.71b±0.35	4.48b±0.25
E2 (500)	20.33a±0.41	5.92c±0.25	3.91c±0.26
E3 (1000)	20.46a±0.71	6.82a±0.17	4.54b±0.14
LSD	2.846	0.56	0.46

Values are means of 240 determinations. Means with same letters within a column are not significantly different ($p < 0.05$).

Effects of Ethephon on Protein Concentration

All ethephon levels significantly increased the protein contents in the roots ($p = 0.001$) (Table C6). Roots treated with E2 had the highest mean protein concentrations of 5.3 g/100g DW while E0 had the lowest protein content (4.1 g/100g DW) (Table 8). Treatments E3 and E1 had mean protein concentrations of 5.1 and 4.9 g/100g DW, respectively. There was a gradual increase in protein content in roots treated with ethephon during the storage period until the 40^{th} day when the concentration in E1 and E3 started declining. Conversely, there was a decline in protein content in the control root samples (Fig. 39).

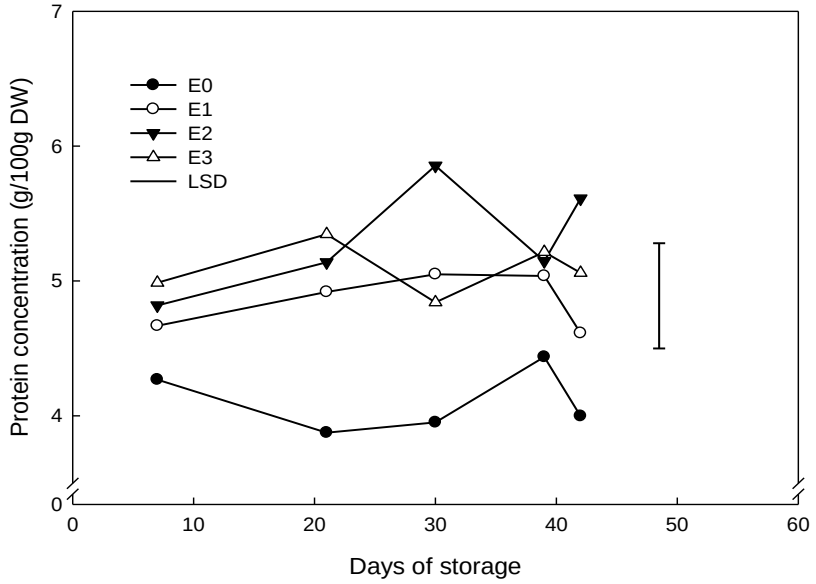

***Figure 39*:** Protein concentration (g/100g DW) of OFSP *Apomuden* roots

Treated 7 days before harvesting with ethephon and stored at mean temperature and relative humidity of 26.3 °C and 88.7 %, respectively. Every data point is the mean of four replications per treatment containing 3 roots per replication (n=12). The LSD bar shows treatment-days interaction effect. ANOVA Table C6.

Effects of Ethephon on Iron Concentration

All ethephon levels significantly increased the iron contents in the roots (p = 0.001) (Table C7). Roots treated with E2 and E3 had significantly higher mean iron concentrations of 2.5 mg/g DW during storage while treatments E1 and E0 had mean iron concentrations of 2.3 and 2.2 mg/g DW, respectively (Table 8). There was a gradual decrease in iron concentration in both ethephon-treated and the control root samples during the storage period (Fig. 40).

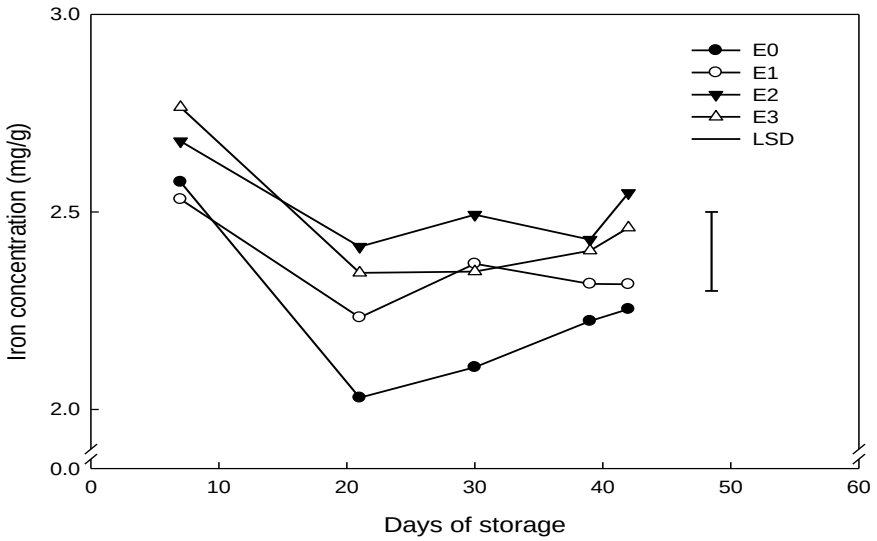

***Figure 40*:** Iron concentration (mg/g DW) of OFSP *Apomuden* roots

Treated 7 days before harvesting with ethephon and stored at mean temperature and relative humidity of 26.3 °C and 88.7 %, respectively. Every data point is the mean of four replications per treatment containing 3 roots per replication (n=12). The LSD bar shows treatment-days interaction effect. ANOVA Table C7.

Effects of Ethephon on Zinc Concentration

All ethephon levels, except E1, significantly increased the zinc concentration in the roots compared to the control ($p = 0.001$) (Table C 8). Roots treated with E2 and E3 had the highest mean zinc concentrations of 1.4 mg/g DW during storage while the treatments E1 and E0 had mean zinc concentrations of 1.3 mg/g DW (Table 8). The zinc concentration in ethephon treated roots remained fairly constant while zinc concentration in the control root samples reduced gradually (1.4-1.3 mg/g) until the 30th day before it increased slightly from 1.3 to 1.4 mg/g at the end of storage period (Fig. 41).

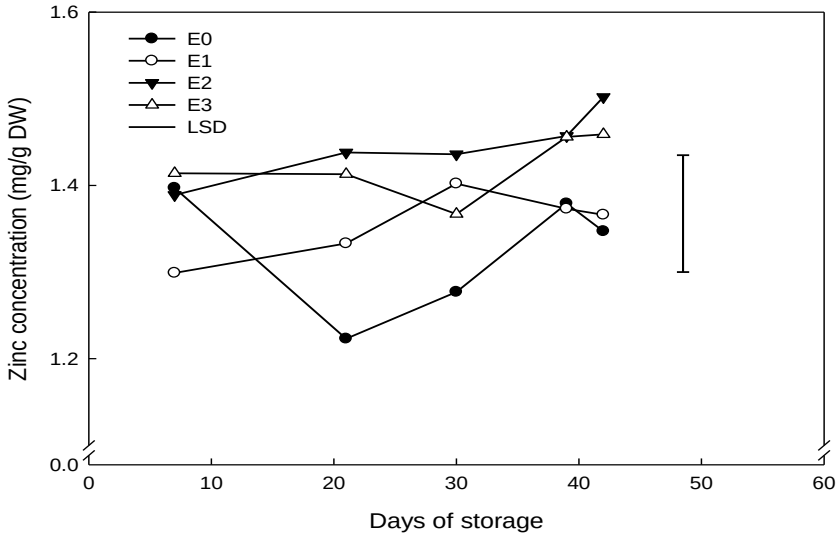

***Figure 41*:** Zinc concentration (mg/g DW) of OFSP *Apomuden* roots

Treated 7 days before harvesting with ethephon and stored at mean temperature and relative humidity of 26.3 °C and 88.7 %, respectively. Every data point is the mean of four replications per treatment containing 3 roots per replication (n=12). The LSD bar shows treatment-days interaction effect. ANOVA Table C7

Table 8: Mean Protein, Iron and Zinc Concentrations of OFSP Roots Treated 7 Days before Harvesting

Treatment (ppm)	Protein (g/100g DW)	Iron (mg/g)	Zinc (mg/g)
E0 (0)	4.106^{c}±0.106	2.238^{c}±0.094	1.325^{b}±0.033
E1 (100)	4.858^{b}±0.092	2.354^{b}±0.050	1.355^{b}±0.018
E2 (500)	5.314^{a}±0.185	2.512^{a}±0.048	1.444^{a} ±0.018
E3 (1000)	5.090a^{b}±0.088	2.464^{a}±0.078	1.422^{a}±0.017
LSD	0.3487	0.0894	0.0604

Values are means of 240 determinations. Means with same letters within a column are not significantly different ($p < 0.05$).

Discussion

The effects of different levels of pre and post-harvest ethephon application on the physiological and biochemical changes of the orange flesh sweet potato *Apomuden* were examined throughout the storage period. The roots for the two experiments were stored at mean temperature of 26.6 °C while mean relative humidity was 89 %. The outcome of the research is discussed in the following sections.

Effect of Ethephon Foliar Application on Sweet Potato Leaves and Vines

Ethephon had a yellowing effect on the leaves which was more pronounced in the highest concentration. This was then followed by abscission of the leaves from the third day after ethephon application. However, the vines were not affected by ethephon and they remained green. The leave senescence and abscission could be linked to the plant hormones ethylene and abscisic acid (ABA). Ethylene and ABA are involved in bud and seed dormancy, leaf senescence and abscission. It inhibits opening of stomata and promotes a variety of plant stress reactions (Fedoroff, 2002).

Enhanced ethylene production plays a role in the ability of abscisic acid to accelerate abscission of isolated abscission zone in plants. Ethylene at concentration of 10 ppm is stated to represent a saturating concentration of the gas for abscission. Ethylene is reported to induce the synthesis of hydrolytic enzymes that dissolves plant cell walls and the middle lamella (Tucker, Sexton, Del Campillo & Lewis, 1988). ABA increases the rate of leave senescence by increasing the rate of ethylene synthesis and induction of cellulose which are actively involved in abscission (Abeles, 1969).

Similar results as in the present study were observed by Chen *et al.* (2010) in a study to determine ethephon-induced effects on leaf senescence and chlorophyll content in sweet potato leaves. Sweet potato leaves detached and senesced earlier in ethephon treatment compared to dark control within three days. The leaves began to turn noticeably yellow on the second day and became nearly wholly yellow on the third day. Sweet potato vines and leaves mean dry weight treated with Ethrel at 250 ppm was lower (2.9 and 4.1%, respectively) than that of the control, and the average dry weight of the vine and leaf treated with Ethrel at 500 ppm was lower (0.6% and 2.2%, respectively) than that of the control (Haizhou,1987).

Weight Loss

Preharvest ethephon application resulted in a lower grand mean weight loss (8.3 %), the effect of which increased with the ethephon concentration while postharvest application had higher root weight loss (8.4 %). Weight loss in storage is as a result of respiration and transpiration, which trigger the conversion of dry matter into energy and water loss, respectively. Weight loss due to transpiration is caused by vapour pressure differential between the outside environment and the root interior (Ke & Kader, 1992). Moreover, root decay contributes to physiological loss of weight as well as reduction of the saleable weight (Kushman & Pope, 1972; Picha, 1986). Rees *et al.* (2003) reported that weight loss in tropical environments during storage is certain and is mainly caused by water loss.

The results from this research are similar to the results by Amoah (2014) who found that ethylene (10 ppm) supplementation resulted in higher root weight loss. Furthermore, De and De (2003) found that ethephon caused a

loss of fresh weight and decreased the length of the seedlings significantly with increase in the concentration of ethephon. The highest decrease in fresh weight (41.7 %) was found at 1600 mg/l concentration and maximum decrease in seedling length (46.3 %) was observed at a 2500 mg/L concentration of ethephon applied on *Trigonella foenum-graecum*. Ethylene also causes expansion of tissue that result in the loss of compartmentalisation of cells (Pratt & Goeschl, 1969). The loosened tissues might aid the loss of water.

Moisture Content

The moisture content of roots treated preharvest fluctuated throughout the storage period. This trend may be due to the relative rates at which individual roots lose their moisture and dry matter, a process which can vary from one root to the other. Abscission of leaves or stomatal closure due to the biosynthesis of abscisic acid is a physiological response to attenuate moisture loss. Thus with the yellowing and subsequent abscission of the leaves of sweet potato, moisture loss was expected to be lower in the ethephon treatments. The opposite effect was, however, observed suggesting that a much more complex mechanism was involved in the moisture loss from the roots.

Ethylene-stimulated respiration of sweet potato and the consequential induction of moisture loss has been reported by Amoah (2014) and possibly, such accelerated respiration by ethephon may be responsible for the higher moisture loss in the roots which also reflected in the higher loss of weight. Kitinoja (1987) cured sweet potato varieties Garnet, Jewel and Jersey in the presence of 2 ppm ethylene or in air at 30 °C. It was similarly found that roots cured in ethylene lost water at higher rates than air-cured roots.

The loss of water from storage roots is a key factor in their storability and quality in the marketing chain. The moisture loss witnessed in this research could be as a result of a number of factors such as wounds on the roots, respiratory activities and the storage conditions of the roots. The mean temperature and relative humidity recorded was 26.3 °C and 88.7 %, respectively. Sweet potato needs lower temperatures (15 °C) and high relative humidity (90 %) for longer storage. The high temperature in the store and slightly lower relative humidity may have contributed to the moisture loss observed during the study.

Sprouting

Preharvest ethephon treated roots had short dormancy period, with E2 having the shortest period of 6 days. In the preharvest roots, the highest sprouting index was in treatment E2 (41.9 %) and the lowest was in the control (18.8 %). Postharvest ethephon dip had higher sprouting index (57.5 %) compared to the control while E2 had the lowest sprouting index (47.08 %). Ethylene has been reported to reduce dormancy period.

Amoah (2014) found out that when sweet potato roots were transferred from ethylene into air after dormancy break, there was vigorous sprout growth in terms of the number of sprouted buds, number of roots that sprouted and the length of the sprouts. The accentuated sprout growth could be as a result the release of ethylene from the ethephon which influenced the bioactivity of other growth regulating hormones like zeatin riboside (Abdi, McGlasson, Holford, Williams & Mizrahi, 1998). Zeatin riboside is a hormone which stimulates growth and is linked to dormancy break and growth of sprouts. However, continuous ethylene exposure from the start of storage and post-

dormant transfer from air storage into ethylene significantly inhibited sprout growth (Amoah, 2014). In his conclusion, the author suggested that once sweet potato roots are predisposed to ethylene, the roots may have to remain in ethylene if quality is to be preserved. This effect was also observed in potatoes by Foukaraki *et al*. (2012).

Similar results have been obtained in ethephon applied on three cultivars of freshly harvested cormels which were studied for 3 years. Ethephon at 400 mg/L was found to be the most effective in altering the days to sprouting, sprout percentage, corm size and development of cormels. The dormancy period was reduced by 17.5 days compared to the control (Ram, Mukherjee & Manuja, 2002). Additionally, in sugarcane experiments involving foliar application of ethephon 7 to 10 days before harvest had stimulatory effect on bud sprouting and growth of stubble buds (Solomon, 1996).

Incidence and Severity of Root Decay

Preharvest study shows that the highest concentration of ethephon (1000 ppm) induced significantly higher decay incidence (45.2 %) and severity of decay (2) than the other treatments. The control had the lowest decay incidence (14.4 %) though it was not significantly different from the ethephon treatments E1 (28.1 %) and E2 (21.9 %). On the other hand, postharvest ethephon application had no significant effect on the root decay.

In all the experiments, the decay mostly affected the proximal ends of the roots and progressed towards the distal ends. Nearly all abiotic and biotic stress situations stimulate ethylene production in plants. The function of ethylene in pathogen-infested plants has been reviewed and further

investigated and the results illustrate the phenomenon as a complex one (Bleecker & Kende, 2000). According to Hoffman, Schmidt, Zheng & Bent (1999), dependent on the pathogen, disease indications seem to be either reduced or heightened by ethylene, or not affected at all. In other words, ethylene seems to facilitate defense responses to some pathogens and to quash them in others. It has been reported that ethylene helps to curb the spread of a pathogen by initiating leaf abscission.

Similar incidences have been observed by Arancibia *et al*. (2013) who used pre-harvest foliar applications of ethephon to determine the possible relationship of tip rot with ethephon-induced stress. After 1-2 months in storage, tip rot occurrence was witnessed mostly in stored roots from ethephon-treated foliage. The increase in tip rot incidence was correlated well with ethephon rate. In addition, when sweet potato varieties 'Garnet', 'Jewel' and 'Jersey' were cured in presence of 2 ppm ethylene at 30 °C it was found that the roots cured in ethylene formed lumps at the root ends which were then infested with fungi unlike the roots cured in air (Kitinoja, 1987). In a similar research Clark *et al*. (2013) treated vines with ethephon 2 weeks before harvest and placed the roots directly in storage at 15 °C without curing. This resulted in greater incidence of tip rots with significant differences in incidence amongst the sweet potato genotypes assessed.

Roots Shrinkage

Preharvest ethephon treatment had significant effect on shrinkage though lower than the shrinkage observed in the postharvest treated roots. Treatment E3 had the highest shrinkage (1.9 %) while E1 had the lowest shrinkage of 1.3 %. The observed shrinkage could be mainly due to water loss

from the treated root tissue. This is further confirmed by the fact that highest dry matter content and lowest weight loss occurred in roots treated with E1.

In experiment 2 the control roots had the highest shrinkage (6.4 %) while E1 had the lowest shrinkage (3.1 %) at the end of storage. The observed shrinkage could be due to water loss from the treated root tissue during storage. Dry matter loss may have also contributed to shrinkage in the control. Water and dry matter losses lead to weight loss which then results in roots shrinkage (Edmunds *et al*., 2008).

Pathogen Identification

In preharvest treated roots the pathogen identified was *Fusarium solani.* This is a fungus which causes *Fusarium* root rot a disease which affects sweet potatoes during storage. It is caused by fungal strains of *Fusarium solani*, which is a soil-borne pathogen carried by wounded roots. The disease typically occurs after harvesting and its symptoms include white spores that penetrate the cortex of the roots. The fungus is soil-borne and can survive in the soil for several years. Contamination is typically through wounds that occur during and after harvesting. The roots may be predisposed in the field when the roots develop growth cracks (Loebenstein *et al*., 2009). The roots might have been infected from the field during harvesting. Fungal infection has been reported to increase the protein concentration of sweet potato roots (Onifade, Atum & Adebolu, 2004).

On the other hand, postharvest roots were infected with *Colletotrichum lini* fungus which causes black dot disease in potato. It has also been reported to attack five varieties of sweet potato in Kenya (Skoglund, 1993). The fungi

can survive on plant debris and its emergence is facilitated by wet weather and warm temperatures.

Biochemical Changes

Dry Matter

Sweet potato has high moisture content but low in the dry matter as in other root crops (Woolfe, 1992). Consumers in Africa have been noted to have a preference for high dry matter varieties of sweet potato. However, sweet potato varieties with high carotenoid contents like OFSP tend to have lower dry matter contents (Kapinga & Carey, 2003). Ethephon had a significant effect on the dry matter content which varied throughout storage time. A fluctuating trend was also observed in the root percentage dry matter content during the different sampling periods. Generally roots treated with ethephon had significantly higher percentage of dry weight compared to the control. This may be attributed to the concomitant moisture loss and this increases the proportion of dry matter in the fresh weight. The increase in the percentage dry weight in ethephon treatment has been observed in sweet potato and other crops.

In a study carried out by Haizhou (1987), spraying ethephon at 250 and 500 ppm on the vines and leaves of sweet potato at the end of the early growth helped to control the unnecessary growth of the vines and leaves and increased the yield of root-tubers. The mean dry weight of the root tuber in treatments with ethephon at 250 and 500 ppm was higher (9.1% and 3.2%), respectively than that of the control. Sweet potato treated with 500 ppm had their yield increased by 14 % and with 250 ppm by 6 %.

De and De (2003) studied the effect of ethylene on growth, diosgenin production and activities of two antioxidant enzymes catalase and peroxidase in *Trigonella foenum-graecum* seedlings. The seedlings were treated with different concentrations of ethephon and kept in sterile lidded glass vessel for 5 days at 15–18 °C. The seedlings were then dried at 45 °C for 48 hours. The length of seedlings and fresh weight decreased significantly but the dry weight of seedlings increased with increase in the concentration of ethephon. With the exception of the ethephon at 100 and 200 mg/L concentrations, the other concentrations of ethephon increased the dry weight of seedlings significantly over the control. Maximum increases (22.7 %) were found at 2000 and 2500 mg/L concentrations. Ethephon application enhanced dry matter accumulation in the non-grain plant parts of barley (Ma & Smith, 1992). The percentage dry matter increase shows that ethephon led to faster rate of moisture loss than dry matter loss. If transpiration is higher than respiration then the dry matter will increase with respect to percentage of the FW and vice versa.

Starch

Starch makes up the bulk of sweet potato roots. Its formation from photosynthesis stops when the roots are harvested hence the roots' living tissues rely on the internally stored reserves for their biochemical activities. Therefore, the stored starch is vital for longer storage life of the roots. Stored starch goes through enzymatic conversion to simple sugars like sucrose, fructose and glucose (Aiyer, 2005). In this study roots treated with 500 ppm had the highest mean starch content of 46.44 g/100g DW while the control had the lowest mean starch contents of 44.4 g/100g DW.

Ethephon has been reported to increase the average dry weight of sweet potato roots which is mainly composed of starch. In a treatment of sweet potato leaves and vines with ethephon (ethrel) at 250 and 500 ppm, higher dry weight was recorded (9.1 %and 3.2 %, respectively) than the control. Generally, there was a noticeable rise in the weight loss of roots treated with ethephon even though the dry matter increased. This confirms that weight loss is predominatly due to water loss (Cheema *et al*., 2013). When treated with 500 ppm the root tuber yield increased by 14 % and with 250 ppm by 6 % (Haizhou, 1987).

In addition, when sweet potato roots are stored in the ambient temperatures, their starch-sugar equilibrium shifts towards starch buildup whereas low storage temperatures leads to an increase in sugar content (Wills *et al*., 1989). This could be the reason why there was an increase in starch content while the sugars content was significantly reduced during storage. The mean storage temperature was at 26.6 ºC which might have favored the observed starch accumulation.

Effects of Ethephon on Sugar Concentrations

The main sugars studied were sucrose, fructose and glucose content. Sucrose formed the bulk (20.5g/100g DW) among the three sugars studied followed by glucose (6.7 g/100g DW) and fructose (4.7 g/100g DW) (Table 7). There was significant effect of the days of storage on the sugar concentrations. Glucose and fructose concentrations decreased with time while sucrose remained relatively high.

Sucrose

Sucrose makes the bulk of sweet potato sugar. When cultivars are analyzed in raw form, the sucrose concentration exceeds the other sugars (Van Den, Biermann & Marlett, 1986). In this study the mean sucrose concentration was the highest among other sugars. The mean sucrose content was approximately 3-fold higher than glucose concentration and 4-fold higher than the fructose concentration. Preharvest treatment had no significant effect on sucrose content ($p = 0.09$). It was also observed that the sucrose content fluctuated as storage time progressed and increased at the end of storage. These results are similar to those found by Cheema *et al.* (2013) who observed that after 4 weeks of storage of "Bushbuck" and "Ibees" sweet potato in 10 µl/L of ethylene and kept at 25^{o}C, there was an increase in sucrose content though it was not significant. In addition, Zhang *et al*. (2002) found that sucrose increased with storage time over the first 60 days. The increase in sugar content may be due to enzymatic conversion of starch to sugars.

Glucose and Fructose

In this study, ethephon significantly suppressed the glucose and fructose concentration in the roots. These results are similar to those found by Cheema *et al*. (2013) who noted that 10 µl L^{-1} ethylene applied on the sweet potato roots at 25 °C led to a decrease in the amount of fructose and glucose in two varieties *viz*. "Bushbuck" and "Ibees" following four weeks in storage (25 ^{o}C) compared to the untreated roots. In addition, Amoah, (2014) found that the 'North Carolina Covington' sweet potato variety treated with ethylene led to a decline in the concentrations of fructose and glucose with time compared to roots stored in air.

The reduction in fructose and glucose could be as a result of respiration induced by ethylene in the roots. Sprout growth requires a certain level of energy substrates and this can be obtained from the hydrolysis of starch. In contrast, Foukaraki *et al*. (2012) found out that exogenous ethylene increases the concentration of sugars in potato which leads to darkening of potatoes when fried.

Effects of Ethephon on Protein Concentration

The results clearly suggest that ethephon treatment can induce protein synthesis in OFSP roots. The highest mean protein concentration obtained in E2 (5.31 g/100g DW) is slightly higher than average crude protein content (4.41 g/l00 g DW) obtained by Ravindran, Sivakanesan and Rajaguru (1995) in 16 cultivars of sweet potato roots on a dry weight basis. Nevertheless, the concentrations determined are within the range of sweet potato samples from Africa (1.3 - 10.0 g/l00g DW) (Onwueme, 1994). Similar results were obtained in a study by De and De (2003) who found out that the protein content increased with increase in concentration of ethephon. Protein content significantly increased over that of control at 1200, 1600, 2000 and 2500 ppm ethephon concentrations in Triogonella seedlings. However, ethephon treatment at low concentration (100 ppm) decreased the protein content a day after application.

Plant growth regulators have been linked to induce protein biosynthesis as well as nucleic acid metabolism (Nooden & Thimann, 1963). It has been found that ethylene induces synthesis of protein in sweet potato slices (Imaseki, Uchiyama & Uritani, 1968). Ethephon (30 ppm) applied 24 and 48 h preharvest boosted precipitable levels of proteins in both roots and

stems of potato. Amino nitrogen concentration increased comparably with protein content in reaction to ethephon, with the tubers displaying considerable stimulation. The results showed that ethephon stimulate protein synthesis and increases cellular metabolic rate and porousness (Palmer, 1985). This could be the reason why significantly higher protein content was found in the ethephon treated OFSP roots than in the control in this study.

Effects of Ethephon on Minerals

Studies have shown that sweet potato tubers are moderately good sources of minerals (Chen, 1996). From this research ethephon treated roots had higher mineral content than the control roots. The highest average minerals; iron (2.5 mg/g) and zinc (1.44 mg/g) were all recorded in E2 while the control had iron content of 2.2 mg/g and zinc content of 1.3 mg/g, respectively. Contrary to the results obtained, ethephon at concentrations of 300, 150 and 75 ppm foliar application on apple fruit trees 1 and 2 weeks before harvesting reduced the concentration of minerals in apples when harvesting was delayed (Drake *et al*., 2005). Information on the effects of ethephon on minerals in OFSP could not be found from literature and thus the results in this study are the first report.

CHAPTER FIVE

SUMMARY, CONCLUSIONS AND RECOMMENDATIONS

Summary

Physiological and biochemical effects of different levels of ethephon (0 ppm, 100 ppm, 500 ppm, 1000 ppm) applied at preharvest and postharvest (physiological) on OFSP during storage were evaluated. The treatments were applied 7 days before harvest and 7 days after harvesting. The average storage temperature and relative humidity was 26.6 °C and 89 %, respectively. The two pretreatments used in this research showed varying physiological effects on the OFSP in storage. Though direct comparison of the two treatment regimes was not conducted, preharvest treated roots stored better than postharvest treated roots.

Preharvest treated roots were more resistant to sprouting, weevil damage and shrinkage but this was not the case in postharvest treated roots. This necessitated further analysis of the preharvest treated roots to determine its effect on some of the nutritional constituents including sugars (sucrose, glucose and fructose), minerals (iron and zinc) and proteins. The two treatments showed similar results in weight loss and decay severity.

Weight Loss and Shrinkage

In this study, physiological weight loss was measured in terms of moisture and dry matter loss in the control and treated wholesome roots. Ethephon treatment applied preharvest at high concentration (1000 ppm), caused a significant loss of OFSP root weight (8.3 %) compared to the lower concentrations and the control. Additionally, the same concentration caused greater root decay (45.3 %); further contributing to the total loss of sellable

weight. On the other hand, postharvest ethephon application had no significant effect on the weight loss of the roots. These results points out to the significance of using appropriate concentrations of ethephon at the right time.

Shrinkage was measured in terms of dimensional changes in the control and treated wholesome roots. Ethephon treatment applied preharvest caused lower shrinkage of OFSP root with the highest shrinkage being 1.93 % in E3. On the other hand, postharvest ethephon application had higher root shrinkage (5.5 %) in E2.

Decay

In this research, decay was measured in terms of the number of decayed roots and the severity of the decay in the control and treated roots. Ethephon treatment applied preharvest at high concentration (1000 ppm), caused a significant decay incidence of OFSP root (45.3 %) compared to the lower concentrations and the control. On the other hand, postharvest ethephon application had no significant effect on the decay of the roots.

Sprouting

Sprouting was measured in terms of the number of sprouted roots, the number of sprouts per root and the maximum sprout length in each root. In preharvest treatment the highest sprouting index was in treatment E2 (41.9 %) and the lowest was in the control (18.8 %). On the other hand, postharvest ethephon application had no significant effect on the sprouting of the roots though unlike the preharvest treatment it had higher sprouting index with highest being recorded in the control (57.5 %) while E1 had the highest sprouting index (50.83 %) among the treated roots.

Pathogen

Two pathogens responsible for the decay were identified as *Fusarium solani* and *Colletotrichum lini* in this study. In preharvest treated roots the pathogen identified was *Fusarium solani.* This is a fungus which causes *Fusarium* rot a disease which affects sweet potatoes during storage. It is caused by fungal strains of *Fusarium solani*, which is a soil-borne pathogen carried by wounded roots. On the other hand, postharvest treated roots were infected with *Colletotrichum lini* fungus which causes black dot disease in potato. It has also been reported to attack five varieties of sweet potato in in Kenya (Skoglund, 1994).

Biochemical Changes

Ethephon applied preharvest significantly preserved most of the desirable nutrients including starch, proteins and minerals (zinc and iron) compared to the control. The mean concentrations of sucrose, glucose and fructose measured in this study ranged between 19.7-20.46 g/100g DW, 5.92-6.82 g/100g DW and 3.91-4.54 g/100g DW, respectively (Table 7). In this study the mean sucrose concentration was the highest among other sugars. The mean sucrose content was approximately 3-fold higher than glucose concentration and 4-fold higher than the fructose concentration. Ethephon significantly reduced the glucose and fructose concentration in the roots Figure 37 and 38, respectively. From this research ethephon treated roots had higher mineral content than the control roots. The highest average minerals; iron (2.5 mg/g) and zinc (1.44 mg/g) were all recorded in E2 while the control had iron content of 2.2 mg/g and zinc content of 1.3 mg/g (Table 8). The

treatment significantly increased protein content of the roots. The highest mean protein concentration was obtained in E2 (5.31 g/100g DW).

Conclusions

This research was conducted to determine the prospects of the use of ethephon applied preharvest and postharvest to improve the storage and shelf life of OFSP sweet potato. This study has shown the efficacy of ethephon foliar application as a means to reduce most of the biochemical deterioration and dry matter loss in sweet potato during storage in the tropics. Ethephon (100 and 500 ppm) application preharvest significantly reduced the dry matter loss compared with the control. In addition, the same level of ethephon significantly preserved most of the nutrients tested in the OFSP roots. The nutrients preserved include starch, protein and minerals (zinc and iron).

The starch retention by the treated roots during storage is desirable especially for sweet potato processing methods like frying. Roots with low sugar content are desirable for frying purposes as it reduces the darkening of chips when fried. Darkening of fried chips is associated with high sugar content of roots mainly stored in the ambient conditions. From this study the roots from the control consignment had the highest sugar content. Glucose and fructose content in the control roots were significantly higher than in the ethephon treated roots. This implies that the starch reserve in the roots is broken down faster to sugars in the untreated roots hence further reducing the root quality in storage.

Whereas the reduced dry matter loss and reduction in sugar accumulation may be beneficial for sweet potato roots stored for marketing and processing respectively, high concentration (1000 ppm) of ethephon

should be avoided as it contributed to higher weight loss and proximal decay. Consequently integrated storage, comprising use of fungicides together with the ethephon foliar can be beneficial in reducing the proximal decay.

Recommendations and Suggestions for Further Research

The treatment used in this study is one of the upcoming methods being used generate ethylene which regulates the growth and development of different plants and crops. Preharvest ethephon treatment (500 ppm) performed better than the other treatments in most determinations conducted. The knowledge on the effects of ethephon on *Apomuden* sweet potatoes roots provides the much needed postharvest data and information on pretreatments to improve the shelf life of OFSP.

More research can be done to determine the optimal day of application for OFSP as the study was based only on one week before and after harvesting. Ethephon can also be used on other sweet potato varieties and other root crops to compare its effect on their shelf life and quality performance. Ethephon may be applied with sprout inhibitors like 1-MCP to determine the effect on sprouting and quality. Finally, the combined effects of ethephon and fungicide should be studied against decay.

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APPENDICES

APPENDIX A

Preharvest Ethephon Treatment ANOVA Tables

Table A1: *ANOVA Table for the Effect of Ethephon on Mean Percentage Weight Loss of OFSP Treated 7days Before Harvesting and Stored for 54 Days at Mean Temperature of 26.3°C and Relative Humidity of 88.7% (Fig.16)*

Source of variation	d.f	s.s	m.s	v.r	F pr
Treatment	3	1830.842	610.281	44.58	<0.001
Block	1	82.012	82.012	5.99	0.015
Treatment. Block	3	179.055	59.685	4.36	0.005
Residual	232	3176.24	13.691	19.82	
Time	5	7191.397	1438.279	2082.32	<0.001
Time. Treatment	15	107.104	7.140	10.34	<0.001
Time. Block	5	18.509	3.702	5.36	0.012
Time. Treatment. Block	15	51.110	3.407	4.93	<0.001
Residual	780	538.753	0.691		
Total	1059	6588.90			

Table A2: *ANOVA Table for the Effect of Ethephon on Mean Moisture Content of OFSP Treated 7days before Harvesting and Stored for 54 Days at Mean Temperature of 26.3°C and Relative Humidity of 88.7% (Fig 17)*

Source of variation	df	s.s	m.s	v.r	F pr
Subject stratum					
Treatment	3	61.806	20.602	4.58	0.008
Block	1	77.496	77.496	17.24	<0.001
Treatment. Block	3	31.154	10.385	2.31	0.091
Residual	40	179.817	4.495	1.01	
Subject. Time stratum					
Time	4	48.798	12.2	2.74	0.04
Time. Treatment	12	58.521	4.877	1.1	0.37
Time. Block	4	9.504	2.376	0.53	0.681
Time. Treatment. Block	12	35.051	2.921	0.66	0.764
Residual	152	676.443	4.45		
Total	231	1148.7			

Table A3: *ANOVA table for the effect of ethephon on mean percentage sprouting index of OFSP treated 7days before harvesting and stored for 54 days at mean Temperature of 26.3°C and Relative Humidity of 88.7% (Fig. 19)*

Source of variation	d.f	s.s	m.s	v.r	F pr
Subject stratum					
Treatment	3	8758.7	2919.6	2.96	0.098
Block	1	357.1	357.1	0.36	0.564
Treatment. Block	3	7582.5	2527.5	2.56	0.128
Residual	8	7885.7	985.7	8.76	
Subject. Time stratum					
Time	6	21785.7	3631	32.26	<0.001
Time. Treatment	18	5674.6	315.3	2.8	0.034
Time. Block	6	2520.6	420.1	3.73	0.036
Time. Treatment. Block	18	1784.1	99.1	0.88	0.541
Residual	48	5403.2	112.6		
Total	111	61752.4			

Table A4: *ANOVA Table for the Effect of Ethephon on Mean Sprout Length of OFSP Treated 7days before Harvesting and Stored for 54 Days at Mean Temperature of 26.3°C and Relative Humidity of 88.7% (Fig. 20)*

Source of variation	d.f	s.s	m.s	v.r	F pr
Subject stratum					
Treatment	3	18593.1	6197.7	4.29	0.006
Block	1	6226.8	6226.8	4.31	0.039
Treatment. Block	3	26496.6	8832.2	6.11	<0.001
Residual	232	335444.8	1445.9	3.24	
Subject. Time stratum					
Time	6	429514.7	71585.8	160.36	<0.001
Time. Treatment	18	12450.2	691.7	1.55	0.195
Time. Block	6	22213.7	3702.3	8.29	0.003
Time. Treatment. Block	18	34372	1909.6	4.28	0.004
Residual	1288	574983.4	446.4		
Total	1575	1380468.3			

Table A5: *ANOVA Table For the Effect of Ethephon On The Mean Number Sprouts Of OFSP Treated 7days Before Harvesting and Stored for 54 Days at Mean Temperature of 26.3°C and Relative Humidity of 88.7% (Fig. 21)*

Source of variation	d.f	s.s	m.s	v.r	F pr
Subject stratum					
Treatment	3	371.266	123.755	11.34	<0.001
Block	1	0.008	0.008	0	0.979
Treatment. Block	3	218.311	72.77	6.67	<0.001
Residual	232	2532.608	10.916	8.27	
Subject. Time stratum					
Time	6	1980.93	330.155	250	<0.001
Time. Treatment	18	88.945	4.941	3.74	<0.001
Time. Block	6	56.219	9.37	7.09	<0.001
Time. Treatment. Block	18	65.729	3.652	2.77	0.003
Residual	996	1315.355	1.321		
Total	1283	6001.672			

Table A6: *ANOVA Table for the Effect of Ethephon on Mean Percentage Root Decay of OFSP Treated 7days before Harvesting and Stored for 54 Days at Mean Temperature of 26.3°C and Relative Humidity of 88.7% (Fig. 22)*

Source of variation	d.f	s.s	m.s	v.r	F pr
Subject stratum					
Treatment	3	12423.61	4141.2	7.54	0.01
Block	1	337.5	337.5	0.61	0.456
Treatment. Block	3	2779.17	926.39	1.69	0.247
Residual	8	4396.3	549.54	8.9	
Subject. Time stratum					
Time	5	29396.76	5879.35	95.2	<0.001
Time. Treatment	15	3623.61	241.57	3.91	0.004
Time. Block	5	259.72	51.94	0.84	0.484
Time. Treatment. Block	15	1279.17	85.28	1.38	0.252
Residual	40	2470.37	61.76		
Total	95	56966.2			

Table A7: *ANOVA Table for the Effect of Ethephon on Average Severity of Root Decay of OFSP Treated 7days Before Harvesting and Stored for 54 Days at Mean Temperature of 26.3°C and Relative Humidity of 88.7% (Fig. 23)*

Source of variation	d.f	s.s	m.s	v.r	F pr
Subject stratum					
Treatment	3	172.3727	57.4576	15.12	<0.001
Block	1	2.5005	2.5005	0.66	0.418
Treatment. Block	3	39.6833	13.2278	3.48	0.017
Residual	232	881.3435	3.7989	5.66	
Subject. Time stratum					
Time	6	610.4336	101.7389	151.55	<0.001
Time. Treatment	18	70.6485	3.9249	5.85	<0.001
Time. Block	6	7.9058	1.3176	1.96	0.161
Time. Treatment. Block	18	31.7163	1.762	2.62	0.044
Residual	1272	853.925	0.6713		
Total	1559	2225.4744			

Table A8: *ANOVA Table for the Effect of Ethephon on Mean Percentage Shrinkage of OFSP Treated 7days Before Harvesting and Stored for 54 Days at Mean Temperature of 26.3°C and Relative Humidity of 88.7% (Fig. 18)*

Source of variation	d.f	s.s	m.s	v.r	F pr
Subject stratum					
Treatment	3	63.69077	21.23026	7.18	<0.001
Block	1	7.97336	7.97336	2.69	0.102
Treatment. Block	3	13.06145	4.35382	1.47	0.223
Residual	232	686.43082	2.95875	35.08	
Subject. Time stratum					
Time	5	609.37416	121.87483	1445.12	<0.001
Time. Treatment	15	6.51192	0.43413	5.15	<0.001
Time. Block	5	0.50024	0.10005	1.19	0.306
Time. Treatment. Block	15	1.92779	0.12852	1.52	0.171
Residual	773	65.19133	0.08434		
Total	1052	881.06289			

APPENDIX B

ANOVA Tables for Postharvest Treatments

Table B1: *ANOVA Table for the Effect of Post-Harvest Ethephon Application on Mean Percentage Weight Loss of OFSP Treated 7days after Harvesting and Stored for 56 days at Temperature of 26.3°C and Relative Humidity of 88.7% (Fig. 25)*

Source of variation	d.f	s.s	m.s	v.r	F pr
Subject stratum					
Treatment	3	193.444	64.481	1.29	0.282
Residual	116	5806.71	50.058	12.56	
Subject. Time stratum					
Time	6	12116.5	2019.41	506.64	<0.001
Time. Treatment	18	87.408	4.856	1.22	0.306
Residual	623	2483.22	3.986		
Total	766	18125.9			

Table B2: *ANOVA Table for the Effect of Ethephon Application on Mean Percentage Sprouting Index of OFSP Treated 7days after Harvesting and Stored for 56 Days at Mean Temperature of 26.3°C and Relative Humidity of 88.7% (Fig. 27)*

Source of variation	d.f	s.s	m.s	v.r	F pr
Subject stratum					
Treatment	4	4312.38	1078.1	1.9	0.205
Residual	8	4550	568.75	8.89	
Subject. Time stratum					
Time	7	88105.2	12586.5	196.7	<0.001
Time. Treatment	28	4124.44	147.3	2.3	0.06
Residual	56	3583.33	63.99		
Total	103	104675			

Table B3: *ANOVA Table for the Effect of Ethephon Application on Mean Sprout Length of OFSP Treated 7days After Harvesting and Stored For 56 Days at Mean Temperature of 26.3°C and Relative Humidity of 88.7% (Fig. 28)*

Source of variation	d.f	s.s	m.s	v.r	F pr
Subject stratum					
Treatment	3	1758.7	586.2	0.61	0.613
Residual	116	112287	968	4.37	
Subject. Time stratum					
Time	7	240826	34403.7	155.45	<0.001
Time. Treatment	21	11922.7	567.7	2.57	0.033
Residual	727	160897	221.3		
Total	874	500329			

Table B4: *ANOVA Table for the Effect of Ethephon Application on Mean Number of Sprouts on OFSP Treated 7days after Harvesting and Stored for 56 Days at Mean Temperature of 26.3°C and Relative Humidity of 88.7% (Fig. 29)*

Source of variation	d.f	s.s	m.s	v.r	F pr
Subject stratum					
Treatment	3	11.729	3.91	0.71	0.545
Residual	116	634.891	5.473	5.46	
Subject. Time stratum					
Time	7	1422.73	203.247	202.69	<0.001
Time. Treatment	21	44.606	2.124	2.12	0.012
Residual	727	728.992	1.003		
Total	874	2746.29			

Table B5: *ANOVA Table for the Effect of Ethephon Application on Mean Percentage Decay Incidence of OFSP Treated 7days after Harvesting and Stored for 56 Days at Mean Temperature of 26.3°C and Relative Humidity of 88.7% (Fig. 31)*

Source of variation	d.f	s.s	m.s	v.r	F pr
Subject stratum					
Treatment	3	3308.3	1102.8	2.7	0.116
Residual	8	3266.7	408.3	2.43	
Subject. Time stratum					
Time	7	20550	2935.7	17.49	<0.001
Time. Treatment	21	8525	406	2.42	0.099
Residual	56	9400	167.9		
Total	95	45050			

Table B6: *ANOVA Table for the Effect of Ethephon Application on Mean Decay Severity of OFSP Treated 7days after Harvesting and Stored for 56 Days at Mean Temperature of 26.3°C and Relative Humidity of 88.7% (Fig. 32)*

Source of variation	d.f	s.s	m.s	v.r	F pr
Subject stratum					
Treatment	3	8.3328	2.7776	1.09	0.354
Residual	116	294.379	2.5377	9.65	
Subject. Time stratum					
Time	7	64.3159	9.188	34.95	<0.001
Time. Treatment	21	7.9951	0.3807	1.45	0.223
Residual	788	207.176	0.2629		
Total	935	493.691			

Table B7: *ANOVA Table for the Effect of Ethephon Application on Mean Percentage Shrinkage of OFSP Treated 7days after Harvesting and Stored for 56 Days at Mean Temperature of 26.3°C and Relative Humidity of 88.7% (Fig. 26)*

Source of variation	d.f	s.s	m.s	v.r	F pr
Subject stratum					
Treatment	4	5370.9	1342.72	9.34	<0.001
Residual	110	15820.6	143.824	16.56	
Subject. Time stratum					
Time	4	290.151	72.538	8.35	<0.001
Time. Treatment	16	1061.86	66.366	7.64	<0.001
Residual	376	3266.49	8.687		
Total	510	24670.9			

APPENDIX C

ANOVA Tables for Biochemical Changes in the Preharvest Treated Roots

Table C1: *ANOVA Table for the Effect of Ethephon on Mean Percentage Dry Matter of OFSP Treated 7days before Harvesting and Stored for 54 Days at Mean Temperature of 26.3°C and Relative Humidity of 88.7% (Fig. 34)*

Source of variation	d.f	s.s	m.s	v.r	F pr
Subject stratum					
Treatment	3	51.199	17.066	4.66	0.007
Block	1	64.876	64.876	17.71	<0.001
Treatment. Block	3	25.863	8.621	2.35	0.086
Residual	40	146.515	3.663	1	
Subject. Time stratum					
Time	5	19990.997	3998.199	1087.22	<0.001
Time. Treatment	15	68.543	4.57	1.24	0.265
Time. Block	5	22.673	4.535	1.23	0.3
Time. Treatment. Block	15	40.8	2.72	0.74	0.697
Residual	192	706.072	3.677		
Total	279	20967.417			

Table C2: *ANOVA Table for the Effect ff Ethephon Application on Mean Starch Concentration (g/100g) of OFSP Roots Treated 7days before Harvesting and Stored for 42 Days at Mean Temperature of 26.3 °C and Relative Humidity of 88.7 % (Fig. 35)*

Source of variation	d.f	s.s	m.s	v.r	F pr
Treatment	3	150.06	50.02	2.48	0.062
Days	4	103.19	25.8	1.28	0.279
Block	1	207.17	207.17	10.28	0.002
Treatment. Days	12	425.12	35.43	1.76	0.058
Treatment. Block	3	91.11	30.37	1.51	0.214
Days. Block	4	73.07	18.27	0.91	0.462
Treatment. Days. Block	12	344.63	28.72	1.42	0.158
Residual	190	3830.76	20.16		
Total	229	5163.72			

Table C3: *ANOVA table for the effect of ethephon application on mean sucrose Concentration (g/100g) of OFSP Roots Treated 7days before Harvesting and Stored for 42 Days at Mean Temperature of 26.3 °C and Relative Humidity of 88.7 % (Fig. 36)*

Source of variation	d.f	s.s	m.s	v.r	F pr
Treatment	3	82.96	27.65	2.21	0.088
Days	4	275.11	68.78	5.5	<0.001
Block	1	172.57	172.57	13.81	<0.001
Treatment. Days	12	150.15	12.51	1	0.449
Treatment. Block	3	157.31	52.44	4.2	0.007
Days. Block	4	21.9	5.47	0.44	0.781
Treatment. Days. Block	12	197.68	16.47	1.32	0.211
Residual	191	2386.64	12.5		
Total	230	3416.41			

Table C4: *ANOVA table for the effect of ethephon application on mean glucose Concentration (g/100g) of OFSP Roots Treated 7days before Harvesting and Stored for 42 Days at Mean Temperature of 26.3 °C and Relative Humidity of 88.7 % (Fig. 37)*

Source of variation	d.f	s.s	m.s	v.r	F pr
Treatment	3	57.915	19.305	7.93	<0.001
Days	4	41.619	10.405	4.27	0.002
Block	1	0.439	0.439	0.18	0.671
Treatment. Days	12	33.019	2.752	1.13	0.338
Treatment. Block	3	3.928	1.309	0.54	0.657
Days. Block	4	21.41	5.352	2.2	0.071
Treatment. Days. Block	12	25.382	2.115	0.87	0.58
Residual	188	457.63	2.434		
Total	227	635.887			

Table C5: *ANOVA table for the effect of ethephon application on mean fructose Concentration (g/100g) of OFSP Roots Treated 7days before Harvesting and Stored for 42 Days at Mean Temperature of 26.3 °C and Relative Humidity of 88.7 % (Fig. 38)*

Source of variation	d.f	s.s	m.s	v.r	F pr
Treatment	3	37.939	12.646	7.86	<0.001
Days	4	16.592	4.148	2.58	0.039
block	1	0.385	0.385	0.24	0.625
Treatment. Days	12	25.811	2.151	1.34	0.201
Treatment. block	3	3.798	1.266	0.79	0.503
Days. block	4	16.727	4.182	2.6	0.038
Treatment. Days. block	12	20.705	1.725	1.07	0.386
Residual	191	307.375	1.609		
Total	230	425.938			

Table C6: *ANOVA table for the effect of ethephon application on mean protein Concentration (g/100g) of OFSP Roots Treated 7days before Harvesting and Stored for 42 Days at Mean Temperature of 26.3 °C and Relative Humidity of 88.7 % (Fig. 39)*

Source of variation	d.f	s.s	m.s	v.r	F pr
Treatment	3	49.59	16.53	17.63	<0.001
Days	4	2.2032	0.5508	0.59	0.672
Block	1	7.0964	7.0964	7.57	0.007
Treatment. Days	12	12.5839	1.0487	1.12	0.347
Treatment. Block	3	7.5524	2.5175	2.69	0.048
Days. Block	4	11.2921	2.823	3.01	0.019
Treatment. Days. Block	12	5.0665	0.4222	0.45	0.94
Residual	191	179.041	0.9374		
Total	230	272.83			

Table C7: *ANOVA table for the effect of ethephon application on mean iron Concentration (g/100g) of OFSP Roots Treated 7days before Harvesting and Stored for 42 Days at Mean Temperature of 26.3 °C and Relative Humidity of 88.7 % (Fig. 40)*

Source of variation	d.f	s.s	m.s	v.r	F pr
Treatment	3	2.69058	0.89686	14.57	<0.001
Days	4	4.10449	1.02612	16.67	<0.001
block	1	0.01026	0.01026	0.17	0.684
Treatment. Days	12	0.60358	0.0503	0.82	0.633
Treatment. block	3	0.99942	0.33314	5.41	0.001
Days. block	4	0.78724	0.19681	3.2	0.014
Treatment. Days. block	12	0.4925	0.04104	0.67	0.782
Residual	191	11.7595	0.06157		
Total	230	21.3371			

Table C8: *ANOVA table for the effect of ethephon application on mean zinc Concentration (g/100g) of OFSP Roots Treated 7days before Harvesting and Stored for 42 Days at Mean Temperature of 26.3 °C and Relative Humidity of 88.7 % (Fig. 41)*

Source of variation	d.f.	s.s.	m.s.	v.r.	F pr.
Treatment	3	0.56579	0.18860	6.70	<0.001
Days	4	0.1687	0.04217	1.50	0.204
Block	1	0.0196	0.01960	0.70	0.405
Treatment. Days	12	0.31008	0.02584	0.92	0.530
Treatment. Block	3	0.28359	0.09453	3.36	0.020
Days. Block	4	0.29061	0.07265	2.58	0.039
Treatment. Days. Block	12	0.20896	0.01741	0.62	0.825
Residual	191	5.37627	0.02815		
Total	230	7.14546			