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Antifungal and antibacterial activity of some medicinal plants used traditionally in Kenya

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Abstract. *Jacqueline I, Okemo P, Maingi J, Bii C. 2018. Antifungal and antibacterial activity of some medicinal plants used traditionally in Kenya. Asian J Ethnobiol 2: 75-90.* Many plants have been used by various communities in Kenya in the treatment of bacterial and fungal infections but they have not been validated. The aim of this study was to determine the efficacy of some medicinal plants used by various communities in Kenya that treat the selected bacterial and the selected fungal diseases in man. An ethnobotanical survey was used to select and collect plants from Mwingi North, Kisii South and Rarieda Districts based on their use to treat infectious diseases such as skin infection, diarrhea and many others. Crude extracts from *Zanthoxylum chalybeum, Boscia angustifolia, Melia volkensii, Zanthoxylum gilletii, Fuerstia africana, Urtica dioica, Vernonia amygdalina, Ricinus communis, Commiphora africana, Psiadia punctulata, Senna didymobotrya, Ormocarpum trichocarpum, Sesbania sesban, Balanites aegyptiaca, Albizia coriaria, Ficus sycomorus, Rhus natalensis* and *Tamarindus indica* believed to contain secondary metabolites were screened against ten microorganisms, including the bacteria: *Salmonella typhi* ATCC 19430, *Escherichia coli* ATCC 25922, *Bacillus subtilis, Staphylococcus aureus* ATCC 25923 and Methicillin-resistant *S. aureus* (MRSA). The fungal strains that were used are; *Aspergillus niger*, *Candida albicans* ATCC 90028, *Microsporum gypseum, Cryptococcus neoformans* ATCC 18310 and *Trichophyton mentagrophyte.* The plants were screened using Kirby Bauer disc diffusion method. Phytochemical screening was carried out to identify the presence or absence of classes of bioactive compounds. Data were analyzed using one way ANOVA, significant means were separated using Tukey's test. Generally, *F. africana, Z. chalybeum, B. aegyptiaca, O. trichocarpum, S. didymobotrya* and *T. indica* gave strong antibacterial results of between 14.5 mm and 20 mm as *A. coriaria, F. sycomorus, C. africana, R. natalensis, S. didymobotrya, P. punctulata,* and *T. indica* produced strong antifungal results of between 15.5 mm and 20.5 mm. The results of MICs and the MBCs/MFCs of the extracts of *A. coriaria, F. sycomorus, S. didymobotrya, P. punctulata, F. africana, B. aegyptiaca* and *T. indica* showed good activity of 0.9375 mg/mL in some test cultures. *S. typhi* ATCC 19430 and *E. coli* ATCC 25922 were the least sensitive bacteria while *C. albicans* ATCC 90028 was the least sensitive fungus. The present study indicates that the majority of the plants tested are an important source of antibacterial agents, especially on Gram-positive bacteria (*S. aureus, B. subtilis* and MRSA) and antifungal agents against the dermatophytes, especially *M. gypseum.* This study recommends that the plant extracts with good antimicrobial activity be subjected to pharmacological and toxicological studies.

Keywords: Kenya, medicinal plants, selected bacterial

INTRODUCTION

Around the world, infectious diseases are the major cause of death (Bandow et al. 2003; Parekh and Chanda 2007). Every year, the biggest infectious illnesses account for more than 11 million deaths (WHO 2005). Infectious diseases account for half of all deaths in tropical countries (Okigbo and Mmeka 2008; Assob et al. 2011). Diseases caused by pathogenic bacteria and fungi remain a major public health concern, particularly in developing countries, due to a variety of factors, such as the emergence of bacterial and fungal strains that are resistant to the majority of commonly used antibiotics; the emergence of pathogenic bacteria and fungi that are resistant to the majority of commonly used antibiotics (Abad et al. 2007; WHO 2007). Conventional medications are too expensive, and western health institutions are inaccessible to rural populations (Matu and Staden 2003; Wagate et al. 2008).

Multiple drug resistance has arisen due to the haphazard application of antimicrobials and the re-emergence of diseases caused by genetically diverse microorganisms (Islam et al. 2006; Wagate et al. 2010). As a result of drug resistance, researchers were forced to look for new antibacterial agents from other sources, such as plants (Pirbalouti et al. 2010). Furthermore, because people have evolved alongside plants and our digestive system and physiology are tuned to digesting and utilizing plant-based foods with medicinal value, herbal treatments are far more effective than chemical chemicals in treating human ailments (Farooq 2005).

Medicinal plants have been utilized to treat and prevent human illnesses since the beginning of time because they contain components that have medicinal potential (Parekh and Chanda 2007; Khodadadi et al. 2015). Animals in natural settings, both domesticated and non-domesticated, automatically treat themselves when sick by consuming various components of medicinal plants, such as leaves, stems, bark, and roots (Sindiga et al. 1995). Furthermore, they can heal their skin ailments by vigorously rubbing themselves against medicinal plants that are suitable for

their condition (Sindiga et al. 1995). According to research, diets rich in plant-based foods and beverages are connected with a lower risk of developing chronic diseases (Njoroge et al. 2012).

WHO believes that up to 80% of the global population uses plants as their primary source of health treatment (Doughari 2006; Turker and Usta 2008; Verma et al. 2011). Medicinal compounds have been isolated from fungi and higher plants and have proven reliable sources of active ingredients (Olila et al. 2001). Antimalarial, anti-cancer, anti-diabetic, and antibiotic chemicals like atropine and ergometrine, isolated from medicinal plants, are among the most effective medicines (Olila et al. 2001; Samie et al. 2005). In addition, many of the active chemicals used in pharmaceuticals are derived from medicinal plants (Maundu and Tengnas 2005). In light of these findings, it's a good idea to examine local plants that have been utilized to cure illnesses of this nature (Atindehou et al. 2002).

Traditional medicine relies heavily on utilizing medicinal plants, which is a highly profitable business in the global market (WHO 2008). Annual sales of herbal medicine and other plant products in Western Europe amounted to US$5 billion in 2003-2004, US$14 billion for China in 2005, US$160 million in Brazil, and US$100 million in Mexico in 2007 (WHO 2008). Every market in the urban and peri-urban areas of Africa sells medicinal plant parts (Rukangira 2001).

The medicinal value of plants used in traditional medicine is derived from the chemical components, which can treat chronic and frequent bacterial illnesses, respectively (Kareru et al. 2008). In plants, these chemicals are secondary metabolites that act as defensive agents against invading microorganisms and predators (Ghdeib and Shtayeh 1999) and aid in the regulation of plant growth (Nwodo et al. 2010). A variety of secondary metabolites found in plants include alkaloids, steroid hormones, tannins, phenol compounds, flavonoids, and fatty acids (Ashokkumar et al. 2010).

The alarming rate at which new and re-emerging infectious diseases are forming and spreading is a cause for concern. In addition, the development of resistance to antibiotics now in clinical use is another issue that must be addressed (Parekh and Chanda 2007). The prevalence of antibiotic resistance among pathogenic bacteria is predicted to be greater than 70%, with at least one of the antibiotics usually used to treat them being particularly high (Okemo et al. 2011). Due to this, there is an urgent and ongoing need to discover novel antimicrobial compounds with diverse chemical structures and novel modes of action (Parekh and Chanda 2007) and the need to investigate antimicrobial chemicals from alternative sources such as plants (Doughari 2006; Duraipandiyan et al. 2006; Parekh and Chanda 2007).

Many studies on the usage of medicinal plants in Kenya have been conducted, with diverse communities being the focus of the research (Maundu and Tengnas 2005; Kokwaro 2009). The ethnobotany of the Kisii, Rarieda, and Mwingi Districts, on the other hand, is relatively unknown. As a result, additional research should be conducted on plants from the Mwingi district. The rich richness of plants present in the Kisii District provides tremendous opportunities to discover natural products containing antibacterial chemicals. The residents of the Rarieda district also have a strong cultural belief in traditional medicine, which is reflected in their attire (Osewe 2011).

The following goals were set for this study: (i) to determine the antifungal activity of crude plant extracts against standard strains of *Candida albicans*, *Aspergillus niger*, *Trichophyton mentagrophyte*, *Cryptococcus neoformans*, and *Microsporum gypseum*; and (ii) to determine the antifungal activity of crude plant extracts against standard strains of *C. albicans, A. niger, T. mentagrophyte*; (ii) to determine the minimum inhibitory concentrations (MICs), minimum bactericidal concentrations (MBCs), and minimum fungicidal concentrations (MFCs) of crude extracts exhibiting antimicrobial activity against the selected bacterial and fungal pathogens; (iii) to identify the phytochemicals present in the crude extracts and quantify their concentrations.

MATERIALS AND METHODS

Plant materials

A survey was conducted in Katse (Mwingi North), Iyabe and Kerina (Kisii South), Lweya, and Ragengni Divisions in the Rarieda District of Kenya (Figure 1) to identify the most commonly utilized medicinal plants for the treatment of fungal illnesses.

The selection of the plants was based on the ethnobotanical knowledge of the consulted herbalists and the accessible literature. With the help of a semi-structured questionnaire, interviews were performed to acquire indigenous knowledge about plants utilized for the survey. Herbalists who are knowledgeable practitioners were found with the aid of locals and the local authorities and selected as survey respondents (Yinerger and Yewhalaw 2007).

Before conducting the interview, however, formal permission was obtained from each informant (Kasolo et al. 2010). In-home interviews were conducted with them. In the study, respondents were asked for the local names of the medicinal plants they used, the sections utilized, the ailments targeted, the method of preparation, the administration of the resulting preparations, and the availability of the plants.

The species choice-value model was favored because it uses a proportion of all informants who cite a certain number of species for a certain category to determine the most often utilized medicinal plant species (Yinerger and Yewhalaw 2007). Therefore, each prescription was only regarded as following valid verification by three independent sources.

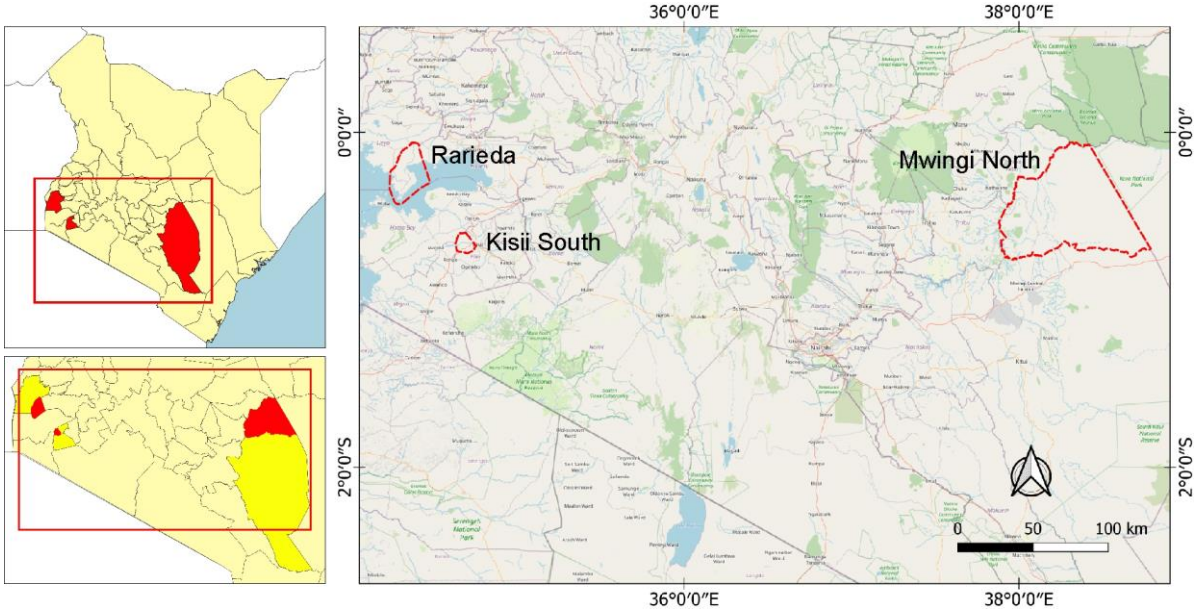

Figure 1. Map of Kenya showing Kisii South, Rarieda, and Mwingi North Districts

Collection and authentication of plant material

In this study, eighteen different medicinal plants, namely, *Zanthoxylum chalybeum* Engl., *Boscia angustifolia* A. Rich, *Melia volkensii* Gürke, *Zanthoxylum gilletii* (De Wild.) Waterman, *Fuerstia africana* T.C.E.Fr., *Urtica dioica* L., *Vernonia amygdalina* Delile, *Ricinus communis* L., *Commiphora africana* (Rich.) Engl., *Psiadia punctulata* (DC.) Oliv. & Hiern ex Vatke, *Senna didymobotrya* (Fresen.) H.S.Irwin & Barneby, *Ormocarpum trichocarpum* (Taub.) Engl., *Sesbania sesban* L., *Balanites aegyptiaca* (L.) Delile, *Albizia coriaria* Welw. ex Oliv., *Ficus sycomorus* L., *Rhus natalensis* Bernh. ex Krauss, and *Tamarindus indica* L. were obtained in Kisii, Rarieda, and Mwingi. The plants gathered were those used to treat gastroenteritis, respiratory ailments, and skin disorders. Fresh plant parts, such as the roots, leaves, and bark, were gathered based on the most frequently employed by traditional healers. Photographs were taken of the selected plant species. A plant taxonomist from the Botany Department, Chiromo, University of Nairobi, Kenya, identified the plants. Collecting voucher specimens and depositing them in the University of Nairobi's herbarium.

Preparation of plant materials

The freshly collected plant parts were thoroughly washed with running tap water, chopped into smaller pieces, and then dried under shade at room temperature for two weeks until completely dry (Matu et al. 2012). Finally, the small pieces were ground into powder at the Botany Department, Chiromo University of Nairobi, using a Wiley mill (model no. 2, USA).

Extraction

The extraction was done at the University of Nairobi, in the Chiromo Department of Chemistry. In a conical flask, 150 grams of each dry powder were mixed with 1000 ml of a 1:1 mixture of methanol and dichloromethane and left overnight (Midiwo 2010; Kitonde et al. 2013). The extracts were then put through a Whatman filter paper No. 1.

Concentration into the solid sample

Dichloromethane and methanol were evaporated in a water bath set to 40°C with a rotary evaporator (Buchii B-205 Switzerland) to make dry organic crude extracts (Omwenga et al. 2009). The paste was put into small vials, then put in an evacuated desiccator with anhydrous copper sulfate to make dry powdered samples. If a bioassay was not done right away, all of the samples were kept in the fridge at a temperature of 4°C.

Microbial test organisms

The microbial test organisms were chosen based on the gathered ethnobotanical information about the disease of interest, their importance as opportunistic pathogens, and their resistance to standard drugs. The chosen microorganisms also cause common infectious diseases that are easy to spread. The standard reference microorganisms and clinical isolates came from the center for Microbiology Research (KEMRI) in Nairobi, Kenya. In this study, the following standard reference microorganisms, environmental microorganisms, and clinical isolates were used:

Fungal isolates

Yeast

C. albicans – ATCC 90028. (i)

C. neoformans – ATCC 18310. (ii)

Dermatophytes

M. gypseum – Clinical isolates. (i)

T. mentagrophytes – Clinical isolates. (ii)

Filamentous fungi

A. niger – Environmental organism. (i)

Maintenance of microbial stock cultures

On Muller Hinton agar no. CMO337, bacterial strains that were already in stock, were grown again (Oxoid Ltd, Basingstoke, Hampshire, England). To get strains that were starting to grow, they were kept at 37°C for 24 hours (Omwenga et al. 2009). Yeasts and molds were grown on Sabaraud Dextrose Agar No. CM 004 by subculturing (Oxoid Ltd Basingstoke, Hampshire, England). Cruz et al. (2007) said that the yeasts were kept at 37°C for 24 hours, the filamentous fungus was kept at 28°C for 48 hours in a humid chamber, and the dermatophytes were kept at 25°C for 72 hours (Korir et al. 2012b). The bacterial and fungal strains were both maintained at a temperature of 4°C.

Antibacterial assays

The antibacterial bioassay was performed by the Kirby Bauer disk diffusion method (Omori et al. 2012). Muller Hinton agar no. CMO337 (Oxoid Ltd, Basingstoke, Hampshire, England) was prepared according to the manufacturer's instructions to culture bacteria. Normal saline solution was used to dilute fresh 24 h culture of bacterial type cultures or clinical isolates to attain a 0.5 McFarland Standard, which gives an equivalent approximate density of 1×10^8 of bacteria (Kitonde et al. 2013). The spread plate method was used to culture 100 µL of the bacterial suspension that was introduced into the petri dishes. Eighteen dry sterile discs (6 mm diameter) were soaked in 100 µL plant extract (made by dissolving 300 mg of each extract in 1000 µL (1 mL) of DMSO). The discs were air-dried and placed aseptically onto the inoculated plates at a distance of 3 mm apart. Discs impregnated with DMSO and then air-dried were used as negative controls, while commercially available discs of chloramphenicol were used as positive control for bacteria. Incubation was carried out at a temperature of 37°C for 24 h. All tests were performed in triplicate. After incubation, bacterial growth inhibition was determined by measuring the diameter zones in millimeters using a transparent ruler and recorded against the corresponding plant extracts (Omwenga et al. 2009; Mariita et al. 2010).

The Kirby Bauer disk diffusion method was used for the antibacterial bioassay (Omori et al. 2012). Oxoid Ltd., Basingstoke, Hampshire, England's Muller Hinton agar no. CMO337 was made according to the manufacturer's instructions so that bacteria could be grown on it. Normal saline solution was used to dilute fresh 24-hour cultures of bacterial type cultures or clinical isolates to reach a 0.5 McFarland Standard, equal to about 1108 bacteria per milliliter (Kitonde et al. 2013). The 100 L of bacterial suspension that was put into the petri dishes was grown using the spread plate method. First, 18 sterile, dry discs with a diameter of 6 mm were soaked in 100 L of plant extract, which was made by dissolving 300 mg of each extract in 1 mL of DMSO. Then, the discs were left to dry in the air and placed 3 mm apart on the inoculated plates cleanly. As negative controls, we used DMSO-soaked and air-dried discs. As positive controls for bacteria, we used discs of chloramphenicol that were available for purchase. During the 24 hours of incubation, the temperature was kept at 37°C. All of the tests were done in triplicate. After incubation, the effectiveness of the plant extracts in inhibiting the development of bacteria was evaluated by using a clear ruler to measure the zone diameter in millimeters and then recording the results against the matching plant extracts (Omwenga et al. 2009; Mariita et al. 2010).

Antifungal assays

To determine the antifungal activity of plant extracts against fungal strains, Sabourand Dextrose Agar no. CM OO4 (Oxoid Ltd, Basingstoke, Hampshire, England) was prepared in accordance with the manufacturer's instructions. Fresh 24 h cultures of fungal type or clinical isolates were diluted with normal saline solution to achieve a McFarland Standard of 0.5. The spread plate method was utilized to culture 100 µL of the fungal suspension that was added to the petri dishes. Eighteen dry, sterile discs (6 mm in diameter) were soaked in 100 µL of plant extract (prepared by dissolving 300 mg of each extract in 1000 µL of DMSO). The discs were air-dried and placed 3 mm apart aseptically on the inoculation plates. Negative controls were discs impregnated with DMSO and air-dried, whereas positive controls were discs containing commercially available miconazole (Bii et al. 2010). The inocula were incubated under conditions where yeast cultures were incubated at 37°C for 24 hours (Cruz et al. 2007), filamentous fungi at 28°C for 48 hours in humid chambers (Costa et al. 2010), and dermatophytes at 25°C for 72 hours in humid chambers (Korir et al. 2012b). In triplicate, all tests were conducted. After incubation, the inhibition of fungal growth was assessed by measuring the diameter zones in millimeters with a transparent ruler and recording the results against the relevant plant extracts (Matu et al. 2012).

Determination of minimum inhibitory concentration

In order to create plant extracts, 300 mg of each crude extract was dissolved in 1000 µL (1 mL) of DMSO. According to the National Committee for Clinical Laboratory Standards (NCCLS), now Clinical Standard Institute (CLSI), the minimum inhibitory concentration of the active crude extracts against the test microorganisms was determined using the broth microdilution method (Korir et al. 2012b).

The experiments were conducted in 96-well microtiter plates. Using serial doubling dilutions, the concentration in each successive well was halved relative to the concentration in the preceding well. Using the disc diffusion method, the MIC was determined only when the plant extract demonstrated high antibacterial activity (≤ 9 mm) (Mariita et al. 2010). Each well received 50 µL of Muller Hinton broth for bacterial strains and 50 µL of Sabourand Dextrose broth. Then, 50 µL of the plant extract (made by dissolving 300 mg of each extract in 1 mL of DMSO) was added to the first well before serial dilutions. The serial dilutions were carried out by transferring 50 µL of the extract-containing Muller Hinton or Sabourand Dextrose broth from the first well to the second, third, and fourth wells. Then, fifty microliters of each test isolate were applied to each well. One row of wells served as the

negative control for microorganism growth in the medium, while 50 µL of the antibiotic (Chloramphenicol/miconazole) served as the positive control. Finally, coated microtitres were applied to plates. In humid chambers, bacteria and yeasts were incubated at 37°C for 24 hours (Kitonde et al. 2013), filamentous fungus at 28°C for 48 hours (Costa et al. 2010), and dermatophytes at 25°C for 72 hours (Korir et al. 2012b). Minimum Inhibitory Concentrations (MIC) were determined by noting the lowest concentration of active extracts that inhibited microbiological growth in comparison to the turbidity of the control broth (Kitonde et al. 2013).

Determination of minimum bactericidal / fungicidal concentrations (MBCs/ MFCs)

Bacteria were subcultured on Mueller Hinton Agar, and fungi were subcultured on Sabourand Dextrose Agar from the wells where MIC findings indicated no growth (not turbid). The bacterial and yeast cultures were incubated at 37°C for 24 hours and at 25°C for 72 hours, respectively (Korir et al. 2012b). The MBC is the lowest concentration of plant extracts that did not produce any colony on a solid medium after sub-culturing and incubation for 24 hours for bacteria, 72 hours for dermatophytes, and 24 hours for yeasts. Each test was conducted in triplicate (Samie et al. 2010; Omori et al. 2012).

Statistical analysis

The data were analyzed with Minitab Statistical Software 13.20, 2000. The data analyzed consisted of the average zone of inhibition values for each test culture acquired from the antibacterial and antifungal assays and expressed as standard deviation means. A one-way ANOVA with a 95% confidence interval was performed to assess the significance between the groups. To determine whether there were significant variations between group means, the Tukey's test was run, and a probability value of ≤ 0.05 was considered significant (Mariita et al. 2010).

RESULTS AND DISCUSSION

Ethnobotanical survey

The results of the ethnobotanical survey are presented in Table 1, in which the plants are arranged in alphabetic synopsis according to families. The local names, parts used, drug preparation methods, diseases treated, and the areas where the plants collected are also presented in Table 1.

In this study, 18 species distributed in 15 families were identified. The family reported with the highest number of medicinal plant species was Fabaceae (3 species). It was followed by Rutaceae (2 species). Capparidaceae, Meliaceae, Lamiaceae, Urticaceae, Asteraceae, Moraceae, Balantiaceae, Papilionaceae, Caesalpinaceae, Burseraceae, Compositae, Anacardiaceae, and Euphorbiaceae had one species each.

Eleven plants were collected from Lweya and Ragengni (Rarieda), 4 from Kerina and Iyabe (Kisii South), and 3 from Mwingi North. Various parts were harvested depending on the parts the communities preferred to use to treat various ailments. The most frequently used preparations for administration were concoctions and decoctions. Only 2 plants were preferred by infusion.

The leaves (9 plants) were the most frequently used parts of the plant, followed by the bark (5 plants) and then roots (4 plants). The diseases that were most frequently treated using herbal medicines were gastrointestinal (37%), respiratory (31%), other ailments (18%), skin infection (6%), urinary tract (4%), and wounds (4%) (Figure 2).

Antibacterial activity of the plant crude extracts against standard strains of bacteria

The results indicated that all the plant extracts tested had bacterial inhibitory effects. However, the inhibition of bacteria by the plant extracts varied with different plants. The zones of inhibition between 7 and 10 mm were referred to as low/weak activity, between 11 and 14 moderate activity and between 15 and 21 high/ strong activity.

The plant extracts significantly inhibited the growth of *Bacillus subtilis* and *Staphylococcus aureus* ATCC 25922 compared to other strains of bacteria (Table 2). A total of five plant extracts had high activity against *B. subtilis*. These include; *F. africana* (17 mm), *S. didymobotrya* (16 mm), *O. trichocarpum* (15.5 mm), *Z. chalybeum* (15 mm) and *T. indica* (15 mm), while nine plant extracts had a moderate activity of between 11 and 13 against *B. subtilis*, *R. communis* (13 mm), *B. angustifolia* (13 mm), *A. coriaria* (12 mm), *B. aegyptiaca* (12 mm), *C. africana* (12 mm), *R. natalensis* (11.5 mm), *S. sesban* (11 mm), *P. punctulata* (11 mm) and *Z. chalybeum* (11 mm). The remaining four plant extracts showed weak activity against *B. subtilis*. The *M. volkensii*, *F. sycomorus*, *U. dioica* and *V. amygdalina* produced a weak antibacterial activity of 10 mm, 9.5 mm, 8 mm and 7 mm respectively. The zones of inhibition were significantly different ($P < 0.05$) (Table 2).

For the case of the *S. aureus* ATCC 25923, *F. africana*, *S. didymobotrya*, *T. indica*, *B. aegyptiaca*, *P. punctulata*, *R. natalensis* and *F. sycomorus* gave antibacterial activity of 19 mm, 16 mm, 14.5 mm, 14 mm and 11.5 mm respectively. On the other hand, six plants produced low activity against *S. aureus* ATCC 25923 with a zone of inhibition ranging between 7.5-10mm. For example, *A. coriaria* (10 mm), *C. africana* (9.5 mm), *O. trichocarpum* (8.5 mm), *S. sesban* (8 mm), *Z. gilletii* (8 mm) and *U. dioica* (7.5 mm). All the remaining plant extracts were completely inactive (6 mm). The zones of inhibition were significantly different ($P < 0.05$) (Table 2).

Fuerstia africana is the only plant that showed strong activity with a zone of inhibition of 20 mm against Methicillin-resistant *S. aureus*. The *A. coriaria*, *C. africana* and *S. didymobotrya* Fresen produced moderate activity of 13 mm, 11.5 mm and 11 mm respectively while *V. amygdalina* and *Z. gilletii* gave a low antibacterial activity of 9 mm. All the remaining 11 plant extracts were completely inactive (6 mm) against this test organism. The zones of inhibition were significantly different ($P < 0.05$) (Table 2). In reference to Table 2, all the plant extracts did not show any activity against *Escherichia coli* ATCC 25922 and *Salmonella typhi* ATCC 19430.

Antifungal activity of the plant crude extracts against standard strains of fungus

In vitro testing was performed using the Kirby Bauer disk diffusion method to examine the antifungal capabilities of 18 different plant species against five different fungi. As a result of the observations, it was established that the antifungal activity lay somewhere between 9 and 20.5 millimeters.

Table 3 summarizes how the DCM and methanol extracts of the plants tested stopped the growth of five strains of fungi on average. Some plants, like *P. punctulata* (20.5 mm), *C. africana* (17.5 mm), *S. didymobotrya* (17 mm), *T. indica* (16 mm), *A. coriaria* (16 mm), *F. sycomorus* (15.5 mm) and *R. natalensis* (15.5 mm). On the other hand, *F. africana* had a moderate activity of 13 mm, while *B. angustifolia*, *R. communis*, and *O. trichocarpum* gave low average inhibition zones of 10 mm, 10 mm, and 8.5 mm against the strain. The remaining plant extracts had no detectable action (6 mm) against *M. gypseum*. Significant differences ($P<0.05$) were found in the areas of inhibition.

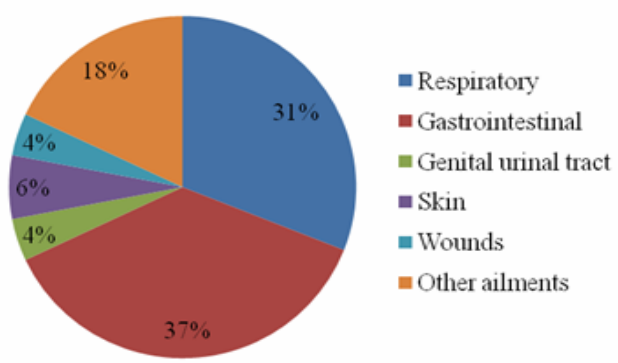

Figure 2. Percentage frequency of diseases treated using herbal drugs in Mwingi North, Kisii South, and Rarieda Districts, Kenya

Table 1. Selected medicinal plants used by the communities from Mwingi North, Kisii South, and Rarieda Districts, Kenya, in treating various bacterial and fungal ailments

Botanical name	Family name and voucher number	Local name	Parts used	Methods	Diseases treated	Area collected
Rhus natalensis Bernh. ex Krauss	Anacardiaceae 2011/IJ018	Sangla (Luo)	Roots	Decoction	Coughs, colds, headache, diarrhea	Lweya (Rarieda)
Vernonia amygdalina Delile	Asteraceae 2011/IJ015	Omosabakwa (Kisii)	Leaves	Concoction	Loss of appetite gastrointestinal problems, diarrhea	Kerina (Kisii South)
Balanites aegyptiaca (L.) Delile	Balantiaceae 2011/IJ05	Othoo (Luo)	Bark	Decoction	Stomach pains, dysentery	Ragengni (Rarieda)
Commiphora africana (Rich.) Engl.	Burseraceae 2011/IJ02	Arupiny (Luo)	Bark	Decoction	Diarrhea fever	Lweya (Rarieda)
Senna didymobotrya (Fresen.) H.S.Irwin & Barneby	Caesalpinaceae 2011/IJ04	Owinu (Luo)	Roots	Decoction	Diarrhea ringworm	Ragengni (Rarieda)
Boscia angustifolia A. Rich	Capparidaceae 2011/IJ017	Mwenzenze (Kamba)	Leaves	Concoction	Chest pains stomachache	Mwingi North
Psiadia punctulata (DC.) Oliv. & Hiern ex Vatke	Compositae 2011/IJ03	Atilili (Luo)	Roots	Decoction	Stomachache, colds, diarrhea	Lweya (Rarieda)
Ricinus communis L.	Euphorbiaceae 2011/IJ01	Odagwa (Luo)	Leaves	Concoction	Stomachache, diarrhea, pains	Lweya (Rarieda)
Tamarindus indica L.	Fabaceae 2011/IJ010	Chwa (Luo)	Bark	Decoction	Diarrhea, cough, fever, tonsils	Ragengni (Rarieda)
Albizia coriaria Welw. ex Oliv.	Fabaceae 2011/IJ08	Ober (Luo)	Bark	Decoction	Cough, diarrhea	Lweya (Rarieda)
Ormocarpum trichocarpum (Taub.) Engl.	Fabaceae 2011/IJ05	Det (Luo)	Leaves	Concoction	Diarrhea, typhoid	Lweya (Rarieda)
Fuerstia africana T.C.E.Fr.	Lamiaceae 2011/IJ011	Ekebunga Baiseke (Kisii)	Leaves	Concoction	Urinary problems, tongue infections, diarrhea, skin infections	Kerina (Kisii South)
Melia volkensii Gürke	Meliaceae 2011/IJ016	Mukau (Kamba)	Leaves	Concoction	Pains in the body	Mwingi North
Ficus sycomorus L.	Moraceae 2011/IJ09	Ngowo (Luo)	Bark	Decoction	Stomach pains, coughs, wounds	Lweya (Rarieda)
Sesbania sesban L.	Papilionaceae 2011/IJ06	Oyieko (Luo)	Roots	Decoction	Diarrheal, diseases, boils	Ragengni (Rarieda)
Zanthoxylum chalybeum Engl.	Rutaceae 2011/IJ013	Mukenea (Kamba)	Leaves	Concoction	Diarrhea, sore throat, coughs, chest pain	Mwingi North
Zanthoxylum gillettii De Wild	Rutaceae 2011/IJ014	Egekoma (Kisii)	Leaves	Infusion	Genitourinary, coughs, mouth ulcers, throat	Kerina (Kisii South)
Urtica dioica L.	Urticaceae 2011/IJ012	Rise (Kisii)	Leaves	Concoction	Blood purification, anemia boils	Iyabe (Kisii South)

Table 2. Zones of inhibition produced by the plant extracts against the selected bacterial strains in mm

Medicinal plants	MRSA	*S. aureus*	*S. typhi*	*E. coli*	*B. subtilis*
Boscia angustifolia	6.0a	6.0a	6.0a	6.0a	13.0b
Fuerstia Africana	20.0d	19.0c	6.0a	6.0a	17.0c
Melia volkensii	6.0a	6.0a	6.0a	6.0a	10.0b
Urtica dioica	6.0a	7.5a	6.0a	6.0a	8.0a
Vernonia amygdalina	9.0b	6.0a	6.0a	6.0a	7.0a
Zanthoxylum chalybeum	6.0a	6.0a	6.0a	6.0a	15.0c
Zanthoxylum gilletti	9.0b	8.0a	6.0a	6.0a	11.0b
Albizia coriaria	13.0c	10.0b	6.0a	6.0a	12.0b
Balanites aegyptiaca	6.0a	14.5c	6.0a	6.0a	12.0b
Commiphora africana	11.5b	9.5a	6.0a	6.0a	12.0b
Ficus sycomorus	8.5b	11.5c	6.0a	6.0a	9.5a
Ormocarpum trichocarpum	6.0a	8.5a	6.0a	6.0a	15.5c
Psiadia punctulata	6.0a	14.0c	6.0a	6.0a	11.0b
Rhus natalensis	6.0a	14.0c	6.0a	6.0a	11.5b
Ricinus communis	6.0a	12.0b	6.0a	6.0a	13.0b
Senna didymobofrya	11.0b	16.0c	6.0a	6.0a	16.0c
Sesbania sesban	6.0a	8.0a	6.0a	6.0a	11.0b
Tamarindus indica	6.0a	16.0c	6.0a	6.0a	15.0c
+ve control	26.0d	24.0d	23.0b	25.5b	26.0d
-ve control	6.0a	6.0a	6.0a	6.0a	6.0a

Table 3. Zones of inhibition produced by plants extracts against fungal strains in mm

Medicinal plants	*C.albicans*	*C. neoformans*	*A. niger*	*T.mentagrophyte*	*M. gypseum*
Boscia angustifolia	6.0a	6.0a	6.0a	6.0a	10.0b
Fuerstia Africana	6.0a	9.0b	8.0a	12.0b	13.0b
Melia volkensii	6.0a	6.0a	6.0a	6.0a	6.0a
Urtica dioica	6.0a	6.0a	6.0a	6.0a	6.0a
Vernonia amygdalina	6.0a	6.0a	6.0a	6.0a	6.0a
Zanthoxylum chalybeum	6.0a	6.0a	6.0a	6.0a	6.0a
Zanthoxylum gilletti	6.0a	6.0a	6.0a	6.0a	6.0a
Albizia coriaria	6.0a	6.0a	6.0a	6.0a	16c
Balanites aegyptiaca	6.0a	6.0a	6.0a	6.0a	6.00a
Commiphora africana	6.0a	6.0a	6.0a	6.0a	17.5c
Ficus sycomorus	6.0a	6.0a	6.0a	6.0a	15.5c
Ormocarpum trichocarpum	6.0a	6.0a	6.0a	6.0a	8.5b
Psiadia punctulata	6.0a	6.0a	6.0a	6.0a	20.5d
Rhus natalensis	6.0a	6.0a	6.0a	6.0a	15.5c
Ricinus communis	6.0a	6.0a	11.5b	6.0a	10.0b
Senna didymobofrya	6.0a	6.0a	6.0a	6.0a	17.0c
Sesbania sesban	6.0a	6.0a	8.0a	6.0a	6.00a
Tamarindus indica	6.0a	6.0a	7.5a	6.0a	16.0c
+ve control	12.5b	13.0c	18,0c	21c	22d
-ve control	6.0a	6.0a	6.0a	6.0a	6.0a

Note: Zones of inhibition in the same column indicated by different letters are significantly different

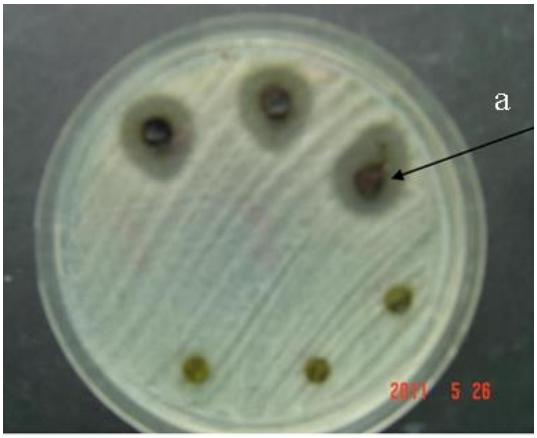

Figure 3. Zones of inhibition of *Commiphora africana* against *Microsporum gypseum.* a. *C. africana*

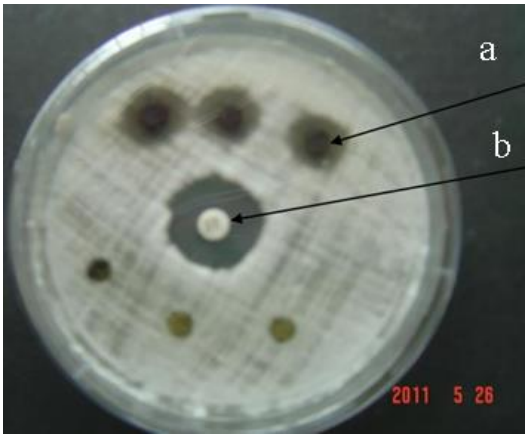

Figure 4. Zones of inhibition of *Rhus natalensis* and that of miconazole control against *Microsporum gypseum.* a. Rhus, b. Miconaz

Table 3 shows that *R. communis* had a moderate effect on *A. niger* (11.5 mm). In contrast, *F. africana, S. sesban*, and *T. indica* had low effects (8 mm, 8 mm, and 7.5 mm, respectively). The other 14 plant extracts, namely *Z. chalybeum, M. volkensii, Z. gilletii, F. africana, U. dioica, V. amygdalina, C. africana, P. punctulata, S. didymobotrya, S. sesban, B. aegyptiaca, A. coriaria, F. sycomorus, R. natalensis* and *T. indica* had no effect on *A. niger* (6 mm). Significant differences ($P < 0.05$) existed between the inhibition zones.

Fuerstia africana showed weak activity against *C. neoformans* ATCC 18310 with a value of 9 mm and moderate activity against *T. mentagrophyte* with 12 mm. All of the other seventeen plant extracts were completely inactive, making a mean zone of 6 mm (Table 3). All eighteen plant extracts were tested against the organism C. albicans ATCC 90028, and none showed any sign of activity. Figures 3 and 4 display the zones of inhibition that plant extracts have produced against several fungus strains.

The Minimum Inhibitory Concentration (MIC) and the Minimum Bactericidal Concentration (MBC)

As shown in Tables 4 and 5, the broth micro dilution method in a 96-well microtitre plate (Figure 5) was used to determine the minimal inhibitory concentration of plant extracts with inhibition diameters of at least 9 mm.

Six plants had MIC and MBC against MRSA. The *A. coriaria* and *C. africana* gave MIC that were equal to MBC of 1.875 mg/mL. The *F. africana* gave MIC and MBC of 0.9375 mg/mL and 1.875 mg/mL, respectively while *V. amygdalina* and *Z. gilleti* gave MIC and MBC of 3.75 mg/mL and 7.5 mg/mL, respectively while *S. didymobotrya* gave MIC and MBC of 1.875 mg/mL and 3.75 mg/mL, respectively. All the tested plants were screened for MIC and MBC against *B. subtilis* except *U. dioica* and *V. amygdalina*, *F. africana* and *S. didymobotrya* gave a low MIC and MBC against *B. subtilis* of 0.9375 mg/mL which is very close to that of the positive control value 0.4688 mg/mL. The *F. sycomorus* and *M. volkensii* had a weak MIC and MBC of 3.75mg/mL and 7.5 mg/mL, respectively. *Z. gilleti, A. coriaria, B. aegyptiaca, P. punctulata, R. natalensis, R. communis, S. didymobotrya, F. africana* had MICs that were equal to MBC.

The *B. aegyptiaca, P. punctulata, R. communis, S. didymobotrya, T. indica* and *F. africana* showed a low MIC and MBC of 0.9375 mg/mL against *S. aureus* ATCC 25923. Although the MIC and MBC for positive control were lower, these are promising plant extracts given that they are crude extract compared to pure compounds of the positive control. The *Z. gilleti* showed a high MIC and MBC of 3.75 mg/mL and 7.5 mg/mL respectively and therefore exhibits the lowest activity.

The best MIC and MBC results came from fungal strains, especially *M. gypseum* (Table 5). The results ranged from 0.9375 mg/mL to 3.75 mg/mL, and 7 plants gave a MIC of 0.9375 mg/mL. The *M. gypseum* was consequently the most sensitive of the tested strains. The *P. punctulata, A. coriaria, C. africana, S. didymobotrya*, and *T. indica* exhibited MIC and MFC values of 0.9375 mg/mL.

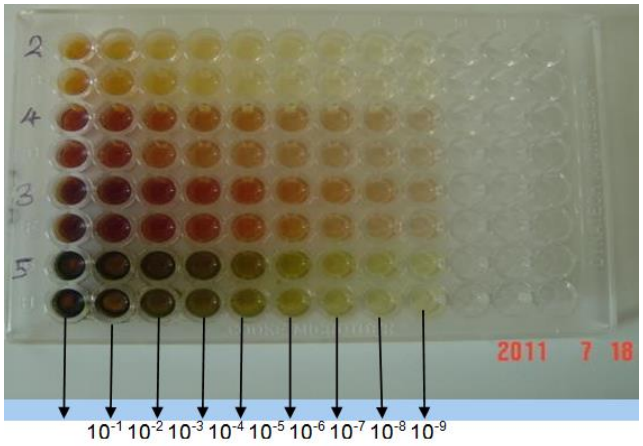

Figure 5. Microtitre plates showing minimum inhibition concentration. The plate shows four extracts at different concentrations beginning from 10^{-1} to 10^{-9} dilution factors

The MIC and MFC values for *F. sycomorus* and *R. natalensis* against *M. gypseum* were different. For both plants, the MIC was 0.9375 mg/mL, and the MFC was 1.875 mg/mL. The activity of *B. angustifolia* and *R. communis* against *M. gypseum* was low, at 3.75 mg/mL for MIC and MFC. It was discovered that only one species of *F. africana*, was effective against both *C. neoformans* and *T. mentagrophyte*. For *C. neoformans*, the MIC and MFC of *F. africana* were 3.75 mg/mL and 7.5 mg/mL. Only *R. communis* was tested for its resistance to *A. niger*. It had a MIC of 3.75 mg/mL and an MFC of 3.75 mg/mL. We did not use any plant extracts for the MIC or MFC screening because every plant tested negative for *C. albicans* ATCC 90028.

Discussion

This study contains information on 18 medicinal plants used to treat, among other conditions, diarrhea, respiratory illness, skin infection, fever, and wounds. The most often mentioned health issue in this study was diarrhea (Figure 2). It could have been caused by drinking polluted water (Jeruto et al. 2008). According to the results of the ethnobotanical study (Table 1), traditional medicine continues to play an important role in treating a variety of disorders for residents of the Mwingi North, Kisii South, and Rarieda Districts.

There are 15 plant families represented by the 18 medicinal plants examined. Concoctions and decoctions were the most commonly utilized preparations for medication methods. A combination of several plants was seen to be used in the preparation of some of the plants. Nanyingi et al. (2008) have backed this up. Because of the synergistic effects of multiple substances in concoctions, the compounds can only be active in combination (Omori et al. 2012).

According to Nanyingi et al. (2008), leaves (9 plants), followed by bark (5 plants), and then roots (4 plants) were the most often used plant parts. The leaves may be chosen since they are the primary photosynthetic organs in plants and produce all of the plant's phytochemical chemicals, which are then transferred to other parts of the plant, such as the bark and roots (Jeruto et al. 2008). As a result of these damaging harvesting procedures, using roots and bark is harmful to the harvested plants' long-term viability (Jeruto et al. 2011).

This investigation suggests significant variation in the antibacterial activity of plant extracts from various species. Some of the medicinal herbs employed by herbalists to treat non-fungal illnesses were also active against certain fungal strains (Tables 2 and 3). Thus, these extracts could be utilized to treat both bacterial and fungal diseases. Examples include *T. indica, S. didymobotrya, R. natalensis, P. punctulata, F. sycomorus, A. coriaria* and *F. africana* showed strong activity against bacterial and fungal strains. In general, among the examined strains of microorganisms, bacteria were more sensitive to several of the chemicals than fungi (Tables 2 and 3). It is consistent with the findings of Korir et al. (2012b), who observed that bacterial strains were more susceptible to most crude extracts than fungal strains.

Table 4. The minimum inhibitory concentration and the minimum bactericidal concentration for bacterial test cultures in mg/mL

Test culture medicinal plants	*MRSA*		*S. aureus*		*B. subtilis*	
	MIC	**MFC**	**MIC**	**MFC**	**MIC**	**MFC**
Boscia angustifolia	ND	ND	ND	ND	1.875	1.875
Fuerstia africana	0.9375	1.875	0.9375	0.9375	0.9375	0.9375
Melia volkensii	ND	ND	ND	ND	3.75	7.5
Urtica dioica	ND	ND	ND	ND	ND	ND
Vernonia amygdalina	3.75	7.5	ND	ND	ND	ND
Zanthoxylum chalybeum	ND	ND	ND	ND	1.875	3.75
Zanthoxylum gilletti	3.75	7.5	ND	ND	3.75	3.75
Albizia coriaria	1.875	1.875	1.875	3.75	1.875	1.875
Balanites aegyptiaca	ND	ND	0.9375	0.9375	1.875	1.875
Commiphora africana	1.875	1.875	3.75	7.5	1.875	3.75
Ficus sycomorus	ND	ND	1.875	3.75	3.75	75.0
Ormocarpum trichocarpum	ND	ND	ND	ND	0.9375	1.875
Psiadia punctulata	ND	ND	0.9375	0.9375	3.75	3.75
Rhus natalensis	ND	ND	1.875	1.875	3.75	3.75
Ricinus communis	ND	ND	1.875	3.75	1.875	1.875
Senna didymobofrya	1.875	3.75	0.9375	0.9375	0.9375	0.9375
Sesbania sesban	ND	ND	ND	ND	3.75	3.75
Tamarindus indica	ND	ND	0.9375	0.9375	0.9375	1.875
Positive control	0.4688	0.4688	0.4688	0.4688	0.4688	0.4688
Negative control	Growth was observed in all the tubes					

Note: MIC= Minimum Inhibitory concentration, MFC= Minimum fungicidal concentration, ND= Not done

Table 5. The minimum inhibitory concentration and the minimum fungicidal concentration for fungal test cultures in mg/mL

Test culture medicinal plants	*C. neoformans*		*T. mentagrophyte*		*M. gypseum*		*A. niger*	
	MIC	**MFC**	**MIC**	**MFC**	**MIC**	**MFC**	**MIC**	**MFC**
Boscia angustifolia	ND	ND	ND	ND	3.75	3.75	ND	ND
Fuerstia Africana	3.75	7.5	3.75	3.75	1.875	3.75	ND	ND
Melia volkensii	ND	ND	ND	ND	ND	ND	ND	ND
Urtica dioica	ND	ND	ND	ND	ND	ND	ND	ND
Vernonia amygdalina	ND	ND	ND	ND	ND	ND	ND	ND
Zanthoxylum chalybeum	ND	ND	ND	ND	ND	ND	ND	ND
Zanthoxylum gilletti	ND	ND	ND	ND	ND	ND	ND	ND
Albizia coriaria	ND	ND	ND	ND	0.9375	0.9375	ND	ND
Balanites aegyptiaca	ND	ND	ND	ND	ND	ND	ND	ND
Commiphora africana	ND	ND	ND	ND	0.9375	0.9375	ND	ND
Ficus sycomorus	ND	ND	ND	ND	0.9375	1.875	ND	ND
Ormocarpum trichocarpum	ND	ND	ND	ND	ND	ND	ND	ND
Psiadia punctulata	ND	ND	ND	ND	0.9375	0.9375	ND	ND
Rhus natalensis	ND	ND	ND	ND	0.9375	1.875	ND	ND
Ricinus communis	ND	ND	ND	ND	3.75	3.75	3.75	3.75
Senna didymobofrya	ND	ND	ND	ND	0.9375	0.9375	ND	ND
Sesbania sesban	ND	ND	ND	ND	ND	ND	ND	ND
Tamarindus indica	ND	ND	ND	ND	0.9375	0.9375	ND	ND
Positive control	0.4688	0.4688	0.4688	0.4688	0.4688	0.4688	0.4688	0.4688
Negative control	Growth was observed in all the tubes							

Note: MIC= Minimum Inhibitory concentration, MFC= Minimum fungicidal concentration, ND= Not done

The antibacterial activity was more pronounced on the Gram-positive bacteria than on the Gram-negative bacteria, for example; *T. indica, S. didymobotrya, R. natalensis, P. punctulata, A. coriaria, B. aegyptiaca* and *F. africana* produced high antibacterial activity against *S. aureus* ATCC 25923 and *B. subtilis* but no plant extract was active on *E. coli* ATCC 25922 and *S. typhi* ATCC 19430 (Table 2). This is in agreement with previous reports by some researchers (Duraipandiyan et al. 2006; Pirbalouti et al. 2010). The probable reason for the variation in the activities between Gram-negative and Gram-positive bacteria could be due to their morphological difference (Maregesi et al. 2008; Pirbalouti et al. 2010). The Gram-negative bacteria have an extra outer membrane in their cell wall which is richer in lipopolysaccharides and acts as a barrier to foreign substances including antibiotic molecules (Maregesi et al. 2008; Pirbalouti et al. 2010). It is also associated with the enzyme found in periplasmic space which can break down the molecules introduced from outside (Pirbalouti et al. 2010).

Bacillus subtilis and *S. aureus* ATCC 25923 were generally more sensitive to the plants extracts (Plates 6 and 8). This may be explained by the cell wall composition of the Gram-positive bacteria (*S. aureus* and *B. subtilis*) which have a relatively thick layer of peptidoglycan sheets of interconnected glycan chains made up of polymer which is fully permeable to many substances and thus sensitive to most plant extracts (Kitonde et al. 2013). This is supported by Samie et al. (2005) who reported that *Bacillus* spp and *S. aureus* were most sensitive whereas *E. coli* and *Salmonella anatum* were more resistant to crude extracts of plants.

The most powerful plant extract was from *F. africana*. The Kisii often uses it to treat diarrhea, mouth infections, urinary tract problems, and skin infections, among other things. It is supported by the fact that the plant extract has a wide range of effects (Table 1). For MRSA, *S. aureus* ATCC 25923, *B. subtilis*, *C. neoformans* ATCC 18310, *T. mentagrophyte*, and *M. gypseum*, the zones of inhibition were 20, 19, 17, 9, 12, and 13 mm, respectively (Table 2 and 3). Based on these results, the extract of *F. africana* could treat bacterial diseases like boils, sores, wounds, and diarrhea caused by *S. aureus*. This result agrees with the findings of another study (Ng'eny et al. 2011) that found *F. africana* to be active against *S. aureus* and MRSA. It also partly agrees with Mariita et al. (2010), who found that the plant had a moderate effect on *S. aureus* but none on *E. coli*, *S. typhi*, and *C. albicans*. Possibly, the difference was caused by harvesting times and solvents utilized during the plant's extraction (Samie et al. 2005; Samie et al. 2010; Assob et al. 2011). In the previous work, Mariita et al. (2010) employed methanol as the solvent for extraction, but DCM and methanol in a ratio of 1:1 were used instead in this investigation.

The phytochemical study revealed the presence of all the examined substances. Therefore, its broad spectrum activity can be linked to its phytoconstituents (Doughari and Manzara 2008). Furthermore, tannins and alkaloids are cytotoxic to bacterial cells, which could explain the broad range of activity of *F. africana* (Omwenga et al. 2009).

Only *F. africana* had low activity (9 mm) against *C. neoformans* ATCC 18310 of the 18 plants examined (Table 3). According to Korir et al. (2012a), *C. neoformans* is resistant to all plant extracts. It may be due to galactoxylomannan and glucuronoxylomannan-based polysaccharide capsules (Susane et al. 2009; Teresa and Alspaugh 2012). The virulence and antimicrobial resistance of *C. neoformans* are attributed to the polysaccharide capsular material (Korir et al. 2012a). For example, the expansion of the capsule has been linked to the protection of the host fungus against host defensive mechanisms such as phagocytosis and oxidative burst (Susane et al. 2009). Additionally, capsular material directly acts against the host. In macrophages, *C. neoformans* releases polysaccharides from its capsule into vesicles surrounding the phagosome; accumulation of these vesicles in the cytoplasm of the cell leads to dysfunction and lysis of macrophages (Hansang and Robin 2009).

Senna didymobotrya was active against *S. aureus* ATCC 25923 and *B. subtilis* at a level of 16 mm, and it was active against MRSA at a level of 11 mm. It also worked very well against *M. gypseum* (17 mm), but it did not affect any tested fungal or bacterial strains. It goes against the previous results, which showed that *S. didymobotrya* was active against *E. coli* and *C. albicans* (Korir et al. 2012b). It could be because the plant species came from different places and were collected at different times (Matu and Staden 2003). Plants picked up during the rainy season will not have the same levels of phytochemicals as those picked up during the dry season (Matu and Staden 2003). The phytochemical screening showed that there were a lot of tannins and terpenoids but not many flavonoids. Tannins from the stem bark of *S. didymobotrya* Fresen are known to kill bacteria and other germs (Chothani and Vaghasiya 2011). Tannins work by stopping the production of secretions and making the mucus in the gut more resistant by making protein tannate (Balogun et al. 2011).

Albizia coriaria exhibited antibacterial activity with a zone of inhibition measuring 13 mm against MRSA, 12 mm against *B. subtilis*, and 10 mm against *S. aureus* ATCC 25923; however, it was inactive against *S. typhi* and *E. coli* (Table 2). The *A. coriaria* also had a high antifungal effect on M. gypseum, with a zone of inhibition of 16 mm, but it did not affect the other fungi tested. It was partly in line with the report by Olila et al. (2007), who found that it worked against *B. subtilis* and *E. coli* but not against *S. aureus*. The *C. africana* is also one of the most active plants, showing antimicrobial activity against four microorganisms with zones of inhibition of 11.5 mm, 9.5 mm, 12 mm, and 17.5 mm against MRSA, *S. aureus* ATCC 25923, *B. subtilis*, and *M. gypseum*, respectively. It was partly in line with what Akor and Anjorin (2009) found when they took root extracts and tested them against *S. aureus*, *E. coli*, and *C. albicans*. The different results might be due to the parts of the used plants, physical factors (temperature, light, and water), contamination by field microbes, different plants, and the location (Okigbo and Mmeke 2008).

Tannins, terpenoids, saponins, cardiac glycosides, and flavonoids were found in *A. coriaria* and *C. africana* in varying concentrations, but alkaloids were absent from *C. africana*, according to the results of a phytochemical analysis. Tannins have antibacterial effects due to their capacity to react with proteins to generate water-soluble compounds that are stable and kill bacteria by breaking their cell membranes. This ability allows tannins to form antibacterial compounds (Mariita et al. 2011).

It has been stated that *F. sycomorus* possesses anti-diarrheal properties (Ahmadu et al. 2007). In this research, the bark extract was active against *S. aureus* ATCC 25923 and *B. subtilis*, with zones of inhibition measuring 11.5 mm, 9.5 mm, and 8.5 mm against MRSA (Table 2). In addition, it displayed 15.5 mm of activity against *M. gypseum*. In prior studies, the crude ethanol extract of *F. sycomorus* demonstrated antibacterial efficacy against *S. aureus*, MRSA, and *S. typhi* (Kubmarawa et al. 2007). It partially corroborates the present findings. However, in the present study, it was ineffective against *S. typhi* ATCC 19430. The *F. sycomorus* had no antifungal activity against the yeast species but was highly active against *M. gypseum*.

It was consistent with the findings of Samie et al. (2010), who also found no antifungal activity against yeast. Differences in geographical location, solvents used in extraction, tested microorganisms, parts used, storage conditions, and analysis techniques may account for the variation (Wagate et al. 2008; Olusesan et al. 2010; Obeidat et al. 2012).

Analysis of the phytochemistry of *F. sycomorus* indicated the presence of secondary metabolites, including tannin, cardiac glycosides, terpenoids, and flavonoids. Possibly, the high content of terpenoids contributed to the antibacterial activity. Since this is corroborated by Chiruvella et al. (2007), who extracted terpenoids from *Soymida febrifuga* (Roxb.) Juss. and reported that the antibacterial activity of terpenoids was caused by terpenes disrupting bacterial cell membranes, other researchers have demonstrated the presence of a portion of these chemicals in previous investigations. For instance, it has been established that terpenes and tannins are present in the leaves (Ahmadu et al. 2007). In addition, it has been shown that stem bark extract contains glycosides, tannins, and flavonoids (Kubmarawa et al. 2007).

Zanthoxylum gilletii was effective against *B. subtilis*, MRSA, and *S. aureus* ATCC 25923 but ineffective against *E. coli* 25922 and *S. typhi* ATCC 19430. In addition, it lacked antifungal efficacy against the studied fungi. Mariita et al. (2010) reported that the plant was inactive against *E. coli, S. typhi, S. aureus*, and *C. albicans*, while Agyare et al. (2006) found that the leaf extract of *Z. gilletii* was active against *E. coli, S. aureus*, *B. subtilis*, and *C. albicans*, but inactive against *A. niger*. The modest discrepancy could be attributable to the solvent employed for extraction and dosing (Okigbo and Mmke 2008) and meteorological and environmental conditions (Kubmarawa et al. 2007; Okigbo and Mmke 2008). Plants harvested at different times and from different areas may have varying antimicrobial properties (Samie et al. 2005). The plant included a high concentration of tannins and flavonoids and a moderate concentration of terpenoids and alkaloids but lacked saponins and cardiac glycosides, as shown by phytochemical analysis. This result is partially consistent with the conclusion reached by Agyare et al. (2006), who discovered that the plant contained alkaloids, tannins, and saponins. Since flavonoids have been shown to have potent antibacterial action, their high content may have been responsible for the observed antimicrobial activity (Olusesan et al. 2010).

Balanites aegyptiaca was effective against *S. aureus* ATCC 25923 and *B. subtilis* but ineffective against MRSA, *S. typhi* ATCC 19430, *E. coli* ATCC 25922, and all tested fungus strains. The flavonoid fractions showed antibacterial activity against *E. coli, B. subtilis*, and *S. aureus*. In contrast, the stem bark of *B. aegyptiaca* revealed a high antifungal activity against *C. albicans* (Maregesi et al. 2008). In contrast, the leaves exhibited high antityphoid activity (Doughari et al. 2007). It was discovered that leaf ethanol extracts were more effective against *E. coli*, *B. subtilis, S. aureus,* and *C. albicans* than stem bark extracts (Gour and Kant 2012). The discrepancy could be due to extraction methods, geographical areas where the plants were taken, the part of plant material used, and the period of plant material collection, all of which affected the number of plant constituents (Ibrahim et al. 2009; Okemo et al. 2011) Moreover, extraction techniques may potentially affect the phytochemical makeup of plants (Korir et al. 2012a).

An examination of the phytochemical compounds of the *B. aegyptiaca* plant revealed the absence of tannins. Still, flavonoids, glycosides, terpenoids, saponins, and traces of alkaloids were found. Flavonoids and terpenoids may have contributed to the antibacterial action (Banson and Adeyemo 2007). The high concentration of flavonoids may have contributed to the antibacterial activity, as flavonoids have been shown to inhibit the enzymes involved in pathogen cell wall formation (Negi et al. 2009). The *B. aegyptiaca* has a long history of traditional use for various illnesses (Chothani and Vaghasiya 2011). Therefore, it may have resulted from secondary metabolites in varying quantities (Nwodo et al. 2010).

Tamarindus indica is a plant with more than one use for most of its parts (Caluwe et al. 2010). In the current study, the stem bark extract of *T. indica* was active against *S. aureus* ATCC 25923, *B. subtilis*, and *M. gypseum*, but not against MRSA, *E. coli* ATCC 25923, *S. typhi* ATCC 19430, or the other fungal strains that were tested. Doughari (2006) found that *T. indica* had antimicrobial action against *E. coli, S. typhi, B. subtilis,* and *S. aureus*. This finding is partially consistent with his findings. Daniyan and Muhammed (2008) found that *T. indica* fruit extract had no effect on *S. typhi* but did affect *E. coli* and *S. aureus*. It is supported by Nwodo et al. (2010), who found that the plant had no effect on *S. typhi* but did have an effect on *E. coli, B. subtilis,* and *S. aureus.* Extraction solvents, collection times, plant ages, the freshness of plant materials, and adulteration may have contributed to these discrepancies in results (Okigbo and Mmeka 2008). It has been demonstrated that various solvents have varying solubility capabilities for certain secondary metabolites (Doughari and Manzara 2008). This study looked at the phytochemical parts like tannins, alkaloids, saponins, cardiac glycosides, and flavonoids. Because of this, the antimicrobial activity can be explained by the phytochemical constituents (Nwodo et al. 2010). Flavonoids have been said to have antimicrobial activity (Olusesan et al. 2010), and their astringent and antimicrobial properties seem to be responsible for their gastric-protective action (Njoroge et al. 2012).

Bacillus subtilis, S aureus ATCC 25923 and *M. gypsum, R. natalensis,* and *P. punctulata* showed considerable activity in this research. The inhibition zone for *R. natalensis* Krauss was found to be 11.5 mm, 14 mm, and 15.5 mm, respectively. In contrast, *P. punctulata* gave 14 mm, 11 mm, and 20.5 mm, respectively (Table 2). The antifungal and antibacterial chemicals extracted from the plants could be employed to treat disorders caused by *M. gypseum, B. subtilis,* and *S. aureus*. It has been said that *R. natalensis* has antiplasmodial activity (Gathirwa et al. 2011). This product can also protect periodontal germs by preventing bacterial enzyme function. It has been said that *P. punctulata* has antileishmanial properties (Githinji

et al. 2009). It was also mildly effective against the fungus that causes coffee berry disease (Midiwo et al. 2002).

The Phytochemical screening indicated that tannins, saponins, cardiac glycosides, terpenoids, and flavonoids were present in *R. natalensis*, but alkaloids were absent. On the other hand, *P. punctulata* possessed tannins, alkaloids, cardiac glycosides, terpenoids and flavonoids but lacked saponins. The known active phytochemical constituents in *P. punctulata* are flavones and phenylpropenoids (Githinji et al. 2010). The antibacterial activity of flavonoids is probably due to their ability to complex with extracellular and soluble proteins and to complex with bacterial cell walls (Banson and Mann, 2008). This could be attributed to the antimicrobial properties of the plant.

The *S. sesban* and *O. trichocarpum* produced zones of inhibition of 11 mm and 15.5 mm, respectively, against *B. subtilis*, indicating moderate activity. Their activity against *S. aureus* ATCC 25923 was mild, at 8 and 8.5 mm. They did not show any action against MRSA, *E. coli* ATCC 25922, *S. typhi* ATCC 19430, the yeast, and dermatophytes, but the leaf extract of *O. trichocarpum* was mildly active against *M. gypseum* (8.5 mm), and the bark extract of *S. sesban* also showed modest activity against *A. niger* of 8 mm. Previous investigations found that the leaves of *S. sesban* had antibacterial activity against *B. cereus, E. coli*, and *A. niger*, which is consistent with the results of this report (Hossain et al. 2007). The *S. sesban* did not affect yeast fungi, but it had a mild activity on filamentous fungi (*A. niger*). This condition may be because the cell walls of the two types of organisms are different (Paiva et al. 2010). The protein in *C. albicans* works as a selective transport system to eliminate wastes and compounds that are destructive to the cell (Nester et al. 2004). An important aspect of medical science is that it permits microorganisms to evade antibiotics and become resistant. The resistance of *C. albicans* ATCC 90028 to crude extracts could be explained in part by this hypothesis.

The phytochemical screening of *O. trichocarpum* showed that it has a lot of terpenoids and flavonoids but no alkaloids or saponins. The *S. sesban* had a lot of tannins, but only a small amount of alkaloids, saponins, cardiac glycosides, and flavonoids. There were no terpenoids. Antimicrobial activity has been attributed to tannins, cardiac glycosides, flavonoids, and saponins (Nwodo et al. 2010). Saponins, for example, disrupt the cell membranes of bacteria, allowing them to enter the body (Omwenga et al. 2009).

Ricinus communis was active against *S. aureus* ATCC 25923, *M. gypsum*, *A. niger*, and *B. subtilis*, with inhibition zones of 12, 13, 11, and 10 millimeters wide, respectively. On the other hand, the plant was inactive against Gram-negative bacteria and yeast when tested. Although Verma et al. (2011) observed that the root extract of *R. communis* had activity against *S. aureus*, *B. subtilis*, and *A. niger*, their findings that it was active against *E. coli* contradict those of *R. communis*. According to Jumbo and Enenebeako's (2007) research, the fermented seed extract of *R. communis* is effective against *S. aureus* and *E. coli*. It, too, is in line with current research findings. Different regions and parts of the plant may be responsible for the discrepancy (Arya et al. 2010).

The phytochemical screening of *R. communis* showed a lot of tannins and flavonoids, a moderate amount of cardiac glycosides and terpenoids, and a small number of saponins but no alkaloids. The antimicrobial activities could be caused by the high concentrations of tannins and flavonoids. The ability of tannins to react with proteins to generate stable, water-soluble compounds have been cited as evidence that they may have antibacterial characteristics (Okemo et al. 2011). Flavonoids can potentially interfere with mitochondrial function and cause metabolic obstructions (Williams et al. 2004). Flavonoids and plant products made from flavonoids have been known for a long time to be antimicrobial defense compounds (Korir et al. 2012b). Tannins, which have antifungal, anti-inflammatory, and healing properties, could be blamed for their antimicrobial properties (Moyo et al. 2010). It could explain why *A. niger* has an inhibiting effect.

Vernonia amygdalina was found to be active against MRSA, not so much against *B. subtilis* in this study. The *S. typhi* ATCC 19430, *S. aureus* ATCC 25923, *E. coli* ATCC 25923, and the tested dermatophytes, yeast, and *A. niger* were all inactive in the presence of this antimicrobial substance. Cheruiyot et al. (2009) and Mariita et al. (2010) also found that this plant could not stop *E. coli, S. typhi*, or *C. albicans* from growing. Furthermore, Uzoigwe and Agwa (2011) found that the leaf and stem extracts of *V. Amygdalina* did not kill *E. coli* and *S. aureus*. It is further confirmed by the findings of Uzoigwe and Agwa (2011), who discovered that *E. coli* and *S. aureus* did not show any signs of susceptibility to the leaf and stem extract of *V. amygdalina*. These findings are in direct conflict with those made by Okigbo and Mmeka (2008) and Ogundare (2011) who found *V. amygdalina* to be active in the treatment of *E. coli, S. aureus*, and *C. albicans*, respectively. In the phytochemical examination, the plant was found to have a high concentration of tannins, a moderate concentration of alkaloids, traces of glycosides, and flavonoids, but no saponins or terpenoids at all. As Ogundare (2011) pointed out, this plant may have antibacterial properties because it contains bioactive compounds.

Bacillus subtilis (13 mm) and *M. gypseum* (10 mm) were killed by the leaf extracts of *B. angustifolia*. It was shown that the plant was very weakly active against *S. aureus* and *B. subtilis* but completely inactive against *S. typhi* and *E. coli* in (Omwenga et al. 2009). This discovery was later refuted by Hassan et al. (2006), who reported that root extracts were effective against *E. coli*. There may have been a disparity since different parts of the plant have varying quantities of active phytochemicals (Gathirwa et al. 2011). While the plant includes a wide range of tannins, alkaloids, and flavonoids in varying concentrations, the phytochemical screening indicated that saponins, cardiac glycosides, and terpenoids were missing from the sample. The development of irreversible compounds between tannins and proteins rich in amino acids inhibits cell protein synthesis (Issazadeh et al. 2012). Alkaloids are well-known for their ability to harm cells in other organisms (Issazadeh et al. 2012).

Both *M. volkensii* and *Z. chalybeum* had a zone of inhibition of 10 mm for *B. subtilis*, they did not affect fungal strains (Tables 2 and 3). It is not in line with Akanga (2008), whose study of *M. volkensii* found that it was active against *E. coli*. This study's findings on *Z. chalybeum* agree with those of Matu and Staden (2003), who found that the stem bark exhibited only moderate antibacterial activity against S. aureus. The antibacterial activity of *Z. chalybeum* against *E. coli* and *S. aureus* has been reported to be zero. Moreover, it demonstrated no antifungal effect against *C. albicans* tests (Olila et al. 2001). One theory is that their antidiarrhoea efficacy is due to a synergistic interaction with components from other plants and certain metabolites (Doughari 2006).

Cardiac glycosides, terpenoids, and saponins were found in moderate abundance in *M. volkensii*'s early phytochemical analysis, but alkaloids and flavonols were absent. The *M. volkensii* has tannins, terpenoids and flavonoids, steroids, alkaloids, cardiac glycosides, and saponins at different levels, according to the phytochemical analysis results of Akanga (2008). For *Z. chalybeum*, the phytochemical analysis discovered low concentrations of alkaloids, cardiac glycosides, and terpenoids but moderate concentrations of tannins and flavonoids with no saponins available. It is possible that the antibacterial elements in *M. volkensii* and *Z. chalybeum* simply slowed the growth of *B. subtilis*. Additionally, the drying process may have altered some of the chemical elements of *M. volkensii* and *Z. chalybeum* (Parekh and Chanda 2007).

Urtica dioica had a zone of inhibition of 7.5 mm on *S. aureus* ATCC 25923 and 8 mm on *B. subtilis*, it had no activity on other fungal species tested. It partially agrees with the findings of Uzun et al. (2004). They discovered that the plant was active against *S. aureus*, inhibited *E. coli* considerably, but had no activity on *C. albicans*. No effect on the fungal strains examined, but *U. dioica* may have a role in transporting active chemicals into the blood, stabilizing body temperature, and reducing harmful effects. For this reason, herbalists administer herbal remedies as a cocktail (Adesina 2005; Samie et al. 2010). Because *U. dioica* had little activity against *B. subtilis* (8 mm) and *S. aureus* (7.5 mm), these microorganisms may have developed tolerance to the herb. The crude extract can be increased in concentration to increase the activity. According to Kitonde et al. (2013), if medications have less activity against the test microorganisms, it suggests that the microorganisms have developed resistance to the drug. Antibacterial properties can be enhanced by raising the concentration of the extract. Diluting crude extracts weakens their antimicrobial activity (Ramamoorthy et al. 2010). The phytochemical screening of this plant revealed the absence of saponins, cardiac glycosides, and terpenoids but found traces of tannins, alkaloids, and flavonoids in the plant.

The low antimicrobial activity of this plant may have been caused by the low concentration of bioactive compounds in it (Nwodo et al. 2010). Other things that could explain the low antimicrobial activity besides the lack of active ingredients are the parts of the plant used, the solvent used for extraction, the location where the plants were collected, and the time when the plants were collected (Hamza et al. 2006). The *U. dioica* extract might not work well because there are not enough diffusible compounds (Ibrahim et al. 2009). Other non-active compounds could have been produced due to disintegration reactions induced by the drying process (Omwenga et al. 2009).

Since herbalists blend multiple herbs, the lack of antibacterial activity does not invalidate the ethnobotanical applications of plants. Due to different phytochemicals in different plants, herbalists employ more than one plant to construct a mixture to treat a certain condition (Omwenga et al. 2009). In herbal medicine, "synergy" means that compounds in plants boost each other's effects (Koroishi et al. 2008). However, one plant was used for each bioassay test in this study. The crude extracts are also less effective because they contain impurities (Ogundare 2011).

The minimal inhibitory concentration is a quantitative experiment that offers further information about the potency of the chemicals present in the extracts (Korir et al. 2012b). No plant extract exhibited activity equivalent to or greater than that of the positive control, although growth was seen in all concentrations of the tubes containing the negative control. The high MIC of 0.9375 mg/mL to 3.75 mg/mL for crude extracts compared to 0.4688 mg/mL for standard pharmaceuticals is a strong indicator that the active ingredient in the crude extract was in low concentration, necessitating the usage of large doses of crude extracts to achieve the desired therapeutic benefits (Korir et al. 2012a). The lower the MIC, the more effective the plant extract is against the tested microorganism (Korir et al. 2012b). On a test against MRSA, both *A. coriaria* and *C. africana* had MICs that were equivalent to MBCs of 1.875 mg/mL. It demonstrates that the concentration of *A. coriaria* that suppresses the growth of MRSA is the same concentration that kills the test organism. The *F. africana* MIC and MBC against MRSA were 0.9375 mg/mL and 1.8775 mg/mL, respectively. While the MIC and MBC of *V. amygdalina* and *Z. gilletii* were 3.75 mg/mL and 7.5 mg/mL, respectively, those of *S. didymobotrya* were 1.875 mg/mL and 3.75 mg/mL (Table 4). Low concentrations of crude extracts of medicinal plants reduce treatment duration, overdose, toxicity, and adverse effects (Kitonde et al. 2013).

Except for *U. dioica* and *V. amygdalina*, all tested plants were screened for MIC and MBC against *B. subtilis*. The *F. africana* and *S. didymobotrya* had MIC and MBC values of 0.9375 mg/mL, which was very close to the positive control value of 0.4688 mg/mL. The MIC and MBC of *F. sycomorus* and *M. volkensii* were low at 3.75 mg/mL and 7.5 mg/mL, respectively. MICs for *Z. gilletii, A. coriaria, B. aegyptiaca, P. punctulata, R. natalensis, R. communis, S. didymobotrya*, and *F. africana* were equivalent to MBC. Certain phytochemicals found in the extracts' chemical screening may be responsible for these antimicrobial activities, possibly in combination, as they have been shown to cause cell membrane disruption, resulting in leakage of cellular components and, finally, the death of microorganisms (Marzouk et al. 2010).

Psidia punctulata, A. coriara, C. africana, S. didymobotrya and *T. indica* produced similar activity on MIC and MFC of 0.9375 mg/mL against *M. gypseum*.

These are very promising plants against *M. gypseum*; therefore, they support the use of these plants for skin conditions.

The best MIC and MFC results were obtained with the fungal strain *M. gypseum*. Seven plants have a MIC of 0.9375 mg/mL, with 0.9375 to 3.75 mg/mL. Consequently, *M. gypseum* was the most sensitive of the examined strains. The *P. punctulata, A. coriara, C. africana, S. didymobotrya* and *T. indica* exhibited comparable activity against *M. gypseum*, with MIC and MFC values of 0.9375 mg/mL. In the fight against *M. gypseum*, these plants have shown great promise, which indicates their potential for use in treating skin problems.

The values for MIC and MFC were different for *F. sycomorus* and *R. natalensis*. For both plants, the MIC was 0.9375 mg/mL, and the MFC was 1.875 mg/mL against *M. gypseum*. The *B. angustifolia* and *R. communis* gave MIC and MFC values of 3.75 mg/mL, which is low. Only the *F. africana* plant extract worked against *C. neoformans* and *T. mentagrophytes*. Its MIC and MFC against *T. mentagrophytes* were both 3.75 mg/mL, and its MIC and MFC against *C. neoformans* 18310 were only 3.75 mg/mL and 7.5 mg/mL, respectively (Table 5). It may result from a high concentration of terpenoids, which have cytotoxic properties against bacteria and fungi (Mamta and Jyoti 2012). The only plant tested against *A. niger* that had a MIC and MFC of 3.75 mg/mL was *R. communis*. Those high values show that the plant extracts have less sensitivity to the microorganisms (Obeidat et al. 2012). Because all plants were negative on *C. albicans* ATCC 90028, no plant extracts were tested for MIC and MFC. It could have been caused by the biofilms that *C. albicans* ATCC 90028 produces when a single cell sticks to a surface and grows into a microcolony. When these microcolonies join, they form a complex 3D structure held by hyphae and an exopolymer matrix (Lafleur 2011).

The highest MIC and MBC/MFC values of the test microbes are an indication that either the plant extracts have low activity on the tested bacterial and fungal strains or that the microorganisms have the potential of developing antibiotic resistance, while the low MIC and MBC/MFC values for the tested microbe is an indication that the plant extracts have the potential to treat any diseases associated with the pathogenic microbes effectively (Doughari 2006; Doughari and Manzara 2008). In this research, all the active plant extracts were bacteriostatic/fungistatic since antibacterial substances are considered bactericidal agents when the ratio of MBC/MIC is ≤ 4 and bacteriostatic agents when the ratio of MBC/MIC is ≥ 4 (Gatsing and Adoga 2007).

The highest MIC and MBC/MFC values of the test microbes indicate either that the plant extracts have limited activity on the tested bacterial and fungal strains or that the microorganisms can develop antibiotic resistance. In contrast, the lowest MIC and MBC/MFC values for the tested microbe indicate that the plant extracts can effectively treat any ailments caused by the pathogenic microbes (Doughari 2006; Doughari and Manzara 2008). All of the active plant extracts in this study were bacteriostatic or fungistatic. Antibacterial substances are called bactericidal agents when the ratio MBC/MIC is ≤ 4 and bacteriostatic agents when the ratio MBC/MIC is ≥ 4 (Gatsing and Adoga 2007).

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