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Chemical and Antibacterial Properties of Lipids Extracted from Some Plant Seeds and Fruits Commonly Used in Cosmetics

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Abstract Over the years, the cosmetic industry has offered a large variety of products that brought out the problems of stability, cost, and scouring couple with the growing effect of bacteria resistance. This research was carried out to determine the chemical and antibacterial properties of lipids extracted from plants commonly used in cosmetics. To achieve this, the physicochemical and phytochemical compositions as well as the antibacterial activity of six oil seeds and fruits: moringa (*Moringa oleifeira*), black seed (*Nigella sativa*), cocoa (*Theobroma cacao*), sesame (*Sesamum indicum*), coconut (*Cocos nucifera*) and avocado (*Persea Americana*) was analysed. The oils were extracted and lipid quality (acid value, saponification value, iodine value, peroxide value, p-anisidine value, and unsaponifiable matter) analysed as well as their phytochemical screening. Antibacterial activity was evaluated by disc diffusion and broth microdilution methods. Results revealed that iodine values of avocado, sesame, moringa, cocoa, coconut, and nigella were 72.89; 74.18; 73.45; 69.54; 70.35, and 61.11(gI_2/100g) respectively. Peroxide values and %FFA ranged between [0.03 to 7.06 meqO_2/Kg] and [8.42 to 20.70%] respectively. The unsaponifiable matter was 0.18; 0.64; 0.21; 0.25; 0.02 and 0.54% for avocado, sesame, moringa, cocoa, coconut and nigella respectively. These values indicate that these oils can be stable during storage. All the seeds and fruits oils extracted contained polyphenols, saponin, alkaloids, and terpenoids reported as classes of metabolites having antioxidant activities. Coconut, sesame, nigella, and moringa oils exhibited high antibacterial activity against selected microorganisms. These results suggest that studied oils may have cosmeceutical and technological applications in cosmetics.

Keywords: *Antibacterial activity, Moringa oleifeira, Nigella sativa, Theobroma cacao, Sesamum indicum, Cocos nucifera, Persea Americana, Polyphenols, Unsaponifiable, Phytochemical*

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1. Introduction

Cosmetics are products that are intended to be applied to the skin to protect it and improve its appearance. Nowadays, the trend of using and seeking natural materials and additives is on the rise; this is most notable for cosmetic products. The reason for the increase in popularity is that the negative effects synthetic materials have on health and the environment have been apparent. Currently, marketing trends are turning towards natural solutions for cosmetics, which have a relation to a healthy lifestyle, and link cosmetic product usage to healthy eating habits [1]. During recent decades, the role of cosmetics degraded on the level of the entirety of society in a way that, many products became a natural part of everyday life [2]. Lipids from plants have been used by humans for centuries as a source of food, medicines, and cosmetic products [3]. Lipids can be obtained from different parts of the plant: seeds, leaves, fruits, or nuts which consist of a large number of active and bioactive substances such as vitamins, sterols, hydrocarbons, triterpene alcohols, carotenoids, and chlorophylls or organic acids including stearic, oleic and palmitic acids which are necessary for the proper functioning of the skin [4]. Lipids and their derivatives form a protective barrier on the skin to keep out external elements and help to keep the skin hydrated [5]. The amount, as well as the presence of ingredients in the lipid, depends mainly on the type of material, its quality, and variety. Some lipids are excellent emollients that make the skin smooth and soft [6]. During diffusion into the skin, lipids have different effects such as: antibacterial, nourishing, smoothing, firming, moisturizing, soothing, or revitalizing [7].

Due to the growing resistance of bacterial and the responsibility of producers to ensure the microbiological stability of cosmetic products, which can be contributed by: the composition of the product, the addition of preservatives, hygiene production, packaging handling, and storage, there is the need for new antibacterial compounds. Some of these seed lipids have not yet been appropriately studied regarding their antibacterial activity [4,8]. In the community, many people favour the use of imported cosmetic oils while nature has gifted us with the amazing plant-producing oils which first of all are duly available, very beneficial with low secondary effects, and very cheap. The microbial contamination of cosmetics may result in the deterioration of its quality: delamination, haze, the appearance of a precipitate, or changes in consistency and colour. To prevent the contamination of cosmetic products huge amounts of preservatives are used during production processes [9,10]. Nowadays the cosmetic industry is under immense consumer pressure to produce safe but innovative natural products. Due to the growing resistance of bacteria, some of these seed oils have not yet been appropriately studied regarding their antibacterial activity and also in search of new skin antibiotics.

Over the years the personal care industry has offered a large variety of multifunctional products consisting of chemical ingredients and synthetic intermediates, and brings out the problems of stability and scouring [9]. Nowadays consumers demand a limitation of chemical synthetic substitutes over cosmetic products containing natural and organic ingredients. Therefore, contaminated cosmetics are a threat to the health of consumers as they may cause dangerous skin infections [10]. For this lucrative industry, new cosmetic ingredients possessing antimicrobial properties are in demand. Plant seeds, nuts, or fruit lipids among all their beneficial features may constitute antimicrobial properties, as it has already been reported in a few kinds of viscous liquids [11,12,13]. So, this study will permit to create awareness, acknowledgment of their properties, and use in this field. Also, Knowledge of these oil sources as well as their properties could add value and encourage their domestication and production of the by-product locally, thereby increasing employment. This work aimed at evaluating the physicochemical, phytochemical, and antimicrobial properties of seed lipids used for cosmetic purposes.

2. Materials and Methods

2.1. Samples Collection

Avocado (*Persea Americana*), coconut (*Cocos nucifera*) fruits, sesame (*Sesamum indicum*), Black seed (*Nigella sativa*), cocoa (*Theobroma cacao*), moringa (*Moringa oleifeira*) seeds were purchased in Buea and transported to the life science laboratory of the University of Buea.

2.2. Sample Preparation and Extraction

Moringa and cocoa seeds were dehulled and dried in an oven at 50ºC for 48 hours. Each of the seeds was crushed in a blender. Avocado was washed, peeled, seed removed, sliced into small pieces, and dried in an oven for 24 hours at 50°C. Coconut was also dehulled and blended using a grater. The extraction was carried out by maceration. The seeds were oven-dried at 50ºC for 48 hours and ground. Two hundred grams of the crushed samples were weighed into different containers respectively, 500 ml of hexane added to it and allowed to stand at room temperature while regularly shaking for 48 hours. The mixtures were filtered and the solvent was removed by rotary evaporation.

2.3. Oil Yield

To determine the oil yield, the method described by Akbar et al., [14] with slight modification was used. The seed and fruits were dried, grounded using a mechanical method, and defatted in a soxhlet apparatus using hexane as solvent. The process continued for 8h. The solvent was removed by vacuum evaporation and exposure to heat in a drying oven at 50°C. The amount of oil recovered was calculated as the percentage of total oil present in the seeds and fruits.

$$\%\ \text{Oil yield} = \frac{(weight\ 1 - weight\ 2)}{weight\ 1} X 100$$

Where,
Weight 1: Weight of seed taken
Weight 2: Weight of extract residue after solvent removal.

2.4. Assessment of Oils Quality Indices

2.4.1. Acid Value (Percentage Free Fatty Acid)

The oil samples were filtered through filter paper to remove impurities. One gram (1g) of each lipid samples was weighed into a 250ml Erlenmeyer flask and 12mL of ethanol was added. Five drops of phenolphthalein indicator were added and the mixture was shaken to dissolve the sample completely. The mixture was then titrated with 0.01N KOH, shaking vigorously until the endpoint was reached. The endpoint was indicated by a slight pink color that persisted for 30seconds [15]. The acid value in terms of free fatty acid was calculated using the following formula:
Where:

$$ACID\ VALUE = \frac{56.1 * C * (Vf - Vi)}{m}$$

$$\%\,\text{FFA} = \frac{2.82 * AV}{56.1} * 100g$$

Vi = consumption of 0.1 mol/l KOH in the main test
Vf = consumption of 0.1 mol/l KOH in the blank test
C= molarity of the KOH
m = oil mass (g)
% FFA = Percent free fatty acid (g/100 g).

2.4.2. Peroxide Value (PV)

Each of the oils was measured (0.4-0.5grams) and 10ml of chloroform, 15ml of acetic acid, and saturated 1ml of potassium iodide were added to it. Each of these solutions was mixed and incubated in the dark for 10 minutes. After

incubation 20ml of distilled water and 0.3ml of starch 1 % was added to the solution and mixed. The solutions were then titrated with thiosulphate 0.01N till transparent color [14] and the peroxide value was calculated as follows.

$$PV = \frac{\text{Cthio}(Vf - Vi)}{m} * 1000$$

Where:
Vf = consumption of 0.01M sodium thiosulfate solution in the main test
Vi = consumption of 0.01M thiosulfate solution in the blank test
C = molarity of the sodium thiosulfate solution
m = weighed portion of substance in grams

2.4.3. P-anisidine Value, AV

Anisidine value was determined by the standard method of the American Oil Chemists' Society Cd 18-90 (p-anisidine value) using a Perkin Elmer UV-Visible Spectrophotometer. Following the method, 0.5-4 g of oil sample was weighed into a 25 mL volumetric flask. The sample was dissolved and diluted to 25 mL of volume with isooctane. Then the absorbance of the solution (Ab) was measured in a cuvette at 350 nm with the spectrophotometer, using the reference cuvette filled with isooctane solvent as a blank. After the measurement of absorbance, 5 ml of this solution and 5 ml of isooctane were transferred into 2 different tests tubes and 1 ml of 0.25% acetic acid solution of p-anisidine was added and the mixtures were shaken on a vortex. After exactly 10 minutes of incubation at room temperature, the absorbance (As) of the solution containing the sample was read at 350 nm, using the second solution as blank. P-anisidine value was calculated as followed:

$$P - An = \frac{25(1.2As - Ab)}{m}$$

Where, As is the absorbance of test solution after reaction with the p-anisidine reagent; Ab is the absorbance of the fat solution; m is the sample weight.

2.4.4. Total Oxidation (TOTOX) Value

A scientist, Holm, suggested a combined expression of peroxides and secondary oxidation products, and therefore developed the concept TOTOX value. Together this established the TOTOX value = 2PV + AV, giving a value of the total oxidation status in oil (Holm, 1972). TOTOX values of oils samples were determined based on the obtained peroxide and P-Anisidine values using the formula; TOTOX = 2PV + AV

2.4.5. Iodine Value, IV

0.1-0.2g of oil sample was weighed in a beaker and 3.8ml of carbon tetrachloride (CCl4) was added. The mixture was shaken for homogeneity and 6ml of Wijs reagent was added and the mixture was stored in the dark for 15 minutes. This was closely followed by the addition of 5ml of 10 percent (10%) potassium iodide solution and 5 drops of starch added until it turns dark. The solution was carefully titrated with a 0.1N sodium thiosulphate till the equivalence point (dark to colourless). Similarly, the procedure was repeated for the flask labeled 'Blank' [15]. The iodine number was calculated using the equation below.

$$\text{Iv} = \frac{12.69(B - S)}{w} XN$$

Where,
B = volume in ml of standard sodium thiosulphate solution required for the blank.
S = volume in ml of standard sodium thiosulphate solution required for the sample.
N = normality of the standard sodium thiosulphate solution.
W = weight in g of the oil sample.

2.4.6. Saponification Value, SV

About 0.5-0.6g of oil was transferred into a test tube and 10mL of 0.5N alcoholic KOH is added into it and mixed well. The flask was fitted with a reflux condenser and refluxed for about one hour (till the reaction is complete and the liquid becomes clear). A blank experiment was simultaneously conducted in the same way without oil. Both the flasks were cooled and washed down the inner side of the reflux condenser into the respective beakers with a minimum quantity of water. 2 drops of phenolphthalein were added and titrated against a standard solution of 0.5N hydrochloric acid until the pink colour disappeared [15]. The saponification value was calculated using the equation below;

$$\text{Sv} = \frac{56.1(B - S)}{M}[HCL]$$

Sv = Saponification value; B= Volume of blank;
S= Volume of sample; [HCL] = concentration of HCL
m= Mass of oil sample

2.5. Unsaponifiable Matter

Five grams (M1) of oil was weighed in a round bottom flask, 50ml of alcoholic potassium hydroxide (2N) was added to it and heated at reflux for an hour. From the top of the round bottom flask 50ml of distilled water was added and the solution mixed. The hydroalcoholic soapy solution was then transferred in a separating funnel and the round bottom flask was rinsed with diethyl ether and added to the separating funnel. The solution in the funnel was mixed vigorously and left to rest for 1 minute. The alcoholic soapy phase was extracted into a second separating funnel. The extraction of the alcoholic soapy phase was repeated and at every time with 50ml of diethyl ether. The ether phase was transferred quantitatively in a round empty bottom flask and weighed. Finally, the ether phase was evaporated till dry using a rotary evaporator at 103°C and the round bottom flask weighed (M2) [16]. The unsaponifiable matter expressed in % was calculated as follow;

$$\%\ \text{Unsaponifiable} = \frac{(M1 - M2)}{M1} X100$$

M1 = mass of oil sample in grams.
M2= mass of dry residue in grams.

2.6. Phytochemical Screening of the Seed Extracts

The presence of major antioxidant secondary metabolite classes, namely; steroids, saponins, alkaloids, cardiac glycosides, phenolics, and triterpenoids were determined using standard phytochemical methods.

2.6.1. Test for Saponins (Frothing Test)

Saponins were tested according to the method described by Banso and Adeyemo [17]. This was done by mixing 0.5mL of the oil in a test tube containing 3mL of hot distilled water. The mixture was shaken vigorously for 1 minute to observe for persistent foaming.

2.6.2. Test for Steroids (Lieberman-Burchard Test)

The method according to Joshi et al. [18] was used. One (1mL) of each oil sample was dissolved in 2mL of chloroform. Three drops of acetic anhydride were added to the test tube and boiled in a water bath for 10 minutes. It was rapidly cooled in running tap water. Two (2mL) of Concentrated H2SO4 was added alongside. It was allowed to stand for 5 minutes for the development of a greenish colouration.

2.6.3. Test for Alkaloids (Dragendorf's Test)

This was done according to the method of Joshi et al. [18]. One (1mL) of oil was stirred with 0.4mL of 1% HCl in a water bath for 5 minutes and filtered. Two (2g) of Potassium iodide and 1.27g of iodine were dissolved in 5mL of distilled water and the solution was diluted to 100 ml with distilled water. Two drops of this iodine solution were added to the filtrate; a brown coloured precipitate indicated the presence of alkaloids.

2.6.4. Test for Phenols (Wagner's Test)

To 1 ml of the oil, one drop of 5% FeCl3 (w/v) was added. The formation of greenish precipitate indicated the presence of phenolics [19].

2.6.5. Test for Terpenoids (Salkowski Test)

The method described by Ayoola et al. [20] was used to test for terpenoids. To 0.2 mL of chloroform, 0.5 mL of oil was added. Concentrated H_2SO_4 (0.3mL) was carefully added to form a layer. A reddish-brown colouration of the interface formed indicated the presence of terpenoids.

2.6.6. Determination of Total Phenolic Content (TPC)

This was determined using the Folin-Ciocalteu colorimetric method as described by Gao et al. [21]. Twenty microliters (20 µL) of the oil were mixed in a test tube with 0.2 mL of Folin-Ciocalteu reagent and 2 mL of distilled water and incubated at room temperature for 3 min. Following this, 1 ml of 20% sodium carbonate solution was added to the mixture and re-incubated for 2 hours at room temperature. The absorbance of the resulting blue color was measured using a quartz cuvette at 765 nm. Gallic acid was used as standard and total phenolic contents were expressed as gram gallic acid equivalents per 100 g of extracts.

2.7. Antibacterial Activity

The three clinical bacterial species (*Staphylococcus aureus*, *Escherichia coli*, and *Salmonella typhi*) used for the antimicrobial assay were obtained from the Buea Regional Hospital. Microscopic, biochemical (catalase, indole, oxidase, and coagulase test), and physiological tests were done on these cells by a laboratory technician of the said hospital and a phenotypical confirmatory test was also done by 2 Ph.D. students of the University of Buea Department of Microbiology. Pure colonies were obtained and were used immediately for the antimicrobial assay.

Determination of the antimicrobial activity of the lipid extracts was performed under sterile conditions. Antibacterial activity was performed using the disc diffusion method [22]. Two concentrations (32 and 64 mg/ml) of each oil extract were prepared in 5% tween 20 and vortex to mix. Bacterial suspension of each strain was prepared and adjusted to MacFarland standard of 0.5 (1.0x108 CFU). The bacteria suspension was spread on a Mueller Hinton agar plate and kept for 3-5 minutes to dry.

Six Whatman paper number 1 were punched and gently fixed at labeled positions on the Mueller Hinton agar surface. Then 10 µL of oil test solution (32 and 64 mg/ml of extract) was transferred onto each disc. Positive (ampicillin and trimetropine) and negative (10 µL of 5% tween 20) controls were included; the plates were kept for 30 min at room temperature and then incubated under incubation conditions. Diameter zones of inhibition were measured after 24 hours of incubation.

The minimum inhibitory concentration (MIC) was determined following the microdilution method as described by Mouekou et al. [23] with some modifications. A stock solution of each oil was prepared in 5% tween 20 by adding 32 mg to 1ml of 5% tween 20. 100 µL of Mueller Hinton broth was transferred into each of the 96 wells of a microtiter plate. Then, 100 ul of the extracts was added to well 1 of row A. a serial dilution was carried out on each extract giving a final concentration of test extracts 3.2, 1.6, 0.8, 0.4, 0.2, 0.1, and 0.05 mg/ml. One hundred µL of bacterial suspension (6 × 105 CFU/mL final d•ensity) was added to each well

3. Results and Discussions

3.1. Oil Yield and Physicochemical Properties

3.1.1. Percentage Oil Yield of Seeds and Fruits

The percentage oil yield for Moringa seed, nigella seed, sesame seed, cocoa, avocado, and coconut are represented in Table 1. Coconut had the highest oil yield followed by moringa and cocoa while nigella had the lowest oil yield. The oil yield of the oil ranging from 15.91 to 54.24 was comparable to that obtained from other studies like those of Ogbunugafor et al. [24], Saeed et al. [25], and Warra et al. [26] on coconut, moringa, and sesame oils.

3.1.2. Physicochemical Properties

The physicochemical properties of *Cocos nucifera* (coconut), *Persea americana* (avocado), *Sesamum indicum*

(sesame), *Moringa oleifera* (moringa), *Nigella sativa* (black seed), and cocoa lipids are presented in Table 1.

Table 1. Percentage oil yield and physicochemical properties of oils

Parameters	Avocado	Sesame	Moringa	Cocoa	Coconut	Nigella
%FFA	0.09 ± 0.02^{a}	0.12 ± 0.00^{a}	0.15 ± 0.09^{a}	0.03 ± 0.00^{a}	0.05 ± 0.00^{a}	7.06 ± 0.73^{b}
IV (gI_2/100g)	72.89 ± 0.21^{a}	74.18 ± 1.71^{a}	73.45 ± 0.22^{a}	69.54 ± 0.00^{a}	63.35 ± 2.47^{a}	75.17 ± 1.90^{b}
PV ($meqO_2$/Kg)	10.75 ± 3.11^{a}	13.00 ± 0.39^{ab}	8.42 ± 0.66^{a}	10.74 ± 3.76^{a}	12.13 ± 0.87^{abc}	20.70 ± 0.51^{bc}
SV (mgKOH/g)	153.96 ± 15.7^{a}	128.79 ± 9.9^{a}	125.61 ± 3.1^{b}	185.35 ± 0^{a}	173.80 ± 17.5^{a}	180.52 ± 47.7^{a}
P-An. Value	5.76 ± 0.13^{a}	4.36 ± 0.44^{a}	5.87 ± 2.31^{a}	2.43 ± 1.42^{a}	2.88 ± 0.96^{a}	8.52 ± 1.36^{b}
TOTOX	27.27 ± 6.09^{abc}	30.36 ± 0.34^{abc}	22.71 ± 5.69^{a}	23.84 ± 0.40^{ab}	27.15 ± 2.70^{abc}	45.95 ± 5.69^{abc}
USM (%)	0.18	0.64	0.21	0.25	0.02	0.54
Oil yield	32.21	36.01	45.60	45.23	54.24	15.91

Mean ± standard deviation carrying different superscripts letters in a row indicates significant difference at P <0.05. IV; Iodine Value, P-An; P anisidine value, SV; Saponification value, PV; Peroxide value, USM; Unsaponifiable matter.

Results obtained for acid value in terms of percentage free fatty acids were compared among the extracted lipids and the values obtained from this work indicate that the FFA of the lipids has a determined range from 0.03 % for cocoa oil to 7.06 % FFA for nigella oil, which was significantly higher compared to the other oils (P<0.05). Low percentage free fatty acids were recorded for cacao and avocado oil. In addition, all the values obtained fall within the range specified by Codex Alimentarius (≤2.5%) [27]. The acid value represents free fatty acid content due to enzymatic activity and is usually indicative of spoilage as rancidity. Its maximum acceptable level is 4 mg KOH/g oil or 2.5 % FFA [27], for recommended international standards for edible oil. In nearly all the cases there are corresponding low levels of free fatty acids in the lipids, which also suggests low levels of hydrolytic and lipolytic activities in the lipids. This could be used to check the level of oxidative deterioration of the oil by enzymatic or chemical oxidation. But on the contrary, the value is high for the nigella oil under study. This acid value can be made fit by subjecting the oil to refining and this may also improve its quality for industrial purposes [28]. The significantly low percentage free fatty acids (P<0.05) of the cocoa, avocado, coconut, sesame, and moringa oils were probably a result of lower hydrolysis of triglycerides thus signified that the oil could have a long shelf life, which allows it to be used in cosmetic formulations.

The result obtained for the Iodine value for the different lipids ranges from 61.17 to 74.18 gI2/100g sample. There was no significant difference between the iodine values of avocado, sesame, moringa, cocoa, and coconut lipids however, there was a significant difference with the other oil samples compared to nigella oil. High iodine observed for nigella, sesame and moringa indicate the increase in the average degree of unsaturation of the lipids, as such, the amount of iodine that can be absorbed by fatty acids would be higher. Lipids are classified as drying and non-drying according to Codex Alimentarius standard based on iodine value [27]. Lipids with iodine values lower than $100gI_2$/100g oil are referred to as non-drying oil. The iodine value obtained from these samples places their lipids in the non-drying groups. These lipids may find application as a raw material in industries for the manufacture of vegetable oil-based foods and cosmetic products [28]. Certainly, those lipids whose values are greater than 100g I_2/100 g oil could be used extensively as lubricants and hydraulic brake fluids. The iodine values obtained here are comparable to the values of castor oils and olive oils, both of which are non-drying oils [29].

The oxidative susceptibility of the extracted lipids was assessed by the determination of peroxide value. These values range from 8.42 to 20.70 meqO2/kg oil. The peroxide value of the moringa seed oil was 8.42 meqO2/kg of oil compared to other samples which are less than 15meqO2/kg of oil, allowed for crude oils by Codex Alimentarius Committee [30]. Nigella seed oil compared to other lipid extracts exhibit a significantly higher (P<0.05) peroxide value. Low values (<15meqO2/kg oil) were obtained especially in moringa, cacao, and avocado. The low values of PV are indicative of low levels of oxidative rancidity of the lipids and also suggest a strong presence or high levels of antioxidants. Certain antioxidants may however be used to reduce rancidity, such as propyl gallate and polyphenol. The peroxide values are low and are pointers to the fact that the lipids may not be easily susceptible to deterioration. Izuagie et al. [31] observed a PV of 1.72 ± 0.01 and 1.42 ± 0.01 mEq/kg for *C. citrullus* and *C. edulis* oil respectively.

Lipids with saponification value within the standard value ≥ 180mgKOH/g according to AOAC [32] are considered as lipids good for soap making. There were significant differences observed in the saponification value of the entire lipid extracts (p<0.05). Saponification values had been reported to be inversely related to the average molecular weight of the fatty acids in the oil fractions. Moringa, sesame, avocado, and coconut lipid extracts have saponification values below the standard value (180mgKOH/g). This implies that these lipids cannot be used in liquid soap making due to the high molecular weight of their fatty acids but can be used in solid soap. Saponification values of oil reported by Saeed et al. gotten from moringa seed, sesame seed, biter kola, melon, and watermelon seed were; 182.89mgKOH/g, 192.70mgKOH/g, 229.545mgKOH/g, 247.95mgKOH/g, and 192.09mgKOH/g respectively [25]. These values of moringa and sesame are not in accordance with the values obtained in this study. The differences observed might be a result of the differences in the method of extraction [14]. Thus, the lipid extracts of nigella and cocoa with SV in the range reported above may be used for soap making, shampoos, and lather shaving creams [28].

P-Anisidine value (p-AV) of the seed oils extracted ranged from 2.43 to 7.55 anisidine units with the highest values obtained with nigella and the lowest with cocoa. The p-AV values suggest the presence of significant amounts of secondary oxidation products in the test oil samples. The significantly high value ($P<0.05$) of nigella oil indicates that considerable oxidative activity might be due to either lipoxygenase or autoxidation [33]. These values are comparable between all the samples.

The primary and secondary oxidation (TOTOX) products of seed oil extracted, were higher with cocoa followed by sesame, coconut, and avocado. Nigella and moringa seed oil show significantly lower oxidation ($P<0.05$). The high TOTOX value obtained with nigella oil is a result of its high p-Anisidine value and high peroxide value can be due to the method of extraction or the low content in natural antioxidants as reported by Zia-Ul-Haq et al. [33]. The low level of unsaturated fatty acid provides the oil with high oxidative stability [33].

The unsaponifiable matter of avocado, moringa, nigella, sesame, coconut, and cacao was found to be in the range of 0.02-0.64% (Table 1). The results suggest that sesame contains the highest amount of unsaponifiable matter followed by nigella, cacao, moringa, avocado, and coconut extract. The high unsaponifiable matter of sesame and nigella may suggest that the oil contains natural antioxidants such as sterols, tocopherols, phenolics, lycopene, and carotene which are considered as the unsaponifiable matter of oil [34]. Hence oil with more abundant amounts of unsaponifiable matter would have higher quantities of these natural antioxidants and higher applications in the cosmeceutical industry.

3.2. Phytochemical Screening of Extracted Lipids

The present study shows the presence of phytochemical constituents like alkaloids, glycosides, saponins, steroids, and terpenoids in different intensities for the six seed oils as shown in Table 2. The qualitative analysis revealed the presence of at least three classes of secondary metabolites in each seed and fruit oil (Table 2). In all oil extracts, except that of *Persea Americana*, phenols were detected.

The amount of total phenols content varied widely in the lipid extracts and ranged from 2.51 to 6.92 g of gallic acid equivalents (GAE) /100 g of extract (Table 2) nigella and coconut with respective values of 5.61 and 6.92g/100 g were found to contain the highest levels while a low level (2.51 g/100 g) was found in avocado oil. The presence of phenols in almost all the lipids could be explained by the fact that phenols are mostly distributed in plants due to their important function in defense mechanisms [35]. Some of these results are following those obtained by Voukeng [36]; Fankam et al. [37]; NGuessan et al. [38] and Lacmata et al., [39] after a chemical screening of the same plants. The differences in the composition of certain seeds observed in this work compared to that of other authors might be attributed to the environmental variations, age of the plant, extraction process, and nature of the extraction solvent [40]. Though the detection of such metabolites does not automatically predict the antioxidant activity of a seed extract, it has clearly been demonstrated that several compounds belonging to the investigated classes of metabolites showed antioxidant activities [41]. From a comparative viewpoint, the TPC found with the oil of coconut was higher than that reported by Nurul et al., [42], with a respective value of 1.56. Oil extracts of moringa and coconut had TPC less than those reported in the literature by Ajavi et al. [43] and Marja et al. [44] with respective values of 5.48 and 72.1g GAE/100g dried weight. Significant differences between the results were likely due to genotypic and environmental differences (namely climate, location, temperature, fertility, diseases, and pest exposure) within species, choice of parts tested, time of taking samples, and determination methods.

3.3. Antibacterial Properties of the Lipid Extracts

The antibacterial activities of the six lipid extracts were tested against three bacteria namely *Escherichia Coli*, *Salmonella typhi*, and *Staphylococcus aureus*. Paper discs containing oil (Figure 1) and the activity are shown in terms of diameters of inhibition and a plot of MIC and MBC are shown in Table 3 and Figure 2 respectively.

Diameter of zones of inhibition of six lipid extracts against three bacterial strains

The impact of the selected oils on the growth of *Staphylococcus aureus*, *Escherichia coli*, and *Salmonella Typhi* is presented in Table 3. The lipid extracts, sesame, nigella, and coconut lipid extracts, demonstrated the highest antibacterial properties at 64 mg/ml against Salmonella Typhi but less than that of the standard antibiotic Trimetoprine. Trimetoprine is specific to *Escherichia coli* and *Salmonella Typhi* and ampicillin was specific to *Staphylococcus aureus*. Cocoa and avocado showed no antibacterial activity on the tested organisms. Moringa, sesame, and coconut oil decreased the growth of S. aureus, to a significant level; this same decrease was observed on *Escherichia coli* by the nigella, moringa, and sesame oils. *Salmonella typhi* activity was also significantly decreased by nigella, sesame, and coconut oils.

Table 2. Phytochemical Screening oil extracts

	Avocado	Sesame	Moringa	Cocoa	Coconut	Nigella
Saponin	+	±	±	+	+	+
Steroids	-	+	-	-	-	+
Alkaloids	+	+	+	±	+	+
Phenols	_	+	+	+	+	+
Terpenoids	+	+	±	+	+	+
TPC(GAE) /100g)	2.51	4.27	3.49	2.74	5.61	6.92

(-): Absent, (+): Present, (±): Mild.

Figure 1. Plates of crude extracts before(A) and after (B) incubation *Staphylococcus aureus*

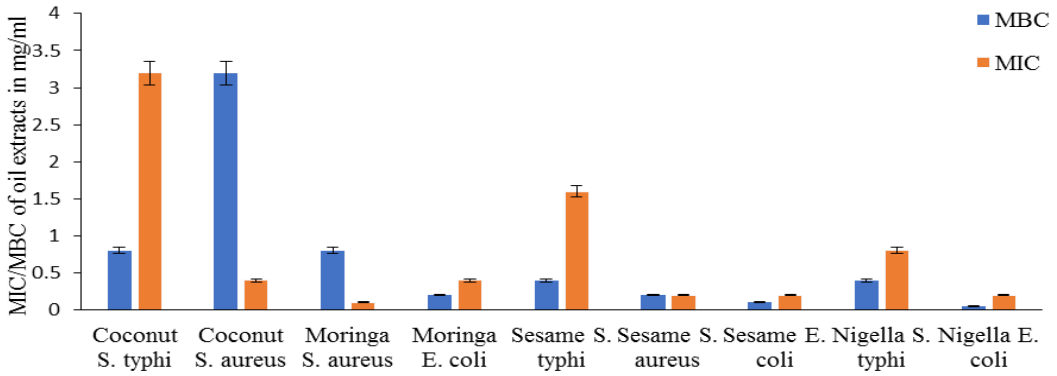

Figure 2. Minimum Inhibitory Concentration (MIC) and Minimum Bactericidal Concentration (MBC)

Table 3. Inhibition zones of the lipids against the selected pathogenic bacterial strains

Bacteria species	Extract concentration (mg/ml)	Diameter of inhibition in mm							
		Nigella	Sesame	Moringa	Avocado	coconut	Cocoa	Trimetroprine	Amplicillin
S.aureus	64	-	20	10	-	16	-	23	25
	32	-	15	8	-	14	-	23	25
S. typhi	64	12	15	-	-	13	-	20	-
	32	7	13	-	-	9	-	20	-
E. coli	64	15	10	9	-	-	-	20	6
	32	15	-	7	-	-	-	20	6

Zones of antagonistic activity towards the growth of *Staphylococcus aureus* were also observed around the paper discs containing the oil of the sesame and coconut (Figure 1). Cocoa, avocado, and coconut oils failed to inhibit *Escherichia coli* like avocado, cocoa, and moringa against *Salmonella typhi*. Nigella oil inhibited efficiently the bacteria *E. coli*.

They conducted laboratory trials indicating differentiated antimicrobial activity of the tested lipids towards indicator microorganisms. The impact of the selected oils on the growth of *Staphylococcus aureus*, *Escherichia coli*, and *Salmonella Typhi* is presented in Table 3. This result is in the same line with those of Katarzyna et al. on the growth of these microorganisms who reported that oil of the nigella seed and tamanu seed oil inhibited *S. aureus* [4]. Comparing the results of the experiments nigella, moringa, coconut, and sesame extracted lipids showed the highest antibacterial activity towards the growth of the selected microorganisms. The antagonistic properties of the selected lipid extracts depended on their type as well as the tested microorganisms. Although several groups of researchers conducted experiments to determine the antibacterial and antifungal properties of different lipids [11,12] there are not many studies concerning the antimicrobial influence of a huge range of seed and fruit lipids dedicated for cosmetic use. The antibacterial activity of nigella seed oil has also been reported by Nair et al. [11]. In our research, this seed oil presented antibacterial activity towards Gram-positive bacteria *S. aureus*. In the research conducted by Hassanein et al. [45], six different cold-pressed black cumin seed oils were tested. These seed oils were supplemented to cheese for an inhibitory effect towards food spoilage bacteria during cold storage. Authors have stated that *in vitro* and *in situ*

supplementation with black cumin oils showed an antibacterial impact on the growth of the *S. enteritidis*, *S. aureus*, *E. coli*, and *L. monocytogenes*. Oyi et al. [46] confirmed that formulated coconut oil creams exhibited both antibacterial and antifungal properties. Among numerous properties of the usage of coconut oil, DebMandal et al. [47] list antibacterial, antifungal, antiviral, and antiparasitic activity. Authors have indicated that coconut oil, received from nuts (concentrations 5-40% of oil) exhibited antagonistic properties against *P. aeruginosa*, *P. vulgaris*, *E. coli*, and *B. subtilis*. However, the result of our studies did not reveal any antagonistic activity of coconut oil. Bacteria including *S. aureus* and *E. coli* are responsible for food intoxications. In addition to suffering and death, these bacteria cause considerable economic losses [48]. *Staphylococcus aureus* and *Salmonella typhi* are two microorganisms responsible for food intoxication and typhoid fever respectively [49]. They were sensitive to oils, thereby indicating that these oils are potent antibacterial agents which could serve either as cosmetic or dietary supplements to fight against infections caused by these bacteria. The antibacterial properties can be justified by the presence of different classes of phytochemicals and fatty acids including PUFA including, linolenic acid, arachidonic acid, palmitoleic acid, and oleic acid as the state in the literature [23].

While MIC is the lowest concentration of an antibacterial agent necessary to inhibit visible growth, minimum bactericidal concentration (MBC) is the minimum concentration of an antibacterial agent that results in bacterial death. The closer the MIC is to the MBC, the more bactericidal the compound. A lower MIC value indicates that less of the oil is required to inhibit the growth of the organism; drugs with lower MIC scores are more effective antimicrobial agents. The MIC and MBC values of the different oils are presented in Figure 2. Oils from nigella and sesame showed MIC lesser than that of coconut against *Salmonella typhi* and *E. coli*; reflecting their activities against bacteria. The oil composition of these two seed species was found to contain significant amounts of PUFA including linolenic acid, arachidonic acid, palmitoleic acid, and oleic acid which could explain their antimicrobial activity. Lesser MIC of nigella and sesame compare to coconut against *Salmonella typhi* and *E. coli* were earlier reported [50-53]. Differences in antibacterial activity of oils could be justified by the presence of different classes of fatty acids including PUFA like linolenic acid, arachidonic acid, and MUFA like palmitoleic, and oleic acid.

Results obtained for the antibacterial activity of nigella oil were in similarity with those informed by Ani et al. [54] and Mohammed et al. [55] when screening antibacterial activity against Candida Albicans and *Escherichia Coli*. The results support the use of nigella oil and its main for their antimicrobial and anti-inflammatory properties. Nigella oil was reported to contain many substances like thymoquinone, dithymoquinone, negillicine, negillidine, and nigellimine [56] that have effects against microorganisms. The results obtained on the low MIC and MBC of sesame on E. coli and Staphylococcus aureus was in accordance with the result reported by Mohammed et al. [55], Anand et al. [57] also reported that sesame oil is found to have antibacterial activity against *Streptococcus mutans*, *Escherichia coli*, and total bacteria. This type of screening by using sesame and nigella oil with very effective antibacterial activity could be promising for future application in cosmetic formulations [57].

4. Conclusion

The present study was carried out on the chemical composition and physicochemical properties of seeds oil suggest that these seeds could be considered as an alternative source of oil and ingredient in cosmetic formulations. Screening of oils revealed the presence of several phytochemicals saponins, steroids, phenols, terpenoids, and phenolic compounds whose pharmacological effects have been documented previously. The TPC and USM which are important criteria for the selection of oil for cosmetics were higher in nigella, coconut, sesame, and moringa. The various oils demonstrate different antagonistic effects toward indicator microorganisms. The highest antibacterial activity was found in nigella, moringa, and sesame seed oils based on their low MIC and MBC. These oils decreased the growth of *Staphylococcus aureus*, *Salmonella typhi*, and *Escherichia coli* to significant levels. The oils of nigella, moringa, and sesame showed antibacterial activity towards all microorganisms. The antibacterial properties of the selected oils depended on their type as well as tested microorganism. From this study, we recommend that: nigella, sesame, and moringa should be preferred to avocado, coconut, and cocoa for cosmetic formulations. The government should encourage the domestication of these seeds and fruit to promote their local use and by-product and product application; Cosmeceutical industries should implement these oils in their product formulations so that the local population can continue using them in their everyday routine.

References

[1] Gubitosa, J. Rizzi, V. Fini, P. Cosma P., Hair Care Cosmetics: From Traditional Shampoo to Solid Clay and Herbal Shampoo. A Review. Cosmetics. 2019; 6, 13.

[2] Nora, A. and Csaba, F. Green Consumer Behavior in the Cosmetics Market, Resources, 2019; 8, 137.

[3] Antonio, M., Rabasco, A., and María, G. R., Lipids in pharmaceutical and cosmetic preparations. Grasas y Aceites; 2000; 51, 74-96.

[4] Timoszuk, M., Bielawska, K., Skrzydlewska, E., Evening Primrose (Oenothera biennis) Biological Activity Dependent on Chemical Composition. Antioxidants (Basel), 2018; 7(8), 108.

[5] Bonnet, C., Lipids, a natural raw material at the heart of cosmetics innovation. Oilseeds Fats Crops Lipids, 2018; 25(5), D501.

[6] Lautenschläger, H., Essential Fatty Acids – Cosmetic from Inside and Outside, 2003.

[7] A. Obiedzińska, B. Waszkiewicz-Robak . Oleje tłoczone na zimno jako żywność ŻYWNOŚĆ. Nauka. Technologia. Jakość, 2012, 1 (80), 27-44.

[8] Rios, J.L. and Recio, M.C. Medicinal Plants and Antimicrobial Activity. Journal of Ethnopharmacology, 2005; 100, 80-84.

[9] Katušin-Ražem, B., Mihaljević, B., Ražem D., Microbial decontamination of Cosmetic Raw Materials and Personal Care Products by Irradiation. Radiation Physics and Chemistry. 2003; 66 (4), 309-316.

[10] Lundov, M.D., Moesby, L., Zachariae, C., Johansen J.D., Contamination versus Preservation of Cosmetics: A Review on Legislation, Usage, Infections, and Contact Allergy, Contact Dermatitis. 2009; 60 (2), 70-78.

[11] Nair M.K.N., Vasudevan, P., and Venkitanarayanan, K., Antibacterial Effect of Black Seed Oil on Listeria Monocytogenes. Food Control. 2005, 16 (5), 395-398.

[12] Idu, M., Erhabor, J.O., Oghale, O.U., Obayagbona, N.O., Antimicrobial qualities, phytochemistry and micronutritional content of Khaya senegalensis (Desr.) A. Juss seed oil. The Journal of Phytopharmacology, 2014; 3(2): 95-101.

[13] Kashmiri, M.A., Yasmin, S., Ahmad, M. Mohy-ud-Din, A. Characterization, Compositional Studies, Antioxidant and Antibacterial Activities of Seeds of Abutilon Indicum and Abutilon Muticum Grown Wild in Pakistan. Acta Chimica Slovenica. 2009; 56, 345-352.

[14] Akbar, E., Yaakob, Z. Kamarudin, S. K. Ismail, M. and Salimon, J. Characteristic and composition of Jatropha Curcas oil seed from Malaysia and its potential as biodiesel feedstock. European Journal of Scientific Research, 2009; 29, 396-403.

[15] AOAC. (2005). Horwitz (Ed.), Official Methods of Analysis of AOAC International (18th ed.) AOAC International. Gaithersburg MD, USA.

[16] AFNOR (Association Française de Normalisation), recueil des norme française des corps gras, graine oléagineuses, produits dérivés 2^{e},1981.

[17] Banso, A., Adeyemo, S., Phytochemical screening and antimalarial assessment of Abutilon mauritianum, Bacopa monnifera and Datura stramonium. Biokemistri, 2006; 18, 39-44.

[18] Joshi, A., Maya, B., and Ahma, S., Phytochemical investigation of roots of *Grewia microscosLinn*. *Journal of chemical and pharmaceutical research*. 2013; 5(7):80-87.

[19] Gibbs R.D., 1974. Chemotaxonomy of Flowering Plants. Vol.1, McGill Queen's University Press, Montreal and London.

[20] Ayoola, G., Coker, H.A., Adesegun, S.A., Adepoju-Bello, A., Obaweya1 K., Ezennia, EC., Atangbayila T., Phytochemical Screening and Antioxidant Activities of Some Selected Medicinal Plants Used for Malaria Therapy in Southwestern Nigeria. *Tropical Journal of Pharmaceutical Research*. 2008; 1019-1024.

[21] Gao X., Ohlander, M., Jeppsson, N., Björk, L., Trajkovski, V., Changes in antioxydant effects and their relationship to phytonutrients in fruits of sea buckthorn (Hippophae rhamnoides L.) during maturation, *Journal of Agriculture and Food Chemistry*, 2000; 48, 1485-149.

[22] Chidozie, V.N., Adoga, G.I., Chukwu, O.C., Chukwu, I.D., Adekeye, A.M., Antibacterial and toxicological effects of the aqueous extract of Mangifera Indica stem bark on albino rats. 2014; Global Journal of Biology, Agriculture & Health Sciences (G.J.B.A.H.S.), Vol.3(3): 237-245.

[23] Mouokeu, R.S., Womeni, H.M., Njike, N.F., Kuiate, J.R., Chemical composition and antibacterial activity of oils from Chrysicthys nigrodigitatus and Hepsetus odoe, two freshwater fishes from Yabassi, Cameroon. *Lipids in Health and Disease*. 2018; 17: 45.

[24] Ogbunugafor, H.A., Eneh, F.U., Ozumba, A.N., Igwo-Ezikpe, M.N., Okpuzor, J., Igwilo, I.O., Adenekan, S.O., and Onyekwelu, O.A., Physico-chemical and Antioxidant Properties of Moringa oleifera Seed Oil. Pakistan Journal of Nutrition. 2011; 10(5), 409-414.

[25] Saeed, M.D. and Shola, E.A., Extraction and physicochemical properties of some edible seed oils sampled in Kano Metropolis, Kano State. Bayero Journal of Pure and Applied Sciences. 2015; 8(2), 239-244.

[26] Warra, A., Cosmetic potentials of African Shea nut (vitellaria paradoxa) butter. *Journal of Current Resources in Chemistry*. 2011; 3: 80-86.

[27] Codex Alimentarus. (1999). Codex Standard for Edible Fats and Oils Not Covered by Individual Standards., Rev. 2.Italy: Codex Alimentarius; 2009.

[28] Oderinde, R.A., Ajayi, I.A., Adewuyi, A., Characterization of seed and seeds oil of Hura Crepitans and the kinetics of degradation of the oil during heating. Electron. *Journal of Environment and Agriculture Food Chemistry*, 2009 8(3): 201-208.

[29] Abayeh, O.J., Aina, E.A., and Okuonghae, C.O., Oil content and oil quality characteristics of some Nigerian oil seeds. *Journal of Pure Applicational Science, 1998*; 1: 17-23.

[30] Firestone D., Physical and Chemical Characteristics of Oils, Fats and Waxes. Champaign, Ill, USA: AOCS Press, 1997.

[31] Izuagie, A., Akpambang, V.O.E., Amoo, A., Comparativecompositional analysis on two varieties of melon (Colocynthis citrullus and Cucumeropsis edulis) and a variety of almond (Prunus amydalus). Res. J. Agric. Biol. Sci., 2008; 4(6), 639-642.

[32] A.O.A.C. (1990) Official Methods of Analysis. 15th Edition, Association of Official Analytical Chemist, Washington DC.

[33] Zia-Ul-Haq, M., Ahmad, M., Iqbal, S., Ahmad, S., and Ali, H., Characterization and Compositional Studies of Oil from Seeds of Desi Chickpea (Cicer arietinum L.) Cultivars Grown in Pakistan. *Journal of the American Oil Chemists' Society*, 2007; 84 (12), 1143-1148.

[34] Ikram, B.A., Nizar, T., Enrique M., Ana, G. P., Rubio Maria C. P. C. Ali A. Sadok B. Content of carotenoids, tocopherols, sterols, triterpenic and aliphatic alcohols, and volatile compounds in six walnuts (Juglans regia L.) varieties. Food Chemistry, 2015; 173, 972-978.

[35] Muanda, F. N., Identification de polyphénols, évaluation de leur activité antioxydante et étude de leurs propriétés biologiques. Thèse de Doctorat, Université Paul Verlaine-Metz. 2010; 55-86.

[36] Voukeng. Activités antibactériennes de onze épices Camerounaises et leurs effets en association avec les antibiotiques sur les bactéries Gram negative multirésistantes in vitro. Thèse de Master II, Université de Dschang, 2011; 71 pp.

[37] Fankam, G. R., Kuete, V., Voukeng, K.I., Kuiate, J. R., and Pages, J. M., Antibacterial of selected Cameroonian spices and their synergistic effets with antibiotic against Multidrug-resistant phenotypes. *Complementary and Alternative Medicine*. 2011; 11, 104.

[38] NGuessan, H. A., Déliko, D.C.E., Békro, M. J. A., and Békro, Y.A., CCM Dextraits selectifs de 10 plantes utilisees dans le traitement traditionnel de l'hypertension arterielle en cote d'ivoire. *European journal of Scientific Research*, 2011 ; 4, 575-585.

[39] Lacmata, S. T., Kuete, V., Dzoyem, J. P. Tankeo, S. B. Ngo, T. G., Kuiate, J. R., and Pages, Antibacterial activities of selected Cameroonian plants and their synergistic effects with antibiotics against bacteria expressing MDR phenotypes. *Complementary and Alternative Medicine*, 2012; 1-11.

[40] Harbone, J. B., Phytochemical methods: A guide to modern techniques of plant analysis. Chapman and Hall Ltd., London. 1973; 116.

[41] Kahouli I. Effet antioxydant d'extraits de plantes (Laurus nobilis L., Rosmarinus officinalis, Origanum majorana, Oléa europea L.) dans l'huile de canola chauffée.Thèse de Master, Université Laval Québec, 2010; 16-52.

[42] Nurul, A., Rizki, M.A., Laras, C. and Rahmahdona, S., Physical and Chemical Characteristic of Virgin Coconut Oil under Mix Culture Fermentation Technique. Journal of Physics: Conference Series 1364. 2019, 012009, IOP Publishing.

[43] Ajayi, A., Ude, A. N., and Balogun, Fulafia. O. J., Qualitative and Quantitative Phytochemical Analysis of Moringa Oleifera and Vernonia Amygdalina. *Journal of Science and Technology*, 2017; 3.

[44] Marja, P.K., Anu, I.H., Heikki, J.V., Jussi-Pekka, R., Kalevi, P., Tytti, S.K., and Marina, H. Antioxidant Activity of Plant Extracts Containing Phenolic Compounds Journal of Agricultural and Food Chemistry. 1999; 47 (10), 3954-3962.

[45] Hassanein, M.M.M., El-Shami, S.M., El-Mallah, M.H., Investigation of, lipids profiles of Nigella, Lupin and artichoke seed oils to be used as healthy oils. J. Oleo. Sci., 2011; 60, 99-107.

[46] Oyi, A.R., Onaolapo, J.A. and Obi, R.C. Formulation and Antimicrobial Studies of Coconut (Cocos nucifera Linne) Oil. Research Journal of Applied Sciences, Engineering and Technology, 2010; 2(2), 133-137.

[47] DebMandal, M., and Mandal, S., Coconut (Cocos nucifera L.: Arecaceae): in health promotion and disease prevention. *Asian Pacific Journal of Tropical Medecin*e, 2011; 4, 241-247.

[48] Abebe, E., Gugsa, G., Ahmed, M., Review on Major Food-Borne Zoonotic Bacterial Pathogens. J Trop Med., 2020; 4674235.

[49] Bintsis, T., Foodborne pathogens. AIMS microbiology, 2017; 3(3), 529-563.

[50] Huang, C.B., George, B., Ebersole, J.L., Antimicrobial activity of n-6, n-7 and n-9 fatty acids and their esters for oral microorganisms. Archives of Oral Biology. 2010; 55(8): 555-60.

[51] Silva L. Antimicrobial peptides from animals: focus on drug discovery. *Letters in Drug Design and Discovery*. 2011; 1(3): 230-6.

[52] Orhan, I.E., Ozcelik, B., Sener, B., Evaluation of antibacterial, antifungal, antiviral, and antioxidant potentials of some edible oils

and their fatty acid profiles. *Turkish Journal of Biology*, 2011; 35, 251-258.

[53] Sugeng, H.S., Saraswati, Sri, H., Ayu, F.I., Fatty Acid Composition of Some Potential Fish Oil from Production Centers in Indonesia. *Oriental Journal of Chemistry*, 2014; 30 (3).

[54] Ani, V., Varadaraj, M.C., and Naidu, K.A., Antioxidant and antibacterial activities of polyphenolic compounds bitter cumin. *European Food Research and Technology*, 2006; 97, 396-403.

[55] Mohammed A. Study of antibacterial activity of Nigella sativa ethanol extract on the growth of Staphylococcus aurus in culture media. *Kufa Journal for Veterinary Medical science*, 2017; 8 (2).

[56] Bakkali, F., Averbeck, S., Averbeck, D., and Idaomar, M., Biological effects of essential oils. *Food and Chemical Toxicology*, 2007; 46: 446-475.

[57] Anand, D.T., Pothiraj, C., Gopinath, R.M., Effect of oil pulling on dental caries causing bacteria. *African Journal of Microbiology research*, 2008; 10, 160-163.