ACS APPLIED MATERIALS & INTERFACES

Research Article

www.acsami.org

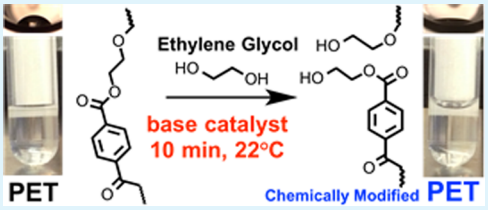
Transforming Plastic Surfaces with Electrophilic Backbones from Hydrophobic to Hydrophilic

Samuel Kim,[†] Raffick A. R. Bowen,[‡] and Richard N. Zare*[,†]

[†]Department of Chemistry and [‡]Department of Pathology, Stanford University, Stanford, California 94305, United States

Supporting Information

ABSTRACT: We demonstrate a simple nonaqueous reaction scheme for transforming the surface of plastics from hydrophobic to hydrophilic. The chemical modification is achieved by base-catalyzed trans-esterification with polyols. It is permanent, does not release contaminants, and causes no optical or mechanical distortion of the plastic. We present contact angle measurements to show successful modification of several types of plastics including poly(ethylene terephthalate) (PET) and polycarbonate (PC). Its applicability to blood analysis is explored using chemically modified PET blood collection tubes and found to be quite satisfactory. We expect this approach will reduce the cost of manufacturing plastic devices with optimized wettability and can be generalized to other types of plastic materials having an electrophilic linkage as its backbone.



KEYWORDS: *surface modification, glycolysis, PET, blood collection devices, wettability, contact angle*

1. INTRODUCTION

Plastics are made of organic polymers and have excellent properties, such as light weight, moldability, chemical and physical durability, and electrical and thermal insulation, in conjunction with low manufacturing cost. Consequently, plastics have found ubiquitous uses in modern life. Unlike glass, however, plastics have poor wettability. This characteristic can sometimes interfere with their use because the plastic easily adsorbs other hydrophobic molecules in contact with its hydrophobic surface.[1,2] Therefore, modification of a hydrophobic polymeric surface into a hydrophilic one is frequently desired.

Conventional approaches for converting hydrophobic plastic surfaces to hydrophilic include plasma treatment,[3−6] UV irradiation,[7,8] and graft polymerization.[9,10] The physical method of exposing surfaces to plasma and photons are widely used as a general, fast, and adjustable approach to modify various types of plastics.[11] The costs for vacuum environment and equipment, however, are higher than other methods. Moreover, in the case of poly(ethylene terephthalate) (PET), a widely used plastic, the plasma-treated surface has been reported to relapse to a certain degree of hydrophobicity over storage time presumably because of the rearrangement of introduced functional groups.[12−14] Grafting technology is also a method for introducing new properties to the surface of a polymeric structure, but surface activation necessary to initiate polymerization of monomers or coupling reactions is often facilitated by plasma or UV treatment, thereby demanding again infrastructure costs. Some manufacturers overcome problems associated with the hydrophobicity of the plastic surface by coating it with surfactants. However, this approach raises the concern that the surfactants may interfere with other uses.[15] More recently, surface modification of PET based on melt blending with polyethylene glycol (PEG) was reported where the addition of polystyrene promoted surface presentation of the more hydrophilic component of PEG.[16] However, mixing with a different type of plastic can alter bulk properties, which may limit the applicability of this approach. Additionally, PEGylation of the PET surface via an adhesive coating has been proposed and shown to be effective for preventing adsorption of biomolecules,[17] with emphasis on biocompatibility in the context of cell culture.

We describe an alternative method for making the surface of plastic materials hydrophilic. It is based on trans-esterification of the polymer surface in contact with liquids containing multiple hydroxyl groups per molecule, such as ethylene glycol and glycerol, in the presence of a base to catalyze the reaction under nonaqueous conditions (Figure 1). Our scheme is effective with organic polymers whose backbones are made of electrophilic linkages. For example, PET is a polymer made of ethylene glycol and terephthalic acid units with ester bond linkages (Figure 1a). When ethylene glycol, as a reactant, is deprotonated by a strong base catalyst, it becomes a nucleophile that attacks the carbonyl carbon of PET in its backbone. The polymeric chain breaks and incorporates hydroxyl groups from the polyol.

This reaction, also called glycolysis, has been explored extensively as a viable method for decomposing PET wastes for recycling purposes.[18−20] It also has been applied to PET fibers to enhance their wettability using inorganic base catalysts.[21,22] In contrast to the harsh reaction conditions used in these previous efforts for complete depolymerization or transformation, our scheme was optimized for a milder condition, especially below the glass transition temperature of PET (about

Received: November 5, 2014
Accepted: January 7, 2015
Published: January 7, 2015

DOI: 10.1021/am507606r
ACS Appl. Mater. Interfaces 2015, 7, 1925−1931

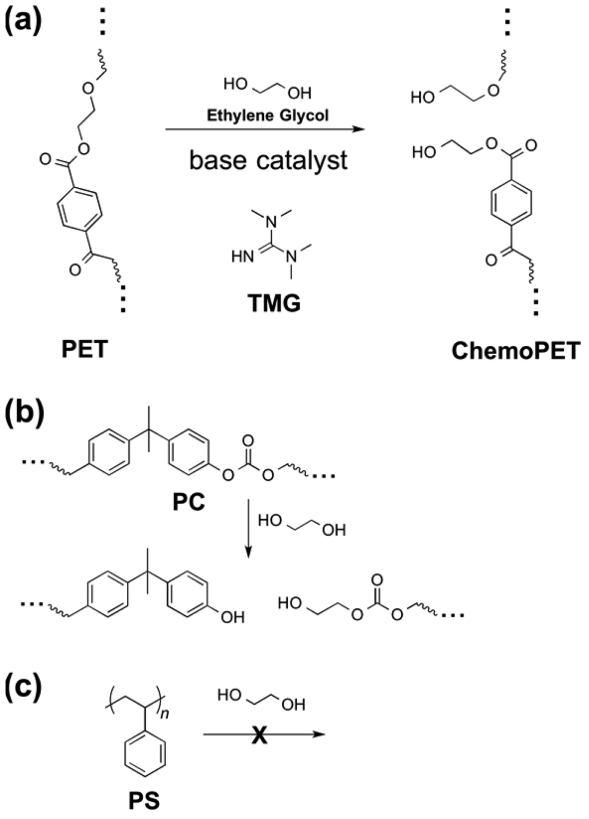


Figure 1. Trans-esterification reactions for transforming the surface of plastics by contact with ethylene glycol (EG) in the presence of a base catalyst to a hydrophilic surface. (a) PET is converted to a chemically modified version, which we call ChemoPET, using the base catalyst, 1,1,3,3-tetramethylguanidine (TMG). (b) Transformation of polycarbonate (PC) can be achieved using a slightly modified reaction conditions. (c) Polystyrene (PS), used as a control, does not contain reactive sites for our scheme and thus is not chemically modified.

70 °C), to modify only the surface layer and to prevent the loss of mechanical strength and optical transparency.

This base-catalyzed esterification approach for surface modification can be also applied to other polymers such as polycarbonate (PC), which contain reactive sites to be targeted by strong nucleophiles, namely the polyol molecules deprotonated by the base catalyst (Figure 1b).

We report the successful transformation of these types of plastic surfaces by measuring contact angles using static sessile drop and captive air bubble methods.[23] Of particular interest is the use of blood collection tubes (BCTs) made from PET as these tubes are the most commonly used ones in clinics and hospitals but must be treated to make them hydrophilic for carrying out various blood tests without the interference from blood components preferentially sticking to the walls of the container. We compare the performance of our modified PET tubes (ChemoPETs) against commercially available BCTs in the context of routine hormone assays to demonstrate practical advantages of using ChemoPETs.

2. MATERIALS AND METHODS

2.1. Surface Modification and Characterization. Ethylene glycol (EG), potassium hydroxide (KOH), 1,5,7-triazabicyclo[4.4.0]-dec-5-ene (TBD), and 1,1,3,3-tetramethylguanidine (TMG) were purchased from Sigma-Aldrich (St. Louis, MO). Glycerol (GL) (>99.5% pure) was obtained from Invitrogen (Carlsbad, CA). The 1 in. × 1 in. flat plastic pieces were cut from square bottles or flat panels for the following plastic types: (1) PET (Corning cat. 46-000-CM, Corning, NY), (2) PET glycol-modified (PETG, a variant of PET with an additional monomeric unit of 1,4-cyclohexanedimethanol) (Nalgene cat. 2019-0500, Penfield, NY), (3) PC (Corning cat. 431430, Corning, NY), and (4) PS (Plaskolite nonglare polystyrene sheet, Columbus, OH). PS pieces were immersed into *n*-hexane to remove residual adhesives or coatings. Base catalyst (KOH, TBD, or TMG) was dissolved in EG at specified concentrations (20−40%). A 5 mL portion of catalyst-containing EG solution was poured into reaction chambers containing 1 in. × 1 in. square plastic pieces or into each blood collection tube. Then, a batch of plastic materials was incubated at either room temperature (22 °C for PET, PETG, PS) or in a 55−60 °C incubator (for some PET tubes and PC) for specified durations. After incubation, the EG solution was collected for repeated use, and the plastic samples were rinsed with deionized water and dried by blowing with a stream of filtered air.

For FT-IR measurements, flat PET plaques were placed in contact with 20% (v/v) TMG solution made in EG, incubated at room temperature for 2 h, rinsed with deionized water, and then dried inside an oven (40 °C) for 6 h to remove any trace amount of water on the surface. Attenuated total reflection (ATR) mode was used to detect changes at the surface of the plastics using an FT-IR spectrometer (VERTEX 70, Bruker Optics Inc., Billerica, MA).

To assess hydrophilicity of the modified surface, the contact angle was measured by recording digital images of a water droplet (5 μL) on a flat plastic surface or an air bubble (5−15 μL) captured under water using a contact angle goniometer (model 100-F0, ramé-hart instrument co., Succasunna, NJ). The obtained images were analyzed by fitting with the DROPimage software (version 2.0.05) provided by the manufacturer. Measurements for each plastic type were repeated 5 times for averaging. Welch's *t*-test[24] was used for assessing statistical significance of the changes in contact angles. For atomic force microscopy (AFM) measurements, five different locations on unmodified and modified PET surfaces were scanned at 60 nm scale for 5 μm × 5 μm range using the tapping mode (Veeco Nanoscope 3100 AFM). These values were used to assess the influence of surface roughness on the measured contact angles.

2.2. PET Blood Collection Tubes Used. The following types of evacuated BCTs were examined in this study: (1) a plastic Vacuette (Greiner Bio-One, gold-top tube with gel separator; 13 mm × 75 mm, cat. 454228; lot B091209, Monroe, NC); (2) a glass tube (Becton Dickinson (BD, Franklin Lakes, NJ), red-top Vacutainer no-additive blood tube; 16 mm × 100 mm, cat. 366441; lot 2219385 from BD); (3) a plastic SST tube (BD, gold-top Vacutainer tube with gel separator; 13 mm × 75 mm, cat. 367983; lot 2258708); (4) a plastic RST tube (BD, orange-top Vacutainer tube with gel separator; 13 mm × 100 mm, cat. 368774; lot 120804); (5) a plastic plain red-top (PRT) tube (BD, Vacutainer tube with no gel separator; 13 mm × 100 mm, cat. 367814; lot 2200653). Chemically modified tubes were made from unmodified PET tubes (BD, 3 mL Vacutainer tubes with no interior coating; 3 mL, cat. no. 366703; lot 2160209).

Glass tubes are considered the controls in this study because this type has been the standard device for collecting serum samples for over five decades. In modern medical practice, however, glass BCTs have been replaced by plastic tubes following the Occupational Safety and Health Administration's guidelines that raise concerns about the dangers of broken glass causing exposure to blood-borne pathogens.[25] The glass tubes contain no clot activator, internal tube coating, or separator gel. The composition and additives for the glass, Vacuette, PRT, RST, and SST tubes are shown in Supporting Information Table S1. All blood collection tubes were used before their expiration dates.

2.3. Chemical Analysis of Quality Control (QC) Materials and Patient Blood Samples. QC materials are used to monitor the performance of the analytical measurement process to ensure that analytical variability meets accuracy and precision requirements that have been established for a measurement procedure and are considered appropriate for patient care. QC materials are typically noncommutable with native patient samples because the matrix of the QC material has been altered from that of native patient specimens. The matrix alteration is due to processing of the QC material during

DOI: 10.1021/am507606r
ACS Appl. Mater. Interfaces 2015, 7, 1925−1931

manufacturing; use of partially purified human and nonhuman analyte additives to achieve the desired concentrations; and various stabilization processes that can alter proteins, cells, and other components.

QC materials were poured and mixed for 30 min in each type of blood collection tube. All QC materials from the different tube types were transferred into 13 mm × 75 mm plastic test tubes. The samples were capped at room temperature if they were tested within 4 h. Alternatively, they were stored between testing intervals at 4 °C for up to 7 days. The thyroid hormones, triiodothyronine (TT_3), thyroxine (TT_4), and cortisol, were shown in our laboratory to be stable for 7 days at 4 °C. After pouring and mixing the QC material from 6 different types of blood collection tube, serum immunoassay analyte levels were measured in random order on an Immulite 1000 analyzer (Siemens Healthcare Global, Malvern, PA), according to the manufacturer's instructions. TT_3, TT_4, and cortisol levels were measured by competitive immunoassays using limited immobilized antibodies and labeled hormones. During the study, one reagent lot and one calibrator lot were used for the Immulite 1000 analyzer.

For patient samples, blood from 5 volunteers was collected via a syringe and slowly dripped into 5 different blood collection tubes. The blood collection tubes were inverted 8 times after the blood was drawn to ensure proper mixing of the blood with tube additives. Serum samples from the tubes were obtained after clotting for 60 min at room temperature followed by centrifugation at 1300*g* for 10 min. Following centrifugation, all tubes were inspected visually for complete barrier formation (except those without separator gels: glass, PRT, and ChemoPET tubes), fibrin, and hemolysis. All serum samples were processed within 2 h of blood collection. The samples were then analyzed in triplicate per patient using the same protocol described above for the QC materials. One-way analysis of variance (ANOVA) and the Bonferroni correction were used to determine statistically significant differences in obtained analyte concentrations.

To determine whether chemistry analytes concentrations in ChemoPET tubes were significantly different than commercially available blood collection tubes (BD glass, RST, PRT, SST; Greiner Vacuette), QC or serum samples from apparently healthy volunteers were poured or collected into the different BCT types, and a comprehensive metabolic panel was run from an aliquot of each tube using a Siemens Dimension RxL analyzer. The QC material and serum from apparently healthy volunteers were analyzed singly in random order and in the same analytical run. The analyte concentrations were not significantly or clinically different among the tube types examined (data not shown). These results are in agreement with a previous study[26] that found that the silicone surfactant, Silwet L-720, did not significantly alter concentrations of routine chemistry analytes.

3. RESULTS AND DISCUSSION

To detect the changes of functional groups at the plastic surface, FT-IR spectra of PET before and after modification were obtained. The two spectra were almost identical except in the region 3100−3700 cm^{-1} (Figure 2a). The calculated difference spectrum revealed that the change of IR absorption in this region corresponds to the vibrational frequency of an alcoholic or phenolic O−H stretch with its typical broad peak shape (Figure 2b). Because the modification occurs only at the top surface of the plastic and the penetration depth of the ATR mode is about 1 μm, the IR signals from added hydroxyl groups are expected to be weak. Multiple measurements at higher resolution confirmed that this difference is reproducible and larger than the noise level of the FT-IR equipment used for data acquisition (Supporting Information Figure S1).

To assess the degree of surface modification quantitatively, we characterized the surface roughness using AFM, and we measured contact angles of water droplets on the prepared plastic materials, correcting for surface morphology using the Wenzel equation.[27,28] Successful hydrophilic transformation is

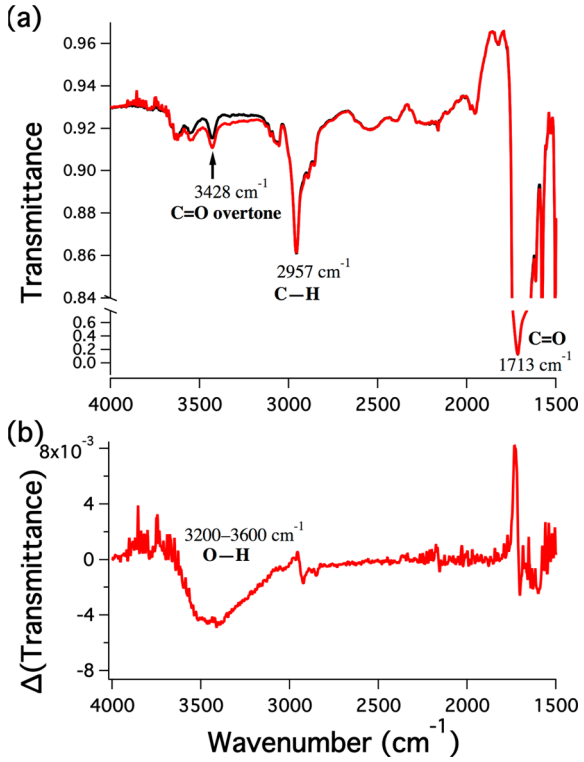


Figure 2. (a) FT-IR spectra of unmodified PET (black) and modified PET (red) obtained using an ATR mode. (b) Difference spectra obtained by subtracting transmittance values of unmodified PET from those of modified PET. Prominent vibrational frequencies and their corresponding chemical bond types are noted.

indicated by the decrease of contact angles as shown in Figure 3.

Table 1 summarizes the contact angle data. In the case of PET, the EG-modified surface had a mean contact angle of 41 ± 4° and was significantly smaller than the bare PET surface of 70 ± 2°, $t(7) = 14.95$, $p < 0.0001$. Similarly, the GL-modified PET had an average contact angle of 58 ± 5°, indicating that trans-esterification resulted in a significant increase in wettability, $t(6) = 5.20$, $p = 0.002$. But the change was not as large as EG, indicating EG has a higher reactivity than GL under the same reaction conditions (20% (v/v) TMG catalyst, 2 h reaction at room temperature). PETG, a variant of PET with an additional monomeric unit of 1,4-cyclohexanedimethanol (CHDM), showed essentially the same trend as PET albeit with a slight difference in the starting contact angle of the unmodified surface.

One might wonder what effect surface morphology plays. For PET and modified PET, AFM measurements yielded the roughness ratio *r*, defined as the ratio of the measured surface area to the projected surface area, of 1.08 and 1.24, respectively (see Supporting Information Figure S4). According to the Wenzel model, the measured contact angle (θ_m) is related to the contact angle for the ideal, flat surface (θ_γ, Young's contact angle) by $\cos \theta_m = r \cos \theta_\gamma$. Simple calculation shows that the corrections are relatively minor. For the bubble method contact angles, $\theta_m = 78°$ for PET and 41° for ChemoPET, the corrected values are $\theta_\gamma = 79°$ for PET and 53° for ChemoPET. Thus, the change in surface roughness induced by chemical modification cannot account for the large change in contact angles we have observed.

DOI: 10.1021/am507606r
ACS Appl. Mater. Interfaces 2015, 7, 1925−1931

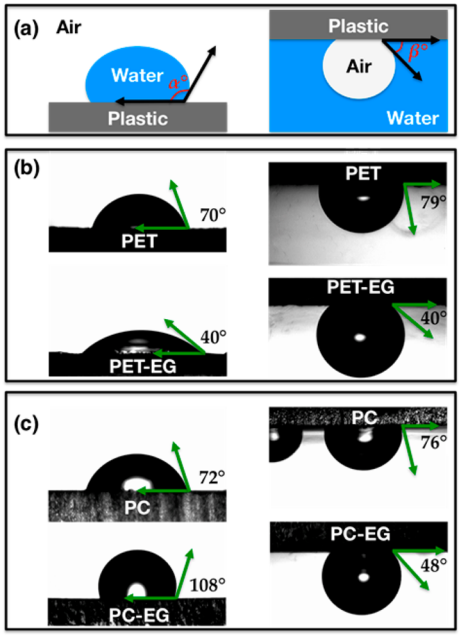


Figure 3. Images showing how contact angle measurements are made using the sessile drop method (left column) and the captive bubble method (right column): (a) schematics of the two contact angle measurement modes; (b) PET and (c) PC, and their chemical modifications using EG. These images are from individual measurements; therefore, the contact angle values differ slightly from the average values presented in Table 1.

Table 1. Average Contact Angles for Plastics before and after Chemical Modification (n = 5; Error Is Standard Deviation)

plastics	droplet method contact angle, α (deg)	bubble method contact angle, β (deg)
PET	70 ± 2[a]	78 ± 2
PET-EG	41 ± 4	41 ± 2
PET-GL	58 ± 5	
PETG	75 ± 1	
PETG-EG	47 ± 1	
PETG-GL	66 ± 4	
PC	71 ± 3	77 ± 2
PC-EG[b]	108 ± 4	44 ± 3
PC-GL[b]	93 ± 7	
PS	80 ± 1	
PS-EG	82 ± 2	
PS-GL	85 ± 3	

[a]All of the reported values are averages ± standard deviations (SD, n = 5). [b]For PC-EG: 40% (v/v) TMG in EG, 2 h incubation at 60 °C. For PC-GL: 20% (v/v) TMG in GL, 2 h incubation at 60 °C. All other samples: 20% (v/v) TMG in EG or GL, 2 h incubation at room temperature.

For PC, the room temperature reaction did not alter contact angle values much. Interestingly, when the reaction temperature was increased to 60 °C, the modified plastic exhibited an even more hydrophobic surface with contact angles of 108° and 93° for EG and GL modifications, respectively, compared to 71° of the native PC surface (Figure 3c). However, during the washing step, the water-liking nature of the modified plastic was evident in that water tended to stay on top of the modified sides of the plastic pieces and avoided the bare PC surface. When the captive bubble method was used so that the PC surface was kept wet during the measurement, chemically modified PC resulted in the contact angle (44 ± 3°) that is significantly smaller than that of the unmodified PC (77 ± 2°, $t(6) = 20.00$, $p < 0.0001$). We speculate that the microstructure of the modified PC surface changes after complete drying to render it hydrophobic. However, the data from the captive bubble method, which simulates better the condition of plastic containers to be used in an aqueous solution, do support the transformation of PC surface property via the base-catalyzed trans-esterification chemistry.

We also examined PS as a control and found almost no change of contact angle (Table 1; Supporting Information Figure S2). Considering the molecular structure (Figure 1c), where no reactive sites can be targeted by a nucleophilic attack, such nominal change of contact angle is an expected result.

Modification of BCTs with Different Base Catalysts. In addition to an inorganic base, KOH, we tested two organic bases for their efficacy as catalysts for this trans-esterification of the PET surface; TBD and TMG are guanidine compounds of strong basicity (their pK_a values in acetonitrile are 25.96 and 23.3, respectively), which can catalyze various types of synthetic reactions.[29] We found that both TBD and TMG catalyzed the reaction to a sufficient degree of transformation into a hydrophilic surface, even at room temperature with a reaction time as short as 10 min. Moreover, as shown in what follows, ChemoPET tubes exhibited a lower analytical bias compared to widely adopted tube types, without the need of surfactant additives, in a number of different clinical chemistry analyses with quality control materials and blood samples from healthy volunteers.

Nonaqueous reactions with ethylene glycol in the presence of base catalysts KOH, TBD, and TMG at concentrations of 1.8, 0.72, and 1.6 M, respectively, resulted in a dramatic change of surface wettability, as observed by examining the water meniscus and contact angle (Figure 4 and Supporting Information Figure S3). The contact angles were approximately 70° for untreated PET tubes and 30° for ChemoPET tubes (Figure 4a,b). As evident in Figure 4b, the meniscus was nearly identical to that seen for water in glass tubes, demonstrating that the PET surface was successfully made hydrophilic. It is also notable that the optical transparency and the shape were unaltered by the chemical reaction. Additionally, the modified plastic tube retained its hydrophilic inner surface property and gas impermeability (that is, ability to hold a vacuum) for at least 12 months, as expected with the stability of covalent bonds and surface-only modification. KOH can be used to achieve a similar level of hydrophilicity as with TBD and TMG, albeit with a longer reaction time and higher temperature. Also, it

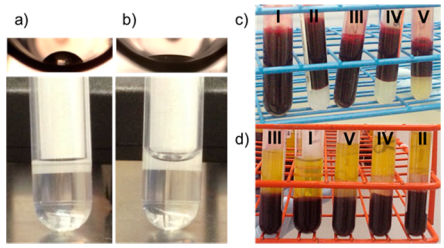


Figure 4. Observation of contact angle and water meniscus formed inside tubes made of (a) untreated PET and (b) ChemoPET treated with EG. Serum tubes containing human blood are shown (c) before and (d) after centrifugation. The five tube types are BD Glass (I), Greiner (II), ChemoPET (III), BD RST (IV), and BD SST (V).

DOI: 10.1021/am507606r
ACS Appl. Mater. Interfaces 2015, 7, 1925−1931

takes longer to dissolve KOH in ethylene glycol, which serves as both a solvent and a reactant; guanidine bases are readily soluble, making it easier to increase the catalyst concentrations.

Characterizing Performance of Prepared BCTs for Thyroid Hormones and Cortisol Assays. To check whether the ChemoPET tubes were compatible for blood storage and serum separation purposes, we collected blood samples from healthy volunteers into various types of BCTs and centrifuged according to tube manufacturers' recommendations. No red blood cell films on the interior wall and no hemolysis in the serum layer were observed in any of the tested tube types (Figure 4c,d). Hemolysis is the rupture of red blood cells and release of cellular constituents into serum. No difference in the mean hemolysis index of all serum samples, which was measured by a spectrophotometer, was found among the different BCT types (data not shown). Additionally, the serum samples collected using ChemoPET tubes were sent to Mayo Medical Laboratories (Rochester, MN) for a volatile chemical screening (for methanol, ethanol, isopropanol, acetaldehyde, and acetone) and ethylene glycol quantification. The ChemoPET tubes did not contain any detectable contaminants for clinical purposes (detection limit: 10 mg/dL). In addition, gravimetric analysis of unmodified and modified PET showed a negligible difference in mass (n = 10 tubes), as expected for modification of only the surface

The performance of ChemoPET tubes was compared with five different types of commercially available BCTs, using two different types of samples: QC materials and blood samples from five apparently healthy volunteers. The concentrations of three analytes (cortisol, total triiodothyronine (TT_3), and total thyroxine (TT_4)) were determined with an automated immunoassay instrument (Immulite 1000) and compared against the values obtained from glass tubes. Glass tubes are considered the control in this study, and contain no clot activator, internal tube coating, or separator gel; therefore, any deviation of analyte concentrations from those obtained with glass tubes indicates interference caused by plastic BCTs and their additives. Table 2 summarizes the results of this experiment. No statistically significant difference is found in comparing ChemoPET tubes with glass tubes whereas this claim cannot be made for some of the other plastic tubes (see Supporting Information Table S3).

For QC materials, ChemoPET and Greiner tubes showed significantly lower relative biases (+1.9% and +5.1% for cortisol; −3.3% and −2.2% for TT_3; −5.0% and −2.5% for TT_4, respectively) than BD SST, RST, and PRT tubes (e.g., for SST, +19.4% for cortisol; +15.0% for TT_3; +21.4% for TT_4, respectively; $p < 0.0001$ from the F-test).

The positive bias values observed from BD PET tubes, which are consistent with previous findings,[26,30] were larger than desirable bias derived from biological variations:[31] 10.26% for cortisol, 3.53% for TT_3, and 3.0% for TT_4. For the ChemoPET tubes, the bias of 5.0% slightly exceeded the desirable bias for TT_4 (Table 2). For blood samples from apparently healthy volunteers, the ChemoPET and Greiner tubes again showed lower relative biases (−3.8% and −1.2% for cortisol; +5.7% and +7.9% for TT_3; +0.2% and −2.7% for TT_4, respectively) than the BD PET tubes (e.g., for SST, +5.9% for cortisol; +17.0% for TT_3; +12.9% for TT_4, respectively). Thus, our ChemoPET tubes showed biases less than those of the BD plastic tubes and had a similar level of biases as the Greiner tubes. Considering that the manufacturing cost of ChemoPET tubes based on chemical modification via trans-esterification will be significantly lower than the BD and Greiner PET tubes, which are made by coating the surface with proprietary surfactants and silica particles, we believe that our method can provide an economical alternative to current market products.

Table 2. Comparison of Cortisol, TT_3, and TT_4 Concentrations in QC Material and from Five Apparently Healthy Volunteers Processed in ChemoPET Tubes and Commercial Brands (Details in Supporting Information Table S1)

	QC material (n = 9)		healthy volunteers (n = 5)	
	conc. (μg/dL)	% bias	conc. (μg/dL)	% bias
Cortisol				
BD Glass	42.8 ± 0.6[a]		8.8 ± 0.9	
ChemoPET	43.6 ± 0.5	+1.9[b]	8.5 ± 0.9	−3.8
BD SST	51.1 ± 0.7	+19.4	9.3 ± 0.9	+5.9
BD RST	52.8 ± 1.2	+23.4	9.4 ± 1.0	+6.1
BD PRT	47.1 ± 0.5	+10.0		
Greiner	45.0 ± 0.7	+5.1	8.7 ± 0.9	−1.2
TT_3[c]				
BD Glass	359 ± 5		83.5 ± 3.0	
ChemoPET	347 ± 5	−3.3	88.2 ± 3.7	+5.7
BD SST	413 ± 10	+15.0	97.6 ± 2.4	+17.0
BD RST	406 ± 5	+13.1	91.9 ± 3.9	+10.1
BD PRT	379 ± 3	+5.6		
Greiner	351 ± 6	−2.2	90.1 ± 2.6	+7.9
TT_4				
BD Glass	15.9 ± 0.4		6.1 ± 0.3	
ChemoPET	15.1 ± 0.3	−5.0	6.1 ± 0.3	+0.2
BD SST	19.3 ± 0.4	+21.4	6.9 ± 0.5	+12.9
BD RST	20.1 ± 0.4	+26.4	6.3 ± 0.4	+3.5
BD PRT	17.2 ± 0.2	+8.2		
Greiner	15.5 ± 0.2	−2.5	5.9 ± 0.3	−2.7

[a]All entries are means ± standard errors. [b]Biases are defined as deviations from values for the BD Glass. [c]TT_3 is reported in ng/dL, rather than μg/dL.

The biases between QC material and specimens from apparently healthy volunteers in TT_3, TT_4, and cortisol concentrations among tube types may be attributable to matrix effects. The QC material used in this study is serum-based, whereas the specimens from apparently healthy adults are serum isolated from whole blood specimens. The cellular material in the whole blood specimens from volunteers may adsorb some of the tube additives, particularly surfactants and/or clot activators, and, therefore, decrease its concentration in the serum layer, producing possibly less interference with components of the immunoassays studied. It is also conceivable that the higher volumes of whole blood from volunteers collected in the tubes (from 3.5 to 10 mL per tube) compared to QC specimens (2 mL per tube) may have diluted out the interferent(s), resulting in minimal alterations in the TT_3, TT_4, and cortisol concentrations. These findings suggests that the effect of tube additives on TT_3, TT_4, and cortisol assays may be greater if specimens from volunteers were partially compared to completely filled to their designated tube volumes with blood. Furthermore, the additives used in commercially available blood collection tubes are likely titrated for whole blood rather than QC specimens. Because QC materials are made from artificial sources that are different from authentic clinical samples, it is not uncommon that these two specimens may give different test results and interpretations as seen in this study.

DOI: 10.1021/am507606r
ACS Appl. Mater. Interfaces 2015, 7, 1925−1931

4. CONCLUSIONS

Nonaqueous trans-esterification with polyols catalyzed by organic bases is an efficient and inexpensive method to prepare glass-like surfaces for plastics having electrophilic backbone linkages. The scheme is effective for both PET and PC. Contact angle measurements show that the chemically modified PET and PC have been transformed from hydrophobic to hydrophilic in the presence of water. The chemically modified plastics are found to retain their optical and mechanical properties, and the modification is permanent and does not leach residue. Emphasis was placed on comparing chemically modified PET blood collection tubes with commercial PET tubes that have been treated by the manufacturers with surfactant coatings. Test results for the standard analysis of TT_3, TT_4, and cortisol in blood showed that our chemically modified tubes are comparable if not superior to those commercially available. Indeed, they behave in a similar fashion to glass blood collection tubes, which are considered to be the gold standard in blood analyses but cannot be used in hospitals because of the risk of cuts from broken glass containers exposed to blood. In consideration of cost and catalytic activity (Supporting Information Table S2), TMG was the best option among the tested catalysts in our study, but a similar organic base can be explored to reduce production costs. It should be noted that these catalysts are not consumed but recycled; also, the amount of ethylene glycol reactant used is minimal per reaction because only the surface layer is modified. Therefore, we believe that our chemical approach can significantly reduce manufacturing costs compared to current methods of spray-drying surfactants onto tube walls. Our approach also appears to give better uniformity and stability of surface modification than the spray-drying approach. Thus, the potential impact of this technology is expected to be wide-ranging, from liquid containers to medical devices.

■ ASSOCIATED CONTENT

Supporting Information

Details for the blood collection tubes examined (Table S1), reaction conditions and material costs of reagents (Table S2), statistical analysis of data from QC materials (Table S3), multiple FT-IR measurement data at a higher frequency resolution (Figure S1), contact angle images of PS (Figure S2), images of water meniscus formed inside ChemoPET tubes (Figure S3), and AFM images and parameters (Figure S4). This material is available free of charge via the Internet at http://pubs.acs.org.

■ AUTHOR INFORMATION

Corresponding Author

*E-mail: zare@stanford.edu.

Notes

The authors declare no competing financial interest.

■ ACKNOWLEDGMENTS

The authors thank Andrew Ingram and Robert Waymouth (Stanford University) for their helpful comments about utilizing organic bases and Brian Sae Yoon Kim (Stanford University) for help with AFM measurements.

■ REFERENCES

(1) Liston, E. M.; Martinu, L.; Wertheimer, M. R. Plasma surface modification of polymers for improved adhesion: a critical review. *J. Adhes. Sci. Technol.* **1993**, *7*, 1091−1127.
(2) Chan, C.-M.; Ko, T.-M.; Hiraoka, H. Polymer surface modification by plasmas and photons. *Surf. Sci. Rep.* **1996**, *24*, 1−54.
(3) Tsuchiya, Y.; Akutu, K.; Iwata, A. Surface modification of polymeric materials by atmospheric plasma treatment. *Prog. Org. Coat.* **1998**, *34*, 100−107.
(4) Chai, J.; Lu, F.; Li, B.; Kwok, D. Y. Wettability interpretation of oxygen plasma modified poly(methyl methacrylate). *Langmuir* **2004**, *20*, 10919−10927.
(5) Ni, W.; Liu, D.; Song, Y.; Ji, L.; Zhang, Q.; Niu, J. Surface modification of PET polymers by using atmospheric-pressure air brush-shape plasma for biomedical applications. *Eur. Phys. J.: Appl. Phys.* **2013**, *61*, 10801.
(6) Perez-Roldan, M. J.; Debarnot, D.; Poncin-Epaillard, F. Processing of plasma-modified and polymer-grafted hydrophilic PET surfaces, and study of their aging and bioadhesive properties. *RSC Adv.* **2014**, *4*, 31409.
(7) Hozumi, A.; Masuda, T.; Hayashi, K.; Sugimura, H.; Takai, O.; Kameyama, T. Spatially defined surface modification of poly(methyl methacrylate) using 172 nm vacuum ultraviolet light. *Langmuir* **2002**, *18*, 9022−9027.
(8) Ye, H.; Gu, Z.; Gracias, D. H. Kinetics of ultraviolet and plasma surface modification of poly(dimethylsiloxane) probed by sum frequency vibrational spectroscopy. *Langmuir* **2006**, *22*, 1863−1868.
(9) Ikada, Y. Surface modification of polymers for medical applications. *Biomaterials* **1994**, *15*, 725−736.
(10) Hu, S.; Ren, X.; Bachman, M.; Sims, C. E.; Li, G. P.; Allbritton, N. Surface modification of poly(dimethylsiloxane) microfluidic devices by ultraviolet polymer grafting. *Anal. Chem.* **2002**, *74*, 4117−4123.
(11) Inagaki, N. *Plasma Surface Modification and Plasma Polymerization*; Technomic Pub. Co: Lancaster, PA, 1996.
(12) Hsieh, Y.-L.; Timm, D. A.; Wu, M. Solvent- and glow-discharge-induced surface wetting and morphological changes of poly(ethylene terephthalate) (PET). *J. Appl. Polym. Sci.* **1989**, *38*, 1719−1737.
(13) Gupta, B.; Hilborn, J.; Hollenstein, C.; Plummer, C. J. G.; Houriet, R.; Xanthopoulos, N. Surface modification of polyester films by RF plasma. *J. Appl. Polym. Sci.* **2000**, *78*, 1083−1091.
(14) Vesel, A.; Mozetic, M.; Zalar, A. XPS study of oxygen plasma activated PET. *Vacuum* **2007**, *82*, 248−251.
(15) Bowen, R. A. R.; Hortin, G. L.; Csako, G.; Otañez, O. H.; Remaley, A. T. Impact of blood collection devices on clinical chemistry assays. *Clin. Biochem.* **2010**, *43*, 4−25.
(16) Rezaei Kolahchi, A.; Ajji, A.; Carreau, P. J. Enhancing hydrophilicity of polyethylene terephthalate surface through melt blending. *Polym. Eng. Sci.* **2014**, n/a−n/a.
(17) Heo, S.; Jeon, Y.-S.; Kim, S. I.; Kim, S. H.; Kim, J.-H. Bioinspired adhesive coating on PET film for antifouling surface modification. *Macromol. Res.* **2014**, *22*, 203−209.
(18) Paszun, D.; Spychaj, T. Chemical recycling of poly(ethylene terephthalate). *Ind. Eng. Chem. Res.* **1997**, *36*, 1373−1383.
(19) Lorenzetti, C.; Manaresi, P.; Berti, C.; Barbiroli, G. Chemical recovery of useful chemicals from polyester (PET) waste for resource conservation: A survey of state of the art. *J. Polym. Environ.* **2006**, *14*, 89−101.
(20) Sinha, V.; Patel, M. R.; Patel, J. V. Pet waste management by chemical recycling: A review. *J. Polym. Environ.* **2010**, *18*, 8−25.
(21) Choi, H.-M. Hygroscopic poly(ethylene terephthalate) by nonaqueous alkaline glycolysis. *Ind. Eng. Chem. Res.* **2007**, *46*, 7891−7895.
(22) Cho, J. Y.; Hong, C.-J.; Choi, H.-M. Microwave-assisted glycolysis for PET with highly hydrophilic surface. *Ind. Eng. Chem. Res.* **2013**, *52*, 2309−2315.
(23) Zhang, W.; Wahlgren, M.; Sivik, B. Membrane characterization by the contact angle technique: II. Characterization of UF-membranes and comparison between the captive bubble and sessile drop as

DOI: 10.1021/am507606r
ACS Appl. Mater. Interfaces 2015, 7, 1925−1931

methods to obtain water contact angles. *Desalination* **1989**, *72*, 263−273.
(24) Welch, B. L. The generalization of "student"s' problem when several different population varlances are involved. *Biometrika* **1947**, *34*, 28−35.
(25) Ernst, D. J. Plastic collection tubes decrease risk of employee injury. *Med. Lab. Obs.* **2001**, *33*, 44−47.
(26) Bowen, R. A. R.; Chan, Y.; Ruddel, M. E.; Hortin, G. L.; Csako, G.; Demosky, S. J.; Remaley, A. T. Immunoassay interference by a commonly used blood collection tube additive, the organosilicone surfactant Silwet L-720. *Clin. Chem.* **2005**, *51*, 1874−1882.
(27) Wenzel, R. N. Surface roughness and contact angle. *J. Phys. Colloid Chem.* **1949**, *53*, 1466−1467.
(28) Kubiak, K. J.; Wilson, M. C. T.; Mathia, T. G.; Carval, P. Wettability versus roughness of engineering surfaces. *Wear* **2011**, *271*, 523−528.
(29) Ishikawa, T. *Superbases for Organic Synthesis: Guanidines, Amidines and Phosphazenes and Related Organocatalysts*; Wiley: Chichester, U.K, 2009.
(30) Bowen, R. A. R.; Chan, Y.; Cohen, J.; Rehak, N. N.; Hortin, G. L.; Csako, G.; Remaley, A. T. Effect of blood collection tubes on total triiodothyronine and other laboratory assays. *Clin. Chem.* **2005**, *51*, 424−433.
(31) Westgard, J. Biological variation database, and quality specifications for imprecision, bias and total error (desirable and minimum). The 2014 update. https://www.westgard.com/biodatabase1.htm (accessed Jul 14, 2014).

DOI: 10.1021/am507606r
ACS Appl. Mater. Interfaces 2015, 7, 1925−1931