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Protein Kinase C Inhibitor Generates Stable Human Tolerogenic Dendritic Cells

Takuya Matsumoto,* Hitoshi Hasegawa,**† Sachiko Onishi,* Jun Ishizaki,* Koichiro Suemori,* and Masaki Yasukawa**†

Tolerogenic dendritic cells (DCs) are a promising tool for a specific form of cellular therapy whereby immunological tolerance can be induced in the context of transplantation and autoimmunity. From libraries of bioactive lipids, nuclear receptor ligands, and kinase inhibitors, we screened conventional protein kinase C inhibitors (PKCIs) bisindolylmaleimide I, G66983, and Ro32-0432 with strong tolerogenic potential. PKCI-treated human DCs were generated by subjecting them to a maturation process after differentiation of immature DCs. The PKCI-treated DCs had a semimature phenotype, showing high production of IL-10, and efficiently induced IL-10-producing T cells and functional Foxp3+ regulatory T cells from naive CD4+ T cells, thus eliciting a strong immunosuppressive function. They also showed CCR7 expression and sufficient capacity for migration toward CCR7 ligands. Additionally, PKCI-treated DCs were highly stable when exposed to inflammatory stimuli such as proinflammatory cytokines or LPS. Conventional PKCIs inhibited NF-κB activation of both the canonical and noncanonical pathways of DC maturation, thus suppressing the expression of costimulatory molecules and IL-12 production. High production of IL-10 in PKCI-treated DCs was due to not only an increase of intracellular cAMP, but also a synergistic effect of increased cAMP and NF-κB inhibition. Moreover, PKCI-treated mouse DCs that had properties similar to PKCI-treated human DCs prevented graft-versus-host disease in a murine model of acute graft-versus-host disease. Conventional PKCI-treated DCs may be useful for tolerance-inducing therapy, as they satisfy the required functional characteristics for clinical-grade tolerogenic DCs. The Journal of Immunology, 2013, 191: 2247–2257.

Dendritic cells (DCs) are a heterogeneous population of APCs that contribute to innate immunity and initiate, coordinate, and regulate adaptive immune responses in infection and inflammation (1). Successful initiation of these adaptive immune responses requires DC maturation. Immature DCs (iDCs) are able to take up soluble Ag but are poorly immunogenic because they express only modest levels of MHC molecules and low levels of costimulatory molecules and proinflammatory cytokines. During maturation, DCs lose their capacity to capture soluble Ags but gain T cell stimulatory capacity due to increased Ag processing and upregulation of MHC, costimulatory molecules, and cytokines (1, 2). Although potentially capable of initiating adaptive immune responses, DCs also play an important role in modulating the induction of tolerance (3–5). Tolerogenic DCs (tDCs) are generally characterized by an immature or semimature phenotype, with a capacity for high Ag uptake and low expression of costimulatory molecules (2, 6). Additionally, tDCs produce low amounts of proinflammatory cytokines and high amounts of anti-inflammatory cytokines. This results in anergy, apoptosis of effector T cells, or induction and expansion of regulatory T cells (7).

For patients with autoimmune diseases and graft rejection, any therapy capable of preventing or reducing the extent immune activation is highly desirable. Ex vivo–generated iDCs are considered to have strong potential for use in cellular therapy for these conditions. In fact, injection of ex vivo–generated tDCs has been shown to be beneficial in animal models of graft rejection (8–10) and autoimmune diseases, including collagen-induced arthritis (11, 12), diabetes (13–16), and experimental autoimmune encephalomyelitis (17). Human tDCs can be cultured in vitro from DC precursors using different compounds (7, 18). For this purpose, anti-inflammatory cytokines such as IL-10 (19–22) and TGF-β (22–24), vitamin D3 (Vit D3) (25–28), neuropeptides, including vasoactive intestinal peptide (17, 29, 30) and α-melanocyte–stimulating hormone (31), and anti-inflammatory/immunosuppressive drugs such as dexamethasone (Dexa) (28, 32, 33) and rapamycin (Rapa) (34–36) are often employed. However, the compound most suitable for clinical application remains undefined from the viewpoint of tolerogenic potential, stability, and capacity for migration toward secondary lymphoid organs, that is, the area where immune responses occur (37–39). Therefore, to obtain compounds that have stronger tolerogenic potential, we screened libraries of bioactive lipids, nuclear receptor ligands, and kinase inhibitors for molecules that would downregulate the costimulatory molecules CD80 and CD86 on DCs and produce the highest level of IL-10. Consequently, we obtained some protein kinase C inhibitors (PKCIs).

Members of the PKC family are serine-threonine protein kinases that are divided into three subfamilies on the basis of their substrate specificity: conventional α, β1, β2, and γ PKCs are diacylglycerol (DAG)- and calcium-dependent; novel ε, δ, μ, θ, and η PKCs are DAG-dependent and calcium-independent; and atypical ζ, ι, and λ PKCs respond to neither DAG nor calcium (40). It has been reported that modulation of cellular individual PKC activity induces
PKC-dependent cell differentiation and cytokine production in many cell types (40). PKC has been reported to play critical roles in mediating progenitor-to-DC differentiation and cytokine production from monocytes/macrophages and DCs (41–46). In the present study, we found that stable iDCs showing high production of IL-10 were generated by addition of PKCI during the process of maturation from iDCs. We also characterized the PKCI-treated iDCs obtained and clarified the mechanism responsible for their induction.

Materials and Methods

Abs and reagents

FITC-conjugated mouse anti-human CD1a (HI149), PE-conjugated mouse anti-human CD11c (B-ly4), allophycocyanin-conjugated mouse anti-human CD4 (RA-4), allophycocyanin-conjugated mouse anti-human CD14 (M6P9), and PE-conjugated rat anti-human IL-10 (JE83-19F1) mAbs were purchased from BD Biosciences (San Diego, CA). PE-conjugated mouse anti-human CD40 (MB89), FITC-conjugated mouse anti-human CD80 (MAB104), FITC-conjugated mouse anti-human CD86 (HB15a), PE-conjugated mouse anti-human CD86 (HA5.2B7), FITC-conjugated mouse anti-human HLA-ABC (B9.12.1), and PE-conjugated mouse anti-human HLA-DR (E8.12.2) mAbs were obtained from Beckman Coulter (Brea, CA). PE-conjugated rat anti-human Foxp3 (PCH101) and mouse anti-human CCR7 (C7R7.6B3) (47) mAbs were obtained from Biolegend (San Diego, CA). FITC-conjugated mouse anti-human CD25 (BC96) mAb was purchased from BioLegend (San Diego, CA). Recombinant human (rh) GM-CSF, rhIL-4, rhIL-1β, rhIL-6, rhTCI19, mouse anti-human IL-10 (25209), and mouse anti-human HLA-DR (E8.12.2) mAbs were obtained from Beckman Coulter (Brea, CA). PE-conjugated rat anti-mouse CD86 (GL1), PE-conjugated mouse anti-human HLA-ABC (B9.12.1), and PE-conjugated rat anti-mouse CD40 (3/23), FITC-conjugated hamster anti-mouse CD80 (2G9) mAbs were purchased from BD Biosciences. Carboxyfluorescein-conjugated rat anti-mouse CCR7 (4B12) mAb, recombinant mouse (rm) CCL19 and rmMIP-1α (9016) mAbs were obtained from R&D Systems (Minneapolis, MN). IFN-γ was purchased from PeproTech (Rocky Hill, NJ). rTNF-α was obtained from Dainippon Pharmaceutical (Osaka, Japan). Five kinds of PKC—βisodinylmethylamide I (Bis I) (48), Ge6993 (49), Ro32-0432 (50), Ge6976, and HBBDE—and antibodies to libraries of bioactive lipids, nuclear receptor ligands, and kinase inhibitors were purchased from Enzo Life Sciences (Plymouth Meeting, PA). CGP33535 was obtained from Tocris Bioscience (Bristol, U.K.). Bis I, Ge69933, and Ro32-0432 are conventional PKCIs, and all share a common inhibition of α, β, and γ isoforms (48–50). Ge6976, CGP33535, and HBBDE are PKCα-specific, PKCβ-specific, and PKCδ- and γ-specific inhibitors, respectively. PGE2 and LPS were obtained from Sigma-Aldrich (St. Louis, MO).

FITC-conjugated hamster anti-mouse CD11c (HL3), FITC-conjugated rat anti-mouse CD40 (3/23), FITC-conjugated hamster anti-mouse CD80 (16A3.1), PE-conjugated rat anti-mouse CD83 (Michel-19), FITC-conjugated rat anti-mouse CD86 (GL1), PE-conjugated mouse anti-human H-2Kd (SF1-1.1), and PE-conjugated rat anti-mouse I-A/I-E (2G9) mAbs were purchased from BD Biosciences. Carboxyfluorescein-conjugated rat anti-mouse CCR7 (4B12) mAb, recombinant mouse (rm) GM-CSF, rmTNF-α, rmIL-1β, and rmCCL19 were obtained from R&D Systems.

Generation of human DCs

Human peripheral blood was obtained from healthy donors after obtaining informed consent in accordance with procedures approved by the Human Ethics Committee, Ehime University. CD14+ cells from PBMCs were isolated using a CD14 isolation kit (Miltenyi Biotec, Bergisch Gladbach, Germany). The purity of the isolated CD14+ monocytes was >95% upon flow cytometric analysis. iDCs were generated from the CD14+ monocytes by culturing them in RPMI 1640 medium supplemented with 10% FBS (Life Technologies, Rockville, MD), 75 mg/ml rhGM-CSF, and 10 ng/ml rhIL-4 for 5 d. To induce maturation (mature DCs (mDCs)), iDCs were incubated with a maturation mixture containing rhTNF-α (10 ng/ml), rhIL-1β (10 ng/ml), and PGE2 (10 μg/ml) for a further 48 h. PKCI-treated DCs were generated by culturing iDCs with a maturation mixture in the presence of PKCI monocytes such as Bis I (10 μM), Ge6993 (10 μM), or Ro32-0432 (10 μM) for 48 h.

Flow cytometric analysis

DC cell surface expression and the expression of Foxp3 and IL-10 in CD4+ T cells were analyzed using each of the mAbs described in the Abs and reagents section labeled with FITC or PE on a FACSCalibur flow cytometer (BD Biosciences), as described previously (51). For intracellular Foxp3 and IL-10 staining, permeabilization was performed in accordance with the manufacturer’s protocol.

Assay for cytokine production from DCs

iDCs, mDCs, and PKCI-treated DCs were adjusted to 1 × 106 per well in 1 ml and cultured in RPMI 1640 medium supplemented with 10% FBS for assay of IL-6, IL-10, IL-12, and IFN-γ, or in X-VIVO medium for assay of TGF-β. The cell supernatants were harvested after 96 h, and cytokines were measured by ELISA. The supernatants were stored at −70°C until use. The supernatants were then assayed using a sandwich ELISA (R&D Systems) (IL-6, IL-10, IL-12, IFN-γ, and TGF-β), in accordance with the manufacturer’s instructions.

Phagocytic ability

Phagocytic ability was measured as the cellular uptake of FITC-latex beads/rabbit IgG using a phagocytosis assay kit (Cayman Chemical, Ann Arbor, MI). DCs were incubated with FITC-latex beads/rabbit IgG for 24 h in a CO2 incubator and then analyzed by flow cytometry in accordance with the manufacturer’s instructions.

Stability of PKCI-treated DCs

Phenotypic stability of PKCI-treated DCs was determined in response to inflammatory stimuli, as described by Harry et al. (37). Briefly, PKCI-treated DCs, iDCs, or mDCs were washed, adjusted to 5 × 105 per well in 1 ml, and cultured in RPMI 1640 medium supplemented with 10% FBS or in X-VIVO medium only for assay of TGF-β in the absence or presence of a mixture of proinflammatory cytokines containing IFN-γ at 1000 U/ml, IL-β, IL-6, and TNF-α each at 10 ng/ml or LPS at 0.1 μg/ml. After 1 and 5 d, mDCs were analyzed for cell surface phenotype by flow cytometry, and production of IL-10 and TGF-β in the culture supernatants was determined by ELISA. To examine the functional properties of the DCs, they were washed after stimulation with a mixture of proinflammatory cytokines or LPS and recultured in RPMI 1640 medium supplemented with 10% FBS for a further 24 h. Then, an MLR was performed as described in the in vitro T cell proliferation assay section.

In vitro T cell proliferation assay

Human CD4+ T cells were isolated from PBMCs using a CD4+ T cell isolation kit II (Miltenyi Biotec). Freshly isolated CD4+ T cells (1 × 105) were cocultured with 1 × 105 (10:1 T cell/DC ratio) or 2 × 105 (10:2) mitomycin C (MMC)-treated allogeneic DCs in a final volume of 200 μl X-VIVO medium supplemented with 10% FBS in 96-well round-bottom plates for 4 d. The wells were pulsed with 1 μCi [3H]thymidine (Amersham Biosciences, Piscataway, NJ) 18 h before harvesting.

For mouse in vitro T cell proliferation assays, mouse CD4+ T cells were isolated from spleens of C57BL/6 mice using a CD4+ T cell isolation kit II (Miltenyi Biotec). MMC-treated allogeneic DCs were prepared from BALB/c mice.

Induction of regulatory T cells by PKCI-treated DCs

Induction of human regulatory T cells by PKCI-treated DCs was described as performed by Sato et al. (21). Briefly, human naive CD4+CD45RA+ T cells were purified from PBMCs with a CD4+ T cell isolation kit II (Miltenyi Biotec) and subsequent negative selection with anti-CD45RA mAb-conjugated immunomagnetic beads (Miltenyi Biotec). Human naive CD4+CD45RA- T cells (5 × 105) were cultured with 5 × 105 MMC-treated allogeneic CD4+CD45RA- T cells and MMC-treated allogeneic CD4+CD45RA- mDCs in X-VIVO medium with 10% FBS for 5 d. Then, Foxp3+CD4+CD25+ T cells and IL-10+CD4+ T cells were analyzed by flow cytometry.

In vitro T regulatory activity

In vitro T regulatory activity was modified as described previously (51) and as reported by Gonzalez-Rey et al. (30). Human Th1 cells were generated from naive CD4+CD45RA- T cells. Briefly, naive CD4+CD45RA- T cells (1 × 105) were cultured with 1 × 105 MMC-treated allogeneic mDCs in X-VIVO medium with 10% FBS. Alloreactive T cells were expanded from day 6 in the presence of 100 μM rhIL-2. Two weeks after priming, T cells were restimulated with mDCs from the same donor under identical culture conditions. CD4+ T cells were isolated 1 wk after restimulation and used as alloreactive Th1 cells.

Human naive CD4+CD45RA- T cells (5 × 105) were cultured with 5 × 105 MMC-treated allogeneic DCs treated with PKCI in X-VIVO medium with 10% FBS for 5 d. Then, CD4+ T cells were purified from the above cocultures with a CD4+ T cell isolation kit II (Miltenyi Biotec). These purified CD4+ T cells (1 × 105) were incubated with syngeneic Th1 cells (responder T cells, 1 × 105) in the presence of MMC-treated allogeneic mDCs (1 × 105) in a final volume of 200 μl X-VIVO medium supplemented with 10% FBS in 96-well round-bottom plates for 4 d. The proliferation of
responder T cells was determined by [3H]thymidine incorporation. In some experiments, cocultures were performed in the presence of blocking mouse anti-IL-10 (40 μg/ml) and/or anti-TGF-β1 (40 μg/ml) mAbs. The anti-IL-10 and anti-TGF-β1 mAbs had been titrated previously. Concentrations resulting in undetectable levels of IL-10 or TGF-β1 following Ab addition were used in the blocking experiments.

Transwell experiments were performed using 24-well plates (Millipore, 0.4 μm; Millipore, Bedford, MA). Th1 cells (5 × 10⁹) together with MMC-treated allogeneic mDCs (5 × 10⁶) were placed in the bottom wells. CD4⁺ T cells (5 × 10⁶) generated in the presence of PKCI-treated DCs were placed in the upper wells. The cultures were performed in the absence or presence of anti-IL-10 and/or anti-TGF-β1 mAbs for 4 d. Then, the basket was removed and the proliferation of the responder T cells was measured.

**ELISA for measurement of RelA and RelB**

Human iDCs (1 × 10⁶/well) were incubated with PKCI for 2 h and then stimulated with TNF-α (10 ng/ml), IL-1β (10 ng/ml), and/or PGE₂ (1 μg/ml). After 4 h of stimulation, nuclear and cytoplasmic extracts from DCs were separated using a nuclear extraction kit (Imgenex, San Diego, CA). Activated NF-κB (RelA or RelB) in the nuclear extracts was measured by oligonucleotide-based ELISA as described by Bhattacharya et al. (52). Briefly, the extracts from DCs were incubated for 1 h at room temperature with digoxigenin in wells of a 96-well microtiter plate that had been coated with the NF-κB consensus nucleotide sequence (5′-GGGAGTTCCC-3′). NF-κB from the cell samples attached to the wells was captured by an Ab specific to either RelA or RelB (Santa Cruz Biotechnology, Santa Cruz, CA). Binding of the specific NF-κB family member was then detected using an anti-rabbit HRP-conjugated IgG. Color was developed with hydrogen peroxide/tetramethylbenzidine chromogenic substrate, and intensity of the developed color was proportional to the quantity of RelA and RelB in each sample. The sample values were normalized against total cell protein determined using a protein assay kit (Pierce, Rockford, IL).

**cAMP assay**

A cAMP assay was performed as described previously (53). After human iDCs (2 × 10⁶/well) had been cultured in a 96-well plate for 12 h, the cells were incubated with PKCI for 1 h. The cells were then washed twice with Krebs-Ringer HEPES (KRH) buffer (1.24 mM NaCl, 5 mM KCl, 1.25 mM MgSO₄, 1.45 mM CaCl₂, 1.25 mM KH₂PO₄, 25 mM HEPES [pH 7.4], and 8 mM glucose), 90 μM KRH buffer containing 10 μM forskolin (Sigma-Aldrich) with or without PKCI was added, and then the cells were stimulated with TNF-α, IL-1β, and/or PGE₂ for 1 h. The cells were lysed by adding 200 μl lysis buffer 1A (a component of the cAMP enzyme immunoassay system; Amersham Biosciences), and 100 μl cell lysate was used for measurement of cAMP produced during incubation with the kit, as recommended by the manufacturer.

**Chemotaxis assay**

Chemotaxis assays for iDCs, mDCs, and PKCI-treated DCs were performed in polycarbonate membrane, 6.5-mm-diameter, 5-μm-pore size Transwell cell culture chambers (Costar, Cambridge, MA), as described previously (54). Aliquots (100 μl) of cells (5 × 10⁶/ml) suspended in RPMI 1640/0.5% BSA were added to the upper chambers. The CCL19 was added to the lower chambers at a concentration of 1 μg/ml. The cells were allowed to migrate for 2 h at 37°C in a 5% CO₂ incubator, after which the filters were fixed with 1% glutaraldehyde in PBS for 30 min and stained with 0.5% toluidine blue overnight. Cell migration was quantified by counting the cells in each lower chamber and cells adhering to the bottom of the polycarbonate filter.

**PKC kinase activity**

iDCs, mDCs, and PKCI-treated DCs (1 × 10⁶) at various PKCI concentrations were lysed with lysis buffer (20 mM MOPS, 50 mM β-glycerophosphate, 50 mM sodium fluoride, 1 mM sodium orthovanadate, 5 mM EGTA, 2 mM EDTA, 1% Nonidet P-40, 1 mM DTT, 1 mM benzamidine, 1 mM PMSF, and 10 μg/ml leupeptin and aprotonin). After centrifugation, each supernatant was analyzed for PKC activity using a PKC kinase activity kit (Enzo Life Sciences), in accordance with the manufacturer’s protocol.

**Mice**

Female BALB/c (H-2ᵇ) and C57BL/6 (H-2ᵇ) mice were purchased from Clea Japan (Tokyo, Japan). The graft-versus-host disease (GVHD) experiment was approved by the Aimal Ethics Committee, Ehime University. The mice used for the GVHD experiment ranged from 10 and 14 wk of age. They were housed in sterilized microisolator cages after bone marrow transplantation (BMT) and received filtered water and autoclaved chow or autoclaved drinking water.

**Generation of mouse DCs**

Mouse iDCs, mDCs, and PKCI-treated DCs were generated from BM cells as described by Chorny et al. (55). Briefly, BM cells (2 × 10⁶) obtained from BALB/c mice were incubated in RPMI 1640 medium with 10% FBS and 20 ng/ml rmGM-CSF for 6 d. Then, nonadherent cells (iDCs) were collected (routinely comprising 80–90% CD11c⁺ cells) and stimulated with LPS (1 μg/ml) or a maturation mixture containing rmTNF-α (50 ng/ml), rmIL-1β (10 ng/ml), and PGE₂ (200 ng/ml) for 48 h to induce maturation. PKCI-treated DCs were generated by culturing iDCs with a maturation mixture in the presence of Bis I (10 μM) for 48 h.

**BMT and GVHD induction**

BMT and GVHD induction were performed as described previously (54) and by the method of Chorny et al. (55). Briefly, recipients (BALB/c) received 10 Gy total body irradiation, split into two doses separated by 12 h to minimize gastrointestinal toxicity. Donor BM (C57BL/6) was re-allowed aseptically from femurs and depleted of T cells with anti-mouse Thy 1.2 mAb (Acris Antibodies, San Diego, CA) and Low-Tox-M rabbit complement (Cedarlane Laboratories, Hornby, ON, Canada). Splenic T cells were prepared by purification over nylon wool for GVHD induction. T cell–depleted BM cells (1.5 × 10⁶) plus 2 × 10⁶ splenic T cells from allogeneic (C57BL/6) donors were resuspended in 0.2 ml PBS and injected i.v. into recipients (BALB/c) on day 0 for induction of acute GVHD. Recipients received a single i.v. injection of host-matched iDCs, mDCs, or PKCI-treated DCs (BALB/c, 2 × 10⁶) 2 d after transplantation.

Recipients were monitored daily from the day of transplantation until they died naturally of GVHD.

**Statistical analysis**

Data are expressed as means ± SD. One-way ANOVA was used to compare multiple groups. When the ANOVA indicated significance, the Tukey-Kramer honestly significant difference test was applied for multiple comparisons. Differences in survival of treatment groups in the GVHD experiment were analyzed using Kaplan-Meier and log-rank tests. A p value of <0.05 was considered to indicate statistical significance.

**Results**

**Characterization of PKCI-treated human DCs**

PKCI-treated human DCs were prepared by culturing iDCs with a maturation mixture (TNF-α, rHL-1β, and PGE₂) for 48 h in the presence of a conventional PKCIs such as Bis I, Go6983, or Ro32-0432. In the preliminary experiment, the optimal concentration of three PKCIs compounds, Bis I, Go66983, and Ro 32-0432, was decided from downregulation of costimulatory molecules (CD40, CD80, and CD86), viability, and inhibition of PKC activity. Among the concentrations of 1, 10, and 30 μM, 10 μM was the optimal one for all 3 PKCIs, resulting in strong downregulation of costimulatory molecules, a viability of >95%, and inhibition of PKC activity that reached a plateau (Supplemental Fig. 1). Therefore, we used 10 μM for all PKCIs in this study.

First, we compared the surface phenotypes of iDCs, mDCs, and PKCI-treated DCs. As shown in Fig. 1A, iDCs showed moderate expression of CD1a and CD83. After inducing maturation with a maturation mixture (iDCs), the expression levels of CD40, CD80, CD86, and MHC class II, and extremely low levels of CD1a and CD83. After inducing maturation with a maturation mixture (mDCs), the expression levels of CD40, CD80, CD86, MHC class I, and MHC class II were increased significantly compared with those of iDCs. DCs treated with PKCI such as Bis I, Go6983, and Ro32-0432 showed significantly decreased levels of expression of CD40, CD80, CD86, and MHC class I, but not CD1a, CD11c, and MHC class II, in comparison with mDCs. CD14, a monocyte marker, was expressed slightly on iDCs but not on mDCs or PKCI-treated DCs (Fig. 1B).

Because migration of DCs to the T cell zone of secondary lymphoid organs is guided by the homing chemokines CCL19 and CCL21 (56), we examined the expression of CCR7, their receptor
on iDCs, mDCs, and PKCI-treated DCs. As shown in Fig. 1A, barely any expression of CCR7 was evident on iDCs. In contrast, upregulation of CCR7 expression was recognized on mDCs and PKCI-treated DCs, although the expression level of CCR7 on PKCI-treated DCs was lower than that on mDCs. Functionally, the capacity of PKCI-treated DCs to migrate toward CCL19 was ~70–80% of that of mDCs (no significant difference versus mDCs), whereas iDCs showed little or no migration (Fig. 1C).

Next, we examined cytokine production of iDCs, mDCs, and PKCI-treated DCs. Cytokines in the 96-h culture supernatants of iDCs, mDCs, and PKCI-treated DCs were measured. As shown in Fig. 2A, after stimulation with a maturation mixture, iDCs matured into cells capable of producing IL-12, but with reduced production of IL-10 and TGF-β. In contrast, PKCI-treated DCs produced a 15- to 20-fold higher concentration of IL-10 and a 3- to 5-fold higher concentration of TGF-β than did mDCs. However, the production of IL-12 by PKCI-treated DCs was much lower than that by mDCs.

We then examined the ability of iDCs, mDCs, and PKCI-treated DCs to induce the proliferation of allogeneic CD4⁺ T cells. As shown in Fig. 2B, iDCs reduced the proliferation of allogeneic CD4⁺ T cells significantly by 25 ± 10 and 48 ± 10% at a ratio of 1:10 and 2:10 (DC/T cell), respectively, in comparison with mDCs. Furthermore, the degree of PKCI-treated DC-induced T cell hyporesponsiveness was higher than that of iDCs (a ratio of 1:10 and 2:10 [DC/T cell]; Bis I, 69 ± 4 and 89 ± 10%; G66983, 77 ± 8 and 83 ± 8%; and Ro32-0432, 79 ± 5 and 78 ± 10%; p < 0.01). PKCI-treated DCs retained their hyporesponsiveness for at least 1 wk. Therefore, this hyporesponsiveness of PKCI-treated DCs was mainly due to downregulation of costimulatory molecules CD80, CD83, and CD86 and high production of IL-10 and TGF-β.

We assessed the phagocytic capacity of PKCI-treated DCs using FITC/latex beads/rabbit IgG. As shown in Fig. 2C, DCs lost their phagocytic ability during maturation. However, PKCI-treated DCs retained their phagocytic ability, similar to iDCs.

For clinical grade DC preparation, iDCs, mDCs, and PKCI-treated DCs were generated using serum-free X-VIVO 15 medium instead of RPMI 1640 medium plus 10% FBS. The iDCs, mDCs, and PKCI-treated DCs prepared in X-VIVO 15 medium had properties similar to those of cells prepared in RPMI 1640 medium with 10% FBS (Supplemental Figs. 2, 3).
PKCI-treated human DCs induce IL-10–producing T cells and functional regulatory T cells

Because PKCI-treated DCs produced IL-10 and TGF-β, we examined whether they would induce IL-10–producing T cells and Foxp3+CD4+CD25+ T cells. Naïve CD4+CD45RA+ T cells were cocultured with iDCs, mDCs, or PKCI-treated DCs for 5 d, and then intracellular IL-10+CD4+ T cells and Foxp3+CD4+CD25+ T cells were analyzed by flow cytometry. As shown in Fig. 3A, PKCI-treated DCs generated >4-fold the number of both IL-10+ CD4+ T cells and Foxp3+CD4+CD25+ T cells than did those induced by mDCs. CD4+ T cells cocultured with PKCI-treated DCs suppressed the proliferation of effector Th1 cells most strongly (Bis I, 48 ± 4%; G6o983, 41 ± 14%; and Ro32-0432, 36 ± 6%), and CD4+ T cells cocultured with iDCs did so to a lesser degree (26 ± 7%), compared with control (Fig. 3B).

Next, we addressed the question of whether this suppression was due to direct cellular contact and/or soluble factors. In the coculture experiments, addition of saturating amounts of anti–IL-10 or anti–TGF-β mAbs (no Abs-C), with only anti–IL-10 mAb, and with only anti–TGF-β mAb: 48 ± 4, 32 ± 8, and 30 ± 5%, respectively). Addition of both anti–IL-10 and anti–TGF-β mAbs to the cocultures reversed the inhibition more markedly, although the inhibition still persisted (21 ± 9%). When effector Th1 cells and CD4+ T cells cocultured with Bis I–treated DCs were separated in the Transwell experiments, the proliferation of effector Th1 cells was still inhibited to a lesser degree (21 ± 5%). Addition of both anti–IL-10 and anti–TGF-β mAbs in the Transwell experiments reversed the inhibitory effect almost completely (Fig. 3C). Similar results were obtained from the experiments using CD4+ T cells cocultured with DCs treated with two other PKCIs, G6o983 and Ro32-0432 (data not shown). These findings indicated that both direct contact and soluble factors mediate the inhibitory effect of CD4+ T cells cocultured with PKCI-treated DCs. Additionally, it was shown that the immunosuppressive function of PKCI-treated DCs was elicited by generating IL-10–producing T cells and functional Foxp3+ regulatory T cells from naïve CD4+ T cells.

Tolerogenic properties of PKCI-treated human DCs are highly stable

A potential risk of ex vivo–generated tDCs is that they may switch to an activating phenotype after encountering danger signals in vivo. To address this issue, the stability of PKCI-treated DCs was examined by stimulation with proinflammatory mediators such as CD80, CD83, and CD86, resulting in loss of T cell hyporesponsiveness upon addition of proinflammatory mediators. These findings indicated that PKCI-treated DCs are refractory to

FIGURE 3. PKCI-treated human DCs induce IL-10–producing T cells and Foxp3+ regulatory T cells. (A) Comparison of IL-10–producing T cells and Foxp3+ T cells induced by iDCs, mDCs, and PKCI-treated DCs. Naïve CD4+CD45RA+ T cells (5 × 105) were cultured with 5 × 105 MMC-treated allogeneic iDCs, mDCs, or PKCI-treated DCs for 5 d. Then, the intracellular expression of IL-10 and Foxp3 in CD4+ T cells and CD4+CD25+ T cells was analyzed by flow cytometry. Numbers in the corners indicate the percentage of positive cells. (B) CD4+ T cells cocultured with PKCI-treated DCs suppressed the proliferation of responder T cells. Naïve CD4+CD45RA+ T cells (5 × 105) were cultured with 5 × 105 MMC-treated allogeneic iDCs, mDCs, or PKCI-treated DCs for 5 d. Then, CD4+ T cells were purified from each coculture. Purified CD4+ T cells (1 × 105) were incubated with syngeneic Th1 cells (responder T cells, 1 × 105) in the presence of MMC-treated allogeneic mDCs (1 × 105) for 4 d. The proliferation of responder T cells was determined using [3H]thymidine incorporation. The proliferation of responder T cells with mDCs (control) was 42,669 ± 2,571 cpm. The proliferation of responder T cells with mDCs by adding CD4+ T cells cocultured with mDCs, iDCs, or PKCI-treated DCs was expressed relative to that of control. Representative data from three experiments are shown. *p < 0.01 versus control. (C) Both direct cellular contact and soluble factors mediate the inhibitory effect of CD4+ T cells cocultured with Bis I–treated DCs. Coculture experiments were also performed without anti–IL-10 and anti–TGF-β mAbs (no Abs-C) or in the presence of saturating blocking anti–IL-10 (40 μg/ml) and/or anti–TGF–β (40 μg/ml) mAbs. The addition of isotype control (IgG) did not alter the effect of CD4+ T cells cocultured with Bis I–treated DCs on the proliferation of responder T cells. Coculture experiments were performed using 24-well plates. Responder T cells (5 × 104) together with MMC-treated allogeneic mDCs (5 × 104) were placed in the bottom wells. CD4+ T cells (5 × 105) generated in the presence of Bis I–treated DCs were placed in the upper wells. The cultures were performed no Abs-C or in the presence of anti–IL-10 and/or anti–TGF–β mAbs for 4 d. Then, the basket was removed and the proliferation of the responder T cells was measured using [3H]thymidine incorporation. The control value in coculture experiment was 42,669 ± 2,571 cpm. The control value in the Transwell experiment was 25,940 ± 1,786 cpm. The proliferation of responder T cells in each condition was expressed relative to that of control. Representative data from three experiments are shown. *p < 0.05 versus no Abs-C, **p < 0.01 versus no Abs-C, 1p < 0.01 versus control (Transwell).
stimulation with proinflammatory mediators and that their tolerogenic properties are highly stable.

**PKC inhibitor (PKCI) reduces maturation stimuli–induced increases in NF-κB family members in DCs**

It has been reported that a crucial pathway for the maturation of DCs upon exposure to proinflammatory stimuli involves the transcription factor NF-κB (57–60). iDCs pretreated with DMSO (control) or PKCIs were exposed to TNF-α, IL-1β, IL-6, and PGE2 and the resulting effects on the nuclear NF-κB family members, RelA (p65) and RelB, were measured. As shown in Fig. 6, TNF-α stimulation increased the expression of RelA and RelB 2.8- and 2.2-fold, whereas pretreatment with PKCIs reduced their expression 1.5- to 2.0- and 1.1- to 1.5-fold, respectively, relative to that of unstimulated iDCs. Alternatively, expression of RelA and RelB was increased 1.5- and 1.3-fold, respectively, upon stimulation with IL-1β and declined to the baseline level upon treatment with PKCI. In contrast, expression of RelA and RelB did not increase significantly upon stimulation with PGE2. Upon stimulation with a maturation mixture containing TNF-α, IL-1β, and PGE2, expression of RelA and RelB was increased synergistically 3.5- and 3.0-fold and was reduced 1.5- to 1.8- and 1.3- to 1.5-fold by PKCI treatment, respectively. This also indicated that expression of RelA and RelB in PKCI-treated DCs is slightly higher than that in iDCs. Additionally, these results demonstrate that conventional PKC is required for both the canonical (RelA) and noncanonical (RelB) pathways of NF-κB activation, and that conventional PKC plays a critical role in DC maturation.

**PKC inhibitor increases intracellular cAMP in DCs upon stimulation with a maturation mixture**

Intracellular cAMP/CREB has been reported to play a critical role in transcriptional regulation of IL-10 in monocytes and DCs (61, 62). iDCs pretreated with DMSO (control) or PKCI were stimulated with TNF-α, IL-1β, IL-6, and TNF-α each at 10 ng/ml or LPS (0.1 μg/ml). After 1 and 5 d, DCs were analyzed for cell surface phenotype by flow cytometry. Values are the means ± SD of mean fluorescence intensity (MFI) of each phenotype on DCs from four experiments (n = 2 or 3 samples/experiment). *p < 0.05 versus mDCs, †p < 0.01 versus mDCs.

**FIGURE 4.** Phenotypic stability of PKCI-treated human DCs upon proinflammatory stimulation. After removal of a maturation mixture and PKCIs, iDCs, mDCs, or PKCI-treated DCs were cultured in the absence or presence of a mixture of proinflammatory cytokines containing IFN-γ (1000 U/ml), IL-1β, IL-6, and TNF-α each at 10 ng/ml, or LPS (0.1 μg/ml). After 1 and 5 d, DCs were analyzed for cell surface phenotype by flow cytometry. Values are the means ± SD of mean fluorescence intensity (MFI) of each phenotype on DCs from four experiments (n = 2 or 3 samples/experiment). *p < 0.05 versus mDCs, †p < 0.01 versus mDCs.

**FIGURE 5.** Stability of PKCI-treated human DCs upon proinflammatory stimulation. (A) Production of IL-10 and TGF-β of PKCI-treated DCs upon proinflammatory stimulation. After removal of a maturation mixture and PKCIs, iDCs, mDCs, or PKCI-treated DCs were cultured in RPMI 1640 medium supplemented with 10% FBS or in X-VIVO medium only for assay of TGF-β in the absence or presence of a mixture of proinflammatory cytokines or LPS. After 5 d, production of IL-10 and TGF-β in the culture supernatants was determined by ELISA. Values are the means ± SD of four experiments (n = 2 or 3 samples/experiment). *p < 0.01. (B) Suppressive properties of PKCI-treated DCs upon proinflammatory stimulation. DCs were washed after stimulation with a mixture of proinflammatory cytokines or LPS and recultured in RPMI 1640 medium supplemented with 10% FBS for a further 24 h. Then, an MLR was performed as described in the in vitro T cell proliferation assay section. T cell proliferation by iDCs and PKCI-treated DCs was expressed relative to that by mDCs. Values are the means ± SD (n = 6 samples). Representative data from four experiments are shown. *p < 0.05 versus mDCs, **p < 0.01 versus mDCs.
relative to DCs stimulated with PGE2 alone. Upon stimulation with a maturation mixture, intracellular cAMP production was reduced significantly compared with that of DCs stimulated with PGE2 alone. However, upon treatment with PKCI, production of cAMP and IL-10 recovered to a level that was the same as, or higher than, that of DCs treated with PGE2 and PKCI. These findings indicated that PGE2 acts antagonistically to TNF-\(\alpha\) or IL-1\(\beta\) in terms of cAMP production by DCs, and that this antagonistic effect is strongly attenuated by PKCI. Additionally, upregulation of intracellular cAMP in DCs does not, in itself, induce IL-10 production, and the presence of PKCI is additionally necessary for this effect.

Characterization of PKCI-treated mouse DCs

First, to examine the effect of PKCI-treated DCs in vivo in a murine model of acute GVHD, we tested whether it would be possible to generate PKCI-treated mouse DCs using a maturation mixture (TNF-\(\alpha\), IL-1\(\beta\), and PGE2). Mouse iDCs were generated by culturing BM cells with GM-CSF for 6 d and then matured with LPS or a maturation mixture for a further 48 h. LPS was used as a maturation control. As shown in Fig. 8A, as well as LPS-induced mDCs, mDCs induced by the maturation mixture showed high expression levels of CD40, CD80, CD83, CD86, MHC class I (H-2K\(d\)), MHC class II (I-A/I-E), and CCR7. Functionally, mDCs induced by the maturation mixture showed migration activity toward CCL19 (Fig. 8B), high IL-12 production (Fig. 9A), sufficient proliferation of allogeneic T cells (Fig. 9B), and low phagocytic capacity (Fig. 9C), similar to LPS-induced mDCs.

PKCI-treated DCs were generated by culturing iDCs with a maturation mixture in the presence of Bis I for 48 h. Bis I–treated DCs showed markedly and significantly decreased levels of expression of CD40, CD80, CD83, CD86, and MHC class I, and slightly decreased levels of expression of MHC class II, in comparison with mDCs induced by the maturation mixture (Fig. 8A). Bis I–treated DCs showed CCR7 expression and migration activity toward CCL19, similar to mDCs (Fig. 8B). Bis I–treated DCs also had high production of IL-10 and TGF-\(\beta\), T cell hyporesponsiveness, and efficient phagocytic capacity, as was the case for human PKCI-treated DCs (Fig. 9A–9C). Thus, using a maturation mixture, we were able to generate mouse PKCI-treated DCs that had properties similar to those of human PKCI-treated DCs.

PKCI-treated DCs prevent acute GVHD

Next, we examined the therapeutic effect of PKCI-treated DCs on acute GVHD following allogeneic BMT. We transplanted lethally irradiated BALB/c recipients (H-2\(d\)) with either T cell–depleted BM cells or with T cell–depleted BM plus spleen T cells (GVHD induction) from C57BL/6 (H-2\(b\)) mice. Two days after BMT, we...
injected iDCs, mDCs, and Bis I–treated DCs obtained from BALB/c mice. As shown in Fig. 10, mice given only T cell–depleted BM cells appeared healthy, and 100% of the animals survived for at least 50 d. Mice that received BM cells plus spleen T cells developed severe signs of GVHD, including weight loss, reduced mobility, hunched posture, diarrhea, and ruffled fur, and they died within 30 d. There were no significant differences of GVHD lethality among untreated, iDC-treated, and mDC-treated mice. In contrast, administration of Bis I–treated DCs protected the mice from lethal GVHD, and 80% of them survived for 50 d (p, 0.01).

From these findings, we confirmed that PKCI-treated DCs were more efficient than iDCs and mDCs for preventing GVHD in vivo.

Discussion
In this study, we demonstrated that human tolerogenic DCs were generated by adding a conventional PKC inhibitor during the maturation process after iDC differentiation, but not during the process of differentiation from progenitors to iDCs. The PKCI-treated DCs had a semimature phenotype with high production of IL-10, and they induced IL-10–producing T cells and functional regulatory T cells from naive CD4+ T cells efficiently, thus eliciting a strong immunosuppressive function. These cells expressed CCR7 and had sufficient capacity to migrate toward CCR7 ligands, especially secondary lymphoid organs. Additionally, PKCI-treated DCs were highly stable upon exposure to inflammatory stimuli. Moreover, PKCI-treated mouse DCs that had properties similar to PKCI-treated human DCs prevented acute GVHD in a murine model.

DCs are a heterogeneous group of APCs involved in the initiation of both adaptive immunity and tolerance induction. Generally, iDCs are characterized by an immature or semimature phenotype, with low expression of costimulatory molecules, low production of proinflammatory cytokines, including IL-12 and TNF-α, and high production of anti-inflammatory cytokines such as IL-10 and TGF-β in comparison with mDCs (2–6). Partial maturation resulting in
intermediate expression of MHC and costimulatory molecules, but little or no production of proinflammatory cytokines, has been termed semimaturiation (2). The mechanisms by which tDCs induce tolerance include 1) deletion of T cells, 2) induction of T cell hyporesponsiveness, 3) deviation of the T cell cytokine profile, and 4) induction of regulatory T cells (7). The anti-inflammatory cytokines IL-10 and TGF-β, neuropeptides including vasoactive intestinal peptide and a melanocyte–stimulating hormone, and several pharmacological agents such as Dexa, Rapa, and Vit D₃ have been reported to promote or restore Ag-specific tolerance in experimental animal models (8–17, 21, 27, 29, 31, 35).

On the basis of these data, clinical trials of ex vivo–generated tDCs are currently being considered. Three functional characteristics are required for clinically applicable tDCs (37–39). First, ex vivo–generated tDCs need to have the capacity for CCR7-dependent migration toward secondary lymphoid organs for induction of suppressive responses in naive T cells. Second, in these organs, tDCs should be capable of inducing the right type of immunosuppressive effector mechanism, especially efficient induction of functional regulatory T cells. Third, it is essential to establish whether ex vivo–generated tDCs are stable and maintain their tolerogenic properties when encountering proinflammatory signals in vivo. Two groups have compared migratory capacity, stability, and functional properties among clinical-grade tDCs generated with Vit D₃, Dexa, Rapa, IL-10, or TGF-β (38, 39). Vit D₃-tDCs, Dexa-tDCs, and IL-10-tDCs had very low CCR7 expression, leading to very poor migration to CCL19 and CCL21, whereas TGF-β-tDCs and Rapa-tDCs had moderate and high CCR7 expression, respectively. Vit D₃-tDCs, Dexa-tDCs, IL-10-tDCs, and TGF-β-tDCs showed an immature or semimature phenotype, whereas the phenotype of Rapa-tDCs resembled that of mDCs. IL-10 production by Vit D₃-tDCs, Dexa-tDCs, and IL-10-tDCs was moderate, whereas that of TGF-β-tDCs and Rapa-tDCs was slight or undetectable. Functionally, IL-10-tDCs and Vit D₃-tDCs suppressed T cell activation, whereas TGF-β-tDCs hardly did so. Two groups have obtained conflicting data regarding the suppressive activity of Dexe-tDCs and Rapa-tDCs. All five tDCs maintained stable tolerogenic properties under proinflammatory conditions. Overall, IL-10-tDCs appear to possess the most powerful tolerogenic characteristics with high IL-10 production and are the best inducers of regulatory T cells, but they show the poorest migration toward CCR7 ligands. In the present study, PKCI-treated tDCs showed moderate expression of CCR7 and had sufficient capacity to migrate toward CCR7 ligands. PKCI-treated tDCs strongly suppressed T cell activation by generating IL-10–producing T cells and functional Foxp₃⁺ regulatory T cells from naive CD4⁺ T cells. Additionally, their tolerogenicity was highly stable in the presence of proinflammatory stimuli. PKCI-treated human DCs prepared in serum-free X-VIVO 15 medium had properties similar to those of cells prepared in RPMI 1640 medium with 10% FBS. From these findings, PKCI-treated human DCs satisfy the above three requirements for clinical application. Moreover, we successfully generated PKCI-treated mouse DCs that had similar properties to PKCI-treated human DCs and demonstrated that PKCI-treated DCs prevented acute GVHD in vivo. Taken together, these findings suggest that PKCI-treated DCs could be clinical-grade tDCs.

Members of the PKC family are serine-threonine protein kinases that are divided into three subfamilies, conventional, novel, and atypical PKCs, depending on their responsiveness to calcium and DAG stimulation (40). PKCs are activated by PMA, an activator of conventional PKC-derived progenitors such as CD34⁺ hematopoietic progenitors and CD14⁺ monocytes, to undergo DC differentiation (41, 44, 45). GM-CSF, IL-4, and TNF-α each activates PKC or PKCB signaling, and addition of these cytokines leads to differentiation of progenitors to DCs (63–65). Other groups have reported that PKCβ₁ and PKCβ₂ play an essential role in DC differentiation and that PKCα signaling is a key driver of monocyte-to-macrophage differentiation (44, 45). Our present study showed that stable, tolerogenic DCs with a semimature phenotype were generated by addition of conventional PKCI during the maturation process. To examine which of the conventional PKCIs affected DC tolerogenicity, we compared the downregulation of costimulatory molecules (CD40, CD80, and CD86) using the specific PKCIs G6976, a PKCα-specific inhibitor, CGP53353 (a PKCβ₁-specific inhibitor), and HBDE (a PKCγ-specific inhibitor). Ins 1–, G6976, CGP53353, and HBDE-treated DCs all showed downregulation of costimulatory molecules. Of these, Bis I–treated DCs showed the strongest downregulation, indicating that PKCα, β, and γ are all involved in the expression of costimulatory molecules on DCs (Supplemental Fig. 4). Taken together, these findings suggest that the conventional PKC subfamily plays a critical role not only in DC differentiation but also maturation in response to exogenous stimuli.

Studies involving culture with cytokines including GM-CSF, IL-4, and TNF-α have indicated that PKCβ₂ plays an essential role in DC differentiation through noncanonical/RelB-mediated NF-κB signaling (44). PKCε is involved in LPS-induced IL-12 production by DCs through IkB-α degradation and subsequent NF-κB activation (42). These findings suggest that NF-κB signaling may be involved in the mechanism by which conventional PKCI induces tDCs. NF-κB is required for proper development and function of DCs, and its activation is upregulated early during differentiation from precursors such as CD34⁺ hematopoietic progenitors and monocytes. Additionally, when matured in the presence of TNF-α alone or LPS, NF-κB activation plays an essential role in IL-12 production and expression of MHC class II and costimulatory.

FIGURE 10. PKCI-treated DCs prevent acute GVHD. Lethally irradiated BALB/c recipients (H-2b) were transplanted with either T cell-depleted BM controls (●) or with T cell-depleted BM plus spleen T cells (GVHD induction) from C57BL/6 (H-2b) mice. Recipients received a single i.v. injection of medium (○), host-matched tDCs (□), mDCs (▲), or Bis I–treated DCs (□) 2 d after GVHD induction. Recipients were monitored daily from the day of transplantation until they died naturally of GVHD. Differences in survival of treatment groups in the GVHD experiment were analyzed using Kaplan–Meier and log-rank test. *p < 0.01 versus any other groups. Similar results were obtained from two independent experiments.
molecules such as CD80, CD86, and CD40 in DCs (58). When matured with a mixture of TNF-α, IL-1β, and PGE2, DCs treated with NF-κB inhibitors BAY 11-7082 or aspirin have been shown to express high levels of MHC class II and CD86 but markedly reduced expression of CD40 and generated anergy and regulatory T cell activity (66). NF-κB family members include RelA (p65), RelB, p52/p100, p50/p105, and c-Rel. The heterodimers RelA/p50 and RelB/p52 are required for activation of thecanonical and noncanonical pathways, respectively. Inhibitory IkBs can be degraded by the proteasome to allow nuclear translocation of NF-κB and activate transcription in the canonical pathway (60). Among the NF-κB family members, RelB is the most crucial NF-κB subunit for expression of CD40, CD86, and MHC class II on DCs during maturation (57, 67, 68). RelB-deficient mice show impaired development of regulatory T cells with inhibited RelB function have been shown to lack typical costimulatory molecules (68). In the present study, DC maturation induced by TNF-α or IL-1β was NF-κB-dependent, whereas that induced by PGE2 was NF-κB-independent. Moreover, conventional PKCs inhibited NF-κB activation of both the canonical and noncanonical pathways of DC maturation, leading to suppression of costimulatory molecule expression and IL-12 production and resulting in a semimature state.

The cAMP/CREB-dependent pathway plays a critical role in IL-10 production by DCs (61, 62). However, an increase of the intracellular level of cAMP by PGE2 reportedly has its own limited effect on IL-10 production (62), and this is reflected in our present data. Additional stimulation is necessary to elicit a synergistic increase of IL-10 production with cAMP. Competition between NF-κB and cAMP/CREB for IL-10 gene transcription has been reported (69, 70), and NF-κB inhibition is associated with an increase of IL-10 production (62, 71, 72). In the present study, upon stimulation with PGE2 alone or a maturation mixture including TNF-α, IL-1β, and PGE2, the amount of intracellular cAMP in PKC-untreated DCs increased only 2- to 3-fold relative to that in PKC-untreated DCs. However, IL-10 production in PKC-treated DCs showed a much greater increase (5-fold) than that in PKC-untreated DCs. This augmentation of IL-10 production may be a synergistic effect of increased cAMP and NF-κB inhibition elicited by conventional PKCs.

tDCs are a promising therapeutic tool for specific cellular induction of immunological tolerance in the context of transplantation and autoimmunity. As there will be a demand for clinical-grade tDCs suited for this purpose.

Disclosures

The authors have no financial conflicts of interest.

References


SUPPLEMENTARY FIGURE LEGEND

Supplementary Fig 1. Optimal concentration of PKCI compounds. A. Downregulation of costimulatory molecules on human DCs treated with three PKCI compounds. PKCI-treated DCs were prepared by culturing iDCs with a maturation cocktail in the presence of 3 PKCI compounds, Bis I, Gö6983, or Ro32-0432 at various concentrations for 48 h. Surface phenotype of costimulatory molecules (CD40, CD80, and CD86) on each DCs was analyzed by flow cytometry. Values are the mean and SD of mean fluorescence intensity (MFI) of each phenotype on DCs from three experiments. *p<0.01. NS, not significant. B. Viability of human DCs treated with three PKCI compounds. Viability of each DCs was examined with trypan-blue staining. *p<0.01. NS, not significant. C. PKC kinase activity of human DCs treated with three PKCI compounds. iDCs, mDCs and PKCI-treated DCs (1 x 10^6) at various PKCI concentrations were lysed with lysis buffer and each supernatant was analyzed for PKC kinase activity. PKC kinase activity of iDCs was used as a control. Each PKC kinase activity was calculated by subtracting the PKC kinase activity of iDCs. The PKC kinase activity of PKCI-treated DCs was expressed relative to that of mDCs. *p<0.01. NS, not significant.

Supplementary Fig 2. Surface phenotype and chemotaxis of human iDCs, mDCs, and PKCI-treated DCs prepared in serum-free X-VIVO15 medium. A. Surface phenotype on iDCs, mDCs, and PKCI-treated DCs. B. CD14 expression on monocytes, iDCs, mDCs, and PKCI-treated DCs. PKCI-treated DCs were generated using X-VIVO15 medium by culturing iDCs with a maturation cocktail in the presence of PKCIs such as Bis I (10 μM), Gö6983 (10 μM), or Ro32-0432 (10 μM) for 48 h.
Surface phenotype on each DCs was analyzed by flow cytometry. Numbers in the corners indicate the mean fluorescence intensity (MFI) of expression level of each surface marker. Representative data from three experiments are shown. C. **Chemotactic activity of iDCs, mDCs, and PKCI-treated DCs to CCL19.** Each assay was performed in triplicate. *p<0.01. NS, not significant.

Supplementary Fig 3. **Characterization of human iDCs, mDCs, and PKCI-treated DCs prepared in serum-free X-VIVO15 medium.** A. **Cytokine production from iDCs, mDCs, and PKCI-treated DCs.** iDCs, mDCs, and PKCI-treated DCs were prepared in serum-free X-VIVO15 medium as described in Materials and Methods. These DCs (1 x 10⁶/mL) were cultured in X-VIVO15 medium for assay of IL-6, IL-10, IL-12, IFN-γ, and TGF-β. The cell supernatants were harvested after 96 h and assayed. Values are the mean and SD of three experiments. *p<0.01. NS, not significant. B. **Comparison of suppressive properties among iDCs, mDCs, and PKCI-treated DCs.** In vitro T cell proliferation assay was performed as described in Materials and Methods. T cell proliferation by iDCs and PKCI-treated DCs was expressed relative to that by mDCs. Values are the mean and SD. Representative data from three experiments are shown. *p<0.05. **p<0.01. NS, not significant. C. **Phagocytic ability of iDCs, mDCs, and PKCI-treated DCs.** DCs were incubated with FITC-latex beads-rabbit IgG for 24 h, and then analyzed by flow cytometry.

Supplementary Fig 4. **Comparison of downregulation of costimulatory molecules on DCs treated with the specific PKCIs.** PKCI-treated DCs were prepared by culturing iDCs with a maturation cocktail in the presence of the specific PKCIs,
Gö6976 (a PKC α- and β1-specific inhibitor), CGP53353 (a PKC βII-specific inhibitor), and HBDDE (a PKC α- and γ-specific inhibitor) at various concentrations for 48 h. Bis I was used at a concentration of 10 μM. Surface phenotype of costimulatory molecules (CD40, CD80, and CD86) on each DCs was analyzed by flow cytometry. Values are the mean and SD of mean fluorescence intensity (MFI) of each phenotype on DCs from three experiments. *p,0.05 vs mDCs. **p,0.01 vs mDCs.