autologous challenge virus SHIV-MN are unlikely to provide information that predicts efficacy in humans, the results presented here demonstrate that recombinant BCG vectors have the potential to deliver a more appropriate immunogen for desirable immune elicitations.

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Cross-Linking Cell Surface Chemokine Receptors Leads to Isolation, Activation, and Differentiation of Monocytes into Potent Dendritic Cells

FUMIKAZU NIMURA,*,† LI FENG ZHANG,* KAZU OKUMA,* REIKO TANAKA,* HAJIME SUNAKAWA,† NAOKI YAMAMOTO,‡ AND YUETSU TANAKA*,[1]

Department of Immunology and †Department of Oral and Maxillofacial Functional Rehabilitation, Graduate School of Medicine, University of the Ryukyus, Okinawa, Japan; and ‡AIDS Research Center, National Institute of Infectious Diseases, Tokyo, Japan

Monocytes express on the cell surface several kinds of chemokine receptors that facilitate chemotaxis followed by differentiation in target tissues. In the present study, we found that a large number of monocytes from peripheral blood mononuclear cells (PBMCs) tightly adhered to plastic cell culture plates precoated with a monoclonal antibody (mAb, clone T312) specific for human CCR5 but not an isotype control after overnight incubation. Soluble T312 did not induce such adhesion, indicating that cross-linking of CCR5 is required for the enhanced adhesion of monocytes. The adhesion was blocked by a PI3-K inhibitor and an anti-CD18 blocking mAb. Following the cross-linking of CCR5, monocytes synthesized high levels of M-CSF, RANTES, MIP-1α, and MIP-1β associated with a readily detectable downmodulation of CD14, CD4, CCR5, and CXCR4 expression. The T312-enriched monocytes differentiated into dendritic cells (DCs) in the presence of interleukin-4 alone. After maturation with β-interferon, the T312-induced DCs stimulated proliferation of allogeneic naïve $CD4^+$ T cells accompanied by the synthesis of high levels of γ-interferon *in vitro*. Furthermore, the T312-induced DCs were capable of stimulating antigen-specific human T- and B-cell immune responses in our hu-PBL-SCID mouse system. Finally, screening of other anti-chemokine receptor mAbs showed that select clones of mAbs against CXCR4 and CCR3 were also capable of facilitating enrichment of monocytes similar to T312. These results show that cross-linking of chemokine receptors on monocytes by appropriate mAbs leads to activation and differentiation of monocytes and that the method described herein provides an alternate simple strategy for adherence-based isolation of monocytes and generation of functional DCs. Exp Biol Med 231:431–443, 2006

Key words: dendritic cell; monocyte; chemokine receptor; human immunodeficiency virus (HIV)

Introduction

Peripheral blood monocytes are derived from bone marrow progenitor cells and are recruited to tissues, where they undergo differentiation into macrophages or myeloid dendritic cells (DCs) (1–4). Recruitment of monocytes from the recirculating pool into normal or inflamed tissues involves a series of cascading events, which include the generation of chemokine gradients and the expression of cell adhesion molecules and their cognate ligands (5). Ligation of the chemokine receptors activates β1- and β2-integrins (6), which in turn mediate adhesion of monocytes within tissues, where they differentiate into DCs. DCs are potent antigen-presenting cells (APCs) and have a central role in the activation and function of both innate and adaptive immune responses against infectious microorganisms (7). Dysfunction and potential loss of DCs have been associated with decreased antigen-specific T-cell responses and synthesis of lower levels of virus-suppressive Type-1 interferon (8–11).

Results from a number of studies have documented the enhanced potential of DCs to process and present antigen, and thus, DCs have been regarded as natural cellular "adjuvants." This functional attribute has led to clinical trials of DC-based immunotherapy not only in a number of animal tumor models and human malignancies (12) but also against a number of infectious disease agents. This view is

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[1] To whom correspondence should be addressed at Department of Immunology, Graduate School of Medicine, University of the Ryukyus, Uehara 207, Nishihara-cho, Nakagami-gun, Okinawa 903-0215, Japan. E-mail: yuetsu@s4.dion.ne.jp

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highlighted by the recent finding that immunization of human immunodeficiency virus Type 1 (HIV-1)–infected patients with autologous DCs sensitized with chemically inactivated autologous HIV-1 led to a marked sustained decrease in viral load (13, 14). Essentially similar data, in terms of decreasing viral loads, were obtained using the simian immunodeficiency virus (SIV)–infected nonhuman primate model of human AIDS following immunization with autologous SIV-pulsed DCs (15); the results of this study were reasoned to be due to enhancement of both T-cell and neutralizing antibody responses. This finding was further supported using the hu-PBL-SCID model, in which human peripheral blood mononuclear cell (PBMC)–engrafted mice, following immunization with inactivated HIV-1–pulsed human DCs, were shown to generate high levels of HIV-1–specific T-cell and B-cell immune responses sufficient to protect these animals against challenge with virulent HIV-1 isolates (16, 17).

Human DCs for such studies are generally derived from culturing enriched populations of monocytes *in vitro* in media supplemented with varying combinations of cytokines, depending on the nature of the studies to be performed. Evidence has been accumulating in support of the notion that there exist subsets of DCs that differ in their expression of cell surface markers, *in vivo* trafficking patterns, and cytokines synthesized, which influences the quality of the T-cell response that is induced by such DCs (18). Thus, *in vitro* culture of monocytes with recombinant human granulocyte/macrophage-colony stimulating factor (GM-CSF) and interleukin-4 (IL-4) leads to their differentiation into myeloid DCs, and *in vitro* culture of those cultured in media containing GM-CSF and IL-3 leads to their differentiation into lymphoid DCs (19). There also appears to be evidence that a common precursor progenitor cell exists that can give rise to either myeloid or plasmacytoid DC subsequent to Flt-3 ligation (20). In the *in vitro* culture of the monocytes, GM-CSF functions as a survival and differentiation factor, whereas IL-4 induces differentiation of DCs by blocking their differentiation into the macrophage lineage (21, 22).

To study the biology of these DCs and to use them for *in vivo* studies, a large number of monocytes need to be isolated from the peripheral blood. This is accomplished using a variety of techniques, such as elutriation centrifugation (23), use of antibody-conjugated immunobeads (21, 24), and the more simple method of adherence of these cells to plastic (5, 24). Although the purity of the preparations using such procedures varies, it is clear that the former two techniques are expensive, requiring unique instrumentation and/or clinically trained staff, and labor intensive. Although the adherence-to-plastic method is simple, the yield of monocytes in such preparations varies among donors, and the results are difficult to reproduce.

We have previously reported that cross-linking of CXCR4 by a monoclonal antibody (mAb) that recognizes the extracellular loop 3 (ECL-3) region of CXCR4 induces homologous adhesion of T cells and enhances HIV-1 infection (25). Since fresh monocytes express a variety of chemokine receptors on their cell surface (26), we hypothesized that cross-linking of these chemokine receptors by an immobilized mAb might induce monocyte adhesion onto culture plates, providing a simple alternative procedure for the enrichment of monocytes from PBMCs.

Indeed, herein we present data that show that cross-linking the chemokine receptors CCR5, CXCR4, and CCR3 on monocytes by appropriate mAbs enhances adhesion of monocytes to plastic plates and that these adherent monocytes can be induced to differentiate along the macrophage or myeloid DC lineages with the use of distinct recombinant cytokines. This procedure will provide a relatively efficient and a more practical alternative for the isolation and study of monocytes and DC lineages.

Materials and Methods

Animals. SCID mice lacking functional T, B, and natural killer (NK) cells and BALB/c–rag2$^{-/-}$ common gamma$^{-/-}$ mice (27) were used in the present study. These mice were kept in the specific-pathogen-free animal facility of the Laboratory Animal Center at the University of the Ryukyus. The protocols for the care and use of the hu-PBL-SCID mice were approved by the Committee on Animal Research of the University of the Ryukyus before initiation of the present study.

Reagents. RPMI-1640 medium was purchased from Sigma Chemical Co. (St. Louis, MO) and supplemented with 5% heat-inactivated fetal calf serum (FCS) (Sigma Chemical) (referred to as RPMI medium). Serum-free medium, AIM-V, was purchased from Life Technology (Grand Island, NY). Recombinant (r) human GM-CSF and IL-4 were produced in 293T cells transfected with pCMhGM-CSF and pCMhIL-4, respectively (RIKEN Gene Bank, Ibaraki, Japan), using the calcium phosphate method. The total protein and cytokine concentrations of the pooled culture supernatants were determined by enzyme-linked immunosorbent assay (ELISA) and a functional assay, as described previously (16). rIL-2 was provided by the U.S. National Institutes of Health (NIH) AIDS Research and Reference Reagent Program. rIFN-β was purchased from Torey (Tokyo, Japan). rIFN-γ and rM-CSF were purchased from Peprotec (London, UK). The PI3-K inhibitor LY294.002 LPS from *Escherichia coli*, OVA, keyhole limpet hemocyanin (KLH), and bovine serum albumin (BSA, fraction V) were all purchased from Sigma Chemical. Fluorescein isothiocyanate (FITC)-labeled *E. coli*, FITC-fibrinogen, FITC-gelatin, and FITC-collagen were purchased from Molecular Probes (Eugene, OR). Monocyte negative isolation kits were purchased from Dynal (Oslo, Norway). Cell proliferation kits were purchased from Roche Diagnostics (Mannheim, Germany). ELISA kits for human IL-1β, IL-4, IL-6, IL-10, IL-12 p70, tumor necrosis factor (TNF)-α, M-CSF, MIP-1α, MIP-1β, and RANTES were

purchased from Biosource (Camarillo, CA). ELISA kits for γ-interferon (IFN–γ) were purchased from R&D Systems, Inc. (Minneapolis, MN). Naïve $CD4^+$ T-cell isolation kit was purchased from MACS (Gladbach, Germany).

The mAbs produced in our laboratory included two rat anti-human CCR5 N-terminus clones (clone T312 immunoglobulin [Ig]G1, clone T227 IgG2b); three rat anti-human CXCR4 (clone A145 IgG1, clone A120 IgG2b, clone A80 IgG1) (25); rat anti-human T-cell leukemia virus (HTLV)-I (clone LAT-27, IgG2b) (28); rat anti-hepatitis C virus (HCV) (clone Mo-8, IgG2b) (29); rat anti-human OX40 (clone W4–54, IgG2b, Tanaka *et al.*, unpublished data), and mouse anti-human OX40L (clone 5A8, IgG1) (30). The other mAbs used included mouse IgG anti-human CD4, SIM-2, and SIM-4 (obtained from the U.S. NIH AIDS Research and Reference Reagent Program), the clones OKT-4, OKT-8, and 60-bca anti-CD14 (obtained from ATCC, Rockville, MD). These mAbs were purified from SCID mouse ascites fluids by Superdex G-200 gel filtration (Amersham Bioscience, Uppsala, Sweden). Commercially available mAbs used were mouse IgG anti-human CD4, CD11c, CD80, CD83, CD86, HLA-DR (Coulter Inc., Hialeah, FL), and mouse IgG anti-human CD11a, CD11b, CD14, CD18, CD29, CD51, and CD61 (BD Pharmingen, San Diego, CA). Additional anti-human chemokine receptor mAbs, such as anti-CCR1 (mouse IgG2b, Cat #MAB145), CCR2 (mouse IgG2b, Cat #MAB150), CCR3 (rat IgG2a, Cat #MAB155), CCR5 (mouse IgG2b, Cat #MAB180), and CCR8 (rat IgG2b, Cat #MAB1429), were purchased from R&D Systems, Inc. (Minneapolis, MN); and the mAbs against CXCR4 (clone 12G5, mouse IgG2a) and CCR5 (clone 2D7, mouse IgG2a) were purchased from BD Pharmingen.

Cultivation of Monocytes. PBMCs were isolated from heparinized (5 U/ml) blood of normal healthy donors by a standard density gradient centrifugation at 400 g using lymphocyte separation medium (Sigma Chemical) for 15 mins at room temperature. The cells at the interface were collected and washed three times in cold phosphate-buffered saline (PBS) containing 0.1% BSA (BSA-PBS). PBMCs were resuspended in RPMI medium or serum-free AIM-V medium at 5×10^6 cells/ml. Then, 1 ml of the cell suspension was dispensed into individual wells of 12-well plates (BD Pharmingen), which were precoated with various mAbs (5 μg/ml) for 1 hr at 37°C. PBMCs were allowed to adhere 2 hrs or overnight at 37°C in a 5% CO_2 humidified incubator. Nonadherent cells were removed by gentle washing three times in BSA-PBS. The remaining adherent cells were then cultured in RPMI medium. For some experiments, monocytes were purified using a monocyte negative isolation kit and were used at $1–2 \times 10^5$ cells/ml. For the generation of macrophages, adherent or immunomagnetic bead-enriched $CD14^+$ monocytes were cultured in the presence of 20 ng/ml M-CSF for 6 days in a 5% CO_2 humidified incubator. For the generation of DCs, the media was supplemented with either GM-CSF (500 ng/ml) and IL-4 (200 ng/ml) or IL-4 (25 ng/ml) alone. Immature DCs were obtained after 5–6 days of culture. For maturation, the immature DCs were cultured in the presence of human β-interferon (IFN-β) (1000 U/ml) for an additional day. For sensitization with antigens, the immature DCs at Day 6 were cultured in the presence of either 100 μg/ml OVA or KLH for 1 day and then matured with IFN-β for an additional day. Viable cell number was assessed on an aliquot of such cells using staining with 0.1% eosin-Y.

Flow Cytometry. Phagocytosis and cell surface markers were determined using fluorescence activated cell sorter (FACS) Calibur and Cell Quest software (BD Pharmingen). Cell samples were Fc-blocked by incubation in media containing 2 mg/ml of human IgG in PBS containing 0.1% NaN_3 and 2% FCS (FACS buffer) on ice for 15 mins and were then stained with appropriate fluorescent dye–conjugated reagents on ice for 30 mins, according to the manufacturer's instructions. After washing with FACS buffer, cells were fixed in 1% paraformaldehyde-containing FACS buffer and analyzed. For quantitation of phagocytosis, sample cells (1×10^6) in 0.2 ml of RPMI medium were cultured in the presence of FITC-labeled *E. coli* at a cell to bacterium ratio of 1:10 to 1:100 for 1 hr at 37°C in a 5% CO_2 humidified incubator. After incubation, these cells were washed once in FACS buffer, fixed with 1% PFA, and then analyzed.

There were significant individual variations in the percentage of CCR5-positive cells in PBMCs. The percentages of T312-positive cells were as follows: 6%–14% of $CD14^+$ monocytes, 11%–43% of $CD3^+$ T cells, 6%–15% of $CD3^+$ $CD4^+$ T cells, and 5%–14% of $CD20^+$ B cells. Overnight incubation of PBMCs in medium alone led to a marked increase in the frequency of monocytes that expressed CCR5 (up to 70%), as detected using the T312 mAb. However, there was no detectable change of CCR5 expression in the other cell subsets (data not shown).

DC Functional Assays. Several assays were employed to determine whether the cell population that was being cultured belonged to the DC lineage. This included quantitation of the levels of IL-12 p70 and IL-10 produced in the culture supernatants of the potential DCs, as determined by ELISA; ability of the cells to induce allo-proliferation; and a unique *in vivo* assay. For the assessment of allo-proliferative potential, the mature or immature DCs to be tested were cultured at 1×10^5 cells/ml in RPMI medium in the presence of LPS (1 μg/ml) and IFN-γ (100 ng/ml) for 24 hrs. The ability to induce allogeneic stimulation was determined by co-culturing naïve $CD4^+$ T cells (1×10^5) with these potential DCs (0.5×10^4) in triplicate in a 96-well plate in a final volume of 0.2 ml in RPMI medium containing 20 U/ml IL-2 for 7 days. The level of proliferation of the $CD4^+$ T cells was assessed by the bromodeoxyuridine (BrdU)–incorporation ELISA method (31).

The *in vivo* function of DCs was determined using the hu-PBL-SCID mouse system, as previously described (16). Briefly, a SCID mouse received antigen-pulsed mature DCs

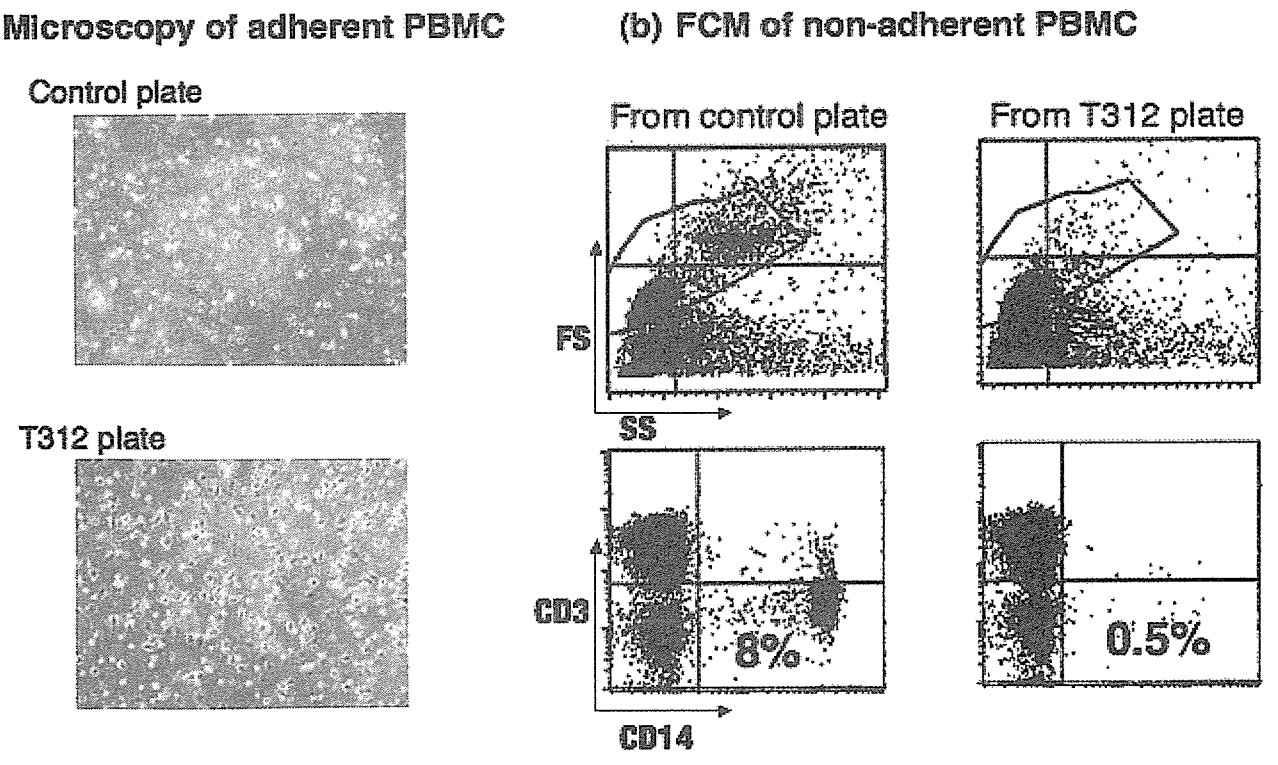

Figure 1. Stable adhesion of monocytes onto the bottom of plastic wells precoated with an anti-human CCR5 mAb. Aliquots (1 ml) of PBMCs at 2×10^6/ml of 5% FCS-RPMI medium were cultured overnight in individual wells of a 12-well plate that had been previously coated either with clone T312 rat IgG1 anti-human CCR5 or an isotype control mAb. (a) The morphology of the adherent cells as seen by phase-contrast microscopy at ×100. (b) Nonadherent PBMCs from each well were collected and analyzed by flow cytometry. The percentages of $CD14^+$ monocytes in the lymphocyte and monocyte gate in nonadherent PBMCs are noted in the double-staining profile with anti-CD3 and anti-CD14 mAbs. FS, forward scatter; SS, side scatter.

(5×10^5 cells) with autologous fresh PBMCs (3×10^6 cells) in a final volume of 0.1 ml in RPMI medium injected directly into the spleen, followed 7 days later with the ip injection of the same number and source of DCs pulsed with the same antigen. Five days later, the mice were sacrificed, and blood was collected by cardiocentesis and human lymphocytes were recovered from both the peritoneal cavity and the spleen. The immune sera were assayed for human antibody titers against OVA or KLH by ELISA using peroxidase-labeled goat anti-human IgG, as described previously (16). For the measurement of antigen-specific human T-cell immune responses, the human lymphocytes (2×10^6 cells) collected from the immunized mice were cultured for 2 days at 37°C in a 5% CO_2 humidified incubator with 2×10^5 autologous APCs (adherent PBMCs) in the presence or absence of either 5 μg/ml OVA or KLH in a volume of 0.5 ml in individual wells of a 24-well microtiter plate (BD Pharmingen). Media used consisted of RPMI medium supplemented with 20 U/ml of IL-2. The concentration of human IFN-γ produced in the culture supernatants was determined by standard ELISA.

Results

Cross-Linked CCR5 Induces Strong Adhesion of Monocytes. Twelve-well microtiter plates were first coated with the rat anti-human CCR5 N-terminus antibody (IgG1, clone T312) or an isotype control mAb. Then, PBMCs (5×10^6/ml) isolated from several normal donors were individually incubated overnight at 37°C in a 5% CO_2 humidified incubator in a volume of 1 ml of RPMI medium in each well of the plate. Following incubation, the nonadherent cells were removed. As shown in Figure1a, there appeared to be a marked increase in the number of cells that remained adherent to the wells precoated with the T312 mAb as compared with the control wells. As seen in Figure 1b, of interest was the finding that the nonadherent cells from the T312 mAb-coated wells were selectively depleted of $CD14^+$ cells (0.5%), compared with the nonadherent cells from the control wells (8%). The input unfractionated PBMCs from the various donors used for this study contained levels of $CD14^+$ cells that were essentially similar to the levels noted in the control wells, indicating that incubation of the PBMCs in the T312 precoated wells led to the select adhesion of the $CD14^+$ cells. Lymphoid cells other than $CD14^+$ monocytes were also present in the population of cells that adhered to the T312 mAb-coated wells, which mainly consisted of T and B cells, constituting up to 20% of the total adherent cells (data not shown). It is important to note that the addition of the T312 mAb to aliquots of the PBMCs from the same donors before

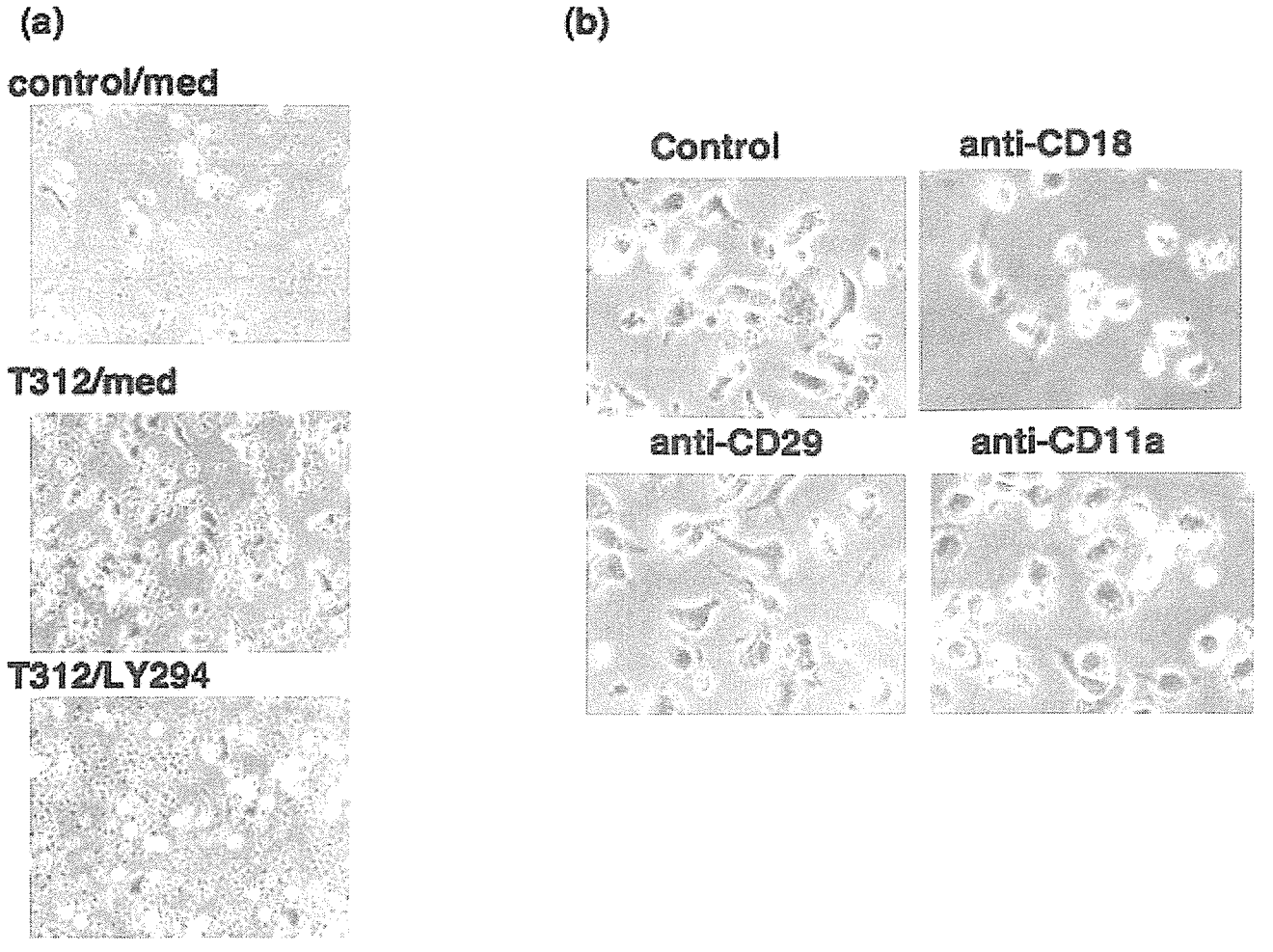

Figure 2. Microscopic analysis of the mechanisms for monocyte adhesion following CCR5 cross-linking. (a) PBMCs were incubated for 2 hrs in control mAb-coated wells (control/med) or overnight in T312 mAb-coated wells in medium alone (T312/med) or in the presence or of 5 μ*M* PI3-K inhibitor, LY294.002 (T312/LY294). (b) PBMCs were cultivated overnight in T312 mAb-coated wells in medium alone (control) or in the presence of 10 μg/ml of each mAb: anti-CD18, CD29, or CD11a. In these experiments, nonadherent cells were removed by gentle washing, and the wells were analyzed microscopically at ×200.

incubation in the microtiter wells or the addition of the same T312 mAb to the PBMCs following dispensing of the cells into the microtiter wells did not lead to increased adhesion of the $CD14^+$ monocytes, providing evidence indicating that the antibody had to be cross-linked for the adhesion to occur. A study of the kinetics of cell adhesion to the T312 mAb-coated wells was also performed. It should also be noted that there was a difference in the incubation time for maximal yield of monocytes following incubation of PBMCs in wells precoated with the T312 mAb, compared with the control antibody. Thus, where a 2-hr incubation was sufficient to obtain the maximum yield of monocytes by incubation of PBMCs in the control antibody-coated wells, it required an overnight or 24-hr incubation in the T312 mAb-coated wells (data not shown). Thus, for all further studies, adherent cells were isolated by incubation of the PBMCs either for 2 hrs in control wells or overnight in T312 mAb-coated wells, unless otherwise noted.

Since binding of CCR5 by its natural ligands is known to induce intracellular signaling via the PI3-K pathway, experiments were carried out to determine if such signaling was a prerequisite for the stable binding of the monocytes to the T312 mAb-coated wells. The above experiment was repeated, with the only difference being that an aliquot of each of the donor PBMCs was incubated in the T312 mAb-coated wells in the presence of 5 μ*M* of the PI3-K inhibitor LY294.002. Representative data shown in Figure 2a demonstrate that the addition of the PI3-K inhibitor completely blocked the cell adhesion. Flow cytometric analysis of the nonadherent cells in the wells containing the PI3-K inhibitor confirmed the presence of a high frequency of $CD14^+$ cells, denoting the lack of adhesion of the CD14-expressing cells (data not shown). It was also of interest that the addition of various concentrations of soluble or immobilized RANTES, MIP-1α, and MIP-1β did not enhance monocyte adhesion (data not shown), denoting that simple ligation of the chemokine receptors was not sufficient to facilitate adhesion.

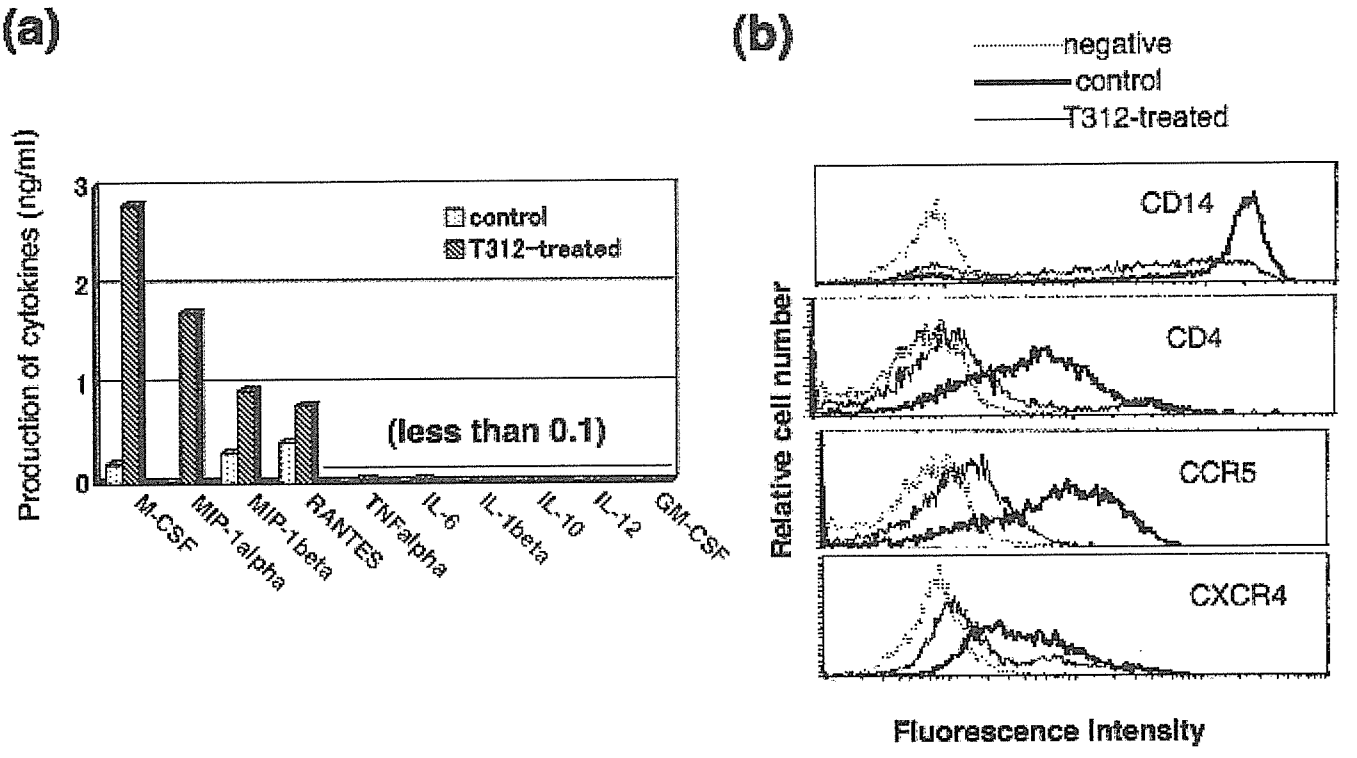

Figure 3. Cytokine profile and phenotypic analysis of purified monocytes following CCR5 cross-linking. (a) CD14 negatively selected monocytes at 2×10^5/ml in RPMI medium in T312 mAb-coated (T312-treated) or control wells (control) were cultured overnight, and cytokines produced in the culture supernatants were assayed by ELISA. Values of <0.1 ng/ml were below the sensitivity of the assay employed. (b) Aliquots of monocytes that were cultured in control wells (thick dark lines) or those cultured in T312 mAb-coated wells (thin dark lines) were analyzed for the expression of CD14, CD4, CCR5, and CXCR4 by routine flow cytometry. The broken lines show background staining of cells with isotype control mAbs.

It has been previously documented that adhesion of monocytes onto plastic plates is mediated by fibrinogen binding by the β2-integrins Mac-1 (CD11b/CD18) and/or p150/95 (CD11c/CD18) (32, 33). As shown in Figure 2b, the T312 mAb-induced adhesion of monocytes was blocked by anti-CD18 blocking mAb (with specificity for the common chain of β2-integrins), but not anti-CD29 mAb (β1-integrin) or anti-CD11a (another component of the β2-integrin, LFA-1), indicating an involvement of the β2-integrins in the enhanced adhesion of monocytes by T312 mAb-mediated CCR5 cross-linking.

Cytokine Profile and Phenotype Analysis of Monocytes Adhered to the T312 mAb-Precoated Wells. In an effort to define the cytokine profile characteristic of the monocyte population, it was reasoned that depletion of nonmonocyte lymphoid cells before analysis would be important. The magnetic bead negative selection technique for $CD14^+$ cells was thus used to deplete such nonmonocyte lymphoid cells, and the subsequent enriched monocytes were then incubated overnight in wells precoated with either T312 or control mAb, and the supernatant fluids were collected. Representative data from three experiments are shown in Figure 3a. As seen, the T312 mAb-stimulated monocytes secreted significant levels of M-CSF, MIP-1α, MIP-1β, and RANTES. The $CD14^+$ monocytes are likely to be the major cell lineage that responded to the T312 mAb stimulation by the production of cytokines, because PBMCs depleted of $CD14^+$ monocytes failed to synthesize detectable levels of the same cytokines. An aliquot of the same supernatants was also screened for levels of GM-CSF, TNF-α, IL-1β, IL-12, or IL-10, but each was found to be below 0.1 ng/ml. A variety of rat and mouse mAbs that had been generated in our laboratory with specificity for HTLV-1, HIV-1, HCV, OX40L, OX40, or IL-2R were also used to precoat the wells and were screened for their ability to induce cytokine synthesis by purified monocytes in parallel with the use of T312 mAb. As expected, only monocytes incubated in the wells precoated with T312 mAb produced M-CSF, MIP-1α, MIP-1β, and RANTES, denoting an element of specificity for the T312 mAb (data not shown).

Phenotypic analysis of monocytes adhered to the T312 mAb-precoated wells consistently showed a marked down-regulation of CD14, CD4, CCR5, and CXCR4 (a representative profile is shown in Figure 3b). Downmodulation of the CD14 on the cell surface was not attributed to the shedding of CD14, since there was no difference in the level of soluble CD14 between the supernatant fluids from the wells precoated with the T312 mAb and those from the control wells, as determined by ELISA (data not shown).

CCR5 Cross-Linking Stimulates Monocyte Dif-

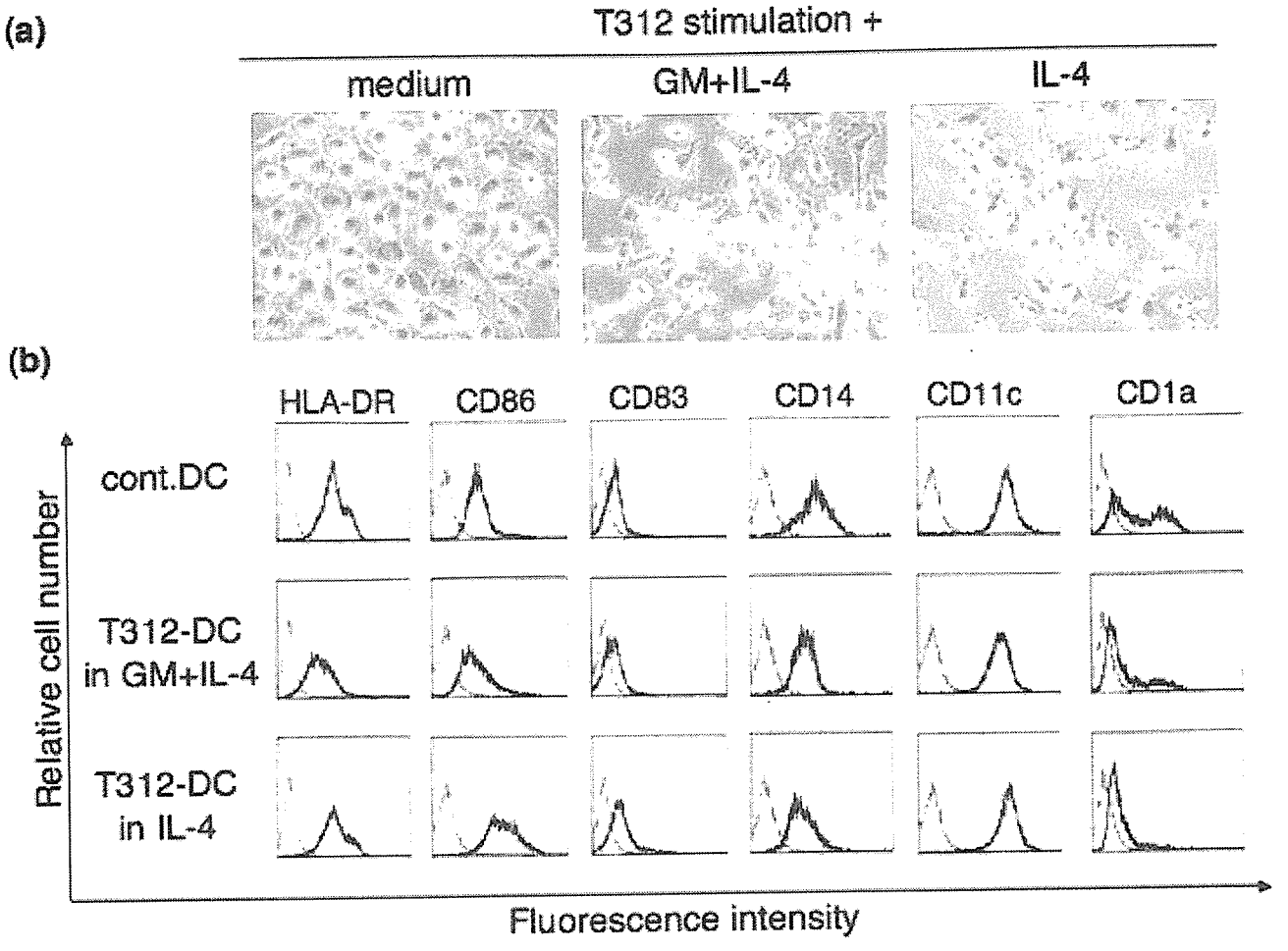

Figure 4. Morphological and flow cytometric analyses of adherent PBMCs following maturation in different culture conditions *in vitro*. (a) PBMCs at 5×10^6/ml in RPMI medium were cultured overnight in wells precoated with T312 mAb. After removal of nonadherent cells, adherent cells were cultured in RPMI medium (medium) or in the presence of 500 ng/ml GM-CSF and 200 ng/ml IL-4 (GM-CSF+IL-4) or 25 ng/ml IL-4 alone (IL-4) for 5 days. Morphology was observed microscopically at ×200. (b) The T312-induced immature DCs were generated from PBMCs adhered to the T312 mAb-coated wells during overnight incubation followed by cultivation in the presence of either GM-CSF and IL-4 (T312-DC in GM+IL-4) or IL-4 alone (T312-DC in IL-4). For comparison, control conventional DCs were generated from 2 hr–adherent PBMCs in control wells followed by cultivation in GM-CSF and IL-4 for 6 days (cont. DC). After maturation for an additional day with IFN-β, aliquots of these cells were subjected to flow cytometric analysis, and the profiles for the expression of HLA-DR, CD86, CD83, CD14, CD11c, and CD1a are shown.

ferentiation into DCs. To test whether the T312 mAb-induced CCR5 cross-linking has the potential to differentiate monocytes into functional DCs, aliquots of PBMCs were first incubated either in the T312 mAb-coated wells overnight or the control mAb-coated wells for 2 hrs (for the generation of conventional DCs), the nonadherent cells were removed, and then the adherent cells were further cultured in the presence or absence of either GM-CSF and IL-4 or IL-4 alone for 5 or 6 days, respectively.

As shown in Figure 4a, the adherent cells incubated in T312-coated wells appeared to have a DC type of morphology, which was confirmed by flow cytometric analysis (see Fig. 4b). Interestingly, the T312 mAb-stimulated monocytes, when cultured in the presence of IL-4, regardless of the presence or absence of GM-CSF, exhibited higher mean density levels of CD86 and lower levels of CD1a than did control conventional DCs that were cultured in media containing both GM-CSF and IL-4. The DCs cultured in the presence of GM-CSF and IL-4 in the T312 mAb-coated wells had a phenotype that was basically similar to that of the ones cultured in IL-4 alone, and these are both representative profiles of immature DCs. Kinetic studies of the differentiation of these T312 mAb-stimulated immature DCs showed that the optimal level of maturation was achieved by cultivation in the presence of IFN-β for an additional day.

We next sought to examine the relative phagocytic activity of monocyte-derived DCs or macrophages that were cultured in various conditions. As seen in Figure 5, macrophages obtained following differentiation of a highly enriched population of $CD14^+$ monocytes in M-CSF–containing media in general gave a higher relative phagocytic index than did DCs generated in media containing GM-CSF and IL-4. In addition, those that were

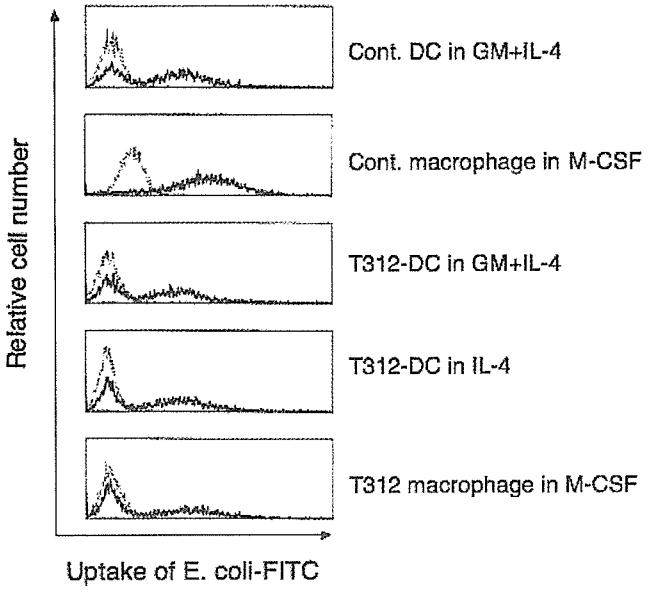

Figure 5. Phagocytic activities of cultured macrophages or immature DCs. Various DCs were generated as described in the legend for Figure 4, and macrophages were generated from 2 hr–adherent PBMCs followed by cultivation in the presence of M-CSF for 6 days. These sample cells in RPMI medium were incubated with a 10-fold excess of FITC-labeled *E. coli* particles for 1 hr at 37°C. After washing, the levels of uptake of FITC-labeled *E. coli* by cells were determined by flow cytometry with gating for macrophages and DCs using standard forward and side scatter.

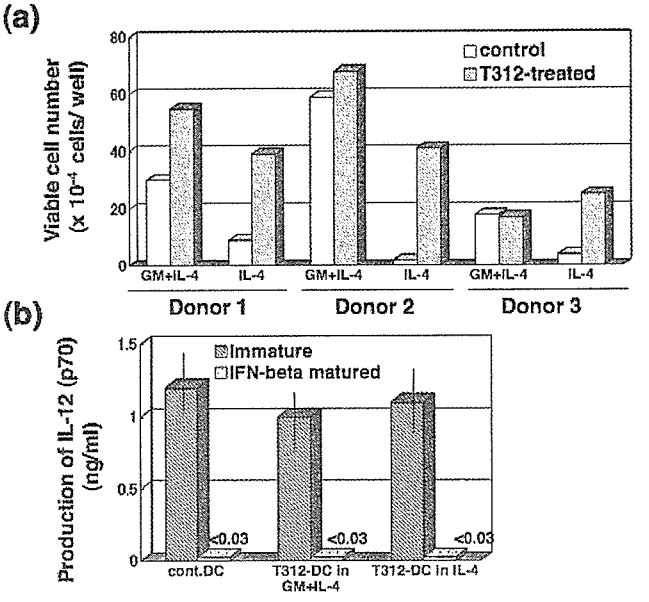

Figure 6. Yields of DCs and IL-12 production. (a) Immature DCs were generated by either incubation of aliquots of PBMCs from three different donors in the control wells (control) or in the T312 mAb-coated wells (T312-treated) and then for 6 or 5 days, respectively, with GM-CSF and IL-4 or IL-4 alone, as described in the legend for Figure 4. Numbers of viable cells that were not stained by eosin-Y were manually counted using a hemocytometer. The means from triplicate determinations are shown. (b) DCs from a donor before (immature) or after IFN-β treatment (mature) were stimulated by 1 μg/ml LPS and 100 ng/ml IFN-γ for 24 hrs. IL-12 p70 levels in the culture supernatants were determined by ELISA.

stimulated with T312 mAb and cultured in media containing either GM-CSF and IL-4 or IL-4 alone were less phagocytic than the control macrophages generated by M-CSF. Interestingly, the T312 mAb-stimulated monocytes, following incubation with media containing M-CSF alone, gave slightly lower phagocytic values than the normal macrophages cultured in the same media. These data seem to indicate that cross-linking of CCR5 influences monocyte differentiation into DC rather than macrophage, even in the presence of M-CSF.

We further determined the relative levels of viability of the cells that adhered either to the T312-coated wells overnight or to control wells for 2 hrs followed by cultivation in medium alone or media containing either GM-CSF and IL-4 or IL-4 alone for 5 or 6 days, respectively. Figure 6a shows, first of all, that whereas there was variation in the number of viable cells recovered among donors of the PBMCs, the yields of viable cells generated in the T312 mAb-coated or control wells followed by cultivation in GM-CSF and IL-4 were basically similar. In contrast, aliquots of the same cells incubated in T312 mAb-coated or control wells gave different yields following incubation with IL-4 alone. The cells stimulated by T312 mAb gave significantly higher viable yields than those incubated in control wells. The overnight adherent monocytes could differentiate into functional DCs in the presence of GM-CSF and IL-4, but the cell yield was lower than those obtained from the 2-hr adherent cells (data not shown).

Aliquots of these *in vitro*–generated immature DCs were subsequently challenged with LPS and IFN-γ for 48 hrs, and supernatant fluids were assayed for levels of IL-12 p70. Controls consisted of aliquots of the conventional DCs obtained from the same PBMCs in wells coated with control IgG1 instead of T312 mAb. As seen in Figure 6b, there were significant levels of IL-12 synthesized by both these populations of immature DCs, and the importance of the levels appeared quite similar. Furthermore, the IL-12–producing activity of these immature DCs was lost after full maturation by incubation with IFN-β.

Function of DCs. In an effort to determine whether the T312 mAb-induced DCs were as competent as conventional DCs, these immature DC populations were matured by cultivation in media containing IFN-β for 1 day and the aliquots dispensed into microtiter wells and standard mixed lymphocyte reaction (MLR) studies performed. The T312 mAb-induced DCs (termed T312/IL-4 or T312/GM-IL-4) and the conventional DCs (termed control/GM-IL-4) were co-cultured with allogeneic naïve $CD4^+$ T cells. Controls consisted of $CD4^+$ T cells cultured alone or DCs cultured alone. The cultures were performed at a DC to T-cell ratio of 1 to 20, and each combination was cultured in triplicate for 7 days and proliferation assessed using BrdU incorporation by the ELISA method. Supernatant fluids from the MLR

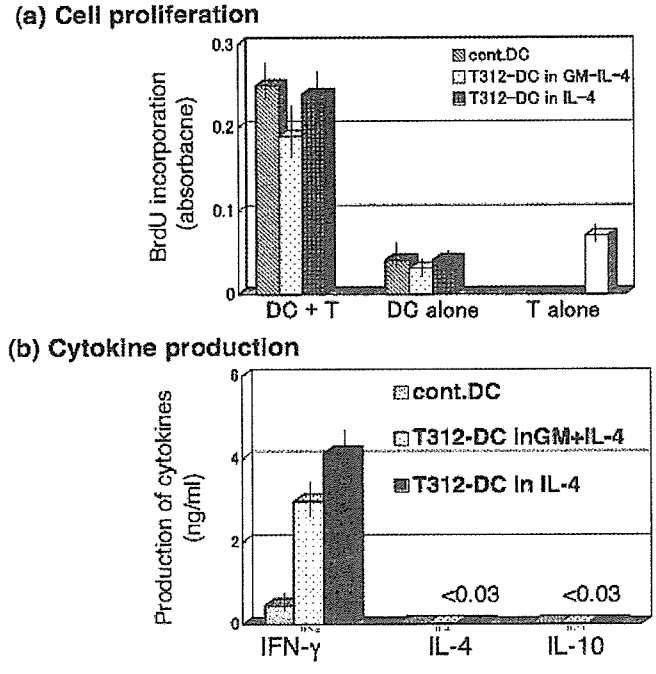

Figure 7. T312 mAb-induced DCs stimulate allogeneic naïve $CD4^+$ T cells to proliferate and produce IFN-γ. Allogeneic naïve $CD4^+$ T cells were co-cultured with IFN-β–matured DCs that had been generated from the adherent PBMCs, as described in the legend for Figure 4 in RPMI medium supplemented with 20 U/ml IL-2 for 7 days. (a) Cell proliferation was determined by BrdU incorporation. Mean values from six wells and standard errors are shown. Controls consisted of culturing either DCs or T cells alone. (b) IFN-γ production in the culture supernatants was determined by ELISA.

cultures were also assayed for levels of IFN-γ, IL-4, and IL-10. As shown in Figure 7a, there was no significant difference in the proliferation-inducing activity among the three preparations of DCs. However, an obvious difference was seen in the levels of IFN-γ synthesized from the naïve $CD4^+$ T cells (Fig. 7b). The T312 mAb-induced DCs generated in the presence of either GM-CSF and IL-4 or IL-4 alone stimulated higher production of IFN-γ than the control DCs. None of these DCs induced production of IL-4 or IL-10 from the $CD4^+$ T cells by co-culture. The T312 mAb-induced DCs did not produce detectable levels of human IFN-γ, even following stimulation with LPS or CD40L (data not shown), indicating that the allogeneic naïve $CD4^+$ T cells are the likely source of IFN-γ following co-culture and stimulation with DCs. These results indicate that the T312 mAb-induced DCs acquired a relatively higher Th1-polarizing capacity than control conventional DCs.

Furthermore, we tested and compared the ability of these DCs to induce human antigen-specific T- and B-cell immune responses *in vivo*. For this purpose, we used our hu-PBL-SCID mouse model (16). SCID mice were engrafted with normal PBMCs together with antigen (OVA)–pulsed autologous T312 mAb-induced or conventional mature DCs. A booster injection with antigen-pulsed homologous source of mature DCs was performed after 7 days. Immune

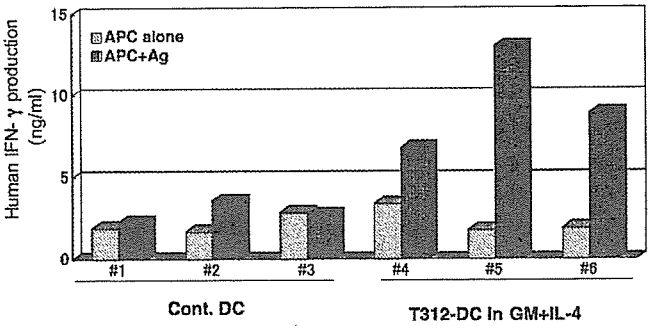

Figure 8. T312 mAb-induced DCs trigger human antigen–specific T-cell responses in hu-PBL-SCID mice. DCs were generated as described in the legend for Figure 4, followed by exposure to antigen (100 μg/ml OVA or KLH) for 24 hrs and by maturation with IFN-β for an additional 24 hrs. These DCs (5×10^5 cells) together with autologous fresh PBMCs (3×10^6 cells) were mixed in 0.1 ml RPMI medium and transplanted into the spleen of SCID mice. Booster immunization was made by injection of the same numbers of antigen-loaded DCs (ip) on Day 7. After 5 days, cells obtained from the spleen and peritoneal cavity of the mice were cultured with autologous APCs (adherent PBMCs during 2-hr incubation in normal wells) in the presence (APC + Ag) or absence (APC alone) of 5 μg/ml OVA for 2 days, and the supernatants were assayed for human IFN-γ by ELISA. Values depicted reflect the mean levels of IFN-γ of triplicate cultures (SD was <10%).

sera and spleen cells were harvested after 5 days following the booster injection. Figure 8 shows that immunization with the T312 mAb-induced and OVA-pulsed DCs was more efficient in inducing antigen-specific T-cell immune responses in the hu-PBL-SCID mice, as determined and defined by human IFN-γ production by the immune cells *in vitro*, than the conventional DCs pulsed with OVA. IL-4 was not produced from those immune cells (data not shown). Human OVA-specific IgG were also induced by both types of the conventional and T312 mAb-induced DCs up to a titer of 1:400, as determined by ELISA (data not shown). Essentially similar results were obtained when mice were immunized with another antigen, KLH (data not shown). These data indicate that the T312 mAb-induced DCs, in general, appear to be relatively more efficient than conventionally derived DCs in inducing antigen-specific IFN-γ responses *in vivo*.

Effect of Cross-Linking by Other mAbs Against Chemokine Receptors. Based on the results from the above studies, additional experiments were carried out to examine the specificity of the ligation of the CCR5 coreceptor by the T312 mAb. Thus, a battery of mAbs against human chemokine receptors expressed by monocytes, including CCR1, CCR2, CCR3, CCR5, CCR8, and CXCR4, were used to precoat microtiter wells, and aliquots of purified monocytes were incubated in such antibody-coated wells. The level of M-CSF synthesized by the cells following overnight incubation was used as an index of monocyte stimulation. As seen in Figure 9 among the anti-human CCR5 mAbs tested, Mab180 (mouse IgG anti-CCR5 N-terminus), but not T227 (rat IgG2a anti-CCR5 N-terminus) or 2D7 (mouse IgG anti-CCR5 ECL-2), was as

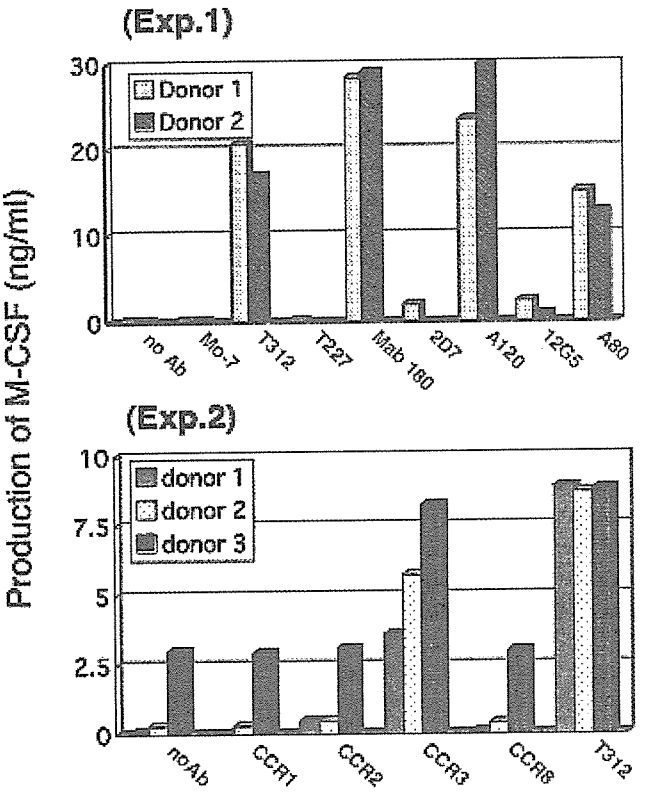

Figure 9. Screening of the other anti-chemokine receptor mAbs that can stimulate monocytes. Purified monocytes at 1×10^5/ml in RPMI medium were cultured in wells precoated with various mAbs overnight, and the culture supernatants were assayed for M-CSF by ELISA. The values depicted represent the mean of triplicate cultures with SD of <10%.

effective as T312 mAb in the stimulation of monocytes. Among the anti-human CXCR4 mAbs, clone A120 (rat IgG2b anti-CXCR4 ECL1&2) and A80 (rat IgG1 anti-CXCR4 ECL-3) were also capable of stimulating monocytes. Another monocyte-stimulating mAb was the rat IgG2a anti-CCR3 mAb. These mAbs that successfully stimulated monocytes also promoted monocyte adhesion, and A120 anti-CXCR4 mAb in particular was very potent in inducing adhesion and differentiation of monocytes similar to the data derived using the T312 mAb (data not shown). Whether subtle differences exist in the adherent cells enriched by the mAbs that were active in such an assay remains to be determined.

Discussion

In the present study, we present data showing that cross-linking of CCR5 as well as select other chemokine receptors (CXCR4 and CCR3) by mAbs immobilized onto plastic microtiter wells leads to enhanced adhesion of monocytes followed by synthesis of M-CSF and a set of β-chemokines and that incubation of these adherent cells with GM-CSF and IL-4 or IL-4 alone leads to their differentiation along the DC lineage. This functional activity observed following chemokine receptor cross-linking is in contrast to the data obtained with FcR cross-linking (34). This enhanced adherence of monocytes apparently improves the efficacy of adhesion-based monocyte enrichment from fresh PBMCs.

One contributing mechanism that facilitates such tight adhesion may involve the enhanced affinity of the β2-integrins Mac-1 and/or p150/95, which bind to fibrinogen, an extracellular matrix protein, since such adhesion was markedly inhibited in the presence of mAb anti-CD18 (a common chain of the β2-integrin) but not anti-CD11a (a component of another β2-integrin, LFA-1). This view is supported by data obtained by flow cytometric analysis of the T312 mAb-stimulated monocytes, compared to those incubated in control mAb-coated wells. Thus, the T312 mAb-stimulated cells but not the control cells showed a markedly increased density of staining with FITC-fibrinogen, but not with FITC-gelatin or collagen (data not shown). Although sensitization of chemokine receptors by their respective cognate chemokine ligands is known to activate β1- and β2-integrins (5), it is important to note that the addition or precoating of wells with chemokines that bind to CCR5 (i.e., RANTES, MIP-1α, and MIP-1β), respectively, did not induce such enhanced monocyte adhesion *in vitro* in the present study (data not shown). Thus, it is likely that antibody-mediated cross-linking of chemokine receptors initiates an enhanced degree of activation of monocytes, probably through an extraordinary degree of polarization of the receptors onto the plates. This is supported by the observations made by Lee *et al.* (35) that the mode of monocyte activation by cross-linking of chemokine receptors by gp120 of HIV-1 particles was stronger than that induced by a simple interaction between CCR5 and CXCR4 with their respective chemokines. The inhibition of the T312 mAb-induced monocyte adhesion by the PI3-K-specific inhibitor LY294.002 indicates that the T312 mAb-mediated CCR5 cross-linking also stimulates the G-protein signal cascade, as documented previously for gp120 of HIV-1 (35) and chemokines (36). Since PI3-K activation by chemokines also induces polarization of adhesion molecules and facilitates migration (5, 37), it is likely that it is either the mere strength of the signal that is induced by cross-linking of CCR5 by T312 mAb or the potential pulling together of additional, as-yet-unknown proteins that distinguishes between enhanced adhesion and mere signaling.

It is worth noting that the tight adhesion was not induced by another rat IgG anti-CCR5 mAb, termed clone T227. The difference between anti-CCR5 clone T312 and T227 is that the T227 mAb recognizes a region located within the N-terminus of the CCR5 molecule that has been further localized to a peptide spanning the CCR5 N-terminus amino acids 1–20 but not 2–21, whereas the clone T312 mAb recognizes both of the peptides, with an affinity similar to the clone T227 mAb (Tanaka *et al.*, unpublished data). These two mAbs compete with each other in binding to CCR5. Thus, these results indicate that activation of monocyte by CCR5 cross-linking is epitope-dependent

rather than antibody affinity-dependent. This view is further supported by the observation that the use of yet another immobilized CCR5-specific clone, ECL-2 mAb, did not lead to enhanced adhesion of monocytes, indicating a requirement for epitope specificity to facilitate adhesion. Similarly, with regard to CXCR4, the clones A145 mAb (anti–N-terminus) and 12G5 (anti-ECL1&2) did not induce significant monocyte adhesion, in contrast to A120 mAb (anti-ECL1&2) and A80 mAb (anti–ECL-3). The clone A120 mAb blocks the binding of SDF-1 (the natural ligand for CXCR4) and inhibits infection by the CXCR4-tropic HIV-1 strains, whereas the clone A80 mAb does not block SDF-1 binding but stimulates homologous T-cell aggregation and thus enhances infection of the CXCR4 and CCR5-tropic HIV-1 strains (25). However, it is still unclear which regions (epitopes) of each chemokine receptor are the hot spots for the induction of strong activation signals on monocytes, and this topic thus requires further study.

Along with the enhanced adhesion facilitated by cross-linking of the chemokine receptor, such cross-linking also induced downmodulation of cell surface expression of CD14, CD4, CCR5, and CXCR4 and cytokine production by monocytes. It is of interest that similar effects on monocytes have been induced by prokineticin 1 (PK1)/endocrine gland-derived vascular endothelial growth factor (EG-VEGF), and its G protein-coupled receptors (PKR1 and PKR2) (38). Downregulation of CD14 was reported to be induced by IL-4 or IL-13 at the transcriptional level (39) or by shedding, which was induced by antibody-mediated cross-linking of CD14 or stimulation by LPS and IFN-γ (40). However, these are not the likely mechanisms in studies reported herein, since there was little if any production of IL-4 and IL-13 and no detectable levels of soluble CD14 in the culture supernatants. It has been shown that downmodulation of cell surface expression of CD14, CD4, and CXCR4, but not CCR5 on monocytes, is induced by PKC activation by phorbol 12-myristate 13-acetate (PMA) (41, 42) and that CCR5 downmodulation is induced by CCR5-binding chemokines at only high concentrations as a result of CCR5 internalization (43). Whether these pathways are activated by cross-linking of the chemokine receptors is currently not known. It has also been shown that there is a molecular association between CD4 and the chemokine receptors or among the various chemokine receptors (44, 45). Thus, it can be speculated that CCR5 or CXCR4 cross-linking may induce homo- or hetero-oligomerization of the cell surface CD14, CD4, CXCR4, and/or CCR5, and is then internalized.

In the early studies on monocyte adhesion (5) it was shown that adherence onto plastic plates stimulates monocytes to activate M-CSF, IL-1β, or TNF-α gene expression and that LPS stimulation further enhances the synthesis of these cytokines. Similarly, activation of monocytes via the PK1 pathway following adhesion requires additional stimulation by LPS to promote the production of IL-12 p70 and TNF-α (46). Thus, it is clear that the mechanisms for cytokine production in the studies described herein likely involve distinct pathways other than those discussed above. In addition, the addition of soluble chemokines or the use of immobilized chemokines with specificity for CCR5 and CXCR4 did not induce cytokine production in the present conditions (data not shown). Differences in the precise pathways that are triggered in the studies reported herein are a subject of current study.

The observation that not only CCR5 but also CXCR4 and CCR3 cross-linking triggered production of M-CSF by monocytes and induced differentiation in the absence of exogenously added M-CSF or GM-CSF was unexpected. For differentiation of monocytes into macrophages *in vitro* in the absence of these cytokines, extracellular matrix proteins are known to have profound influence (47). It has been previously reported that human monocytes differentiated into CD14-low macrophages in a fibronectin-containing medium without the addition of exogenous cytokines (48). Thus, bovine fibronectin in our RPMI medium may have some additive effects on chemokine receptor–stimulated monocyte differentiation, although the use of serum-free medium for monocyte cultivation in T312 mAb-coated wells did not influence the present results (data not shown). Although cross-linking of CCR5, CXCR4, or CCR3 resulted in M-CSF production from monocytes, anti-M-CSF– or GM-CSF–neutralizing mAbs did not block monocyte differentiation into macrophages (data not shown). Thus, we assume that chemokine receptor cross-linking may bypass the requirement of M-CSF stimulation. Since the ligation of M-CSF receptor (CD115) stimulates PI3-K (49, 50), the PI3-K activation as a result of the chemokine receptor cross-linking may be responsible for the *de novo* monocyte differentiation into macrophages. The mechanisms by which cross-linking of chemokine receptors on monocytes induced the differentiation of these cells into DCs without the addition of exogenous GM-CSF in the presence of IL-4 are also under current study. The cross-linking may function as survival and differentiation factor for the generation of DCs from monocytes, because monocytes in the absence of chemokine receptor cross-linking failed to survive when cultured in media containing IL-4 alone.

The primary objective of the present study was to find an alternate, more-efficient and practical method for the *in vitro* generation of functional DCs with potent Th1-inducing capacity *in vivo*. Various cytokine cocktails have been applied for generation of functional DCs, including combinations of IL-3 and IFN-β, Flt3L and IL-4, CD40L alone, or GM-CSF, IL-4, and the proinflammatory cytokines GM-CSF and Type I IFN (51–56). In addition, it has been reported that the methods of monocyte isolation, such as adherence or $CD14^+$ enrichment by MACS (which results in CD14 cross-linking), influence cytokine production from generated DCs (i.e., adherent monocyte-derived DCs were more potent in producing IL-12, IL-10, and TNF-α and in stimulating Tc1 than those from CD14 positively selected

monocytes) (24). Because the DCs generated in the studies reported herein by CCR5 (or CXCR4 or CCR3) cross-linking in the presence of either GM-CSF and IL-4 or IL-4 alone were relatively more potent than conventional DCs in stimulating IFN-γ production from allogeneic naïve $CD4^+$ T cells *in vitro* and OVA- or KLH-specific T cells *in vivo*, it is indicated that in studies that require Th1-polarizing DCs, the cross-linking of CCR5, CXCR4, or CCR3 on monocytes should be considered. The higher density of expression of CD86 on the chemokine receptor cross-linked DCs may have some influence for Th1-inducing capacity, although the mechanism at present still remains unknown.

Based on the results of the present study, we suggest that the use of cross-linking of CCR5, CXCR4, or other chemokine receptors for the generation of functional DCs *ex vivo* provides an alternate and simple tool that will facilitate the therapeutic use of DCs. Thus, studies of DC-based immunotherapy in patients with HIV-1 infection that have been reasoned to require induction of maintenance of Th1 type of antiviral immune response could benefit from such an approach. In addition, cross-linking of the chemokine receptors could also facilitate limited entry of HIV-1 into such cells because of downmodulation of the HIV-1 receptors. Such an approach has already been initiated, and preliminary studies have shown that inactivated HIV-1–pulsed DCs generated from CCR5– and CXCR4–cross-linked monocytes could trigger highly potent human anti–HIV-1 T-cell responses using our hu-PBL-SCID mouse model. These studies are currently in progress.

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Potential role of natural killer cells in controlling growth and infiltration of AIDS-associated primary effusion lymphoma cells

Md. Zahidunnabi Dewan,[1,2] Hiroshi Terunuma,[3,4] Masakazu Toi,[5] Yuetsu Tanaka,[6] Harutaka Katano,[7] Xuewen Deng,[3] Hiroyuki Abe,[4] Tadashi Nakasone,[2] Naoki Mori,[8] Tetsutaro Sata[7] and Naoki Yamamoto[1,2,9]

[1]Department of Molecular Virology, Graduate School, Tokyo Medical and Dental University, 1-5-45 Yushima, Bunkyo-ku, Tokyo 113-8519; [2]AIDS Research Center, National institute of Infectious Disease, 1-23-1 Toyama, Shinjuku-ku, Tokyo 162-8640; [3]Biotherapy Institute of Japan, 2-4-8 Edagawa, Koutou-ku, Tokyo 135-0051; [4]Kudan Clinic, 1-9-5 Kudankita, Chiyoda-ku, Tokyo 102-0073; [5]Division of Clinical Trials and Research, Breast Cancer Research and Treatment Program, Tokyo Metropolitan Komagome Hospital, Tokyo Medical Center for Cancer and Infectious Disease, 3-18-22 Honkomagome, Bunkyo-ku, Tokyo 113-8677; [6]Department of Immunology, Faculty of Medicine, University of the Ryukyus, 207 Uehara, Nishihara, Okinawa 903-0215; [7]Department of Pathology, National Institute of Infectious Diseases, 1-23-1 Toyama, Shinjyuku-ku, Tokyo 162-8640; [8]Division of Molecular Virology and Oncology, Graduate School of Medicine, University of the Ryukyus, 207 Uehara, Nishihara, Okinawa 903-0215, Japan

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Natural killer (NK) cells are an important component of the innate immune response against microbial infections and tumors. Direct involvement of NK cells in tumor growth and infiltration has not yet been demonstrated clearly. Primary effusion lymphoma (PEL) cells were able to produce tumors and ascites very efficiently with infiltration of cells in various organs of T-, B- and NK-cell knock-out NOD/SCID/γc^{null} (NOG) mice within 3 weeks. In contrast, PEL cells formed small tumors at inoculated sites in T- and B-cell knock-out NOD/SCID mice with NK-cells while completely failing to infiltrate into various organs. Immunosupression of NOD/SCID by treatment with an antimurine TM-β1 antibody, which transiently abrogates NK cell activity *in vivo*, resulted in enhanced tumorigenicity and organ infiltration in comparison with non-treated NOD/SCID mice. Activated human NK cells inhibited tumor growth and infiltration in NOG mice. Our results suggest that NK cells play an important role in growth and infiltration of PEL cells, and activated NK cells could be a promising immunotherapeutic tool against tumor or virus-infected cells either alone or in combination with conventional therapy. The rapid and efficient engraftment of PEL cells in NOG mice also suggests that this new animal model could provide a unique opportunity to understand and investigate the mechanism of pathogenesis and malignant cell growth. (*Cancer Sci* 2006; 97: 1381–1387)

Primary effusion lymphoma (PEL) was originally identified in AIDS-associated immunodeficient patients and has been recognized by the World Health Organization as a distinct AIDS-related form of B-cell lymphoproliferative disorder.[1–3] PEL is a non-Hodgkin's type lymphoma derived from postgerminal center B cells.[4] The tumor clone is characteristically infected by the Kaposi's sarcoma-associated herpesvirus, formerly called human herpesvirus type 8 (HHV-8),[5] and most cases are coinfected with Epstein–Barr virus.[6,7] PEL shows a peculiar presentation involving lymphomatous effusions of serous cavities and only occasionally presents with a definable mass.[5]

Immunodeficient mouse models of human malignancy have contributed significantly to understanding the pathogenesis of diseases as well as therapeutic purpose. The congenitally athymic and hairless nude mouse lacking functional T cells has been utilized as a host for human xenotransplantations for 30 years.[8] Thereafter, severe combined immunodeficiency (SCID) mice were found to have a genetic defect preventing functional development of T and B lymphocytes,[9,10] and can be engrafted successfully with a variety of normal hematopoietic and neoplastic cells.[11,12] In comparison with conventional SCID, the NOD-SCID strain appears to be more promising as a tool for xenotransplantion of human tumors. However, the NOD-SCID mouse strain retains natural killer (NK) cell activity, macrophage function, complement activity and functional dendritic cells.[13] NK cells might play an important role in the rejection of implanted tissues or cells in SCID mice.[14–17] Although several models using mainly conventional nude and SCID mice are available, there are some major drawbacks: the requirement of long time periods, repeated transplantation, total body irradiation of mice, hormone supplements, etoposide pretreatment and anti-NK monoclonal antibodies required for tumor formation. These problems appear to hinder wider use of these animal models. Due to the low engraftment efficiency of hematopoietic and tumor cells transplanted in SCID mice, T, B and NK knock-out NOD/SCID/γc^{null} (NOG) mice were used in the present study to investigate the role of NK in tumor growth and metastasis.[13]

Natural killer cells are a type of lymphocyte that comprises up to 15% of peripheral blood lymphocytes and mediates innate immunity against pathogens and tumors.[17] In addition, NK cells are an important source of cytokines that regulate hematopoiesis and link the innate to the adaptive immune response through a bidirectional cross-talk with dendritic cells.[18,19] NK cells were originally discovered because of their ability to kill tumor and virally infected cells *in vitro*. NK-cell activity against these *in vitro* targets is spontaneous; it is readily apparent in individuals who have not been previously exposed to the target cell antigens. A clear involvement of NK cells in antitumor immunity *in vivo*, and the involvement of major histocompatibility complex (MHC) class I in NK-cell recognition, was shown in 1986 by Karre and colleagues.[20] They showed that the RMA T-cell lymphoma, derived from the Rauscher virus-induced murine cell line RBL-5, grew progressively in syngenic mice, but that an MHC class I-negative variant, RMA-S, was rejected by host NK cells. In many different situations, NK cells were shown to kill certain tumor cell lines *in vitro*, despite significant levels of MHC class I on their cell surface.[21,22] This implied that killing of MHC class I^+ tumor cells was mediated by activating receptors that were either not impaired by the inhibitory NK receptors for MHC class I or provided sufficient stimulation to overcome the negative regulation.

One cohort study showed that individuals with low natural cytotoxic activity of peripheral blood lymphocytes are at a

[9]To whom correspondence should be addressed. E-mail: yamamoto.mmb@tmd.ac.jp

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significantly higher risk of cancer, compared with those of median or high activity.[23] It has been reported recently that NK cells isolated from HIV-infected individuals are impaired in their ability to kill the virus-infected autologous cells, as well as tumor cell lines.[24–27] Previous studies also reported that NK-cell activity controls PEL and Kaposi's sarcoma (KS) development associated with HHV-8 infection.[28,29] The ability of the NK cells to kill relevant targets, such as tumor or virally infected cells, depends on the delicate balance of the patterns of expression of MHC class I-specific inhibitory NK receptors and activating receptors.[30,31] As there is no animal model in which NK-cell activities are genetically and selectively deficient to rule out the function of NK cells in viral infection and tumor growth and metastasis, most studies have relied on depleting NK cells in mice using monoclonal or polyclonal antibodies.[32,33] Depletion of NK cells *in vivo* by anti-NK antibody leads to enhanced tumor formation in several mouse tumor models.[15,34] Therefore, the role of NK cells in the course of tumor growth and infiltration as well as viral infection remains one of the major topics in tumor immunology.

In the present study, we investigated the direct involvement of NK cells in growth and infiltration of PEL cells using T, B and NK knock-out NOG mice,[13,35,36] and T and B knock-out NOD/SCID mice. NK knock-out NOG mice were most efficient in the formation of large tumors, massive ascites and infiltration within 3 weeks in comparison with NK-bearing NOD/SCID mice. We also provide evidence that activated human NK cells inhibit tumor growth and infiltration in NOG mice. These results suggest that NK cells play an important role in tumor growth and infiltration, and activated NK cells could be a promising immunotherapeutic strategy against AIDS-associated PEL or other malignancies either alone or in combination with conventional therapy.

Materials and Methods

Mice and inoculation of cell lines. NOG and NOD/SCID mice were obtained from the Central Institute for Experimental Animals (Kawasaki, Japan). All mice were maintained under specific pathogen-free conditions at the Animal Center of Tokyo Medical and Dental University (Tokyo, Japan). The Ethical Review Committee of the institute approved the experimental protocol.

Primary effusion lymphoma cell lines BCBL-1[37] and TY-1,[38] NK cell line KHYG-1 and bcr-abl$^+$ leukemic cell line K562 were cultured in RPMI-1640 medium supplemented with 2% heat-inactivated fetal bovine serum (FBS; JRH Biosciences, Lenexa, KS, USA), 100 U/mL penicillin and 10 μg/mL streptomycin. BCBL-1 and TY-1 cells were washed twice with serum-free RPMI-1640 and resuspended in fresh RPMI-1640. Mice were anesthetized with ether and cells were inoculated either subcutaneously (sc) in the postauricular region or intraperitoneally (ip) in the abdominal region of mice at doses of 1×10^7 and 2×10^6 cells per mouse, respectively. BCBL-1 cells were also inoculated either sc in the postauricular region or ip in the abdominal region of NOD/SCID mice with or without pretreatment with TMβ1 antibody, or in NOG mice. All mice were killed 3 weeks after inoculation with PEL cells. We measured tumor size, collected ascites from the abdomen of mice, and measured the volume of ascites.

Isolation and culture of NK cells. Blood was collected after obtaining informed consent from healthy volunteers. Peripheral blood mononuclear cells (PBMC) were isolated from the blood by Ficoll-Hypaque gradient centrifugation (Amersham Biosciences, Uppsala, Sweden), washed twice with RPMI-1640, and the number of cells counted. To generate activated NK cells, PBMC were cultured in anti-CD16-coated flasks with AIM-V medium (Invitrogen, Tokyo, Japan) supplemented with 5% auto-plasma, 700 U/mL interleukin (IL)-2 (Chiron, Amsterdam, the Netherlands), and 1 μL/mL OK432 (Chugai Pharmaceutical, Tokyo, Japan) for 24 h at 39°C, and then the cultured cells were centrifuged at 550 *g* for 10 min and the supernatants were discarded. Cells were again cultured in anti-CD16-uncoated flasks with AIM-V medium supplemented with 5% auto-plasma, and 700 U/mL IL-2 at 37°C for 2–3 weeks. During culture periods, we added medium several times for expansion and maintenance of activated NK cells. The purity of NK cells was 92–95%.

Flow cytometric analysis and cytotoxic activity. For five-color flow cytometric analysis (Cytomics FC500; Beckman Coulter, Miami, FL, USA), freshly isolated and activated NK cells were stained with monoclonal antibodies (ECD-labeled anti-CD3, PC5-labeled anti-CD4, PC7-labeled anti-CD8, PC7-labeled anti-CD16, PE or PC7-labeled anti-CD45, PC5-labeled anti-CD56, and PE-labeled anti-CD69 [Immunotech, Marseile, France]) and appropriate anti-isotypic monoclonal antibodies stained as negative controls. Data were analyzed by using CXP Analysis software version 1.1.

Freshly isolated and activated NK cells were tested for cytotoxic activity at various effector-to-target (E/T) ratios in a calcein-AM release assay using TERASCAN VP (Minerva Tech., Tokyo, Japan). We labeled the target cells with the immunofluorescent dye Calcein-AM solution (Do Jindo Laboratory, Kumamoto, Japan) and incubated them for 30 min. The cells were then washed with phosphate-buffer saline (PBS)(–) and the fluorescence intensity checked. Target cells and effector cells were suspended in RPMI-1640 with 10% FBS at various E/T ratios, added into 96-well plates and incubated for 2 h, and the fluorescence intensity was again checked.

Inoculation of activated NK cells into mice and collection of samples. Mice were inoculated with BCBL-1 (2×10^6) cells ip in the abdominal region of NOG mice. Three days after inoculation of PEL cells, mice were treated with either RPMI-1640 (control mice) or activated NK cells (1×10^7) ip on days 4, 10 and 17. All mice were killed 3 weeks after inoculation with PEL cells. We measured tumor size, collected ascites from the abdomen of mice, and measured the volume of ascites. Tissues and various organs of mice were also collected and fixed with 10% buffered formalin (Streck Tissue Fixative, Omaha, NE, USA), then processed to paraffin-embedded sections for staining with hematoxylin and eosin (HE) and immunostaining.

Immunohistochemistry. Paraffin sections of various organs were deparaffinized and hydrated in xylene or clearing agents and a graded alcohol series, then rinsed for 5 min in water. Deparaffinized samples were incubated with 0.025% trypsin/PBS for 30 min followed by washing, and then incubated with 0.3% H_2O_2 in methanol for 30 min at room temperature before being washed twice with PBS. Immunostaining was done for PEL cells with a 1:500 dilution of primary rabbit polyclonal antibody specific for HHV-8-encoded LANA.[39] This was followed by washing in PBS and incubation with a secondary antibody, biotinylated antirabbit IgG, after which cells were again washed in PBS and incubated with horseradish peroxidase-conjugated streptavidin for 30 min at room temperature. After two washes in PBS, the amplification procedure was carried out using kits according to the manufacturer's instructions (catalyzed signal amplification system kit; DAKO, Copenhagen, Denmark). The signal was visualized using 0.2 mg/mL diaminobenzidine and 0.015% H_2O_2 in 0.05 M Tris-HCl, pH 7.6. Positive staining was visualized after incubation of these samples with a mixture of 0.05% 3,3′-diaminobenzidine tetrahydrochloride in 50 mM Tris-HCl buffer and 0.01% H_2O_2 for 5 min. The samples were counterstained with hematoxylin for 2 min, dehydrated completely, cleaned in xylene and then mounted. HE and immunostaining were visualized and photographed under light microscopy (BX41 and DP70; Olympus, Tokyo, Japan).

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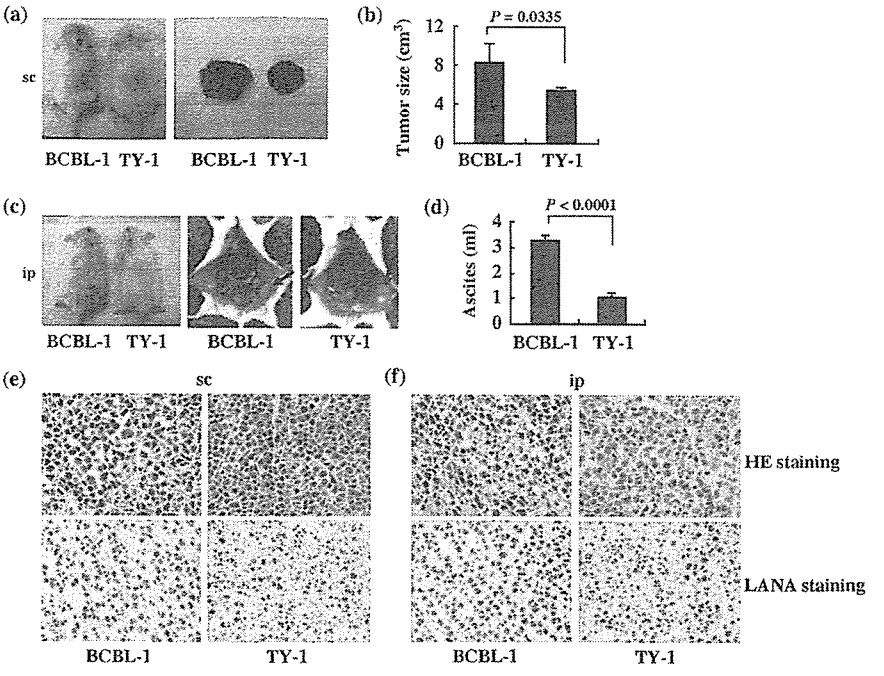

Fig. 1. Successful engraftment and tumor marker of primary effusion lymphoma (PEL) cells in T, B and natural killer (NK) knock-out NOG mice. (a) Photograph of mice inoculated with BCBL-1 and TY-1 cells subcutaneously in the postauricular region (left panel) and those of subcutaneously formed BCBL-1 and TY-1 tumor 3 weeks after inoculation of cells (right panel). (b) Subcutaneous tumor size of mice inoculated with BCBL-1 and TY-1 cells, shown as the mean ± s.e.m. from five mice ($P = 0.0335$). (c) Photograph of ascites-bearing mice inoculated with BCBL-1 and TY-1 cells intraperitonealy in the abdominal region (left panel) and peritoneal cavity of mice 21 days after inoculation of BCBL-1 (middle panel) and TY-1 cells (right panel). Arrow head indicates the tumor in mice inoculated intraperitonealy. (d) Volume of ascites in mice inoculated with various BCBL-1 and TY-1 cells, shown as the mean ± s.e.m. from five mice ($P < 0.0001$). (e,f) Hematoxylin–eosin (HE) and immunohistochemical staining of tumor tissue of BCBL-1 and TY-1 cells injected mice. Upper panels represent HE staining. Immunohistochemical staining was conducted using rabbit anti-LANA (lower panels). Left and right panels represent results with BCBL-1 and TY-1, respectively (magnification, ×40). Data are from (e) mice inoculated subcutaneously and (f) mice inoculated intraperitonealy.

Statistical analysis. The statistical analysis was carried out using StatView J-4.5 (Hulinks, Tokyo, Japan).

Results

Rapid tumor and massive ascites formation and infiltration of PEL cells in T, B and NK knock-out NOG mice. To investigate *in vivo* growth, PEL cell lines (BCBL-1 and TY-1) were inoculated sc in the postauricular region of NOG mice (Fig. 1a,b). Mice inoculated with cell lines BCBL-1 and TY-1 produced a visible tumor within 3 weeks in all NOG mice. The BCBL-1 cell line was very efficient in the formation of a large tumor (Fig. 1a,b), as well as development of clinical signs of near-death, such as pilorection, weight loss and cachexia in mice at the time of killing. The average tumor size in NOG mice inoculated with BCBL-1 and TY-1 was 8.25 cm^3 and 5.43 cm^3, respectively. PEL is an AIDS-associated non-Hodgkin's lymphoma that is characterized by lymphomatous effusions of serous cavities and rarely presents with a definable tumor mass.[5,7] To establish a clinically relevant PEL model, we inoculated BCBL-1 and TY-1 cells ip in the abdominal region of NOG mice (Fig. 1c,d). BCBL-1 and TY-1 produced massive ascites and a small tumor mass in the peritoneal cavity within 3 weeks of inoculation in all mice. The BCBL-1 cell line was most efficient in the formation of massive ascites (Fig. 1c,d), as well as development of clinical signs of near-death. The average volume of ascites in NOG mice inoculated with BCBL-1 and TY-1 was 3.26 mL and 1.05 mL, respectively. To test whether tumors maintain original histomorphology and expression patterns of tumor markers in NOG, we carried out HE and immunostaining of tumor tissues and various organs obtained from mice inoculated with BCBL-1 and TY-1 cells. Histological and immunological analysis revealed that *in vivo* tumor cells had well-preserved morphology as well as expression of the viral gene *LANA* (Fig. 1e,f). These results show that PEL cell lines inoculated either sc into the postauricular region or ip in the abdominal region of NOG mice were able to produce a large tumor and ascites very efficiently. Interestingly, ip-inoculated PEL cells were found to form clinically relevant lymphomatous effusions in the peritoneal cavity as well as a small definable mass.

To assess the tissue distribution of PEL cells, we carried out histological examinations of the different organs of NOG mice after inoculation of the cells. Infiltration of tumor cells was found not only in primary tumor tissues, but also to a lesser extent in the lung of NOG mice inoculated sc with BCBL-1 and TY-1 (Fig. 2a,b). We found that mice inoculated ip with BCBL-1 cells exhibited infiltration in the lung, liver and spleen (Fig. 2c), whereas TY-1 cells did so to a lesser extent only in the lung and liver (Fig. 2d). HE and immunohistochemical staining showed a degree of infiltration of tumor cells at the site of inoculation and various organs with BCBL-1 and TY-1 (Fig. 2). Furthermore, BCBL-1 was most efficient at infiltrating the lung (Fig. 2a,c). Interestingly, ip-inoculated PEL cells appeared to infiltrate various organs of mice more aggressively and massively than sc inoculation. This extremely rapid tumor formation and infiltration in all mice is one of the hallmarks of our clinically relevant animal model without changes in histomorphology or tumor marker expression.

Role of NK cells in the growth and infiltration of PEL cell *in vivo*. Severe combined immunodeficiency mice lack functional T and B lymphocytes, but NK-cell activity remains normal.[10,17,40] Despite severe immunological defects, SCID mice have the ability to reject xenografts. Further, immunosupression of SCID by treatment with etoposide, irradiation or an anti-NK antibody, which transiently abrogates NK-cell activity *in vivo*, results in enhanced tumor growth in mice.[41–47] To determine the possibility that NK-cell activity suppresses tumorigenesis in conventional SCID mice, the PEL cell line BCBL-1 was inoculated either sc in the postauricular region or ip in the abdominal region of T and B knock-out NOD/SCID mice with or without pretreatment of with TMβ1 antibody, or T, B and NK knock-out NOG mice (Fig. 3a–g). BCBL-1 cells were able to produce tumors at inoculation sites in NOD/SCID mice with common γ-chain. Immunosupression of NOD/SCID by treatment with an antimurine TMβ1 antibody, which transiently abrogates natural killer cell activity *in vivo*, resulted in induction of larger tumor and ascites formation in comparison with non-treated NOD/SCID mice. NOG mice lacking common γ-chain inoculated with BCBL-1 cells were most efficient in the formation of large tumor and massive ascites within 3 weeks.

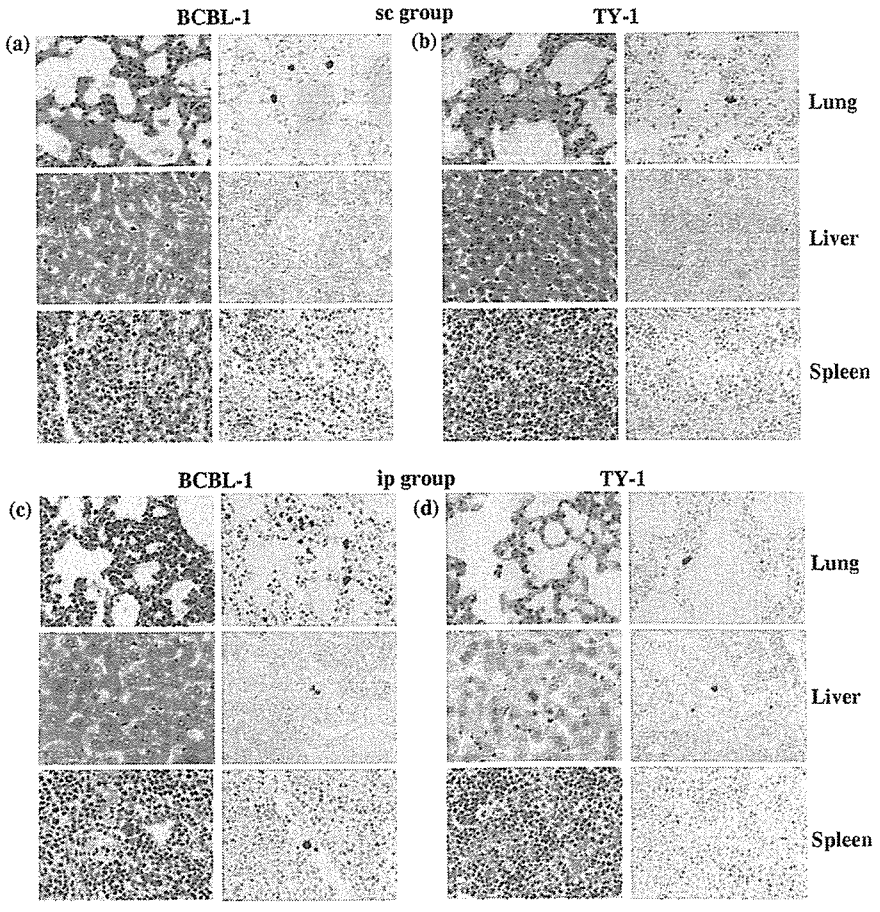

Fig. 2. Metastasis of primary effusion lymphoma (PEL) cells in various organs of T, B and natural killer (NK) knock-out NOG mice. (a–d) Histological analysis of lung, liver and spleen of mice inoculated with BCBL-1 and TY-1 cells either (a,b) subcutaneously (sc) or (c,d) intraperitonealy (ip). Immunohistochemical staining was conducted using anti-LANA. Data are from (a,c) BCBL-1-inoculated mice and (b,d) TY-1-inoculated mice. Left and right panels of all figures represent hematoxylin–eosin and immunostaining, respectively (magnification, ×40).

NOG mice have a defective common cytokine receptor, γ chain. Mutation in the common cytokine receptor γ chain leads to life-threatening, X-linked, severe combined immunodeficiency disease (XSCID) in humans, characterized by an extremely low number of T and NK cells.[48,49] These results suggest that NK cells are responsible for the formation of a progressively growing rapid large tumor and massive ascites of PEL cells in SCID mice at inoculation sites.

Severe combined immunodeficiency mice have NK cells, an important immune effector population implicated in protection against tumor metastasis and viral infection.[17,50] It has been reported recently that individuals with low natural cytotoxic activity of peripheral blood lymphocytes are at a significantly higher risk of cancer, compared with those of median or high activity, as well as functional impairment of NK cells in viral infection.[23] To assess the infiltration of PEL cells, we carried out histological examinations of tumor tissue and the different organs of mice inoculated with BCBL-1 cells (Fig. 3h). Infiltration of tumor cells was found in various organs of NOG mice inoculated with BCBL-1 cells. We found that NOD/SCID inoculated with BCBL-1 cells exhibited no infiltrate in any organs. NOD/SCID mice immunosuppressed by pretreatment with anti-NK antibody showed infiltration of PEL cells to a lesser extent in various organs of mice inoculated with BCBL-1 cells. HE and immunohistochemical staining showed a degree of infiltration of tumor cells in the lung of mice inoculated with BCBL-1 (Fig. 3h). These results suggest that NK cells play an important role in the infiltration of cancer cells in various organs.

Activated NK cells inhibit tumor growth and infiltration *in vivo*. As the above results suggested the potential role of NK cells in tumor growth and metastasis, we next examined whether adoptive transfer of activated NK cells could inhibit tumor growth and infiltration of xenografted PEL cells in the NOG mouse model. For this purpose, freshly isolated PBMC from the blood of healthy donors were cultured for 2–3 weeks to generate NK cells. NK cells were expanded *ex vivo* by several hundred to 2500-fold after 2 weeks cultivation and the expression level of CD69, an activated marker of NK cells, was increased dramatically. The purity of the activated NK cells used in the present study was 92–95% (data not shown). NK cells use cytoplasmic granules containing perforins and granzymes to kill the target cells. Using a highly sensitive flow cytometry-based intracellular cytokine assay, we next investigated the expression of intracellular perforins and granzymes in NK cells. Intracellular perforin and granzyme expression was increased in activated culture cells in comparison to freshly isolated cells from healthy donors (data not shown). PBMC, NK cell line KHYG-1 and activated NK cells were analyzed for cytotoxic activity against the NK-susceptible K562 erythroleukemia cell line (Fig. 4a,b). Cytotoxic activity of cells cultured for 2 weeks was increased significantly compared with freshly isolated PBMC from healthy donors (Fig. 4b). Activated NK cells also killed PEL cells efficiently *in vitro* at various E/T ratios, but the NK cell line KHYG-1 did not (Fig. 4c).

To examine the antitumor effect of activated NK cells against PEL, we injected the PEL cell line BCBL-1 (2×10^6) ip into the abdominal region of NOG mice. Three days after inoculation, mice were treated with either RPMI-1640 (as control) or activated NK cells (1×10^7) ip on days 4, 10 and 17. BCBL-1 cell inoculation promoted the development of massive ascites in the peritoneal cavity of all control mice within 3 weeks of inoculation. In contrast, activated NK-treated mice appeared to be

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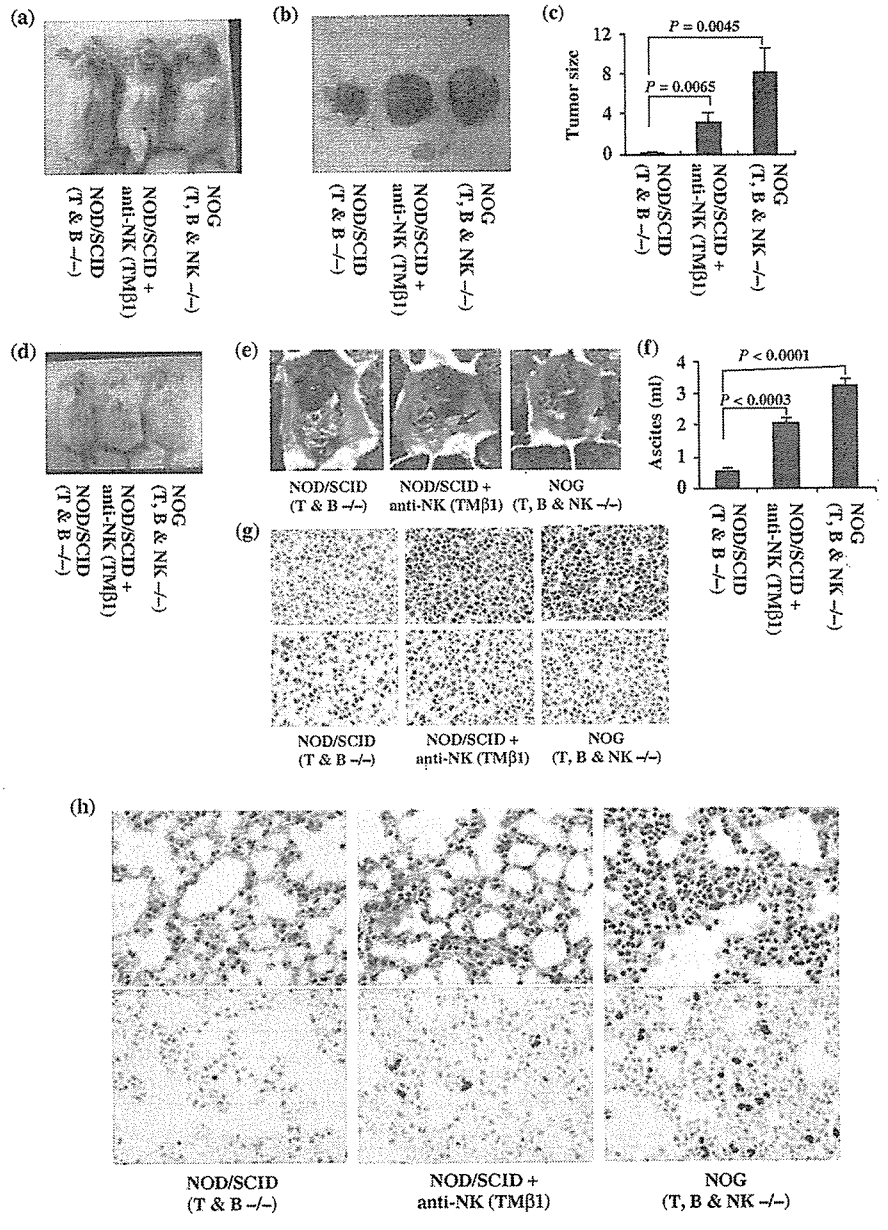

Fig. 3. Natural killer (NK) cells in tumor growth and infiltration. BCBL-1 cells were inoculated subcutaneously in the postauricular region or intraperitonealy in the abdominal region of T and B knock-out NOD/SCID, TMβ1-pretreated T and B knock-out NOD/SCID and T, B and NK knock-out NOG mice. (a) Photograph of mice inoculated with BCBL-1 cells subcutaneously in the postauricular region. (b) Photograph of BCBL-1 tumor 3 weeks formed subcutaneously after inoculation of cells. (c) Subcutaneous tumor size of mice inoculated with BCBL-1 cells, shown as the mean ± s.e.m. from six mice. Tumor size of TMβ1-pretreated NOD/SCID mice was significantly larger than NOD/SCID ($P = 0.0065$) and that of NOG mice was more significant than NOD/SCID ($P = 0.0045$). (d) Photograph of ascites-bearing mice inoculated with BCBL-1 cells intraperitonealy in the abdominal region. (e) Photograph of the peritoneal cavity of mice 3 weeks after inoculation of BCBL-1. Left, middle and right panels represent the T and B knock-out NOD/SCID, TMβ1-pretreated T and B knock-out NOD/SCID and T, B and NK knock-out NOG mice, respectively. Arrow head indicates the tumor in mice inoculated intraperitonealy. (f) Volume of ascites in mice inoculated with BCBL-1 cells, shown as the mean ± s.e.m. from six mice. Volume of ascites in TMβ1-pretreated NOD/SCID mice was significantly higher than NOD/SCID ($P = 0.0003$) and that of NOG mice was more significant than NOD/SCID ($P < 0.0001$). Hematoxylin–eosin (HE) and immunohistochemical staining of (g) lung tissue and (h) tumor tissue of BCBL-1-injected mice. Upper panels represent HE staining. Immunohistochemical staining was conducted using rabbit anti-LANA (lower panels). Left, middle and right panels represent results from T and B knock-out NOD/SCID, TMβ1-pretreated T and B knock-out NOD/SCID and T, B and NK knock-out NOG mice, respectively. Magnification, ×40.

healthy and had a significantly lower volume of ascites (Fig. 5a,b). Clinical evaluation of organ infiltration 3 weeks after injection of PEL cells showed that activated NK treatment inhibited their infiltration into the lung. In contrast, all control mice showed massive infiltration of tumor cells into the lung (Fig. 5c). Organ infiltration of tumor cells was analyzed and evaluated by HE and immunostaining of LANA. These data indicate that activated NK cells significantly inhibit the growth and infiltration of PEL cells *in vivo* (Fig. 5).

Discussion

Natural killer cells form a first line of defense against pathogens or host cells that are stressed or cancerous. To execute the concept of using activated NK cells in order to prevent cancer, it is indispensable to know how NK cells are important for tumor growth and infiltration. There have been a number of reports about the contribution of NK cells in tumor growth and metastasis. In particular, whole-body irradiation has been reported to suppress NK activity and increase the ability of human and murine tumors to be transplanted into SCID mice.[42–46] Treatment of mice with murine anti-NK antibody, which transiently inhibits NK-cell activity, results in efficient engraftment of tumor cells in SCID mice.[15,47] In the present study, we demonstrated the direct role of NK cells in tumor growth and metastasis using T, B and NK knock-out NOG and T and B knock-out NOD/SCID mice. PEL cells were able to produce a large tumor and massive ascites very efficiently at inoculated sites and infiltrate various organs in T, B and NK nock-out NOG mice. We found that T and B knock-out NOD/SCID mice inoculated with PEL cells formed small tumors and a lower volume of ascites, but completely failed to infiltrate. T and B knock-out NOD/SCID mice were further immunosuppressed by pretreatment with anti-NK antibody, which enhanced tumor and ascites formation as well as organ infiltration. These results demonstrate the critical role of NK cells in tumor growth and infiltration using NK knock-out mice. It is of particular importance that ip-inoculated PEL cells were found to form clinically relevant lymphomatous effusions in the peritoneal cavity and small tumor mass as well as infiltration. This clinically relevant